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National Rivers and Streams Assessment

Field Operations Manual

April 2009

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NOTICE

The intention of the National Rivers and Streams Assessment project is to provide a comprehensive "State of the Flowing Waters" assessment for rivers and streams across the United States. The complete documentation of overall project management, design, methods, and standards is contained in four companion documents:

- National Rivers and Streams Assessment: *Quality Assurance Project Plan (EPA-841-B-07-007)*
- National Rivers and Streams Assessment: *Site Evaluation Guidelines (EPA-841-B-07-008)*
- National Rivers and Streams Assessment: *Field Operations Manual (EPA-841-B-07-009)*
- National Rivers and Streams Assessment: *Laboratory Methods Manual (EPA-841-B-07-010)*

This document (*Field Operations Manual*) contains a brief introduction and procedures to follow at the base location and on-site, including methods for sampling water chemistry (grabs and *in situ* measurements), periphyton, benthic macroinvertebrates, sediment enzymes, fish composition, fish tissue (at non-wadeable sites), a fecal indicator, and physical habitat. These methods are based on the guidelines developed and followed in the Western Environmental Monitoring and Assessment Program (Baker, et al., 1997), the methods outlined in Concepts and Approaches for the Bioassessment of Non-wadeable Streams and Rivers (Flotemersch, et al., 2006), and methods employed by several key states that were involved in the planning phase of this project. Methods described in this document are to be used specifically in work relating to the National Rivers and Streams Assessment. All Project Cooperators must follow these guidelines. Mention of trade names or commercial products in this document does not constitute endorsement or recommendation for use. Details on specific methods for site evaluation and sample processing can be found in the appropriate companion document.

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ACRONYMS/ABBREVIATIONS

AFDM	ash-free dry mass
ANC	acid neutralizing capacity
APA	acid/alkaline phosphatase activity
CPR	cardiopulmonary resuscitation
DBH	diameter at breast height
DI	deionized
DO	dissolved oxygen
DOC	dissolved organic carbon
EMAP	Environmental Monitoring and Assessment Program
EPA	Environmental Protection Agency
ETOH	ethyl alcohol
GIS	geographic information system
GPS	global positioning device
HDPE	high density polyethylene
IBI	Index of Biotic Integrity
LWD	large woody debris
NAD	North American Datum
NAWQA	National Water-Quality Assessment Program
NHD	National Hydrography Dataset
NH_4	ammonium
NIST	National Institute of Standards
NO_3	nitrate
NRSA	National Rivers and Streams Assessment
O/E	“observed” over “expected”
OSHA	Occupational Safety and Health Administration
PFD	personal floatation device
P-Hab	physical habitat
PSI	pounds per square inch
PVC	polyvinyl chloride
QAPP	Quality Assurance Project Plan
QA/QC	quality assurance/quality control
SOPs	Standard Operating Procedures
TN	total nitrogen
TOC	total organic carbon
TP	total phosphorus
TSS	total suspended solids
USGS	United States Geological Survey
WSA	Wadeable Streams Assessment

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1.0 BACKGROUND

This manual describes field protocols and daily operations for crews to use in the National Rivers and Streams Assessment (NRSA). The NRSA is a probability-based survey of our Nation's rivers and streams and is designed to:

- Assess the condition of the Nation's rivers and streams
- Establish a baseline to compare future rivers and streams surveys for trends assessments
- Evaluate changes in condition from the 2004 Wadeable Streams Assessment
- Help build State and Tribal capacity for monitoring and assessment and promote collaboration across jurisdictional boundaries

This is one of a series of water assessments being conducted by states, tribes, the U.S. Environmental Protection Agency (EPA), and other partners. In addition to rivers and streams, the water assessments will also focus on coastal waters, lakes, and wetlands in a revolving sequence. The purpose of these assessments is to generate statistically-valid reports on the condition of our Nation's water resources and identify key stressors to these systems.

The goal of the NRSA is to address two key questions about the quality of the Nation's rivers and streams:

- What percent of the Nation's rivers and streams are in good, fair, and poor condition for key indicators of water quality, ecological health, and recreation?
- What is the relative importance of key stressors such as nutrients and pathogens?

The NRSA is designed to be completed during the index period of late May through the end of September. Field crews will collect a variety of measurements and samples from predetermined sampling locations (located with an assigned set of coordinates), and from randomized stations along the sampling reach.

1.1 Survey Design

EPA selected sampling locations using a probability based survey design. Sample surveys have been used in a variety of fields (e.g., election polls, monthly labor estimates, forest inventory analysis) to determine the status of populations or resources of interest using a representative sample of a relatively few members or sites. Using this survey design allows data from the subset of sampled sites to be applied to the larger target population, and assessments with known confidence bounds to be made.

The objectives, or design requirements, for the National Rivers and Streams Assessment are to produce:

- estimates of the 2008-2009 status of all flowing waters nationally and regionally (9 aggregated Omernik ecoregions),
- estimates of the 2008-2009 status of wadeable streams and non-wadeable rivers nationally and regionally (9 aggregated Omernik ecoregions),
- estimates of the 2008-2009 status of urban flowing waters nationally, and

- estimates of the change in status in wadeable streams between 2008-2009 and 2004, nationally and regionally (nine aggregated Omernik ecoregions).

With input from the states and other partners, EPA used an unequal probability design to select 900 wadeable streams and 900 non-wadeable rivers. For purposes of this study, a wadeable stream segment is defined being >50% wadeable; if it is <50% wadeable, it is defined as non-wadeable. To evaluate change in wadeable streams from the 2004 WSA, 450 of the 900 wadeable sites were selected using an unequal probability design from the WSA original sites. The result was the selection of 1800 river and stream sites, with approximately 10%, or 200, of these sites scheduled for revisits. **The NRSA design is explicitly stratified by state.** An "oversample" of additional sites also is available so that any state wishing to conduct a state scale assessment could be accommodated.

1.1.1 Target Population and Sample Frame

The target population consists of all streams and rivers within the 48 contiguous states that have flowing water during the study index period excluding portions of tidal rivers up to head of salt. The study index period extends from late May to the end of September and is characterized by base flow conditions. The target population includes the Great Rivers. Run-of-the-river ponds and pools with a residency time of less than 7 days, are included while reservoirs are excluded. Tidal freshwater rivers and streams are included above the head of salt. For the purposes of this study the head of salt is < .05ppt. Please refer to the Site Evaluation Guidelines *(EPA-841-B-07-008)* and the NRSA Web site (http://www.epa.gov/owow/riverssurvey/index.html) for more detailed information on the target population.

The sample frame was derived from the National Hydrography Dataset, NHD-Plus, from 1:100,000 scale maps. Attributes that are used in the NRSA design include:

- State
- EPA Region
- NAWQA Mega Region
- Omernik Ecoregion Level 3
- WSA aggregated ecoregions
- Strahler order (1st through 8th+)
- Strahler order categories
- Urban (site is within "urban" boundary)

1.1.2 Replacing Sites

Sites are organized to be replaced within each state. If a stream or river site is evaluated and it is determined that it cannot be sampled, then it is to be replaced by another site within the state. Sites that are coded as 1st through 4th order are to be replaced by oversample sites that are coded 1st through 4th order, ignoring order **within this range**. For example, a 2nd order stream would be replaced by the next 1st, 2nd, 3rd or 4th order stream on the state list. Sites that are coded as 5th through 10th order are to be replaced by oversample sites that are coded 5th through 10th order, again ignoring order within the range. For example, a 5th order river would be replaced by the next 5th, 6th, 7th, 8th, 9th, or 10th order river on the state list. In each case the **next site** that is **within the Strahler order range** is used for the replacement. Please refer to the *Site Evaluation Guidelines (EPA-841-B-07-008)* for more detailed information.

1.2 Selection of NRSA Indicators

As part of the indicator selection process, EPA worked with state and tribal partners and technical expert consultants through technical conferences and indicator workgroup teleconferences. The Agency formed a National Rivers and Streams Assessment Steering Committee with state and regional representatives to develop and refine methodologies. This section summarizes the Steering Committee recommendations to EPA for selecting NRSA indicators.

The EPA and partners developed screening and evaluation criteria and identified potential indicators based on recommendations received at the Large Rivers Assessment Planning Meeting in San Antonio, Texas (January 10-12, 2007), and the National Rivers and Streams Planning Session held in Washington, D.C, (April 12, 2007). Key screening and evaluation criteria included indicator applicability on a national scale, the ability of an indicator to reflect various aspects of ecological condition, repeatability, and cost-effectiveness.

Participants in indicator discussions included partners and consultants with a technical background in water monitoring program design and execution, as well as those with knowledge of state and regional water monitoring programs. Workgroup participants provided feedback on indicators, field protocols, and analytical procedures for the NRSA. EPA, states, tribes, and others discussed approaches and options on the chemical, physical, and biological parameters to be measured. Participants explored the technical and budgetary feasibility of sampling and analysis methods, the use of specialized technologies (e.g., remote sensing), practical considerations for completing the assessment (e.g., use of volunteers, availability of labs, timeframes, funding), and emerging pollutants and contaminant issues.

The remainder of this section briefly describes the indicators that will be used for the NRSA to assess water quality, ecological integrity, recreational value, and site characteristics (also see Table 1-1 and Table 1.2).

1.3 Description of NRSA Indicators

In Situ Water Quality Measurements

Measurements for temperature, pH, dissolved oxygen (DO), and conductivity will be taken with a calibrated water quality probe meter or multi-probe sonde at the X-site (center) transect in each river or stream. This information will be used to detect extremes in condition that might indicate impairment.

Secchi Disk Transparency

A Secchi disk is a commonly used black and white patterned disk used to measure the clarity of water in visibility distance.

Water Chemistry and Associated Measurements

Water chemistry measurements will be used to determine the acidic conditions and nutrient enrichment, as well as classification of water chemistry type.

Sediment Enzymes

Benthic organisms are in intimate contact with river sediments, and they are influenced by the physical and chemical properties of the sediment. Sediment enzyme activity serves as a functional indicator of key ecosystem processes. Analytical tests include DIN, DIC, TP and TN.

Chlorophyll a

Chlorophyll a is the pigment that makes plants and algae green. Its measurement is used to determine algal biomass in the water.

Periphyton Assemblage

Periphyton are diatoms and soft-bodied algae that are attached or otherwise associated with channel substrates. They can contribute to the physical stability of inorganic substrate particles, and provide habitat and structure. Periphyton are useful indicators of environmental condition because they respond rapidly and are sensitive to a number of anthropogenic disturbances, including habitat destruction, contamination by nutrients, metals, herbicides, hydrocarbons, and acidification.

Benthic Macroinvertebrate Assemblage

Benthic macroinvertebrates are bottom-dwelling animals without backbones ("invertebrates") that are large enough to be seen with the naked eye ("macro"). Examples of macroinvertebrates include: crayfish, snails, clams, aquatic worms, leeches, and the larval and nymph stages of many insects, including dragonflies, mosquitoes, and mayflies. Populations in the benthic assemblage respond to a wide array of stressors in different ways so that it is often possible to determine the type of stress that has affected a macroinvertebrate assemblage (Klemm et al., 1990). Because many macroinvertebrates have relatively long life cycles of a year or more and are relatively immobile, the structure of the macroinvertebrate assemblage is a response to exposure of present or past conditions.

Fish Assemblage

Monitoring of the fish assemblage is an integral component of many water quality management programs. The assessment will measure specific attributes of the overall structure of the ichthyofaunal community to evaluate biological integrity and water quality.

Physical Habitat Assessment

The physical habitat assessment of the sampling reach and the riparian zone (the region lying along a bank) will serve three purposes. First, habitat information is essential to the interpretation of what ecological condition is expected to be like in the absence of many types of anthropogenic impacts. Second, the habitat evaluation is a reproducible, quantified estimate of habitat condition, serving as a benchmark against which to compare future habitat changes that might result from anthropogenic activities. Third, the specific selections of habitat information collected aid in the diagnosis of probable causes of ecological degradation in rivers and streams. For example, some of the data collected will be used to calculate relative bed stability (RBS). RBS is an estimate of stream stability that is calculated by comparing the mean sediment size present to the sediment size predicted by channel and slope.

In addition to information collected in the field by the physical habitat assessment, the physical habitat description of each site includes many map-derived variables such as stream order and drainage area. Furthermore, an array of information, including watershed topography and land use, supplements the physical habitat information. Together with water chemistry, the habitat measurements and observations describe the variety of physical and chemical conditions that are necessary to support biological diversity and foster long-term ecosystem stability.

Fecal Indicator (Enterococci)

Enterococci are bacteria that are endemic to the guts of warm blooded creatures. These bacteria, by themselves, are not considered harmful to humans but often occur in the presence of potential human pathogens (the definition of an indicator organism). Epidemiological studies of marine and fresh water bathing beaches have established a direct relationship between the density of *enterococci* in water and the occurrence of swimming-associated gastroenteritis.

Fish Tissue

The fish tissue contaminants indicator, which measures bioaccumulation of persistent toxics, is used to estimate national risks of fish consumption to humans. Various studies have been done on fish tissue contaminants focusing on different parts of the fish (e.g., whole fish, fillets, livers). The NRSA will focus on fillets because of its emphasis on human health.

Other Indicators / Site Characteristics

Pharmaceuticals and Personal Care Products (PPCP) will be sampled from fish tissue and water column at 154 pre-selected sites. These sites are defined as urban, boatable sites and will have an additional water grab taken to look at these emerging contaminants. Observations and impressions about the site and its surrounding catchment by field teams will be useful for ecological value assessment, development of associations and stressor indicators, and data verification and validation.

Table 1-1. Summary table of indicators for non-wadeable sites

Indicator	Specs/Location in Sampling Reach
In Situ measurements (pH, DO, temperature, conductivity)	Measurements taken at X site at midchannel; readings are taken at 0.5 m depth
Water chemistry (TP, TN [NH_4, NO_3), basic anions and cations, alkalinity [ANC], DOC, TOC, TSS, conductivity	Collected from a depth of 0.5 m at the cross site at the center of the stream
Secchi Disk transparency	Measured at X site at midchannel
Chlorophyll a	Collected as part of water chemistry and periphyton samples
Sediment enzymes	Collected from 11 locations systematically placed at each site and combined into a single composite sample
Periphyton	Collected from 11 locations systematically placed at each site and combined into a single composite sample
Benthic macroinvertebrate assemblage (Littoral)	Collected from 11 locations systematically placed at each site and combined into a single composite sample
Fish Assemblage	Sampled throughout the sampling reach at specified locations
Physical habitat assessment	Measurements collected throughout the sampling reach at specified locations
Fecal indicator (*enterococci*)	Collected at the last transect one meter off the bank
Fish Tissue	Target species collected throughout the sampling reach as part of fish assemblage sampling
Drainage area	Done at desktop, and used in target population selection
Characteristics of watershed	Done at desktop using GIS and verified by state agencies
PPCP (Only at pre-defined urban sites)	Collected only at specified sites at the X site

Table 1-2. Summary table of indicators for wadeable sites

Indicator	Specs/Location in Sampling Reach
In Situ measurements (pH, DO, temperature, conductivity)	One set of measurements taken at the X site in the center of the stream; readings are taken at 0.5 m depth
Water chemistry (TP, TN [NH_4, NO_3), basic anions and cations, alkalinity [ANC], DOC, TOC, TSS, conductivity	Collected from a depth of 0.5 m at the X site at the center of the stream
Chlorophyll a	Collected as part of water chemistry and periphyton samples
Sediment enzymes	Collected from 11 locations systematically placed at each site and combined into a single composite sample
Periphyton	Collected from 11 locations systematically placed at each site and combined into a single composite sample
Benthic macroinvertebrate assemblage (Littoral)	Collected from 11 locations systematically placed at each site and combined into a single composite sample
Fish Assemblage	Sampled throughout the sampling reach at specified locations
Physical habitat assessment	Measurements collected throughout the sampling reach at specified locations
Fecal indicator (*enterococci*)	Collected at the last transect one meter off the bank
Drainage area	Done at desktop, and used in target population selection
Characteristics of watershed	Done at desktop using GIS and verified by state agencies

1.4 Supplemental Material to the Field Operations Manual

The Field Operations Manual describes field protocols and daily operations for crews to use in the NRSA. Following these detailed protocols will ensure consistency across regions and reproducibility for future assessments. Before beginning sampling at a site, crews should prepare a packet for each site containing pertinent information to successfully conduct sampling. This includes a road map and set of directions to the site, topographic maps, land owner access forms, sampling permits (if needed), site evaluation forms and other information necessary to ensure an efficient and safe sampling day.

Field crews will also receive a quick-reference handbook that contains tables and figures summarizing field activities and protocols from the Field Operations Manual. This waterproof handbook will be the primary field reference used by field teams after completing the required field training session. The field teams are also required to keep the field operations manual available in the field for reference and for possible protocol clarification.

Large-scale and/or long-term monitoring programs such as those envisioned for national surveys and assessments require a rigorous QA program that can be implemented consistently by all participants throughout the duration of the monitoring period. Quality assurance is a required element of all EPA-sponsored studies that involve the collection of environmental data (USEPA 2000a, 2000b). Field teams will be provided a copy of the integrated Quality Assurance

and Project Plan (QAPP). The QAPP contains more detailed information regarding QA/QC activities and procedures associated with general field operations, sample collection, measurement data collection for specific indicators, and data reporting activities. For more information on the Quality Assurance procedures, refer to the *National Rivers and Streams Assessment: Quality Assurance Project Plan (EPA 841-B-07-007).*

Related NRSA documents include the following: National Rivers and Streams Assessment: Quality Assurance Project Plan (EPA 841-B-07-007), National Rivers and Streams Assessment: Site Evaluation Guidelines (EPA 841-B-07-008), and National Rivers and Streams Assessment: Laboratory Methods Manual (EPA 841-B-07-010 or 841-B-07-011). These documents are available at: http://www.epa.gov/owow/riverssurvey/index.html.

2.0 DAILY OPERATIONS SUMMARY

This Field Operations Manual will be used for sampling at both wadeable and non-wadeable sites. The same indicators will be collected (with the exception of Secchi transparency and fish tissue, which are only collected at non-wadeable sites), but the sampling will be conducted with different protocols and equipment. This section presents a general overview of the activities that a field team is to conduct during a typical 1-day sampling visit to a site, whether wadeable or non-wadeable. General guidelines for recording data using standardized field data forms and sample labels are also presented. Finally, safety and health considerations and guidelines related to field operations are described.

2.1 Sampling Scenario

The Field methods for the NRSA are designed to be completed in one field day for most sites. Depending on the time needed for both the sampling and travel for the day, an additional day may be needed to complete sampling or for pre-departure and post-sampling activities (e.g., cleaning equipment, repairing gear, shipping samples, and traveling to the next site). Remote sites with lengthy or difficult approaches may require more time, and field crews will need to plan accordingly.

Each field team should define roles and responsibilities for each team member to organize field activities efficiently. Minor modifications to the sampling scenario may be made by teams; however the ***sequence of sampling events presented in the Figures 2-1 and 2-2 cannot be changed and is based on the need to protect some types of samples from potential contamination and to minimize holding times once samples are collected***.

2.1.1 Non-wadeable Sites

A field crew for a non-wadeable field team typically will consist of four or five people in 2 boats. A minimum of two people are always required in a boat together to execute the sampling activities and to ensure safety. Typically, in non-wadeable sites, two crew members will work in the “habitat” boat, and two or three will work in the “fish” boat. One crew member on each boat is primarily responsible for boat operation and navigation. Any additional team members may either help collect samples, or may remain on the bank to provide logistical support. A daily field sampling scenario showing how the work load may be split between team members is presented in Figure 2-1. The following sections further define the sampling sequence and the protocols for sampling activities.

2.1.2 Wadeable Sites

A field crew for wadeable sites will typically consist of four people. Any additional team members may either help collect samples, or may remain on the bank to provide logistical support. A daily field sampling scenario showing how the work load may be split between team members is presented in Figure 2-2. The following sections further define the sampling sequence and the protocols for sampling activities.

The field team arrives at the site in the early morning to complete the sampling in a single day. The sampling sequence is to:

- verify site and locate x-site (whole crew),

Divide into 2 groups and:

- conduct *in situ* measurements of dissolved oxygen, pH, temperature, and conductivity
- take Secchi disk transparency depth measurements at non-wadeable sites,
- collect water chemistry and *chlorophyll a,*
- conduct physical habitat characterization,
- collect periphyton samples,
- collect benthic samples,
- collect sediment enzyme samples,
- collect fish samples,
- collect fish tissue samples at non-wadeable sites,
- collect fecal indicator sample,
- filter fecal indicator, *chlorophyll a,* and periphyton samples,
- preserve and prepare all samples for shipment,
- review field forms,
- report sampling event,
- ship time-sensitive samples.

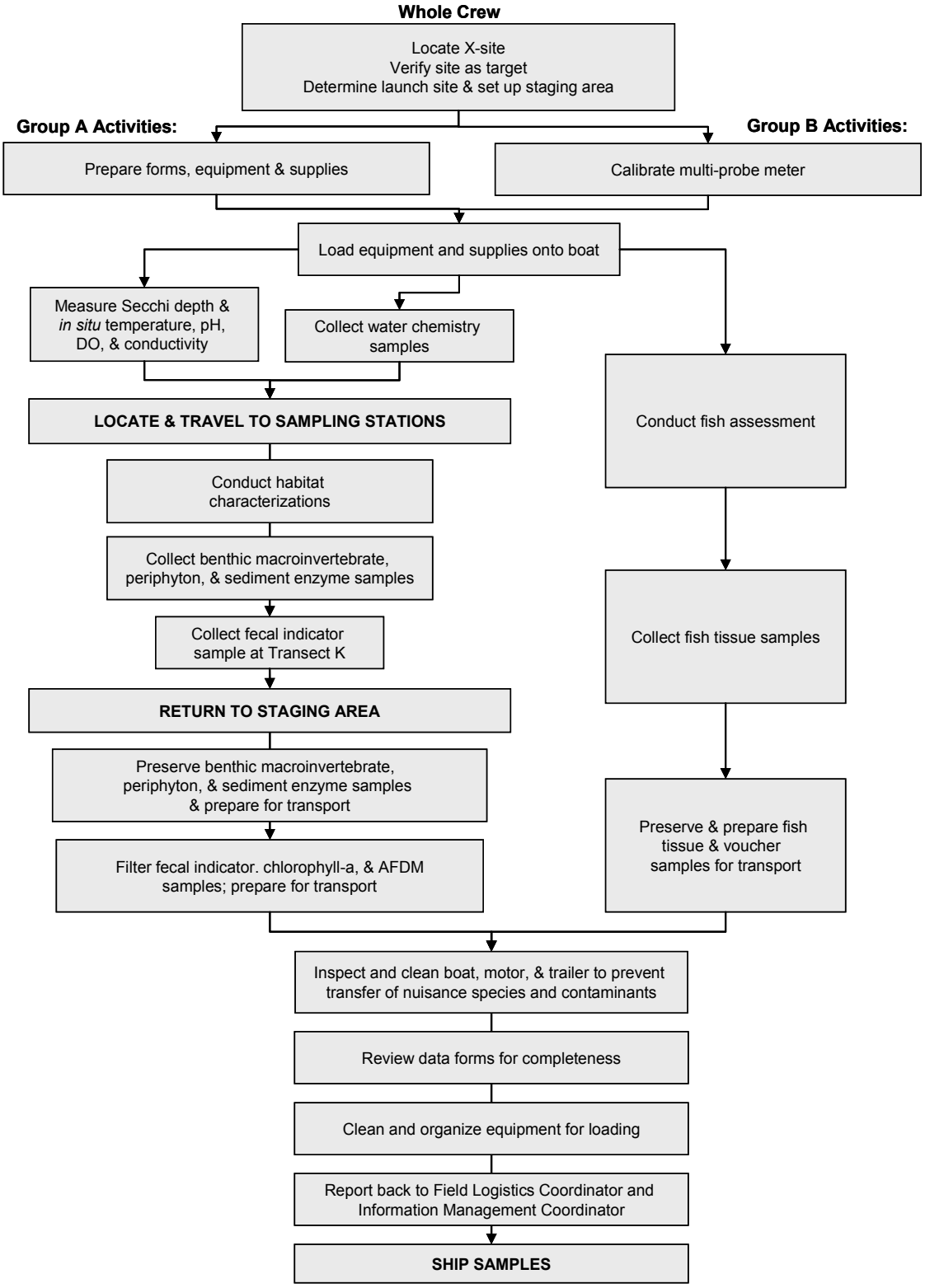

Figure 2-1. Field sampling scenario for non-wadeable sites.

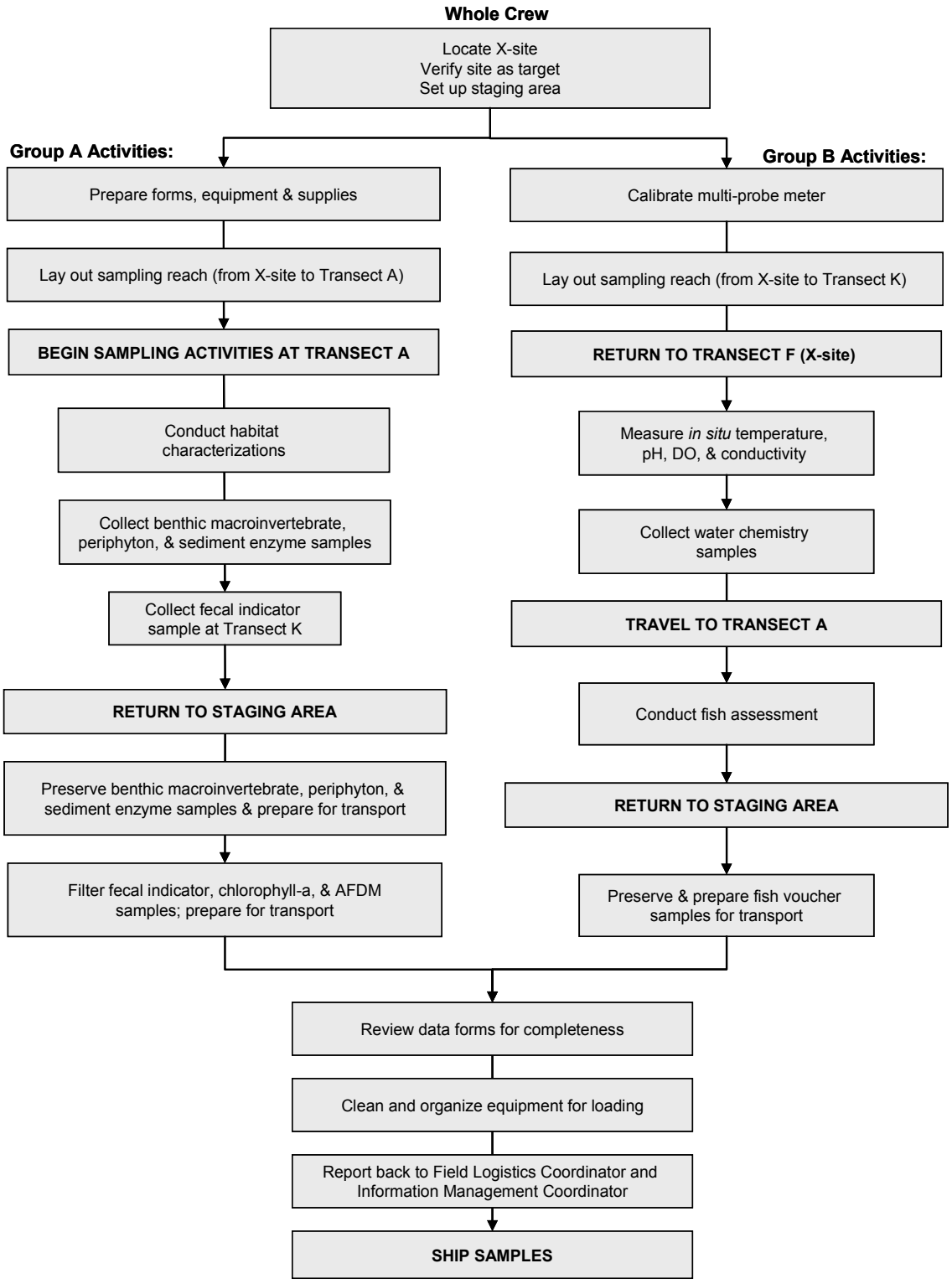

Figure 2-2. Field sampling scenario for wadeable sites.

2.2 Recording Data and Other Information

All samples need to be identified and tracked, and associated information for each sample must be recorded. To assist with sample identification and tracking, labels are preprinted with sample ID numbers (Figure 2-3).

Figure 2-3. Example sample labels for sample tracking and identification.

It is imperative that field and sample information be **recorded accurately, consistently, and legibly**. The cost of a sampling visit coupled with the short index period severely limits the ability to resample a site if the initial information recorded was inaccurate or illegible. Guidelines for recording field measurements are presented in Table 2-1.

Table 2-1. Guidelines for recording field measurements and tracking information

Activity	Guidelines
Field Measurements	
Data Recording	Record measurement values and observations on data forms preprinted on water-resistant paper. Use No. 2 pencil only (fine-point indelible markers can be used if necessary) to record information on forms. Record data and information using correct format as provided on data forms. Be sure to accurately record site IDs and sample numbers. For revisit samples use (*site ID*)-R to indicate the samples are from revisit sites. For duplicate samples, use (site ID)-D to indicate the samples are duplicates. Print legibly (and as large as possible). Clearly distinguish letters from numbers (e.g., 0 versus O, 2 versus Z, 7 versus T or F, etc.), but do not use slashes. In cases where information is recorded repeatedly on a series of lines (e.g., physical habitat characteristics), do not use "ditto marks" (") or a straight vertical line. Record the information that is repeated on the first and last lines, and then connect these using a wavy vertical line. When recording comments, print or write **legibly**. Make notations in comments field only; avoid marginal notes. Be concise, but avoid using abbreviations or "shorthand" notations. If you run out of space, attach a sheet of paper with the additional information, rather than trying to squeeze everything into the space provided on the form.
Data Qualifiers (Flags)	Use only defined flag codes and record on data form in appropriate field. K = Measurement not attempted or not recorded. Q = Failed quality control check; remeasurement not possible. U = Suspect measurement; remeasurement not possible. F*n* = Miscellaneous flags (*n* = 1, 2, etc.) assigned by a field team during a particular sampling visit (also used for qualifying samples). Explain reason for using each flag in comments section on data form.
Sample Labels	Use adhesive labels with preprinted ID numbers and follow the standard recording format for each type of sample. Use a pencil to record information on label. Cover the completed label with clear tape. Record sample ID number from label and associated collection information on sample collection form preprinted on water-resistant paper.

Activity	Guidelines
Sample Collection and Tracking	
Sample Qualifiers (Flags)	Use only defined flag codes and record on sample collection form in appropriate field. K = Sample not collected or lost before shipment; resampling not possible. U = Suspect sample (e.g., possible contamination, does not meet minimum acceptability requirements, or collected by non-standard procedure). F*n* = Miscellaneous flags (*n*=1, 2, etc.) assigned by a field team during a particular sampling visit (also used for field measurements). Explain reason for using flags in "Comments" on sample collection form.
Review of Labels and Data Collection Forms	Compare information recorded on labels and sample collection form for accuracy before leaving site. Review labels and data collection forms for accuracy, completeness, and legibility before leaving site. The Field Team Leader must review all labels and data collection forms for consistency, correctness, and legibility before transfer to the Information Management Center.

2.3 Safety and Health

Collection and analysis of samples can involve significant risks to personal safety and health. This section describes recommended training, communications, and safety considerations, safety equipment and facilities, and safety guidelines for field operations.

2.3.1 General Considerations

Important considerations related to field safety are presented in Table 2-2. It is the responsibility of the state or contractor project leader to ensure that the necessary safety courses are taken by all field personnel and that all safety policies and procedures are followed. Please follow your own agency's health and safety protocols, or refer to the *Health and Safety Guidance for Field Sampling: National Rivers and Streams Assessment* (available from the EPA Regional Coordinator) and *Logistics of Ecological Sampling on Large Rivers (*Flotemersch, et al. (editors) 2000). Additional sources of information regarding safety-related training include the American Red Cross (1979), the National Institute for Occupational Safety and Health (1981), U.S. Coast Guard (1987) and Ohio EPA (1990).

Field crew members should become familiar with the hazards involved with sampling equipment and establish appropriate safety practices prior to using them. Make sure all equipment is in safe working condition. Personnel must consider and prepare for hazards associated with the operation of motor vehicles, boats, winches, tools, and other incidental equipment. Boat operators should meet any state requirements for boat operation and be familiar with U.S. Coast Guard rules and regulations for safe boating contained in a pamphlet, "*Federal Requirements for Recreational Boats*," available from a local U.S. Coast Guard

Director or Auxiliary or State Boating Official (U.S. Coast Guard, 1987). Life jackets must be worn by crew members at all times on the water. All boats with motors must have fire extinguishers, boat horns, life jackets or flotation cushions, and flares or communication devices. Boats should stay in visual contact with each other, and should use 2-way radios to communicate.

Primary responsibility for safety while electrofishing rests with the crew chief. Electrofishing units may deliver a fatal electrical shock, and should only be used by qualified, experienced operators. Field crew members using electrofishing equipment must be insulated from the water, boat, and electrodes via rubber boots and linesman gloves. Use chest waders with nonslip soles and linesman gloves. DO NOT wear breathable waders while electrofishing. If waders become wet inside, stop fishing until they are thoroughly dry or use a dry pair. Avoid contact with the anode and cathode at all times due to the potential shock hazard. If you perspire heavily, wear polypropylene or some other wicking and insulating clothing instead of cotton. If it is necessary for a team member to reach into the water to pick up a fish or something that has been dropped, do so only after the electrical current is off and the anode is removed from the water. Do not resume electrofishing until all individuals are clear of the electroshock hazard. The backpack electrofishing equipment is equipped with a 45° tilt switch that interrupts the current. Do not make any modifications to the electrofishing unit that would hinder this safety switch. Avoid electrofishing near unprotected people, pets, or livestock. Discontinue activity during thunderstorms or rain. Team members should keep each other in constant view or communication while electrofishing. For each site, know the location of the nearest emergency care facility. Although the team leader has authority, each team member has the responsibility to question and modify an operation or decline participation if it is unsafe.

Table 2-2. General health and safety considerations.

Recommended Training
▪ First aid and cardiopulmonary resuscitation (CPR)
▪ Vehicle safety (e.g., operation of 4-wheel drive vehicles)
▪ Boating and water safety; whitewater safety if applicable
▪ Field safety (weather, personal safety, orienteering, site reconnaissance of prior to sampling
▪ Equipment design, operation, and maintenance
▪ Handling of chemicals and other hazardous materials
Communications
▪ Check-in schedule
▪ Sampling itinerary (vehicle used & description, time of departure & return, travel route)
▪ Contacts for police, ambulance, hospitals, fire departments, search and rescue personnel
▪ Emergency services available near each sampling site and base location
▪ Cell (or satellite) phone and VHF radio if possible
Personal Safety
▪ Field clothing and other protective gear including lifejackets for all team members
▪ Medical and personal information (allergies, personal health conditions)
▪ Personal contacts (family, telephone numbers, etc.)
▪ Physical exams and immunizations

A communications plan to address safety and emergency situations is essential. All field personnel need to be fully aware of all lines of communication. Field personnel should have a daily check-in procedure for safety. An emergency communications plan should include contacts for police, ambulance, fire departments, hospitals, and search and rescue personnel.

Proper field clothing should be worn to prevent hypothermia, heat exhaustion, sunstroke, drowning, or other dangers. Field personnel must be able to swim, and personal flotation devices must be used. Chest waders made of rubberized or neoprene material must always be worn with a belt to prevent them from filling with water in case of a fall. A personal flotation device (PDF) and suitable footwear must be worn at all times while on board a boat.

Many hazards lie out of sight in the bottoms of rivers and streams. Broken glass or sharp pieces of metal embedded in the substrate can cause serious injury if care is not exercised when walking or working with the hands in such environments. Infectious agents and toxic substances that can be absorbed through the skin or inhaled may also be present in the water or sediment. Personnel who may be exposed to water known or suspected to contain human or animal wastes that carry causative agents or pathogens must be immunized against tetanus, hepatitis, typhoid fever, and polio. Biological wastes can also be a threat in the form of viruses, bacteria, rickettsia, fungi, or parasites.

2.3.2 Safety Equipment

Appropriate safety apparel such as waders, linesman gloves, safety glasses, etc. must be available and used when necessary. First aid kits, fire extinguishers, and blankets must be readily available in the field. Cellular or satellite telephones and/or portable radios should be provided to field teams working in remote areas in case of an emergency. Supplies (e.g., clean water, anti-bacterial soap, ethyl alcohol) must be available for cleaning exposed body parts that may have been contaminated by pollutants in the water.

2.3.3 Safety Guidelines for Field Operations

General safety guidelines for field operations are presented in Table 2-3. Personnel participating in field activities should be in sound physical condition and have a physical examination annually or in accordance with organizational requirements. All surface waters and sediments should be considered potential health hazards due to potential toxic substances or pathogens. Persons must become familiar with the health hazards associated with using chemical fixing and/or preserving agents. Chemical wastes can be hazardous due to flammability, explosiveness, toxicity, causticity, or chemical reactivity. All chemical wastes must be discarded according to standardized health and hazards procedures (e.g., National Institute for Occupational Safety and Health [1981]; U.S. EPA [1986]).

During the course of field research activities, field teams may observe violations of environmental regulations, may discover improperly disposed hazardous materials, or may observe or be involved with an accidental spill or release of hazardous materials. In such cases it is important that the proper actions be taken and that field personnel do not expose themselves to something harmful. The following guidelines should be applied:

First and foremost, protect the health and safety of all personnel. Take necessary steps to avoid injury or exposure to hazardous materials. If you have been trained to take action such as cleaning up a minor fuel spill during fueling of a boat, do it. However, you should always err on the side of personal safety.

Field personnel should never disturb or retrieve improperly disposed hazardous materials from the field to bring back to a facility for “disposal”. To do so may worsen the impact, incur personal liability for the team members and/or their respective organizations, cause personal injury, or cause unbudgeted expenditure of time and money for proper treatment and disposal of the material. Notify the appropriate authorities so they may properly respond to the incident.
For most environmental incidents, the following emergency telephone numbers should be provided to all field teams: State or Tribal department of environmental quality or protection, U.S. Coast Guard, and the U.S. EPA regional office. In the event of a major environmental incident, the National Response Center may need to be notified at 1-800-424-8802.

Table 2-3. General safety guidelines for field operations

- Two crew members must be present during all sample collection activities, and no one should be left alone while in the field. Boats should proceed together down the river.
- Use caution when sampling in swift or deep water. Wear a suitable PFD and consider using a safety tether held by an assistant.
- Use extreme care walking on riprap. Rocks can shift unexpectedly and serious falls are possible.
- Field crew members using electrofishing equipment must be insulated from the water, boat, and electrodes via non-breathable waders and linesman gloves. Use chest waders with nonslip soles.
- Electrofishing units may deliver a fatal electrical shock, and should only be used by qualified, experienced operators.
- Do not attempt to collect samples from vertical or near vertical banks.
- Professional-quality breathable waders with a belt are recommended for littoral sampling only, and at a safe distance from the electrofishing sampling. Neoprene boots are an alternative, but should have sturdy, puncture-resistant soles.
- Use caution using the Ponar-type samplers. The jaws are sharp and may close unexpectedly.
- Exposure to water and sediments should be minimized as much as possible. Use gloves if necessary, and clean exposed body parts as soon as possible after contact.
- All electrical equipment must bear the approval seal of Underwriters Laboratories and must be properly grounded to protect against electric shock.
- Use heavy gloves when hands are used to agitate the substrate during collection of benthic macroinvertebrate samples.
- Use appropriate protective equipment (e.g., gloves, safety glasses) when handling and using hazardous chemicals.
- Crews working in areas with poisonous snakes must check with the local Drug and Poison Control Center for recommendations on what should be done in case of a bite from a poisonous snake.
- Any person allergic to bee stings, other insect bites, or plants (i.e., poison ivy, oak, sumac, etc.) must take proper precautions and have any needed medications handy.
- Field personnel should also protect themselves against deer or wood ticks because of the potential risk of acquiring pathogens that cause Rocky Mountain spotted fever and Lyme disease.
- Field personnel should be familiar with the symptoms of hypothermia and know what to do in case symptoms occur. Hypothermia can kill a person at temperatures much above freezing (up to 10^{o}C or 50^{o}F) if he or she is exposed to wind or becomes wet.
- Field personnel should be familiar with the symptoms of heat/sun stroke and be prepared to move a suffering individual into cooler surroundings and hydrate immediately.
- Handle and dispose of chemical wastes properly. Do not dispose any chemicals in the field.

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3.0 BASE SITE ACTIVITIES

Field teams conduct a number of activities at their base site (i.e., office or laboratory, camping site, or motel). These include tasks that must be completed both before departure to the site and after return from the field (Figure 3-1). Close attention to these activities is required to ensure that the field teams know (1) where they are going, (2) that access is permissible and possible, (3) that equipment and supplies are available and in good working order to complete the sampling effort, and (4) that samples are packed and shipped appropriately.

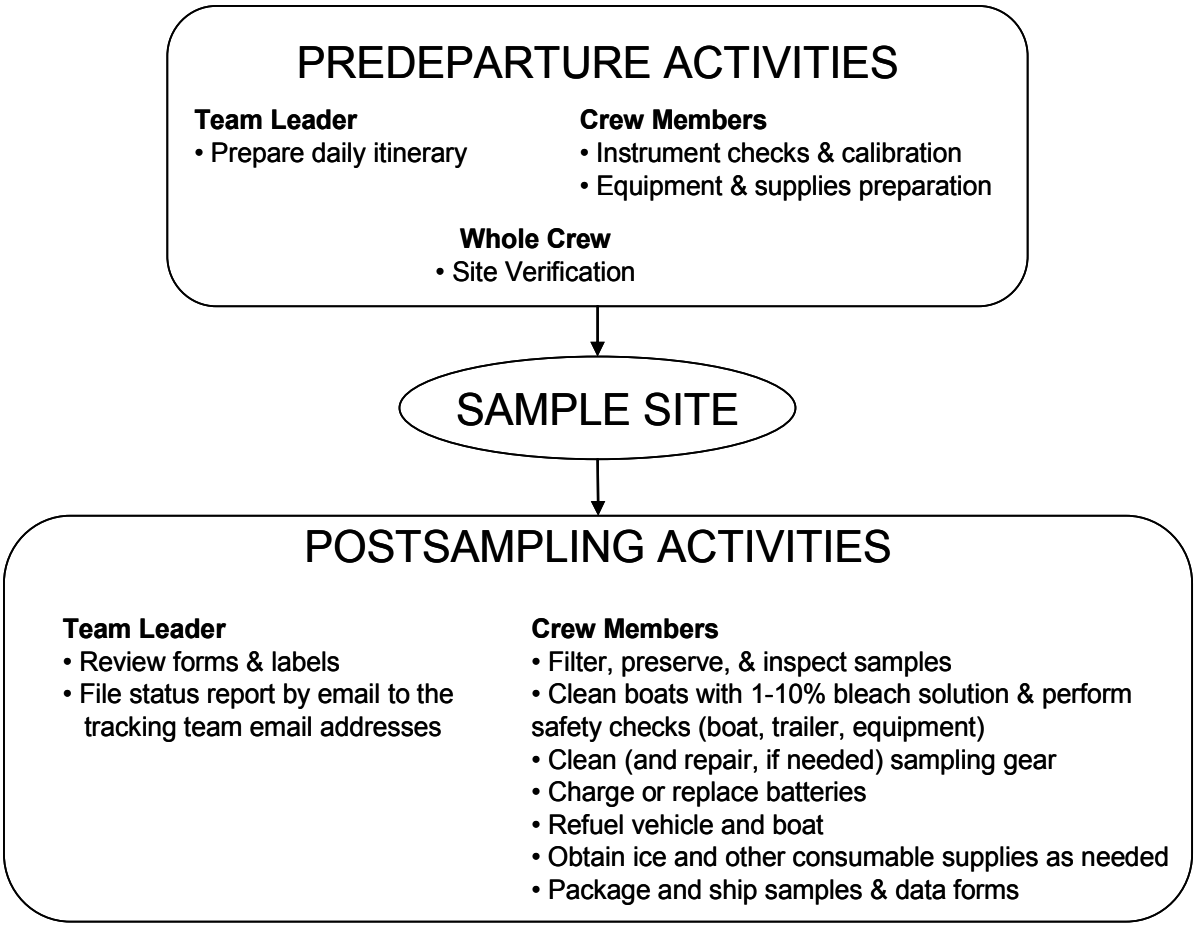


Figure 3-1. Overview of base site activities.

3.1 Predeparture Activities

Predeparture activities include the development of a daily itinerary, instrument checks and calibration, and equipment and supply preparation. Procedures for these activities are described in the following sections.

3.1.1 Daily Itineraries

The Field Team Leaders are responsible for developing daily itineraries. This entails compiling maps, contact information, copies of permission letters, and access instructions (a "site packet"). Additional activities include confirming the best access routes, calling the landowners or local contacts, confirming lodging plans, and coordinating rendezvous locations with individuals who must meet with field teams prior to accessing a site. Changes in the itinerary during the week, such as canceling a sampling day, must be relayed by the crew leader to the Field Logistics Coordinator as soon as possible.

3.1.2 Instrument Checks and Calibration

Each field team must test and calibrate instruments prior to sampling. Calibration can be conducted prior to departure for the site or at the site, with the exception of dissolved oxygen (DO) calibration. Because of the potential influence of altitude, DO calibration is to be performed only at the site. Field instruments include a global positioning system (GPS) receiver, a multiprobe unit for measuring DO, pH, temperature, and conductivity, and electrofishing equipment. Field teams should have access to backup instruments if any instruments fail the manufacturer performance tests or calibrations. Prior to departure, field teams must:

- Turn on the GPS receiver and check the batteries. Replace batteries immediately if a battery warning is displayed.

- Test and calibrate the multi-probe meter. Each field team should have a copy of the manufacturer's calibration and maintenance procedures. All meters should be calibrated according to manufacturer specifications provided along with the meter. Once a week, crews should check their multiprobe against the provided Quality Check Solution. This QCS is provided to all crews in their base kits and is used to check pH and conductivity measurements.

- Turn on the electrofishing unit and check the batteries. Be sure to have fully charged backup batteries. If using a gas powered electrofishing unit, check the oil and gas supply.

3.1.3 Equipment and Supply Preparation

Field teams must check the inventory of supplies and equipment prior to departure using the equipment and supplies checklists provided in Appendix A; use of the lists is mandatory. Specific equipment will be used for wadeable vs. non-wadeable sites; be sure to bring both sets of equipment if you are unsure what type of site you will be visiting that day. Pack meters, probes, and sampling gear in such a way as to minimize physical shock and vibration during transport. Pack stock solutions as described in Table 3-1. Follow the regulations of the Occupational Safety and Health Administration (OSHA).

Table 3-1. Stock solutions, uses, and methods for preparation.

Solution	Use	Preparation
Bleach (1-10%)	Clean nets, gear, and inside of boat	Add 10 -100 mL bleach to 1 L distilled water.
Calibration QCS	QCS for pH and conductivity calibration	None (included in site kits)
Lugol's	Preserve periphyton ID samples	None (included in site kits)
95% Ethanol	Preserve benthic samples	None
Formalin	Preserve fish voucher samples	None

Site kits of consumable supplies for each sampling site will be delivered based on the schedule each crew provides prior to the sampling season. **Field crew leaders MUST provide a schedule in order to receive the site kits.** If your schedule changes, report the change as soon as possible to the Field Logistics Coordinator (Jennifer Pitt: jennifer.pitt@tetratech.com; copy Tara.Kolodiej@tetratech.com; 410-356-8993). The site kit will include data forms, labels, sample jars, bottles, filters, and other supplies (see complete list in Appendix A). The teams must inventory these site kits before departure. The teams should also label and package the sample containers into site kits prior to departure. Container labels should not be covered with clear tape until all information is completed during sampling at the river/stream. Store extra site kits of sampling supplies in the vehicles. Inventory these extra site kits prior to each site visit.

3.2 Post Sampling Activities

Upon return to the launching location after sampling, the team must review all completed data forms and labels for accuracy, completeness, and legibility and make a final inspection of samples. If information is missing from the forms or labels, the Field Team Leader is to provide the missing information. The Field Team Leader is to initial all data forms after review. If obtainable samples are missing, the site should be rescheduled for complete sampling. Other post sampling activities include: inspection and cleaning of sampling equipment, supply inventory, sample and data form shipment, and communications.

3.2.1 Review Data Forms and Labels

The field crew leader is ultimately responsible for reviewing all data forms and labels for accuracy, completeness, and legibility. Ensure that written comments use no "shorthand" or abbreviations. The data forms must be thoroughly reviewed. Upon completing the review, the field crew leader must initial the field forms to indicate that they are ready to be sent to the Information Management Center. Each sample label must also be checked for accuracy, completeness, and legibility. The field crew leader must cross-check the sample numbers on the labels with those recorded on the data forms.

3.2.2 Inspect and Prepare Samples

All samples need to be inspected and appropriately preserved and packaged for transport. Check that all samples are labeled, and all labels are completely filled in. Check that each label is covered with clear plastic tape. Check the integrity of each sample container, and

be sure there are no leaks. Make sure that all sample containers are properly sealed. Make sure that all sample containers are properly preserved for storage or immediate shipment.

3.2.3 Equipment Cleanup and Check

All equipment and gear must be cleaned and disinfected between sites to reduce the risk of transferring nuisance species and pathogens. Species of primary concern in the U.S. include Eurasian watermilfoil (*Myriophyllum spicatum*), zebra mussels (*Dreissena polymorpha*), New Zealand mud snails (*Potamopyrgus antipodarum*), *Myxobolus cerebralis* (sporozoan parasite that causes salmonid whirling disease), and *Batrachochytrium dendrobatidis* (a chytrid fungus that threatens amphibian populations). Field crews must be aware of regional species of concern, and take appropriate precautions to avoid transfer of these species. There are several online resources regarding invasive species, including information on cleaning and disinfecting gear, such as the Whirling Disease Foundation (www.whirling-disease.org), the USDA Forest Service (*Preventing Accidental Introductions of Freshwater Invasive Species*, available from http://www.fs.fed.us/invasivespecies/documents/Aquatic_is_prevention.pdf), and the California Dept. of Fish and Game (Hosea and Finlayson 2005). General information about freshwater invasive species is available from the U.S. Geological Survey Nonindigenous Aquatic Species website (http://nas.er.usgs.gov), the *Protect Your Waters* website that is co-sponsored by the U.S. Fish and Wildlife Service (http://www.protectyourwaters.net/hitchhikers), and the Sea Grant Program (http://www.sgnis.org).

Handle and dispose of disinfectant solutions properly, and take care to avoid damage to lawns or other property. Table 3-2 describes equipment care. Inspect all equipment, including nets, boat, and trailer, and clean off any plant and animal material. Prior to leaving a site, drain all bilge water and live wells in the boat. Inspect, clean, and handpick plant and animal remains from vehicle, boat, motor, and trailer. Before moving to the next site, if a commercial car wash facility is available, wash vehicle, boat, and trailer and thoroughly clean (hot water pressurized rinse--no soap). Rinse equipment and boat with 1% - 10% bleach solution to prevent the spread of exotics. Note that many organizations now recommend ***against*** using felt-soled wading boots in affected areas due to the difficulty in removing myxospores and mudsnails.

3.2.4 Supply Inventory

A **site kit** containing field forms, labels, and consumable supplies (see App. A) will be provided to the field crews for each sampling site. Site kits will be shipped out based on the schedule that each field crew provides prior to the start of the sampling season. **Field crew leaders MUST provide a schedule in order to receive the site kits. Crews should include in this schedule the primary fish taxonomist at each site.** If your schedule changes, please report the change as soon as possible to the Field Logistics Coordinator (Jennifer Pitt: jennifer.pitt@tetratech.com; copy Tara.Kolodiej@tetratech.com; 410-356-8993). Prior to sampling, inspect each site kit to ensure all supplies are included. Store an extra, complete backup site kit in the vehicle. Check the inventory of supplies and equipment at the end of the day using the checklists provided in Appendix A. Make sure specific supplies are not running low due to sampling errors, accidental loss, or increased demand at certain sites (e.g., some sites may require extra benthic macroinvertebrate bottles). Make sure you have enough site kits for sites that will require duplicate samples.

Table 3-2. Postsampling equipment care

1. Clean for biological contaminants.

- Prior to departing site, drain all water from live wells and buckets used to hold and process fish, and drain all bilge water from the boat.
- Inspect motor, boat, trailer, sampling gear, waders, boots, etc. for evidence of mud, snails, plant fragments, algae, animal remains, or debris, and remove using brushes or other tools.
- At the base location, inspect and rinse periphyton sampling equipment, dip nets, kick nets, waders, and boots with water and dry. Use one of the procedures below to disinfect gear if necessary.

Additional precautions to prevent transfer of Whirling Disease spores, New Zealand mudsnails, and amphibian chytrid fungus.

Before visiting the site, consult the site dossier and determine if it is in an area where whirling disease, New Zealand mud snails, or chytrid fungus are known to exist. Contact the local State fishery biologist to confirm the existence or absence of these organisms.

- If the stream is listed as "positive" for any of the organisms, or no information is available, *avoid using felt-soled wading boots*, and, after sampling, disinfect **all** fish and benthos sampling gear and other equipment that came into contact with water or sediments (i.e., waders, boots, etc.) by one of the following procedures:

Option A:

1. Soak gear in a 10% household bleach solution for at least 10 minutes, or wipe or spray on a 50% household bleach solution and let stand for 5 minutes
2. Rinse with clean water (do not use stream water), and remove remaining debris
3. Place gear in a freezer overnight or soak in a 50% solution of Formula 409® antibacterial cleaner for at least 10 minutes or soak gear in 120°F (49°C) water for at least 1 minute.
4. Dry gear in direct sunlight (at least 84 °F) for at least 4 hours.

Option B:

1. Soak gear in a solution of Sparquat® (4-6 oz. per gallon of water) for at least 10 minutes (Sparquat is especially effective at inactivating whirling disease spores).
2. Place gear in a freezer overnight or soak in 120°F (49°C) water for at least 1 min.
3. Dry gear in direct sunlight (at least 84 °F) for at least 4 hours.

2. Clean and dry other equipment prior to storage.

- Rinse coolers with water to clean off any dirt or debris on the outside and inside.
- Rinse periphyton sampling equipment with tap water at the base location.
- Make sure conductivity meter probes are rinsed with deionized water and stored moist.
- Rinse carboy and all beakers used to collect water chemistry samples three times with deionized water. Place beakers in a 1-gallon sealable plastic bag with a cube container for use at the next stream.
- Check nets for holes and repair or locate replacements.

3. Inventory equipment and supply needs and relay orders to the Field Logistics Coordinator.

4. Remove GPS, multi-probe meter, and electrofishing unit from carrying cases and set up for predeparture checks and calibration. Examine the oxygen membranes for cracks, wrinkles, or bubbles. Replace if necessary, allowing sufficient time for equilibration.

5. Recharge/replace batteries as necessary.

6. Replenish fuel and oil; if a commercial car wash facility is available, thoroughly clean vehicle and boat (hot water pressurized rinse—no soap).

3.2.5 Shipment of Samples and Forms

The field team must ship or deliver time-sensitive samples (i.e., water chemistry, *chlorophyll a*) to the appropriate analytical laboratories as soon as possible after collection. Other samples (see App. C) may be shipped or delivered in batches provided they can be adequately preserved. Batched samples should be shipped every two weeks. Field teams are to fill out one sample tracking form for each sample shipment. On each sample tracking form, the following information must be recorded:

- Airbill or package tracking number
- Date sample(s) were sent
- Site ID where each sample was collected
- Sample type code:

CHEM – Chemistry	**ENTE** – Enterococci
CHLA – *Chlorophyll a*	**BERW** – Benthos (reach-wide sample)
SEDE– Sediment enzymes	**BELG** – Benthos (low gradient)
PERI – Periphyton	**FTIS** – Fish Tissue
PAPA – Periphyton APA	**VOUC**-- Fish voucher sample

- Date when the sample(s) was collected (1st day if sampling took >1 day)
- Site visit number (e.g., 1 for first visit, 2 for re-visit)
- Sample ID number encoded on label
- Number of containers for each sample
- For Fish Tissue samples (FTIS), record species and length of each fish specimen under Comments
- Any additional comments

Packaging and shipping guidelines for each type of sample are summarized in Figure 3-3. **Detailed sample shipping instructions are presented in Appendix C.**

After checking the Field Forms for completeness and accuracy, the Field Crew Leader will make copies of all Field Forms and retain the copies. **The <u>original</u> forms will be mailed to Marlys Cappaert in the FedEx envelope provided in the site kit.** A pre-addressed airbill will be provided. The original forms must be sent because they are printed specifically to be used in a scanner for automated data entry. Field forms may be retained and mailed in batches throughout the field season (about every 2 weeks) when it is convenient to make the copies.

3.2.6 Status Reports and Communications

After each sampling event, the field crew leader must file a status report via email. This status report email must be sent before the water chemistry/*chlorophyll* sample is shipped, and no later than the following morning after **each** sampling event. An electronic tracking and sample status report form will be emailed to the field crew leaders after their training session. **Complete the tracking and sample status report form for each site, even sites that are**

visited but not sampleable, and email the form to SampleTracking@epa.gov. If you are not able to fill out the electronic form, the Tracking and Sample Status form provided in the field kits can be faxed on a non VOIP fax machine or called into the number provided on the bottom of the TSS form.

The separate, scanable Tracking and Sample Status form (Fig 3.2) provided in the set of field forms must be filled out first; the information from this form will be used to fill out the status report form. The scanable Tracking and Sample Status form will then be shipped in the container with the samples. A tracking form must accompany every sample.

You must follow a standardized naming convention when naming the electronic status report files. The naming convention for fresh samples is "labid_siteid_datecollected.doc:"

ex. WRS_FW08OR123_05_05_2008.doc

For batch/retained samples, the naming convention is "BR_siteid_datecollected.doc:"

ex. BR_FW08OR123_05_05_2008.doc (*in this case, the site id and date collected will refer to the first sample on the page*)

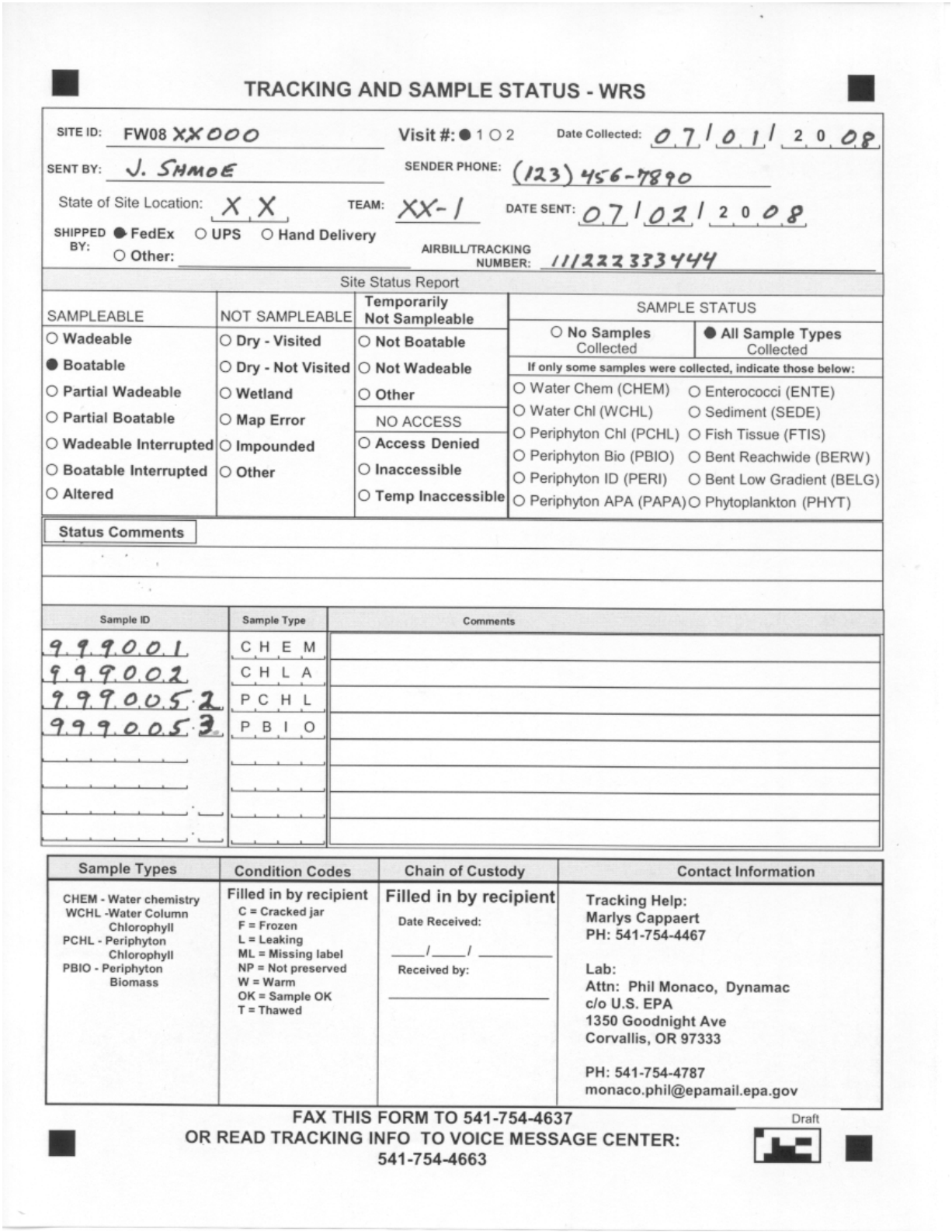
It is very important to complete the status report **after every sampling event**. This will enable the Field Logistics Coordinator to track sampling progress. More importantly, it will enable the Information Management Center to track which samples were collected at each site, and to immediately track the shipment of the time-sensitive water chemistry and *chlorophyll* samples that will be shipped after each sampling event. If the form cannot be emailed by the following morning after sampling, fax the scanable Tracking and Sample Status form (Fig 3.2) or phone in ALL of the information (read the ENTIRE form to the voice mail machine) to the Information Management Coordinator:

Information Management Coordinator: Marlys Cappaert
Sample Tracking (phone): 541-754-4663; Sample Tracking (fax): 541-754-4637

A second form will be provided to track batched and retained samples while they are being held and when they are in transit to the appropriate laboratory. This form must be filled out and emailed right away when samples are brought into your lab or holding facility, and then again when the samples are shipped. The scanable Tracking (Batched and Retained) Form (Fig 3.3) will be filled out and shipped in the container with the samples.

The field crews should call or email the Field Logistics Coordinator (Jennifer Pitt; 410-356-8993; Jennifer.Pitt@tetratech.com) to report any problems encountered. The Field Logistics Coordinator monitors all aspects of field sampling activities. The Field Logistics Coordinator and Information Management Coordinator will contact the EPA Headquarters Coordinator regularly to provide regional updates throughout the sampling period. The EPA Headquarters Coordinator will maintain a database of all sampling activities and reconnaissance information. For questions or problems related to fish tissue or PPCP water sampling, contact Leanne Stahl or Blaine Snyder. See Appendix E for contact information.

The EPA Regional Coordinator serves as the central point of contact for information exchange among field teams, the management and QA staffs, the information management team, and the public. A list of EPA Regional Coordinators and their contact information can be found at the beginning of this manual on page xv.



TRACKING AND SAMPLE STATUS - WRS

SITE ID: FW08 XX000 Visit #: ● 1 ○ 2 Date Collected: 07/01/2008

SENT BY: J. SHMOE SENDER PHONE: (123) 456-7890

State of Site Location: X X TEAM: XX-1 DATE SENT: 07/02/2008

SHIPPED BY: ● FedEx ○ UPS ○ Hand Delivery ○ Other: AIRBILL/TRACKING NUMBER: 111222333444

Site Status Report

SAMPLEABLE	NOT SAMPLEABLE	Temporarily Not Sampleable
○ Wadeable	○ Dry - Visited	○ Not Boatable
● Boatable	○ Dry - Not Visited	○ Not Wadeable
○ Partial Wadeable	○ Wetland	○ Other
○ Partial Boatable	○ Map Error	NO ACCESS
○ Wadeable Interrupted	○ Impounded	○ Access Denied
○ Boatable Interrupted	○ Other	○ Inaccessible
○ Altered		○ Temp Inaccessible

SAMPLE STATUS

○ No Samples Collected ● All Sample Types Collected

If only some samples were collected, indicate those below:

○ Water Chem (CHEM)	○ Enterococci (ENTE)
○ Water Chl (WCHL)	○ Sediment (SEDE)
○ Periphyton Chl (PCHL)	○ Fish Tissue (FTIS)
○ Periphyton Bio (PBIO)	○ Bent Reachwide (BERW)
○ Periphyton ID (PERI)	○ Bent Low Gradient (BELG)
○ Periphyton APA (PAPA)	○ Phytoplankton (PHYT)

Status Comments

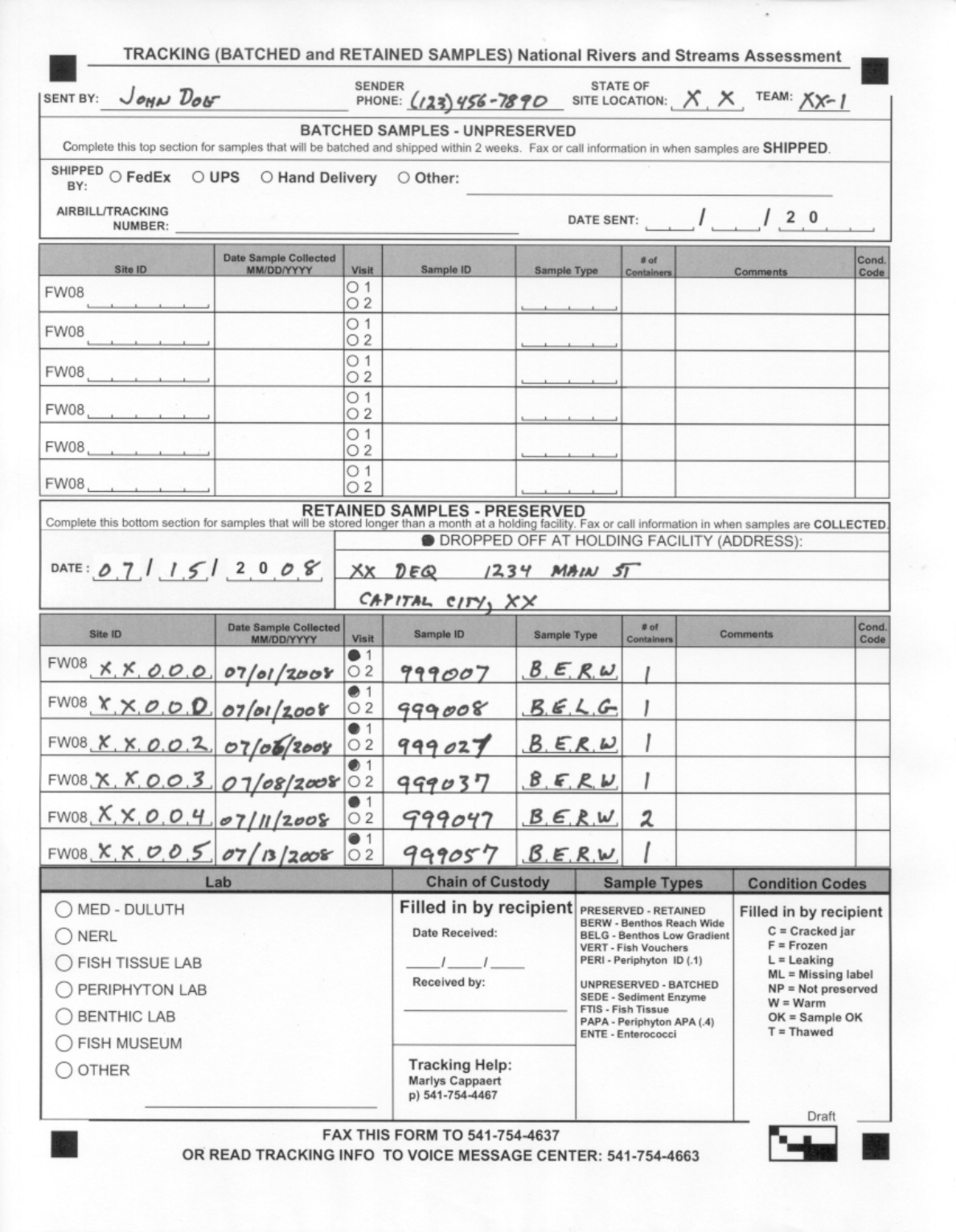
Sample ID	Sample Type	Comments
9.9.9.0.0.1	CHEM	
9.9.9.0.0.2	CHLA	
9.9.9.0.0.5.2	PCHL	
9.9.9.0.0.5.3	PBIO	

Sample Types	Condition Codes	Chain of Custody	Contact Information
CHEM - Water chemistry WCHL -Water Column Chlorophyll PCHL - Periphyton Chlorophyll PBIO - Periphyton Biomass	Filled in by recipient C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK = Sample OK T = Thawed	Filled in by recipient Date Received: ____/____/ _____ Received by: _____	Tracking Help: Marlys Cappaert PH: 541-754-4467 Lab: Attn: Phil Monaco, Dynamac c/o U.S. EPA 1350 Goodnight Ave Corvallis, OR 97333 PH: 541-754-4787 monaco.phil@epamail.epa.gov

FAX THIS FORM TO 541-754-4637
OR READ TRACKING INFO TO VOICE MESSAGE CENTER:
541-754-4663

Draft

Figure 3-2. Tracking and Sample Status Form



TRACKING (BATCHED and RETAINED SAMPLES) National Rivers and Streams Assessment

SENT BY: John Doe SENDER PHONE: (123) 456-7890 STATE OF SITE LOCATION: X X TEAM: XX-1

BATCHED SAMPLES - UNPRESERVED

Complete this top section for samples that will be batched and shipped within 2 weeks. Fax or call information in when samples are **SHIPPED**.

SHIPPED BY: ○ FedEx ○ UPS ○ Hand Delivery ○ Other: _____

AIRBILL/TRACKING NUMBER: _____ DATE SENT: ___/___/20___

Site ID	Date Sample Collected MM/DD/YYYY	Visit	Sample ID	Sample Type	# of Containers	Comments	Cond. Code
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					

RETAINED SAMPLES - PRESERVED

Complete this bottom section for samples that will be stored longer than a month at a holding facility. Fax or call information in when samples are **COLLECTED**.

DATE: 07/15/2008

● DROPPED OFF AT HOLDING FACILITY (ADDRESS):

XX DEQ 1234 MAIN ST
CAPITAL CITY, XX

Site ID	Date Sample Collected MM/DD/YYYY	Visit	Sample ID	Sample Type	# of Containers	Comments	Cond. Code
FW08 XX0000	07/01/2008	● 1 ○ 2	999007	BERW	1		
FW08 XX0000	07/01/2008	● 1 ○ 2	999008	BELG	1		
FW08 XX0002	07/06/2008	● 1 ○ 2	999027	BERW	1		
FW08 XX0003	07/08/2008	● 1 ○ 2	999037	BERW	1		
FW08 XX0004	07/11/2008	● 1 ○ 2	999047	BERW	2		
FW08 XX0005	07/13/2008	● 1 ○ 2	999057	BERW	1		

Lab	Chain of Custody	Sample Types	Condition Codes
○ MED - DULUTH ○ NERL ○ FISH TISSUE LAB ○ PERIPHYTON LAB ○ BENTHIC LAB ○ FISH MUSEUM ○ OTHER _____	**Filled in by recipient** Date Received: ___/___/___ Received by: _____ **Tracking Help:** Marlys Cappaert p) 541-754-4467	PRESERVED - RETAINED BERW - Benthos Reach Wide BELG - Benthos Low Gradient VERT - Fish Vouchers PERI - Periphyton ID (.1) UNPRESERVED - BATCHED SEDE - Sediment Enzyme FTIS - Fish Tissue PAPA - Periphyton APA (.4) ENTE - Enterococci	**Filled in by recipient** C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK = Sample OK T = Thawed

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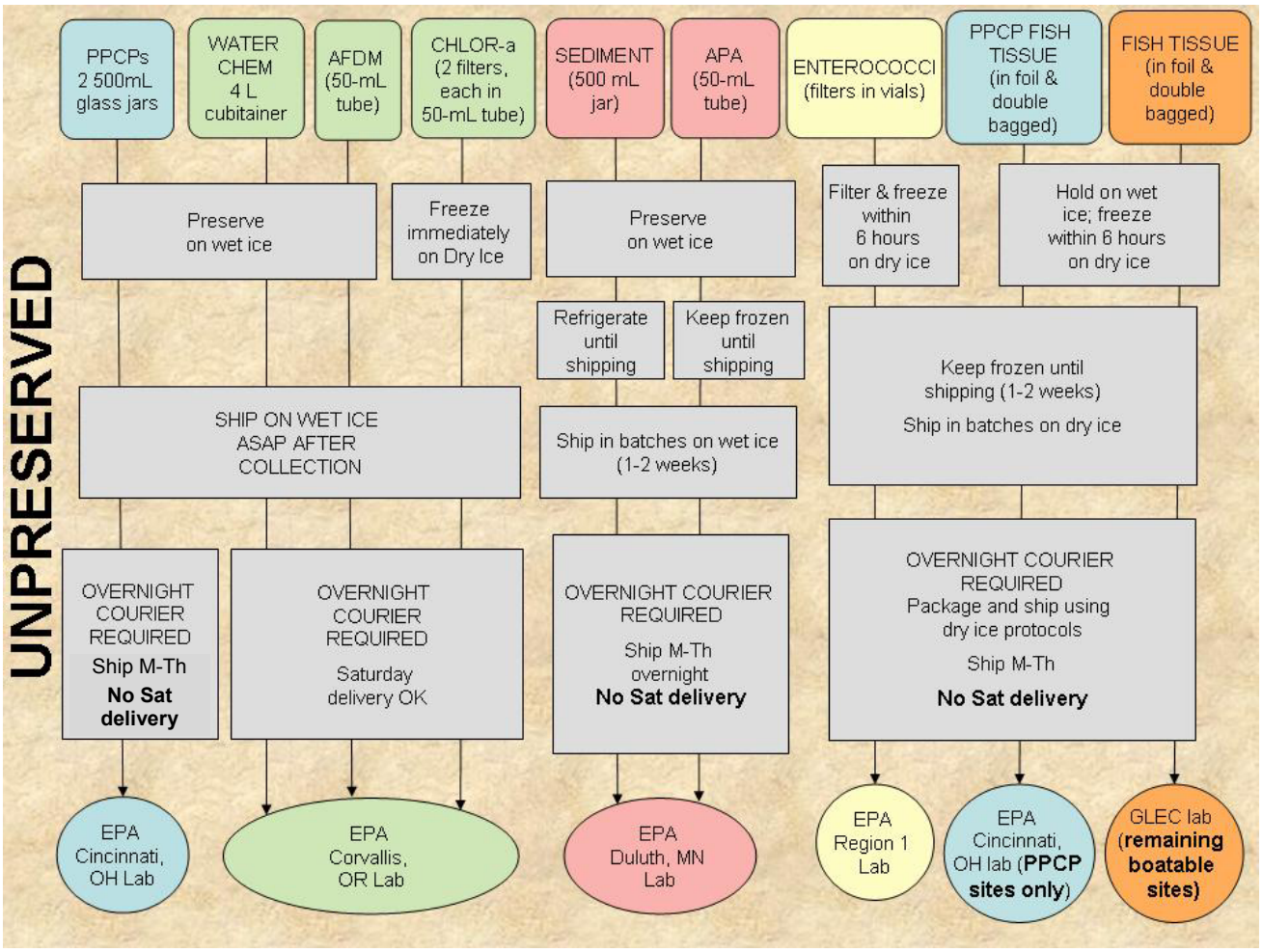
FAX THIS FORM TO 541-754-4637
OR READ TRACKING INFO TO VOICE MESSAGE CENTER: 541-754-4663

Figure 3-3. Tracking (Batched and Retained) Form



*PPCP samples are only collected at a subset of pre-selected sites

Figure 3-4. Sample packaging and shipping procedures for unpreserved samples

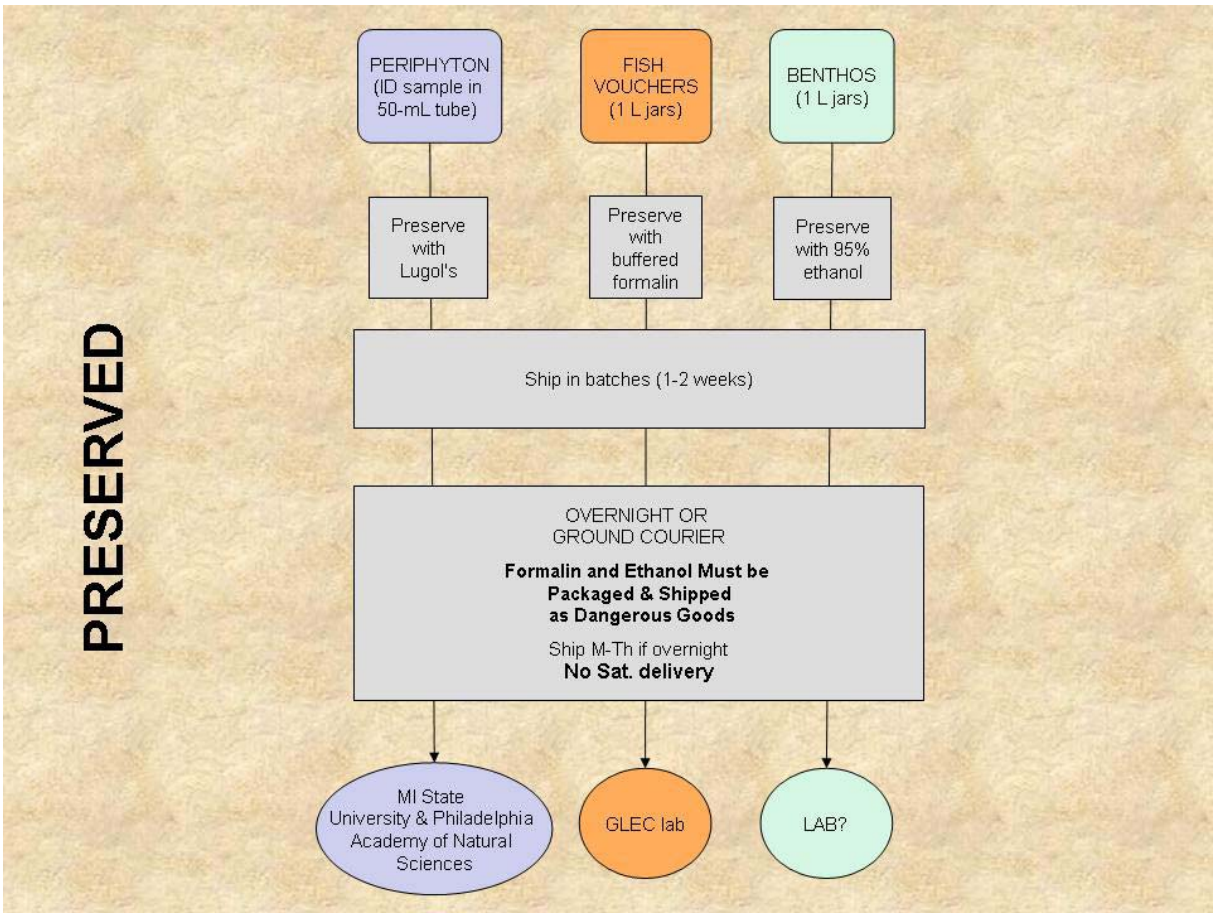


Figure 3-5. Sample packaging and shipping procedures.

4.0 INITIAL SITE PROCEDURES

When you arrive at a site, you must first confirm you are at the correct site, and then determine if the site meets the criteria for sampling and data collection activities (See Site Evaluation Guidelines EPA-841-B-07-008). Inspect the selected reach for appropriate access, safety, and general conditions. Decide whether the site is at base flow condition and not unduly influenced by rain events which could affect the representativeness of field data and samples. If you determine that the site can be sampled, lay out a defined reach within which all sampling and measurement activities are conducted.

4.1 Site Verification Activities

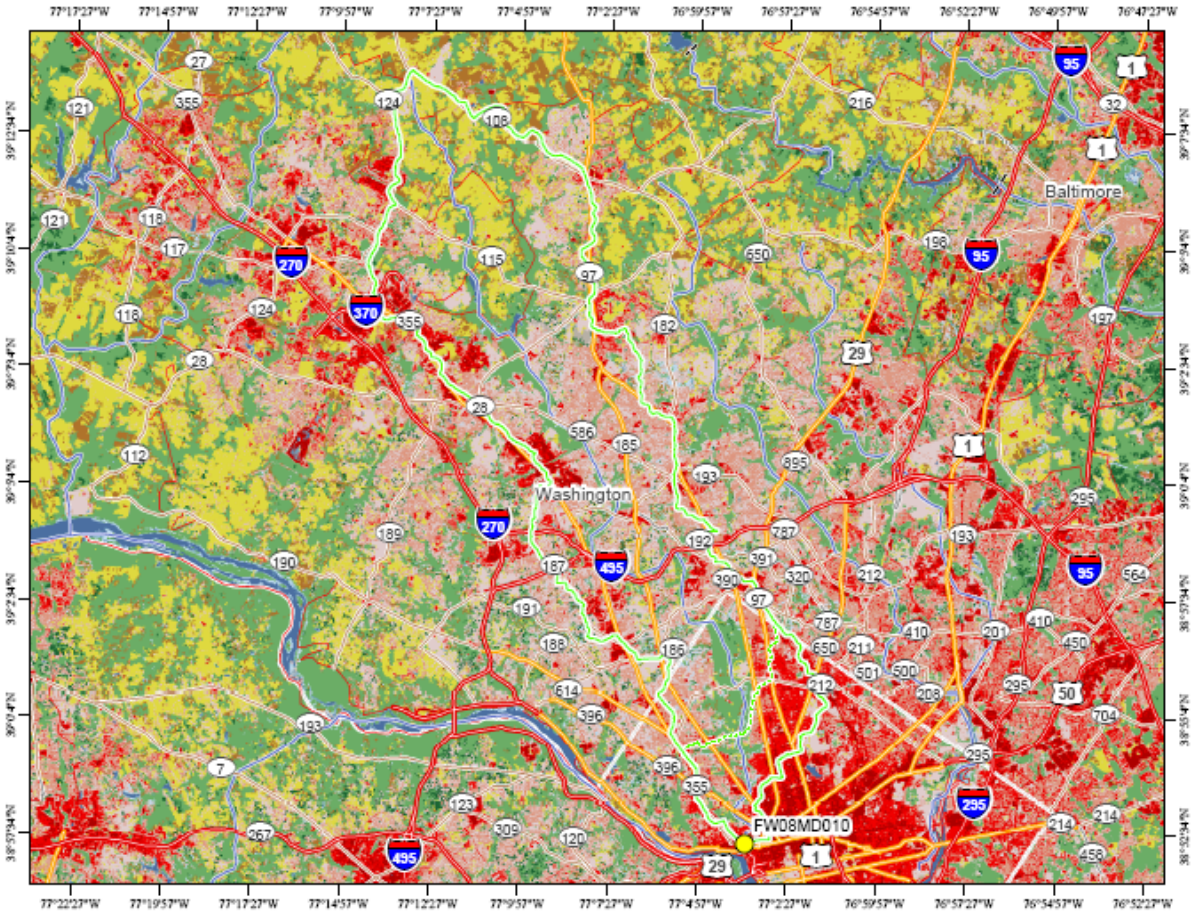
4.1.1 Locating the X-Site

River and stream sampling points were chosen using the National Hydrography Dataset (NHD), in particular NHD-Plus, following a systematic randomized selection process (Stevens and Olsen, 2004). Each point is referred to as the "X-site." The "X-site" is the mid-point of the sampling reach, and it will determine the location and extent for the rest of the sampling reach. The latitude/longitude of the "X-site" is listed on the site spreadsheet that was distributed by the EPA Regional Coordinators.

Conditions encountered at rivers and streams across the country will vary tremendously. To orient the crews and help them anticipate sampling and access challenges, EPA MED prepared site dossiers for all of the sampling sites. Each dossier contains maps with the X-site plotted, and they show general conditions at each site at two scales. The "watershed" scale page shows the position of the site in the landscape and stream network. The "site" scale page shows the area around the site where samples will be taken.

Watershed Page Overview

The watershed page (Figure 4-1) shows land cover (National Land Cover Data 2001), cities, major roads, stream networks, and county, state, watershed and catchment boundaries of the site's watershed. The map scale and level of detail for this page varies according to watershed size. Catchments (nominally, a site's local watershed) are spatially nested within the stream's watershed. Catchment boundaries and hydrologic connectivity were defined in the National Hydrography Dataset Plus (http://www.horizon-systems.com/nhdplus/; NHDPlus) using a Digital Elevation Model (DEM). Watersheds are aggregates of all the catchments upstream from a site. In small watersheds, the catchment may be the entire watershed. In large watersheds, the catchment may not be visible. Pour-points are the downstream end of the watershed. Catchment and watershed attributes (Table 4-1) include areas downstream of the site to the pour-point.



NLCD DESCRIPTION	Catchment	Watershed
11 - Open Water	0.000	0.237
12 - Perennial Ice, Snow	0.000	0.000
21 - Developed, Open Space	12.979	24.030
22 - Developed, Low Intensity	23.563	20.494
23 - Developed, Medium Intensity	39.995	10.902
24 - Developed, High Intensity	8.568	3.540
31 - Barren Land, Rock, Sand, Clay	0.100	0.808
41 - Deciduous Forest	10.014	18.874
42 - Evergreen Forest	0.740	1.488
43 - Mixed Forest	0.000	0.011
52 - Shrub, Scrub	0.000	0.000
71 - Grassland, Herbaceous	0.000	0.000
81 - Pasture, Hay	1.694	13.843
82 - Cultivated Crops	0.614	4.152
90 - Woody Wetlands	1.707	1.468
95 - Emergent Herbaceous Wetlands	0.026	0.152
-- Percent Area in Canada	0.000	0.000
-- Percent Area in Mexico	0.000	0.000
Total	100.000	100.000

RIVER - Rock Creek	
Stream Order	3
Catchment Area (square km)	20.671
Watershed Area (square km)	198.228
FLOW, ELEVATION, and SLOPE	
Mean Annual Flow (cfs) - Unit Runoff Method	87.337
Mean Annual Flow (cfs) - Vogel Method	85.779
Mean Annual Velocity (fps) - Unit Runoff Method	1.264
Mean Annual Velocity (fps) - Vogel Method	1.263
Incremental Flow for Flowline (cfs) - Unit Runoff Method	9.108
Minimum Elevation (meters) - Smoothed	1.680
Maximum Elevation (meters) - Smoothed	19.660
Slope of Flowline (cm/cm)	0.00254
PRECIPITATION and TEMPERATURE	
Area Weighted Mean Annual Precipitation at bottom of Flowline (mm)	1061.872
Area Weighted Mean Annual Temperature at bottom of Flowline (C x 10)	124.034
Mean Annual Precipitation for Catchment (mm)	1073.056
Mean Annual Temperature for Catchment (C x 10)	131.482

Watershed Catchment Pour Point

Map Information
Projection: Albers
Central Meridian: -96.0
1st Standard Parallel: 29.5
2nd Standard Parallel: 45.5
Latitude of Origin: 23.0
Datum: NAD83

Watershed: FW08MD010
Map Scale: 1:236,000

0 3 6 9 12 Kilometers

EPA United States Environmental Protection Agency
Map layout produced under the FAIR II Contract - 2/15/2008
Page 1 of 2

Figure 4-1. Watershed page

Table 4-1. Landscape and NHDPlus attributes for the watershed page (data were summarized from NHDPlus and NLCD2001)

Measure	Scale	Units
Area	Catchment Watershed	km^2
NLCD2001 land cover classes	Catchment Watershed	% area
Mean annual precipitation	Catchment	mm
Mean annual temperature	Catchment	C° x 10
Stream order	Stream (flowline)	Strahler units
Flow	Stream (flowline)	cfs
Velocity	Stream (flowline)	fps
Elevation	Stream (flowline)	meters
Slope	Stream (flowline)	cm/cm
Area-weighted mean annual precipitation	Stream (flowline)	mm
Area-weighted mean annual temperature	Stream (flowline)	C° x 10

Site Page Overview

The site page (Figure 4-2) shows the area immediately surrounding the sampling site. The sampling site, roads, and stream lines are labeled on an aerial photograph. Aerial imagery is provided by ArcGIS Online and features i-cubed Nationwide Select imagery. This dataset consists of imagery from various sources and time periods. For more information on the imagery in these maps, please see http://arcgisonline.esri.com (Layer name: ESRI_Imagery_World_2D). Road data is provided by the U.S. EPA and features 2007 Tele Atlas North America data. The catchment boundary and pour-point are noted. The map scale is fixed at 1:8,000. In some wide rivers, the scale ratio was reduced in order to show shorelines. Sampling stations within the site are distributed according to mean channel width (refer to National Rivers and Streams Assessment Field Operations Manual; EPA 841-B-07-009, 2008). Tabular information includes Site ID, river name, stream order, state, county, latitude and longitude coordinates of the site. An inset map locates the site in the state.

Table 4-2 is the checklist for equipment and supplies required to conduct site verification protocols described in this section. It is a subset of the checklist in Appendix A that is used at a base site to assure that all equipment and supplies are taken to and available at the site. While traveling from a base location to a site, record a detailed description of the route taken on page 1 of the Verification Form (Figure 4-3). This information will help others find the site again in the future. Upon reaching the X-site, confirm its location and verify that you are at the correct stream. Use all available means to accomplish this, including map coordinates, locational data from the GPS, and any other evidence such as signs or conversations with local residents, and record the information on page 1 of the Verification Form (Figure 4-3). Complete a verification

form for each site visited (regardless of whether you end up sampling it), following the procedures described in Table 4-3.

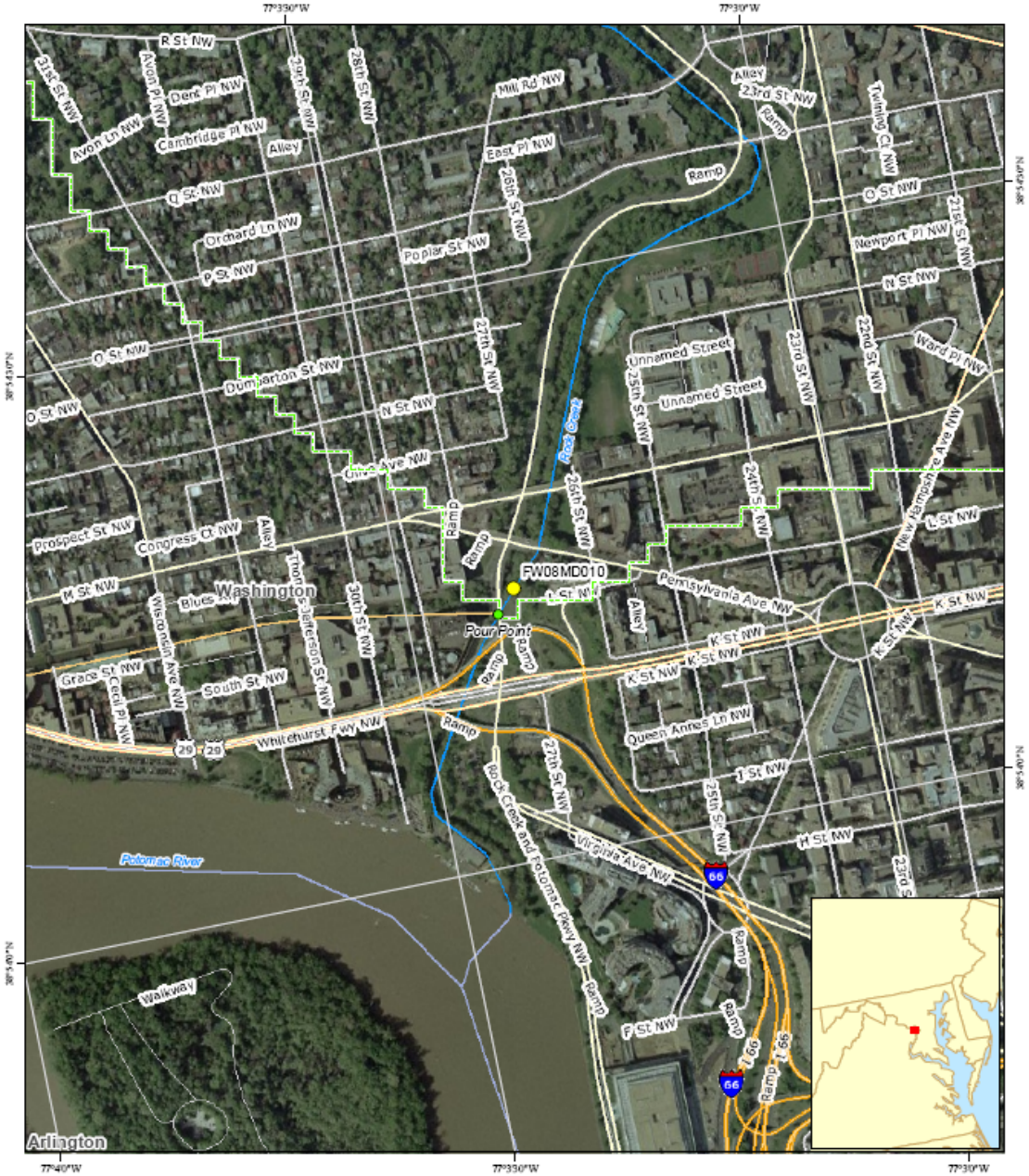


Figure 4-2. Site page.

Table 4-2. Equipment and supplies list for site verification.

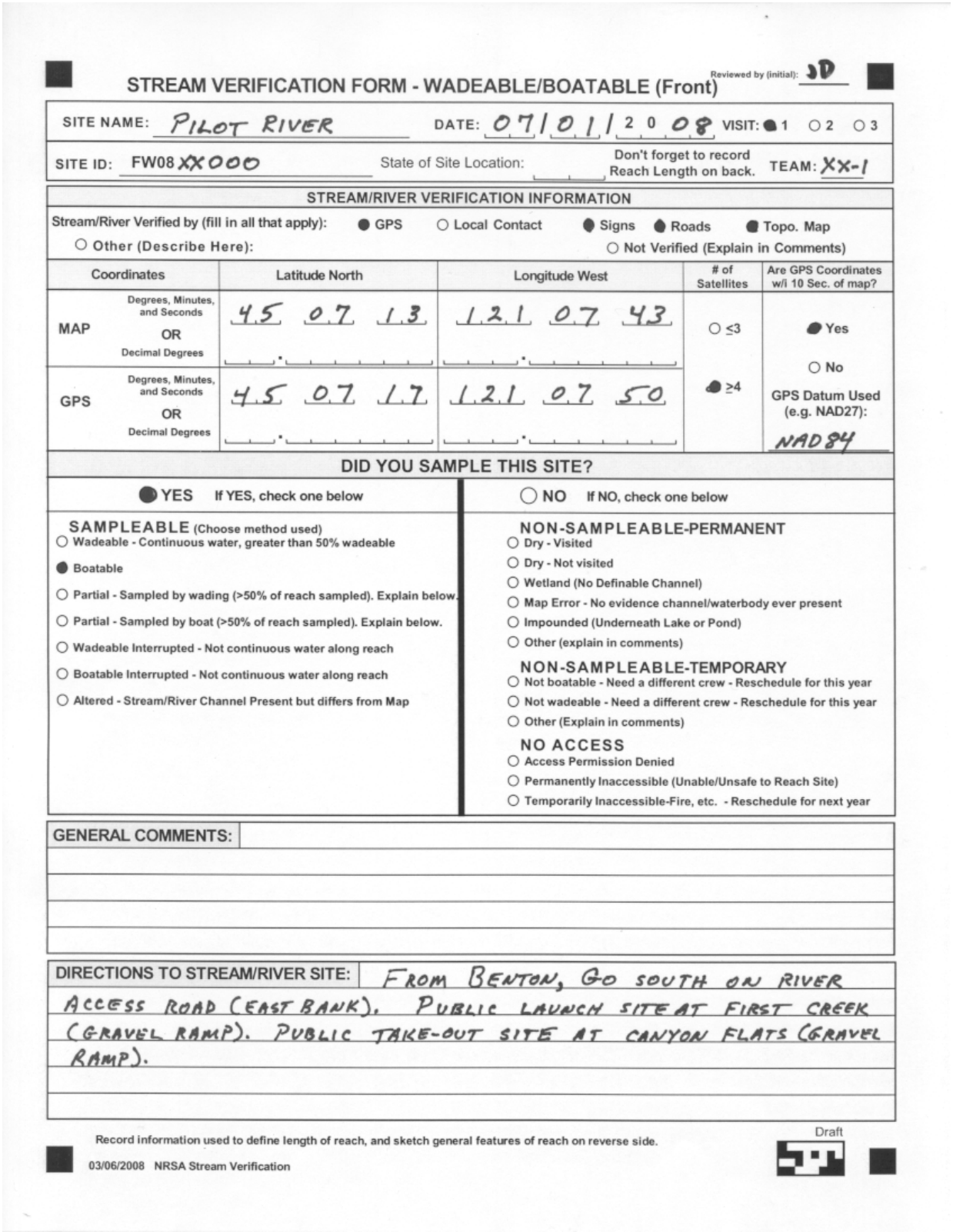
For locating and verifying site	▪ Sampling permit and landowner access(if required) ▪ Field Operations Manual and/or laminated quick reference guide ▪ Site dossier, including access information, site spreadsheet with map coordinates, street and/or topographic maps with "X-site" marked ▪ NRSA Fact Sheets ▪ GPS unit (preferably one capable of recording waypoints) with manual, reference card, extra battery pack ▪ Surveyor's flagging tape (to mark transects if not using GPS waypoints) ▪ Laser rangefinder ▪ 50 m or 100 m measuring tape with reel (if not using rangefinder)
For recording measurements	▪ Clipboard ▪ #2 pencils ▪ Site Verification Form ▪ Fine-tipped indelible markers to write on flagging

4.1.2 Determining the Sampling Status of a Stream

After you confirm the location of the X-site, evaluate the stream reach surrounding the X-site and classify the stream into one of three major sampling status categories: sampleable, non-sampleable, or no access (Table 4-3). The primary distinction between "Sampleable" and "Non-Sampleable" streams is based on the presence of a defined stream channel, water content during base flow, and adequate access to the site.

Even if there is no water at the X-site coordinates, you may still sample the site as an "interrupted flow" stream (Section 4.3.1). If the channel is dry at the X-site, determine if there is water present anywhere within the sampling reach. ***There must be greater than 50% water throughout the channel reach***. If there are isolated pools of water within the reach that equal greater than 50% of the reach length, proceed to sample using the modified procedures outlined in Section 4.3.1. If less than 50% of the reach has water , classify the site as "Dry-visited" on the verification form. NOTE: Do not "slide" the reach (Section 4.2) for the sole purpose of obtaining more water to sample (e.g., the downstream portion of the reach has water, but the upstream portion does not).

Record the sampling status and pertinent site verification information on the Verification Form (Figure 4-3). If the site is non-sampleable or inaccessible, no further sampling activities are conducted. Replace the site with the first oversample site on the state list within the appropriate Strahler order category (Section 1.1.2). Notify the EPA Regional Coordinator and Field Logistics Coordinator (Section 3.2.6) that the site was replaced.



STREAM VERIFICATION FORM - WADEABLE/BOATABLE (Front)

Reviewed by (initial): JD

SITE NAME: PILOT RIVER DATE: 07/01/2008 VISIT: ● 1 ○ 2 ○ 3

SITE ID: FW08XX000 State of Site Location: Don't forget to record Reach Length on back. TEAM: XX-1

STREAM/RIVER VERIFICATION INFORMATION

Stream/River Verified by (fill in all that apply): ● GPS ○ Local Contact ● Signs ● Roads ● Topo. Map ○ Other (Describe Here): ○ Not Verified (Explain in Comments)

Coordinates		Latitude North	Longitude West	# of Satellites	Are GPS Coordinates w/i 10 Sec. of map?
MAP	Degrees, Minutes, and Seconds OR Decimal Degrees	45 07 13	121 07 43	○ ≤3	● Yes ○ No
GPS	Degrees, Minutes, and Seconds OR Decimal Degrees	45 07 17	121 07 50	● ≥4	GPS Datum Used (e.g. NAD27): NAD84

DID YOU SAMPLE THIS SITE?

● YES If YES, check one below

SAMPLEABLE (Choose method used)
- ○ Wadeable - Continuous water, greater than 50% wadeable
- ● Boatable
- ○ Partial - Sampled by wading (>50% of reach sampled). Explain below.
- ○ Partial - Sampled by boat (>50% of reach sampled). Explain below.
- ○ Wadeable Interrupted - Not continuous water along reach
- ○ Boatable Interrupted - Not continuous water along reach
- ○ Altered - Stream/River Channel Present but differs from Map

○ NO If NO, check one below

NON-SAMPLEABLE-PERMANENT
- ○ Dry - Visited
- ○ Dry - Not visited
- ○ Wetland (No Definable Channel)
- ○ Map Error - No evidence channel/waterbody ever present
- ○ Impounded (Underneath Lake or Pond)
- ○ Other (explain in comments)

NON-SAMPLEABLE-TEMPORARY
- ○ Not boatable - Need a different crew - Reschedule for this year
- ○ Not wadeable - Need a different crew - Reschedule for this year
- ○ Other (Explain in comments)

NO ACCESS
- ○ Access Permission Denied
- ○ Permanently Inaccessible (Unable/Unsafe to Reach Site)
- ○ Temporarily Inaccessible-Fire, etc. - Reschedule for next year

GENERAL COMMENTS:

DIRECTIONS TO STREAM/RIVER SITE: FROM BENTON, GO SOUTH ON RIVER ACCESS ROAD (EAST BANK). PUBLIC LAUNCH SITE AT FIRST CREEK (GRAVEL RAMP). PUBLIC TAKE-OUT SITE AT CANYON FLATS (GRAVEL RAMP).

Record information used to define length of reach, and sketch general features of reach on reverse side.

Draft

03/06/2008 NRSA Stream Verification

Figure 4-3. Verification Form (page 1).

Table 4-3. Site Verification Procedures

1. Find the stream/river location in the field corresponding to the X-site coordinates and the "X" marked on the maps prepared for each site in the site dossier. Record the routes taken and other directions on the Verification Form so that others can visit the same location in the future. If the site is non-wadeable, locate public or private launch sites.
2. Use a GPS receiver to confirm the latitude and longitude at the X-site with the coordinates provided for the site (datum = NAD 27). Record these on the Verification Form.
3. Use all available means to insure you are at the correct stream/river as marked on the map, including 1:24,000 USGS maps, topographic landmarks, road maps, signs, local contacts, etc.
4. Scan the channel upstream and downstream from the X-site, decide if the site is sampleable, and mark the appropriate circle on the verification form. If the channel is dry at the X-site, determine if water is present within 75 m upstream and downstream of the X-site. Assign one of the following sampling status categories to the stream. Record the category on the Verification Form.

SAMPLEABLE CATEGORIES

- Wadeable - Continuous water, ≥50% wadeable
- Boatable
- Partial - Sampled by wading (>50% of reach sampled)
- Partial - Sampled by boat (>50% of reach sampled)
- Wadeable Interrupted: not continuous water along reach
- Boatable Interrupted: not continuous water along reach
- Altered Channel: Stream/river channel present but differs from map.

NON-SAMPLEABLE CATEGORIES

Permanent

- Dry Channel: Less than 50% water within the reach. Record as "Dry-Visited." If site was determined to be dry (or otherwise non-perennial) from another source and/or field verified before the actual sampling visit, record as "Dry-Not visited".
- Wetland: Standing water present, but no definable stream channel. If wetland is surrounding a stream channel, define the site as Target but restrict sampling to the stream channel.
- Map Error: No evidence that a water body or stream channel was ever present at the X-site.
- Impounded stream: Stream is submerged under a lake or pond due to man-made or natural (e.g., beaver dam) impoundments. If the impounded stream is still wadeable, record it as "Altered" and sample.
- Other: Examples would include underground pipelines, or a non-target canal. A sampling site must meet both of the following criteria to be classified as a non-target canal:
  - The channel is constructed where no natural channel has ever existed.
  - The sole purpose/usage of the reach is to transfer water. There are no other uses of the waterbody by humans (e.g., fishing, swimming, boating).

Temporary

- Not Boatable – need a different crew
- Not Wadeable – need a different crew
- Other: The site could not be sampled on that particular day, but is still a target site. Examples might include a recent precipitation event that has caused unrepresentative conditions.

NO ACCESS TO SITE CATEGORIES

- Access Permission Denied: You are denied access to the site by the landowners.

- Permanently Inaccessible: Site is unlikely to be sampled by anyone due to physical barriers that prevent access to the site (e.g., cliffs).
- Temporarily Inaccessible: Site cannot be reached due to barriers that may not be present at a future date (e.g. forest fire, high water, road temporarily closed, unsafe weather conditions)

5. Do not sample non-target or "Non-sampleable" or "No Access" sites. Fill in the "NO" circle for "Did you sample this site?" and check the appropriate circle in the "Non-Sampleable" or "No Access" section of the Verification Form; provide detailed explanation in comments section.

4.1.3 Sampling During or After Rain Events

Avoid sampling during high flow rainstorm events. It is often unsafe to be in the water during such times. In addition, biological and chemical conditions during such episodes are often quite different from those during baseflow. On the other hand, sampling cannot be restricted to only strict baseflow conditions. It would be next to impossible to define "strict baseflow" with any certainty at an unstudied site. Such a restriction would also greatly shorten the index period when sampling activities can be conducted. Thus, some compromise is necessary regarding whether to sample a given stream because of storm events. To a great extent, this decision is based on the judgment of the field team. Some guidelines to help make this decision are presented in Table 4-4. The major indicator of the influence of storm events will be the condition of the stream itself. If you decide a site is unduly influenced by a storm event, do not sample the site that day. Notify the Field Logistics Coordinator or other central contact person to reschedule the stream for another visit.

Table 4-4. Guidelines to determine the influence of rain events

- If it is running at bank full discharge or the water seems much more turbid than typical for the class of stream do not sample it that day.
- Do not sample that day if it is unsafe to be in the water.
- Keep an eye on the weather reports and rainfall patterns. Do not sample a stream during periods of prolonged heavy rains.
- If the stream seems to be close to normal summer flows, and does not seem to be unduly influenced by storm events, sample it even if it has recently rained or is raining.

4.1.4 Site Photographs

Taking site photographs is an optional activity, but should be considered if the site has unusual natural or man-made features associated with it. If you do take photographs with a digital camera at a site, date-stamp the photograph and include the site ID. Alternatively, start the sequence with one photograph of an 8.5 × 11 inch piece of paper with the site ID, waterbody name, and date printed in large, thick letters. After the photo of the site ID information, take at least two photographs at the X-site, one in the upstream direction and one downstream. Take any additional photos you find interesting after these first three pictures. Keep a log of your photographs and briefly describe each one.

4.2 Laying out the sampling reach

Unlike chemistry, which can be measured at a point, most of the biological and habitat structure measures require sampling a certain length of a stream to get a representative picture of the ecological community. A length of 40 times the channel width is necessary to characterize

the habitat and several biotic assemblages associated with the sampling reach. Establish the sampling reach about the X-site using the procedures described in Tables 4-5a (non-wadeable sites) and 4-5b (wadeable sites). It is **highly recommended** that you lay out the sampling reach for large, non-wadeable sites before you go in the field using maps, aerial photos, and/or GIS software. This will save time on the field day.

Scout the sampling reach to make sure it is clear of obstacles that would prohibit sampling and data collection activities. Record the channel width used to determine the reach length, and the sampling reach length upstream and downstream of the X-site on page 2 of the Verification Form as shown in Figure 4-4. Figures 4-5 and 4-6 illustrate the principal features of the established sampling reach for both non-wadeable and wadeable sites, including the location of 11 cross-section transects used for collecting samples and physical habitat measurements. The figures also show the specific sampling stations on each cross-section transect at the two different types of sites for collection of sediment enzyme, periphyton, and benthic macroinvertebrate samples.

Before leaving the stream, complete a rough sketch map of the stream reach you sampled on page 2 of the Verification Form (Figure 4-4). In addition to any other interesting features that should be marked on the map, note any landmarks/directions that can be used to find the X-site for future visits.

Table 4-5a. Laying out the sampling reach at non-wadeable sites

Laying out the sampling reach at the base site (recommended at boatable sites)

1. On an aerial photo or a 1:100:000 topographic map, locate the X-site using the coordinates provided for the site and the maps prepared in the site dossier for the site.
2. Determine the average wetted width of the channel at the X-site using maps and/or aerial photographs. To get an average, determine the wetted width of the channel at 5 places of "typical" width within approximately 5 channel widths upstream and downstream from the X-site. Average the 5 readings together and round to the nearest 1 m.
3. Multiply the average wetted width by 40 to determine the reach length. If the average width is <4 m, use 150 m as a **minimum** reach length. If the average width is >100 m, use 4 km as a **maximum** reach length.
4. From the X-site, measure a distance of 20 channel widths downstream using GIS software. Be careful to measure all of the bends of the river/stream; do not artificially straighten out the line of measurement. The downstream endpoint is marked as Transect K. Measure 20 channel widths upstream from the X-site; the upstream end of the reach is marked as Transect A.
5. Measure 1/10 of the reach length downstream from Transect A, and mark this spot as Transect B. Continue marking the 11 transects A - K in increments of 1/10 of the reach length. Enter the waypoints for the transects into a GPS unit so the transects are easy to find on the sampling day.
6. Assign the sampling station at Transect A randomly (e.g., use the seconds display on a digital watch to select the initial sampling station: 1 - 5 = Left Bank, 6 – 9 = Right Bank). From here, three stations will be on the first (randomly selected) side of the river, then 2 on the other, then 2 on the first side, and so on through Transect K (as shown in Figure 4-5).
7. When you are at the site, "ground truth" the wetted width measurements and proceed to Steps 9 & 10 to see if the layout needs to be adjusted.

Laying out the sampling reach in the field

8. Use a laser range finder to determine the wetted width of the channel at 5 places of "typical" width within approximately 5 channel widths upstream and downstream from the X-site. Average the 5 readings together and round to the nearest 1 m. If the average width is <4 m, use 150 m as a

minimum reach length. If the average width is >100 m, use 4 km as a maximum reach length. Record this width on page 2 of the Site Verification Form.

For channels with "interrupted flow", estimate the width based on the unvegetated width of the channel (again, with a 150 m minimum and 4 km maximum).

9. Check the condition of the stream about the X-site by having one team member go upstream and one downstream. Each person proceeds until they can see the stream to a distance of 20 times the average channel width (equal to one-half the sampling reach length) determined in Step 1.
10. Determine if the reach needs to be adjusted about the X-site due to confluences with higher order streams (downstream), or a change to a lower order streams (upstream), impoundments (lakes, reservoirs, ponds), physical barriers (e.g., falls, cliffs), or because of access restrictions to a portion of the initially-determined sampling reach. Refer to Table 4-6 for specific instructions.
11. Starting at the X-site (or the new midpoint of the reach if it had to be adjusted as described in Step 10), measure a distance of 20 channel widths downstream using a GPS unit, laser rangefinder, or tape measure. Be careful to measure all of the bends of the river/stream; do not artificially straighten out the line of measurement. Enter the channel to make measurements only when necessary to avoid disturbing the stream channel prior to sampling activities. The downstream endpoint is flagged as Transect K. The upstream end of the reach is flagged as Transect A.
12. **Sampling Stations at non-wadeable sites**

    At Transect A, use the seconds display on a digital watch to select the initial sampling station for transect samples: 1 - 5 = Left Bank, 6 – 9 = Right Bank. Mark "L" or "R" on the transect flagging.
13. Measure 1/10 of the reach length downstream from Transect A. Flag this spot as Transect B. Assign the sampling station systematically after the first random selection as shown in Figure 4-5. Three stations will be on the first side of the river, then 2 on the other, then 2 on the first side, and so on through Transect K.
14. Proceed downstream with a GPS unit, laser rangefinder, or tape measure and flag the positions of 9 additional transects (labeled "C" through "K" as you move upstream) at intervals equal to 1/10 of the reach length. Continue to assign the sampling stations systematically.

Table 4-5b. Laying out the sampling reach at wadeable sites

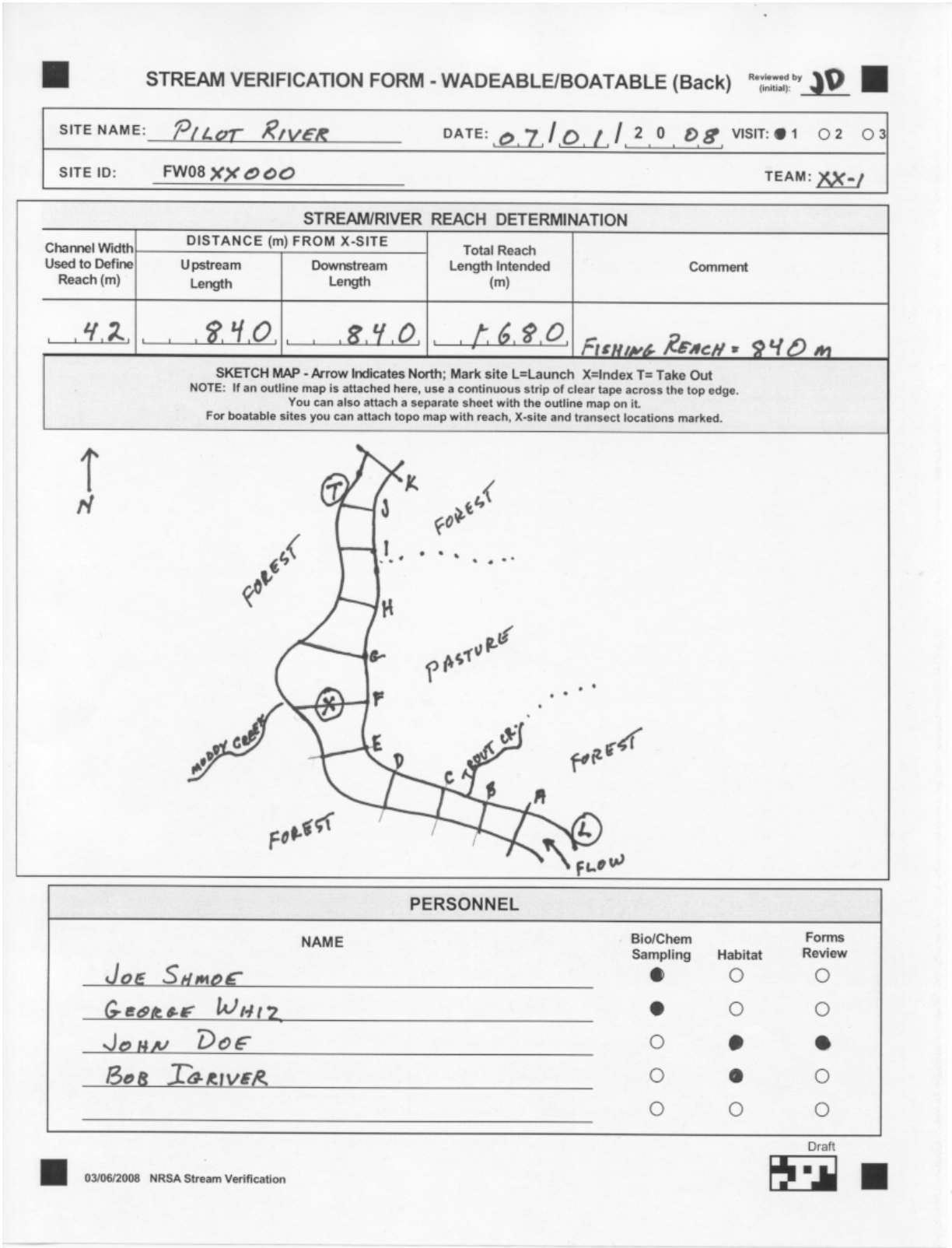
1. Use a surveyor's rod, tape measure, or laser range finder to determine the wetted width of the channel at 5 places of "typical" width within approximately 5 channel widths upstream and downstream from the X-site. Average the 5 readings together and round to the nearest 1 m. If the average width is <4 m, use 150 m as a minimum reach length. If the average width is >100 m, use 4 km as a maximum reach length. Record this width on page 2 of the Site Verification Form.

   For channels with "interrupted flow", estimate the width based on the unvegetated width of the channel (again, with a 150 m minimum and 4 km maximum).

2. Check the condition of the stream about the X-site by having one team member go upstream and one downstream. Each person proceeds until they can see the stream to a distance of 20 times the average channel width (equal to one-half the sampling reach length) determined in Step 1.
3. Determine if the reach needs to be adjusted about the X-site due to confluences with higher order streams (downstream), a change to a lower order streams (upstream), impoundments (lakes, reservoirs, ponds), physical barriers (e.g., falls, cliffs), or because of access restrictions to a portion of the initially-determined sampling reach. Refer to Table 4-7.
4. Starting at the X-site (or the new midpoint of the reach if it had to be adjusted as described in Step 3), measure a distance of 20 channel widths down one side of the stream using a GPS unit, laser rangefinder, or tape measure. Be careful not to "cut corners". Enter the channel to make measurements only when necessary to avoid disturbing the stream channel prior to sampling activities. This endpoint is the downstream end of the reach, and is flagged as Transect "A".
5. **Sampling Stations at wadeable sites:**

   At Transect A, use the seconds display on a digital watch to select the initial sampling station for standard transect samples: 1-3="Left", 4-6="Center", 7-9=Right. Mark "L", "C", or "R" on the transect flagging; the 3 potential collection points are roughly equivalent to 25%, 50%, and 75% of the channel width, respectively.

6. Measure 1/10 of the required reach length upstream from transect A. Flag this spot as transect B. Assign the sampling station systematically after the first random selection (Figure 4-6 & Table 4-6).
7. Proceed upstream with the tape measure and flag the positions of 9 additional transects (labeled "C" through "K" as you move upstream) at intervals equal to 1/10 of the reach length. Continue to assign the sampling stations systematically.
8. Benthic macroinvertebrates at "low gradient" streams: A second, separate composite is collected at low gradient streams to include the edge habitats (0%, 50%, and 100% channel width). The initial sampling station will be the first to the right of the one selected for the standard sample (Table 4-5). For example, if the sampling station for transect A (standard), was "C", then the initial transect A sampling station for the second sample would be "R". This second pattern would be R, L, C, R, ….



STREAM VERIFICATION FORM - WADEABLE/BOATABLE (Back) Reviewed by (initial): JD

SITE NAME: PILOT RIVER DATE: 07/01/2008 VISIT: ● 1 ○ 2 ○ 3

SITE ID: FW08XX000 TEAM: XX-1

STREAM/RIVER REACH DETERMINATION

Channel Width Used to Define Reach (m)	DISTANCE (m) FROM X-SITE		Total Reach Length Intended (m)	Comment
	Upstream Length	Downstream Length		
4.2	840	840	1680	FISHING REACH = 840 m

SKETCH MAP - Arrow Indicates North; Mark site L=Launch X=Index T= Take Out
NOTE: If an outline map is attached here, use a continuous strip of clear tape across the top edge.
You can also attach a separate sheet with the outline map on it.
For boatable sites you can attach topo map with reach, X-site and transect locations marked.

PERSONNEL

NAME	Bio/Chem Sampling	Habitat	Forms Review
JOE SHMOE	●	○	○
GEORGE WHIZ	●	○	○
JOHN DOE	○	●	●
BOB IGRIVER	○	●	○
	○	○	○

Draft

03/06/2008 NRSA Stream Verification

Figure 4-4. Verification Form (page 2)

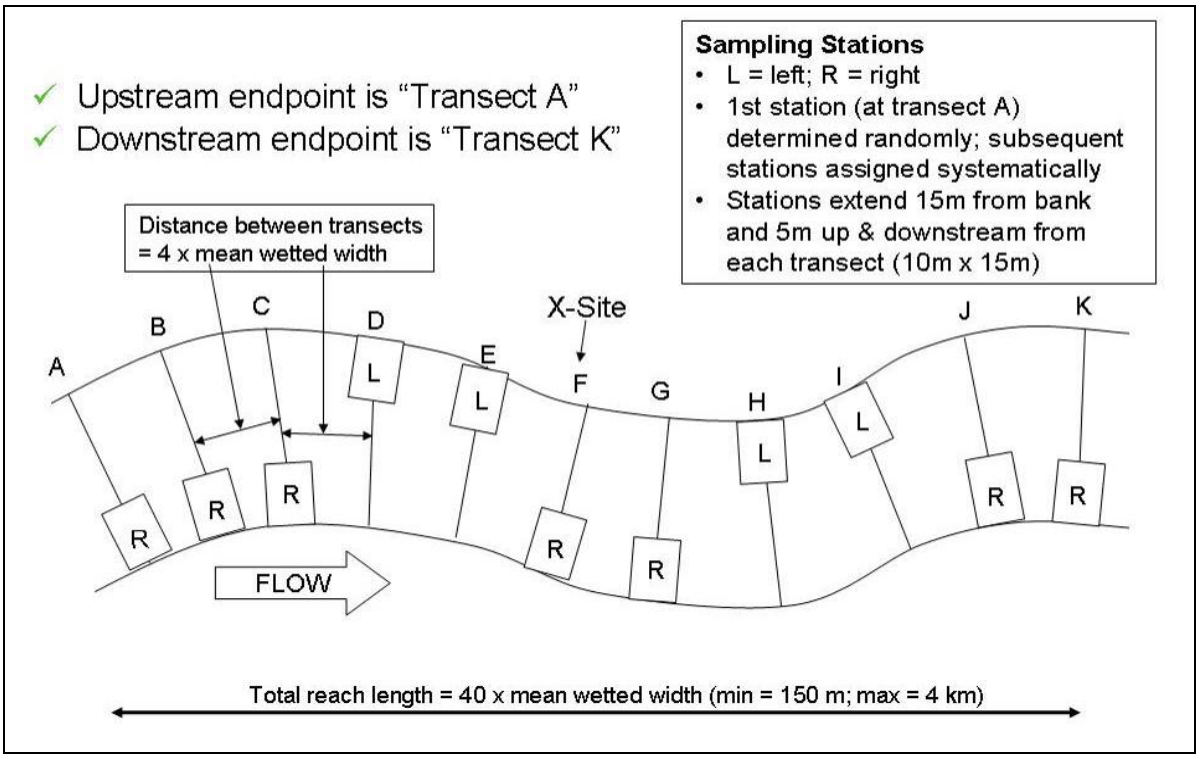


Figure 4-5. Sampling reach features for a non-wadeable site.

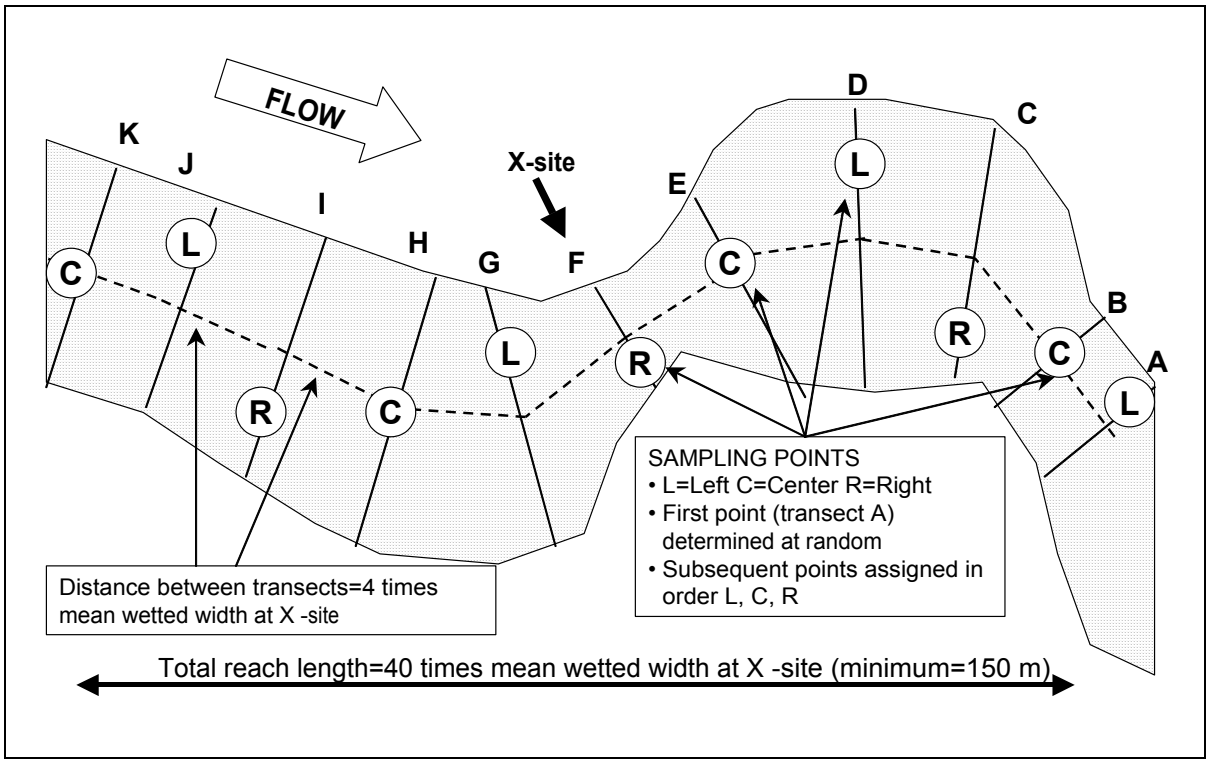


Figure 4-6. Sampling reach features for a wadeable site.

Table 4-6. Sample point distribution in wadeable streams (the Transect A sample point for the standard sample is randomly selected; the secondary sample point distribution is used only to collect the second benthic macroinvertebrate sample in low gradient wadeable streams (L=left, C=center, R=right))

PRIMARY SAMPLE											
Transect A	Transect….	B	C	D	E	F	G	H	I	J	K
If you randomly select "LEFT"	Then continue sequence….	C	R	L	C	R	L	C	R	L	C
SECONDARY SAMPLE- Low gradient benthic macroinvertebrate only											
Transect A	Transect….	B	C	D	E	F	G	H	I	J	K
Select next in sequence to start 2nd pattern "CENTER"	Then continue sequence….	R	L	C	R	L	C	R	L	C	R

There are some conditions that may require sliding the reach about the X-site (i.e., the X-site is no longer located at the midpoint of the reach) to avoid features we do not wish to or physically cannot sample across. Sliding the reach involves noting the distance of the barrier, confluence, or other restriction from the X-site, and flagging the restriction as the endpoint of the reach. Add the distance to the other end of the reach, such that the total reach length remains the same, but it is no longer centered about the X-site. Table 4-7 describes when you should and should not slide the sampling reach.

Table 4-7. Sliding the sampling reach

1. Slide the reach if you run into an impoundment (lake, pond, or reservoir), so that the lake/stream confluence is at one end.
2. Slide the reach if you run into an impassible barrier (e.g., waterfall, cliff, navigation dam) so that the barrier is at one end.
3. When you are denied access permission to a portion of the reach, you can slide the reach to make it entirely accessible; use the point of access restriction as the endpoint of the reach.
4. Note the distance of the barrier, confluence, or other restriction from the X-site, and flag the restriction as the endpoint of the reach. Add the distance to the other end of the reach, so the total reach length remains the same, but it is no longer centered about the X-site.
5. **Do not slide the reach so that the X-site falls outside of the reach boundaries**.
6. **Do not proceed upstream into a lower order stream or downstream into a higher order stream** when laying out the stream reach (order is based on 1:100,000 scale maps).
7. **Do not slide a reach to avoid man-made obstacles** such as bridges, culverts, rip-rap, or channel-ization. These represent important features and effects to study.
8. **Do not slide a reach to gain more water** to sample if the flow is interrupted (Section 4.3.1).
9. **Do not slide a reach to gain better habitat** for benthos or fish,

4.3 Modifying Sample Protocols for High or Low Flows

4.3.1 Streams with Interrupted Flow

You cannot collect the full complement of field data and samples from streams that are categorized as "Interrupted" (Table 4-8). Note that no data should be collected from streams that are completely "Dry" as defined in Table 4-8. Interrupted streams will have some cross-sections amenable to biological sampling and habitat measurements and some that are not. To be considered target, streams must have greater than 50% water in the reach length within the channel (can be isolated pools). Modified procedures for interrupted streams are presented in Table 4-8. Samples for water chemistry (Section 5) will be collected at the X-site (even if the reach has been adjusted by "sliding" it). If the X-site is dry and there is water elsewhere in the sample reach, collect the sample from a location having water with a surface area >1 m^2 and a depth >10 cm.

Collect data for the physical habitat indicator along the entire sample reach from interrupted streams, regardless of the amount of water present at the transects. Obtain depth measurements along the deepest part of the channel (the "thalweg") along the entire sampling reach to provide a record of the "water" status of the stream for future comparisons (e.g., the percent of length with intermittent pools or no water). Other measurements associated with characterizing riparian condition, substrate type, etc., are useful to help infer conditions in the stream when water is flowing.

Table 4-8. Reach layout modifications for interrupted streams

- Streams with less than 50% of reach length containing water (not necessarily continuous) are considered dry and are not sampled.
- If more than 50% of the channel has water and if the X-site is dry but there is flowing water or a pool of water having a surface area > 1 m^2 and a depth > 10 cm somewhere along the defined sampling reach, take the water sample at the pool or flowing water location that is nearest to the X-site. Note that the sample was not collected at the X-site and where on the reach the sample was collected on the field data form.
- Do not collect a water sample if there is no acceptable location within the sampling reach. Record a "K" flag for the chemistry sample on the sample collection form and explain why the sample was not collected in the comments section of the form.

Physical Habitat, Periphyton, Sediment Enzymes, and Benthic Macroinvertebrates

- Obtain a complete thalweg profile for the entire reach. At points where channel is dry, record depth as 0 cm and wetted width as 0 m.
- At each of the transects (cross-sections), sample the stream depending on flow status:

DRY CHANNEL: No surface water anywhere in cross-section; collect all physical habitat data. Use the unvegetated area of the channel to determine the channel width and the subsequent location of substrate sampling points. Record the wetted width as 0 m. Record substrate data at the sampling points located in the unvegetated, but dry, channel. Do not collect periphyton, sediment enzymes, or benthic macroinvertebrates from this transect.

DAMP CHANNEL: No flowing water at transect, only puddles of water < 10 cm deep; collect all physical habitat data. Do not collect periphyton, sediment enzymes, or benthic macroinvertebrates from this transect.

WATER PRESENT: Transect has flow or pools > 10 cm deep; collect all data and measurements for physical habitat, periphyton, sediment enzymes, benthic macroinvertebrate, and fish indicators, using standard procedures.

4.3.2 Partially Wadeable Sites

Some wadeable sites will have sections that are too deep or swift to wade safely, and it will be impossible to do all of the wadeable sampling protocols at every transect. At these sites, keeping safety in mind, try to do as much sampling and data collection as you can with the wadeable procedures. The amount of sampling that can actually be done while wading will depend on the extant conditions. Only sample or measure what can be done **safely**. Make detailed comments on the Verification Form describing what the conditions were like and where sampling occurred. Use the sketch map on the back of the Verification Form to indicate problem areas and where samples were collected if you had to go off transect. If barriers prohibit physically reaching the X-site, then the site is not a Sampleable site; it should be coded as "No Access - Inaccessible" on the Verification Form.

4.3.3 Braided Rivers and Streams

Depending upon the geographic area and/or the time of the sampling visit, you may encounter a stream having "braided" channels, which are characterized by numerous sub-channels that are generally small and short, often with no obvious dominant channel. If you encounter a braided stream, establish the sampling reach using the procedures presented in Table 4-9. Figuring the mean width of extensively braided rivers and streams for purposes of setting up the sample reach length is challenging. For braided channels, measure the mean width and bankfull width as defined in the physical habitat protocols (Sections 5.2 and 6.2). For relatively small streams (mean bankfull width ≤15 m) the sampling reach is defined as 40 times the mean bankfull width. For larger streams (>15 m), sum the actual wetted width of all the braids and use that as the width for calculating the 40 channel width reach length. If there is any question regarding an appropriate reach length for the braided system, it is better to overestimate. Make detailed notes and sketches on the Verification Form (Fig. 4-3 and Fig. 4-4) about what you did. It is important to remember that the purpose of the 40 channel width reach length is to sample enough stream to incorporate the variability in habitat types. Generally, the objective is to sample a long enough stretch of a stream to include 2 to 3 meander cycles (about 6 pool-riffle habitat sequences). In the case of braided systems, the objective of this protocol modification is to avoid sampling an excessively long stretch of stream. In a braided system where there is a 100 m wide active channel (giving a 4 km reach length based on the standard procedure) and only 10 m of wetted width (say five, 2 m wide braids), a 400 m long sample reach length is likely to be sufficient, especially if the system has fairly homogenous habitat throughout its length.

Table 4-9. Modifications for braided streams

1. Estimate the mean width as the bankfull channel width as defined in the physical habitat protocol.
   - If the mean width is ≤15 m, set up a 40 x channel width sample reach in the normal manner.
   - If >15 m, sum up the actual wetted width of all the braids and use that as the width for calculating the 40 x channel width reach length. Remember the minimum reach length is always 150 m.
   - If the reach length seems too short for the system in question, set up a longer sample reach, taking into consideration that the objective is to sample a long enough stretch of a stream to include at least 2 to 3 meander cycles (about 6 pool-riffle habitat sequences).
2. Make detailed notes and sketches on the Verification Form about what you did.

5.0 NON-WADEABLE RIVERS

5.1 Water Quality

This section describes the procedures and methods for the field collection and analysis of the water quality indicators (in-situ measurements, water chemistry, Secchi Disk transparency, and sediment enzymes) from **non-wadeable** streams and rivers. Refer to Appendix E for PPCP water sampling procedures at the designated urban river sites.

5.1.1 In Situ Measurements of Dissolved Oxygen, pH, Temperature, and Conductivity

5.1.1.1 Summary of Method

Measure dissolved oxygen (DO), pH, temperature, and conductivity using a calibrated multi-parameter water quality meter (or sonde). Take the measurements mid-channel at the X-site. Take the readings at 0.5 m depth. Measure the site depth accurately before taking the measurements. Take care to avoid the probe contacting bottom sediments, as the instruments are delicate.

5.1.1.2 Equipment and Supplies

Table 5.1-1 provides the equipment and supplies needed to measure dissolved oxygen, pH, temperature, and conductivity. Record the measurements on the Field Measurement Form, as seen in Figure 5.1-1.

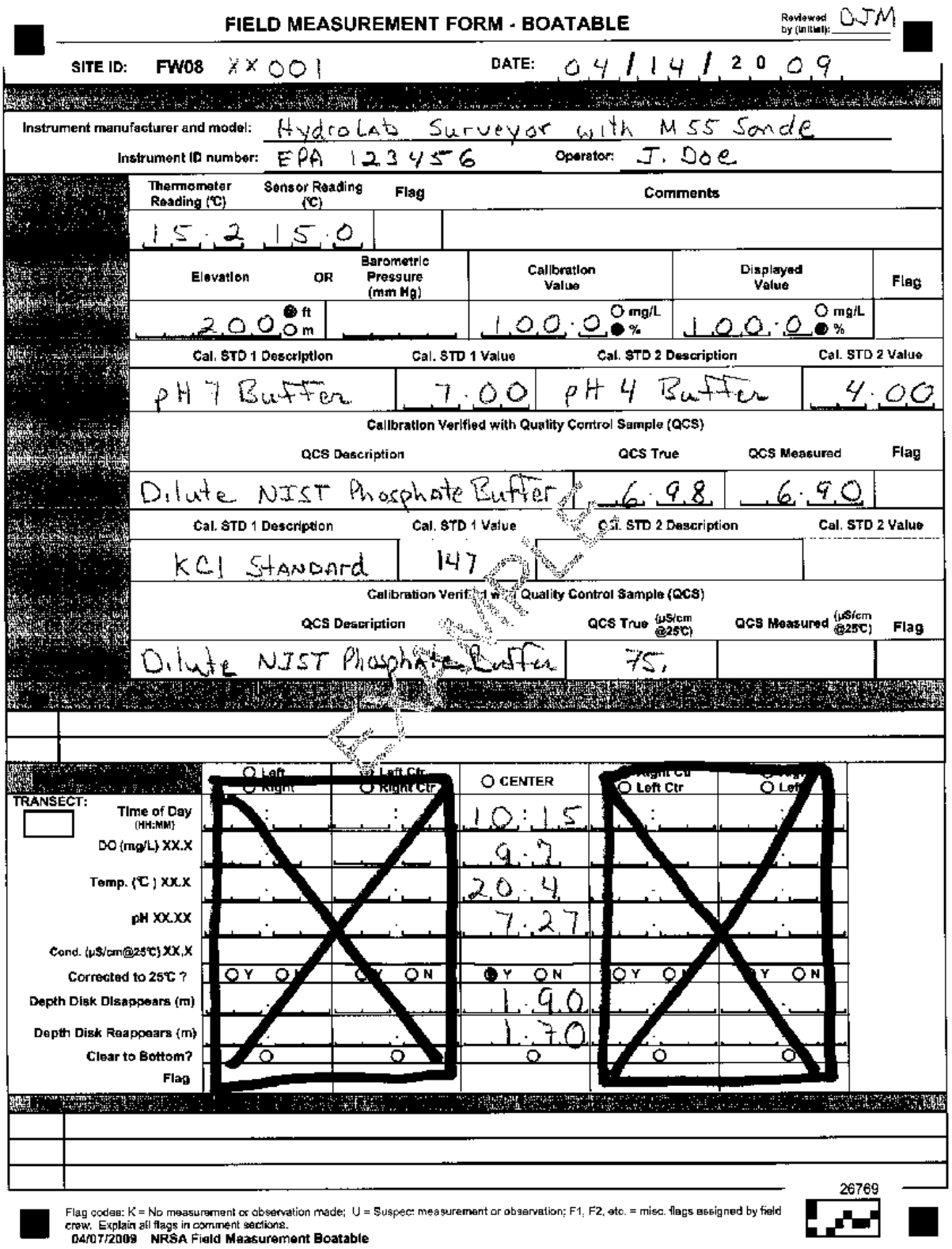
Table 5.1-1. Equipment and supplies—DO, pH, temperature, and conductivity

For taking measurements and calibrating the water quality meter	▪ Multi-parameter water quality meter with pH, DO, temperature, and conductivity probes. ▪ Extra batteries ▪ De-ionized and tap water ▪ Calibration cups and standards ▪ QCS calibration standard ▪ Barometer or elevation chart to use for calibration
For recording measurements	▪ Field Measurement Form ▪ Pencils (for data forms)

5.1.1.3 Multi-Probe Sonde

Dissolved Oxygen Meter

Calibrate the DO meter prior to each sampling event. It is recommended that the probe be calibrated in the field against an atmospheric standard (ambient air saturated with water) prior to launching the boat. In addition, manufacturers typically recommend periodic comparisons with a DO chemical analysis procedure (e.g., Winkler titration) to check accuracy and linearity.



FIELD MEASUREMENT FORM - BOATABLE

Reviewed by (initial): DJM

SITE ID: FW08 XX001 DATE: 04/14/2009

Instrument manufacturer and model: HydroLab Surveyor with MS5 Sonde

Instrument ID number: EPA 123456 Operator: J. Doe

Thermometer Reading (°C)	Sensor Reading (°C)	Flag	Comments
15.2	15.0		

Elevation	OR	Barometric Pressure (mm Hg)	Calibration Value	Displayed Value	Flag
200 ● ft ○ m			100.0 ○ mg/L ● %	100.0 ○ mg/L ● %	

Cal. STD 1 Description	Cal. STD 1 Value	Cal. STD 2 Description	Cal. STD 2 Value
pH 7 Buffer	7.00	pH 4 Buffer	4.00

Calibration Verified with Quality Control Sample (QCS)

QCS Description	QCS True	QCS Measured	Flag
Dilute NIST Phosphate Buffer	6.98	6.90	

Cal. STD 1 Description	Cal. STD 1 Value	Cal. STD 2 Description	Cal. STD 2 Value
KCl Standard	147		

Calibration Verified with Quality Control Sample (QCS)

QCS Description	QCS True (µS/cm @25°C)	QCS Measured (µS/cm @25°C)	Flag
Dilute NIST Phosphate Buffer	75.		

TRANSECT:	○ Left ○ Right	○ Left Ctr ○ Right Ctr	○ CENTER	○ Right Ctr ○ Left Ctr	○ Left
Time of Day (HH:MM)	:	:	10:15	:	:
DO (mg/L) XX.X			9.2		
Temp. (°C) XX.X			20.4		
pH XX.XX			7.27		
Cond. (µS/cm@25°C) XX.X					
Corrected to 25°C ?	○ Y ○ N	○ Y ○ N	● Y ○ N	○ Y ○ N	○ Y ○ N
Depth Disk Disappears (m)			1.90		
Depth Disk Reappears (m)			1.70		
Clear to Bottom?	○	○	○	○	○
Flag					

26769

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = misc. flags assigned by field crew. Explain all flags in comment sections.

04/07/2009 NRSA Field Measurement Boatable

Figure 5.1-1. Field Measurement Form.

pH Meter

Calibrate the pH meter prior to each sampling event. Calibrate the meter in accordance with the manufacturer's instructions and with the team agency's existing SOP. You must also conduct a quality control check with the provided standard to verify the calibration and periodically evaluate instrument precision (see Section 3.1.2). Once a week, each crew must check their multi-probe against the QCS that was in each base kit. Any irregularities must be reported to the Field logistics coordinator immediately.

Temperature Meter

Check the accuracy of the sensor against a thermometer that is traceable to the National Institute of Standards (NIST) at least once per sampling season. The entire temperature range encountered in the NRSA should be incorporated in the testing procedure and a record of test results kept on file.

Conductivity Meter

Calibrate the conductivity meter prior to each sampling event. Calibrate the meter in accordance with the manufacturer's instructions. The entire conductivity range encountered in the NRSA should be incorporated in the testing procedure and a record of test results kept on file. You must also conduct a quality control check with the provided standard to verify the calibration and periodically evaluate instrument precision (see Section 3.1.2). Once a week, each crew must check their multi-probe against the QCS that was in each base kit. Any irregularities must be reported to the Field logistics coordinator immediately.

5.1.1.4 Sampling Procedure

Table 5.1-2 presents step-by-step procedures for measuring dissolved oxygen, pH, temperature, and conductivity.

Table 5.1-2. Sampling procedure—temperature, pH, conductivity and dissolved oxygen.

1. Check meter and probes and calibrate according to manufacturer's specifications.
2. Check the calibration against the provided QCS solution for pH and conductivity and record the results on the field sheet as the QCS Measured value. This should be done at least once a week.
3. Record the true value of the QCS solution from the stock solution container on the field sheet as QCS True.
4. Samples are taken mid-channel, at the X site, at a depth of 0.5 meters or at a mid-depth if less than 1 meter deep.
5. Lower the sonde in the water and measure DO, pH, temperature, and conductivity at 0.5 m depth.
6. Record the measurements on the Field Measurement Form.
7. Flag any measurements that the team feels needs further comment or when a measurement cannot be made.
8. If sampling at the X-site is not possible, move to another part of the reach to take the measurements (as close to the X-site as possible), record the letter of the nearest transect in the "TRANSECT" box and more detailed reasons and/or information in the Comments section.

5.1.2 Water Chemistry Sample Collection and Preservation

5.1.2.1 Summary of Method

The water chemistry samples will be analyzed for total phosphorus (TP), total nitrogen (TN), total ammonia-nitrogen (NH_4), nitrate (NO_3), basic anions, cations, total suspended solids (TSS), turbidity, acid neutralizing capacity (ANC, alkalinity), dissolved organic carbon (DOC), and total organic carbon (TOC). You will also collect a 2-L sample in an amber Nalgene bottle to be filtered on shore for later analysis of *chlorophyll a* (See Section 7 for filtration procedure). Store all samples in darkness on ice in a closed cooler. After you filter the *chlorophyll a* samples, the filters must be kept frozen until ready to ship.

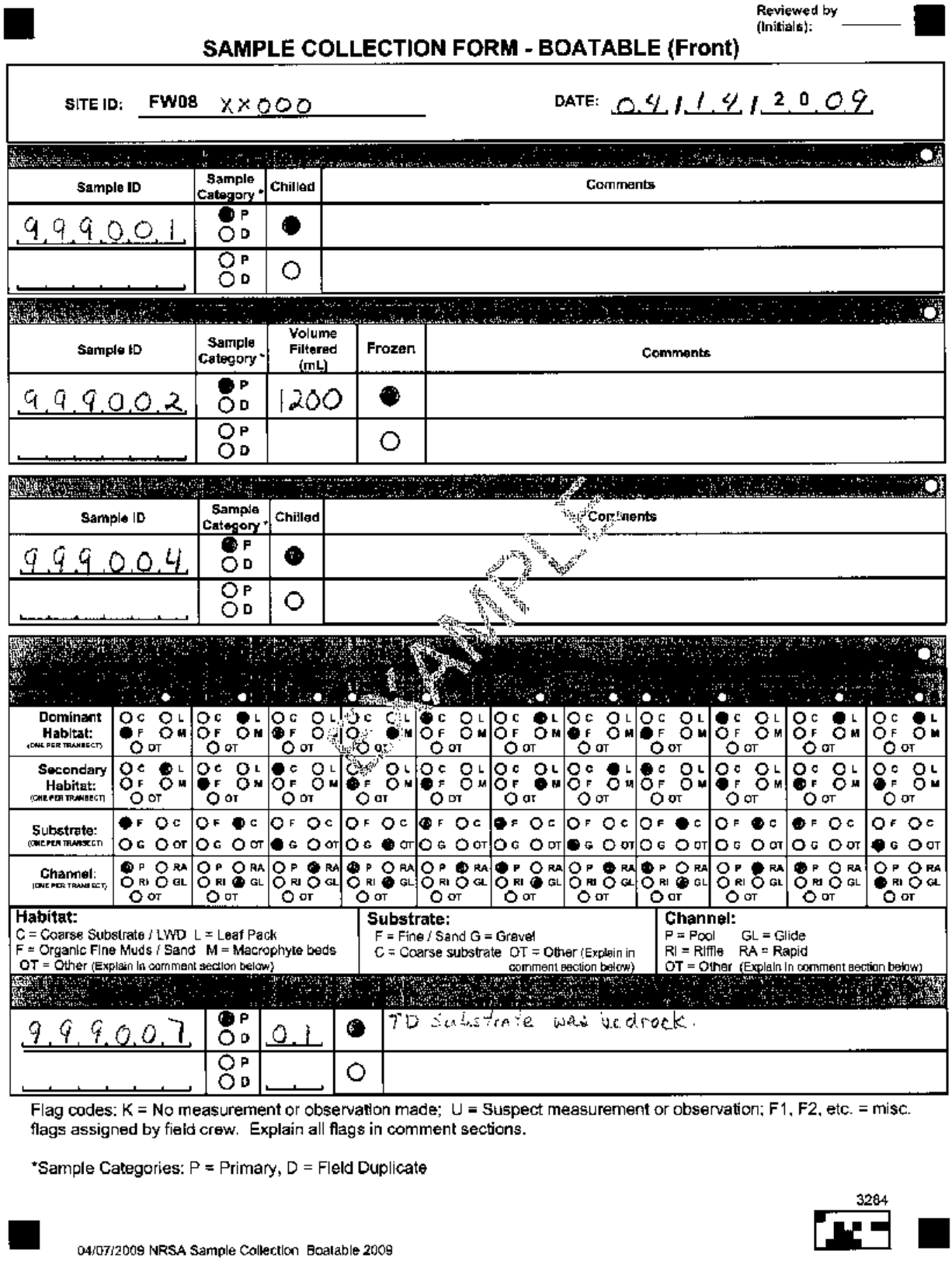
Collect the samples at mid-channel at the X-site of the river from a depth of 0.5 meters. Use the 3 L Nalgene beaker to fill the individual sample bottles. The 3 L Nalgene beaker will be rinsed and re-used at each sampling location.

5.1.2.2 Equipment and Supplies

Table 5.1-3 provides the equipment and supplies needed to collect water samples at the index site. Record the Water Sample Collection and Preservation data on the Sample Collection Form, Side 1 as seen in Figure 5.1-2.

Table 5.1-3. Equipment and supplies—water chemistry sample collection and preservation

For collecting samples	▪ Laser Rangefinder ▪ Nitrile gloves ▪ one 2-L amber Nalgene bottle (*chlorophyll*) ▪ 4-L cube container ▪ 3 L Nalgene beaker ▪ Cooler with ice ▪ Field Operations Manual and/or laminated Quick Reference Guide
For recording measurements	▪ Sample Collection Form ▪ Field Measurement Form ▪ Pencils (for data forms) ▪ fine-tipped indelible markers (for labels)



Reviewed by (Initials): _____

SAMPLE COLLECTION FORM - BOATABLE (Front)

SITE ID: FW08 XX000 DATE: 04/14/2009

Sample ID	Sample Category*	Chilled	Comments
999001	● P ○ D	●	
	○ P ○ D	○	

Sample ID	Sample Category*	Volume Filtered (mL)	Frozen	Comments
999002	● P ○ D	1200	●	
	○ P ○ D		○	

Sample ID	Sample Category*	Chilled	Comments
999004	● P ○ D	●	
	○ P ○ D	○	

Dominant Habitat: (ONE PER TRANSECT)	○ C ○ L ● F ○ M ○ OT	○ C ● L ○ F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	○ C ○ L ○ F ● M ○ OT	● C ○ L ○ F ○ M ○ OT	○ C ● L ○ F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	● C ○ L ○ F ○ M ○ OT	○ C ● L ○ F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	○ C ● L ○ F ○ M ○ OT
Secondary Habitat: (ONE PER TRANSECT)	○ C ● L ○ F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	○ C ○ L ○ F ● M ○ OT	○ C ● L ○ F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT	○ C ○ L ● F ○ M ○ OT
Substrate: (ONE PER TRANSECT)	● F ○ C ○ G ○ OT	○ F ● C ○ G ○ OT	○ F ○ C ● G ○ OT	○ F ○ C ○ G ● OT	● F ○ C ○ G ○ OT	● F ○ C ○ G ○ OT	○ F ○ C ● G ○ OT	○ F ● C ○ G ○ OT	○ F ● C ○ G ○ OT	○ F ● C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ● G ○ OT
Channel: (ONE PER TRANSECT)	● P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ● GL ○ OT	○ P ● RA ○ RI ○ GL ○ OT	● P ○ RA ○ RI ● GL ○ OT	○ P ● RA ○ RI ○ GL ○ OT	● P ○ RA ○ RI ● GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ● RA ○ RI ○ GL ○ OT	● P ○ RA ○ RI ● GL ○ OT	○ P ● RA ○ RI ○ GL ○ OT	● P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ● RI ○ GL ○ OT

Habitat:
C = Coarse Substrate / LWD L = Leaf Pack
F = Organic Fine Muds / Sand M = Macrophyte beds
OT = Other (Explain in comment section below)

Substrate:
F = Fine / Sand G = Gravel
C = Coarse substrate OT = Other (Explain in comment section below)

Channel:
P = Pool GL = Glide
RI = Riffle RA = Rapid
OT = Other (Explain in comment section below)

999007	● P ○ D	0.1	●	TD Substrate was bedrock.
	○ P ○ D		○	

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = misc. flags assigned by field crew. Explain all flags in comment sections.

*Sample Categories: P = Primary, D = Field Duplicate

3284

04/07/2009 NRSA Sample Collection Boatable 2009

Figure 5.1-2. Sample Collection Form, Side 1.

5.1.2.3 Sampling Procedure

Table 5.1-4 describes the sampling procedures for collecting water chemistry samples in non-wadeable streams and rivers. Refer to Appendix E for PPCP water sampling procedures at the designated urban river sites.

Table 5.1-4. Sampling procedure for non-wadeable sites—water chemistry sample collection

1. Collect the water samples from the X-site in a flowing portion near the middle of the stream.
2. Put on nitrile gloves. Make sure not to handle sunscreen or other chemical contaminants until after the sample is collected.
3. Rinse the 3-L Nalgene beaker three times with water, and discard the rinse downstream.
4. Remove the cube container lid and expand the cube container by pulling out the sides. **NOTE: DO NOT BLOW into the cube container to expand them, this will cause contamination.**
5. Fill the 3-liter beaker with water and slowly pour 30 - 50 mL into the cube container. Cap the cube container and rotate so that the water contacts all the surfaces. Discard the water downstream. Repeat this rinsing procedure 2 more times.
6. Fill the beaker with water and pour into the cube container. Repeat as necessary to fill the cube container. Let the weight of the water expand the cube container. Pour the water slowly as the cube container expands. Fill the cube container to at least three-fourths of its maximum volume. Rinse the cube container lid with water. Eliminate any air space from the cube container, and cap it tightly. Make sure the cap is tightly sealed and not on at an angle.
7. Fill the 3-liter beaker with water and slowly pour 30 - 50 mL into the 2 L amber Nalgene bottle. Cap the bottle and rotate so that the water contacts all the surfaces. Discard the water downstream. Repeat this rinsing procedure 2 more times.
8. Fill the beaker with water and pour into the 2 L amber Nalgene bottle. Cap the bottle tightly
9. Place the cube container and bottle in a cooler (on ice or water) and shut the lid. If a cooler is not available, place the cube container in an opaque garbage bag and immerse it in the stream.
10. Record the Sample ID on the Sample Collection Form along with the pertinent stream information (stream name, ID, date, etc.). Note anything that could influence sample chemistry (heavy rain, potential contaminants) in the Comments section. If sampling at the X-site is not possible, move to another part of the reach to collect the sample (as close to the X-site as possible), record the letter of the nearest transect and more detailed reasons and/or information in the Comments section.

5.1.3 Secchi Disk Transparency at Non-Wadeable Sites

5.1.3.1 Summary of Method

A Secchi disk is a black and white patterned disk used to measure water clarity (see Figure 5.1-3). A Secchi disk transparency reading will be collected mid-channel at the X-site. The Secchi disk will be affixed to the end of a solid metered rod (e.g., Schedule 80 PVC pipe, or equivalent) and lowered into the water until it disappears from sight. Measurements are recorded at the depth that the disk disappears and again when it reappears. The reading is taken on the shady side of the boat, without sunglasses, hat or view aids.

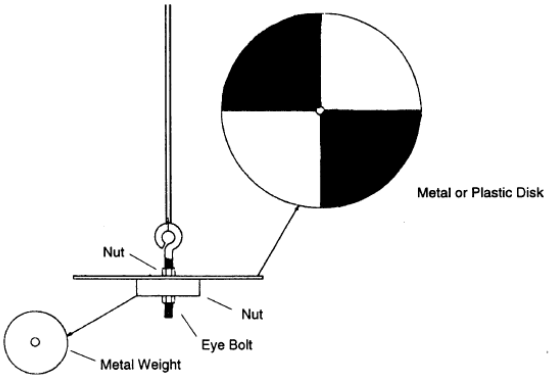


Figure 5.1-3. Secchi disk diagram (EPA, 1991).

5.1.3.2 Equipment and Supplies

Table 5.1-5 lists the equipment and supplies needed to measure Secchi disc transparency. Record the Secchi disk readings on the Field Measurement Form, Side 1 as seen in Figure 5.1-1.

Table 5.1-5. Equipment and supplies—Secchi disc transparency

For taking measurements and calibrating the water quality meter	• 20 cm diameter Secchi disk and calibrated sounding rod (marked in half centimeter intervals) • Tape measure (in centimeters)
For recording measurements	• Field Measurement Form • Pencils (for data forms)

5.1.3.3 Sampling Procedure

Because different people measuring Secchi disk transparency at the same site may obtain different results (due to differences in vision and interpreting disk disappearance and reappearance), one team member will conduct Secchi disk measurements for all sites. Table 5.1-6 lists the procedure for Secchi disk transparency at non-wadeable sites.

If the water is shallow and clear, the Secchi disk might reach the bottom and still be visible. If this is the case, it is important to not stir up the bottom sediments while anchoring the boat. Be sure to move the boat away from the anchor before taking the reading. If the disk is visible at the bottom, indicate this on the form.

Table 5.1-6. Sampling procedure at non-wadeable sites—Secchi disk transparency

1. Measure Secchi disk transparency mid-channel at the X-site.
2. Confirm that the lowering rod is firmly attached to the Secchi disk.
3. Remove sunglasses and hats. Also, **do not** use view scopes or other visual aids. If wearing prescription sunglasses, temporarily replace them with regular clear lens prescription glasses.
4. Lower the Secchi disk over the shaded side of the boat until it disappears.
5. Read the depth indicated on the lowering rod. If the disappearance depth is <1.0 meter, determine the depth to the nearest 0.05 meter by marking the line at the nearest depth marker and measuring the remaining length with a tape measure. Otherwise, estimate the disappearance depth to the nearest 0.1 meter. Record the disappearance depth on the Sample Collection Form.
6. Lower the disk a bit farther and then slowly raise the disk until it reappears and record the reappearance depth on the Field Measurement Form.
7. Note any conditions that might affect the accuracy of the measurement in the comments field.

5.1.4 Sediment Enzymes

5.1.4.1 Summary of Method

Collect sediment samples at the 11 sampling stations at each site and combine all stations at a site, resulting in a single 500 mL sample per site. Collect fine surface sediments (top 5 cm) using a spoon or dredge. Store samples on ice until shipment to the laboratory for processing. Samples will be analyzed for available DIN, NH_4, DIP, TP, TN, total carbon (TC), and enzyme activity.

5.1.4.2 Equipment and Supplies

Table 5.1-7 lists the equipment and supplies needed to collect sediment enzyme samples. Record collection data on Side 2 of the Sample Collection Form, as seen in Figure 5.1-4.

Table 5.1-7. Equipment and supplies—sediment enzymes

For collecting samples	▪ Petite Ponar sampler with plastic tub, drop line, and spare pinch pin Standard Ponar may substitute. ▪ Graduated plastic bucket with lid	▪ Large stainless steel spoon for collecting & mixing sediment composite ▪ 500-mL plastic bottle for storing sediment sample
For recording measurements	▪ Sample Collection Form ▪ Sample labels ▪ Pencils	▪ Fine-tipped indelible markers (for labels) ▪ Clear tape strips

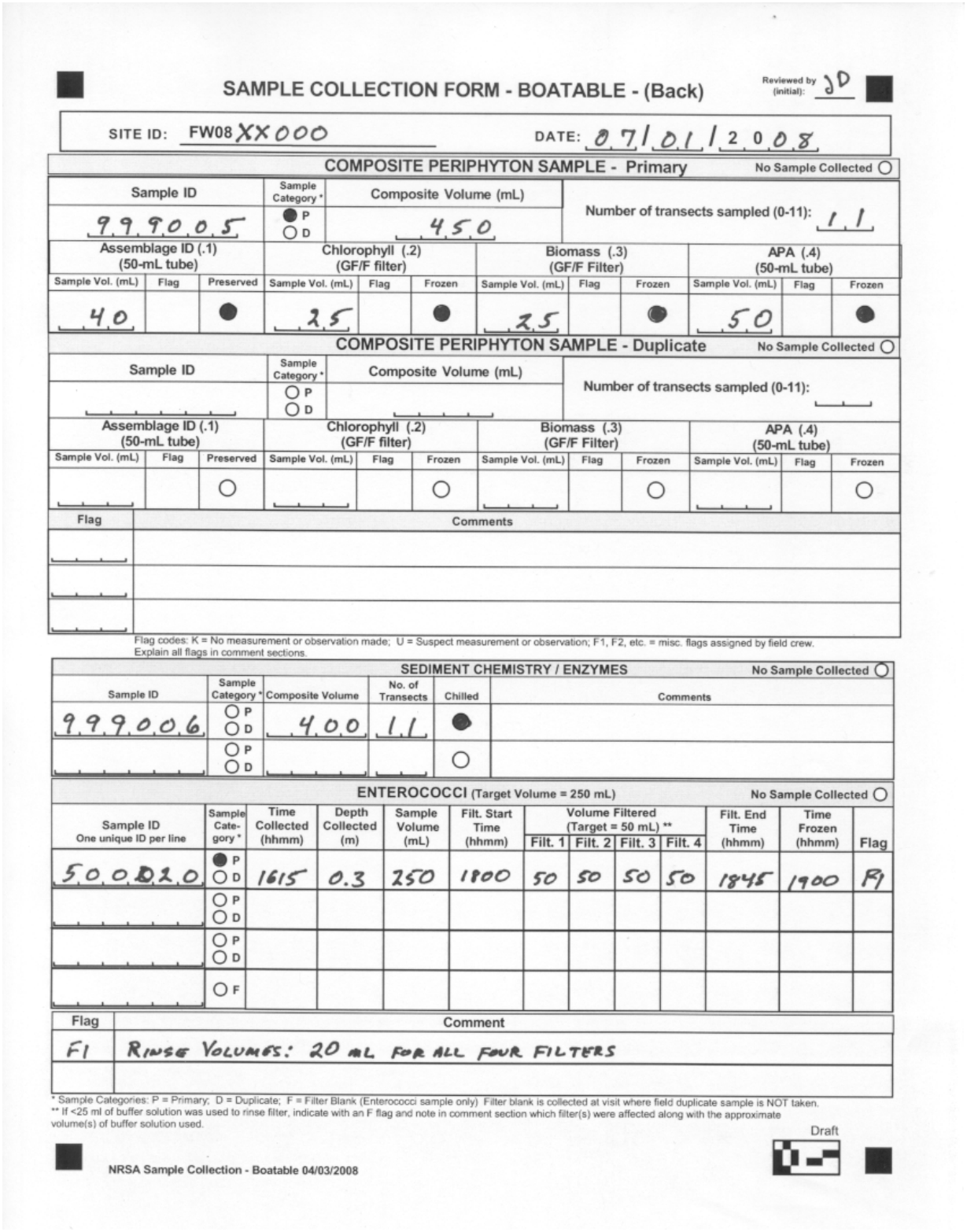
5.1.4.3 Sampling Procedure

Near each of the macroinvertebrate and periphyton sampling locations, collect a fine-grained sediment sample using a spoon. If the depth is too great to reach the bottom with the spoon, a “petite Ponar” grab sampler can be used to collect sediment and the stainless steel

spoon can take the sample to be added to the composite bucket from the ponar. The objective is to collect a 500-mL composite sample that is representative of depositional areas at the site. The composite sample will be subsampled in the laboratory for multiple analyses. Table 5.1-8 presents step-by-step procedures for collecting sediment enzyme samples.

Table 5.1-8. Sampling procedure—sediment enzymes

1. Collect a sediment sample at each of the 11 transect sampling stations, near the periphyton and macroinvertebrate sample locations. Make sure each subsample comprises an approximately equal portion of the total composite. You may collect sediment between stations to insure at least 500 mL of composite volume (note any deviations from standard procedure in a comment.)
2. Locate sediment samples in areas or patches of fine-grained substrate (silty sand, silt, clay, muck) in a zone bounded on the shore side by the apparent low-water mark from daily flow fluctuations (often detected by the presence of periphyton or attached filamentous algae just below the low-water mark) and bounded on the river side by the 0.3-m depth contour (recommended maximum sample depth; deeper sampling may be possible). If samples cannot be safely collected by wading at a station due to vertical banks or other reason go to step 5.
3. Avoid the area that has just been kick sampled for macroinvertebrates. Sampling up-stream from the kick sample location is recommended. If fine substrates are not present within 5 m up- or downstream from the station, flag the station on the form.
4. If fine substrate is present, use the stainless steel spoon to collect a sample (approximately one spoonful of sediment) from the top 5 cm of substrate. Place the sample in a clean bucket. Use gloves for handling sediment. Do not assume rip rapped shorelines lack fine-grained sediment. Look for fines between the large rocks.
5. If the littoral zone cannot be waded, use a petite Ponar (or similar) sampler deployed from the boat to collect a sediment sample adjacent to the station. (*Use caution with Ponar samplers. The jaws are sharp and may close unexpectedly. Replace frayed lines and worn parts.)* Raise the Ponar sampler from the water and into a plastic tub rather than from the boat deck. This prevents feet from getting under the sampler. Release the petite Ponar sample into a tub and use the scoop to collect about 15 x 15 cm (6 x 6 inches) of the top 5 cm of the sample. Using the stainless steel spoon, take a one spoon grab from the top layer of sediment captured in the Ponar. Place this in the composite bucket and discard the rest.
6. Repeat steps 2-5 at each of the 11 littoral stations. Record the total number of replicates (stations) included in the composite. Note in a comment the stations at which sediment was collected using a non-wading method.
7. It is important that a sufficient sediment (not less than 500 mL) composite sample for analysis be collected. If multiple stations have no fine sediment, it is permissible to collect extra sample at stations that do have fine sediment or between stations. *Be sure to note this in a comment.*
8. Using the stainless steel spoon, thoroughly mix the composite sample and transfer 500 mL into the 500 mL plastic bottle. Place in a in a cooler with ice for final labeling and preservation.
9. Prepare a label for the sample jar. Using a fine-point indelible marker, fill in the site # and sample date. Place the label on the jar and cover it with clear tape. Record the sample ID and other data on sampling form. Place the sample on ice or in a refrigerator. Do not freeze sediment samples. The sediment enzyme sample has a two week holding time.



SAMPLE COLLECTION FORM - BOATABLE - (Back)

Reviewed by (initial): JD

SITE ID: FW08XX000 DATE: 07/01/2008

COMPOSITE PERIPHYTON SAMPLE - Primary — No Sample Collected ○

Sample ID	Sample Category *	Composite Volume (mL)	Number of transects sampled (0-11):
999005	● P ○ D	450	11

Assemblage ID (.1) (50-mL tube)			Chlorophyll (.2) (GF/F filter)			Biomass (.3) (GF/F Filter)			APA (.4) (50-mL tube)		
Sample Vol. (mL)	Flag	Preserved	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen
40		●	25		●	25		●	50		●

COMPOSITE PERIPHYTON SAMPLE - Duplicate — No Sample Collected ○

Sample ID	Sample Category *	Composite Volume (mL)	Number of transects sampled (0-11):
	○ P ○ D		

Assemblage ID (.1) (50-mL tube)			Chlorophyll (.2) (GF/F filter)			Biomass (.3) (GF/F Filter)			APA (.4) (50-mL tube)		
Sample Vol. (mL)	Flag	Preserved	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen
		○			○			○			○

Flag	Comments

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = misc. flags assigned by field crew. Explain all flags in comment sections.

SEDIMENT CHEMISTRY / ENZYMES — No Sample Collected ○

Sample ID	Sample Category *	Composite Volume	No. of Transects	Chilled	Comments
999006	○ P ○ D	400	11	●	
	○ P ○ D			○	

ENTEROCOCCI (Target Volume = 250 mL) — No Sample Collected ○

Sample ID One unique ID per line	Sample Category *	Time Collected (hhmm)	Depth Collected (m)	Sample Volume (mL)	Filt. Start Time (hhmm)	Volume Filtered (Target = 50 mL) ** Filt. 1	Filt. 2	Filt. 3	Filt. 4	Filt. End Time (hhmm)	Time Frozen (hhmm)	Flag
500020	● P ○ D	1615	0.3	250	1800	50	50	50	50	1845	1900	F1
	○ P ○ D											
	○ P ○ D											
	○ F											

Flag	Comment
F1	RINSE VOLUMES: 20 mL FOR ALL FOUR FILTERS

* Sample Categories: P = Primary; D = Duplicate; F = Filter Blank (Enterococci sample only) Filter blank is collected at visit where field duplicate sample is NOT taken.
** If <25 ml of buffer solution was used to rinse filter, indicate with an F flag and note in comment section which filter(s) were affected along with the approximate volume(s) of buffer solution used.

Draft

NRSA Sample Collection - Boatable 04/03/2008

Figure 5.1-4. Sample Collection Form, Side 2.

5.2 Physical Habitat Characterization in Non-Wadeable Rivers and Streams

Physical habitat in rivers includes all those physical attributes that influence or provide sustenance to river organisms. Physical habitat varies naturally; thus, expectations differ even in the absence of anthropogenic disturbance. Within a given physiographic-climatic region, river drainage area and channel gradient are likely to be strong natural determinants of many aspects of river habitat, because of their influence on discharge, flood stage, and stream power (the product of discharge times gradient). Kaufmann (1993) identified 7 physical habitat attributes important in influencing stream ecology that are likely applicable in rivers as well. They include:

- Channel Dimensions
- Channel Gradient
- Channel Substrate Size and Type
- Habitat Complexity and Cover
- Riparian Vegetation Cover and Structure
- Anthropogenic Alterations
- Channel-Riparian Interaction

The protocol defines the length of each sampling reach proportional to river wetted width and then systematically places measurements to statistically represent the entire reach. Stream thalweg depth measurements, habitat classification, and mid-channel substrate observations are made at very tightly spaced intervals; whereas channel "littoral" and riparian stations for measuring or observing substrate, fish cover, large woody debris, bank characteristics and riparian vegetation structure are spaced further apart. The tightly spaced depth measures allow calculation of indices of channel structural complexity, objective classification of channel units such as pools, and quantification of residual pool depth, pool volume, and total stream volume.

5.2.1 Equipment and Supplies

Table 5.2-1 lists the equipment and supplies required to conduct all the activities described for characterizing physical habitat. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the river. Use this checklist to ensure that equipment and supplies are organized and available at the river site in order to conduct the activities efficiently.

Table 5.2-1. Checklist of equipment and supplies for physical habitat

For making measurements	▪ Surveyor's telescoping leveling rod (round profile, metric scale, 7.5m extended) ▪ Convex spherical canopy densiometer (Lemmon Model B), modified with taped "V" ▪ GPS ▪ 1 roll each colored surveyor's flagging tape (2 colors) ▪ 2 pair chest waders ▪ 1 or 2 fisherman's vest with lots of pockets and snap fittings. ▪ Digital camera with extra memory card & battery ▪ 50 m or 100 m measuring tape with reel ▪ Meter stick for bank angle measurements ▪ SONAR unit ▪ Laser rangefinder (400 ft. distance range) and clear waterproof bag ▪ Clinometer

	▪ Binoculars ▪ Field Operations Manual and/or laminated quick reference guide ▪ Laminated invasive species guide
For recording data	▪ 2 covered clipboards (lightweight, with strap or lanyard) ▪ Soft (#2) lead pencils ▪ 11 plus extras Channel/Riparian Transect Forms ▪ 11 plus extras Thalweg Profile Forms ▪ 1+ extras field Form: Stream Verification Form ▪ 1+ extras field Form: Field Measurement Form ▪ 1+ extras field Form: Sample Collection Form ▪ 1+ extras field Form: Riparian "Legacy" Trees and Invasive Alien Plants ▪ 1+ extras field Form: Channel Constraint ▪ 1+ extras field Form: Fish Gear and Voucher/Tissue Information Form ▪ 1+ extras field Form: Fish Collection Form ▪ 1+ extras field Form: Visual Assessment Form

5.2.2 Components of the Field Habitat Assessment

Field data collection for the physical habitat assessment is accomplished in a single float down each sampling reach. River sample reach lengths are defined as 40 x the wetted width at the x-site, with a minimum of 150m and maximum of 4km. To characterize mid-channel habitat (Table 5.2.2), they measure a longitudinal thalweg (or mid-channel) depth profile, record the presence of snags and off-channel habitats, classify main channel habitat types, characterize mid-channel substrate, and locate the 11 transect locations for littoral/riparian sampling and other habitat observations. At each of the 11 transects (A-K), they measure channel wetted width, bankfull channel dimensions, incision, GPS lat/long, and then assess near-shore, shoreline, and riparian physical habitat characteristics by measuring or observing littoral depths, riparian canopy cover, substrate, large woody debris, fish cover, bank characteristics, riparian vegetation structure, presence of large ("legacy") riparian trees, non-native riparian and aquatic species, and evidence of human activities. After all the thalweg and littoral/riparian measurements and observations are completed, the crews estimate the extent and type of channel constraint.

Table 5.2-2. Components of river physical habitat protocol

Thalweg Profile:

At 10 equally spaced intervals between each of 11 transects (100 along entire reach):

- Classify habitat type, record presence of backwater and off-channel habitats.

Determine dominant substrate visually or using sounding rod.

At 10 equally spaced intervals between each of 11 transects (100 along entire reach**):**

Record the presence of mid-channel snags
Measure thalweg (maximum) depth using Sonar or rod
Littoral/Riparian Cross-Sections: @ 11 transects at equal intervals along reach length:

Measure/estimate from one chosen bank on 11 transects :

Wetted width and Mid-channel bar width (laser range finder).
Bankfull width (laser) and height (pole and clinometer used as level).
Incision height (pole and clinometer used as level).
Bank angle (estimate)
Riparian canopy cover (densiometer) in four directions from chosen bank.
Shoreline Substrate in the first 1m above waterline (dominant and subdominant size class).

In 20m long Littoral Plot extending streamward 10m from chosen bank : [1]

Littoral depth at 5 locations systematically-spaced within plot (Sonar or sounding rod).
Dominant and Subdominant substrate size class at 5 systematically-spaced locations (visual or sounding rod).
Tally large woody debris in littoral plot and in bankfull channel by size and length class.
Areal cover class of fish concealment and other features, including:

filamentous algae	overhanging vegetation	aquatic macrophytes
undercut banks	large woody debris	boulders and rock ledges
brush/small woody debris	live trees or roots	artificial structures

In 20m long Riparian Plot extending 10m landward starting at bankfull margin--both sides of river:[1]

Estimate areal cover class and type (e.g., woody) of riparian vegetation in Canopy, Mid-Layer, and Ground Cover layers
Observe and record human activities and disturbances and their proximity to the channel.
Record species of alien (non-native) trees, shrubs, grasses visible within riparian plot.

Looking upstream and downstream from each Transect (both sides of river):

Look for largest visible tree within 100m from the water's edge or as far as you can see, if less:
Estimate diameter (Dbh), height, species, and distance from river edge.
For the whole sampling reach, after completing thalweg and littoral/riparian measurements:*

- Classify channel type and degree of constraint, identify features causing constraint, estimate the percentage of constrained channel margin for the whole reach, and estimate the bankfull and valley widths.

[1]Note: Boundaries for visual observations are estimated by eye.

5.2.3 Summary of Workflow

Table 5.2-3 lists the activities performed at and between each transect for the physical habitat characterization. The activities are performed along the chosen river bank and mid-channel (thalweg profile).

Table 5.2-3. Summary of workflow—river physical habitat characterization

A. At the chosen bank on first transect (farthest upstream):

Read GPS Lat./Long. and record it in the Transect (Shoreline) space on the field form.

Move boat in a "loop" within 10 x 20 m littoral plot, measuring 5 littoral depths and probing substrate.

Estimate dominant and subdominant littoral substrate, based on probing the 5 locations.

Estimate areal cover of fish concealment features in 10 x 20 meter littoral plot.

Tally LWD within or partially within the 10 x 20 meter littoral plot.

Do densiometer measurements at bank (facing upstream, downstream, left, right).

Choose bank angle class, estimate bankfull height, width and channel incision. (Note that width and incision estimates incorporate both left and right banks.).

Tally LWD entirely out of water but at least partially within the bankfull channel.

Estimate and record distance to riparian vegetation on the chosen bank.

Make visual riparian vegetation cover estimates for the 10 x 20 meter riparian plot on both sides of the channel. (Riparian plot starts where perennial vegetation begins or at bankfull channel margin, whichever is closest to the wetted river margin. The plot continues 10m back from the bankfull line).

Identify taxa, height, diameter at breast height (Dbh), and distance from riverbank of largest tree as far as you can see confidently upstream and downstream within 100m of the wetted river margin.

From a regional listing, record alien invasive tree, shrub, or grass taxa within in the 10m x 20m riparian plots on either side of the river.

Make visual human disturbance tally on both sides of the river. Use the same plot dimensions as for riparian vegetation -- except that if a disturbance item is observed in the river or within the bankfull channel, the proximity code is "B", the closest rating; "C" if within the riparian plot. If the item is only observed beyond (outside) the riparian plot, the proximity code is "P".

Get out far enough from the bank so you can see downstream. Then use the laser rangefinder to sight and record the distance to the intended position of the next downstream transect.

B. Thalweg Profile:

As soon as you get out from the bank after doing transect activities, take the first of 10 thalweg depth measurements and substrate/snag probes using sonar and pole -- also classify habitat type and record presence of side-channels and backwaters.

Estimate thalweg measurement distance increments using the GPS course-tracking and trip-meter functions. Alternatively, estimate these distances by keeping track of boat lengths or channel-width distances traversed; each one is 1/10th the distance between transects (also one-half channel-width, which can help you keep track of your downstream progress).

C. Repeat the Whole Process (for the remaining 10 transects and spaces in between).

D. Channel Constraint Assessment

After completing the Thalweg Profile and Littoral-Riparian measurements and observations at all 11 Transects, complete the classification and estimation of channel constraint type, frequency of contact with constraining features, and the width ratio of bankfull channel divided by valley width. You may wish to refer to the individual transect assessments of incision and constraint.

5.2.4 Habitat Sampling Locations on the Study Reach

Measurements are made at two scales of resolution along the mid-channel length of the reach; the results are later aggregated and expressed for the entire reach, a third level of resolution (Figure 5.2-1). Section 4 describes the procedures for locating the X-site, or the midpoint of the sample reach. This sampling location is marked on the maps provided to the field crews in the site dossiers prior to sampling. Sections 4.2 and 5.2.3 describe the protocol for delineating a sample reach that is 40 times its width. Those sections also describe the protocol for measuring out (with a laser range finder or GIS software) and locating the 11 littoral/riparian stations where many habitat measurements will be made (Figure 5.2-3). The distance between each of these transects is 1/10th the total length of the sample reach.

The thalweg profile measurements are spaced as evenly as practicable over the entire sample reach length. In addition, they must be sufficiently close together to not "miss" deep areas and habitat units that are in a size range of about 1/3 to 1/2 of the average channel width. To set the interval between thalweg profile measurements, measure the wetted channel width with a laser rangefinder at 5 locations near the X-site and multiply the average width by 40 to set the river sample reach length. Then divide that reach length by 100 to set the thalweg increment distance. Following these guidelines, you will be making 100 evenly-spaced thalweg profile measurements, 10 between each detailed channel cross-section where littoral/riparian observations are made. If the thalweg is too deep or not physically possible to be measured to, estimate the depth to the best of your ability and flag it on the field form.

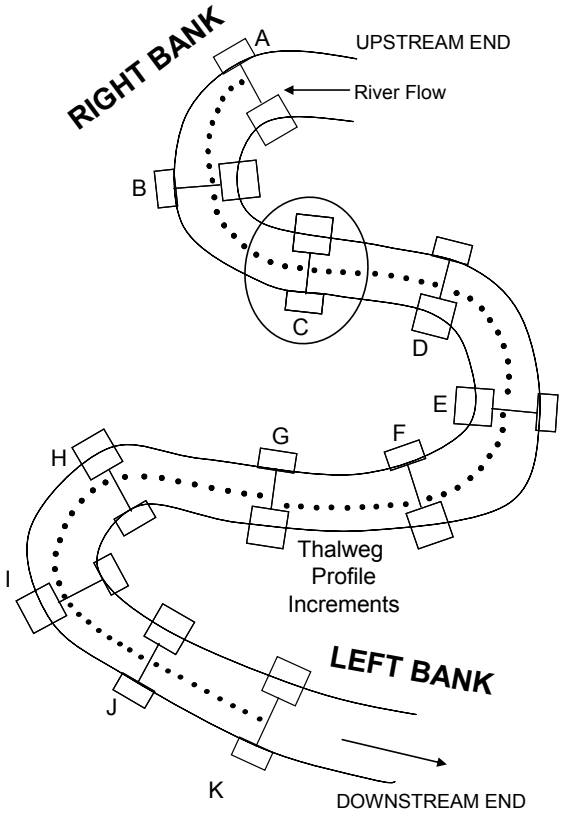


Figure 5.2-1. River reach sample layout.

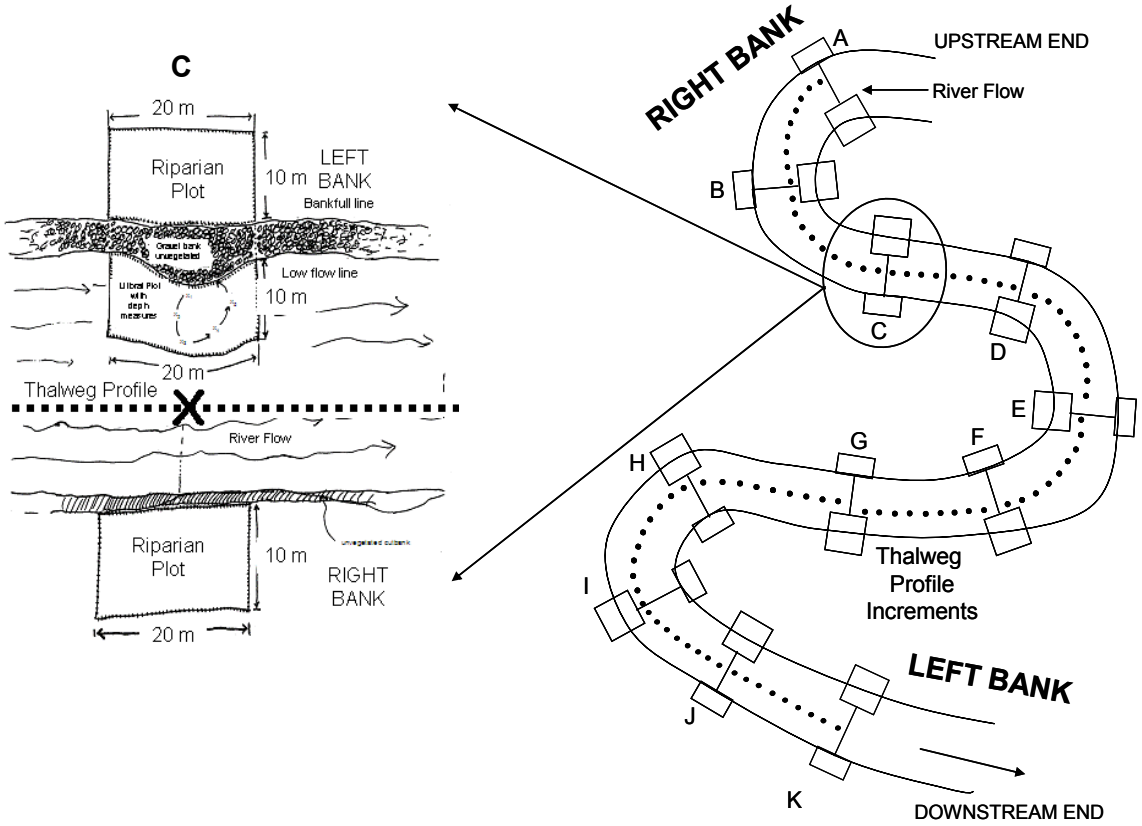


Figure 5.2-2. Littoral-Riparian Plots for characterizing riparian vegetation, human influences, fish cover, littoral substrate, and littoral depths.

5.2.5 Work Flow and Reach Marking

After finding adequate put-in and take-out locations, the team may opt to mark the upstream end of the sample reach end with colored flagging. In a single midstream float down the 40 channel-width reach, the 2-person habitat team accomplishes a reconnaissance, a sonar/pole depth profile, and a pole-drag to tally snags and characterize mid-channel substrate. The float is interrupted by stops at 11 transect locations for littoral/riparian observations. They determine (and mark – optional, but recommended) the intended position of each successive downstream transect using a global positioning system (GPS) or a laser range finder. Each transect is located 4 channel-width's distance from the preceding transect immediately upstream. The crew then floats downstream along the thalweg to the new transect location, making thalweg profile measurements and observations at 10 evenly-spaced increments along the way. When they reach the new downstream transect location, they stop to do cross-section, littoral, and riparian measurements, recording the actual GPS latitude/longitude of the transect position. In addition, while they are stopped at a cross-section station, the crew can fill out the habitat "typing" entries retrospectively and prospectively for the portion of the stream distance that is visible up- and downstream. They will also collect biological and sediment samples.

GPS coordinates are determined for the actual locations of each transect stop. **If GPS unit also has course tracking, trip-meter (accumulated distance and bearing), and waypoint setting/navigation features,** we recommend using it to locate thalweg measurement

points (use course tracking and trip meter). Equipping the boat with a bow or stern anchor to stop at transect locations can greatly ease the shore marking operation and shoreline measurement activities, though such equipment can be dangerous in white-water rivers.

5.2.6 Reconnaissance

The habitat crew will also record reconnaissance and safety notes at this time. They will inform the second boat of the route, craft, and safety precautions needed during its subsequent electrofishing activities. They also assist the electrofishing boat crew over jams and help to conduct shuttles (this can take considerable time where put-ins and take-outs are distant). As the team floats downstream, they may choose and communicate to the electrofishing crew the most practical path to be used when fishing with a less maneuverable boat, taking into consideration multiple channels, blind channels, backwaters, alcoves, impassible riffles, rapids, jams, and hazards such as dams, bridges and power lines. They determine if and where tracking or portages are necessary.

5.2.7 Thalweg Profile

"Thalweg" refers to the flow path of the deepest water in a river channel. The thalweg profile is a longitudinal survey of maximum depth and several other selected characteristics at 100 near-equally spaced points along the centerline of the river between the two ends of the river reach (Figure 5.2-1). For practical reasons, field crews will approximate a thalweg profile by sounding along the river course that they judge is deepest, but also safely navigable. **Locations for observations and measurements along the path of this profile are determined using the GPS course-tracking and trip-meter features (recommended)**, or by visually estimating distances based upon the river width. Data from the thalweg profile allows calculation of indices of residual pool volume, river size, channel complexity, and the relative proportions of habitat types such as riffles and pools. The procedure for obtaining thalweg profile measurements is presented in Table 5.2-2. Record data on the Thalweg Profile Form as shown in Figure 5.2-3.

5.2.7.1 Thalweg Depth Profile

A thalweg depth profile of the entire 40 channel-width reach is approximated by a sonar or sounding rod depth profile while floating downstream along the deepest part of the channel (or closest navigable path). In the absence of a recording fathometer (sonar depth sounder with strip-chart output or electronic data recorder), the crew records depths at frequent, relatively evenly-spaced downstream intervals while observing a sonar display and holding a surveyor's rod off the side of the boat (see Section 5.2.7.2). The sonar screen is mounted so that the crewmember can read depths on the sonar and the rod at the same time. The sonar sensor may need to be mounted at the opposite end of the boat to avoid mistaking the rod's echo for the bottom, though using a narrow beam (16 degree) sonar transducer minimizes this problem. It is easy to hold the sounding rod vertically if you are going at the same speed as the water. If the thalweg is too deep to safely be recorded, estimate the depth and note on comments form.

5.2.7.2 Pole Drag for Snags and Substrate Characteristics

The procedure for dragging the thalweg pole to detect underwater snags and substrate characteristics is presented in Table 5.2-4. While floating downstream, one crewmember holds a calibrated PVC sounding rod or surveying rod down vertically from the gunwale of the boat,

dragging it lightly on the bottom to simultaneously "feel" the substrate, detect snags, and measure depth with the aid of sonar. The crewmember shall record the dominant substrate type sensed by dragging the rod along the bottom (bedrock/hardpan, boulder, cobble, gravel, sand, silt & finer) on the Thalweg Profile Form (Figure 5.2-3). Substrate characteristics are recorded at every thalweg depth measurement (e.g., 10 determinations between transects A and B). In shallow, fast-water situations, where pole-dragging might be hazardous, crews will estimate bottom conditions the best they can visually and by using paddles and oars. If unavoidable, suspend measurements until out of whitewater situations, but make notes and appropriately flag observations concerning your best judgments of depth and substrate.

Table 5.2-4. Thalweg profile procedure

1. Determine the interval between transects based on the mean wetted width used to determine the reach length. Transects are at 4 channel-width spacings; thalweg depth, snags, off-channel habitats and other downstream longitudinal profile observations are recorded at intervals of 0.4 channel-width.

2. Complete header information on the Thalweg Profile Form, noting transect pair (up- to downstream).

3. Begin at the upstream transect (station "1" of "10"). Determine the locations to take measurements using the course-tracking and trip-meter functions of the GPS. Alternatively, estimate your position.

Thalweg Depth Profile

a) While floating downstream along the thalweg, record depths at frequent, even-spaced intervals while observing a sonar display and holding a surveyor's rod off the side of the boat.

b) A depth recording every 0.4 channel-width distance is required, yielding 10 measurements between channel/riparian cross-section transects.

c) If the depth is >0.5 meters, or contains a lot of air bubbles, the sonar fathometer will not give reliable depth estimates. In this case, record depths using a calibrated sounding rod. In shallow, fast-water situations depths may have to be visually estimated to the nearest 0.5 m.

d) Measure depths to nearest 0.1 m and record in the "SONAR" or "POLE" column.

Pole Drag for Snags and Substrate Characteristics

From the gunwale of the boat, hold a surveying rod or calibrated PVC sounding rod down vertically into the water. (CAUTION: Hold the rod over the side or stern of the raft; otherwise it could be jerked out of your hands if it catches on an obstruction in fast water.)

Lightly drag the rod on the river bottom to "feel" the substrate and detect snags.

Record the presence of snags hit by the rod or seen visually, plus the dominant substrate type sensed by dragging the rod along the bottom.

Circle the appropriate "SUBSTRATE" type and record the presence/absence of "SNAGS".

If it is too deep to safely measure the substrate type, estimate the type based on knowledge and surrounding measurements and flag the date.

Channel Habitat Classification

Classify and record the channel habitat type at increments of every 0.4 channel width.

Check for off-channel and backwater habitat at increments of every 0.4 channel width.

If channel is split by a bar or island, navigate and survey the channel with the most flow.

When a side channel is encountered, circle "Y" in the "OFF-CHANNEL" column beginning with the point of divergence from the main channel, continuing downriver until the side channel converges with the main channel.

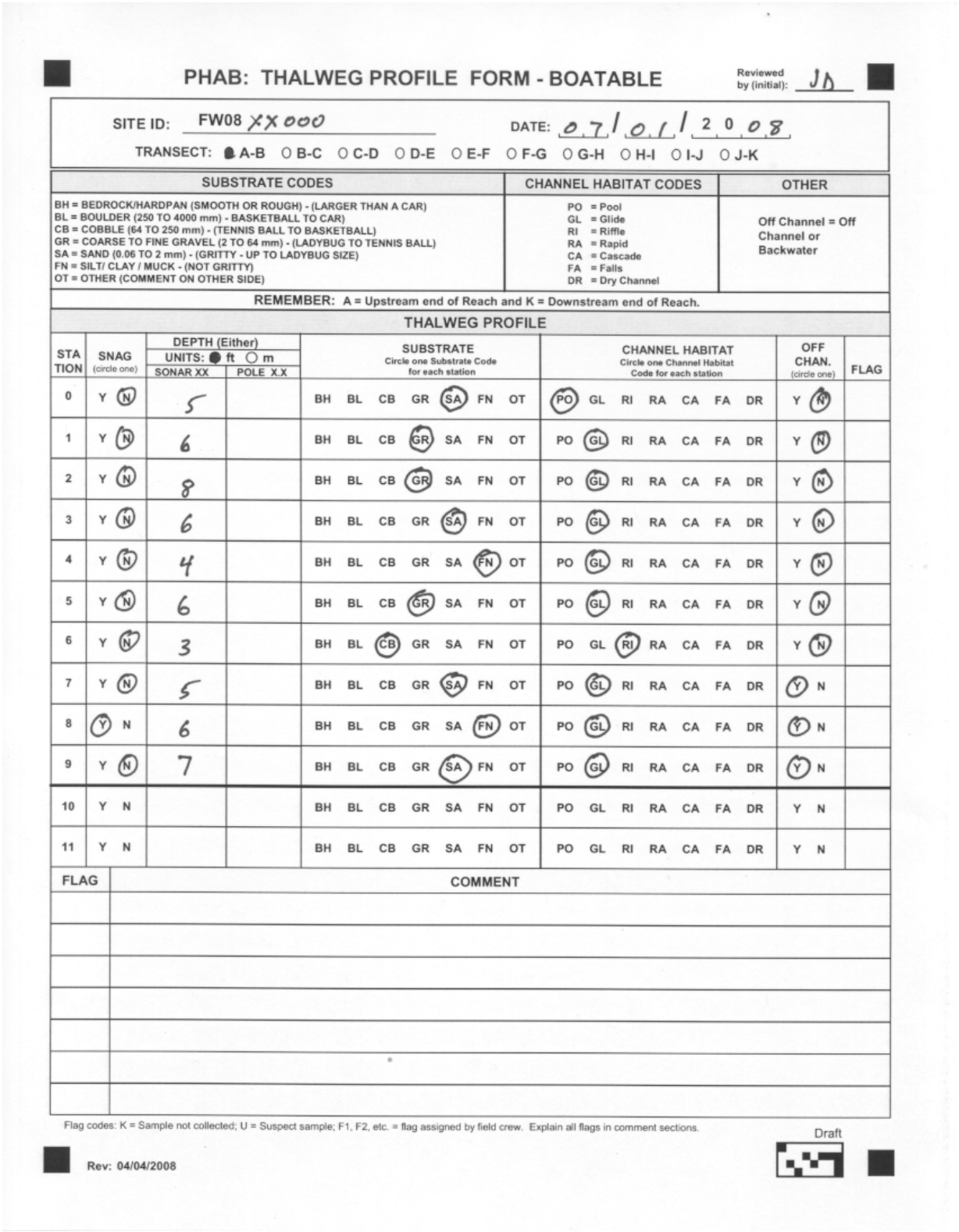
Circle the "CHANNEL HABITAT" and record side channels as described in (d) above.

Proceed downriver to the next station, and repeat the above procedures.

Record GPS waypoint (Lat/Long) midstream and at shoreline location on each transect in decimal degrees.

Repeat the above procedures until you reach next transect. Set a waypoint location for the transect location midstream and at the adjacent bank. Record waypoints that you set for channel bends, transect mid-stream, and transect shoreline locations on the Channel-Riparian Transect Form corresponding to the downstream end of the thalweg sub-reach you just traversed.

After completing activities at the shoreline, prepare a new Thalweg Profile Form, then repeat the above procedures for each of the reach segments, until you reach the downriver end of the reach (Transect "K").



PHAB: THALWEG PROFILE FORM - BOATABLE

Reviewed by (initial): JD

SITE ID: FW08 XX000 DATE: 07/01/2008

TRANSECT: ● A-B ○ B-C ○ C-D ○ D-E ○ E-F ○ F-G ○ G-H ○ H-I ○ I-J ○ J-K

SUBSTRATE CODES	CHANNEL HABITAT CODES	OTHER
BH = BEDROCK/HARDPAN (SMOOTH OR ROUGH) - (LARGER THAN A CAR) BL = BOULDER (250 TO 4000 mm) - BASKETBALL TO CAR) CB = COBBLE (64 TO 250 mm) - (TENNIS BALL TO BASKETBALL) GR = COARSE TO FINE GRAVEL (2 TO 64 mm) - (LADYBUG TO TENNIS BALL) SA = SAND (0.06 TO 2 mm) - (GRITTY - UP TO LADYBUG SIZE) FN = SILT/ CLAY / MUCK - (NOT GRITTY) OT = OTHER (COMMENT ON OTHER SIDE)	PO = Pool GL = Glide RI = Riffle RA = Rapid CA = Cascade FA = Falls DR = Dry Channel	Off Channel = Off Channel or Backwater

REMEMBER: A = Upstream end of Reach and K = Downstream end of Reach.

THALWEG PROFILE

STATION	SNAG (circle one)	DEPTH (Either) UNITS: ● ft ○ m — SONAR XX	DEPTH (Either) UNITS: ● ft ○ m — POLE X.X	SUBSTRATE Circle one Substrate Code for each station	CHANNEL HABITAT Circle one Channel Habitat Code for each station	OFF CHAN. (circle one)	FLAG
0	Y (N)	5		BH BL CB GR (SA) FN OT	(PO) GL RI RA CA FA DR	Y (N)	
1	Y (N)	6		BH BL CB (GR) SA FN OT	PO (GL) RI RA CA FA DR	Y (N)	
2	Y (N)	8		BH BL CB (GR) SA FN OT	PO (GL) RI RA CA FA DR	Y (N)	
3	Y (N)	6		BH BL CB GR (SA) FN OT	PO (GL) RI RA CA FA DR	Y (N)	
4	Y (N)	4		BH BL CB GR SA (FN) OT	PO (GL) RI RA CA FA DR	Y (N)	
5	Y (N)	6		BH BL CB (GR) SA FN OT	PO (GL) RI RA CA FA DR	Y (N)	
6	Y (N)	3		BH BL (CB) GR SA FN OT	PO GL (RI) RA CA FA DR	Y (N)	
7	Y (N)	5		BH BL CB GR (SA) FN OT	PO (GL) RI RA CA FA DR	(Y) N	
8	(Y) N	6		BH BL CB GR SA (FN) OT	PO (GL) RI RA CA FA DR	(Y) N	
9	Y (N)	7		BH BL CB GR (SA) FN OT	PO (GL) RI RA CA FA DR	(Y) N	
10	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
11	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	

FLAG	COMMENT

Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, etc. = flag assigned by field crew. Explain all flags in comment sections.

Draft

Rev: 04/04/2008

Figure 5.2-3. Thalweg Profile Form.

5.2.7.3 Channel Habitat Classification

Classify and record channel habitat types shown in Table 5.2-5 at a spatial resolution of about 0.5 channel-widths and check presence of off-channel and backwater habitat at every 0.4 channel-width increment. The procedures for classifying channel habitat are presented in Table 5.2-2. Designate side channels, backwaters and other off-channel areas independent of the main-channel habitat type. Main channel habitat units are at least half as long as the channel is wide. (e.g., if there is a small, deep, pool-like area at the thalweg within a large riffle area, don't record it as a pool unless it occupies an area about half as wide or long as the channel is wide).

Table 5.2-5 Channel unit categories

Class (Code)[a]	Description
Pools (PO):	Still water, low velocity, smooth, surface, deep compared to other parts of channel
Glide (GL)	Water moving slowly, with a smooth, unbroken surface. Low turbulence.
Riffle (RI)	Water moving, with small ripples, waves and eddies—waves not breaking, surface tension not broken. Sound: "babbling", "gurgling".
Rapid (RA)	Water movement rapid and turbulent, surface with intermittent whitewater with breaking waves. Sound: continuous rushing, but not as loud as cascade.
Cascade (CA)	Water movement rapid & very turbulent over steep channel bottom. Most of the water surface is broken in short, irregular plunges, mostly whitewater. Sound: roaring.
Falls (FA)	Free falling water over vertical or near vertical drop into plunge, water turbulent and white over high falls. Sound: splash to roar. (Do not navigate raft over a waterfall!).
Dry channel (DR)	No water in the channel.
Off-channel	Side-channels, sloughs, backwaters, and alcoves separated from the main channel.

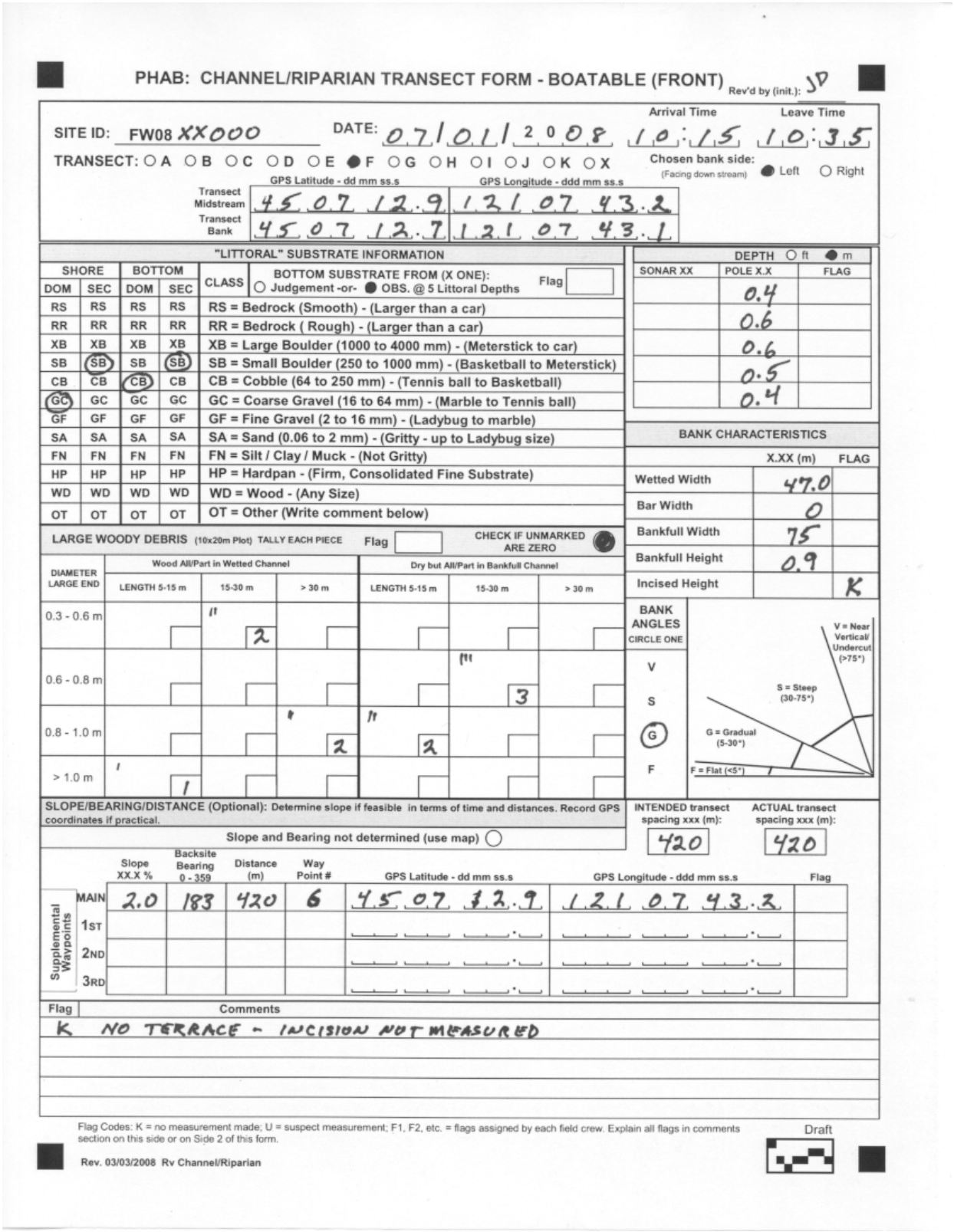
[a] In order for a channel habitat unit to be distinguished, it must be at least half as wide or long as the channel is wide.

Mid-channel bars, islands, and side channels within a thalweg profile require some guidance. Mid-channel bars are defined as channel features below the bankfull flow level that are dry during baseflow conditions (Section 5.2.8.3 defines bankfull channel). Islands are channel features that are dry even when the river is at bankfull flow. If a mid-channel feature is as high as the surrounding flood plain, it is considered an island. Both mid-channel bars and islands cause the river to split into side channels. If a bar or island is encountered along the thalweg profile, navigate and survey the channel that carries the most flow. Note side channels are present but do not sample them.

When side channels are present, on the Thalweg Profile form check the "Off-Channel" column. These checkmarks will begin at the point of divergence from the main channel, continuing downstream to the point of convergence with the main channel. In the case of a slough or alcove, the "off-channel" checkmarks should continue from the point of divergence downstream to where the off-channel feature is no longer evident. When major side channels occur, flag the "Off-Channel" checkmarks and indicate in the comments section that the feature is a side channel. For dry and intermittent rivers, record zeros for depth and wetted width in places where no water is in the channel. Record habitat type as dry channel (DR).

5.2.8 Channel Margin ("Littoral") and Riparian Measurements

This section covers channel margin depth and substrate, large woody debris, bank angle, channel cross-section morphology, canopy cover, riparian vegetation structure, fish cover, and human influences. Record measurements on the Channel/Riparian Transect Form (Figures 5.2-4 and 5.2-5).



PHAB: CHANNEL/RIPARIAN TRANSECT FORM - BOATABLE (FRONT) Rev'd by (init.): JP

SITE ID: FW08 XX000 DATE: 07/01/2008 Arrival Time 10:15 Leave Time 10:35

TRANSECT: ○A ○B ○C ○D ○E ●F ○G ○H ○I ○J ○K ○X Chosen bank side: (Facing down stream) ● Left ○ Right

	GPS Latitude - dd mm ss.s	GPS Longitude - ddd mm ss.s
Transect Midstream	45 07 12.9	121 07 43.2
Transect Bank	45 07 12.7	121 07 43.1

"LITTORAL" SUBSTRATE INFORMATION

SHORE DOM	SHORE SEC	BOTTOM DOM	BOTTOM SEC	CLASS
RS	RS	RS	RS	RS = Bedrock (Smooth) - (Larger than a car)
RR	RR	RR	RR	RR = Bedrock (Rough) - (Larger than a car)
XB	XB	XB	XB	XB = Large Boulder (1000 to 4000 mm) - (Meterstick to car)
SB	(SB)	SB	(SB)	SB = Small Boulder (250 to 1000 mm) - (Basketball to Meterstick)
CB	CB	(CB)	CB	CB = Cobble (64 to 250 mm) - (Tennis ball to Basketball)
(GC)	GC	GC	GC	GC = Coarse Gravel (16 to 64 mm) - (Marble to Tennis ball)
GF	GF	GF	GF	GF = Fine Gravel (2 to 16 mm) - (Ladybug to marble)
SA	SA	SA	SA	SA = Sand (0.06 to 2 mm) - (Gritty - up to Ladybug size)
FN	FN	FN	FN	FN = Silt / Clay / Muck - (Not Gritty)
HP	HP	HP	HP	HP = Hardpan - (Firm, Consolidated Fine Substrate)
WD	WD	WD	WD	WD = Wood - (Any Size)
OT	OT	OT	OT	OT = Other (Write comment below)

BOTTOM SUBSTRATE FROM (X ONE): ○ Judgement -or- ● OBS. @ 5 Littoral Depths Flag

DEPTH ○ ft ● m

SONAR XX	POLE X.X	FLAG
	0.4	
	0.6	
	0.6	
	0.5	
	0.4	

BANK CHARACTERISTICS

	X.XX (m)	FLAG
Wetted Width	47.0	
Bar Width	0	
Bankfull Width	75	
Bankfull Height	0.9	
Incised Height		K

LARGE WOODY DEBRIS (10x20m Plot) TALLY EACH PIECE Flag CHECK IF UNMARKED ARE ZERO

DIAMETER LARGE END	Wood All/Part in Wetted Channel: LENGTH 5-15 m	15-30 m	> 30 m	Dry but All/Part in Bankfull Channel: LENGTH 5-15 m	15-30 m	> 30 m
0.3 - 0.6 m		II 2				
0.6 - 0.8 m					III 3	
0.8 - 1.0 m			I 2	II 2		
> 1.0 m	I 1					

BANK ANGLES CIRCLE ONE: V, S, (G), F

SLOPE/BEARING/DISTANCE (Optional): Determine slope if feasible in terms of time and distances. Record GPS coordinates if practical.

Slope and Bearing not determined (use map) ○

INTENDED transect spacing xxx (m): 420 ACTUAL transect spacing xxx (m): 420

	Slope XX.X %	Backsite Bearing 0 - 359	Distance (m)	Way Point #	GPS Latitude - dd mm ss.s	GPS Longitude - ddd mm ss.s	Flag
MAIN	2.0	183	420	6	45 07 12.9	121 07 43.2	
Supplemental Waypoints 1ST							
2ND							
3RD							

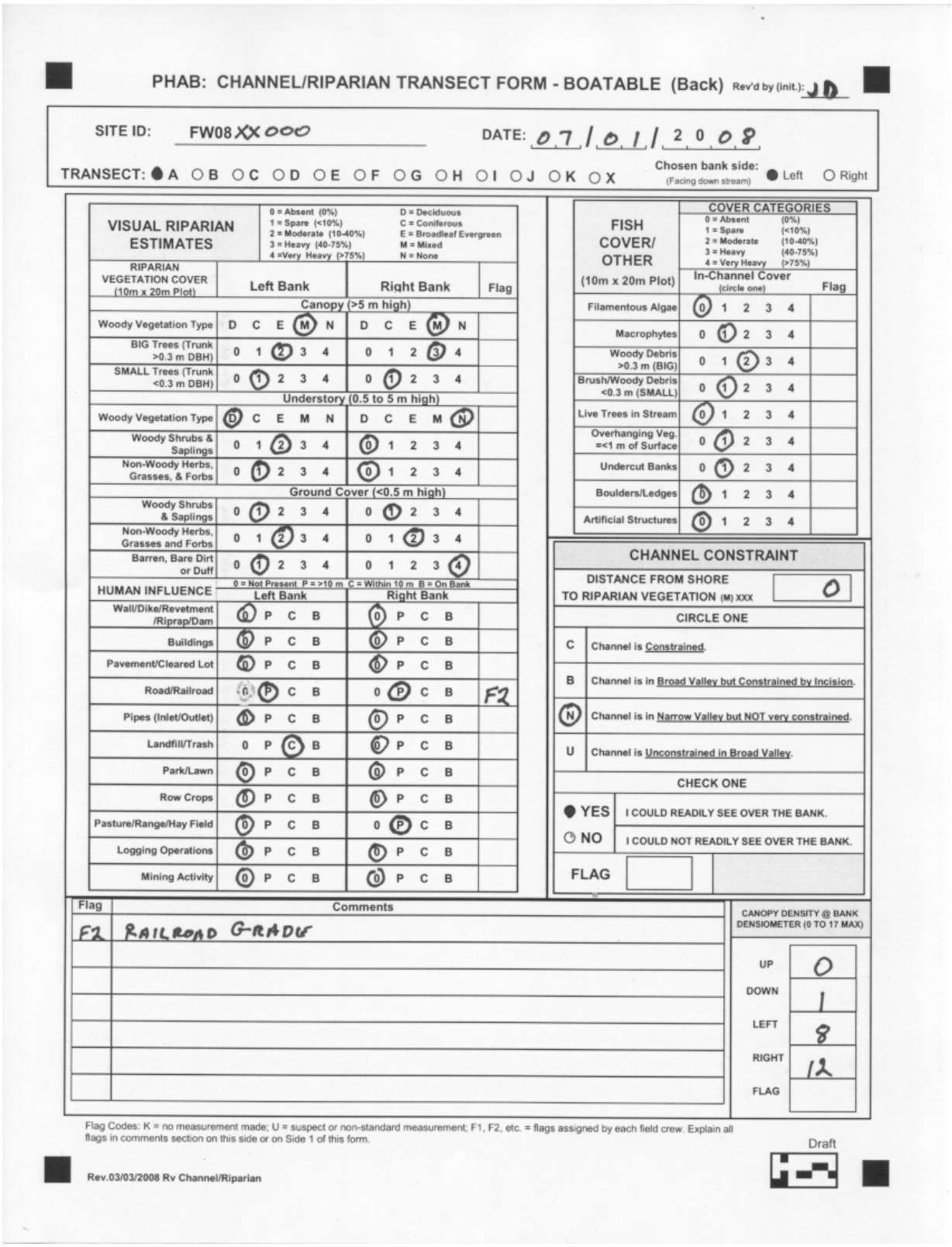
Flag	Comments
K	NO TERRACE - INCISION NOT MEASURED

Flag Codes: K = no measurement made; U = suspect measurement; F1, F2, etc. = flags assigned by each field crew. Explain all flags in comments section on this side or on Side 2 of this form.

Rev. 03/03/2008 Rv Channel/Riparian

Draft

Figure 5.2-4. Channel/Riparian Transect Form, page 1 (front side).



PHAB: CHANNEL/RIPARIAN TRANSECT FORM - BOATABLE (Back) Rev'd by (init.): JD

SITE ID: FW08XX000 DATE: 07/01/2008

TRANSECT: ● A ○ B ○ C ○ D ○ E ○ F ○ G ○ H ○ I ○ J ○ K ○ X Chosen bank side: (Facing down stream) ● Left ○ Right

VISUAL RIPARIAN ESTIMATES

0 = Absent (0%); 1 = Spare (<10%); 2 = Moderate (10-40%); 3 = Heavy (40-75%); 4 = Very Heavy (>75%)
D = Deciduous; C = Coniferous; E = Broadleaf Evergreen; M = Mixed; N = None

RIPARIAN VEGETATION COVER (10m x 20m Plot)	Left Bank	Right Bank	Flag
Canopy (>5 m high)			
Woody Vegetation Type	D C E (M) N	D C E (M) N	
BIG Trees (Trunk >0.3 m DBH)	0 1 (2) 3 4	0 1 2 (3) 4	
SMALL Trees (Trunk <0.3 m DBH)	0 (1) 2 3 4	0 (1) 2 3 4	
Understory (0.5 to 5 m high)			
Woody Vegetation Type	(D) C E M N	D C E M (N)	
Woody Shrubs & Saplings	0 1 (2) 3 4	(0) 1 2 3 4	
Non-Woody Herbs, Grasses, & Forbs	0 (1) 2 3 4	(0) 1 2 3 4	
Ground Cover (<0.5 m high)			
Woody Shrubs & Saplings	0 (1) 2 3 4	0 (1) 2 3 4	
Non-Woody Herbs, Grasses and Forbs	0 1 (2) 3 4	0 1 (2) 3 4	
Barren, Bare Dirt or Duff	0 (1) 2 3 4	0 1 2 3 (4)	
HUMAN INFLUENCE	0 = Not Present P = >10 m C = Within 10 m B = On Bank		
	Left Bank	Right Bank	
Wall/Dike/Revetment /Riprap/Dam	(0) P C B	(0) P C B	
Buildings	(0) P C B	(0) P C B	
Pavement/Cleared Lot	(0) P C B	(0) P C B	
Road/Railroad	(0) (P) C B	0 (P) C B	F2
Pipes (Inlet/Outlet)	(0) P C B	(0) P C B	
Landfill/Trash	0 P (C) B	(0) P C B	
Park/Lawn	(0) P C B	(0) P C B	
Row Crops	(0) P C B	(0) P C B	
Pasture/Range/Hay Field	(0) P C B	0 (P) C B	
Logging Operations	(0) P C B	(0) P C B	
Mining Activity	(0) P C B	(0) P C B	

FISH COVER/ OTHER (10m x 20m Plot)

COVER CATEGORIES: 0 = Absent (0%); 1 = Spare (<10%); 2 = Moderate (10-40%); 3 = Heavy (40-75%); 4 = Very Heavy (>75%)

In-Channel Cover (circle one)		Flag
Filamentous Algae	(0) 1 2 3 4	
Macrophytes	0 (1) 2 3 4	
Woody Debris >0.3 m (BIG)	0 1 (2) 3 4	
Brush/Woody Debris <0.3 m (SMALL)	0 (1) 2 3 4	
Live Trees in Stream	(0) 1 2 3 4	
Overhanging Veg. =<1 m of Surface	0 (1) 2 3 4	
Undercut Banks	0 (1) 2 3 4	
Boulders/Ledges	(0) 1 2 3 4	
Artificial Structures	(0) 1 2 3 4	

CHANNEL CONSTRAINT

DISTANCE FROM SHORE TO RIPARIAN VEGETATION (M) XXX: 0

CIRCLE ONE

- C — Channel is Constrained.
- B — Channel is in Broad Valley but Constrained by Incision.
- (N) — Channel is in Narrow Valley but NOT very constrained.
- U — Channel is Unconstrained in Broad Valley.

CHECK ONE

- ● YES — I COULD READILY SEE OVER THE BANK.
- ○ NO — I COULD NOT READILY SEE OVER THE BANK.

FLAG:

Flag	Comments
F2	RAILROAD GRADE

CANOPY DENSITY @ BANK DENSIOMETER (0 TO 17 MAX)

UP	0
DOWN	1
LEFT	8
RIGHT	12
FLAG	

Flag Codes: K = no measurement made; U = suspect or non-standard measurement; F1, F2, etc. = flags assigned by each field crew. Explain all flags in comments section on this side or on Side 1 of this form.

Draft

Rev.03/03/2008 Rv Channel/Riparian

Figure 5.2-5. Channel/Riparian Transect Form, page 2 (back side).

5.2.8.1 Channel Margin Depth and Substrate

Channel margin depths are measured along the designated shoreline at each transect within the 10m x 20m littoral plot that is centered on the transect. Dominant and sub-dominant bottom substrates are determined and recorded at 5 systematically-spaced locations that are located by eye within the 10m x 20m plot. The procedure for obtaining channel margin depth and substrate measurements is described in more detail in Table 5.2-6. Record these measurements on the Channel/Riparian Transect Form as shown in Figure 5.2-4. Identify the dominant and subdominant substrate present along a shoreline swath 20 meters long and 1 meter back from the waterline. The substrate size class choices are as shown in Table 5.2-6.

Table 5.2-6. Channel margin depth and substrate procedure

1. Fill in the header information on page 1 of a Channel/Riparian Transect Form. Be sure to indicate the letter designating the transect location.
2. Measure depth and observe bottom substrates within the 10m x 20 m littoral plot that is centered on each transect location.
3. Determine and record the depth and the dominant and subdominant substrate size class at 5 systematically-spaced locations estimated by eye within this 10m x 20m plot and 1m back from the waterline. **If the substrate particle is "artificial" (e.g. concrete, asphalt), choose the appropriate size class, flag the observation and note that it is artificial in the comment space.**

Code	Size Class	Size Range (mm)	Description
RS	Bedrock (Smooth)	>4000	Smooth surface rock bigger than a car
RR	Bedrock (Rough)	>4000	Rough surface rock bigger than a car
XB	Large Boulders	>1000 to 4000	Meter stick to Car size
SB	Small Boulders	>250 to 1000	Basketball to Meter stick size
CB	Cobbles	>64 to 250	Tennis ball to basketball size
GC	Gravel (Coarse)	>16 to 64	Marble to tennis ball size
GF	Gravel (Fine)	> 2 to 16	Ladybug to marble size
SA	Sand	>0.06 to 2	Gritty – up to ladybug size,
FN	Fines	<0.06	Silt Clay Muck (not gritty between fingers)
HP	Hardpan		Firm, consolidated fine substrate
WD	Wood	Regardless of Size	Wood & other organic particles
OT	Other	Regardless of Size	Concrete, metal, tires, etc. (note in comments)

4. On page 1 of the Channel/Riparian Transect Form, circle the appropriate shore and bottom substrate type and record the depth measurements ("SONAR" or "POLE" columns).
5. Repeat Steps 1 through 4 at each new cross-section transect.

5.2.8.2 Large Woody Debris

Large Woody Debris (LWD) is defined as woody material with small end diameter of ≥30 cm (1ft) and length of ≥5 m (15 ft). These size criteria are larger than those used in wadeable streams because of the lesser role that small wood plays in controlling velocity and morphology of larger rivers. The procedure for tallying LWD is presented in Table 5.2-7. For each tally (Wood All/Part in Wetted Channel and Dry but All/Part in Bankfull Channel), the field form (Figure 5.2-4) provides 12 entry boxes for tallying debris pieces visually estimated within three length and four diameter class combinations. Tally each LWD piece in only one box. Do not tally

woody debris in the area between channel cross-sections, but the presence and location of large debris dams and accumulations should be mapped (sketched) and noted in the thalweg profile comments.

For each LWD piece, first visually estimate its length and its large and small end diameters and place it in one of the diameter and length categories. The diameter classes on the field form (Figure 5.2-4) refer to the large end diameter. Sometimes LWD is not cylindrical, so it has no clear "diameter". In these cases visually estimate what the diameter would be for a piece of wood with circular cross-section that would have the same volume. When evaluating length, include only the part of the LWD piece that has a diameter >0.3m (1 ft). Count each of the LWD pieces as one tally entry and include the whole piece when assessing dimensions, even if part of it is outside of the bankfull channel. If you encounter massive, complex debris jams, estimate their length, width, and height. Estimate the diameter and length of large "key" pieces and judge the average diameter and length of the other pieces making up the jam. Record this information in the comments section of the form.

Table 5.2-7. Procedure for tallying large woody debris

Note: Tally pieces of large woody debris (LWD) within the 11 transects of the river reach at the same time the shoreline measurements are being determined. Include all pieces whose large end is located within the transect plot in the tally. Tally wood that is at least partially within the wetted channel separately from that that is not presently wetted, but still within or directly above (bridging) the bankfull channel

1. LWD is tallied in 11 "plots" systematically spaced over the entire length of the stream reach. These plots are each 20 m long in the upstream-downstream direction (10m up, 10m down). They are positioned along the chosen bank and extend from the shore in 10m towards mid-channel and then all the way to the bankfull margin.
2. Tally all LWD pieces within the plot that are at least partially within the presently wetted (baseflow) channel. First, determine if a piece is large enough to be classified as LWD (**small end diameter 30 cm [1 ft.]; length 5 m [15 ft.]**)
3. For each piece of LWD, determine its diameter class **based on the diameter of the large end** (0.3 m to < 0.6 m, 0.6 m to <0.8 m, 0.8 m to <1.0 m, or >1.0 m), and the **length class of the LWD pieces based on the part of its length that has diameter ≥30 cm.** Length classes are 5m to <15m, 15m to <30m, or >30m.
   - If the piece is not cylindrical, visually estimate what the diameter would be for a piece of wood with circular cross-section that would have the same volume.
   - When estimating length, include only the part of the LWD piece that has a diameter >0.3 m (1 ft.)
4. Place a tally mark in the appropriate diameter × length class tally box in the "**WOOD ALL/PART IN WETTED CHANNEL**" section of the Channel/Riparian Transect Form.
5. Tally all shoreline LWD pieces along the littoral plot that are at least partially within or above (bridging) the bankfull channel, but not in the wetted channel. For each piece, determine the diameter class based on the diameter of the **large end** (0.3 m to < 0.6 m, 0.6 m to <0.8 m, 0.8 m to <1.0 m, or >1.0 m), and the **length class based on the length of the piece that has diameter ≥30 cm.** Length classes are 5m to <15m, 15m to <30m, or >30m.
6. Place a tally mark for each piece in the appropriate diameter × length class tally box in the "**DRY BUT ALL/PART IN BANKFULL CHANNEL**" section of the Channel/Riparian Transect Form.
7. After all pieces within the segment have been tallied, write the total number of pieces for each diameter × length class in the small box at the lower right-hand corner of each tally box.
8. Repeat Steps 1 through 7 for the next river transect, using a new Channel/Riparian Transect Form.

5.2.8.3 Bank Angle and Channel Cross-Section Morphology

Bank angles of undercut, vertical, steep, and gradual are visually estimated as defined on the field form (Figure 5.2-4). Observations are made from the wetted channel margin up 5 m (a canoe's length) into the bankfull channel margin on the previously chosen side of the stream.

You will measure or estimate the wetted width, mid-channel bar width, bankfull height and width, the amount of incision, and the degree of channel constraint. These are assessed for **the whole channel (left and right banks)** at each of the 11 cross-section transects. Record each on the Channel/Riparian Transect Form (Figure 5.2-4). The procedures for obtaining bank angle and measurements of channel cross-section morphology are presented in Table 5.2-8.

Wetted width is the width of the channel containing free-standing water; if >15 m, it can be measured with a laser rangefinder. **Mid-channel bar width**, the width of exposed mid-channel gravel or sand bars, is included within the wetted width, but is also recorded separately. In channel cross-section measurements, the wetted and bankfull channel boundaries include mid-channel bars. Therefore, the wetted width is measured as the distance between wetted left and right banks. Measure across and over mid-channel bars and boulders. If islands are present, treat them like bars, but flag these measurements and indicate in the comments that the "bar" is an island. If you are unable to see across the full width of the river when an island separates a side channel from the main channel, record the width of the main channel, flag the observation, and note in the comments section that the width pertains only to the main channel.

Table 5.2-8. Procedure for bank angle and channel cross-section

1. Visually estimate the bank angle (undercut, vertical, steep, gradual), as defined on the field form. Bank angle observations refer to the area from the wetted channel margin up 5 m (canoe's length) into the bankfull channel margin on the previously chosen side of the river. Circle the angle in the "BANK ANGLES" section of the Channel/Riparian Transect Form.
2. Hold the surveyor's rod vertically, with its base planted at the water's edge. Examine both banks, then determine the channel *incision* as the *height up from the water surface to elevation of the **first terrace** of the valley floodplain* (Note this is at or above the bankfull channel height). Whenever possible, use the clinometer as a level (positioned so it reads 0% slope) to measure this height by transferring (backsighting) it onto the surveyor's rod. Record this value in the *INCISED HEIGHT* field of the bank characteristics section on the field data form.
3. While still holding the surveyor's rod as a guide, and sighting with the clinometer as a level, examine both banks to measure and record the *height of bankfull flow above the present water level*. Look for evidence on one or both banks such as:
   - An obvious slope break that differentiates the channel from a relatively flat floodplain terrace higher than the channel.
   - A transition from exposed stream sediments to terrestrial vegetation.
   - Moss growth on rocks along the banks.
   - Presence of drift material caught on overhanging vegetation.
   - A transition from flood- and scour-tolerant vegetation to that which is relatively intolerant of these conditions.
4. Record the *wetted width* value determined when locating substrate sampling points in the *BANK CHARACTERISTICS* section of the field data form. Also determine the *bankfull channel width* and the *width of exposed mid-channel bars* (if present).
5. Repeat Steps 1 through 6 at each cross-section transect, (including any additional side channel transects established when islands are present). Record data for each transect on a separate field data form.

Bankfull flows are large enough to erode the stream bottom and banks, but frequent enough (every 1 to 2 years) to not allow substantial growth of upland terrestrial vegetation. Consequently, in many regions, it is these flows that have determined the width and depth of the channel. Estimates of the bankfull dimensions of stream channels are extremely important in EMAP surveys. They are used to calculate shear stress and bed stability (see Kaufmann et al., 1999). Unfortunately, we have to depend upon evidence visible during the low-flow sampling season. If available, consult published rating curves relating expected bankfull channel dimensions to stream drainage area within the region of interest. Graphs of these rating curves can help you get a rough idea of where to look for field evidence to determine the level of bankfull flows. Curves such as these are available from the USGS for streams in most regions of the U.S. (e.g., Dunne and Leopold 1978; Harrelson et al. 1994, Leopold 1994). To use them, you need to know the contributing drainage area to your sample site. Interpret the expected bankfull levels from these curves as a height above the streambed in a riffle, but remember that your field measurement will be a height above the present water surface of the stream. Useful resources to aid your determination of bankfull flow levels in streams in the United States are video presentations produced by the USDA Forest Service for western streams (USDA Forest Service 1995) and eastern streams (USDA Forest Service 2002).

After consulting rating curves that show where to expect bankfull levels in a given size of stream, estimate the bankfull flow level by looking at the following indicators:

- First look at the stream and its valley to determine the active floodplain. This is a depositional surface that frequently is flooded and experiences sediment deposition under the current climate and hydrological regime.
- Then look specifically for:
- An obvious break in the slope of the banks.
- A change from water-loving and scour-tolerant vegetation to more drought-tolerant vegetation.
- A change from well-sorted stream sediments to unsorted soil materials.

In the absence of clear bankfull indications, consider the previous season's flooding as the best evidence available (note: you could be wrong if very large floods or prolonged droughts have occurred in recent years.). Look for:

- Drift debris (“sticky wickets” left by the previous seasons flooding).
- The level where deciduous leaf-fall is absent on the ground (carried away by previous winter flooding).
- Unvegetated sand, gravel or mud deposits from previous year's flooding.

In years that have experienced large floods, drift material and other recent high flow markers may be much higher than other bankfull indicators. In such cases, base your determination on less-transient indicators such as channel form, perennial vegetation, and depositional features. In these cases, flag your data entry and also record the height of drift material in the comments section of the field data form.

We use the vertical distance (height) from the observed water surface up to the level of the first major valley depositional surface (Figure 5.2-6) as a measure of the degree of *incision* or *downcutting* of the stream below the general level of its valley. This value is recorded in the

incised height field. It may not be evident at the time of sampling whether the channel is downcutting, stable, or aggrading (raising its bed by depositing sediment). However, by recording incision heights measured in this way and monitoring them over time, we will be able to tell if streams are incising or aggrading.

If the channel is not greatly incised, bankfull channel height and incision height will be the same. However, if the channel is incised greatly, the bankfull level will be below the level of the first terrace of the valley floodplain, making "Bankfull Height" smaller than "Incision" (Figure 5.2-6). **Bankfull height is never greater than incision height**. Look for evidence of recent flows (within about 1 year) to distinguish bankfull and incision heights, though recent flooding of extraordinary magnitude may be misleading. In cases where the channel is cutting a valley sideslope and has oversteepened and destabilized that slope, the bare "cutbank" against the steep hillside at the edge of the valley is not necessarily an indication of recent incision. In such a case, the opposite bank may be lower, with a more obvious terrace above bankfull height; choose that bank for your measurement of incised height. Examine both banks to determine incision height and bankfull height. Remember that incision height is measured as **vertical distance to the first terrace above bankfull; if terrace heights differ on left and right banks, choose the lower of the two terraces.** Even when quite constrained by their valley sideslopes, large rivers often have flood terraces above bankfull height. In some cases, though, your sample reach may be in a steep "V" shaped valley or gorge formed over eons, and the slopes of the channel banks simply extend uphill indefinitely, not reaching a terrace before reaching the top of a ridge. In such cases, record incision height values equal to bankfull values and make appropriate comments that no terrace is evident. Similarly, when the river is extremely incised below an ancient terrace or plateau,(e.g., the Colorado River in the Grand Canyon), you may crudely estimate the terrace height if it is the first one above bankfull level. If you cannot estimate the terrace height, make appropriate comments describing the situation.

Finally, assess the **local degree of river channel constraint** (i.e., at the transect) by following the guidelines on the form (Figure 5.2-5) regarding the relationships among channel incision, valley sideslope, and width of the valley floodplain. You will also do an overall assessment of channel constraint for the whole river reach; see Section 5.2.9 for a discussion of constraint concepts and assessment procedures.

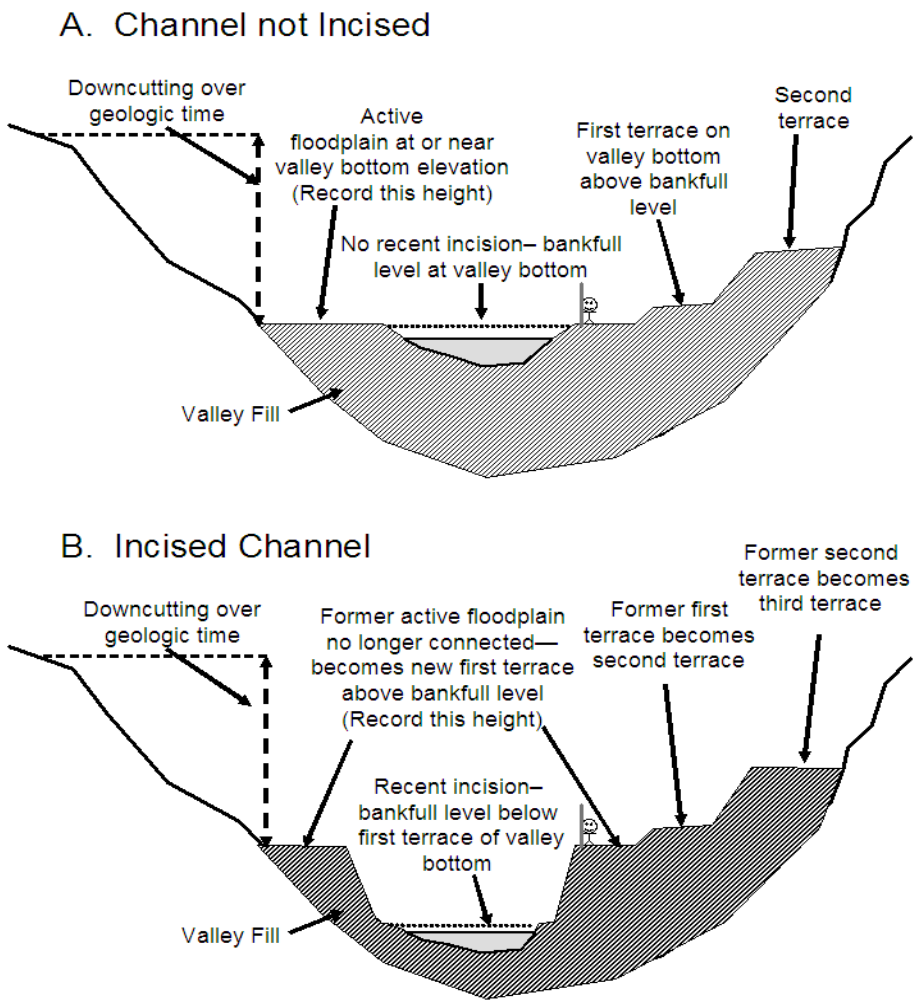


Figure 5.2-6. Schematic showing bankfull channel and incision for channels. (A) not recently incised, and (B) recently incised into valley bottom. Note level of bankfull stage relative to elevation of first terrace on valley bottom (stick figure included for scale)

5.2.8.4 Canopy Cover (Densiometer)

Measure vegetative cover over the reach at the chosen bank at each of the 11 transects (A-K). with a Convex Spherical Densiometer. Tape the densitometer exactly as shown in Figure 5.2-7 to limit the number of grid intersections to 17. Densiometer readings can range from 0 (no canopy cover) to 17 (maximum canopy cover). Four measurements are obtained at each cross-section transect (upriver, downriver, left, and right). The procedure for obtaining canopy cover data is presented in Table 5.2-8. Record the counts in the "Canopy Density @ Bank" section of the Channel/Riparian Transect Form as shown in Figure 5.2-4.

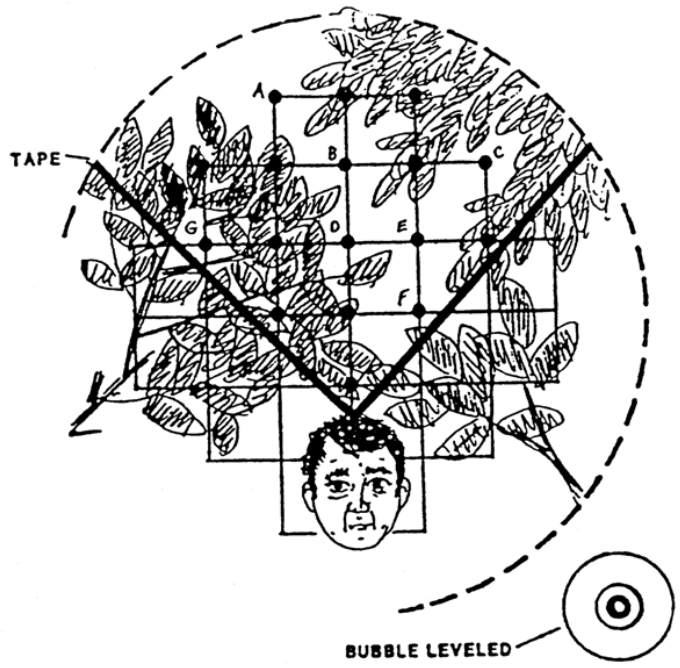


Figure 5.2-7. Schematic of modified convex spherical canopy densiometer (From Mulvey et al., 1992). In this example, 10 of the 17 intersections show canopy cover, giving a densiometer reading of 10. Note proper positioning with the bubble leveled and face reflected at the apex of the "V."

Table 5.2-9. Procedure for canopy cover measurements

1. Take densiometer readings at a cross-section transect while anchored or tied up at the river margin.
2. Hold the densiometer 0.3 m (1 ft) above the surface of the river. Holding the densiometer level using the bubble level, move it in front of you so your face is just below the apex of the taped "V".
3. At the channel margin measurement locations, count the number of grid intersection points within the "V" that are covered by either a tree, a leaf, a high branch, or the bank itself.
4. Take 1 reading each facing upstream (UP), downstream (DOWN), left bank (LEFT), and right bank (RIGHT). Right and left banks are defined with reference to an observer facing downstream.
5. Record the UP, DOWN, LEFT, and RIGHT values (0 to 17) in the "CANOPY COVER @ BANK" section of the Channel/Riparian Transect Form.
6. Repeat Steps 1 through 5 at each cross-section transect. Record data for each transect on a separate field data form.

5.2.8.5 Riparian Vegetation Structure

Riparian vegetation observations apply to the riparian area upstream 10 m and downstream 10 m from each of the 11 transects. They include the visible area from the river bankfull margin back a distance of 10 m (30 ft) shoreward from both the left and right banks, creating a 10m X 20m riparian plot on each side of the river (Figure 5.2-2). The riparian plot dimensions are estimated, not measured. Table 5.2-9 presents the procedure for characterizing

riparian vegetation structure and composition. Figure 5.2-5 illustrates how measurement data are recorded in the "Visual Riparian Estimates" section of the Channel/Riparian Transect Form, side 2.

Table 5.2-10. Procedure for characterizing riparian vegetation structure

1. Anchor or tie up at the river margin at a cross-section transect; then make the following observations to characterize riparian vegetation structure.
2. Estimate the distance from the shore to the edge of the riparian vegetation plot; record it just below the title "Channel Constraint" on the Channel/Riparian Transect Form, side 2.
3. Facing the left bank (left as you face downstream), estimate a distance of 10 m back into the riparian vegetation, beginning at the bankfull channel margin. Estimate the cover and structure of riparian vegetation within an estimated 10 m x 20 m plot centered on the transect, and starting where perennial vegetation begins or at the bankfull river margin (whichever is closest to the river shoreline). On steeply-sloping channel margins, estimate the riparian plot dimensions as if they were projected down from an aerial view.
4. Within this 10 m × 20 m area, conceptually divide the riparian vegetation into 3 layers: a CANOPY (>5m high), an UNDERSTORY (0.5 to 5 m high), and a GROUND COVER layer (<0.5 m high).
5. Within this 10 m × 20 m area, determine the dominant **woody** vegetation type for the CANOPY LAYER (vegetation > 5 m high) as either Deciduous, Coniferous, broadleaf Evergreen, Mixed, or None. Consider the layer "Mixed" if more than 10% of the areal coverage is made up of the alternate vegetation type. If the dominant vegetation type in the canopy layer is not woody, record the vegetation type as "None". Indicate the appropriate vegetation type in the "VISUAL RIPARIAN ESTIMATES" section of the Channel/Riparian Cross-section and Thalweg Profile Form.
6. Determine separately the areal cover class of large trees (> 0.3 m [1 ft] diameter at breast height [DBH]) and small trees (< 0.3 m DBH) within the canopy layer. Estimate areal cover as the amount of shadow that would be cast by a particular layer alone if the sun were directly overhead. Record the appropriate cover class on the field data form ("0" = absent, zero cover; "1" = sparse, <10%; "2" = moderate, 10-40%; "3" = heavy, 40-75%; or "4" = very heavy, >75%).
7. Look at the UNDERSTORY layer (vegetation between 0.5 and 5 m high). Determine the dominant **woody** vegetation type for the understory layer as described in Step 5 for the canopy layer. If the dominant vegetation type in the understory is not woody (e.g., herbaceous), record the vegetation type as "None".
8. Determine the areal cover class for woody shrubs and saplings separately from non-woody vegetation within the understory, as described in Step 6 for the canopy layer.
9. Look at the GROUND COVER layer (vegetation < 0.5 m high). Determine the areal cover class for woody shrubs and seedlings, non-woody vegetation, and the amount of bare ground or duff (dead organic material) present as described in Step 6 for large canopy trees.
10. Repeat Steps 1-9 for all transects, using a separate field data form for each transect.

You will estimate the areal cover separately in each of the three vegetation layers. Note that the areal cover can be thought of as the amount of shadow cast by a particular layer alone when the sun is directly overhead. The maximum cover in each layer is 100%, so the sum of the areal covers for the combined three layers could add up to 300%. When rating vegetation cover types, mixtures of two or more subdominant classes might all be given sparse ("1") moderate ("2") or heavy ("3") rankings. One very heavy cover class with no clear subdominant class might be ranked "4" with all the remaining classes either moderate ("2"), sparse ("1") or absent ("0"). Two heavy classes with 40-75% cover can both be ranked "3".

5.2.8.6 Fish Cover, Algae, Aquatic Macrophytes

Over a defined length and distance from shore at the sampling locations, crews shall estimate by eye and by sounding the proportional cover of fish cover features and trophic level indicators including large woody debris, rootwads and snags, brush, live trees in the wetted channel, undercut banks, overhanging vegetation, rock ledges, aquatic macrophytes, filamentous algae, and artificial structures.

The procedure to estimate the types and amounts of fish cover is outlined in Table 5.2-10. Record data in the "Fish Cover/Other" section of the Channel/Riparian Transect Form as shown in Figure 5.2-5. Crews will estimate the areal cover of all of the fish cover and other listed features that are in the water and on the banks within the 10m x 20m plot (refer to Figure 5.2-2).

Table 5.2.11. Procedure for estimating fish cover

1. Stop at the designated shoreline at a cross-section transect and estimate a 10 m distance upstream and downstream (20 m total length), and a 10 m distance out from the banks to define a 20 m x 10 m littoral plot.
2. Examine the water and the banks within the 20 m x 10 m littoral plot for the following features and types of fish cover: filamentous algae, aquatic macrophytes, large woody debris, in-channel live trees or roots, brush and small woody debris, overhanging vegetation, undercut banks, boulders, and artificial structures.
3. For each cover type, estimate its areal cover by eye and/or by sounding with a pole. Record the appropriate cover class in the "FISH COVER/OTHER" section of the Channel/Riparian Transect Form ("0"=absent: zero cover, "1"=sparse: <10%, "2"=moderate: 10-40%, "3"=heavy: 40-75%, or "4"=very heavy: >75%).
4. Repeat Steps 1 through 3 at each cross-section transect, recording data from each transect on a separate field data form.

Filamentous algae pertains to long streaming algae that often occur in slow moving waters. Aquatic macrophytes are water loving plants in the river, including mosses, that could provide cover for fish or macroinvertebrates. If the river channel contains live wetland grasses, include these as macrophytes. Woody debris are the larger pieces of wood that can provide cover and influence stream morphology (i.e., those pieces that would be included in the large woody debris tally [Section 5.2.8.2]). Brush/woody debris pertains to the smaller wood that primarily affects cover but not morphology. The entry for trees or brush within one meter of the surface is the amount of brush, twigs, small debris etc. that is not in the water but is close to the stream and provides cover. "Live Trees or Roots" are living trees that are within the channel -- estimate the areal cover provided by the parts of these trees or roots that are inundated. For ephemeral channels, estimate the proportional cover of these trees that is inundated during bankfull flows. Boulders are typically basketball to car sized particles. Many streams contain artificial structures designed for fish habitat enhancement. Streams may also have in-channel structures discarded (e.g., cars or tires) or purposefully placed for diversion, impoundment, channel stabilization, or other purposes. Record the cover of these structures on the form.

5.2.8.7 Human Influences

For the left and right banks at each of the 11 detailed Channel/Riparian Cross-Sections, evaluate the presence/absence and the proximity of 11 categories of human influences outlined

in Table 5.2-11. Record human influences on the Channel/Riparian Transect Form (Figure 5.2-5). You may mark "P" more than once for the same human influence observed outside of more than one riparian observation plot (e.g. at both Transect D and E). **The rule is that you count human disturbance items as often as you see them, BUT NOT IF you have to site through a previously counted transect or its 10x20 meter riparian plot.**

Table 5.2-12. Procedure for estimating human influence

1. Stop at the designated shoreline at a cross-section transect, look toward the left bank (left when facing downstream), and estimate a 10m distance upstream and downstream (20 m total length). Also, estimate a distance of 10 m back into the riparian zone to define a riparian plot area.
2. Examine the channel, bank and riparian plot area adjacent to the defined river segment for the following human influences: (1) walls, dikes, revetments, riprap, & dams; (2) buildings; (3) cleared lot, pavement (e.g., paved, graveled, dirt parking lot, foundation); (4) roads or railroads, (5) inlet or outlet pipes; (6) landfills or trash (e.g., cans, bottles, trash heaps); (7) parks or maintained lawns; (8) row crops; (9) pastures, rangeland, or hay fields; (10) logging; and (11) mining (include gravel mining).
3. For each type of influence, determine if it is present and what its proximity is to the river and riparian plot area. Consider human disturbance items as present if you can see them from the cross-section transect. Do not include them if you have to site through another transect or its 10 m × 20 m riparian plot.
4. For each type of influence, record the proximity class in the "HUMAN INFLUENCE" part of the "VISUAL RIPARIAN ESTIMATES" section of the Channel/Riparian Transect Form. Proximity classes are:
   - ·B ("Bank") Present within the defined 20 m river segment and located in the stream or on the wetted or bankfull bank.
   - ·C ("Close") Present within the 10 × 20 m riparian plot area, but above the bankfull level.
   - ·P ("Present") Present, but observed outside the riparian plot area.
   - ·O ("Absent") Not present within or adjacent to the 20 m river segment or the riparian plot area at the transect
5. Repeat Steps 1 through 4 for the opposite bank.
6. Repeat Steps 1 through 5 for each cross-section transect, recording data for each transect on a separate field form.

5.2.8.8 Riparian "Legacy" Trees and Invasive Alien Species

At each littoral-riparian station (A-K), search for the largest tree visible. Confine your search to within 100m (or as far as you can see) from the wetted bank on either side of the river from each transect upstream and downstream. Classify this tree as broadleaf deciduous, coniferous, or broadleaf evergreen (classify western larch as coniferous). Identify, if possible, the species or the taxonomic group of this tree from the list provided in Table 5.2-12 (also on field form) and estimate its height, diameter at breast height (dbh) and distance from the wetted margin of the river. You may need to use binoculars to make these determinations. Enter this information on the left hand column of the field form for Riparian "Legacy" Trees and Invasive Alien Plants (Figure 5.2-8). If the largest tree is a dead "snag", enter "Snag" as the taxonomic group. Note that the tree you choose may not truly be a "Legacy" tree; we use this data to determine if there are Legacy Trees along the stream reach.

Search in the 10 m x 20 m riparian and littoral plots on both banks for the presence of any invasive alien species listed in the NRSA Invasive Species Guide provided to each field crew. Document the species observed on the Riparian "Legacy" Trees and Invasive Alien Plants

form (Figure 5.2-8), answering the question of whether each of the target species is present in the plot. If you have a camera, document the species with a photograph. If you observe no alien taxa within the riparian and littoral plots, but can confidently identify them outside of the plots, include your observations in the comments portion of the form. If the river is too wide to effectively observe the far bank at a transect, record what you observe for the plot on the near bank, record a "U" flag, and explain in the comments section of the form.

Table 5.2-13. Procedure for identifying riparian legacy trees and alien invasive species

Legacy Trees:

Beginning at Transect A, look upstream and downstream as far as you can see within the 100m of the wetted bank but look no further downstream than half of the distance to the next transect. Locate the legacy tree from within that area.

Classify this tree as broadleaf deciduous, coniferous, or broadleaf evergreen (classify western larch as coniferous). Identify, if possible, the species or the taxonomic group of this tree from the list below.

1. Acacia/Mesquite
2. Alder/Birch
3. Ash
4. Cedar/Cypress/Sequoia
5. Fir (including Douglas Fir, Hemlock)
6. Juniper
7. Maple/Boxelder
8. Oak
9. Pine
10. Poplar/Cottonwood
11. Snag (Dead Tree of Any Species)
12. Spruce
13. Sycamore
14. Willow
15. Unknown, other Broadleaf Evergreen
16. Unknown or Other Conifer
17. Unknown or Other Deciduous
18. Elm

NOTE: If the largest tree is a dead "snag", enter "Snag" as the taxonomic group.

Estimate the height of the potential legacy tree, its diameter at breast height (dbh) and its distance from the wetted margin of the stream. Enter this information on the left hand column of the Riparian "Legacy" Trees and Invasive Alien Plants field form.

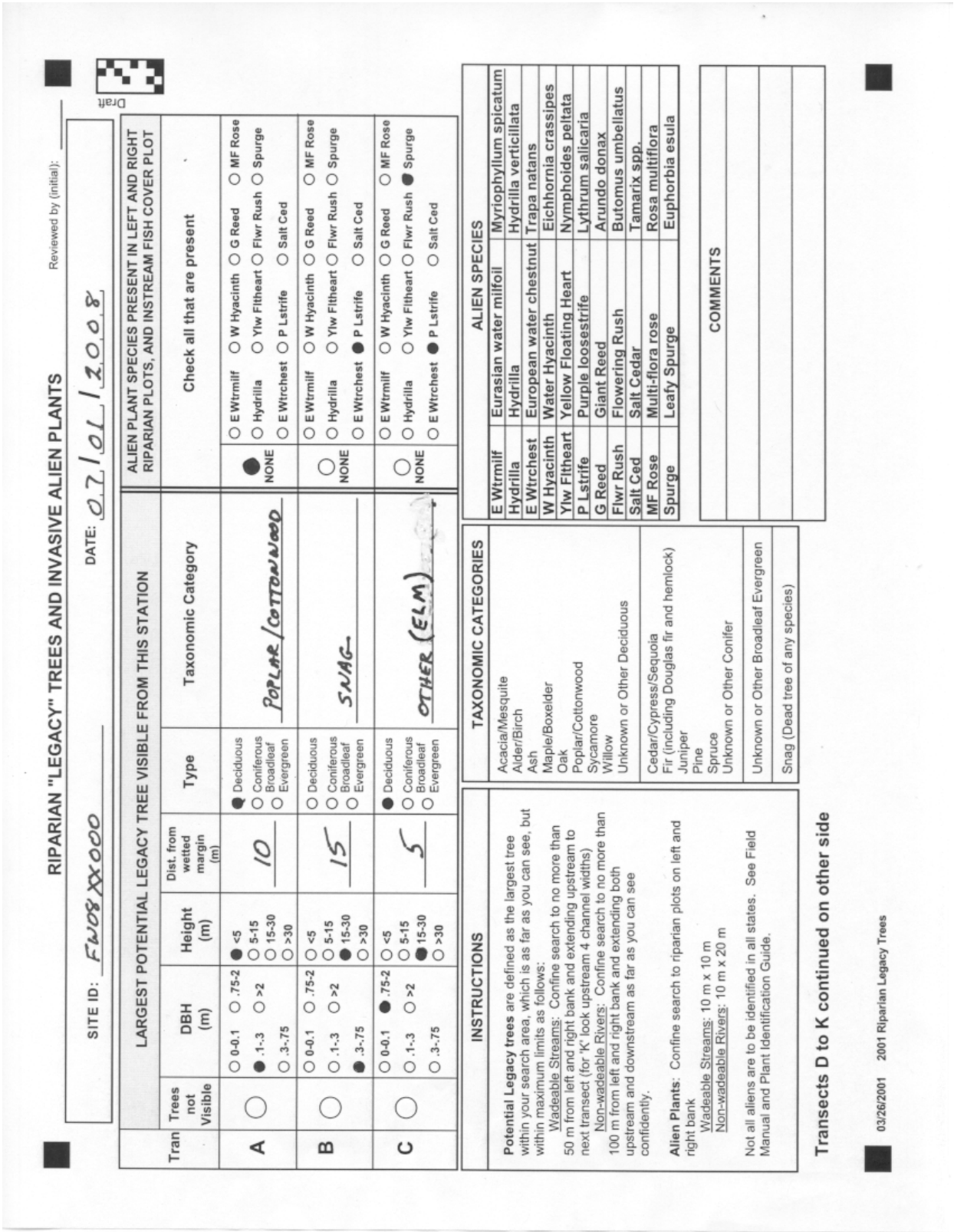
Alien Invasive Species:

Examine the 10m x 20m riparian and littoral plots on both banks for the presence of alien species. (Species lists will be provided)

Record the presence of any species listed within the plots on either the left or right bank on the Riparian "Legacy" Trees and Invasive Alien Species field form. If none of the species listed is present in the plots at a given transect, fill in the circle indicating "None" for this transect.

Repeat for each remaining transect (B through K). At transect "K", look upstream a distance of 4 channel widths) when locating the legacy tree.

Any invasive species seen but not included on this list should be written in the comments section.



RIPARIAN "LEGACY" TREES AND INVASIVE ALIEN PLANTS

Reviewed by (initial): ____

SITE ID: FW08XX000 DATE: 07/01/2008

Draft

Tran	Trees not Visible	LARGEST POTENTIAL LEGACY TREE VISIBLE FROM THIS STATION: DBH (m)	Height (m)	Dist. from wetted margin (m)	Type	Taxonomic Category	ALIEN PLANT SPECIES PRESENT IN LEFT AND RIGHT RIPARIAN PLOTS, AND INSTREAM FISH COVER PLOT: Check all that are present
A	○	○ 0-0.1 ○ .75-2 ● .1-.3 ○ >2 ○ .3-.75	● <5 ○ 5-15 ○ 15-30 ○ >30	10	● Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen	POPLAR/COTTONWOOD	● NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fitheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
B	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ● .3-.75	○ <5 ○ 5-15 ● 15-30 ○ >30	15	○ Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen	SNAG	○ NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fitheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ● P Lstrife ○ Salt Ced
C	○	○ 0-0.1 ● .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ● 15-30 ○ >30	5	● Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen	OTHER (ELM)	○ NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fitheart ○ Flwr Rush ● Spurge ○ E Wtrchest ● P Lstrife ○ Salt Ced

INSTRUCTIONS

Potential Legacy trees are defined as the largest tree within your search area, which is as far as you can see, but within maximum limits as follows:
Wadeable Streams: Confine search to no more than 50 m from left and right bank and extending upstream to next transect (for 'K' look upstream 4 channel widths)
Non-wadeable Rivers: Confine search to no more than 100 m from left and right bank and extending both upstream and downstream as far as you can see confidently.

Alien Plants: Confine search to riparian plots on left and right bank
Wadeable Streams: 10 m x 10 m
Non-wadeable Rivers: 10 m x 20 m

Not all aliens are to be identified in all states. See Field Manual and Plant Identification Guide.

TAXONOMIC CATEGORIES

- Acacia/Mesquite
- Alder/Birch
- Ash
- Maple/Boxelder
- Oak
- Poplar/Cottonwood
- Sycamore
- Willow
- Unknown or Other Deciduous
- Cedar/Cypress/Sequoia
- Fir (including Douglas fir and hemlock)
- Juniper
- Pine
- Spruce
- Unknown or Other Conifer
- Unknown or Other Broadleaf Evergreen
- Snag (Dead tree of any species)

ALIEN SPECIES

E Wtrmilf	Eurasian water milfoil	Myriophyllum spicatum
Hydrilla	Hydrilla	Hydrilla verticillata
E Wtrchest	European water chestnut	Trapa natans
W Hyacinth	Water Hyacinth	Eichhornia crassipes
Ylw Fitheart	Yellow Floating Heart	Nymphoides peltata
P Lstrife	Purple loosestrife	Lythrum salicaria
G Reed	Giant Reed	Arundo donax
Flwr Rush	Flowering Rush	Butomus umbellatus
Salt Ced	Salt Cedar	Tamarix spp.
MF Rose	Multi-flora rose	Rosa multiflora
Spurge	Leafy Spurge	Euphorbia esula

COMMENTS

Transects D to K continued on other side

03/26/2001 2001 Riparian Legacy Trees

Figure 5.2-8. Field form for Riparian "Legacy" Trees and Invasive Alien Plants (Page 1)

5.2.9 Channel Constraint Assessment

After completing the thalweg profile and littoral-riparian measurements and observations, visualize the stream at bankfull flow and evaluate the degree, extent and type of channel constraint, following the procedure in Table 5.2-12. Figure 5.2-9 illustrates anastomosing and braided channel types. Use the definitions on the Channel Constraint Assessment form (Figure 5.2-10) to classify the channel. Estimate the percent of the channel margin in contact with constraining features (for unconstrained channels, this is 0%). To aid in this estimate, you may wish to refer to the individual transect assessments of incision and constraint. Finally, estimate the "typical" bankfull channel width and visually estimate the average width of the valley floor. (valley floor width can often be determined from 1:24,000-scale topographic maps).

Table 5.2-14. Procedures for assessing channel constraint

NOTE: These activities are conducted after completing the thalweg profile and littoral-riparian measurements and observations, and represent an evaluation of the entire stream reach. Record this information on the Channel Constraint Form.

***CHANNEL CONSTRAINT*:** Determine the degree, extent, and type of channel constraint based on envisioning the stream at **bankfull flow**.

Classify the stream reach channel pattern as predominantly **one** channel, an **anastomosing** channel, or a **braided** channel.

> ***One channel*** may have occasional in-channel bars or islands with side channels, but feature a predominant single channel, or a dominant main channel with a subordinate side channel.
>
> ***Anastomosing channels*** have relatively long major and minor channels branching and rejoining in a complex network separated by vegetated islands, with no obvious dominant channel.
>
> ***Braided channels*** also have multiple branching and rejoining channels, separated by unvegetated bars. Subchannels are generally small, short, and numerous, often with no obvious dominant channel.

After classifying the channel pattern, determine whether the channel is constrained within a narrow valley, constrained by local features within a broad valley, unconstrained and free to move about within a broad floodplain, or free to move about, but within a relatively narrow valley floor.

Then examine the channel to ascertain the bank and valley features that constrain the stream. Entry choices for the type of constraining features are bedrock, hillslopes, terraces/alluvial fans, and human land use (e.g., a road, a dike, landfill, rip-rap, etc.).

Estimate the percent of the channel margin in contact with constraining features (for unconstrained channels, this is 0%).

Finally, estimate the "typical" bankfull channel width. To aid in this estimate, you may wish to refer to the individual transect assessments of incision and constraint that were recorded on the Channel/Riparian Cross-Section Forms.

Visually estimate the average width of the valley floor. If the valley is wider than you can directly estimate, record the distance you can see and mark the box on the field form.

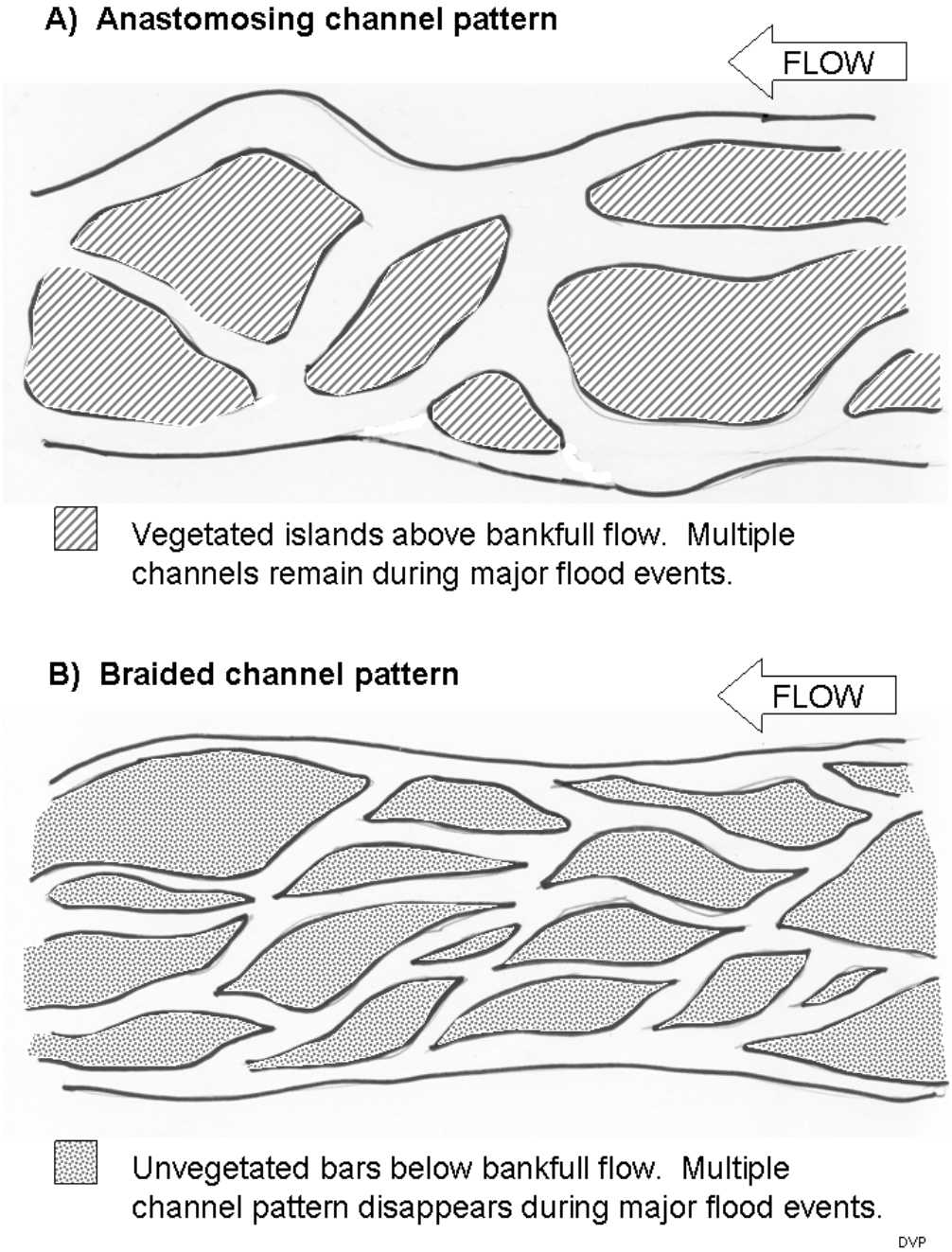
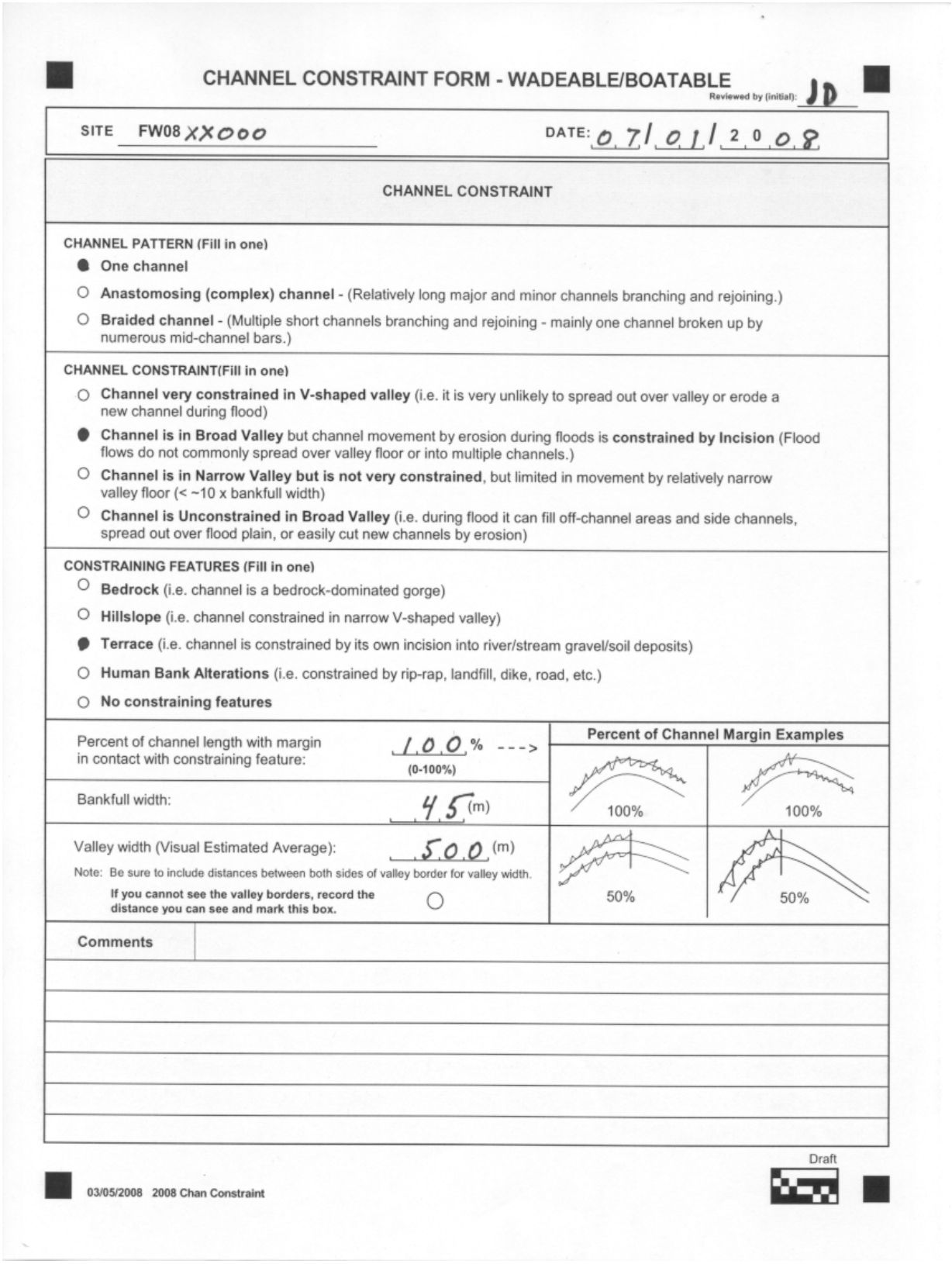


Figure 5.2-9. Types of multiple channel patterns.



CHANNEL CONSTRAINT FORM - WADEABLE/BOATABLE

Reviewed by (initial): JD

SITE FW08XX000 DATE: 07/01/2008

CHANNEL CONSTRAINT

CHANNEL PATTERN (Fill in one)

- ● One channel
- ○ **Anastomosing (complex) channel** - (Relatively long major and minor channels branching and rejoining.)
- ○ **Braided channel** - (Multiple short channels branching and rejoining - mainly one channel broken up by numerous mid-channel bars.)

CHANNEL CONSTRAINT(Fill in one)

- ○ **Channel very constrained in V-shaped valley** (i.e. it is very unlikely to spread out over valley or erode a new channel during flood)
- ● **Channel is in Broad Valley** but channel movement by erosion during floods is **constrained by Incision** (Flood flows do not commonly spread over valley floor or into multiple channels.)
- ○ **Channel is in Narrow Valley but is not very constrained**, but limited in movement by relatively narrow valley floor (< ~10 x bankfull width)
- ○ **Channel is Unconstrained in Broad Valley** (i.e. during flood it can fill off-channel areas and side channels, spread out over flood plain, or easily cut new channels by erosion)

CONSTRAINING FEATURES (Fill in one)

- ○ **Bedrock** (i.e. channel is a bedrock-dominated gorge)
- ○ **Hillslope** (i.e. channel constrained in narrow V-shaped valley)
- ● **Terrace** (i.e. channel is constrained by its own incision into river/stream gravel/soil deposits)
- ○ **Human Bank Alterations** (i.e. constrained by rip-rap, landfill, dike, road, etc.)
- ○ **No constraining features**

Percent of channel length with margin in contact with constraining feature: 100 % ---> (0-100%)

Bankfull width: 4.5 (m)

Valley width (Visual Estimated Average): 500 (m)

Note: Be sure to include distances between both sides of valley border for valley width.

If you cannot see the valley borders, record the distance you can see and mark this box. ○

Percent of Channel Margin Examples

Comments

Draft

03/05/2008 2008 Chan Constraint

Figure 5.2-10. Channel Constraint Form.

5.2.10 Debris Torrents and Recent Major Floods

Debris torrents, or *lahars*, differ from conventional floods in that they are flood waves of higher magnitude and shorter duration, and their flow consists of a dense mixture of water and debris. Their high flows of dense material exert tremendous scouring forces on streambeds. For example, in the Pacific Northwest, flood waves from debris torrents can exceed 5 meters deep in small streams normally 3 m wide and 15 cm deep. These torrents move boulders in excess of 1 m diameter and logs >1 m diameter and >10 m long. In temperate regions, debris torrents occur primarily in steep drainages and are relatively infrequent, occurring typically less than once in several centuries. They are usually set into motion by the sudden release of large volumes of water upon the breaching of a natural or human-constructed impoundment, a process often initiated by mass hillslope failures (landslides) during high intensity rainfall or snowmelt. Debris torrents course downstream until the slope of the stream channel can no longer keep their viscous sediment suspension in motion (typically <3% for small streams); at this point, they "set up", depositing large amounts of sediment, boulders, logs, and whatever else they were transporting. Upstream, the *torrent track* is severely scoured, often reduced in channel complexity and devoid of near-bank riparian vegetation. As with floods, the massive disruption of the stream channel and its biota are transient, and these intense, infrequent events will often lead to a high-quality complex habitat within years or decades, as long as natural delivery of large wood and sediment from riparian and upland areas remains intact.

In arid areas with high runoff potential, debris torrents can occur in conjunction with flash flooding from extremely high-intensity rainfall. They may be nearly annual events in some steep ephemeral channels where drainage area is sufficient to guarantee isolated thunderstorms somewhere within their boundaries, but small enough that the effect of such storms is not dampened out by the portion of the watershed not receiving rainfall during a given storm.

Because they may alter habitat and biota substantially, infrequent major floods and torrents can confuse the interpretation of measurements of stream biota and habitat in regional surveys and monitoring programs. Therefore, it is important to determine if a debris torrent or major flood has occurred within the recent past. After completing the thalweg profile and channel/riparian measurements and observations, examine the stream channel along the entire sample reach, including its substrate, banks, and riparian corridor, checking the presence of features described on the Torrent Evidence Assessment Form (Figure 5.2-11). It may be advantageous to look at the channel upstream and downstream of the actual sample reach to look for areas of torrent scour and massive deposition to answer some of the questions on the field form. For example, you may more clearly recognize the sample reach as a torrent deposition area if you find extensive channel scouring upstream. Conversely, you may more clearly recognize the sample reach as a torrent scour reach if you see massive deposits of sediment, logs, and other debris downstream.

Reviewed by (Initials): JD

TORRENT EVIDENCE ASSESSMENT FORM

SITE ID: FW08 XX000 DATE: 07/01/2008

TORRENT EVIDENCE	
Please fill in any of the following that are evident.	
EVIDENCE OF TORRENT SCOURING:	
○	01 - Stream channel has a recently devegetated corridor two or more times the width of the low flow channel. This corridor lacks riparian vegetation with possible exception of fireweed, even-aged alder or cottonwood seedlings, grasses, or other herbaceous plants.
○	02 - Stream substrate cobbles or large gravel particles are NOT IMBRICATED. (Imbricated means that they lie with flat sides horizontal and that they are stacked like roof shingles -- imagine the upstream direction as the top of the "roof.") In a torrent scour or deposition channel, the stones are laying in unorganized patterns, lying "every which way." In addition many of the substrate particles are angular (not "water-worn.")
○	03 - Channel has little evidence of pool-riffle structure. (For example, could you ride a mountain bike down the channel?)
○	04 - The stream channel is scoured down to bedrock for substantial portion of reach.
○	05 - There are gravel or cobble berms (little levees) above bankfull level.
○	06 - Downstream of the scoured reach (possibly several miles), there are massive deposits of sediment, logs, and other debris.
○	07 - Riparian trees have fresh bark scars at many points along the stream at seemingly unbelievable heights above the channel bed.
○	08 - Riparian trees have fallen into the channel as a result of scouring near their roots.
EVIDENCE OF TORRENT DEPOSITS:	
○	09 - There are massive deposits of sediment, logs, and other debris in the reach. They may contain wood and boulders that, in your judgement, could not have been moved by the stream at even extreme flood stage.
○	10 - If the stream has begun to erode newly laid deposits, it is evident that these deposits are "MATRIX SUPPORTED." This means that the large particles, like boulders and cobbles, are often not touching each other, but have silt, sand, and other fine particles between them (their weight is supported by these fine particles -- in contrast to a normal stream deposit, where fines, if present, normally "fill-in" the interstices between coarser particles.)
NO EVIDENCE:	
●	11 - No evidence of torrent scouring or torrent deposits.

COMMENTS

Draft

03/06/2008 NRSA Torrrent Evidence

Figure 5.2-11. Torrent Evidence Form.

5.3 Periphyton

5.3.1 Summary of Method

Collect periphyton from the near-shore shallows at each of the sampling stations located on the 11 cross-section transects ("A" through "K") established within the sampling reach. Collect periphyton samples at the same time as sediment enzyme samples (Section 5.1.4) and benthic macroinvertebrate samples (Section 5.4). Prepare one composite sample of periphyton for each site. At the completion of the day's sampling activities, but before leaving the site, prepare four types of laboratory samples (an ID/enumeration sample to determine taxonomic composition and relative abundances, a *chlorophyll* sample, a biomass sample (for ash-free dry mass [AFDM]), and an acid/alkaline phosphatase activity [APA] sample) from the composite periphyton sample.

5.3.2 Equipment and Supplies

Table 5.3-1 is a checklist of equipment and supplies required to conduct periphyton sample collection and processing activities. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the river.

Table 5.3-1. Equipment and supplies list for periphyton at non-wadeable sites

For collecting samples	▪ Large Funnel (15-20 cm diameter) ▪ 12-cm^2 area delimiter (3.8 cm diameter pipe, 3 cm tall) ▪ Stiff-bristle toothbrush with handle bent at 90˚ angle ▪ 1-L wash bottle for stream water ▪ 500-mL plastic bottle for the composite sample with marked volume gradations ▪ 60-mL plastic syringe with 3/8" hole bored into the end ▪ Aspirator ▪ Cooler with bags of ice ▪ Field Operations Manual or laminated Quick Reference Guide
For recording measurements	▪ Sample Collection Form ▪ Soft (#2) lead pencils for recording data on field forms ▪ Fine-tipped indelible markers for sample labels ▪ Sample labels (4 per set) with the sample ID number ▪ Clear tape strips for covering labels

5.3.3 Sampling Procedure

At each of the 11 transects, collect samples from the sampling station assigned during the layout of the reach. Collect the substrate selected for sampling from a depth no deeper than 0.5 m. If you cannot collect a sample because the location is too deep, skip the transect. The procedure for collecting samples and preparing a composite sample is presented in Table 5.3-2. Collect one sample from each of the transects and composite in one bottle to produce one composite sample for each site. Record the volume of the sample on the Sample Collection Form as shown in Figure 5.1-4.

Table 5.3-2. Procedure for collecting composite index samples of periphyton at non-wadeable sites

1. Starting with Transect "A", collect a single sample from the assigned sampling station using the procedure below.
   a) Collect a sample of hard substrate (rock or wood) that is small enough (< 15 cm diameter) and can be easily removed from the river. Place the substrate in a plastic funnel which drains into a 500-mL plastic bottle with volume graduations marked on it.
   b) Use the area delimiter to define a 12-cm^2 area on the upper surface of the substrate. Dislodge attached periphyton from the substrate within the delimiter into the funnel by brushing with a stiff-bristled toothbrush for 30 seconds. Take care to ensure that the upper surface of the substrate is the surface that is being scrubbed, and that the entire surface within the delimiter is scrubbed.
   c) Fill a wash bottle with river water. Wash the dislodged periphyton from the piece of substrate, brush, delimiter and funnel into the 500-mL bottle. Use an appropriate amount of water to bring the sample up to the next gradation. Doing so should result in collecting approximately 45mL of sample at each transect.
   d) If no coarse sediment (cobbles or larger) are present:
      - Use the area delimiter to confine a 12-cm^2 area of soft sediments.
      - Either:
        Vacuum the top 1 cm of sediment from within the delimited area into a de-tipped 60- mL syringe.

        Use an aspirator to suction the top 1 cm of sediment from within the delimited area into the sample bottle.
      - Empty the syringe into the same 500-mL plastic bottle as above.
   e) Put the bottle in a cooler on ice while you travel between transects and collect the subsequent samples. (The samples need to be kept cool and dark because a chlorophyll sample will be filtered from the composite.)
2. Repeat Step 1 for transects "B" through "K". Place the sample collected at each sampling site into the single 500-mL bottle to produce the composite index sample.
3. After samples have been collected from all 11 transects, thoroughly mix the 500-mL bottle regardless of substrate type.
4. Record the total volume of the composite sample in the periphyton section of the Sample Collection Form.
5. If you are unable to collect a sample at any location, mark it on the field form and record the volume of overall sample collected.

5.3.4 Sample Processing in the Field

You will prepare four different types of laboratory samples from the composite index samples: an **ID/enumeration sample** (to determine taxonomic composition and relative abundances), a **chlorophyll sample**, a **biomass sample** (for ash-free dry mass [AFDM]), and an **acid/alkaline phosphatase activity** (APA) sample. All the sample containers required for an individual site should be sealed in plastic bags until use to avoid external sources of contamination (e.g., dust, dirt, or mud) that are present at site shorelines. Please refer to Sections 7.2.5 and 7.2.6 processing the periphyton samples.

5.4 Benthic Macroinvertebrates

5.4.1 Summary of Method

Collect benthic macroinvertebrate composite samples using a D-frame net with 500 µm mesh openings. Take the samples from the sampling stations at the 11 transects equally distributed along the targeted reach. Composite all sample material and field-preserve with ~95% ethanol.

5.4.2 Equipment and Supplies

Table 5.4-1 shows the checklist of equipment and supplies required to complete the collection of benthic macroinvertebrates at non-wadeable sites. This checklist is similar to the checklist presented in Appendix A, which is used at the base location to ensure that all of the required equipment is brought to the site.

Table 5.4-1. Equipment and supplies list for benthic macroinvertebrate collection at non-wadeable sites

For collecting samples	▪ Modified kick net (D-frame with 500 µm mesh) and 4-5 ft handle ▪ Spare net(s) and/or spare bucket assembly for end of net ▪ Buckets, plastic, 8- to 10-qt ▪ Sieve bucket with 500 µm mesh openings (U.S. std No. 35) ▪ Watchmakers' forceps ▪ Wash bottle, 1-L capacity labeled "STREAM WATER" ▪ Funnel, with large bore spout	▪ Small spatula, spoon, or scoop to transfer sample ▪ Sample jars, 1-L HDPE plastic suitable for use with ethanol ▪ 95% ethanol, in a proper container ▪ Cooler (with absorbent material) for transporting ethanol & samples ▪ Plastic electrical tape ▪ Scissors ▪ Field Operations Manual or laminated Quick Reference Guide
For recording measurements	▪ Composite benthic sample labels with & without preprinted ID numbers ▪ Blank labels on waterproof paper for inside of jars	▪ Soft (#2) lead pencils ▪ Fine-tip indelible markers ▪ Clear tape strips ▪ Sample Collection Form

5.4.3 Sampling Procedure

Collect benthic macroinvertebrate samples at the 11 transects and within the sampling stations for non-wadeable streams. The process for selecting the sample stations is described in the Initial Site Procedures Section (Section 4). Collect all benthic samples at non-wadeable sites from the dominant habitat type within the 10 m x 15 m randomly selected sampling station at each transect (Figure 5.4-1). Take 1 linear meter sweep at the dominant habitat type. Record the benthic macroinvertebrate collection data on the Sample Collection Form, Side 1 as seen in Figure 5.1-2.

The sampling process for collecting benthic samples from non-wadeable sites is illustrated in Figure 5.4-2 and described in Table 5.4-2.

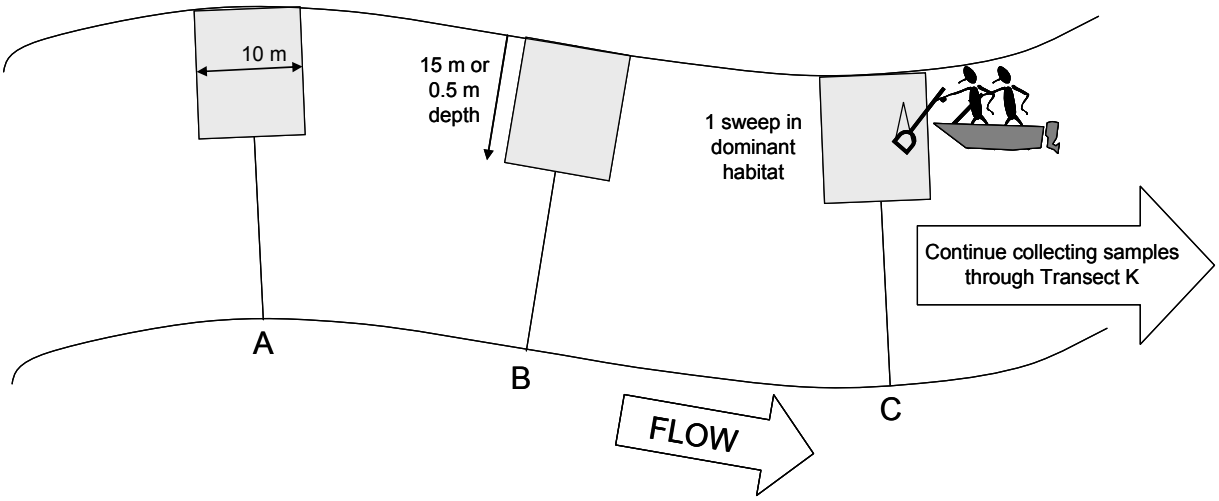


Figure 5.4-1. Transect sample design for collecting benthic macroinvertebrates at non-wadeable sites.

5.4.4 Sample Processing in Field

Use a 500 μm mesh sieve bucket placed inside a larger bucket full of site water while sampling to carry the composite sample as you travel around the site. It is recommended that teams carry a sample bottle containing a small amount of ethanol with them to enable them to immediately preserve larger predaceous invertebrates such as helgramites and water beetles. Doing so will help reduce the chance that other specimens will be consumed or damaged prior to the end of the field day. Once the sample from all stations is composited, sieved and reduced in volume, store in a 1-liter jar and preserve with 95% ethanol. Multiple jars may be required if detritus is heavy (Table 5.4-3). It is suggested that no more than 5 1-L jars be used at any site. If more than one jar is used for a composite sample, use the "extra jar" label provided; record the SAME sample ID number on this "extra jar" label. **DO NOT use two different sample numbers on two jars containing one single sample**. Remove any inorganic material (rocks, debris, etc) before preserving sample. Cover the labels with clear tape. The sample ID number is also recorded with a No. 2 lead pencil on a waterproof label that is placed inside each jar. Be sure the inside label and outside label describe the same sample. If there is a large amount of organic material in the sample, or there are adverse field conditions (i.e. hot, humid weather), place sample in a 1-L jar with ethanol after each station.

Record information for each composite sample on the Sample Collection Form as shown in Figure 5.1-2. If a sample requires more than one jar, make sure the correct number of jars for the sample is recorded on the Sample Collection Form. **Do not fill out the collection form until you have collected (or confirmed at the site that you will collect) samples.** If forms are filled out before you arrive at the site, and then no samples are collected, a lot of time is wasted by others later trying to find samples that do not exist. If you are unable to collect a sample at any station, make note of it on the sample collection form. Place the samples in a cooler or other secure container for transporting and/or shipping to the laboratory (see Appendix C).

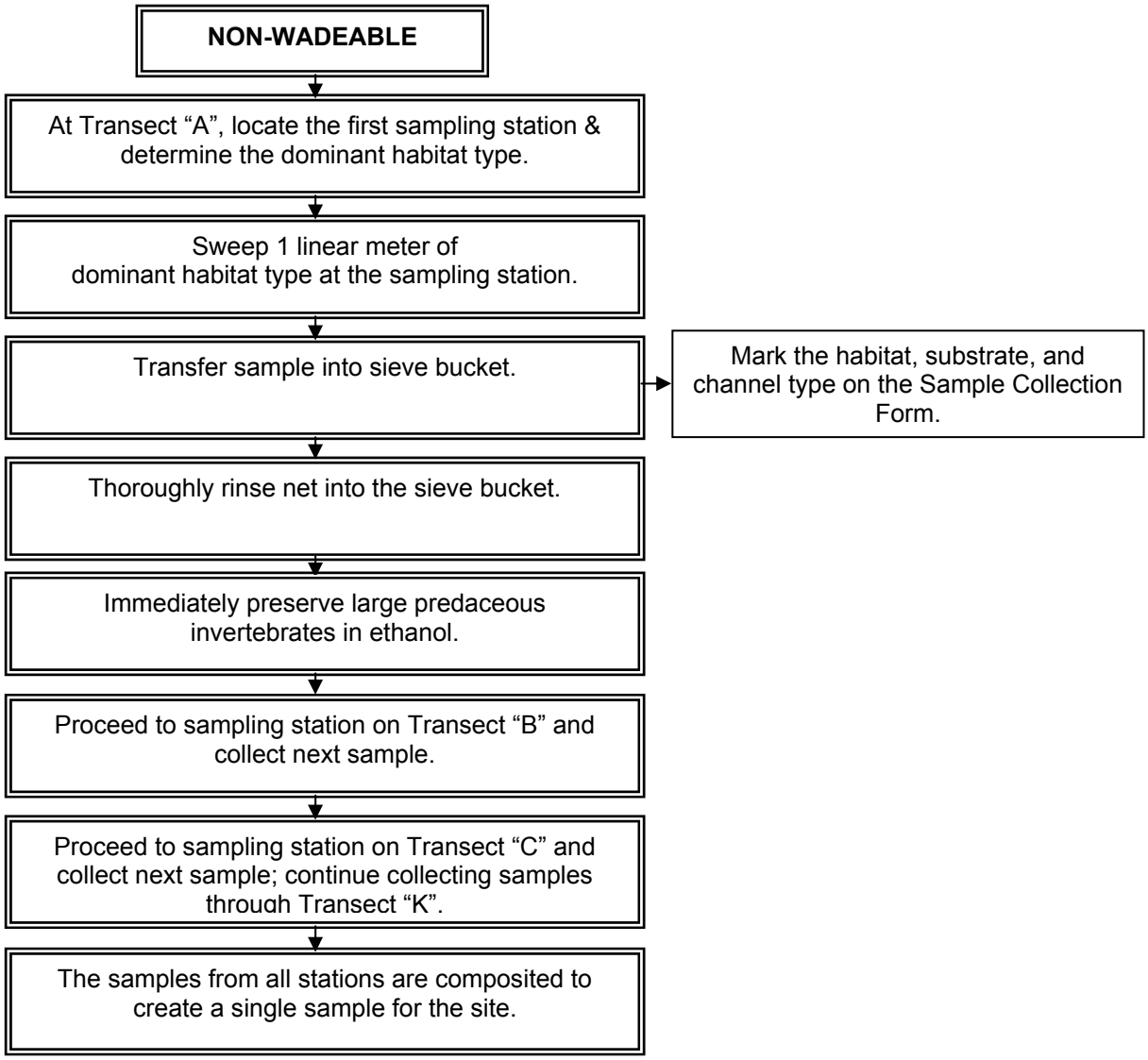


Figure 5.4-2. Benthic macroinvertebrate collection at non-wadeable sites.

Table 5.4-2. Procedure for benthic macroinvertebrate sampling at non-wadeable sites

1. After locating the sampling station site according to procedures described in the physical habitat section, identify the dominant habitat type within the plot:
   - Rocky/cobble/gravel/large woody debris
   - Organic fine mud or sand
   - Macrophyte beds
   - Leaf Pack
2. Use the D-frame dip net (equipped with 500 µm mesh) to sweep through 1 linear meter of the most dominant habitat type within the 10m x 15m sampling station, making sure to disturb the substrate enough to dislodge organisms.
   - If the dominant habitat is rocky/cobble/large woody debris it may be necessary to exit the boat and disturb the substrate (e.g., overturn rocks, logs) using your feet while sweeping the net through the disturbed area.
   - Because a dip-net is being used for sampling, the maximum depth for sampling will be approximately 0.5 m; therefore, in cases in which the depth of the river quickly drops off it may be necessary to sample in the nearest several meters to the shore.
3. After completing the 1 linear meter sweep, remove all organisms and debris from net and place them in a bucket following sample processing procedures described in the following section.
4. Record the sampled habitat type on the Sample Collection Form.
   a) **F**ine/sand: not gritty (silt/clay/muck <0.06 mm diam.) to gritty (up to ladybug sized 2 mm diam.)
   b) **G**ravel: fine to coarse gravel (ladybug to tennis ball sized; 2 mm to 64 mm diam.)
   c) **C**oarse: Cobble to boulder (tennis ball to car sized; 64 mm to 4000 mm)
   d) **O**ther: bedrock (larger than car sized; > 4000 mm), hardpan (firm, consolidated fine substrate), wood of any size, aquatic vegetation, etc.). Note "other" substrate in comments on field form.
5. Identify the channel habitat type where the sampling sweep was located. Mark the appropriate channel habitat type for the transect on the Sample collection Form.
   a) **P**ool; Still water; low velocity; smooth, glassy surface; usually deep compared to other parts of the channel
   b) **GL**ide: Water moving slowly, with smooth, unbroken surface; low turbulence
   c) **RI**ffle: Water moving, with small ripples, waves, and eddies; waves not breaking, and surface tension is not broken; "babbling" or "gurgling" sound.
   d) **RA**pid: Water movement is rapid and turbulent; surface with intermittent "white water" with breaking waves; continuous rushing sound.
6. Proceed to the next sampling station and repeat steps 1-5. The organisms and detritus collected at each station on the river should be combined in a single bucket to create a single composite sample for the river. After sampling at all 11 stations is completed, process the composite sample in the bucket according to procedures described in the following section.
7. If the sample contains primarily organic material, or if adverse weather conditions exist (i.e. hot humid weather) process the sample at each station by placing it in a 1-L nalgene jar with ethanol. Follow instructions in Table 5.4-3.
8. Immediately preserve larger predaceous invertebrates such as helgramites and water beetles in ethanol.

Table 5.4-3. Procedure for compositing samples for benthic macroinvertebrates at non-wadeable sites

Estimate the total volume of the sample in the sieve and determine how large a jar will be needed for the sample (500-mL or 1-L) and how many jars will be required. It is suggested that no more than 5 1-L jars are used at each site.

Fill in a sample label with the Sample ID and date of collection. Attach the completed label to the jar and cover it with a clear tape strip. Record the Sample ID for the composite sample on the Sample Collection Form. For each composite sample, make sure the number on the form matches the number on the label.

Wash the contents of the sieve to one side by gently agitating the sieve in the water. Wash the sample into a jar using as little water from the wash bottle as possible. Use a large-bore funnel if necessary. If the jar is too full pour off some water through the sieve until the jar is not more than 1/3 full, or use a second jar if a larger one is not available. Carefully examine the sieve for any remaining organisms and use watchmakers' forceps to place them into the sample jar. Remove any inorganic material, such as gravel, by rinsing the material, examining it and removing it from the sample.

- If a 2nd jar is needed, fill in a label that does not have a pre-printed ID # on it. Record the ID # from the pre-printed label prepared above in the "SAMPLE ID" field of the label. Attach the label to the 2nd jar and cover it with a strip of clear tape. Record the number of jars on the Sample Collection Form. **Make sure the number you record matches the actual number of jars used.** Write "Jar *N* of *X*" on each sample label using a waterproof marker. **Try to use no more than 5 jars per site.**

Place a waterproof label inside each jar with the following information written with a #2 lead pencil:

- Site ID
- Type of sampler and mesh size used
- Name of site
- Date of collection
- Collectors initials
- Number of stations sampled
- Jar "N" of "X"

Completely fill the jar with 95% ethanol (no headspace). It is very important that sufficient ethanol be used, or the organisms will not be properly preserved. Existing water in the jar should not dilute the concentration of ethanol below 70%.

- NOTE: Composite samples can be transported back to the vehicle before adding ethanol if necessary. In this case, fill the jar with stream water, then drain using the net (or sieve) across the opening to prevent loss of organisms, and replace with ethanol at the vehicle.

Replace the cap on each jar. Slowly tip the jar to a horizontal position, then gently rotate the jar to mix the preservative. Do not invert or shake the jar. After mixing, seal each jar with plastic tape.

Store labeled composite samples in a container with absorbent material that is suitable for use with 70% ethanol until transport or shipment to the laboratory.

5.5 Fish

5.5.1 Summary of Method

The fish sampling method is designed to provide a representative sample of the fish community, collecting all but the rarest fish inhabiting the site. It is assumed to accurately represent species richness, species guilds, relative abundance, size, and anomalies. The goal is to collect fish community data that will allow the calculation of an Index of Biotic Integrity (IBI) and Observed/Expected (O/E) models. Boat electrofishing is the preferred method of sampling. If electrofishing is not possible due to safety concerns, high turbidity, or extremes in conductivity, complete the "Not Fished" section of the field form and comment why.

The time and effort necessary to sample the reach in its entirety is prohibitive in the context of the survey, thus sub-sampling is required. Electrofishing will occur in a downstream direction at all habitats along alternating banks (see section 5.5.3), over a length of 20 times the mean channel width (Transects A through F). Collection of a minimum of 500 fish is required. If this target is not attained, sampling will continue until 500 individuals are captured or the downstream extent of the site (transect K) is reached. Identification and processing of fish should occur at the completion of each transect. If sampling cannot happen at any individual transect, record it on the field collection form.

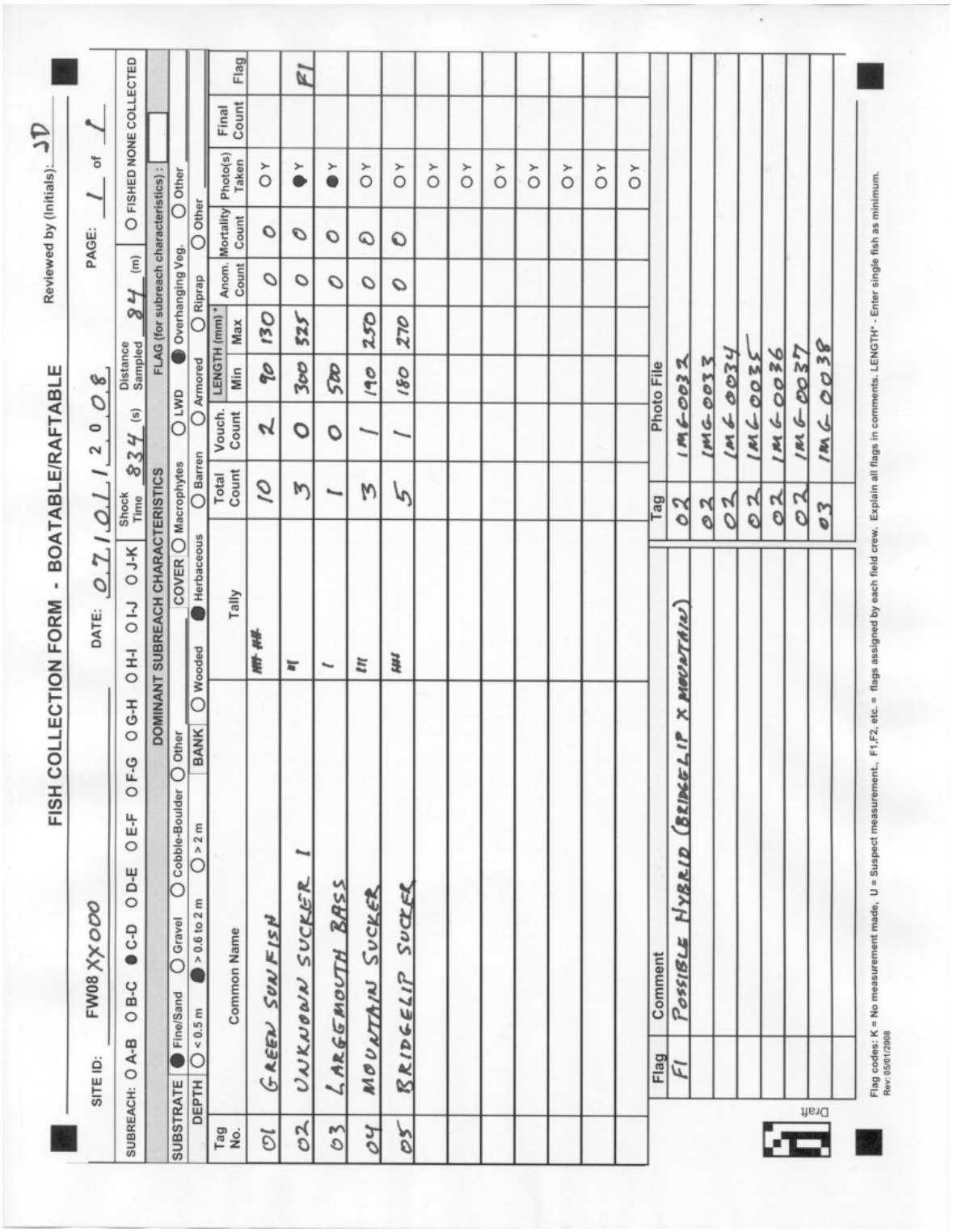
5.5.2 Equipment and Supplies

Table 5.5-1 shows the checklist of equipment and supplies required to complete the non-wadeable fish assessment. This checklist is similar to the one presented in Appendix A, which is used at the base location to ensure that all of the required equipment is brought to the site. Record fish collection data on the Fish Collection Form, Side 1 (Fig. 5.5-1). Additional sheets may be necessary – remember to indicate the transect on each form.

Table 5.5-1. Equipment and supplies — fish assessment at non-wadeable sites.

For collecting samples	• Boat, motor, and trailer (and necessary safety equipment)
	• Gasoline and oil (if using a 2 cycle)
	• Boat electrofishing equipment
	  ・ Pulsator Control Box
	  ・ Foot Pedal
	  ・ Anode Droppers
	  ・ Generator
	  ・ Linesman's Gloves
	  ・ Hearing Protection
	• Tow barge electrofishing equipment
	  ・ Probes with extensions.
	  ・ Appropriate switching box
	• Dip nets (non-conductive handles) ¼" mesh
	• Scientific collection permit
		• GPS with transect waypoints preloaded
		• Several Leak-proof HDPE jars for fish voucher specimens (various sizes from 250 mL – 4L)
		• 1scalpel for slitting open large fish before preservation
		• 1 container of 10% buffered formalin
		• 1 Minnow net for dipping small fish from live well
		• 2 measuring boards (3 cm size classes)
		• 1 set Fish ID keys
		• Field Operations Manual and/or laminated Quick Reference Guide
		• Digital camera with extra memory card & battery

For recording measurements	• Sample labels • Sample Collection Form • Clear tape strips	• Soft (#2) lead pencils • Fine-tip indelible markers



FISH COLLECTION FORM - BOATABLE/RAFTABLE

SITE ID: FW08XX000 DATE: 07/01/2008 Reviewed by (Initials): JD

SUBREACH: ○ A-B ○ B-C ● C-D ○ D-E ○ E-F ○ F-G ○ G-H ○ H-I ○ I-J ○ J-K Shock Time 834 (s) Distance Sampled 84 (m) ○ FISHED NONE COLLECTED PAGE: 1 of 1

DOMINANT SUBREACH CHARACTERISTICS FLAG (for subreach characteristics) :

SUBSTRATE ● Fine/Sand ○ Gravel ○ Cobble-Boulder ○ Other ____ COVER ○ Macrophytes ○ LWD ● Overhanging Veg. ○ Other ____

DEPTH ○ < 0.5 m ● > 0.6 to 2 m ○ > 2 m BANK ○ Wooded ● Herbaceous ○ Barren ○ Armored ○ Riprap ○ Other ____

Tag No.	Common Name	Tally	Total Count	Vouch. Count	LENGTH (mm) * Min	Max	Anom. Count	Mortality Count	Photo(s) Taken	Final Count	Flag
01	GREEN SUNFISH	||| |||	10	2	90	130	0	0	○ Y		
02	UNKNOWN SUCKER 1	|||	3	0	300	525	0	0	● Y		F1
03	LARGEMOUTH BASS			1	0	500		0	0	● Y		
04	MOUNTAIN SUCKER	|||	3	1	190	250	0	0	○ Y		
05	RIDGELIP SUCKER	||||	5	1	180	270	0	0	○ Y		

Flag	Comment	Tag	Photo File
F1	POSSIBLE HYBRID (RIDGELIP X MOUNTAIN)	02	IMG-0032
		02	IMG-0033
		02	IMG-0034
		02	IMG-0035
		02	IMG-0036
		02	IMG-0037
		03	IMG-0038

Draft

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = flags assigned by each field crew. Explain all flags in comments. LENGTH* - Enter single fish as minimum.
Rev: 05/01/2008

Figure 5.5-1. Fish Collection Form, Side 1.

5.5.3 Sampling Procedure

Sampling will begin at the upstream half of the overall site, representing 20 times the mean channel width. The total distance fished will depend upon the number of individuals captured. Shoreline electrofishing will begin at transect A and proceed in a downstream direction, alternating banks and terminating with the completion of subreach E-F (Figure 5.5-2). Determination of the initial stream bank sampling location at transect A (i.e., right or left bank) corresponds to the sequence established for physical habitat sampling and is determined at random. Subreaches A-B, B-C, and C-D are sampled along the same bank before alternating to the opposite bank to complete subreaches D-E and E-F. Each subreach is sampled for a maximum of 700 seconds per subreach. Identification and processing of the sample should be completed prior to beginning the next subreach. A minimum of 500 specimens is required. If fewer than 500 individuals are captured, sampling must continue on alternating banks (again following the pattern laid out for physical habitat sampling) until the minimum number is attained or the downstream extent of the site (transect K) is reached (Figure 5.5-2).

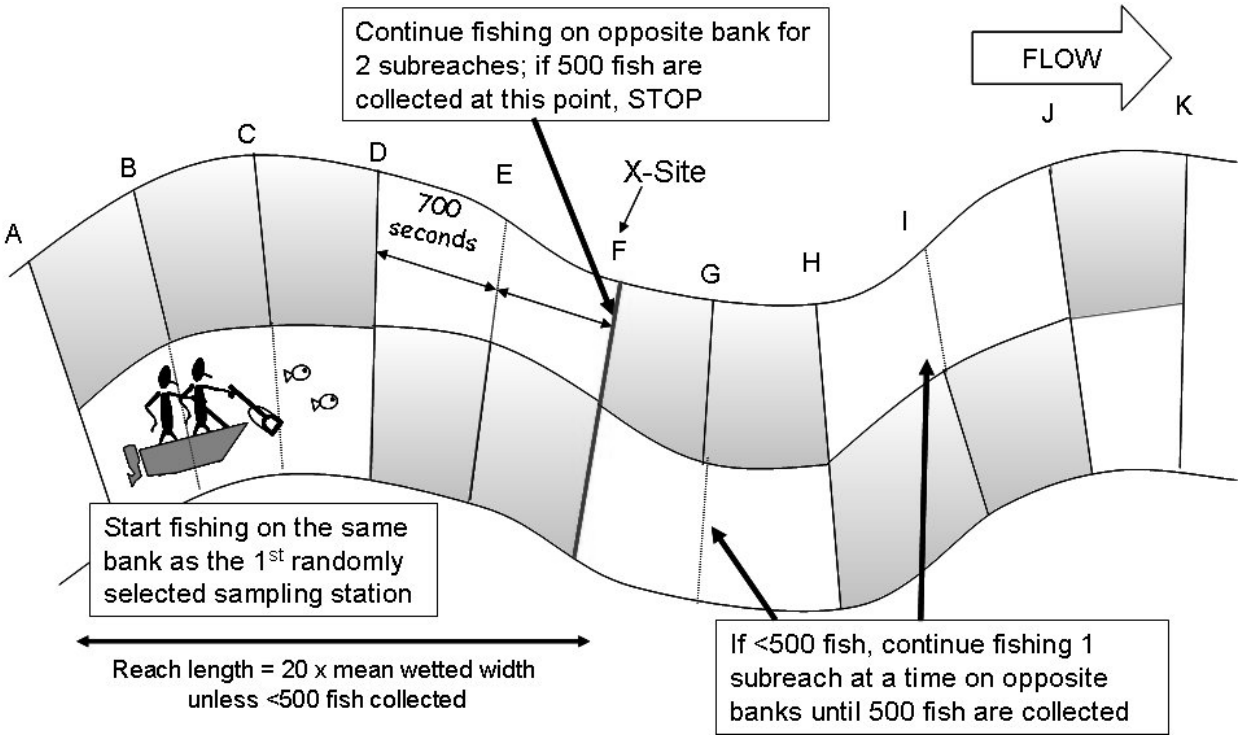


Figure 5.5-2. Transect sampling design for fish sampling at non-wadeable sites.

The sampling crew should consist of one boat operator (also controlling the electrofishing unit) and one dip-netter (1/4" mesh dip nets) situated at the bow. Prior to sampling each subreach, the crew should determine the most appropriate gear for the segment (e.g., boat or barge electrofishing units). Electrofishing should proceed downstream at a pace equal to or slightly greater than the prevailing current to maximize capture efficiency. It may be necessary to maneuver the electrofishing unit in and around complex habitat; however,

discretion should be used in sampling these areas in order to maintain equal effort between subreaches. Total effort expended (i.e., button time) over the five subreaches should be approximately 3500 seconds. If additional subreaches are sampled, additional time will be spent. To reduce stress and mortality, immobilized fish should be netted immediately and deposited into a live-well for processing. For safety, all crew members are required to wear personal floatation devices and insulated gloves. Polarized sunglasses and caps to aid vision are also required. Table 5.5-2 provides the procedure for electrofishing in non-wadeable streams.

Table 5.5-2. Procedure for electrofishing at non-wadeable sites.

1. Review all collecting permits to determine if any sampling restrictions are in effect for the site. In some cases, you may have to cease sampling if you encounter certain State- or Federally-listed species.
2. Boat electrofishing will be used in non-wadeable streams, and the direction of fishing will be downstream. If conductivity, turbidity, or safety precludes electrofishing, complete the "NOT FISHED" field on the Fish Collection Form and comment why.
3. The sampling reach is defined as 20 times the mean channel width, corresponding to transects A through F unless < 500 individuals are captured.
4. Shoreline electrofishing between each transect will occur on alternating banks following the sequence established in the physical habitat procedures. Sampling will begin on the bank selected at random and continue from transect A downstream for 700 seconds or until the next transect is reached. Subreaches B-C and C-D are fished similarly; subreaches D-E and E-F will then be sampled on the opposite bank. If fewer than 500 individuals are captured, sampling should continue until the minimum catch is attained or the last subreach (J-K) is fished. Follow the systematic rotation of banks such that up to two subreaches would be fished on the same bank prior to switching to the opposite bank. Crews must complete each of the additional subreaches as described above, do not stop in the middle of any subreach, even if the 500 fish minimum is attained before the end of the subreach.
5. Set unit to pulsed DC and test settings outside of the sampling area. Start the electrofisher, set the timer, and depress the switch to begin fishing. Typical settings are: 500-1000VDC; 8-20A; and 120 Hz. If fishing success is poor, increase the pulse width first and then the voltage. Increase the pulse rate last to minimize mortality or injury to large fish. If mortalities occur, first decrease pulse rate, then voltage, then pulse width.
6. Once the settings on the electrofisher are adjusted to sample effectively and minimize injury and mortality, begin sampling at the upstream reach (Transect A). Electrofishing proceeds downstream in close proximity to the bank and at a pace equal to or slightly greater than the prevailing current to maximize capture efficiency. Crews may "nose in" to habitat to effectively sample but should not remain in that habitat for too long. Generally effort (i.e., button time) should be 700 seconds per subreach. At sites with maximum reach length (4km) it is likely that the entire subreach (400m) will not be fished. Depending upon the habitat complexity, variable distances may be fished in the time allotted. Distance sampled is recorded on the Fish Collection Form.
7. Recommended mesh size on dip nets is 6mm (1/4"). Dip netters should actively capture stunned fish, removing them from the electric field and immediately placing them in the livewell. Special attention should be devoted to netting small and benthic fishes as well as fishes that may respond differently to the current.
8. Process fish at the completion of each subreach to reduce mortality and track sampling effort. Release fish in a location that eliminates the likelihood of recapture.
9. Complete header information on the Fish Collection Form. Record the number of seconds fished and the estimated distance fished (as tracked by GPS or measured by range finder).

10. Repeat Steps 6 through 8 until subreach E-F and 500 individuals are captured or at a maximum, subreach J-K is finished.

5.5.4 Processing Fish

Process fish when fish show signs of stress (e.g., loss of righting response, gaping, gulping air, excessive mucus). Change water or stop fishing and initiate processing as soon as possible. Similarly, State- and Federally-listed threatened or endangered species or large game fish should be processed and released as they are captured. If periodic processing is required, fish should be released in a location that prevents the likelihood of their recapture.

Use the Fish Collection Form – Large Wadeable/Boatable/Raftable. If several forms are needed, use an extra form and note the page number on the top of the form as well as the subreach sampled (i.e. Page 1 of 3). Taxonomic identification and processing should only be completed on specimens greater than 25 mm total length and by crew members designated as "fish taxonomic specialists" by EPA regional coordinators. Fish are tallied by species, evaluated for maximum and minimum length, and examined for the presence of DELT (Deformities, Eroded Fins, Lesions and Tumors) anomalies. Common names of species should follow those established under the American Fisheries Society's publication, "Common and Scientific Names of Fishes from the United States, Canada and Mexico" (Nelson, et al. 2004). A list of species common to freshwater systems of the United States is presented in Appendix D.

Species not positively identified in the field should be separately retained (up to 20 individuals per species) for laboratory identification. Common names for retained species should be assigned as "unknown", followed by its common family name and sequential lettering to designate separate species (e.g., UNKNOWN SCULPIN A). Following positive laboratory identification, field form information should be updated to reflect the actual species count and number in the Final Count field. For example, if a sample of 20 specimens of species A is later identified as 15 individuals of species A and 5 of species B, the Final Count of species A should be corrected by assigning 25% to species B and 75% to species A. Table 5.5-3 presents the procedure for processing fish.

Table 5.5-3. Procedure for processing fish at non-wadeable sites.

1.	Complete all header information accurately and completely. If no fish were collected, complete the "NONE COLLECTED" field on the Fish Collection Form.
2.	Complete the information on the Fish Gear and Voucher/Tissue Sample Information Form.
3.	Only identify and process individuals > 25mm in total length, ideally handling specimens only once. Record the common name on the first blank line in the "COMMON NAME" Field of the Fish Collection Form.
4.	Fill in the Tag Number. The tag number is a number starting with 01 and continuing sequentially to a number equal to the total number of species collected within the entire sample reach. Each reoccurrence of a species within the entire reach should be assigned the same tag number as it was assigned initially. For example, if a bluegill is assigned tag number 01 when processing fish from the first subreach, all bluegills from the other subreaches will also be assigned tag number 01. The purpose of the tag number is to connect species identifications with subsequent verification and voucher collections.
5.	If a species cannot be positively identified, assign it a sequential tag number in the Tag Number Field and leave the "COMMON NAME" Field Blank. Flag this line and indicate in the "COMMENT"

field its common family name (e.g., UNKNOWN SCULPIN A). Retain a maximum subsample of 20 individuals for in-house laboratory identification of Unknowns. Do not include the number of each species retained solely for in-house lab verification in the Voucher Count column of the fish collection form. This column is reserved only for those fish that are to be sent in for independent re-identification as part of a complete voucher collection.

6. Process species listed as threatened and endangered first and return individuals immediately to the stream. Photograph specimens for verification purposes if conditions permit and stress to individuals will be minimal. Indicate if photographed on Fish Collection Form. If individuals are killed, prepare them as verification specimens and preserve them in field.
7. Tally the number of individuals of each species collected in the "TALLY" box on the Fish Collection Form and record the total number in the "TOTAL COUNT" field on the form. Do not enter a total for fishes that must be identified in the laboratory.
8. Measure the total length of the largest and smallest individual to provide a size range for the species. Record these values in the "LENGTH" area of the Fish Collection Form. If only one fish is collected, leave the maximum field blank.
9. Examine each individual for external anomalies and tally those observed. Readily identify external anomalies including missing organs (eye, fin), skeletal deformities, shortened operculum, eroded fins, irregular fin rays or scales, tumors, lesions, ulcerous sores, blisters, cysts, blackening, white spots, bleeding or reddening, excessive mucus, and fungus. After all of the individuals of a species have been processed, record the total number of individuals affected in the "ANOMALIES" Field of the Fish Collection Form.
10. Record the total number of mortalities due to electrofishing or handling on the Fish Collection Form.
11. Follow the appropriate procedure to prepare voucher specimens and/or to select specimens for tissue samples. Release all remaining individuals so as to avoid their recapture.
12. For any line with a fish name, ensure that all spaces on that line are filled in with a number, even if it is zero.

5.5.5 Taxonomic Quality Assurance/Quality Control

5.5.5.1 Sample Preservation

Fish retained for laboratory identification or voucher purposes should be placed in a large sample jar containing a 10% buffered formalin solution in a volume equal to or greater than the total volume of specimens. Individuals larger than 200 mm in total length should be slit along the right side of the fish in the lower abdominal cavity to allow penetration of the solution.

Fish retained for laboratory identification or as vouchers should be preserved in the field following the precautions outlined in the MSDS. All personnel handling 10% buffered formalin must read the MSDS (Appendix D). **Formalin is a potential carcinogen and should be used with extreme caution, as vapors and solution are highly caustic and may cause severe irritation on contact with skin, eyes, or mucus membranes. Wear vinyl or nitrile gloves and safety glasses, and always work in a well-ventilated area.**

5.5.5.2 Laboratory Identification of Fish

Fish that are difficult to identify in the field should be kept for laboratory identification or to verify difficult field identifications. Table 5.5-4 outlines the laboratory identification process and completing the Fish Collection Form. Field crews may use a supplemental Fish Identification Lab sheet such as that shown in Figure 6.5-4 for internal laboratory use only.

Crews should retain the Fish verification sample – contact your regional EPA coordinator if you cannot store the samples at your facility.

Do not include the number of each species retained solely for in-house lab verification in the Voucher Count column of the fish collection form. This column is reserved only for those fish that are to be sent in for independent re-identification as part of a complete voucher collection.

Field crews should not retain the Fish Collection Form(s) if the laboratory identification process cannot be completed within a short period of time. If the time needed to complete the identification/verification is expected to exceed two weeks, make copies of the Fish Collection Form(s) and send the entire pack of original data forms to the Information Management Coordinator. When the identification/verification process is complete, make the necessary changes to the copied Fish Collection Form(s) and send them as soon as possible to the Information Management Coordinator as well.

Table 5.5-4. Procedure for laboratory identification of fish samples.

1. Fish may be retained for routine laboratory identification and verification purposes. Fish tags are provided with each site kit. Crews may use these tags at their discretion in order to identify fish at their laboratory.
2. Retained fish should be placed in a large sample jar containing a 10% buffered formalin solution in a volume equal to or greater than the total volume of specimens. Individuals larger than 200mm in total length should be slit along the right side of the fish in the lower abdominal cavity to allow penetration of the solution.
3. Following fixation for 5 to 7 days, the volume of formalin should be properly discarded and replaced with tap water for soaking specimens over a 4-5 day period. Soaking may require periodic water changes and should continue until the odor of formalin is barely detectable. Final storage of specimens is done in 45%-50% isopropyl alcohol or 70% ethanol. Formalin is a potential carcinogen and should be used with extreme caution, as vapors and solution are highly caustic and may cause severe irritation on contact with skin, eyes, or mucus membranes. Wear vinyl or nitrile gloves and safety glasses, and always work in a well-ventilated area.
4. Formalin must be disposed of properly. Contact your regional EPA coordinator if your laboratory does not have the capability of handling waste formalin.
5. Unknown fish are identified to species in the laboratory. You may use a Fish Identification Lab Sheet such as the one presented in Figure 6.5-4.
6. Fill in the Unknown species name in the “COMMON NAME” field of the Fish Collection Form and make certain the “FINAL COUNT” field is correct.
7. If species field identifications were incorrect, correct the “COMMON NAME” Field by completely erasing the Common Name and replacing the correct name. Add an additional Common Name if needed. Make certain the “FINAL COUNT” field is correct. If the “COMMON NAME” Field was incorrect or cannot be cleanly erased, cross out the line of data and fill out a new line with the correct “COMMON NAME” and “FINAL COUNT”.

5.5.5.3 Voucher Specimens

Approximately 10% of each field crews' sites will be randomly pre-selected for re-identification by an independent taxonomist. A minimum of one complete voucher is required for each person performing field taxonomy and will consist of either preserved specimen(s) or digital images representative of all species in the sample, including common species. Multiple specimens per species can be used as vouchers, if necessary (i.e., to document different life or growth stages, or sexes). Note that a complete sample voucher does not mean that all individuals of each species will be vouchered, only enough so that independent verification can be achieved.

Digital images should be taken as voucher documentation for species that are recognized as Rare, Threatened, or Endangered – they should not be killed. Digital images should also be taken of fish specimens too large for preservation.

Certain states or regions may require that more fish vouchers are taken. Check with your state/regional coordinators to determine if your team will be required to collect complete vouchers at more than 10% or your sites.

For the sample voucher, specimen containers should be labeled with the sample number, site ID number, site name, and collection date. There should be no taxonomic identification labels in or on the container, or in any of the digital photos.

Choose individual specimens that are intact and in good condition, such that re-identification will be possible. Fish that are damaged, have significant scale loss or those that have been dead for a significant amount of time prior to preservation should be avoided if possible. Fish in pristine condition and those possessing clear identification characteristics are preferred. Additionally, fish that are preserved while still live will typically flare their fins and gills and will allow for easier re-identification in the laboratory.

Place one or more representative specimens of each species in plastic mesh sleeves along with one of the corresponding tag number labels provided in your site kit. (Several fish may be placed in a single mesh sleeve, as long as they are of the same species). Ensure that the tag numbers in the voucher collection match the tag numbers on the fish collection data forms. Seal both ends of the mesh sleeve with zip ties and place it inside the voucher collection jar with the appropriate preservative. Unknown fish may be identified in the laboratory as described in section 5.5.5.2 and subsequently included in the voucher collection.

Record the total number of each fish species retained for voucher purposes in each subreach on the fish collection form. Record the voucher sample ID number on the fish gear / voucher / fish tissue collection form. If no voucher is prepared for the site, fill in the "no vouchers preserved" circle on the fish gear form.

Table 5.5-5. Procedure for vouchering of fish samples.

1. Approximately 10% of each field crews' sites will be randomly pre-selected for re-identification by an independent taxonomist. A minimum of one complete voucher is required for each person performing field taxonomy and will consist of either preserved specimen(s) and/or digital images representative of all species in the sample, even common species.
2. Take digital images as voucher documentation for species that are recognized as Rare, Threatened, or Endangered; or when fish specimens are too large for preservation.
3. For the sample voucher, label the specimen containers with the sample number, site ID number, site name, and collection date. Do not put taxonomic identification labels in or on the container.
4. Place one or more representative specimens of each species in plastic mesh sleeves along with one of the corresponding tag number labels provided in your site kit. (Several fish may be placed in a single mesh sleeve, as long as they are of the same species).
5. Ensure that the tag numbers in the voucher collection match the tag numbers on the fish collection data forms.
6. Seal both ends of the mesh sleeve with zip ties and place it inside the voucher collection jar with the appropriate preservative.
7. Unknown fish may be identified in the laboratory as described in section 5.5.5.2 and subsequently included in the voucher collection.
8. Record the total number of each fish species retained for voucher purposes in each subreach on the fish collection form.
9. Record the voucher sample ID number on the fish gear / voucher / fish tissue collection form.
10. If no voucher is prepared for the site, fill in the "no vouchers preserved" circle on the fish gear form.

5.5.5.4 Photovouchering

Digital imagery should be used for fish species that cannot be retained as preserved specimens (e.g., RTE species; or very large bodied fish). Views appropriate and necessary for an independent taxonomist to accurately identify the specimen should be the primary goal of the photography. Additional detail for these guidelines is provided in Stauffer et al. (2001), and is provided to all field crews as a handout.

The recommended specifications for digital images to be used for photovouchering include: 16-bit color at a minimum resolution of 1024x768 pixels; macro lens capability allowing for images to be recorded at a distance of less than 4 cm; and built-in or external flash for use in low-light conditions. Specimens should occupy as much of the field of view as possible, and the use of a fish board is recommended to provide a reference to scale (i.e., ruler or some calibrated device) and an adequate background color for photographs. Information on Station ID, Date and TAG NUMBER should also be captured in the photograph, so that photos can be identified if file names become corrupted. All photovouchered species should have at least a full-body photo (preferably of the left side of the fish) and other zoom images as necessary for individual species, such as lateral line, ocular/oral orientation, fin rays, gill arches, or others. It may also be necessary to photograph males, females, or juveniles.

Images should be saved in medium- to high-quality jpeg format, with the resulting file name of each picture noted one the Fish Collection Form. It is important that time and date stamps are accurate, as this information can also be useful in tracking the origin of photographs. Because close-up photography is difficult in the best of conditions with typical point and shoot cameras, it might be best to take high quality pictures at a greater distance so that the image can be zoomed with a PC. It is recommended that images stored in the camera be transferred to a PC or storage device at the first available opportunity. At this time the original file should be renamed to follow the logic presented below:

F01_CT003_20080326_A.jpg

Where:
F = fish
01 = TAG NUMBER
CT003 = state (Connecticut) and site number
20080326 = date (yyyymmdd)
A = first of several pictures of same fish (e.g., A, B, C)

Field crews should maintain files for the duration of the sampling season. Notification regarding the transfer of all images to the existing database will be provided at the conclusion of the sampling. Only keep photos that are useful for identifications. If photos are to be submitted as vouchers, burn a CD of those photos that can be submitted along with the voucher jar.

5.6 Fish Tissue

5.6.1 Summary of Method

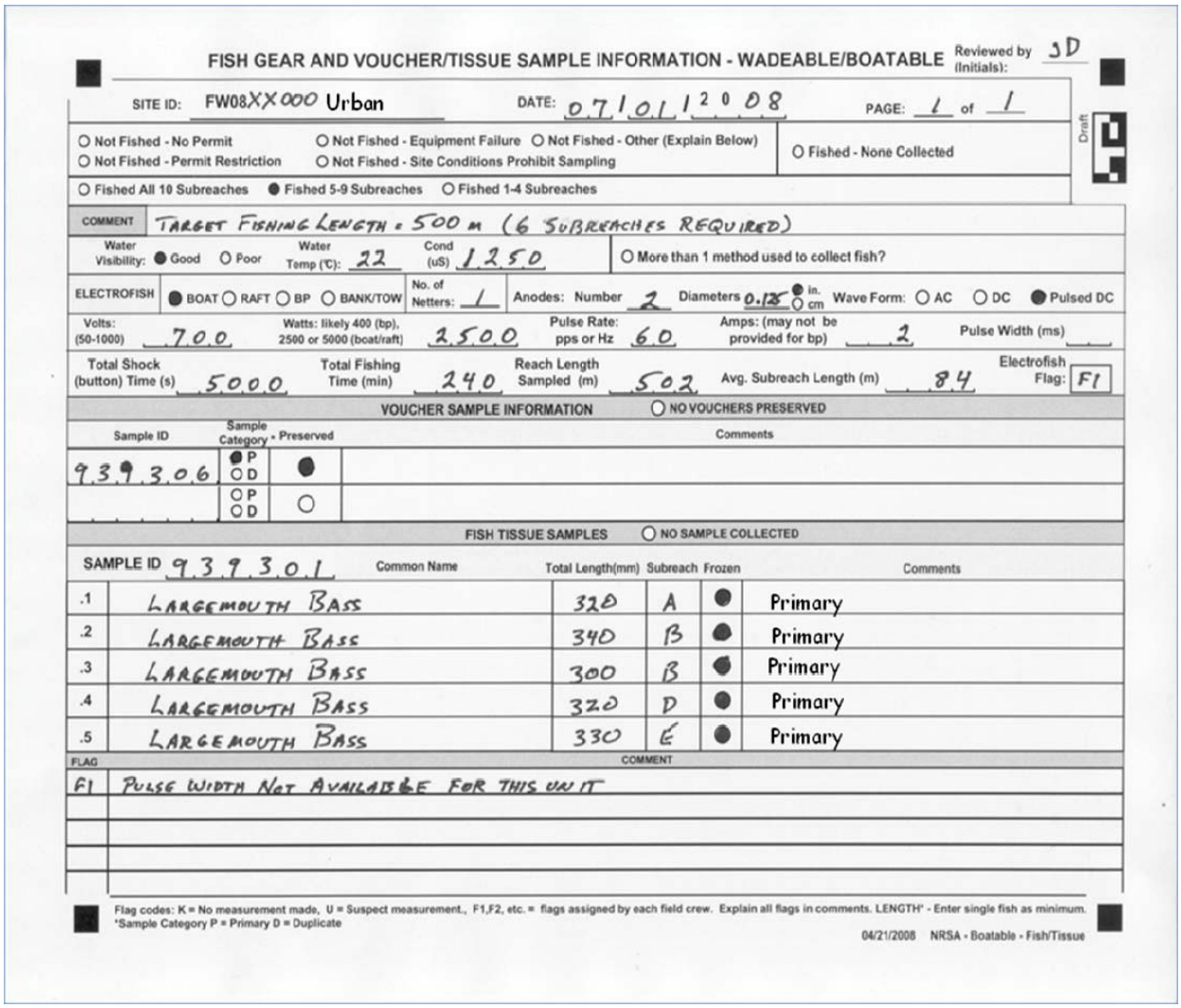
You will collect one predator species composite from each target site for human health related analyses. The focus is on fish species that commonly occur throughout the region of interest, and that are sufficiently abundant within a sampling reach. Each composite sample will consist of five adult fish of the same species that are similar in size (the smallest individual in the composite is no less than 75% of the total length of the largest individual). Collection occurs in the sampling reach.

5.6.2 Equipment and Supplies

Table 5.6-1 lists the equipment and supplies necessary for field crews to collect fish tissue samples. This list is comparable to the checklist presented in Appendix A, which provides information to ensure that field teams bring all of the required equipment to the site. Record the fish tissue sampling data on the Fish Gear and Voucher/Tissue Sample Information Form (Figure 5.6-1).

Table 5.6-1. Equipment and supplies—fish tissue collection at non-wadeable sites

For collecting fish composite sample	▪ Electrofishing equipment (including variable voltage pulsator unit, wiring cables, generator, electrodes, dip nets, protective gloves, boots, and necessary safety equipment) ▪ Scientific collection permit ▪ Sampling vessel (including boat, motor, trailer, oars, gas, and all required safety equipment)	▪ Coast Guard-approved personal floatation devices ▪ Maps of target sites & access routes ▪ Global Positioning System (GPS) unit ▪ Livewell and/or buckets ▪ Measuring board (millimeter scale) ▪ Clean nitrile gloves
For storing and preserving fish composite sample	▪ Aluminum foil (solvent-rinsed and baked) ▪ Heavy-duty food grade polyethylene tubing ▪ Large plastic (composite) bags	▪ Knife or scissors ▪ Dry Ice ▪ Plastic cable ties ▪ Coolers
For documenting the fish composite sample	▪ Fish Collection Forms ▪ Clipboard	▪ Sample Identification Labels ▪ #2 pencils ▪ Fine tipped indelible markers
For shipping the fish composite samples	▪ Preaddressed FedEx airbill ▪ Coolers	▪ Tracking Form ▪ Chain-of-custody labels ▪ Packing/strapping tape



FISH GEAR AND VOUCHER/TISSUE SAMPLE INFORMATION - WADEABLE/BOATABLE Reviewed by (Initials): JD

SITE ID: FW08XX000 Urban DATE: 07/01/2008 PAGE: 1 of 1

Draft

○ Not Fished - No Permit ○ Not Fished - Equipment Failure ○ Not Fished - Other (Explain Below)
○ Not Fished - Permit Restriction ○ Not Fished - Site Conditions Prohibit Sampling
○ Fished - None Collected

○ Fished All 10 Subreaches ● Fished 5-9 Subreaches ○ Fished 1-4 Subreaches

COMMENT: TARGET FISHING LENGTH = 500 m (6 SUBREACHES REQUIRED)

Water Visibility: ● Good ○ Poor | Water Temp (°C): 22 | Cond (uS) 1250 | ○ More than 1 method used to collect fish?

ELECTROFISH: ● BOAT ○ RAFT ○ BP ○ BANK/TOW | No. of Netters: 1 | Anodes: Number 2 | Diameters 0.25 ● in. ○ cm | Wave Form: ○ AC ○ DC ● Pulsed DC

Volts: (50-1000) 700 | Watts: likely 400 (bp), 2500 or 5000 (boat/raft) 2500 | Pulse Rate: pps or Hz 60 | Amps: (may not be provided for bp) 2 | Pulse Width (ms)

Total Shock (button) Time (s) 5000 | Total Fishing Time (min) 240 | Reach Length Sampled (m) 502 | Avg. Subreach Length (m) 84 | Electrofish Flag: F1

VOUCHER SAMPLE INFORMATION ○ NO VOUCHERS PRESERVED

Sample ID	Sample Category	Preserved	Comments
939306	● P ○ D	●	
	○ P ○ D	○	

FISH TISSUE SAMPLES ○ NO SAMPLE COLLECTED

SAMPLE ID 939301

	Common Name	Total Length(mm)	Subreach	Frozen	Comments
.1	LARGEMOUTH BASS	320	A	●	Primary
.2	LARGEMOUTH BASS	340	B	●	Primary
.3	LARGEMOUTH BASS	300	B	●	Primary
.4	LARGEMOUTH BASS	320	D	●	Primary
.5	LARGEMOUTH BASS	330	E	●	Primary

FLAG	COMMENT
F1	PULSE WIDTH NOT AVAILABLE FOR THIS UNIT

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = flags assigned by each field crew. Explain all flags in comments. LENGTH' - Enter single fish as minimum.
'Sample Category P = Primary D = Duplicate

04/21/2008 NRSA - Boatable - Fish/Tissue

Figure 5.6-1. Fish Gear and Voucher/Tissue Sample Information

5.6.3 Sampling Procedure

The fish tissue indicator will be collected using the same gear and procedures used to collect the fish community assemblage. Collection of individuals for fish tissue occurs in the sample reach during the fish community assemblage sampling. If the five fish are not collected during the community sampling, sample for up to one additional hour. If the sample is still not collected, call the Logistics Coordinator at the end of the day and record on the field collection form. If the target species are unavailable, the fisheries biologist will select an alternative species (i.e., a species that is commonly consumed in the study area, with specimens of harvestable or consumable size, and in sufficient numbers to yield a composite) to obtain a fish composite sample from the species that are available. Recommended target species, listed in order of preference, are given in Table 5.6-2.

Table 5.6-2. Recommended target species for fish tissue collection (in order of preference) at non-wadeable sites

Predator/Gamefish Species (in order of preference)	Family name	Common name	Scientific name	Length Guideline (Estimated Minimum)
	Centrarchidae	Largemouth bass	*Micropterus salmoides*	~280 mm
		Smallmouth bass	*Micropterus dolomieu*	~300 mm
		Black crappie	*Pomoxis nigromaculatus*	~330 mm
		White crappie	*Pomoxis annularis*	~330 mm
	Ictaluridae	Channel Catfish	*Ictalurus punctatus*	~300 mm
		Blue Catfish	*Ictalurus furcatus*	~300 mm
		Flathead Catfish	*Pylodictis olivaris*	~350 mm
	Percidae	Walleye/sauger	*Sander vitreus /S. canadensis*	~380 mm
		Yellow perch	*Perca flavescens*	~330 mm
	Percichthyidae	White bass	*Morone chrysops*	~330 mm
	Esocidae	Northern pike	*Esox lucius*	~430 mm
	Salmonidae	Brown trout	*Salmo trutta*	~300 mm
		Rainbow trout	*Oncorhynchus mykiss*	~300 mm
		Brook trout	*Salvelinus fontinalis*	~330 mm

The procedures for collecting and processing fish composite samples are presented in Table 5.6-3.

Table 5.6-3. Sampling procedure for fish composite samples at non-wadeable sites

1. Put on clean nitrile gloves before handling the fish. Do not handle any food, drink, sunscreen, or insect repellant until after the composite sample has been collected, measured, and wrapped.
2. Rinse potential target species/individuals in ambient water to remove any foreign material from the external surface and place in clean holding containers (e.g., livewells, buckets). Return non-target fishes or small specimens to the river or stream.
3. Retain one predator species composite from each site. The composite must consist of 5 fish of

adequate size to provide a total of 500 grams of edible tissue for analysis (refer to Table 5.6-2 for minimum species length guidelines). Select fish for each composite based on the following criteria:

- all are of the same species,
- all satisfy legal requirements of harvestable size (or weight) for the sampled river, or at least be of consumable size if no legal harvest requirements are in effect,
- all are of similar size, so that the smallest individual in a composite is no less than 75% of the total length of the largest individual, and
- all are collected at the same time, i.e., collected as close to the same time as possible, but no more than one week apart (Note: Individual fish may have to be frozen until all fish to be included in the composite are available for delivery to the designated laboratory).

Accurate taxonomic identification is essential in assuring and defining the organisms that have been composited and submitted for analysis. Under no circumstances should individuals from different species be used in a single composite sample.

4. Measure each individual fish to determine total body length. Measure total length of each specimen in millimeters, from the anterior-most part of the fish to the tip of the longest caudal fin ray (when the lobes of the caudal fin are depressed dorsoventrally).
5. Record sample number, species retained, specimen length, site ID, and sampling date on the Fish Collection Form (Figure 5.5-1) in black ink. Mark site type ("Urban" or "Non-urban") next to the site identification number at the top left of the fish form, and write primary or duplicate in the comment section. Make sure the sample identification numbers recorded on the collection form match those on the sample labels.
6. Remove each fish retained for analysis from the clean holding container(s) (e.g., livewell) using clean nitrile gloves. Dispatch each fish using a clean wooden bat (or equivalent wooden device).
7. Wrap each fish in extra heavy-duty aluminum foil with the dull side in (foil provided by EPA as solvent-rinsed, oven-baked sheets).
8. Prepare a Sample Identification Label for each sample, ensuring that the label information matches the information recorded on the Fish Collection Form. **Be sure to include fish species and specimen length on each label.**
9. Cut a length of food grade tubing (provided by EPA) that is long enough to contain each individual fish and to allow extra length on each end to secure with cable ties. Place each foil-wrapped specimen into the appropriate length of tubing. Seal each end of the tubing with a plastic cable tie. Attach the fish sample label to the outside of the food-grade tubing with clear tape and secure the label by taping around the entire fish (so that tape sticks to tape).
10. Place all the wrapped fish in the composite from each site in a large plastic bag and seal with another cable tie.
11. After each sample is packaged, place it immediately on dry ice for shipment. If samples will be carried back to a laboratory or other facility to be frozen before shipment, wet ice can be used to transport wrapped and bagged fish samples in the coolers to a laboratory or other interim facility.
12. If possible, keep all (five) specimens designated for a particular composite in the same shipping container (ice chest) for transport.
13. Samples may be stored temporarily on dry ice (replenishing the dry ice daily). You have the option, depending on site logistics, of:

    - shipping the samples packed on dry ice in sufficient quantities to keep samples frozen for up to 48 hours (50 pounds are recommended), via priority overnight delivery service (e.g., Federal Express), so that they arrive at the sample preparation laboratory within less than 24 hours from the time of sample collection, or

- freezing the samples within 24 hours of collection at ≤-20°C, and storing the frozen samples until shipment within 2 weeks of sample collection (frozen samples will subsequently be packed on dry ice and shipped to the sample preparation laboratory via priority overnight delivery service).

14. Ship fish tissue samples from urban sites to the EPA NERL lab in Cincinnati, OH and from non-urban sites to the GLEC lab in Traverse City, MI on Monday through Thursday.

5.7 Fecal Indicator (Enterococci)

5.7.1 Summary of Method

Collect a fecal indicator sample at the last transect (Transect K) after all other sampling is completed. Samples must be filtered and the filters must be frozen within 6 hours of collection. Use a pre-sterilized, 250 ml bottle and collect the sample approximately 1 m off the bank at about 0.3 meter (12 inches) below the water surface. Following collection, place the sample in a cooler, chill for at least 15 minutes, and maintain on ice prior to filtration of four 50 mL volumes. (Samples must be filtered and frozen on dry ice within 6 hours of collection). In addition to collecting the sample, look for signs of disturbance throughout the reach that would contribute to the presence of fecal contamination to the waterbody. Record these disturbances on the Site Assessment Form (Figure 7-2).

5.7.2 Equipment and Supplies

Table 5.7-1 provides the equipment and supplies needed to collect the fecal indicator sample. Record the sample data on the Sample Collection Form, Side 2 (Figure 5.1-4).

Table 5.7-1. Equipment and supplies list for fecal indicator sampling at non-wadeable sites

For collecting samples	▪ nitrile gloves ▪ pre-sterilized, 250 ml sample bottle	▪ sodium thiosulfate tablet ▪ Wet ice ▪ cooler
For recording measurements	▪ Sample Collection Form ▪ Fecal Indicator sample labels (4 vial labels and 1 bag label) ▪ Pencils (for data forms)	▪ Fine tipped indelible markers (for labels) ▪ Clear tape strips

5.7.3 Sampling Procedure

The procedure for collecting the fecal indicator sample is presented in table 5.7-2.

Table 5.7-2. Procedure for fecal indicator (Enterococci) sample collection at non-wadeable sites

1. Put on nitrile gloves.
2. Select a sampling location at transect K that is approximately 1 m from the bank and approximately 0.3m deep. Approach the sampling location slowly from downstream or downwind.
3. Lower the un-capped, inverted 250 ml sample bottle to a depth of 1 foot below the water surface, avoiding surface scum, vegetation, and substrates. Point the mouth of the container away from the body or boat. Right the bottle and raise it through the water column, allowing bottle to fill completely.

If the depth does not reach 0.3m along the transect at 1 m from the bank, take the sample and flag it on the field form.

4. After removing the container from the water, discard a small portion of the sample to allow for proper mixing before analyses.
5. Add the sodium thiosulfate tablet, cap, and shake bottle 25 times.
6. Store the sample in a cooler on ice to chill (not freeze). Chill for at least 15 minutes and do not hold samples longer than 6 hours before filtration and freezing.

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6.0 WADEABLE STREAMS

6.1 Water Quality

This section describes the procedures and methods for the field collection and analysis of the water quality indicators (in-situ measurements, water chemistry, and sediment enzymes) from wadeable streams and rivers.

6.1.1 In Situ Measurements of Dissolved Oxygen, pH, Temperature, and Conductivity

6.1.1.1 Summary of Method

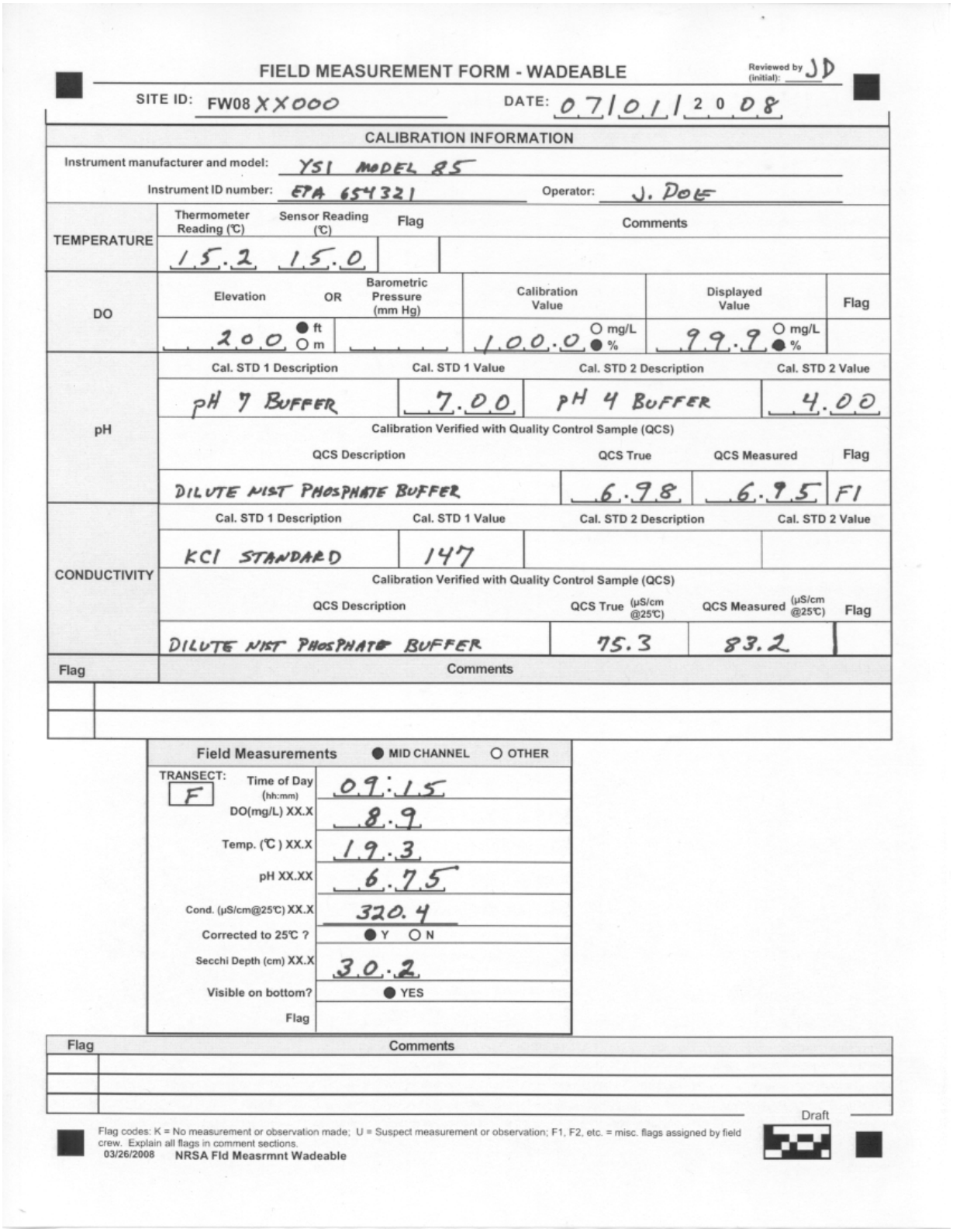
You will measure dissolved oxygen (DO), pH, temperature, and conductivity by using a multi-parameter water quality meter (or sonde). Take all measurements at the X site at 0.5 m depth, or mid-depth if depth is <1 m. The site depth must be accurately measured before taking the measurements, and care should be taken to avoid the probe contacting bottom sediments.

6.1.1.2 Equipment and Supplies

Table 6.1-1 provides the equipment and supplies needed to measure dissolved oxygen, pH, temperature, and conductivity. Record the measurements on the Field Measurement Form, as seen in Figure 6.1-1.

Table 6.1-1. Equipment and supplies—DO, pH, temperature, and conductivity

For taking measurements and calibrating the water quality meter	▪ Multi-parameter water quality meter with DO, pH, temperature, and conductivity probes. ▪ Extra batteries ▪ De-ionized and tap water ▪ Calibration cups and standards ▪ QC calibration standard ▪ Barometer or elevation chart to use for calibration
For recording measurements	▪ Field Measurement Form ▪ Pencils (for data forms)



FIELD MEASUREMENT FORM - WADEABLE

Reviewed by (initial): JD

SITE ID: FW08XX000 DATE: 07/01/2008

CALIBRATION INFORMATION

Instrument manufacturer and model: YSI MODEL 85

Instrument ID number: EPA 654321 Operator: J. DOE

	Thermometer Reading (°C)	Sensor Reading (°C)	Flag	Comments
TEMPERATURE	15.2	15.0		

	Elevation	OR	Barometric Pressure (mm Hg)	Calibration Value	Displayed Value	Flag
DO	200 ● ft ○ m			100.0 ○ mg/L ● %	99.9 ○ mg/L ● %	

	Cal. STD 1 Description	Cal. STD 1 Value	Cal. STD 2 Description	Cal. STD 2 Value
pH	pH 7 BUFFER	7.00	pH 4 BUFFER	4.00

Calibration Verified with Quality Control Sample (QCS)

QCS Description	QCS True	QCS Measured	Flag
DILUTE NIST PHOSPHATE BUFFER	6.98	6.95	F1

	Cal. STD 1 Description	Cal. STD 1 Value	Cal. STD 2 Description	Cal. STD 2 Value
CONDUCTIVITY	KCl STANDARD	147		

Calibration Verified with Quality Control Sample (QCS)

QCS Description	QCS True (µS/cm @25°C)	QCS Measured (µS/cm @25°C)	Flag
DILUTE NIST PHOSPHATE BUFFER	75.3	83.2	

Flag	Comments

Field Measurements ● MID CHANNEL ○ OTHER

TRANSECT: F	
Time of Day (hh:mm)	09:15
DO(mg/L) XX.X	8.9
Temp. (°C) XX.X	19.3
pH XX.XX	6.75
Cond. (µS/cm@25°C) XX.X	320.4
Corrected to 25°C ?	● Y ○ N
Secchi Depth (cm) XX.X	30.2
Visible on bottom?	● YES
Flag	

Flag	Comments

Draft

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = misc. flags assigned by field crew. Explain all flags in comment sections.

03/26/2008 NRSA Fld Measrmnt Wadeable

Figure 6.1-1. Field Measurement Form.

6.1.1.3 Multi-Probe Sonde

Dissolved Oxygen Meter

Calibrate the DO meter prior to each sampling event. We recommend that the probe be calibrated in the field against an atmospheric standard (ambient air saturated with water) prior to sampling. In addition, manufacturers typically recommend periodic comparisons with a DO chemical analysis procedure (e.g., Winkler titration) to check accuracy and linearity.

pH Meter

Calibrate the pH meter prior to each sampling event. Calibrate the meter in accordance with the manufacturer's instructions and with the team agency's existing SOP. You must also conduct a quality control check with the provided standard to verify the calibration and periodically evaluate instrument precision (see Section 3.1.2). Crews must check their probe once a week against the provided Quality Control Standard (QCS) and record the information on the data forms.

Temperature Meter

You must check the accuracy of the sensor against a thermometer that is traceable to the National Institute of Standards (NIST) at least once per sampling season. The entire temperature range encountered in the NRSA should be incorporated in the testing procedure and a record of test results kept on file.

Conductivity Meter

Calibrate the conductivity meter prior to each sampling event. Calibrate the meter in accordance with the manufacturer's instructions. The entire conductivity range encountered in the NRSA should be incorporated in the testing procedure and a record of test results kept on file. You must also conduct a quality control check with the provided standard to verify the calibration and periodically evaluate instrument precision (see Section 3.1.2). Crews must check their probe once a week against the provided QCS and record the information on the data forms.

6.1.1.4 Sampling Procedure

Table 6.1-2 presents step-by-step procedures for measuring dissolved oxygen, pH, temperature, and conductivity.

Table 6.1-2. Sampling procedure—temperature, pH, conductivity and dissolved oxygen

1. Check meter and probes and calibrate according to manufacturer's specifications.
2. **Wadeable Sites:** Measurements are taken at the X site at a depth of 0.5 meters or at mid-depth if less than 1 meter deep.
3. Lower the sonde in the water and measure DO, pH, temperature, and conductivity at 0.5 m depth.
4. Record the measurements on the Field Measurement Form.
5. If sampling at the X-site is not possible, move to another part of the reach to collect the sample (as close to the X-site as possible), record the letter of the nearest transect in the "TRANSECT" box and more detailed reasons and/or information in the Comments section.
6. Flag any measurements that need further comment (or when a measurement cannot be made).

6.1.2 Water Chemistry Sample Collection and Preservation

6.1.2.1 Summary of Method

The water chemistry samples will be analyzed for total phosphorus (TP), total nitrogen (TN), total ammonia-nitrogen (NH_4), nitrate (NO_3), basic anions, cations, total suspended solids (TSS), turbidity, acid neutralizing capacity (ANC, alkalinity), dissolved organic carbon (DOC), and total organic carbon (TOC). You will collect a grab sample in one 4-L cube container and in one 2-L amber Nalgene bottle from the X site at the center of the reach. Store all samples on ice in a closed cooler.

6.1.2.2 Equipment and Supplies

Table 6.1-3 provides the equipment and supplies needed to collect water samples at the index site. Record the Water Sample Collection and Preservation data on the Sample Collection Form, as seen in Figure 6.1-2.

Table 6.1-3. Equipment and supplies—water chemistry sample collection and preservation

For collecting samples	▪ Nitrile gloves ▪ 4-L cube container for wadeable sites ▪ 2-L amber Nalgene bottle ▪ 3 L Nalgene beaker ▪ Cooler with ice ▪ DI water (for cleaning beaker and carboy between sites) ▪ Field Operations Manual and/or laminated Quick Reference Guide
For recording measurements	▪ Sample Collection Form ▪ Field Measurement Form ▪ Pencils (for data forms) ▪ Fine tipped indelible markers

6.1.2.3 Sampling Procedure

Table 6.1-4 presents step-by-step procedures for collecting water chemistry samples at wadeable sites.

Table 6.1-4. Sampling procedure for wadeable sites—water chemistry sample collection

1. Collect the water samples from the X-site in a flowing portion near the middle of the stream.
2. Put on nitrile gloves. Make sure not to handle sunscreen or other chemical contaminants until after the sample is collected.
3. Rinse the 3-L Nalgene beaker three times with water, and discard the rinse downstream.
4. Remove the cube container lid and expand the cube container by pulling out the sides. **NOTE: DO NOT BLOW into the cube container to expand them, this will cause contamination.**
5. Fill the 3-liter beaker with water and slowly pour 30 - 50 mL into the cube container. Cap the cube container and rotate so that the water contacts all the surfaces. Discard the water downstream. Repeat this rinsing procedure 2 more times.
6. Fill the beaker with water and pour into the cube container. Repeat as necessary to fill the cube container. Let the weight of the water expand the cube container. Pour the water slowly as the cube container expands. Fill the cube container to at least three-fourths of its maximum volume. Rinse the cube container lid with water. Eliminate any air space from the cube container, and cap it tightly. Make sure the cap is tightly sealed and not on at an angle.
7. Fill the 3-liter beaker with water and slowly pour 30 - 50 mL into the 2 L amber Nalgene bottle. Cap the bottle and rotate so that the water contacts all the surfaces. Discard the water downstream. Repeat this rinsing procedure 2 more times.
8. Fill the beaker with water and pour into the 2 L amber Nalgene bottle. Cap the bottle tightly
9. Place the cube container and bottle in a cooler (on ice or water) and shut the lid. If a cooler is not available, place the cube container in an opaque garbage bag and immerse it in the stream.
10. Record the Sample ID on the Sample Collection Form along with the pertinent stream information (stream name, ID, date, etc.). Note anything that could influence sample chemistry (heavy rain, potential contaminants) in the Comments section. If sampling at the X-site is not possible, move to another part of the reach to collect the sample (as close to the X-site as possible), record the letter of the nearest transect and more detailed reasons and/or information in the Comments section.

6.1.3 Sediment Enzymes

6.1.3.1 Summary of Method

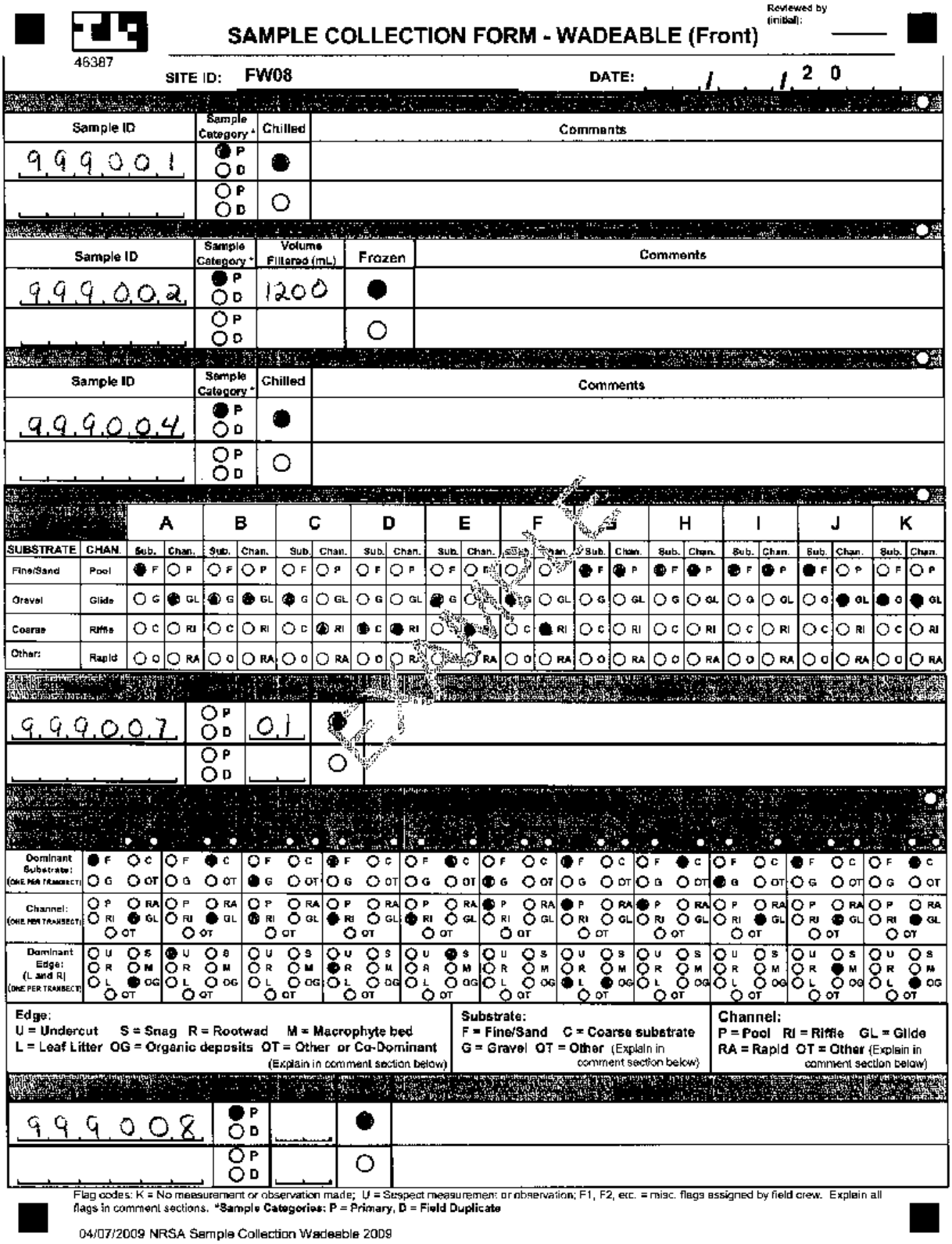
Collect sediment samples at the 11 sampling stations along each reach and combine for all stations at a site, resulting in a single 500 mL sample per site. Collect fine surface sediments (top 5 cm) using a scoop, spoon or dredge. Store samples on ice until shipment to the laboratory. Samples will be analyzed for available DIN, NH_4, DIP, TP, TN, total carbon (TC) and enzyme activity.

6.1.3.2 Equipment and Supplies

Table 6.1-5 lists the equipment and supplies needed to collect sediment enzyme samples. Record collection data on the Sample Collection Form, as seen in Figure 6.1-2.

Table 6.1-5. Equipment and supplies—sediment enzymes

For collecting samples	▪ 4 L graduated plastic bucket ▪ Large stainless steel spoon for mixing sediment composite ▪ 500 mL plastic jar for storing sediment sample
For recording measurements	▪ Sample Collection Form ▪ Sample labels ▪ Pencils ▪ Fine tipped indelible markers ▪ Clear tape strips



SAMPLE COLLECTION FORM - WADEABLE (Front)

Reviewed by (initial): _____

46387

SITE ID: FW08 DATE: / / 20

Sample ID	Sample Category*	Chilled	Comments
999001	P / D	●	
	P / D	○	

Sample ID	Sample Category*	Volume Filtered (mL)	Frozen	Comments
999002	P / D	1200	●	
	P / D		○	

Sample ID	Sample Category*	Chilled	Comments
999004	P / D	●	
	P / D	○	

		A		B		C		D		E		F		G		H		I		J		K	
SUBSTRATE	CHAN.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.
Fine/Sand	Pool	● F	○ P	○ F	○ P	○ F	○ P	○ F	○ P	○ F	○ P	○ F	○ P	● F	● P	● F	● P	● F	● P	● F	○ P	○ F	○ P
Gravel	Glide	○ G	● GL	● G	● GL	● G	○ GL	○ G	○ GL	● G	○ GL	● G	○ GL	○ G	○ GL	○ G	○ GL	○ G	○ GL	○ G	● GL	● G	● GL
Coarse	Riffle	○ C	○ RI	○ C	○ RI	○ C	● RI	● C	● RI	○ C	● RI	○ C	● RI	○ C	○ RI	○ C	○ RI	○ C	○ RI	○ C	○ RI	○ C	○ RI
Other:	Rapid	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA

999007	P / D	0.1	●
	P / D		○

	A	B	C	D	E	F	G	H	I	J	K
Dominant Substrate: (ONE PER TRANSECT)	● F ○ C ○ G ○ OT	○ F ● C ○ G ○ OT	○ F ○ C ● G ○ OT	● F ○ C ○ G ○ OT	○ F ● C ○ G ○ OT	○ F ○ C ● G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	● F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ● C ○ G ○ OT
Channel: (ONE PER TRANSECT)	○ P ○ RA ○ RI ● GL ○ OT	○ P ○ RA ○ RI ● GL ○ OT	● P ○ RA ● RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	● P ○ RA ○ RI ○ GL ○ OT	● P ○ RA ○ RI ○ GL ○ OT	● P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ● GL ○ OT	● P ○ RA ● RI ○ GL ○ OT	○ P ○ RA ○ RI ● GL ○ OT
Dominant Edge: (L and R) (ONE PER TRANSECT)	○ U ○ S ○ R ○ M ○ L ● OG ○ OT	● U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ● R ○ M ○ L ○ OG ○ OT	○ U ● S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ● OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ● M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ● OG ○ OT

Edge:
U = Undercut S = Snag R = Rootwad M = Macrophyte bed
L = Leaf Litter OG = Organic deposits OT = Other or Co-Dominant (Explain in comment section below)

Substrate:
F = Fine/Sand C = Coarse substrate
G = Gravel OT = Other (Explain in comment section below)

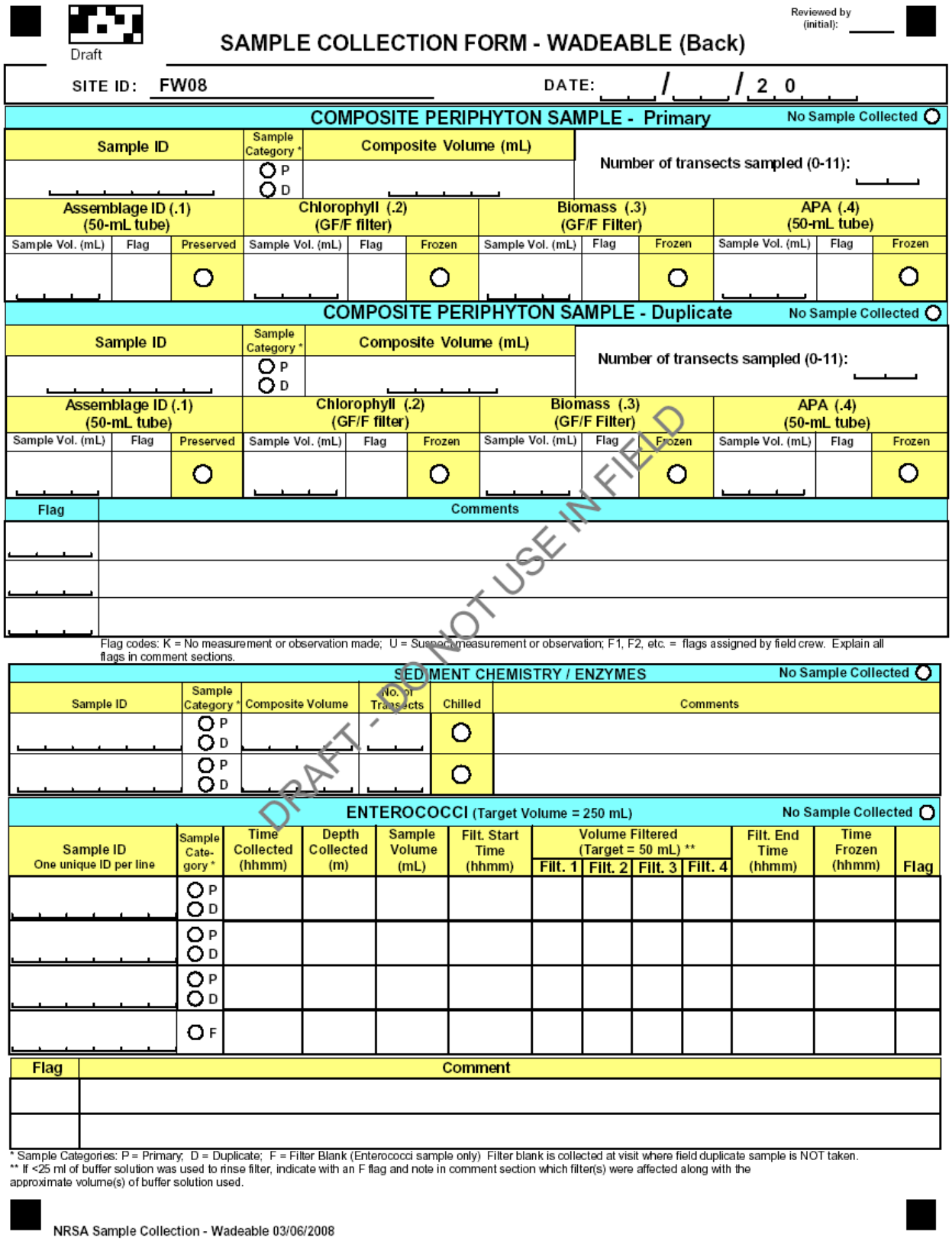
Channel:
P = Pool RI = Riffle GL = Glide
RA = Rapid OT = Other (Explain in comment section below)

999008	P / D		●
	P / D		○

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = misc. flags assigned by field crew. Explain all flags in comment sections. *Sample Categories: P = Primary, D = Field Duplicate

04/07/2009 NRSA Sample Collection Wadeable 2009

Figure 6.1-2. Sample Collection Form, Side 1.



Draft

Reviewed by (initial): ____

SAMPLE COLLECTION FORM - WADEABLE (Back)

SITE ID: FW08 DATE: ___/___/20___

COMPOSITE PERIPHYTON SAMPLE - Primary — No Sample Collected ○

Sample ID	Sample Category *	Composite Volume (mL)	Number of transects sampled (0-11):
	○ P ○ D		

Assemblage ID (.1) (50-mL tube)			Chlorophyll (.2) (GF/F filter)			Biomass (.3) (GF/F Filter)			APA (.4) (50-mL tube)		
Sample Vol. (mL)	Flag	Preserved	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen
		○			○			○			○

COMPOSITE PERIPHYTON SAMPLE - Duplicate — No Sample Collected ○

Sample ID	Sample Category *	Composite Volume (mL)	Number of transects sampled (0-11):
	○ P ○ D		

Assemblage ID (.1) (50-mL tube)			Chlorophyll (.2) (GF/F filter)			Biomass (.3) (GF/F Filter)			APA (.4) (50-mL tube)		
Sample Vol. (mL)	Flag	Preserved	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen
		○			○			○			○

Flag	Comments

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = flags assigned by field crew. Explain all flags in comment sections.

SEDIMENT CHEMISTRY / ENZYMES — No Sample Collected ○

Sample ID	Sample Category *	Composite Volume	No. of Transects	Chilled	Comments
	○ P ○ D			○	
	○ P ○ D			○	

ENTEROCOCCI (Target Volume = 250 mL) — No Sample Collected ○

Sample ID One unique ID per line	Sample Category *	Time Collected (hhmm)	Depth Collected (m)	Sample Volume (mL)	Filt. Start Time (hhmm)	Volume Filtered (Target = 50 mL) ** Filt. 1	Filt. 2	Filt. 3	Filt. 4	Filt. End Time (hhmm)	Time Frozen (hhmm)	Flag
	○ P ○ D											
	○ P ○ D											
	○ P ○ D											
	○ F											

Flag	Comment

* Sample Categories: P = Primary; D = Duplicate; F = Filter Blank (Enterococci sample only) Filter blank is collected at visit where field duplicate sample is NOT taken.
** If <25 ml of buffer solution was used to rinse filter, indicate with an F flag and note in comment section which filter(s) were affected along with the approximate volume(s) of buffer solution used.

NRSA Sample Collection - Wadeable 03/06/2008

Figure 6.1-3. Sample Collection Form, Side 2.

6.1.3.3 Sampling Procedure

Near each of the macroinvertebrate and periphyton sampling locations, collect a fine-grained sediment sample using either a hand scoop or spoon sampler. The objective is to collect a 500-mL composite sample that is representative of depositional areas at the site. The composite sample will be subsampled in the lab for multiple analyses. Table 6.1-6 presents step-by-step procedures for collecting sediment enzyme samples.

Table 6.1-6. Sampling procedure—sediment enzymes

1. Collect a sediment sample at each of the macroinvertebrate and periphyton sample locations. Make sure each of the subsamples comprises an approximately equal portion of the total composite. It is permissible to collect sediment between stations to insure a composite volume of at least 500 mL. (Note any deviations from standard procedure in a comment.)
2. Locate sediment samples in areas or patches of fine-grained substrate (silty sand, silt, clay, muck) in a zone bounded on the shore side by the apparent low-water mark from daily flow fluctuations and bounded on the river side by the 0.3-m (usually about mid-biceps) depth contour (recommended maximum sample depth; deeper sampling may be possible). The low-water mark at a site can often be detected by the presence of periphyton or attached filamentous algae just below the low-water mark. If samples cannot be safely collected by wading at a station due to vertical banks or other reason go to step 5.
3. Be sure to avoid the area that has just been kick sampled for macroinvertebrates. Sampling up-stream from the kick sample location is recommended. If fine substrates are not present within 5 m up- or downstream from the station, flag the station on the form.
4. If fine substrate is present, use a stainless steel spoon to collect a sample of about 50ml or one spoonful from the top 5 cm of substrate. Place the sample in a clean bucket. Use gloves for handling sediment. Do not assume rip rapped shorelines lack fine-grained sediment. Look for fines between the large rocks.
5. Repeat steps 2-4 at each of the 11 littoral stations. Record the total number of replicates (stations) included in the composite. Note in a comment the stations at which sediment was collected using a non-wading method.
6. It is important that a sufficient sediment (not less than 500 mL) sample for analysis be collected. If multiple stations have no fine sediment, it is permissible to collect extra sample at stations that do have fine sediment or between stations. Be sure to note this in a comment.
7. Using the stainless steel spoon, thoroughly mix the composite sample and transfer 500 mL into the 500 mL plastic bottle. Place in a cooler with ice for final labeling and preservation.
8. Prepare a label for the sample jar. Using a fine-point indelible marker, fill in the site # and sample date. Place the label on the jar and cover it with clear tape. Record the sample ID and other data on sampling form. Place the sample on ice or in a refrigerator. Do not freeze sediment samples. The sediment enzyme samples have a 2 week holding time.

6.2 Physical Habitat Characterization—Wadeable Streams

Physical habitat in streams includes all those physical attributes that influence or provide sustenance to organisms within the stream. The physical habitat of a stream varies naturally, thus expectations differ even in the absence of anthropogenic disturbance. Within a given physiographic-climatic region, stream drainage area and overall stream gradient are likely to be strong natural determinants of many aspects of stream habitat. This is because of their influence on discharge, flood stage, and stream power (the product of discharge times gradient). Kaufmann (1993) identified seven general physical habitat attributes important in influencing stream ecology:

- Channel Dimensions
- Channel Gradient
- Channel Substrate Size and Type
- Habitat Complexity and Cover
- Riparian Vegetation Cover and Structure
- Anthropogenic Alterations
- Channel-Riparian Interaction

The procedures are employed on a support reach length 40 times its baseflow wetted width, as described in Section 4. Measurement points are systematically placed to statistically represent the entire reach. Stream depth and wetted width are measured at very tightly spaced intervals, whereas channel cross-section profiles, substrate, bank characteristics and riparian vegetation structure are measured at larger intervals. Woody debris is tallied along the full length of the sampling reach, and discharge is measured at one location. The tightly spaced depth and width measures allow calculation of indices of channel structural complexity, objective classification of channel units such as pools, and quantification of residual pool depth, pool volume, and total stream volume.

6.2.1 Components of the Habitat Characterization

There are five components of the physical habitat characterization (Table 6.2-1). Measurements are recorded on 11 copies of a two-sided field form, and separate forms for recording slope and bearing measurements, recording observations concerning riparian *legacy* (large) trees and alien invasive riparian plants, assessing the degree of channel constraint, and recording evidence of debris torrents or recent major flooding. The *thalweg profile* is a longitudinal survey of depth, habitat class, presence of deposits of soft/small sediments, and presence of off-channel habitats at 100 equally spaced stations (150 in streams less than 2.5 m wide) along the centerline between the two ends of the sampling reach. *Thalweg* refers to the flow path of the deepest water in a stream channel. Wetted width is measured and substrate size is evaluated at 21 equally spaced cross-sections (at 11 regular transects [A through K], and 10 supplemental cross-sections spaced midway between each of these). Data for the second component, the *woody debris tally*, are recorded for each of 10 segments of stream located between the 11 regular transects. The third component, the *channel and riparian characterization*, includes measures and/or visual estimates of channel dimensions, substrate, fish cover, bank characteristics, riparian vegetation structure, presence of large (legacy) riparian trees, nonnative (alien) riparian plants, and evidence of human disturbances. These data are obtained at each of the 11 equally-spaced transects established within the sampling reach. In addition, measurements of the stream slope and compass bearing between stations are obtained, providing information necessary for calculating reach gradient, residual pool volume, and channel sinuosity. The fourth component, *assessment of channel constraint, debris*

torrents, and major floods, is an overall assessment of these characteristics for the whole reach, and is undertaken after the other components are completed.

Table 6.2-1. Components of physical habitat characterization

Component	Description
Thalweg Profile (Section 6.2.4.1)	▪ Measure maximum depth, classify habitat and pool-forming features, and check presence of backwaters, side channels and loose, soft deposits of sediment particles at 10-15 equally spaced intervals between each of 11 transects (100 or 150 individual measurements along entire reach). ▪ Measure wetted width and evaluate substrate particle size classes at 11 cross-section transects and midway between them (21 width measurements and substrate cross-sections).
Woody Debris Tally (Section 6.2.4.2)	▪ Between each of the channel cross-sections, tally large woody debris numbers within and above the bankfull channel according to specified length and diameter classes (10 separate tallies).
Channel and Riparian Characterization (Section 6.2.5)	▪ At 11 transects (21 for substrate size) placed at equal intervals along reach: ▪ Measure: channel cross-section dimensions, bank height, bank undercut distance, bank angle, slope and compass bearing (backsight), and riparian canopy density (densiometer). ▪ Visually Estimate[a]: substrate size class and embeddedness; areal cover class and type (e.g., woody trees) of riparian vegetation in Canopy, Mid-Layer and Ground Cover; areal cover class of fish concealment features, aquatic macrophytes and filamentous algae. ▪ Observe & Record[a]: Presence and proximity of human disturbances, presence of large trees, and presence of invasive riparian plants.
Assessment of Channel Constraint, Debris Torrents, and Major Floods (Section 6.2.6)	▪ After completing thalweg and transect measurements and observations, identify features causing channel constraint, estimate the percentage of the channel margin that is constrained for the whole reach, and estimate the ratio of bankfull/valley width. Check evidence of recent major floods and debris torrent scour or deposition.
Discharge (Section 6.2.6.3)	▪ Measure water depth and velocity at 0.6 depth at 15 to 20 equally spaced intervals across one carefully chosen channel cross-section. ▪ In very small streams, measure discharge by timing the passage of a neutrally buoyant object through a segment whose cross-sectional area has been estimated or by timing the filling of a bucket.

[a] Substrate size class is estimated for a total of 105 particles taken at 5 equally-spaced points along each of 21 cross-sections. Depth is measured and embeddedness estimated for the 55 particles located along the 11 regular transects A through K. Cross-sections are defined by laying the surveyor's rod or tape to span the wetted channel. Woody debris is tallied over the distance between each cross-section and the next cross-section upstream. Riparian vegetation and human disturbances are observed 5m upstream and 5m downstream from the cross-section transect. They extend shoreward 10m from left and right banks. Fish cover types, aquatic macrophytes, and algae are observed within the channel 5m upstream and 5m downstream from the cross-section stations. These boundaries for visual observations are estimated by eye.

6.2.2 Habitat Sampling Locations within the Reach

Measurements are made at two scales of resolution along the length of the reach; the results are later aggregated and expressed for the entire reach, a third level of resolution. Figure 6.2-1 illustrates the locations within the reach where data for the different components of the physical habitat characterization are obtained. Many channel and riparian features are characterized on 11 cross-sections and pairs of riparian plots spaced at 4 channel-width intervals (i.e., transect spacing = 1/10th the total reach length). The thalweg profile measurements must be spaced evenly over the entire support reach. In addition, they must be sufficiently close together that they do not miss deep areas and major habitat units. Follow these guidelines for choosing the increment between thalweg profile measurements:

- Channel Width < 2.5 m — increment = 1.0 m
- Channel Width 2.5 to 3.5 m — increment = 1.5 m
- Channel Width > 3.5 m — increment = 0.01 × (reach length)

Following these guidelines, make 150 evenly spaced thalweg profile measurements in the smallest category of streams, 15 between each detailed channel cross-section. In all of the larger stream sizes, you will make 100 measurements, 10 between each cross-section.

6.2.3 Logistics and Work Flow

The five components (Table 6.2-1) of the habitat characterization are organized into four grouped activities:

1. *Thalweg Profile and Large Woody Debris Tally (Section 6.2.4)*. Two people proceed upstream from the downstream end of the sampling reach (see Figure 6.2-1) making observations and measurements at the chosen increment spacing. One person is in the channel making width and depth measurements, and determining whether soft/small sediment deposits are present under his/her staff. The other person records these measurements, classifies the channel habitat, records presence/absence of side channels and off-channel habitats (e.g., backwater pools, sloughs, alcoves), and tallies large woody debris. Each time this team reaches a flag marking a new cross-section transect, they start filling out a new copy of the Thalweg Profile and Woody Debris Form. They interrupt the thalweg profile and woody debris tallying activities to complete data collection at each cross-section transect as it comes. When the crew member in the water makes a width measurement at channel locations midway between regular transects (i.e., *A*, *B*, *K*), she or he also locates and estimates the size class of the substrate particles on the left channel margin and at positions 25%, 50%, 75%, and 100% of the distance across the wetted channel. Procedures for this substrate tally are the same as for those at regular cross-sections, but data are recorded on the thalweg profile side of the field form.

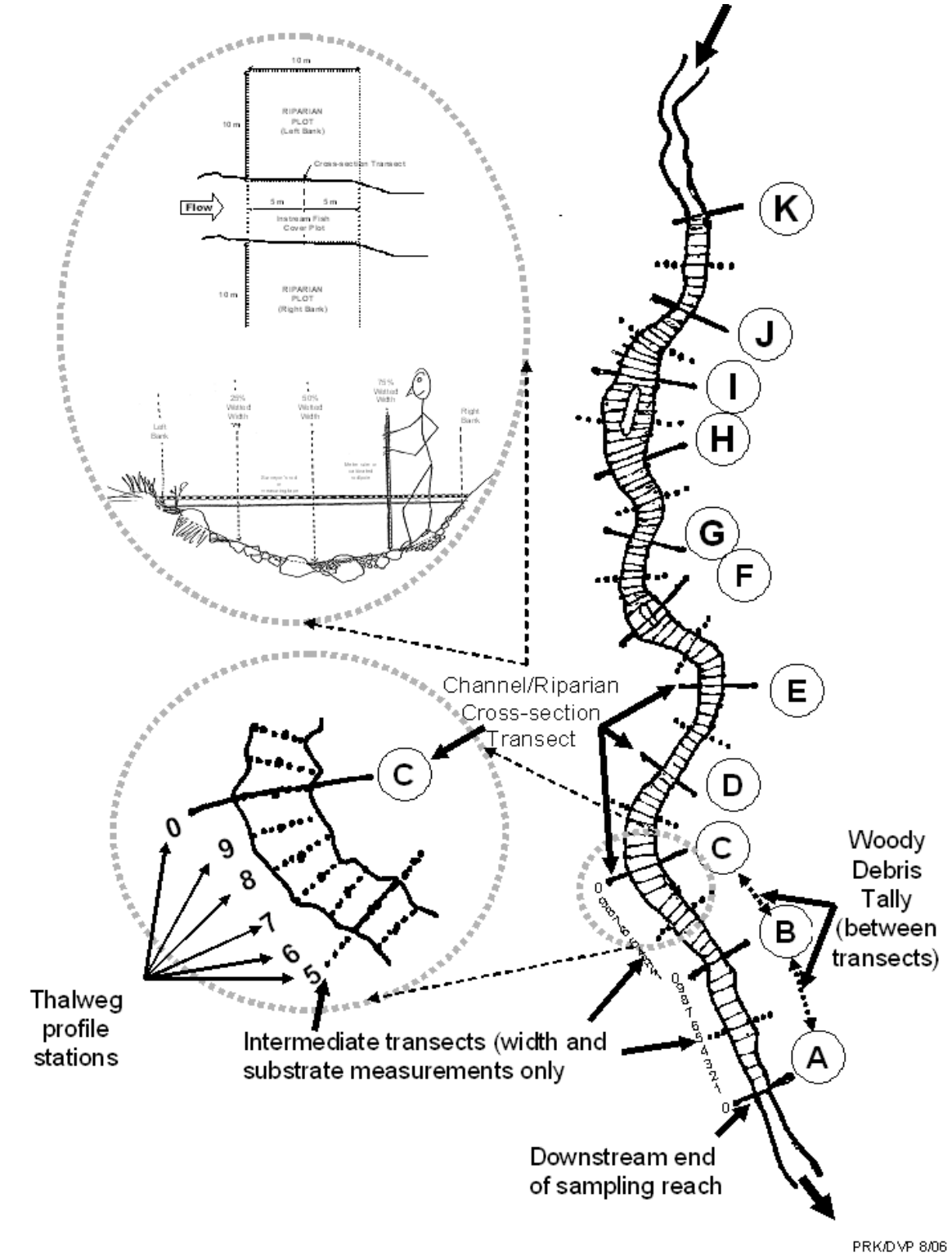


Figure 6.2-1. Reach layout for physical habitat measurements (plan view).

2. *Channel/Riparian Cross-Sections (Section 6.2.5)*. One person proceeds with the channel cross-section dimension, substrate, bank, and canopy cover measurements. The second person records those measurements on the Channel/ Riparian Cross-section Form while making visual estimates of riparian vegetation structure, instream fish cover, and human disturbance specified on that form. They also make observations to complete the riparian "legacy" tree field form. Slope is measured by measuring the difference in elevation between each transect and bearing is determined by backsighting to the previous transect. Supplementary points may need to be located and flagged (using a different color) if the stream is extremely brushy, sinuous, or steep to the point that you cannot sight for slope and bearing measures between two adjacent transects.

   The work flow for the thalweg profile and channel cross described above can be modified by delaying the measurements for slope and bearing and the woody debris tally until after reaching the upstream end of the reach. Backsighting and wood tallies can be done on the way back down (Note that in this case, the slope and bearing data form would have to be completed in reverse order).

3. *Channel Constraint and Torrent Evidence (Section 6.2.6)*. After completing observations and measurements along the thalweg and at all 11 transects, the field crew completes the overall reach assessments of channel constraint and evidence of debris torrents and major floods.

4. *Stream Discharge*. Discharge measurements are made after collecting the water chemistry sample. They are done at a chosen optimal cross-section (but not necessarily at a transect) near the X-site. However, do not use the electromagnetic current meter close to where electrofishing is taking place. Furthermore, if a lot of channel disruption is necessary and sediment must be stirred up, wait on this activity until all chemical and biological sampling has been completed.

6.2.4 Thalweg Profile and Large Woody Debris Measurements

6.2.4.1 Thalweg Profile

Thalweg refers to the flow path of the deepest water in a stream channel. The thalweg profile is a longitudinal survey of maximum flow path depth and several other selected characteristics at 100 or 150 equally spaced points (termed *stations*) along the length of the reach measured along the centerline of the channel. Data from the thalweg profile allows calculation of indices of residual pool volume, stream size, channel complexity, and the relative proportions of habitat types such as riffles and pools. One person walks upstream carrying a fiberglass telescoping (1.5 to 7.5 m) surveyor's rod and a 1-m metric ruler (or a calibrated rod or pole, such as a ski pole, shovel handle, wooden dowel, or old billiard cue). A second person on the bank or in the stream carries a clipboard with 11 copies of the field data form.

The procedure for obtaining thalweg profile measurements is presented in Table 6.2-2. Record data on the Thalweg Profile and Woody Debris Data Form as shown in Figure 6.2-2. Use the surveyor's rod and a metric ruler or calibrated rod or pole to make the required depth and width measurements at each station, and to measure off the distance between stations as you proceed upstream. You may need to make minor adjustments to align each 10^{th} measurement to be one increment short of the next transect. In streams with average widths less than 2.5 m, make thalweg measurements at 1-meter increments. Because the minimum reach length is set

at 150 meters, there will be 15 measurements on a field data form: Station 0 at the transect plus 14 additional stations between it and the next transect upstream. Use the five extra lines on the thalweg profile portion of the data form (Figure 6.2-2) to record these measurements.

Table 6.2-2. Thalweg profile procedure

1. Determine the increment distance between measurement stations based on the wetted width used to determine the length of the reach. Using a laser rangefinder or surveyor's rod:
   - For widths ≤ 2.5 m, establish stations every 1 m (150 total).
   - For widths > 2.5 and ≤3.5 m, establish stations every 1.5 m (100 total).
   - For widths > 3.5 m, establish stations at increments equal to 0.01 times the reach length (100 total).
2. Complete the header information on the Thalweg Profile and Woody Debris Form, noting the transect pair (downstream to upstream). Record the increment distance determined in Step 1 in the *INCREMENT* field on the field data form.
3. Begin at the downstream end (*station 0*) of the first transect (transect *A*).
4. Measure the wetted width at station *0*, and at either station *5* (if the stream width defining the reach length is ≥ 2.5 m), or station *7* (if the stream width defining the reach length is < 2.5 m). Wetted width is measured across and over mid-channel bars and boulders. Record the width on the field data form to the nearest 0.1 m. For streams with interrupted flow, where no water is in the channel at the station or transect, record zeros for wetted width.

NOTE: If a mid-channel bar is present at a station where wetted width is measured, measure the wetted width across and including the bar, but also measure the bar width and record it on the field data form.

5. At station *5* or *7* (see above) classify the size of the bed surface particle at the tip of your depth measuring rod at the left wetted margin and at positions 25%, 50%, 75%, and 100% of the distance across the wetted width of the stream. This procedure is identical to the substrate size evaluation procedure described for regular channel cross-sections (transects *A* - *K*), except that for these midway supplemental cross-sections, substrate size is entered on the thalweg profile side of the field form.
6. At each thalweg profile station, use a calibrated pole or rod to locate the deepest point within the deepest flow path (*the thalweg*), which may not always be found at mid-channel (and may not always be the absolute deepest point in every channel cross-section). Measure the thalweg depth to the nearest cm from the substrate surface to the water surface, and record it on the thalweg profile form. Read the depth on the **side** of the rod to avoid inaccuracies due to the wave formed by the rod in moving water.

NOTE: For streams with interrupted flow - if there is no water at a transect, record zeros for depth.

*NOTE: Obtain thalweg depths at all stations. If the thalweg is too deep to measure directly, stand in shallower water and extend the surveyor's rod or pole at an angle to reach the thalweg. Determine the angle by resting the clinometer on the upper surface of the rod and reading the angle on the external scale of the clinometer. Leave the depth reading for the station blank, and record a U flag to indicate a non-standard procedure was used. Record the water level on the rod and the rod angle in the comments section of the field data form. For deeper depths, use the same procedure with a taut string as the measuring device. Tie a weight to one end of a length of string or fishing line, and toss the weight into the deepest channel location. Draw the string up tight and measure the length of the line that is under water. Measure the string angle with the clinometer exactly as done for the surveyor's rod. If a direct measurement cannot be obtained, make the **best estimate** you can of the thalweg depth, and use a U flag to identify it as an estimated measurement.*

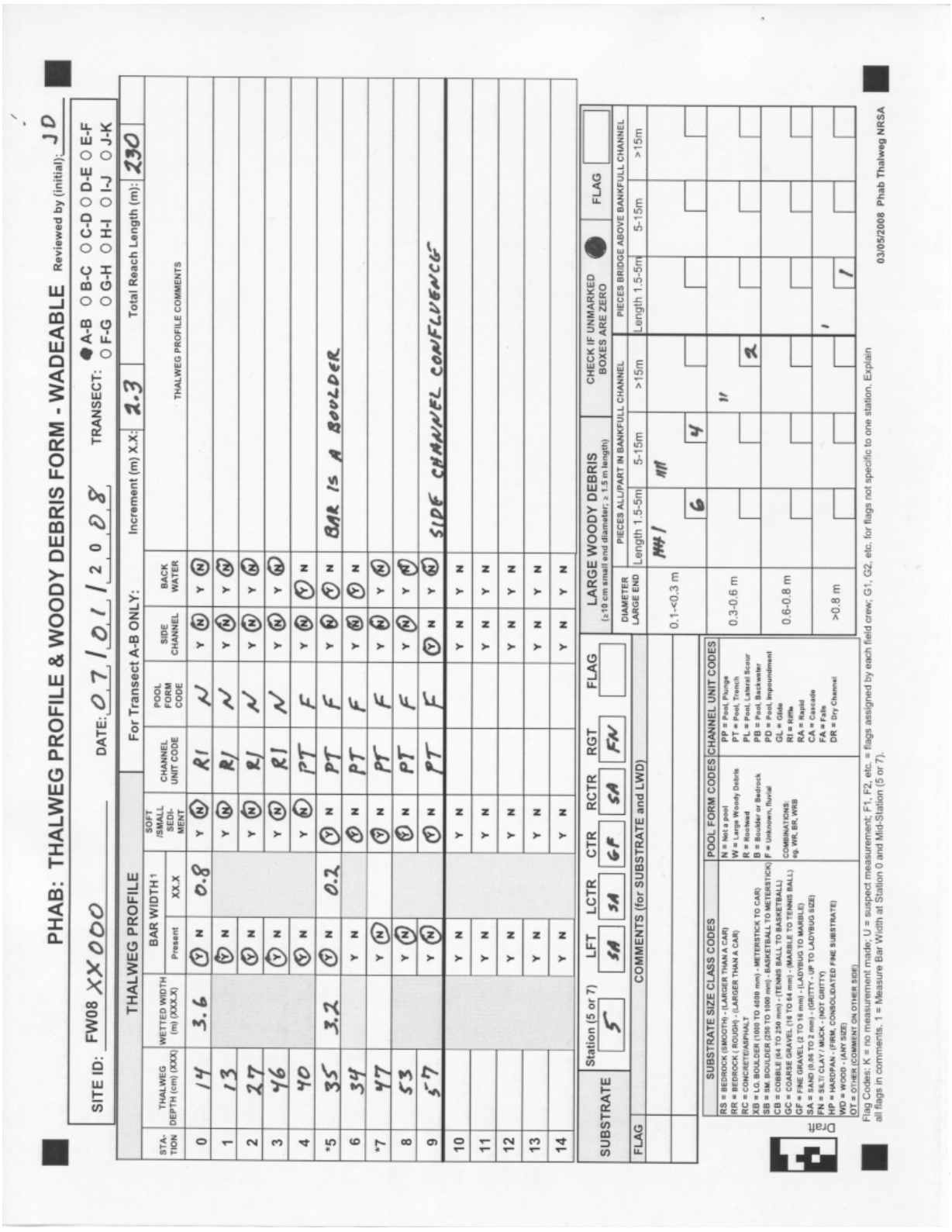
7. At the point where the thalweg depth is determined, observe if unconsolidated, loose (*soft*) deposits of small diameter (≤16mm) sediments are present directly beneath your ruler, rod, or pole.

Soft/ small sediments are defined here as fine gravel, sand, silt, clay or muck readily apparent by "feeling" the bottom with the rod. Record presence or absence in the *SOFT/SMALL SEDIMENT* field on the field data form. *Note: A thin coating of fine sediment or silty algae coating the surface of cobbles should not be considered soft/small sediment. However, fine sediment coatings should be identified in the comments section of the field form when determining substrate size and type*.

8. Determine the channel unit code and pool forming element codes for the station. Record these on the field data form using the standard codes provided. For dry and intermittent streams, where no water is in the channel, record habitat type as dry channel (*DR*).
9. If the station cross-section intersects a mid-channel bar, indicate the presence of the bar in the *BAR WIDTH* field on the field data form.
10. Record the presence or absence of a side channel at the station's cross-section in the *SIDE CHANNEL* field on the field data form.

    Record the presence or absence of quiescent off-channel aquatic habitats, including sloughs, alcoves and backwater pools in the *BACKWATER* column of the field form.
11. Proceed upstream to the next station, and repeat Steps 2 through 11.
12. Repeat Steps 2 through 12 until you reach the next transect. At this point complete Channel/ Riparian measurements at the new transect (Section 6.2.5). Then prepare a new Thalweg Profile and Woody Debris Form and repeat Steps 2 through 12 for each of the reach segments, until you reach the upstream end of the sampling reach (transect *K*). At transect *K*, you will have completed 10 copies of the Thalweg Profile and Woody Debris Form, one for each segment (*A* to *B*, *B* to *C*, etc.).

Measure thalweg depths at **all** stations. Missing depths at the end of the reach (e.g., due to the stream flowing into or out of a culvert or under a large pile of debris) can be tolerated, but those in the middle of the reach are more difficult to deal with. Flag any missing measurements using a *K* code and explain the reason in the comments section of the field data form. At points where a direct depth measurement cannot be made, make your best estimate of the depth, record it on the field form, and flag the value using a *U* code (nonstandard measurement), explaining that it is an estimated value in the comments section of the field data form. *Where the thalweg points are too deep for wading*, measure the depth by extending the surveyor's rod at an angle to reach the thalweg point. Record the water level on the rod, and the rod angle, as determined using the external scale on the clinometer (vertical = 90°). In analyzing these data we calculate the thalweg depth as the length of the rod (or string) under water multiplied by the trigonometric *sine* of the rod angle. (For example, if 3 meters of the rod are under water when the rod held at 30 degrees (*sine*=0.5), the actual thalweg depth is 1.5 meters.) These calculations are done after field forms are returned for data analysis. On the field form, crews are required only to record the wetted length of the rod under the water, a *U* code in the flag field (to indicate a nonstandard technique), and a comment to the right saying "*depth taken at an angle of xx degrees*." If a direct measurement of the thalweg depth is not possible, make the best estimate you can of the depth, record it, and use a *U* flag and a comment to note it is an estimated value.



PHAB: THALWEG PROFILE & WOODY DEBRIS FORM - WADEABLE

SITE ID: FW08 XX000 DATE: 07/01/2008 TRANSECT: ● A-B ○ B-C ○ C-D ○ D-E ○ E-F ○ F-G ○ G-H ○ H-I ○ I-J ○ J-K

Reviewed by (initial): JD

THALWEG PROFILE — For Transect A-B ONLY: Increment (m) X.X: 2.3 Total Reach Length (m): 230

STATION	THALWEG DEPTH (cm) (XXX)	WETTED WIDTH (m) (XXX.X)	BAR WIDTH[1] Present	BAR WIDTH[1] XX.X	SOFT /SMALL SEDIMENT	CHANNEL UNIT CODE	POOL FORM CODE	SIDE CHANNEL	BACK WATER	THALWEG PROFILE COMMENTS
0	14	3.6	Y (N)	0.8	Y (N)	RI	N	Y (N)	Y (N)	
1	13		Y (N)		Y (N)	RI	N	Y (N)	Y (N)	
2	27		Y (N)		Y (N)	RI	N	Y (N)	Y (N)	
3	46		Y (N)		Y (N)	RI	N	Y (N)	Y (N)	
4	40		Y (N)		Y (N)	PT	F	Y (N)	(Y) N	
*5	35	3.2	(Y) N	0.2	(Y) N	PT	F	Y (N)	(Y) N	BAR IS A BOULDER
6	34		(Y) N		(Y) N	PT	F	Y (N)	(Y) N	
*7	47		Y (N)		(Y) N	PT	F	Y (N)	Y (N)	
8	53		Y (N)		(Y) N	PT	F	Y (N)	Y (N)	
9	57		Y (N)		(Y) N	PT	F	(Y) N	Y (N)	SIDE CHANNEL CONFLUENCE
10			Y N		Y N			Y N	Y N	
11			Y N		Y N			Y N	Y N	
12			Y N		Y N			Y N	Y N	
13			Y N		Y N			Y N	Y N	
14			Y N		Y N			Y N	Y N	

SUBSTRATE	Station (5 or 7)	LFT	LCTR	CTR	RCTR	RGT	FLAG
	5	SA	SA	GF	SA	FN	

FLAG — COMMENTS (for SUBSTRATE and LWD)

LARGE WOODY DEBRIS (≥10 cm small end diameter; ≥ 1.5 m length) — CHECK IF UNMARKED BOXES ARE ZERO — FLAG

DIAMETER LARGE END	PIECES ALL/PART IN BANKFULL CHANNEL Length 1.5-5m	5-15m	>15m	PIECES BRIDGE ABOVE BANKFULL CHANNEL Length 1.5-5m	5-15m	>15m
0.1-<0.3 m	6	IIII				
0.3-0.6 m			11			
0.6-0.8 m			2			
>0.8 m		1		1		

SUBSTRATE SIZE CLASS CODES
RS = BEDROCK (SMOOTH) - (LARGER THAN A CAR)
RR = BEDROCK (ROUGH) - (LARGER THAN A CAR)
RC = CONCRETE/ASPHALT
XB = LG. BOULDER (1000 TO 4000 mm) - METERSTICK TO CAR)
SB = SM. BOULDER (250 TO 1000 mm) - BASKETBALL TO METERSTICK)
CB = COBBLE (64 TO 250 mm) - (TENNIS BALL TO BASKETBALL)
GC = COARSE GRAVEL (16 TO 64 mm) - (MARBLE TO TENNIS BALL)
GF = FINE GRAVEL (2 TO 16 mm) - (LADYBUG TO MARBLE)
SA = SAND (0.06 TO 2 mm) - (GRITTY - UP TO LADYBUG SIZE)
FN = SILT/ CLAY / MUCK - (NOT GRITTY)
HP = HARDPAN - (FIRM, CONSOLIDATED FINE SUBSTRATE)
WD = WOOD - (ANY SIZE)
OT = OTHER (COMMENT ON OTHER SIDE)

POOL FORM CODES
N = Not a pool
W = Large Woody Debris
R = Riverbed
B = Boulder or Bedrock
F = Unknown, fluvial
COMBINATIONS:
eg. WR, BR, WRB

CHANNEL UNIT CODES
PP = Pool, Plunge
PT = Pool, Trench
PL = Pool, Lateral Scour
PB = Pool, Backwater
PD = Pool, Impoundment
GL = Glide
RI = Riffle
RA = Rapid
CA = Cascade
FA = Falls
DR = Dry Channel

Draft

Flag Codes: K = no measurement made; U = suspect measurement; F1, F2, etc. = flags assigned by each field crew; G1, G2, etc. for flags not specific to one station. Explain all flags in comments. 1 = Measure Bar Width at Station 0 and Mid-Station (5 or 7).

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Figure 6.2-2. Thalweg Profile and Woody Debris Form.

At every thalweg station, determine by sight or feel whether deposits of *soft/small sediments* are present on the channel bottom. These particles are defined as substrate equal to or smaller than fine gravel (≤ 16 mm diameter). These soft/small sediments are **different** from *Fines* described when determining the substrate particle sizes at the cross-section transects (Section 6.2.5.2). If the channel bottom is not visible, determine if soft/small sediment deposits are readily obvious by feeling the bottom with your boot, the surveyor's rod, or a calibrated rod or pole.

Measure wetted width at each transect (station *0*), and midway between transects (station *5* for larger streams having 100 measurement points, or station *7* for smaller streams having 150 measurement points). The wetted width boundary is the point at which substrate particles are no longer surrounded by free water. Estimate substrate size for five particles evenly spaced across each midway cross-section using procedures described for substrate at regular cross-sections (Section 6.2.5.2), but at the supplemental cross-sections, only the size class (not distance and depth) data are recorded.

While recording the width and depth measurements and the presence of soft/small sediments, the second person evaluates and records the habitat class and the pool forming element (Table 6.2-3) applicable to each of the 100 (or 150) measurement points along the length of the reach. Make channel unit scale habitat classifications at the thalweg of the cross-section. The habitat unit itself must meet a minimum size criteria in addition to the qualitative criteria listed in Table 6.2-3. Before being considered large enough to be identified as a channel-unit scale habitat feature, the unit should be at least as long as the channel is wide. For instance, if there is a small deep (pool-like) area at the thalweg within a large riffle area, do not record it as a pool unless it occupies an area about as wide or long as the channel is wide. If a backwater pool **dominates the channel**, record *PB* as the dominant habitat unit class. If the backwater is a pool that **does not dominate** the main channel, or if it is an **off-channel** alcove or slough (large enough to offer refuge to small fishes), circle *Y* to indicate presence of a backwater in the *BACKWATER* column of the field form, but classify the main channel habitat unit type according to characteristics of the main channel. *Sloughs* are backwater areas having marsh-like characteristics such as vegetation, and *alcoves* (or *side pools*) are deeper areas off the main channel that are typically wide and shallow (Helm 1985, Bain and Stevenson 1999). When trying to identify the pool forming element for a particular pool, remember that most pools are formed at high flows, so you may need to look for elements that are dry at baseflow, but still within the bankfull channel (e.g., boulders or large woody debris).

Table 6.2-3. Channel unit and pool forming element categories

Channel Unit Habitat Classes [a]

Class (Code)	Description
Pools: Still water, low velocity, a smooth, glassy surface, usually deep compared to other parts of the channel:	
Plunge Pool (*PP*)	Pool at base of plunging cascade or falls
Trench Pool (*PT*)	Pool-like trench in the center of the stream
Lateral Scour Pool (*PL*)	Pool scoured along a bank
Backwater Pool (*PB*)	Pool separated from main flow off the side of the channel (large enough to offer refuge to small fishes). Includes sloughs (backwater with marsh characteristics such as vegetation), and alcoves (a deeper area off a wide and shallow main channel)
Impoundment Pool(*PD*)	Pool formed by impoundment above dam or constriction.
Pool (*P*)	Pool (unspecified type)
Glide (*GL*)	Water moving slowly, with a *smooth, unbroken surface*. Low turbulence.
Riffle (*RI*)	Water moving, with *small ripples, waves and eddies* -- waves not breaking, *surface tension not broken*. Sound: babbling, gurgling.
Rapid (*RA*)	Water movement rapid and turbulent, surface with *intermittent whitewater* with breaking waves. Sound: continuous rushing, but not as loud as cascade.
Cascade (*CA*)	Water movement rapid and very turbulent over steep channel bottom. Much of the water surface is broken in *short, irregular plunges, mostly whitewater.* Sound: roaring.
Falls (*FA*)	*Free falling water* over a vertical or near vertical drop into plunge, water turbulent and white over high falls. Sound: from splash to roar.
Dry Channel (*DR*)	No water in the channel, or flow is submerged under the substrate (*hyporheic flow*).

[a] Note that in order for a channel habitat unit to be distinguished, it must be at least as wide or long as the channel is wide (except for off channel backwater pools, which are noted as present regardless of size).

Categories of Pool-forming Elements [b]

Code	Category
N	Not Applicable, Habitat Unit is not a pool
W	Large Woody Debris.
R	Rootwad
B	Boulder or Bedrock
F	Unknown cause (unseen fluvial processes)
WR, RW, RBW	Combinations
OT	Other (describe in the comments section of field form)

[b] In determining the pool forming element, remember that most pools are formed at high flows, so you may need to look at features, such as large woody debris, that are dry at baseflow, but still within the bankfull channel.

6.2.4.2 Large Woody Debris Tally

Large Woody Debris is defined here as woody material with a small end diameter of at least 10 cm (4 in.) **and** a length of at least 1.5 m (5 ft.). The procedure for tallying LWD is presented in Table 6.2-4. The tally includes all pieces of LWD that are at least partially in the baseflow channel (Zone 1), in the *bankfull channel* (Zone 2, flood channel up to bankfull stage), or spanning above the bankfull channel (Zone 3), as shown in Figure 6.2-3. The *bankfull channel* is defined as the channel that is filled by moderate sized flood events that typically recur every one to two years. LWD in or above the bankfull channel is tallied over the entire length of the reach, including the area between the channel cross-section transects. Pieces of LWD that are not at least partially within Zones 1, 2, or 3 are not tallied.

Table 6.2-4. Procedure for tallying large woody debris

Note: Tally pieces of large woody debris (LWD) within each segment of stream while the thalweg profile is being determined. Include all pieces in the tally whose large end is found within the segment.

1. Scan the stream segment between the two cross-section transects where thalweg profile measurements are being made.
2. Tally all LWD pieces within the segment that are at least partially within the bankfull channel. Determine if a piece is LWD (*small end diameter ≥10 cm [4 in.], **and** length ≥1.5 m [5 ft.]*)
3. For each piece of LWD, determine the class based on the *diameter of the **large end*** (0.1 m to < 0.3 m, 0.3 m to <0.6 m, 0.6 m to <0.8 m, or >0.8 m), **and** the class based on the ***length** of the piece* (1.5m to <5.0m, 5m to <15m, or >15m).
   - If the piece is not cylindrical, visually estimate what the diameter would be for a piece of wood with circular cross-section that would have the same volume.
   - When estimating length, include only the part of the LWD piece that has a diameter >10 cm (4 in)
4. Place a tally mark in the appropriate diameter × length class tally box in the *PIECES ALL/PART IN BANKFULL CHANNEL* section of the Thalweg Profile and Woody Debris Form.
5. Tally all LWD pieces within the segment that are not actually within the bankfull channel, but are at least partially spanning (bridging) the bankfull channel. For each piece, determine the class based on the diameter of the **large end** (0.1 m to < 0.3 m, 0.3 m to <0.6 m, 0.6 m to <0.8 m, or >0.8 m), **and** the class based on the **length** of the piece (1.5 m to <5.0 m, 5 m to <15 m, or >15 m).
6. Place a tally mark for each piece in the appropriate diameter × length class tally box in the *PIECES BRIDGE ABOVE BANKFULL CHANNEL* section of the Thalweg Profile and Woody Debris Form.
7. After all pieces within the segment have been tallied, write the total number of pieces for each diameter × length class in the small box at the lower right-hand corner of each tally box.
8. Repeat Steps 1 through 7 for the next stream segment, using a new Thalweg Profile and Woody Debris Form.

6.2.5 Channel and Riparian Measurements at Cross-Section Transects

6.2.5.1 Slope and Bearing

Measure bearing by *sighting* between transects (e.g., transect *B* and *A*, *C* and *B*, etc.) as shown in Figure 6.2-4. To measure the bearing between adjacent transects, follow the procedure presented in Table 6.2-5. Record bearing data on the Slope and Bearing Form as shown in Figure 6.2-5.

Slope is typically measured by two people, one holding a surveyor's rod and the second sighting through the surveyor's level. Be sure that the person is standing (or holding the marked pole) at the water's edge holding the rod at the surface of the water. The intent is to get a measure of the *water surface* slope, which may not necessarily be the same as the bottom slope. The surveyor's level is leveled according to the manufacturer's recommendations which is generally to adjust the three screw leveling feet until the bubble is centered. Level is checked in all planes to be measured. If the level does not "self level" in all measured planes the user should check the instruction manual for suggested options. Elevation readings are made at each transect and the difference between each elevation reading is recorded as the change in elevation. NOTE: Multiple transect elevations can often be made for each setup of the level, but every time the transit is moved requires re-measuring the last transect elevation from the last setup. You cannot use elevations from previous setups because the relative height of the transit has changed.

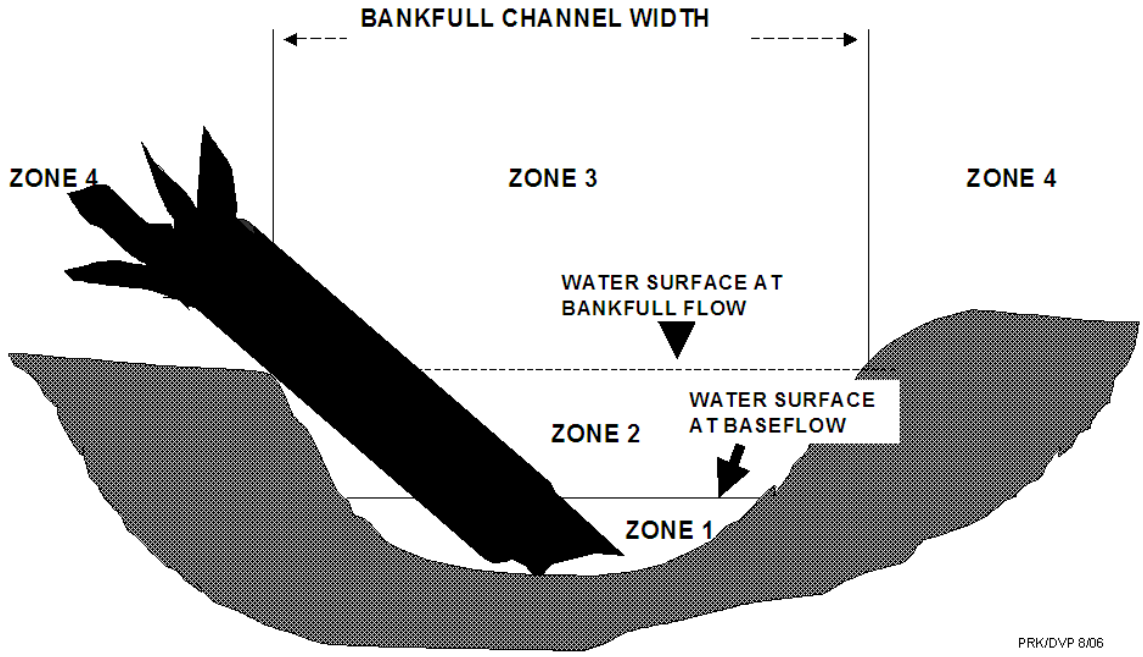
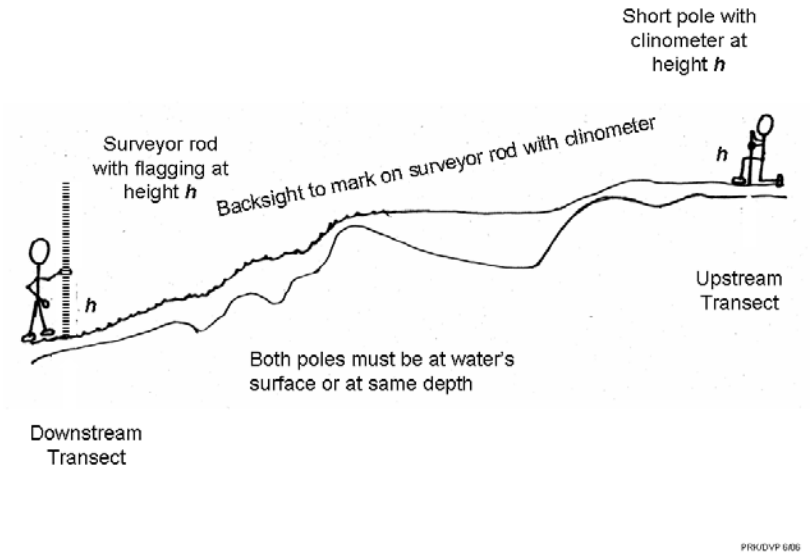


Figure 6.2-3. Large woody debris influence zones (modified from Robison and Beschta, 1990).

To calculate sinuosity from bearing measurements, it does not matter whether or not you adjust your compass bearings for magnetic declination, but it is important that you are **consistent** in the use of magnetic or true bearings throughout all the measurements you make on a given reach. Note in the comments section of the Slope and Bearing Form which type of bearings you are taking, so the measurements can be used to describe reach aspect. Also, guard against recording *reciprocal bearings* (erroneous bearings 180 degrees from what they should be). The best way to do this is to know where the primary (cardinal) directions are in the field: (north [0 degrees], east [90 degrees], south [180 degrees], and west [270 degrees]), and insure that your bearings "make sense."

As stated earlier, it may be necessary to set up intermediate (supplemental) slope and bearing points between a pair of cross-section transects if you do not have direct line-of-sight along (and within) the channel between stations (see Figure 6.2-4). This can happen if brush is too heavy, or if there are sharp slope breaks or tight meander bends. *If you would have to sight across land to measure slope or bearing between two transects, then you need to make one or more supplemental measurements* (i.e., do not "short-circuit" a meander bend). Mark these supplemental locations with a different color of plastic flagging than used for the cross-section transects to avoid confusion. Record these supplemental slope and bearing measurements, along with the proportion of the stream segment between transects included in each supplemental measurement, in the appropriate sections of the Slope and Bearing Form (Figure 6-5). Note that the main slope and bearing observations are always downstream of supplemental observations (i.e., from or to the downstream transect). Similarly, first supplemental observations are always downstream of second supplemental observations.



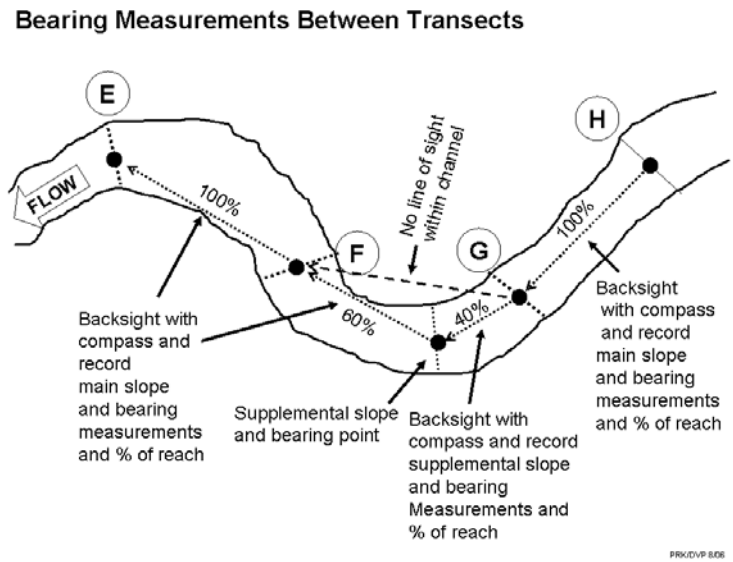


Figure 6.2-4. Channel slope and bearing measurements.

Because of ease of use, portability, and cost, hand-held clinometers were previously used to determine slope. In this instance, the field crews will have access to more sophisticated instrumentation (e.g., surveyor's level), and have field personnel who are experienced in the use of these instruments. The Slope and Bearing Form (Figure 6-5) is designed to allow for different methods and/or different units of measuring slope. Mark the appropriate method circle (instead of *CL*; method codes are identified in Tables 6.2-5 and 6.2-6), and mark the *CM* circle (instead of the *%* circle) if the method or instrument measures the change in elevation rather than the percent slope.

Table 6.2-5. Procedure for obtaining slope and bearing data

1. Determine a location at transect K to hold a surveyor's rod that will be visible from a point between transect J and transect K:
   a) Set up the instrument at a point approximately halfway between points J and K and where a clear line of sight is possible.
   b) Position the staff at point K, holding the bottom of the staff at the water level and the staff as vertical as possible and the numbers facing the instrument.
   c) Site the staff and record the reading to the nearest centimeter.
   d) Move the staff to point J and gently swivel the instrument to face the next reading. Hold the staff as before, vertically, with the bottom at the water level and the numbers facing the instrument.
   e) Site the staff and record the reading to the nearest centimeter.
   f) Repeat measurements between each transect.
   g) The difference in the readings is the height difference or gradient.

Note: In small streams with a clear line of site it may be possible to set the instrument up once and make readings to several transects from a single set up. Simply record the readings for each transect and do not skip transects.

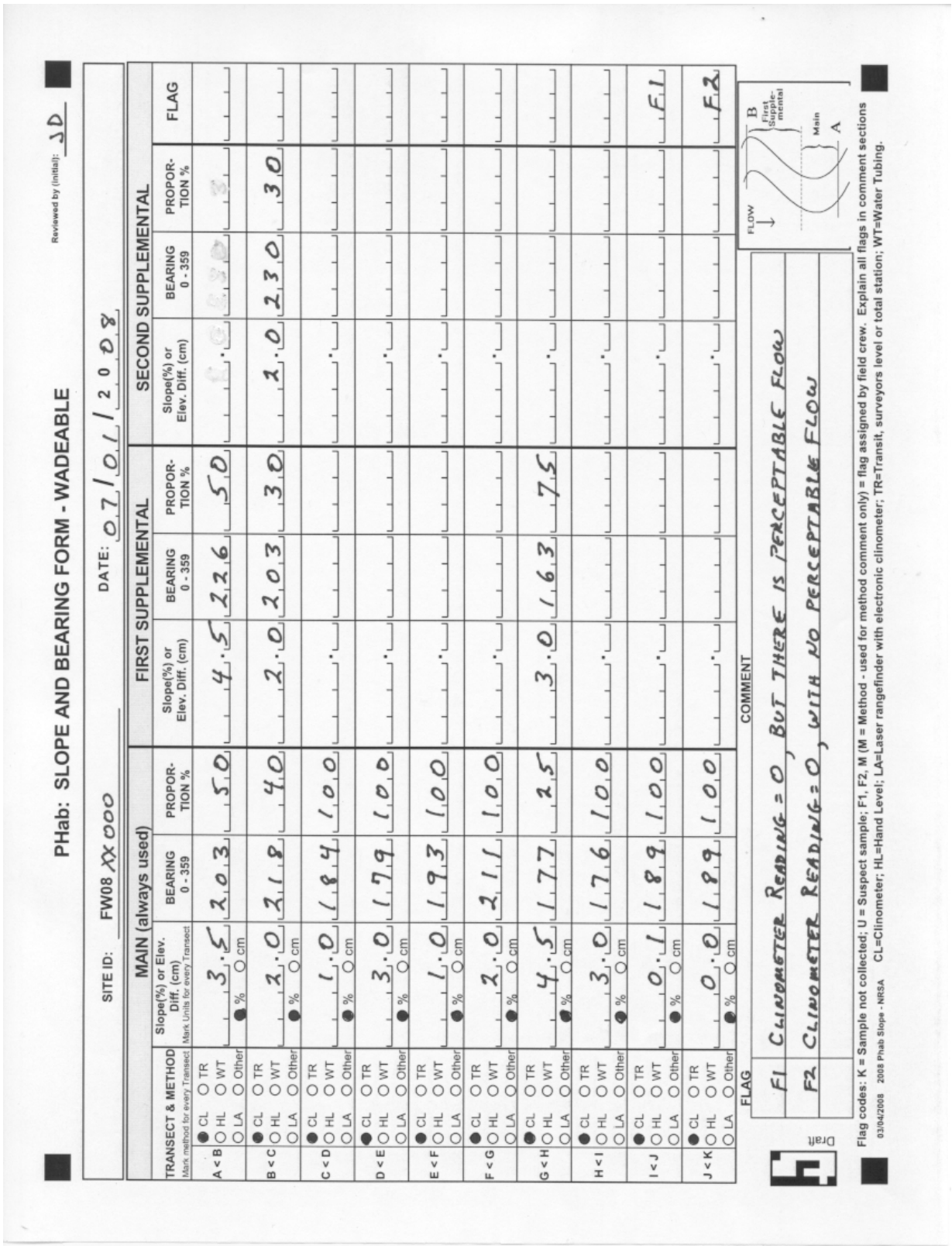
- If you are backsighting from a supplemental point, record the bearing in the appropriate *SUPPLEMENTAL* section of the Slope and Bearing Form.

2. Proceed to the next cross-section transect (or supplementary point), and repeat Steps a - g above.

Instrument Setup:

   a) Extend the tripod legs to approximately eye level and set the legs firmly into the ground; adjust the legs so that they form a regular triangle and are firmly set with no wobble. Adjust the legs so that the base plate is approximately level.
   b) Hold the instrument on the tripod and start the centering screw. Ensure the adjustable feet are roughly evenly adjusted. While the centering screw is still loose slide the instrument on the base plate until the bubble is approximately centered in the circular level. Tighten the centering screw.
   c) Adjust the leveling foot screws until the bubble is exactly level in the center circle.
   d) Self Leveling instruments can now be swiveled gently on the base plate and maintain level as long as the tripod remains steady.
   e) Adjust focus, brightness and parallax according to manufactures specifications.
   f) The instrument is ready to make measurements.

[a] Method codes are: *CL*=clinometer, *TR*=transit, *HL*=hand level, *WT*=Water tube, *LA*=laser level, *OTHER*=method not listed (describe in comments section of form).



PHab: SLOPE AND BEARING FORM - WADEABLE

Reviewed by (initial): JD

SITE ID: FW08XX000 DATE: 07/01/2008

TRANSECT & METHOD (Mark method for every Transect)	MAIN (always used) Slope(%) or Elev. Diff. (cm) (Mark Units for every Transect)	MAIN (always used) BEARING 0 - 359	MAIN (always used) PROPOR-TION %	FIRST SUPPLEMENTAL Slope(%) or Elev. Diff. (cm)	FIRST SUPPLEMENTAL BEARING 0 - 359	FIRST SUPPLEMENTAL PROPOR-TION %	SECOND SUPPLEMENTAL Slope(%) or Elev. Diff. (cm)	SECOND SUPPLEMENTAL BEARING 0 - 359	SECOND SUPPLEMENTAL PROPOR-TION %	FLAG
A < B ● CL ○ HL ○ LA ○ TR ○ WT ○ Other	3.5 ● % ○ cm	203	50	4.5	226	50				
B < C ● CL ○ HL ○ LA ○ TR ○ WT ○ Other	2.0 ● % ○ cm	218	40	2.0	203	30	2.0	230	30	
C < D ● CL ○ HL ○ LA ○ TR ○ WT ○ Other	1.0 ● % ○ cm	184	100							
D < E ● CL ○ HL ○ LA ○ TR ○ WT ○ Other	3.0 ● % ○ cm	179	100							
E < F ● CL ○ HL ○ LA ○ TR ○ WT ○ Other	1.0 ● % ○ cm	193	100							
F < G ● CL ○ HL ○ LA ○ TR ○ WT ○ Other	2.0 ● % ○ cm	211	100							
G < H ● CL ○ HL ○ LA ○ TR ○ WT ○ Other	4.5 ● % ○ cm	177	25	3.0	163	75				
H < I ● CL ○ HL ○ LA ○ TR ○ WT ○ Other	3.0 ● % ○ cm	176	100							
I < J ● CL ○ HL ○ LA ○ TR ○ WT ○ Other	0.1 ● % ○ cm	189	100							F1
J < K ● CL ○ HL ○ LA ○ TR ○ WT ○ Other	0.0 ● % ○ cm	189	100							F2

COMMENT

FLAG

F1 CLINOMETER READING = 0, BUT THERE IS PERCEPTABLE FLOW

F2 CLINOMETER READING = 0, WITH NO PERCEPTABLE FLOW

Draft

Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, M (M = Method - used for method comment only) = flag assigned by field crew. Explain all flags in comment sections

03/04/2008 2008 Phab Slope - NRSA CL=Clinometer; HL=Hand Level; LA=Laser rangefinder with electronic clinometer; TR=Transit, surveyors level or total station; WT=Water Tubing.

Figure 6.2-5. Slope and Bearing Form.

Table 6.2-6. Modified procedure for obtaining slope and bearing data

Use this procedure if you are starting at the **upstream transec**t (*K*), after completing the thalweg profile and other cross-section measurements at transects *A* through *K*.

1. Stand in the center of the channel at the upstream cross-section transect. Determine if you can see the center of the channel at the next cross-section transect downstream without sighting across land (i.e., do not "short-circuit" a meander bend). If not, you will have to take supplementary slope and bearing measurements.

   Mark a surveyor's rod and a calibrated rod (or meter ruler) at the same height. If a shorter pole or ruler is used, measure the height from the ground to the opening of the clinometer when it is resting on top.

2. Have one person take the marked surveyor's rod to the downstream transect. Hold the rod vertical with the bottom at the same level as the water surface. If no suitable location is available at the stream margin, position the rod in the water and note the depth.

   - If you have determined in Step 1 that supplemental measurements are required for this segment, walk downstream to the furthest point where you can stand in the center of the channel and still see the center of the channel at the upstream cross-section transect . Remember that your line of sight cannot "cross land." Mark this location with a different color flagging than that marking the cross-section transects.

3. Place the base of the calibrated rod at the level as the surveyor's rod (either at the water surface or at the same depth in the water).

4. Place the clinometer on the calibrated rod at the height determined in Step 2. With the clinometer, sight back downstream to the flagged height on the surveyor's rod at the downstream transect (or at the supplementary point).

   - If you are sighting to the next downstream transect, read and record the **percent** slope in the *MAIN* section on the Slope and Bearing Form for the **downstream transect** (e.g., *J* < *K*), *which is at the **bottom** of the form* (i.e., you are completing the form in reverse order). Record the *PROPORTION* as 100%.

   - If you are backsighting from a supplemental point, record the slope (%) and proportion (%) of the stream segment that is included in the measurement in the appropriate *SUPPLEMENTAL* section of the Slope and Bearing Form. The last sighting to a downstream transect (from either the upstream transect or the nearest upstream supplemental point) is always recorded as the *MAIN* reading.

5. Stand in the middle of the channel at upstream transect (or at a supplemental point), and sight with your compass to the middle of the channel at the downstream transect (or at a supplemental point). Record the bearing (degrees) in the same section of the Slope and Bearing form (Supplemental or Main) as you recorded the slope in Step 6.

6. Proceed to the next cross-section transect (or to a supplementary point), and repeat Steps 3 through 7 above.

6.2.5.2 Substrate Size and Channel Dimensions

Substrate size and embeddedness are evaluated at 5 points at each of the 11 transects (refer to Figure 6.2-6). Substrate size is also evaluated at 10 additional cross-sections located midway between each of the 11 regular transects (*A*-*K*). In the process of measuring substrate particle sizes at each channel cross-section, the wetted width of the channel and the water depth at each substrate sample point are measured (at the 10 midway cross-sections, only substrate size and wetted width are recorded). If the wetted channel is split by a mid-channel bar (see Section 6.2.4.1), the five substrate points are centered between the wetted width boundaries regardless of the mid-channel bar in between. Consequently, substrate particles selected in some cross-sections may be "high and dry". *For cross-sections that are entirely dry, make measurements across the* ***unvegetated portion*** *of the channel.*

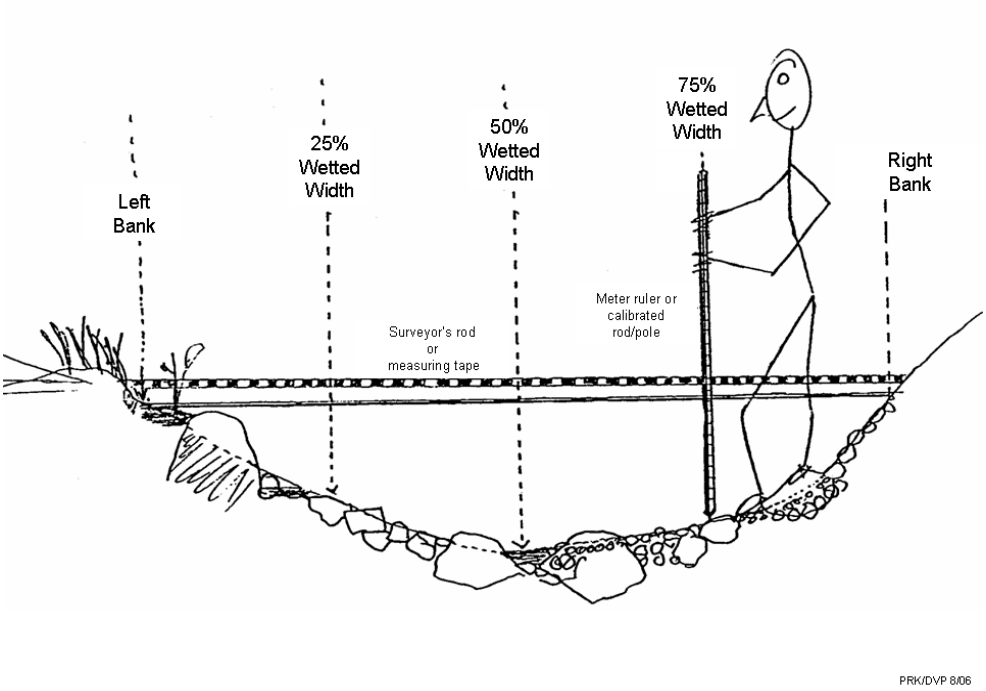


Figure 6.2-6. Substrate sampling cross-section.

The substrate sampling points along the cross-section are located at 0, 25, 50, 75, and 100 percent of the measured wetted width, with the first and last points located at the water's edge just within the left and right banks. The procedure for obtaining substrate measurements is described in Table 6.2-7 (including all particle size classifications). Record these measurements on the Channel/Riparian Cross-section side of the field form, as shown in Figure 6.2-7. For the supplemental cross-sections midway between regular transects,

Table 6.2-7. Substrate measurement procedure

1. Fill in the header information on page 1 of a Channel/Riparian Cross-section Form. Indicate the cross-section transect. At the transect, extend the surveyor's rod or metric tape across the channel perpendicular to the flow, with the "zero" end at the left bank (facing downstream).

NOTE: If a side channel is present, and contains 16 - 49% of the total flow, establish a secondary cross-section transect. Use a separate field data form to record data for the side channel, designating it as a secondary transect by marking both the X-TRA SIDE CHANNEL circle and the associated primary transect letter (e.g., XA, XB, etc.). Collect all channel and riparian cross-section measurements from the side channel.

2. Divide the wetted channel width channel by 4 to locate substrate measurement points on the cross-section. In the *DISTLB* fields of the form, record the distances corresponding to 0% (*LFT*), 25% (*LCTR*), 50% (*CTR*), 75% (*RCTR*), and 100% (*RGT*) of the measured wetted width. Record these distances at Transects *A-K*, but just the wetted width at midway cross-sections.

3. Place your sharp-ended meter stick or calibrated pole at the *LFT* location (0 m). Measure the depth and record it on the field data form. (Cross-section depths are measured only at regular transects *A-K*, not at the 10 midway cross-sections).

   - Depth entries at the left and right banks may be 0 (zero) if the banks are gradual.
   - If the bank is nearly vertical, let the base of the measuring stick fall to the bottom (i.e., the depth at the bank will be > 0 cm), rather than holding it suspended at the water surface.

4. Pick up the substrate particle that is at the base of the meter stick (unless it is bedrock or boulder), and visually *estimate its particle size*, according to the following table. Classify the particle according to its **median diameter** (the middle dimension of its length, width, and depth). Record the size class code on the field data form. (Cross-section side of form for transects *A-K*; special entry boxes on Thalweg Profile side of form for midway cross-sections.)

Code	Size Class	Size Range (mm)	Description
RS	Bedrock (Smooth)	>4000	Smooth surface rock bigger than a car
RR	Bedrock (Rough)	>4000	Rough surface rock bigger than a car
HP	Hardpan	>4000	Firm, consolidated fine substrate
LB	Boulders (large)	>1000 to 4000	Yard/meter stick to car size
SB	Boulders (small)	>250 to 1000	Basketball to yard/meter stick size
CB	Cobbles	>64 to 250	Tennis ball to basketball size
GC	Gravel (Coarse)	>16 to 64	Marble to tennis ball size
GF	Gravel (Fine)	> 2 to 16	Ladybug to marble size
SA	Sand	>0.06 to 2	Smaller than ladybug size - gritty between fingers
FN	Fines	≤0.06	Silt Clay Muck (not gritty between fingers)
WD	Wood	Regardless of Size	Wood & other organic particles
RC	Concrete	Regardless of size	Record size class in comment field
OT	Other	Regardless of Size	Metal, tires, car bodies etc. (describe in comments)

5. Evaluate substrate embeddedness as follows at each transects. For particles larger than sand, examine the surface for stains, markings, and algae. Estimate the average % embeddedness of particles in the

10 cm circle around the measuring rod. Record this value on the field data form. For sand and smaller particles, you will not be able to pick up an individual particle, but a "pinch" of fine particles between your fingers. Determine and record the dominant size of particles in the "pinch." By definition, sand and fines are embedded *100%*; bedrock and hardpan are embedded *0%*.

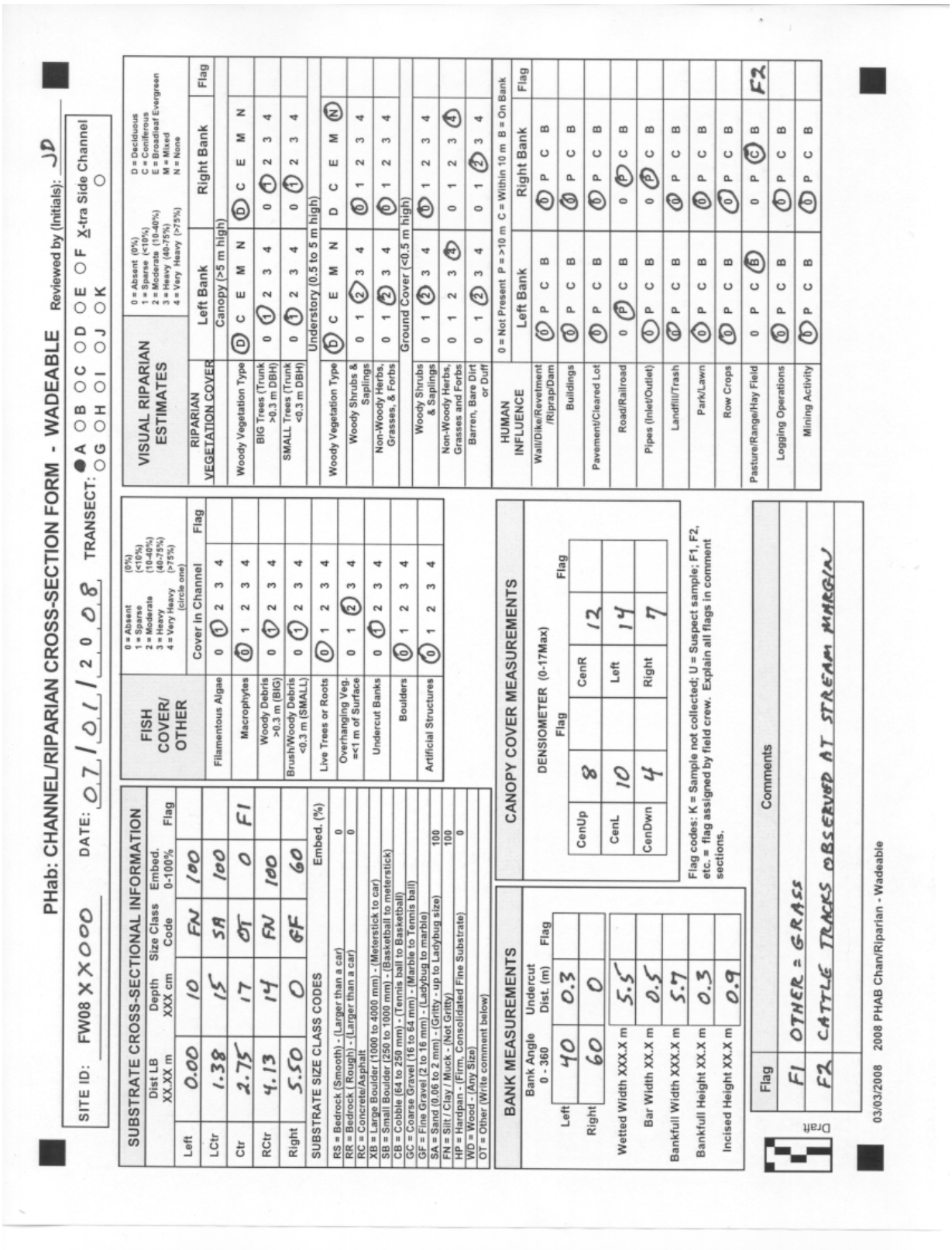
6. Move to the next location on the transect, and repeat Steps 4 - 6 at each location. Repeat Steps 1 - 6 at each transect, including any additional side channel transects established if islands are present.

record substrate size and wetted width data on the thalweg profile side of the field form. To minimize bias in selecting a substrate particle for size classification, it is important to concentrate on correct placement of the measuring stick along the cross-section, and to select the particle right at the bottom of the stick (not, for example, a more noticeable large particle that is just to the side of the stick). Classify the particle into one of the size classes listed on the field data form (Figure 6.2-7) based on the middle dimension of its length, width, and depth. This *median dimension* determines the sieve size through which the particle can pass. When you record the size class as *Other*, assign an *Fn* flag on the field data form and describe the substrate type in the comments section of the field form, as shown in Figure 6.2-7.

At substrate sampling locations on the 11 regular transects (*A-K*), examine particles larger than sand for surface stains, markings, and algal coatings to estimate embeddedness of all particles in the 10 cm diameter circle around the substrate sampling point. Embeddedness is the fraction of a particle's volume that is surrounded by (embedded in) sand or finer sediments on the stream bottom. By definition, record the embeddedness of sand and fines (silt, clay, and muck) as *100 percent*, and record the embeddedness of hardpan and bedrock as *0 percent*.

6.2.5.3 Bank Characteristics

The procedure for obtaining bank and channel dimension measurements is presented in Table 6.2-8. Data are recorded in the *BANK MEASUREMENTS* section of the Channel/Riparian Cross-section Form as shown in Figure 6.2-7. Bank angle and bank undercut distance are determined on the left and right banks at each cross-section transect. Figure 6.2-8 illustrates how bank angle is determined for several different situations. The scale at which bank angle is characterized is approximately 0.5 m. A short (approx. 1-m long) pole is used to determine bank angle. The angle is determined based on the pole resting on the ground for about 0.5 m. Other features include the wetted width of the channel (as determined in Section 6.2.5.2), the width of exposed mid-channel bars of gravel or sand, estimated incision height, and the estimated height and width of the channel at bankfull stage as described in Table 6-8. *Bankfull height* and *incised height* are both measured relative to the present water surface (i.e. the level of the wetted edge of the stream). This is done by placing the base of the small measuring rod at the bankfull elevation and sighting back to the survey rod placed at the water's edge using the clinometer as a level (i.e., positioned so the slope reading is *0%*.). The height of the clinometer above the base of the smaller rod is subtracted from the elevation sighted on the surveyor's rod.



PHab: CHANNEL/RIPARIAN CROSS-SECTION FORM - WADEABLE

SITE ID: FW08XX000 DATE: 07/01/2008 TRANSECT: ● A ○ B ○ C ○ D ○ E ○ F X-tra Side Channel ○ G ○ H ○ I ○ J ○ K

SUBSTRATE CROSS-SECTIONAL INFORMATION

	Dist LB XX.XX m	Depth XXX cm	Size Class Code	Embed. 0-100%	Flag
Left	0.00	10	FN	100	
LCtr	1.38	15	SA	100	
Ctr	2.75	17	OT	0	F1
RCtr	4.13	14	FN	100	
Right	5.50	0	GF	60	

SUBSTRATE SIZE CLASS CODES — Embed. (%)

RS = Bedrock (Smooth) - (Larger than a car) 0
RR = Bedrock (Rough) - (Larger than a car) 0
RC = Concrete/Asphalt
XB = Large Boulder (1000 to 4000 mm) - (Meterstick to car)
SB = Small Boulder (250 to 1000 mm) - (Basketball to meterstick)
CB = Cobble (64 to 250 mm) - (Tennis ball to Basketball)
GC = Coarse Gravel (16 to 64 mm) - (Marble to Tennis ball)
GF = Fine Gravel (2 to 16 mm) - (Ladybug to marble)
SA = Sand (0.06 to 2 mm) - (Gritty - up to Ladybug size) 100
FN = Silt / Clay / Muck - (Not Gritty) 100
HP = Hardpan - (Firm, Consolidated Fine Substrate) 0
WD = Wood - (Any Size)
OT = Other (Write comment below)

FISH COVER/ OTHER

0 = Absent (0%) 1 = Sparse (<10%) 2 = Moderate (10-40%) 3 = Heavy (40-75%) 4 = Very Heavy (>75%) (circle one)

Cover in Channel					Flag
Filamentous Algae	0	(1)	2	3 4	
Macrophytes	(0)	1	2	3 4	
Woody Debris >0.3 m (BIG)	0	(1)	2	3 4	
Brush/Woody Debris <0.3 m (SMALL)	0	(1)	2	3 4	
Live Trees or Roots	(0)	1	2	3 4	
Overhanging Veg. =<1 m of Surface	0	1	(2)	3 4	
Undercut Banks	0	(1)	2	3 4	
Boulders	(0)	1	2	3 4	
Artificial Structures	(0)	1	2	3 4	

VISUAL RIPARIAN ESTIMATES

0 = Absent (0%) 1 = Sparse (<10%) 2 = Moderate (10-40%) 3 = Heavy (40-75%) 4 = Very Heavy (>75%)
D = Deciduous C = Coniferous E = Broadleaf Evergreen M = Mixed N = None

RIPARIAN VEGETATION COVER	Left Bank					Right Bank					Flag
Canopy (>5 m high)											
Woody Vegetation Type	(D)	C	E	M	N	(D)	C	E	M	N	
BIG Trees (Trunk >0.3 m DBH)	0	(1)	2	3	4	0	(1)	2	3	4	
SMALL Trees (Trunk <0.3 m DBH)	0	(1)	2	3	4	0	(1)	2	3	4	
Understory (0.5 to 5 m high)											
Woody Vegetation Type	(D)	C	E	M	N	D	C	E	M	(N)	
Woody Shrubs & Saplings	0	1	(2)	3	4	(0)	1	2	3	4	
Non-Woody Herbs, Grasses, & Forbs	0	1	(2)	3	4	(0)	1	2	3	4	
Ground Cover (<0.5 m high)											
Woody Shrubs & Saplings	0	1	(2)	3	4	(0)	1	2	3	4	
Non-Woody Herbs, Grasses and Forbs	0	1	2	3	(4)	0	1	2	3	(4)	
Barren, Bare Dirt or Duff	0	1	(2)	3	4	0	1	(2)	3	4	

HUMAN INFLUENCE	0 = Not Present P = >10 m C = Within 10 m B = On Bank										
	Left Bank					Right Bank					Flag
Wall/Dike/Revetment /Riprap/Dam	(0)	P	C	B		(0)	P	C	B		
Buildings	(0)	P	C	B		(0)	P	C	B		
Pavement/Cleared Lot	(0)	P	C	B		(0)	P	C	B		
Road/Railroad	0	(P)	C	B		0	(P)	C	B		
Pipes (Inlet/Outlet)	(0)	P	C	B		0	(P)	C	B		
Landfill/Trash	(0)	P	C	B		(0)	P	C	B		
Park/Lawn	(0)	P	C	B		(0)	P	C	B		
Row Crops	(0)	P	C	B		(0)	P	C	B		
Pasture/Range/Hay Field	0	P	C	(B)		0	P	(C)	B		F2
Logging Operations	(0)	P	C	B		(0)	P	C	B		
Mining Activity	(0)	P	C	B		(0)	P	C	B		

BANK MEASUREMENTS

	Bank Angle 0 - 360	Undercut Dist. (m)	Flag
Left	40	0.3	
Right	60	0	

Wetted Width XXX.X m	5.5
Bar Width XX.X m	0.5
Bankfull Width XXX.X m	5.7
Bankfull Height XX.X m	0.3
Incised Height XX.X m	0.9

CANOPY COVER MEASUREMENTS

DENSIOMETER (0-17Max)

		Flag			Flag
CenUp	8		CenR	12	
CenL	10		Left	14	
CenDwn	4		Right	7	

Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, etc. = flag assigned by field crew. Explain all flags in comment sections.

Flag	Comments
F1	OTHER = GRASS
F2	CATTLE TRACKS OBSERVED AT STREAM MARGIN

Draft

03/03/2008 2008 PHAB Chan/Riparian - Wadeable

Figure 6.2-7. Channel/Riparian Cross-section Form.

Table 6.2-8. Procedure for measuring bank characteristics

1. To measure *bank angle*, lay a meter ruler or a short (approx. 1-m long) rod down against the left bank (determined as you face downstream), with one end at the water's edge. At least 0.5 m of the ruler or rod should be *resting comfortably* on the ground to determine bank angle. Lay the clinometer on the rod, and read the bank angle in degrees from the external scale on the clinometer. Record the angle in the field for the left bank in the *BANK MEASUREMENT* section of the Channel/Riparian Cross-section Form.
    - A *vertical bank* is 90°, *overhanging banks* have angles >90° approaching 180°, and more gradually sloped banks have angles <90°. To measure bank angles >90°, turn the clinometer (which only reads 0 to 90°) over and subtract the angle reading from 180°.
    - If there is a large boulder or log present at the transect, measure bank angle at a nearby point where conditions are more representative.
2. If the bank is *undercut*, measure the horizontal distance of the undercutting to the nearest 0.01 m. The undercut distance is the distance from the water's edge out to the point where a vertical plumb line from the bank would hit the water's surface. Record the distance on the field data form. Measure submerged undercuts by thrusting the rod into the undercut and reading the length of the rod that is hidden by the undercutting.
3. Repeat Steps 1 and 2 on the right bank.
4. Hold the surveyor's rod vertical, with its base planted at the water's edge. Examine both banks, then determine the channel *incision* as the *height up from the water surface to elevation of the* ***first terrace*** *of the valley floodplain* (Note this is at or above the bankfull channel height). Whenever possible, use the clinometer as a level (positioned so it reads 0% slope) to measure this height by transferring (backsighting) it onto the surveyor's rod. Record this value in the *INCISED HEIGHT* field of the bank measurement section on the field data form.
5. While still holding the surveyor's rod as a guide, and sighting with the clinometer as a level, examine both banks to measure and record the *height of bankfull flow above the present water level*. Look for evidence on one or both banks such as:
    - An obvious slope break that differentiates the channel from a relatively flat floodplain terrace higher than the channel.
    - A transition from exposed stream sediments to terrestrial vegetation.
    - Moss growth on rocks along the banks.
    - Presence of drift material caught on overhanging vegetation.
    - A transition from flood- and scour-tolerant vegetation to that which is relatively intolerant of these conditions.
6. Record the *wetted width* value determined when locating substrate sampling points in the *WETTED WIDTH* field in the bank measurement section of the field data form. Also determine the *bankfull channel width* and the *width of exposed mid-channel bars* (if present). Record these values in the *BANK MEASUREMENT* section of the field data form.
7. Repeat Steps 1 through 6 at each cross-section transect, (including any additional side channel transects established when islands are present). Record data for each transect on a separate field data form.

Bankfull flows are large enough to erode the stream bottom and banks, but frequent enough (every 1 to 2 years) to not allow substantial growth of upland terrestrial vegetation. Consequently, in many regions, it is these flows that have determined the width and depth of the

channel. Estimates of the bankfull dimensions of stream channels are extremely important in EMAP surveys. They are used to calculate shear stress and bed stability (see Kaufmann et al., 1999). Unfortunately, we have to depend upon evidence visible during the low-flow sampling season. If available, consult published rating curves relating expected bankfull channel dimensions to stream drainage area within the region of interest. Graphs of these rating curves can help you get a rough idea of where to look for field evidence to determine the level of bankfull flows. Curves such as these are available from the USGS for streams in most regions of the U.S. (e.g., Dunne and Leopold 1978; Harrelson et al. 1994, Leopold 1994). To use them, you need to know the contributing drainage area to your sample site. Interpret the expected bankfull levels from these curves as a height above the streambed in a riffle, but remember that your field measurement will be a height above the present water surface of the stream. Useful resources to aid your determination of bankfull flow levels in streams in the United States are video presentations produced by the USDA Forest Service for western streams (USDA Forest Service 1995) and eastern streams (USDA Forest Service 2002).

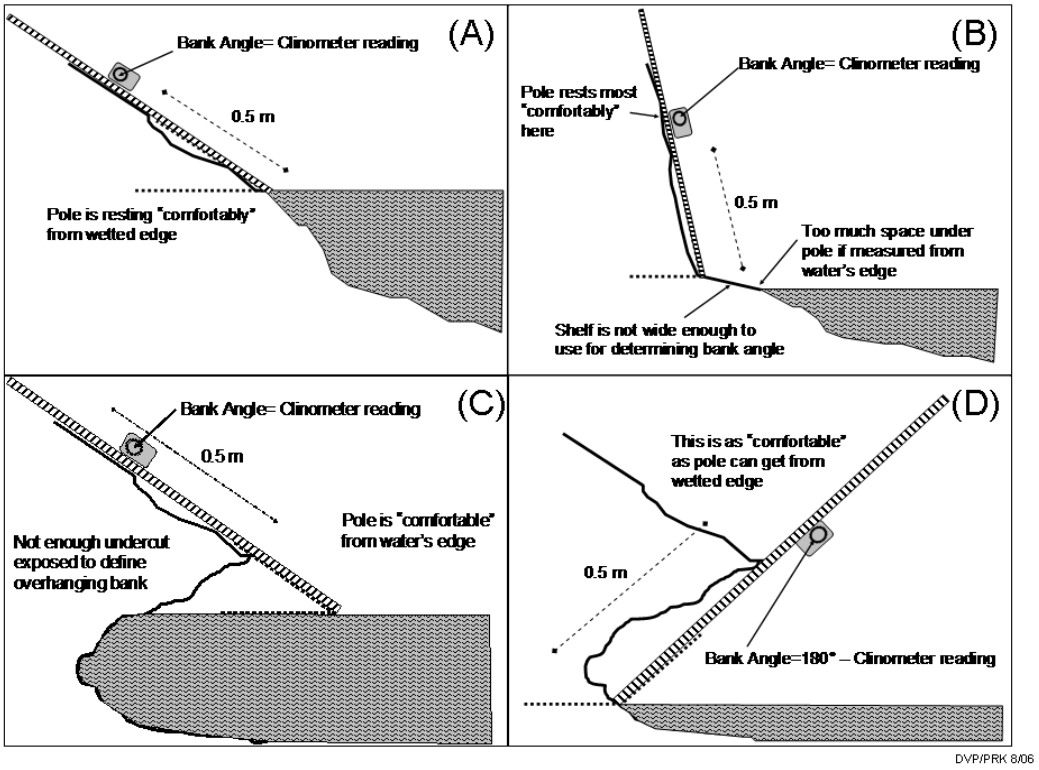


Figure 6.2-8. Determining bank angle under different types of bank conditions. (A) typical, (B) incised channel, (C) undercut bank, and (D) overhanging bank.

After consulting rating curves that show where to expect bankfull levels in a given size of stream, estimate the bankfull flow level by looking at the following indicators:

- First look at the stream and its valley to determine the active floodplain. This is a depositional surface that frequently is flooded and experiences sediment deposition under the current climate and hydrological regime.
- Then look specifically for:
- An obvious break in the slope of the banks.
- A change from water-loving and scour-tolerant vegetation to more drought-tolerant vegetation.
- A change from well-sorted stream sediments to unsorted soil materials.

In the absence of clear bankfull indications, consider the previous season's flooding as the best evidence available (note: you could be wrong if very large floods or prolonged droughts have occurred in recent years.). Look for:

- Drift debris ("sticky wickets" left by the previous seasons flooding).
- The level where deciduous leaf-fall is absent on the ground (carried away by previous winter flooding).
- Unvegetated sand, gravel or mud deposits from previous year's flooding.

In years that have experienced large floods, drift material and other recent high flow markers may be much higher than other bankfull indicators. In such cases, base your determination on less-transient indicators such as channel form, perennial vegetation, and depositional features. In these cases, flag your data entry and also record the height of drift material in the comments section of the field data form.

We use the vertical distance (height) from the observed water surface up to the level of the first major valley depositional surface (Figure 6.2-9) as a measure of the degree of *incision* or *downcutting* of the stream below the general level of its valley. This value is recorded in the *INCISED HEIGHT* field. It may not be evident at the time of sampling whether the channel is downcutting, stable, or aggrading (raising its bed by depositing sediment). However, by recording incision heights measured in this way and monitoring them over time, we will be able to tell if streams are incising or aggrading.

If the channel is not greatly incised, bankfull channel height and incision height will be the same (i.e., the first valley depositional surface is the active floodplain). However, if the channel is incised greatly, the bankfull level will be below the level of the first terrace of the valley floodplain, making bankfull channel height less than incision height (Figure 6.2-10). *Bankfull height is **never** greater than incision height.* You may need to look for evidence of recent flows (within about one year) to distinguish bankfull and incision heights. In cases where the channel is cutting a valley sideslope and has oversteepened and destabilized that slope, the bare "cutbank" against the steep hillside at the edge of the valley is not necessarily an indication of recent incision. In such a case, the opposite bank may be lower, with a more obvious terrace above bankfull height; choose that bank for your measurement of incised height. Examine both banks to more accurately determine incision height and bankfull height. Remember that incision height is measured as *the vertical distance to the first major depositional surface above bankfull* (whether or not it is an active floodplain or a terrace. If terrace heights differ on left and right

banks (both are above bankfull), choose the lower of the two terraces. In many cases your sample reach may be in a "V" shaped valley or gorge formed over eons, and the slope of the channel banks simply extends uphill indefinitely, not reaching a terrace before reaching the top of a ridge (Figure 6.2-10). In such cases, record incision height values equal to bankfull values and make appropriate comment that no terrace is evident. Similarly, when the stream has extremely incised into an ancient terrace, (e.g., the Colorado River in the Grand Canyon), you may crudely estimate the terrace height if it is the first one above bankfull level. If you cannot estimate the terrace height, make appropriate comments describing the situation.

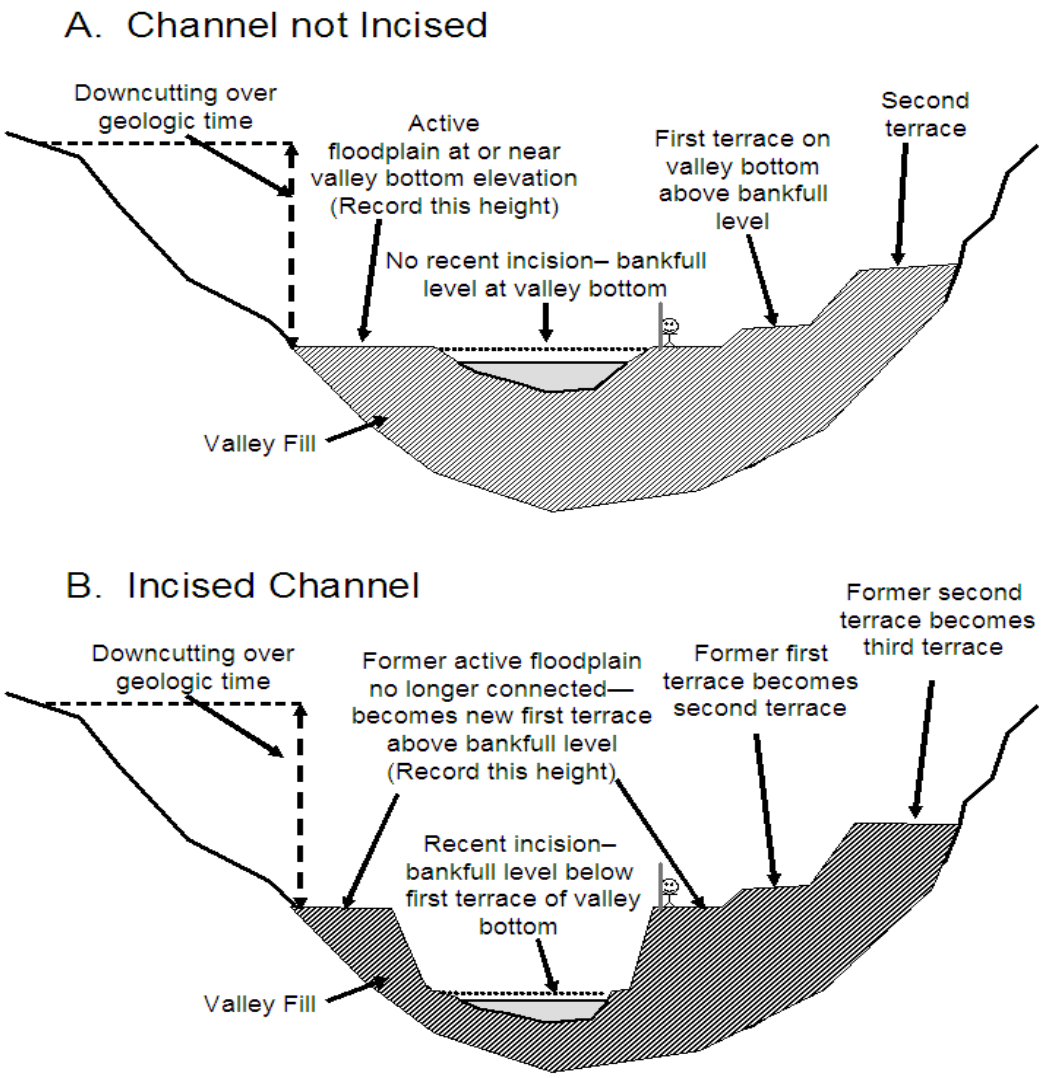


Figure 6.2-9. Schematic showing relationship between bankfull channel and incision. (A) not recently incised, and (B) recently incised into valley bottom. Note level of bankfull stage relative to elevation of first terrace (abandoned floodplain)on valley bottom. (Stick figure included for scale).

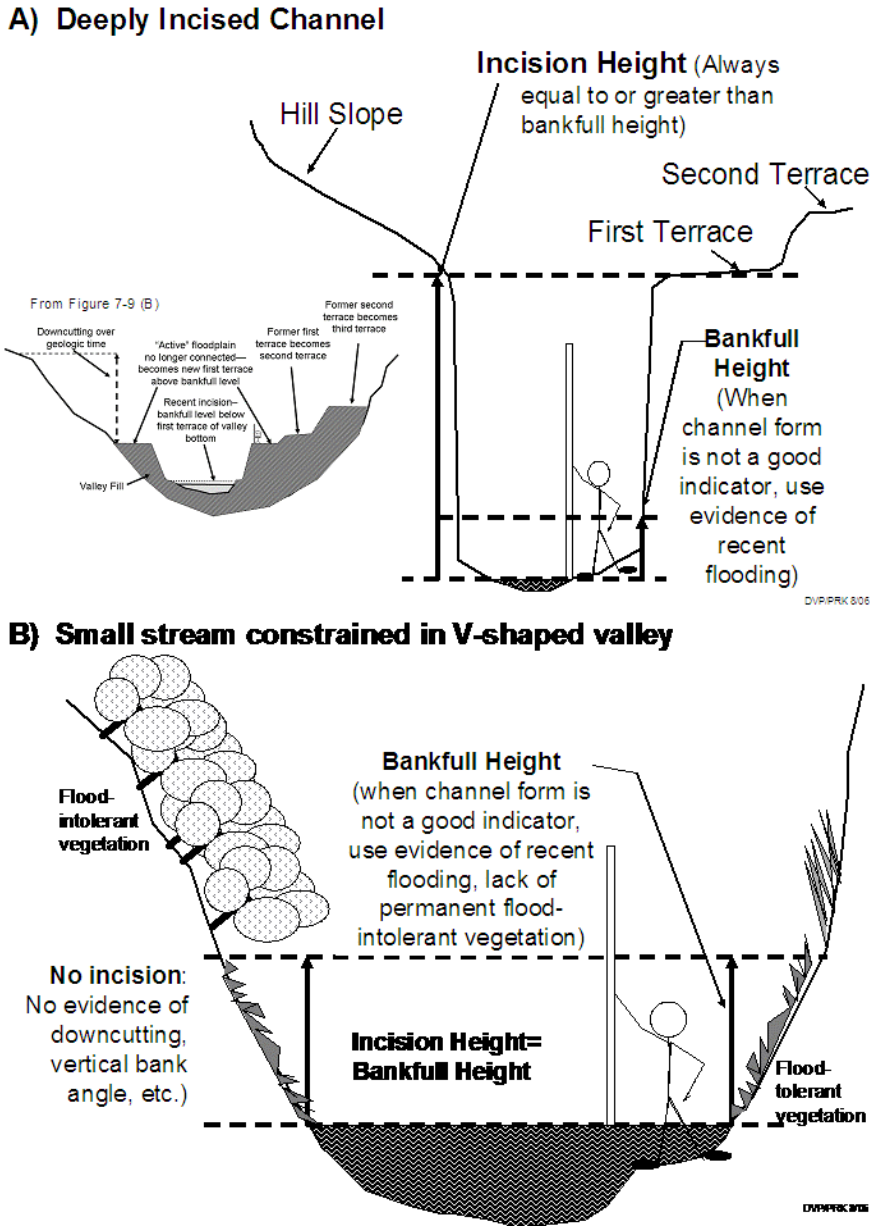


Figure 6.2-10. Determining bankfull and incision heights for (A) deeply incised channels, and (B) streams in deep V-shaped valleys. (Stick figure included for scale).

6.2.5.4 Canopy Cover Measurements

Canopy cover over the stream is determined at each of the 11 cross-section transects. A spherical densiometer (model *A*- **convex type**) is used (Lemmon 1957). Mark the densiometer with a permanent marker or tape exactly as shown in Figure 6.2-11 to limit the number of square grid intersections read to 17. Densiometer readings can range from 0 (no canopy cover) to 17 (maximum canopy cover). Six measurements are obtained at each cross-section transect (four measurements in each of four directions at mid-channel and one at each bank).

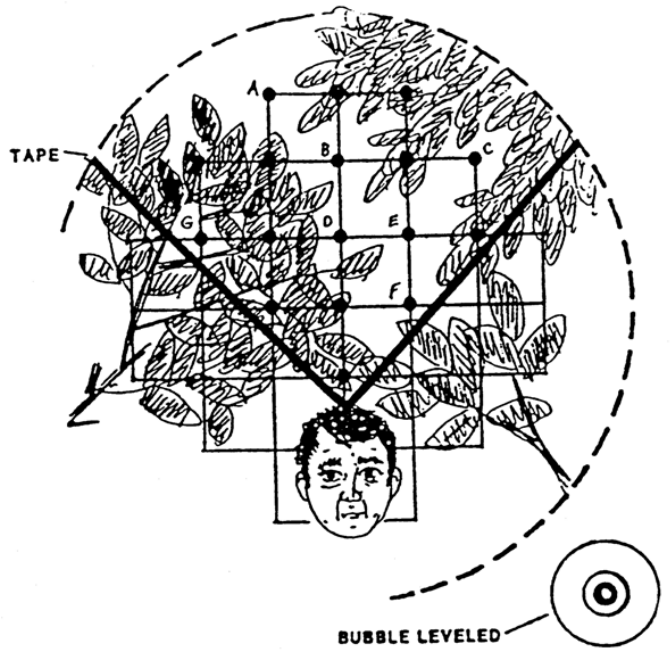


Figure 6.2-11. Schematic of modified convex spherical canopy densiometer. From Mulvey et al. (1992). Note proper positioning with the bubble leveled and face reflected at the apex of the "V". In this example, 10 of the 17 intersections show canopy cover, giving a densiometer reading of *10*.

The procedure for obtaining canopy cover data is presented in Table 6.2-9. Hold the densiometer level (using the bubble level) 0.3 m above the water surface with your face reflected just below the apex of the taped "V", as shown in Figure 6.2-11. Concentrate on the 17 points of grid intersection on the densiometer that lie within the taped "V". If the reflection of a tree or high branch or leaf overlies any of the intersection points, that particular intersection is counted as having cover. For each of the six measurement points, record the number of intersection points (maximum=17) that have vegetation covering them in the *CANOPY COVER MEASUREMENT* section of the Channel/Riparian Cross-section Form as shown in Figure 6.2-7.

Table 6.2-9. Procedure for canopy cover measurements

1. At each cross-section transect, stand in the stream at mid-channel and face upstream.
2. Hold the densiometer 0.3 m (1 ft) above the surface of the stream. Level the densiometer using the bubble level. Move the densiometer in front of you so your face is just below the apex of the taped "V".
3. Count the number of grid intersection points within the "V" that are covered by either a tree, a leaf, or a high branch. Record the value (0 to 17) in the *CENUP* field of the canopy cover measurement section of the Channel/Riparian Cross-section and Thalweg Profile Form.
4. Face toward the left bank (left as you face downstream). Repeat Steps 2 and 3, recording the value in the *CENL* field of the field data form.
5. Repeat Steps 2 and 3 facing downstream, and again while facing the right bank (right as you look downstream). Record the values in the *CENDWN* and *CENR* fields of the field data form.
6. Move to the water's edge (either the left or right bank). Repeat Steps 2 and 3 again, this time facing the bank. Record the value in the *LFT* or *RGT* fields of the field data form. Move to the opposite bank and repeat.
7. Repeat Steps 1 through 6 at each cross-section transect (including any additional side channel transects established when islands are present). Record data for each transect on a separate field data form.

6.2.5.5 Riparian Vegetation Structure

The previous section (6.2.5.4) described methods for quantifying the cover of canopy over the stream channel. The following visual estimation procedures supplement those measurements with a semi-quantitative evaluation of the type and amount of various types of riparian vegetation. Additional measures within the riparian zone (legacy trees and invasive riparian plants) are described in Section 6.2.5.9.

Riparian vegetation observations apply to the riparian area upstream 5 meters and downstream 5 meters from each of the 11 cross-section transects (refer to Figure 6.2-1). They include the visible area from the stream back a distance of 10m (~30 ft) shoreward from both the left and right banks, creating a 10 m $\times$ 10 m riparian plot on each side of the stream (Figure 6.2-12). The riparian plot dimensions are estimated, not measured. On steeply sloping channel margins, the 10 m $\times$ 10 m plot boundaries are defined as if they were projected down from an aerial view.

Table 6.2-10 presents the procedure for characterizing riparian vegetation structure and composition. Figure 6.2-7 illustrates how measurement data are recorded on the Channel/Riparian Cross-section Form. Conceptually divide the riparian vegetation into 3 layers: the *Canopy* layer (> 5 m high), the *Understory* layer (0.5 to 5 m high), and the *Ground cover* layer (< 0.5 m high). Note that several vegetation types (e.g., grasses or woody shrubs) can potentially occur in more than one layer. Similarly note that some things other than vegetation are possible entries for the *Ground cover* layer (e.g., barren ground).

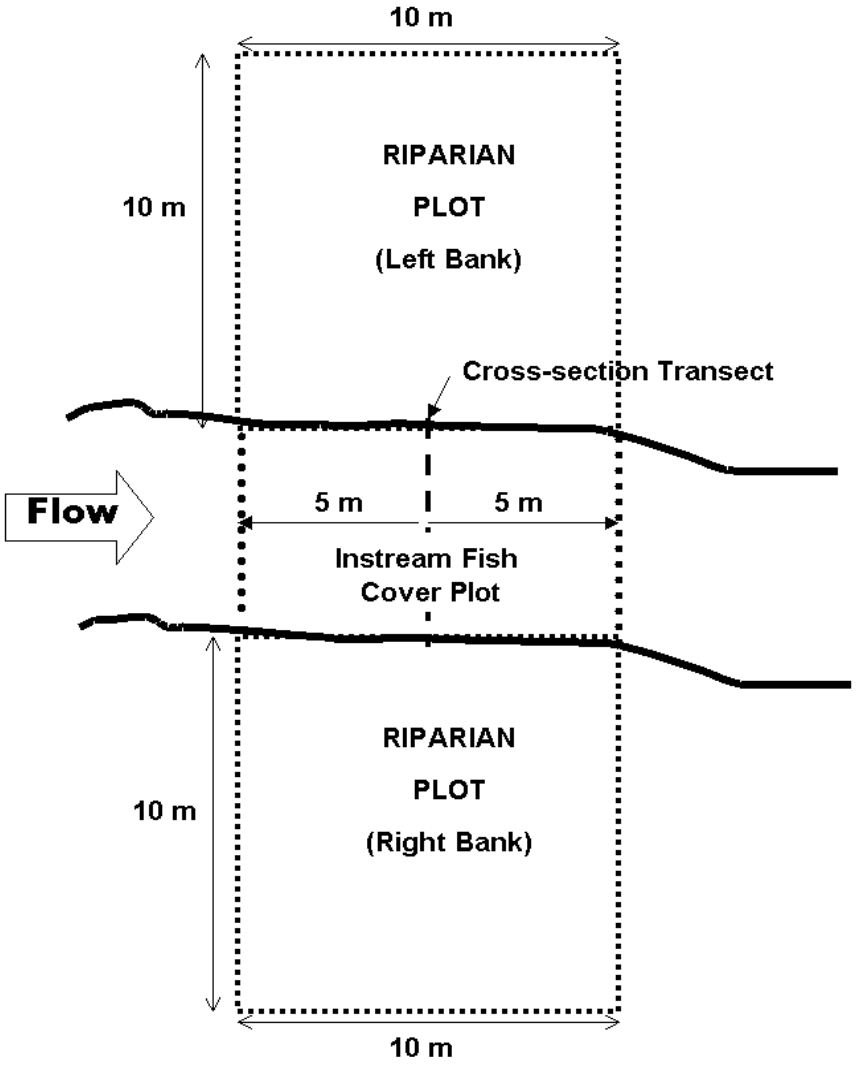


Figure 6.2-12. Riparian zone and instream fish cover plots for a stream cross-section transect.

Table 6.2-10. Procedure for characterizing riparian vegetation structure

1. Standing in mid-channel at a cross-section transect, estimate a 5 m distance upstream and downstream (10 m total length).
2. Facing the left bank (left as you face downstream), estimate a distance of 10 m back into the riparian vegetation.

   On steeply-sloping channel margins, estimate the distance into the riparian zone as if it were projected down from an aerial view.
3. Within this 10 m × 10 m area, conceptually divide the riparian vegetation into 3 layers: a *Canopy Layer* (>5 m high), an *Understory* (0.5 to 5 m high), and a *Ground Cover* layer (<0.5 m high).
4. Within this 10 m × 10 m area, determine the dominant vegetation type for the *CANOPY LAYER* (vegetation >5 m high) as either ***D**eciduous*, ***C**oniferous*, *broadleaf **E**vergreen*, ***M**ixed*, or ***N**one*. Consider the layer *Mixed* if more than 10% of the areal coverage is made up of the alternate vegetation type. Indicate the appropriate vegetation type in the *VISUAL RIPARIAN ESTIMATES* section of the Channel/Riparian Cross-section Form.
5. Determine separately the areal cover class of large trees (≥0.3 m [1 ft] diameter at breast height [dbh]) and small trees (<0.3 m dbh) within the canopy layer. Estimate areal cover as the amount of shadow that would be cast by a particular layer alone if the sun were directly overhead. Record the appropriate cover class on the field data form (*0=absent*: zero cover, *1=sparse*: <10%, *2=moderate*: 10-40%, *3=heavy*: 40-75%, or *4=very heavy*: >75%).
6. Look at the *UNDERSTORY* layer (vegetation between 0.5 and 5 m high). Determine the dominant **woody** vegetation type for the understory layer as described in Step 4 for the canopy layer. If there is no woody vegetation in the understory layer, record the type as ***N**one.*
7. Determine the areal cover class for woody shrubs and saplings separately from non-woody vegetation within the understory, as described in Step 5 for the canopy layer.
8. Look at the *GROUND COVER* layer (vegetation <0.5 m high). Determine the areal cover class for woody shrubs and seedlings, non-woody vegetation, and the amount of bare ground present as described in Step 5 for large canopy trees.
9. Repeat Steps 1 through 8 for the right bank.
10. Repeat Steps 1 through 9 for all cross-section transects (including any additional side channel transects established when islands are present). Use a separate field data form for each transect.

Before estimating the areal coverage of the vegetation layers, record the type of *woody* vegetation (*broadleaf **D**eciduous*, ***C**oniferous*, *broadleaf **E**vergreen*, ***M**ixed*, or ***N**one*) in each of the two taller layers (Canopy and Understory). Consider the layer *Mixed* if more than 10% of the areal coverage is made up of the alternate vegetation type. If there is no woody vegetation in the understory layer, record the type as *None*.

Estimate the areal cover separately in each of the three vegetation layers. Note that the areal cover can be thought of as the amount of shadow cast by a particular layer alone when the sun is directly overhead. *The maximum cover in each layer is 100%, so the sum of the areal covers for the combined three layers could add up to 300%.* The four areal cover classes are *Absent*, *Sparse* (<10%), *Moderate* (10 to 40%), *Heavy* (40 to 75%), and *Very Heavy* (>75%). These cover classes and their corresponding codes are shown on the field data form (Figure 6.2-7). When rating vegetation cover types for a single vegetation layer, mixtures of two or more subdominant classes might all be given *Sparse* (*1*), *Moderate* (*2*), or *Heavy* (*3*) ratings. One *Very Heavy* cover class with no clear subdominant class might be rated *4* with all the remaining classes rated as either *Moderate* (*2*), *Sparse* (*1*) or *Absent* (*0*). Note that within a

given vegetation layer, two cover types with 40-75% cover can both be rated *3*, but no more than one cover type could receive a rating of *4*.

6.2.5.6 Instream Fish Cover, Algae, and Aquatic Macrophytes

The procedure to estimate the types and amounts of instream fish cover is outlined in Table 6.2-11. Data are recorded on the Channel/Riparian Cross-section Form as shown in Figure 6.2-7. Estimate the areal cover of all of the fish cover and other listed features that are in the water and on the banks 5 m upstream and downstream of the cross-section (see Figure 6.2-12). The areal cover classes of fish concealment and other features are the same as those described for riparian vegetation (Section 6.2.5.5).

The entry *FILAMENTOUS ALGAE* refers to long streaming algae that often occur in slow moving waters. *AQUATIC MACROPHYTES* are water-loving plants, including mosses, in the stream that could provide cover for fish or macroinvertebrates. If the stream channel contains live wetland grasses, include these as aquatic macrophytes. *WOODY DEBRIS* are the larger pieces of wood that can influence cover and stream morphology (i.e., those pieces that would be included in the large woody debris tally [Section 6.2.4]). *BRUSH/WOODY DEBRIS* refers to smaller wood pieces that primarily affect cover but not morphology. *LIVE TREES OR ROOTS* are living trees that are within the channel – estimate the areal cover provided by the parts of these trees or roots that are inundated. *OVERHANGING VEGETATION* includes tree branches, brush, twigs, or other small debris that is not in the water but is close to the stream (within 1 m of the surface) and provides potential cover. *BOULDERS* are typically basketball- to car-sized particles. *ARTIFICIAL STRUCTURES* include those designed for fish habitat enhancement, as well as in-channel structures that have been discarded (e.g., concrete, asphalt, cars, or tires) or deliberately placed for diversion, impoundment, channel stabilization, or other purposes.

Table 6.2-11. Procedure for estimating instream fish cover

1. Standing mid-channel at a cross-section transect, estimate a 5m distance upstream and downstream (10 m total length).
2. Examine the water and both banks within the 10-m segment of stream for the following features and types of fish cover: *filamentous algae, aquatic macrophytes, large woody debris, brush and small woody debris, in-channel live trees or roots, overhanging vegetation, undercut banks, boulders, and artificial structures*.
3. For each cover type, estimate the areal cover. Record the appropriate cover class in the *FISH COVER/OTHER* section of the Channel/Riparian Cross-section Form:

   0=absent: zero cover,

   1=sparse: <10%,

   2=moderate: 10-40%,

   3=heavy: >40-75%, or

   4=very heavy: >75%).
4. Repeat Steps 1 through 3 at each cross-section transect (including any additional side channel transects established when islands are present). Record data from each transect on a separate field data form.

6.2.5.7 Human Influence

For the left and right banks at each of the 11 detailed Channel and Riparian Cross-sections, evaluate the presence/absence and the proximity of 11 categories of human influences with the procedure outlined in Table 6.2-12. Relate your observations and proximity evaluations to the stream and riparian area within 5 m upstream and 5 m downstream from the station (Figure 6.2-12). Four proximity classes are used: In the stream or on the bank within 5 m upstream or downstream of the cross-section transect, present within the 10 m × 10 m riparian plot but not in the stream or on the bank, present outside of the riparian plot, and absent. Record data on the Channel/Riparian Cross-section Form as shown in Figure 6.2-7. If a disturbance is within more than one proximity class, record the one that is closest to the stream (e.g., *C* takes precedence over *P*).

A particular influence may be observed outside of more than one riparian observation plot (e.g., at both transects *D* and *E*). Record it as present at every transect where you can see it without having to sight through another transect or its 10 m × 10 m riparian plot.

Table 6.2-12. Procedure for estimating human influence

1. Standing mid-channel at a cross-section transect, look toward the left bank (left when facing downstream), and estimate a 5 m distance upstream and downstream (10 m total length). Also, estimate a distance of 10 m back into the riparian zone to define a riparian plot area.
2. Examine the channel, bank and riparian plot area adjacent to the defined stream segment for the following human influences: (1) *walls, dikes, revetments, riprap, and dams*; (2) *buildings*; (3) *pavement/cleared lots* (e.g., paved, gravelled, dirt parking lot, foundation); (4) *roads or railroads*, (5) *inlet or outlet pipes*; (6) *landfills or trash* (e.g., cans, bottles, trash heaps); (7) *parks or maintained lawns*; (8) *row crops*; (9) *pastures, rangeland, hay fields*, or *evidence of livestock*; (10) *logging*; and (11) *mining* (including gravel mining).
3. For each type of influence, determine if it is present and what its proximity is to the stream and riparian plot area. Consider human disturbance items as present if you can see them from the cross-section transect. Do not include them if you have to sight through another transect or its 10 m ×10 m riparian plot.
4. For each type of influence, record the appropriate proximity class in the *HUMAN INFLUENCE* part of the *VISUAL RIPARIAN ESTIMATES* section of the Channel/Riparian Cross-section Form. Proximity classes are:

B (*Bank*)	Present within the defined 10 m stream segment and located in the stream or on the stream bank.
C (*Close*)	Present within the 10 × 10 m riparian plot area, but away from the bank.
P (*Present*)	Present, but outside the riparian plot area.
0 (*Absent*)	Not present within or adjacent to the10 m stream segment or the riparian plot area at the transect

5. Repeat Steps 1 through 4 for the right bank.
6. Repeat Steps 1 through 5 for each cross-section transect, (including any additional side channel transects established when islands are present). Record data for each transect on a separate field form.

6.2.5.8 Cross-section Transects on Side Channels

If the wetted channel is split by an island, and the estimated flow in the side channel is less than or equal to 15% of the total flow, the bank and riparian measurements are made at each side of the main channel (the minor side channel is ignored other than to note its presence on the thalweg profile form), so one riparian plot is established on the island as shown in Figure 6.2-13. If an island is present that creates a major side channel containing **more than 15%** of the total flow (Section 6.2.4.1), an additional cross-section transect is established for the side channel as shown in Figure 6.2-13. Separate substrate, bank and riparian measurements are made for side channel transects. Data from the additional side channel transect are recorded on a separate Channel/Riparian Cross-section Form as shown in Figure 6.2-14. Riparian plots established on the island for each transect may overlap (and be < 10 m shoreward) if the island is less than 10 m wide at the transect.

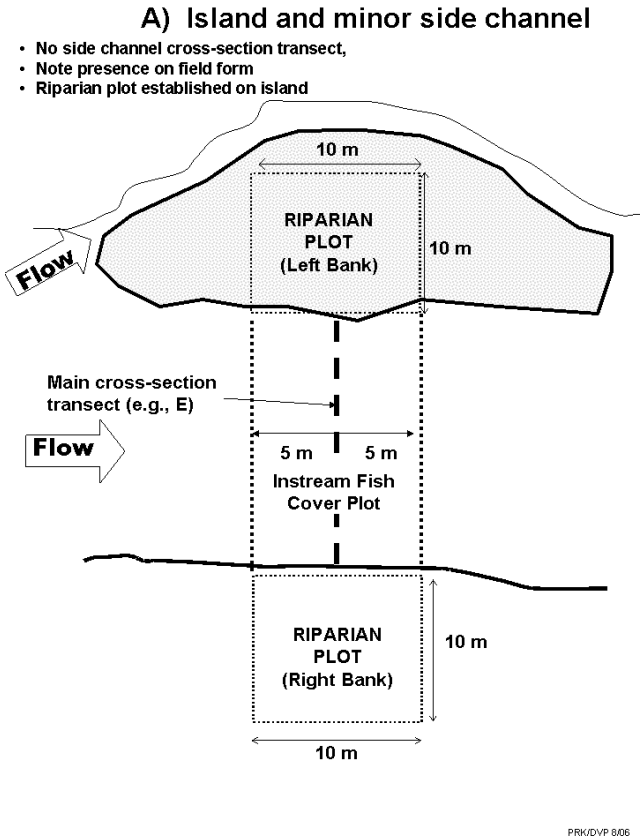


Figure 6.2-13. Riparian and instream fish cover plots for a stream with minor and major side channels.

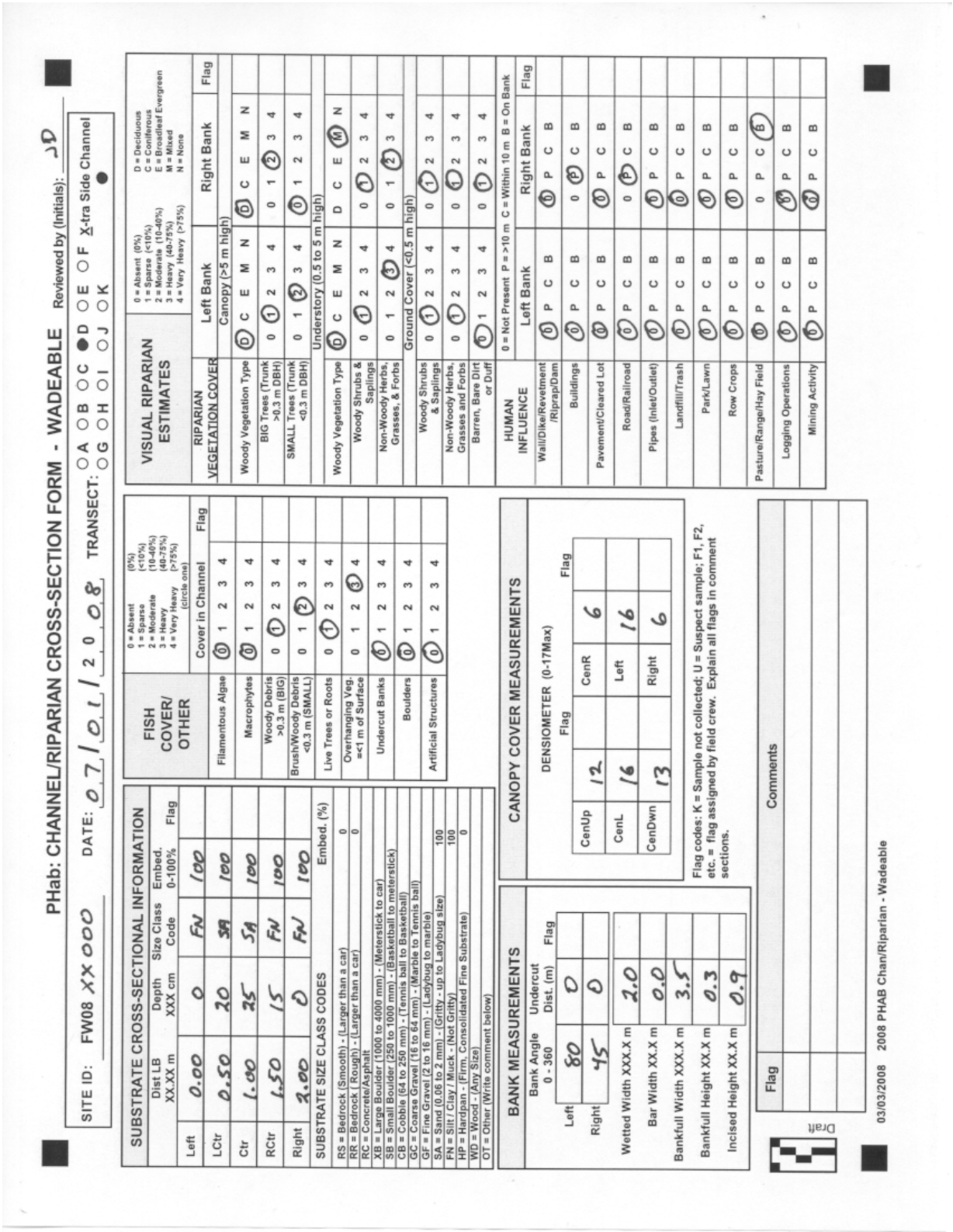


Figure 6.2-14. Channel/Riparian Cross-section Form for an additional major side channel transect.

6.2.5.9 Riparian "Legacy" Trees and Invasive Alien Species

Follow the procedures in Table 6.2-13 to locate the **largest** tree associated with each transect. The tree you choose may not truly be an old *legacy* tree – just choose the largest you see. We use these data to determine if there are true legacy trees somewhere within the support reach. Note that only one tree is identified for each transect between that transect and the next one upstream; at transect *K*, look upstream a distance of 4 channel widths. Record the type of tree, and, if possible, the taxonomic group (using the list provided in Table 6.2-13) on the left-hand column of the Riparian "Legacy" Trees and Invasive Alien Plants form (Figure 6.2-15). Estimate the height of the tree and the diameter at breast height (dbh), and mark the appropriate height and dbh classes on the form. Estimate and record the distance of the legacy tree from the wetted margin of the stream.

Search in the 10 m x 10 m riparian and littoral plots on both banks for the presence of any invasive alien species listed in the NRSA Invasive Species Guide provided to each field crew. Document the species observed on the Riparian "Legacy" Trees and Invasive Alien Plants form (Figure 5.2-8), answering the question of whether each of the target species is present in the plot. If you have a camera, document the species with a photograph. If you observe no alien taxa within the riparian and littoral plots, but can confidently identify them outside of the plots, include your observations in the comments portion of the form. If the river is too wide to effectively observe the far bank at a transect, record what you observe for the plot on the near bank, record a "U" flag, and explain in the comments section of the form.

Table 6.2-13. Procedure for identifying riparian legacy trees

Legacy Trees:

- Beginning at Transect A, look upstream and downstream as far as you can see confidently. Search both sides of the stream downstream to the next transect. Locate the largest tree visible within 100m (or as far as you can see, if less) from the wetted bank.
- Classify this tree as broadleaf deciduous, coniferous, or broadleaf evergreen (classify western larch as coniferous). Identify, if possible, the species or the taxonomic group of this tree from the list below.

1. Acacia/Mesquite	10. Poplar/Cottonwood
2. Alder/Birch	11. Snag (Dead Tree of Any Species)
3. Ash	12. Spruce
4. Cedar/Cypress/Sequoia	13. Sycamore
5. Fir (including Douglas Fir, Hemlock)	14. Willow
6. Juniper	15. Unknown, other Broadleaf Evergreen
7. Maple/Boxelder	16. Unknown or Other Conifer
8. Oak	17. Unknown or Other Deciduous
9. Pine	

NOTE: If the largest tree is a dead "snag", enter "Snag" as the taxonomic group.

Estimate the height of the potential legacy tree, its diameter at breast height (dbh) and its distance from the wetted margin of the stream. Enter this information on the left hand column of the Riparian "Legacy" Trees and Invasive Alien Plants field form.

Alien Invasive Plants:
Examine the 10m x 10m riparian and littoral plots on both banks for the presence of alien species. (Species lists will be provided)
Record the presence of any species listed within the plots on either the left or right bank on the Riparian "Legacy" Trees and Invasive Alien Species field form. If none of the species listed is present in the plots at a given transect, fill in the circle indicating "None" for this transect. Repeat for each remaining transect (B through K). At transect "K", look upstream a distance of 4 channel widths when locating the legacy tree.

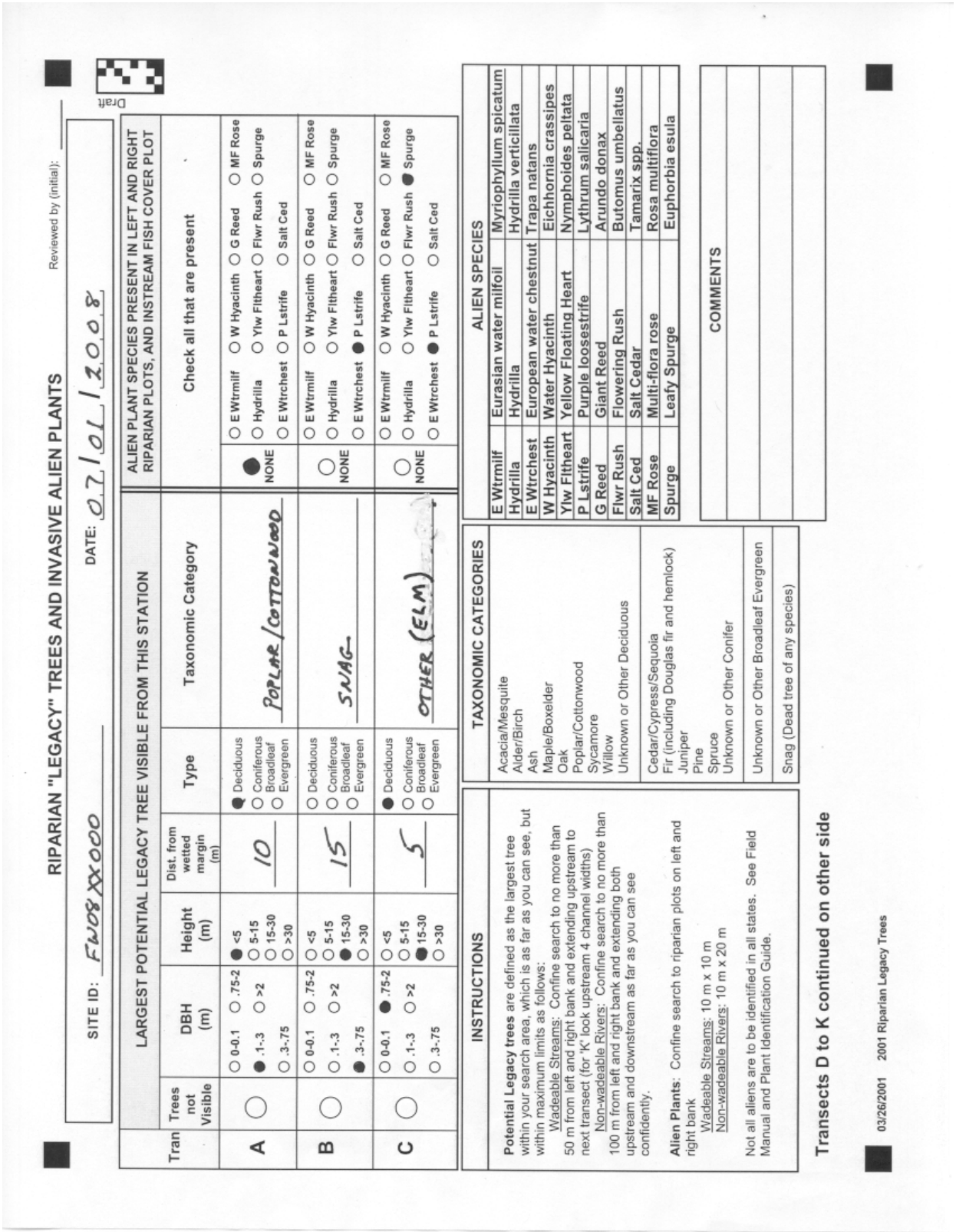
6.2.6 Channel Constraint, Debris Torrents, Recent Floods, and Discharge

6.2.6.1 Channel Constraint

After completing the thalweg profile and riparian/channel cross-section measurements and observations, envision the stream at bankfull flow and evaluate the degree, extent and type of channel constraint, using the procedures presented in Table 6.2-14. Record data on the Channel Constraint Assessment Form (Figure 6.2-16). First, classify the stream reach channel pattern as predominantly a *single channel*, an *anastomosing channel*, or a *braided channel* (Figure 6.2-17):

1. *Single channels* may have occasional in-channel bars or islands with side channels, but feature a *predominant single channel*, or a *dominant main channel with a subordinate side channel*.
2. *Anastomosing channels* have relatively long *major and minor channels* (but no predominant channel) in a complex network, diverging and converging around many *vegetated islands*. Complex channel pattern remains even during major floods.
3. *Braided channels* also have multiple branching and rejoining channels, (but no predominant channel) *separated by unvegetated bars*. Channels are generally smaller, shorter, and more numerous, often with no obvious dominant channel. During major floods, a single continuous channel may develop

After classifying the channel pattern, determine whether the channel is constrained within a narrow valley, constrained by local features within a broad valley, unconstrained and free to move about within a broad floodplain, or free to move about, but within a relatively narrow valley floor. Then examine the channel to ascertain the bank and valley features that constrain the stream. Entry choices for the type of constraining features are bedrock, hillslopes, terraces/alluvial fans, and human land use (e.g., a road, a dike, landfill, rip-rap, etc.). Estimate the percent of the channel margin in contact with constraining features (for unconstrained channels, this is 0%). To aid in this estimate, you may wish to refer to the individual transect assessments of incision and constraint. Finally, estimate the "typical" bankfull channel width and estimate the average width of the valley floor either with a topographic map or visually. If you cannot directly estimate the valley width (e.g., it is further than you can see, or if your view is blocked by vegetation), record the distance you can see and mark the appropriate circle on the field form.



RIPARIAN "LEGACY" TREES AND INVASIVE ALIEN PLANTS

Reviewed by (initial): ____

SITE ID: FW08XX000 DATE: 07/01/2008

Draft

Tran	Trees not Visible	LARGEST POTENTIAL LEGACY TREE VISIBLE FROM THIS STATION: DBH (m)	Height (m)	Dist. from wetted margin (m)	Type	Taxonomic Category	ALIEN PLANT SPECIES PRESENT IN LEFT AND RIGHT RIPARIAN PLOTS, AND INSTREAM FISH COVER PLOT: Check all that are present
A	○	○ 0-0.1 ○ .75-2 ● .1-.3 ○ >2 ○ .3-.75	● <5 ○ 5-15 ○ 15-30 ○ >30	10	● Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen	POPLAR/COTTONWOOD	● NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fitheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
B	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ● .3-.75	○ <5 ○ 5-15 ● 15-30 ○ >30	15	○ Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen	SNAG	○ NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fitheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ● P Lstrife ○ Salt Ced
C	○	○ 0-0.1 ● .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ● 15-30 ○ >30	5	● Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen	OTHER (ELM)	○ NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fitheart ○ Flwr Rush ● Spurge ○ E Wtrchest ● P Lstrife ○ Salt Ced

INSTRUCTIONS

Potential Legacy trees are defined as the largest tree within your search area, which is as far as you can see, but within maximum limits as follows:
Wadeable Streams: Confine search to no more than 50 m from left and right bank and extending upstream to next transect (for 'K' look upstream 4 channel widths)
Non-wadeable Rivers: Confine search to no more than 100 m from left and right bank and extending both upstream and downstream as far as you can see confidently.

Alien Plants: Confine search to riparian plots on left and right bank
Wadeable Streams: 10 m x 10 m
Non-wadeable Rivers: 10 m x 20 m

Not all aliens are to be identified in all states. See Field Manual and Plant Identification Guide.

TAXONOMIC CATEGORIES

- Acacia/Mesquite
- Alder/Birch
- Ash
- Maple/Boxelder
- Oak
- Poplar/Cottonwood
- Sycamore
- Willow
- Unknown or Other Deciduous
- Cedar/Cypress/Sequoia
- Fir (including Douglas fir and hemlock)
- Juniper
- Pine
- Spruce
- Unknown or Other Conifer
- Unknown or Other Broadleaf Evergreen
- Snag (Dead tree of any species)

ALIEN SPECIES

E Wtrmilf	Eurasian water milfoil	Myriophyllum spicatum
Hydrilla	Hydrilla	Hydrilla verticillata
E Wtrchest	European water chestnut	Trapa natans
W Hyacinth	Water Hyacinth	Eichhornia crassipes
Ylw Fitheart	Yellow Floating Heart	Nymphoides peltata
P Lstrife	Purple loosestrife	Lythrum salicaria
G Reed	Giant Reed	Arundo donax
Flwr Rush	Flowering Rush	Butomus umbellatus
Salt Ced	Salt Cedar	Tamarix spp.
MF Rose	Multi-flora rose	Rosa multiflora
Spurge	Leafy Spurge	Euphorbia esula

COMMENTS

Transects D to K continued on other side

03/26/2001 2001 Riparian Legacy Trees

Figure 6.2-15. Riparian "Legacy" Tree and Invasive Alien Plants Form (Page 1)

Table 6.2-14. Procedures for assessing channel constraint

NOTE: These activities are conducted after completing the thalweg profile and littoral-riparian measurements and observations, and represent an evaluation of the entire stream reach.

***CHANNEL CONSTRAINT*:** Determine the degree, extent, and type of channel constraint based on envisioning the stream at **bankfull flow**.

Classify the stream reach channel pattern as predominantly a **single** channel, an **anastomosing** channel, or a **braided** channel.

> ***Single channels*** *may have occasional in-channel bars or islands with side channels*, but feature a predominant single channel, or a dominant main channel with a subordinate side channel.
>
> ***Anastomosing channels*** *have relatively long major and minor channels* branching and rejoining in a complex network separated by vegetated islands, with no obvious dominant channel.
>
> ***Braided channels*** *also have multiple branching and rejoining channels,* separated by unvegetated bars. Subchannels are generally small, short, and numerous, often with no obvious dominant channel.

After classifying the channel pattern, determine whether the channel is constrained within a narrow valley, constrained by local features within a broad valley, unconstrained and free to move about within a broad floodplain, or free to move about, but within a relatively narrow valley floor.

Then examine the channel to ascertain the bank and valley features that constrain the stream. Entry choices for the type of constraining features are bedrock, hillslopes, terraces/alluvial fans, and human land use (e.g., a road, a dike, landfill, rip-rap, etc.).

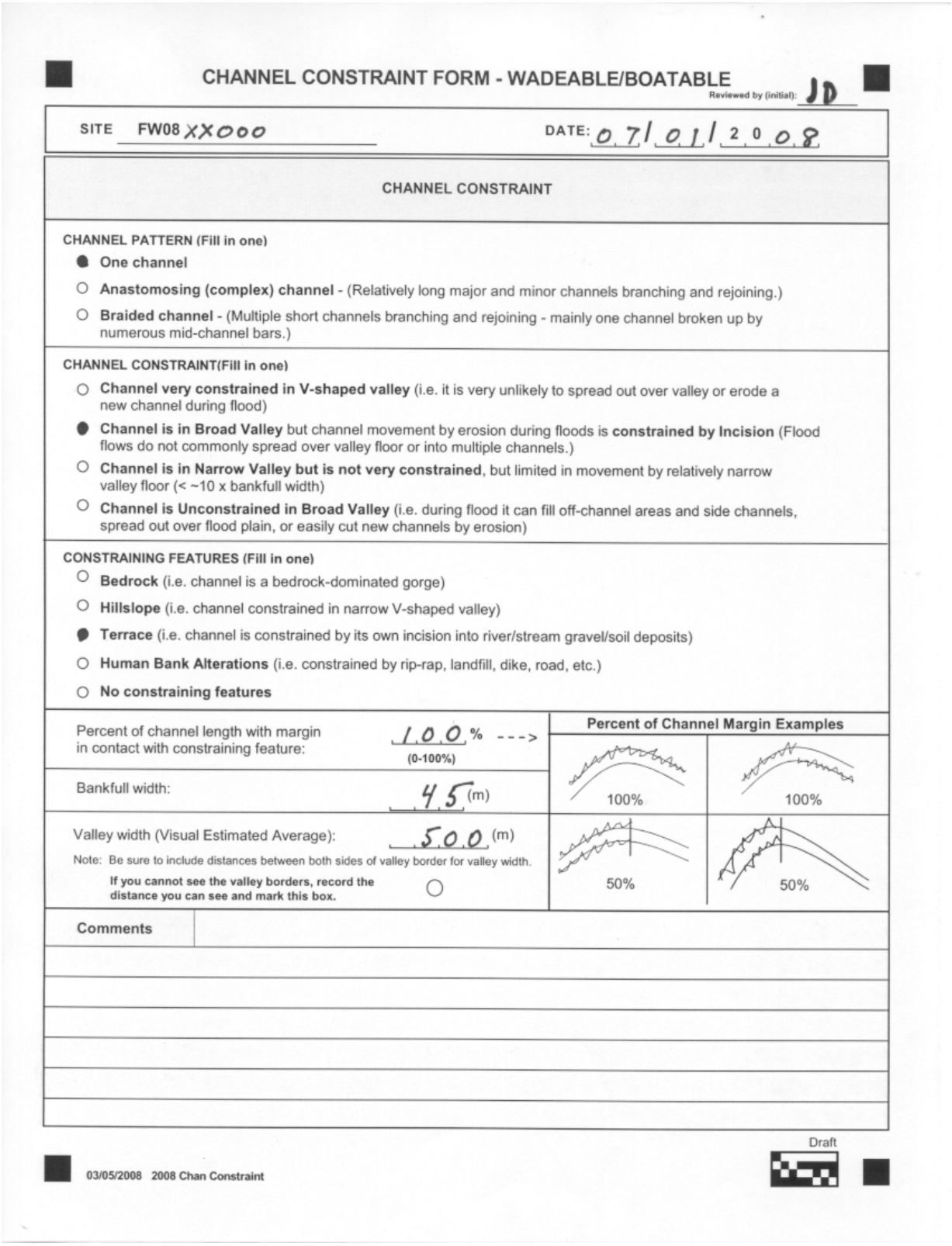
Based on your determinations from Steps 1 through 3, select and record one of the constraint classes shown on the Channel Constraint Form.

Estimate the percent of the channel margin in contact with constraining features (for unconstrained channels, this is 0%). Record this value on the Channel Constraint Form.

Finally, estimate the “typical” bankfull channel width, and visually estimate the average width of the valley floor. Record these values on the Channel Constraint Form.

NOTE: To aid in this estimate, you may wish to refer to the individual transect assessments of incision and constraint that were recorded on the Channel/Riparian Cross-Section Forms.

> NOTE: If the valley is wider than you can directly estimate, record the distance you can see and mark the circle on the field form.



CHANNEL CONSTRAINT FORM - WADEABLE/BOATABLE

Reviewed by (initial): JD

SITE FW08 XX000 DATE: 07/01/2008

CHANNEL CONSTRAINT

CHANNEL PATTERN (Fill in one)

- ● One channel
- ○ **Anastomosing (complex) channel** - (Relatively long major and minor channels branching and rejoining.)
- ○ **Braided channel** - (Multiple short channels branching and rejoining - mainly one channel broken up by numerous mid-channel bars.)

CHANNEL CONSTRAINT(Fill in one)

- ○ **Channel very constrained in V-shaped valley** (i.e. it is very unlikely to spread out over valley or erode a new channel during flood)
- ● **Channel is in Broad Valley** but channel movement by erosion during floods is **constrained by Incision** (Flood flows do not commonly spread over valley floor or into multiple channels.)
- ○ **Channel is in Narrow Valley but is not very constrained**, but limited in movement by relatively narrow valley floor (< ~10 x bankfull width)
- ○ **Channel is Unconstrained in Broad Valley** (i.e. during flood it can fill off-channel areas and side channels, spread out over flood plain, or easily cut new channels by erosion)

CONSTRAINING FEATURES (Fill in one)

- ○ **Bedrock** (i.e. channel is a bedrock-dominated gorge)
- ○ **Hillslope** (i.e. channel constrained in narrow V-shaped valley)
- ● **Terrace** (i.e. channel is constrained by its own incision into river/stream gravel/soil deposits)
- ○ **Human Bank Alterations** (i.e. constrained by rip-rap, landfill, dike, road, etc.)
- ○ **No constraining features**

Percent of channel length with margin in contact with constraining feature: 100 % ---> (0-100%)

Bankfull width: 4.5 (m)

Valley width (Visual Estimated Average): 500 (m)

Note: Be sure to include distances between both sides of valley border for valley width.

If you cannot see the valley borders, record the distance you can see and mark this box. ○

Percent of Channel Margin Examples

Comments

Draft

03/05/2008 2008 Chan Constraint

Figure 6.2-16. Channel Constraint Form, showing data for channel constraint.

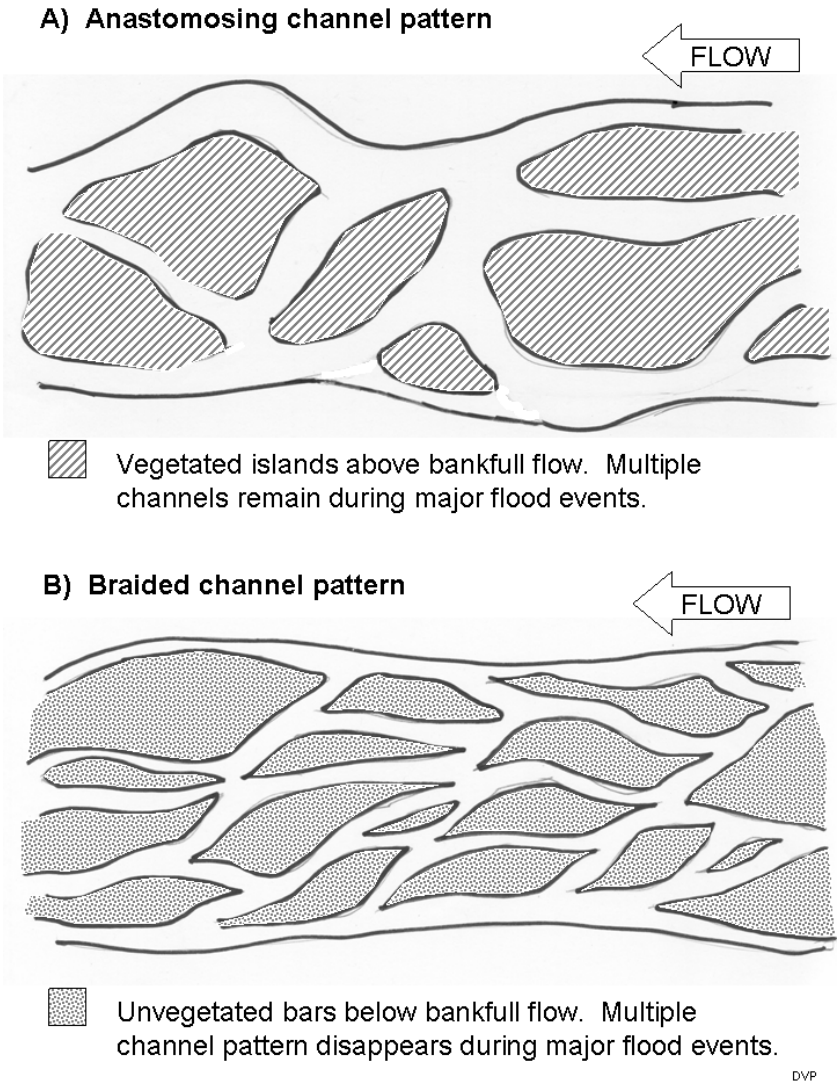


Figure 6.2-17. Types of multiple channel patterns.

6.2.6.2 Debris Torrents and Recent Major Floods

Debris torrents, or *lahars*, differ from conventional floods in that they are flood waves of higher magnitude and shorter duration, and their flow consists of a dense mixture of water and debris. Their high flows of dense material exert tremendous scouring forces on streambeds. For example, in the Pacific Northwest, flood waves from debris torrents can exceed 5 meters deep in small streams normally 3 m wide and 15 cm deep. These torrents move boulders in excess of 1 m diameter and logs >1 m diameter and >10 m long. In temperate regions, debris torrents occur primarily in steep drainages and are relatively infrequent, occurring typically less than once in several centuries. They are usually set into motion by the sudden release of large volumes of water upon the breaching of a natural or human-constructed impoundment, a process often initiated by mass hillslope failures (landslides) during high intensity rainfall or snowmelt. Debris torrents course downstream until the slope of the stream channel can no longer keep their viscous sediment suspension in motion (typically <3% for small streams); at this point, they “set up”, depositing large amounts of sediment, boulders, logs, and whatever

else they were transporting. Upstream, the *torrent track* is severely scoured, often reduced in channel complexity and devoid of near-bank riparian vegetation. As with floods, the massive disruption of the stream channel and its biota are transient, and these intense, infrequent events will often lead to a high-quality complex habitat within years or decades, as long as natural delivery of large wood and sediment from riparian and upland areas remains intact.

In arid areas with high runoff potential, debris torrents can occur in conjunction with flash flooding from extremely high-intensity rainfall. They may be nearly annual events in some steep ephemeral channels where drainage area is sufficient to guarantee isolated thunderstorms somewhere within their boundaries, but small enough that the effect of such storms is not dampened out by the portion of the watershed not receiving rainfall during a given storm.

Because they may alter habitat and biota substantially, infrequent major floods and torrents can confuse the interpretation of measurements of stream biota and habitat in regional surveys and monitoring programs. Therefore, it is important to determine if a debris torrent or major flood has occurred within the recent past. After completing the thalweg profile and channel/riparian measurements and observations, examine the stream channel along the entire sample reach, including its substrate, banks, and riparian corridor, checking the presence of features described on the Torrent Evidence Assessment Form (Figure 6.2-18). It may be advantageous to look at the channel upstream and downstream of the actual sample reach to look for areas of torrent scour and massive deposition to answer some of the questions on the field form. For example, you may more clearly recognize the sample reach as a torrent deposition area if you find extensive channel scouring upstream. Conversely, you may more clearly recognize the sample reach as a torrent scour reach if you see massive deposits of sediment, logs, and other debris downstream.

6.2.6.3 Stream Discharge

Stream discharge is equal to the product of the mean current velocity and vertical cross-sectional area of flowing water. Discharge measurements are critical for assessing trends in streamwater acidity and other characteristics that are very sensitive to streamflow differences. Discharge should be measured at a suitable location within the sample reach that is as close as possible to the location where chemical samples are collected, so that these data correspond. Discharge is usually determined after collecting water chemistry samples.

No single method for measuring discharge is applicable to all types of stream channels. The preferred procedure for obtaining discharge data is based on “velocity-area” methods (e.g., Rantz and others, 1982; Linsley et al., 1982). For streams that are too small or too shallow to use the equipment required for the velocity-area procedure, two alternative procedures are presented. One procedure is based on timing the filling of a volume of water in a calibrated bucket. The second procedure is based on timing the movement of a neutrally buoyant object (e.g., an orange or a small rubber ball) through a measured length of the channel, after measuring one or more cross-sectional depth profiles within that length.

Reviewed by (Initials): JD

TORRENT EVIDENCE ASSESSMENT FORM

SITE ID: FW08 XX000 DATE: 07/01/2008

TORRENT EVIDENCE	
Please fill in any of the following that are evident.	
EVIDENCE OF TORRENT SCOURING:	
○	01 - Stream channel has a recently devegetated corridor two or more times the width of the low flow channel. This corridor lacks riparian vegetation with possible exception of fireweed, even-aged alder or cottonwood seedlings, grasses, or other herbaceous plants.
○	02 - Stream substrate cobbles or large gravel particles are NOT IMBRICATED. (Imbricated means that they lie with flat sides horizontal and that they are stacked like roof shingles -- imagine the upstream direction as the top of the "roof.") In a torrent scour or deposition channel, the stones are laying in unorganized patterns, lying "every which way." In addition many of the substrate particles are angular (not "water-worn.")
○	03 - Channel has little evidence of pool-riffle structure. (For example, could you ride a mountain bike down the channel?)
○	04 - The stream channel is scoured down to bedrock for substantial portion of reach.
○	05 - There are gravel or cobble berms (little levees) above bankfull level.
○	06 - Downstream of the scoured reach (possibly several miles), there are massive deposits of sediment, logs, and other debris.
○	07 - Riparian trees have fresh bark scars at many points along the stream at seemingly unbelievable heights above the channel bed.
○	08 - Riparian trees have fallen into the channel as a result of scouring near their roots.
EVIDENCE OF TORRENT DEPOSITS:	
○	09 - There are massive deposits of sediment, logs, and other debris in the reach. They may contain wood and boulders that, in your judgement, could not have been moved by the stream at even extreme flood stage.
○	10 - If the stream has begun to erode newly laid deposits, it is evident that these deposits are "MATRIX SUPPORTED." This means that the large particles, like boulders and cobbles, are often not touching each other, but have silt, sand, and other fine particles between them (their weight is supported by these fine particles -- in contrast to a normal stream deposit, where fines, if present, normally "fill-in" the interstices between coarser particles.)
NO EVIDENCE:	
●	11 - No evidence of torrent scouring or torrent deposits.

COMMENTS

Draft

03/06/2008 NRSA Torrrent Evidence

Figure 6.2-18. Torrent Evidence Assessment Form.

6.2.6.4 Velocity-Area Procedure

Because velocity and depth typically vary greatly across a stream, accuracy in field measurements is achieved by measuring the mean velocity and flow cross-sectional area of many increments across a channel (Figure 6.2-19). Each increment gives a subtotal of the stream discharge, and the whole is calculated as the sum of these parts. Discharge measurements are made **at only one carefully chosen channel cross-section within the sampling reach**. It is important to choose a channel cross-section that is as much like a canal as possible. A glide area with a "U" shaped channel cross-section that is free of obstructions provides the best conditions for measuring discharge by the velocity-area method. You may remove rocks and other obstructions to improve the cross-section before any measurements are made. However, because removing obstacles from one part of a cross-section affects adjacent water velocities, you must not change the cross-section once you commence collecting the set of velocity and depth measurements.

The procedure for obtaining depth and velocity measurements is outlined in Table 6.2-15. Record the data from each measurement on the Stream Discharge Form as shown in Figure 6.2-20. In the field, data will be recorded using only one of the available procedures.

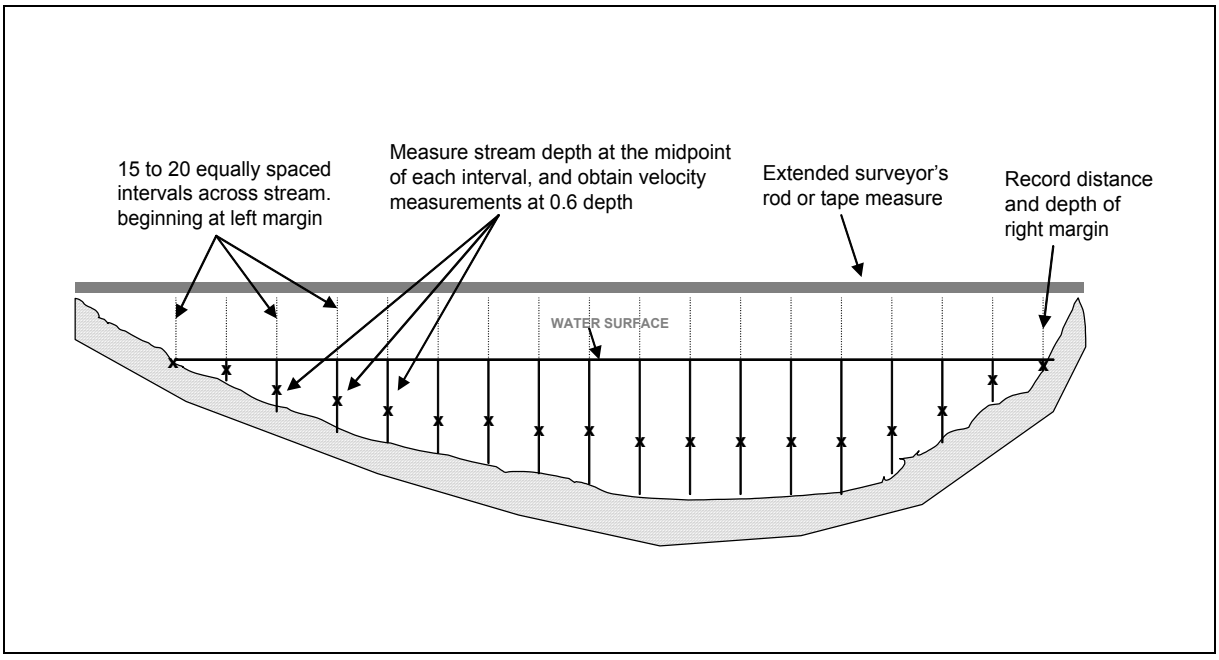


Figure 6.2-19. Layout of channel cross-section for obtaining discharge data by the velocity-area procedure.

Table 6.2-15. Velocity-Area procedure for determining stream discharge

1. Locate a cross-section of the stream channel for discharge determination that has most of the following qualities (based on Rantz and others, 1982):

    - Segment of stream above and below cross-section is straight
    - Depths mostly greater than 15 centimeters, and velocities mostly greater than 0.15 meters/second. Do not measure discharge in a pool.
    - "U" shaped, with a uniform streambed free of large boulders, woody debris or brush, and dense aquatic vegetation.
    - Flow is relatively uniform, with no eddies, backwaters, or excessive turbulence.

2. Lay the surveyor's rod (or stretch a measuring tape) across the stream perpendicular to its flow, with the "zero" end of the rod or tape on the left bank, as viewed when looking downstream. Leave the tape tightly suspended across the stream, approximately one foot above water level.

3. Attach the velocity meter probe to the calibrated wading rod. Check to ensure the meter is functioning properly and the correct calibration value is displayed. Calibrate (or check the calibration) the velocity meter and probe as directed in the meter's operating manual. Fill in the "VELOCITY AREA" circle on the Stream Discharge Form.

4. Divide the total wetted stream width into 15 to 20 equal-sized intervals. To determine interval width, divide the width by 20 and round up to a convenient number. Intervals should not be less than 10 cm wide, even if this results in less than 15 intervals. The first interval is located at the left margin of the stream (left when looking downstream), and the last interval is located at the right margin of the stream (right when looking downstream).

5. Stand downstream of the rod or tape and to the side of the first interval point (closest to the left bank if looking downstream).

6. Place the wading rod in the stream at the interval point and adjust the probe or propeller so that it is at the water surface. Fill in the appropriate "Distance Units" and "Depth Units" circles on the Stream Discharge Form. Record the distance from the left bank and the depth indicated on the wading rod on the Stream Discharge Form.

    Note for the first interval, distance equals 0 cm, and in many cases depth may also equal 0 cm. For the last interval, distance will equal the wetted width (in cm) and depth may again equal 0 cm.

7. Stand downstream of the probe or propeller to avoid disrupting the stream flow. Adjust the position of the probe on the wading rod so it is at 0.6 of the measured depth below the surface of the water. Face the probe upstream at a right angle to the cross-section, even if local flow eddies hit at oblique angles to the cross-section.

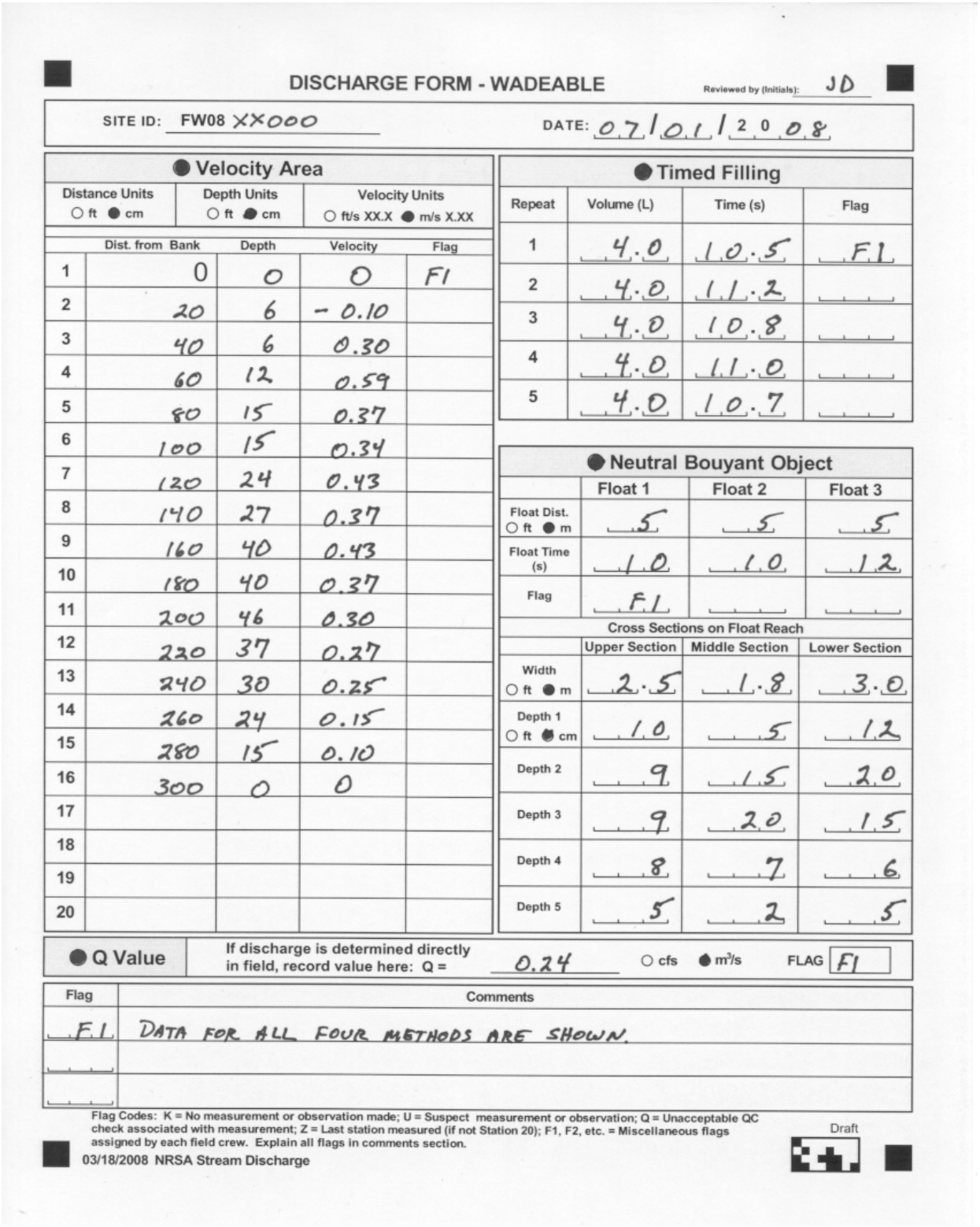
8. Wait 20 seconds to allow the meter to equilibrate, then measure the velocity. Fill in the appropriate "Velocity Units" circle on the Stream Discharge Form. Record the value on the Stream Discharge Form. Note for the first interval, velocity may equal 0 because depth will equal 0.

    - For the electromagnetic current meter (e.g., Marsh-McBirney), use the lowest time constant scale setting on the meter that provides stable readings.
    - For the impeller-type meter (e.g., Swoffer 2100), set the control knob at the mid-position of "DISPLAY AVERAGING". Press "RESET" then "START" and proceed with the measurements.

9. Move to the next interval point and repeat Steps 6 through 8. Continue until depth and velocity measurements have been recorded for all intervals. Note for the last interval (right margin), depth and velocity values may equal 0.

10. At the last interval (right margin), record a "Z" flag on the field form to denote the last interval sampled.

11. If using a meter that computes discharge directly, check the "Q" circle on the discharge form, and record calculated discharge value. In this case, you do not have to record the depth and velocity data for each interval.



DISCHARGE FORM - WADEABLE

Reviewed by (Initials): JD

SITE ID: FW08 XX000 DATE: 07/01/2008

Velocity Area

Distance Units ○ ft ● cm | Depth Units ○ ft ● cm | Velocity Units ○ ft/s XX.X ● m/s X.XX

	Dist. from Bank	Depth	Velocity	Flag
1	0	0	0	F1
2	20	6	- 0.10	
3	40	6	0.30	
4	60	12	0.59	
5	80	15	0.37	
6	100	15	0.34	
7	120	24	0.43	
8	140	27	0.37	
9	160	40	0.43	
10	180	40	0.37	
11	200	46	0.30	
12	220	37	0.27	
13	240	30	0.25	
14	260	24	0.15	
15	280	15	0.10	
16	300	0	0	
17				
18				
19				
20				

Timed Filling

Repeat	Volume (L)	Time (s)	Flag
1	4.0	10.5	F1
2	4.0	11.2	
3	4.0	10.8	
4	4.0	11.0	
5	4.0	10.7	

Neutral Bouyant Object

	Float 1	Float 2	Float 3
Float Dist. ○ ft ● m	5	5	5
Float Time (s)	10	10	12
Flag	F1		

Cross Sections on Float Reach

	Upper Section	Middle Section	Lower Section
Width ○ ft ● m	2.5	1.8	3.0
Depth 1 ○ ft ● cm	10	5	12
Depth 2	9	15	20
Depth 3	9	20	15
Depth 4	8	7	6
Depth 5	5	2	5

Q Value If discharge is determined directly in field, record value here: Q = 0.24 ○ cfs ● m^3/s FLAG F1

Flag	Comments
F1	DATA FOR ALL FOUR METHODS ARE SHOWN.

Flag Codes: K = No measurement or observation made; U = Suspect measurement or observation; Q = Unacceptable QC check associated with measurement; Z = Last station measured (if not Station 20); F1, F2, etc. = Miscellaneous flags assigned by each field crew. Explain all flags in comments section.

03/18/2008 NRSA Stream Discharge

Draft

Figure 6.2-20. Discharge Form, showing data recorded for all discharge measurement procedures.

6.2.6.5 Timed Filling Procedure

In channels too "small" for the velocity-area method, discharge can sometimes be measured by filling a container of known volume and timing the duration to fill the container.

"Small" is defined as a channel so shallow that the current velocity probe cannot be placed in the water, or where the channel is broken up and irregular due to rocks and debris, and a suitable cross-section for using the velocity area procedure is not available. This can be an extremely precise and accurate method, but requires a natural or constructed spillway of freefalling water. If obtaining data by this procedure will result in a lot of channel disturbance or stir up a lot of sediment, wait until after all biological and chemical measurements and sampling activities have been completed.

Choose a cross-section of the stream that contains one or more natural spillways or plunges that collectively include the entire stream flow. A temporary spillway can also be constructed using a portable V-notch weir, plastic sheeting, or other materials that are available onsite. Choose a location within the sampling reach that is narrow and easy to block when using a portable weir. Position the weir in the channel so that the entire flow of the stream is completely rerouted through its notch (Figure 6-3). Impound the flow with the weir, making sure that water is not flowing beneath or around the side of the weir. Use mud or stones and plastic sheeting to get a good waterproof seal. The notch must be high enough to create a small spillway as water flows over its sharp crest.

The timed filling procedure is presented in Table 6.2-16. Make sure that the entire flow of the spillway is going into the bucket. Record the time it takes to fill a measured volume on the Discharge Measurement Form as shown in Figure 6-2. Repeat the procedure 5 times. If the cross-section contains multiple spillways, you will need to do separate determinations for each spillway. If so, clearly indicate which time and volume data replicates should be averaged together for each spillway; use additional Stream Discharge Form if necessary.

Table 6.2-16. Timed filling procedure for determining stream discharge

NOTE: If measuring discharge by this procedure will result in significant channel disturbance or will stir up sediment, delay determining discharge until all biological and chemical measurement and sampling activities have been completed.

1. Choose a cross-section that contains one or more natural spillways or plunges, or construct a temporary one using on-site materials, or install a portable weir using a plastic sheet and on-site materials.
2. Fill in the "TIMED FILLING" circle in the stream discharge section of the Stream Discharge Form.
3. Position a calibrated bucket or other container beneath the spillway to capture the entire flow. Use a stopwatch to determine the time required to collect a known volume of water. Record the volume collected (in liters) and the time required (in seconds) on the Stream Discharge Form.
4. Repeat Step 3 a total of 5 times for each spillway that occurs in the cross-section. If there is more than one spillway in a cross-section, you must use the timed-filling approach on all of them. Additional spillways may require additional data forms

6.2.6.6 Neutrally-Buoyant Object Procedure

In very small, shallow streams with no waterfalls, where the standard velocity-area or timed-filling methods cannot be applied, the neutrally buoyant object method may be the only way to obtain an estimate of discharge. The required pieces of information are the mean flow velocity in the channel and the cross-sectional area of the flow. The mean velocity is estimated by measuring the time it takes for a neutrally buoyant object to flow through a measured length of the channel. The channel cross-sectional area is determined from a series of depth measurements along one or more channel cross-sections. Since the discharge is the product of mean velocity and channel cross-sectional area, this method is conceptually very similar to the standard velocity-area method.

The neutrally buoyant object procedure is described in Table 6.2-17. Examples of suitable objects include plastic golf balls (with holes), small sponge rubber balls, or small sticks. The object must float, but very low in the water. It should also be small enough that it does not "run aground" or drag bottom. Choose a stream segment that is roughly uniform in cross-section, and that is long enough to require 10 to 30 seconds for an object to float through it. Select one to three cross-sections to represent the channel dimensions within the segment, depending on the variability of width and/or depth. Determine the stream depth at 5 equally spaced points at each cross-section. Three separate times, measure the time required for the object to pass through the segment that includes all of the selected cross-sections. Record data on the Stream Discharge Form as shown in Figure 6.2-20.

Table 6.2-17. Neutrally buoyant object procedure for determining stream discharge

1. Fill in the "NEUTRALLY BUOYANT OBJECT" circle on the Stream Discharge Form.
2. Select a segment of the sampling reach that is deep enough to float the object freely, and long enough that it will take between 10 and 30 seconds for the object to travel. Mark the units used and record the length of the segment in the "FLOAT DIST." field of the Stream Discharge Form.
3. If the channel width and/or depth change substantially within the segment, measure widths and depths at three cross-sections, one near the upstream end of the segment, a second near the middle of the segment, and a third near the downstream end of the segment.

   If there is little change in channel width and/or depth, obtain depths from a single "typical" cross-section within the segment.
4. At each cross-section, measure the wetted width using a surveyor's rod or tape measure, and record both the units and the measured width on the Stream Discharge Form. Measure the stream depth using a wading rod or meter stick at points approximately equal to the following proportions of the total width: 0.1, 0.3, 0.5, 0.7, and 0.9. Record the units and the depth values (not the distances) on the Stream Discharge Form.
5. Repeat Step 4 for the remaining cross-sections.
6. Use a stopwatch to determine the time required for the object to travel through the segment. Record the time in the "FLOAT TIME" field of the Stream Discharge Form.
7. Repeat Step 6 two more times. The float time may differ somewhat for the three trials.

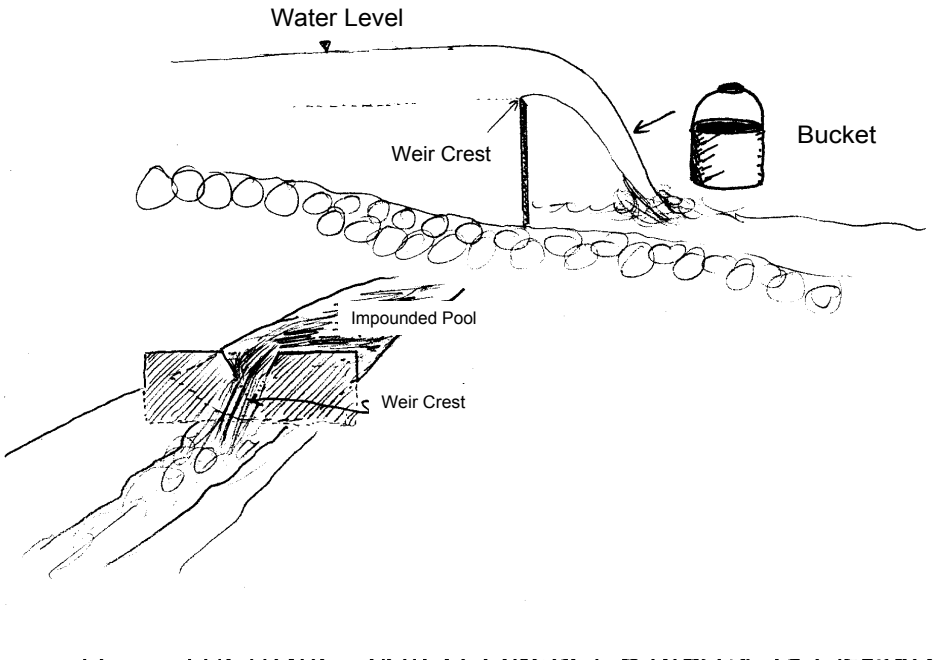


Figure 6.2-21. Use of a portable weir in conjunction with a calibrated bucket to obtain an estimate of stream discharge.

6.2.7 Equipment and Supplies

Table 6.2-18 lists the equipment and supplies required to conduct all the activities described for characterizing physical habitat. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the stream.

Table 6.2-18. Checklist of equipment and supplies for physical habitat

For taking measurements	▪ Surveyor's telescoping leveling rod (round profile, metric scale, 7.5 m extended) ▪ 50 m or 100 m measuring tape & reel ▪ Laser rangefinder (400 ft. distance range) and clear waterproof bag ▪ Digital camera with extra memory card & battery ▪ Two ½-inch diameter PVC pipe, 2-3 m long: Two of these, each marked at the same height (for use in slope determinations involving two persons) ▪ Meter stick, or a short rod or pole (e.g., a ski pole) with cm markings for thalweg measurements, or the PVC pipe for slope determinations can be marked in cm ▪ 1 roll each colored surveyor's flagging tape (2 colors) ▪ Convex spherical canopy densiometer (Lemmon Model A), modified with taped "V" ▪ Clinometer ▪ Bearing compass (Backpacking type) ▪ Binoculars ▪ 1 or 2 fisherman's vest with lots of pockets and snap fittings. Used to hold the various measurement equipment (densiometer, clinometer, compass, etc.). ▪ 2 pair chest waders (hip waders can be used in shallower streams). ▪ Current velocity meter, probe, and operating manual ▪ Top-set wading rod for use with current velocity meter ▪ Portable Weir with 60° "V" notch (optional) and plastic sheeting to use with weir ▪ Plastic bucket (or similar container) with volume graduations ▪ Stopwatch ▪ Neutrally buoyant object (e.g., plastic golf ball with holes, small rubber ball, stick) ▪ Field Methods Manual and/or laminated quick reference guide
For recording data	▪ Covered clipboards (lightweight, with strap or lanyard) ▪ Soft (#2) lead pencils (mechanical are acceptable) ▪ 11 plus extras Channel/Riparian Cross-section Forms ▪ 11 plus extras Thalweg Profile and Woody Debris Forms ▪ 1+ extras field Form: Stream Verification Form ▪ 1+ extras field Form: Field Measurement Form ▪ 1+ extras field Form: Discharge Form ▪ 1+ extras field Form: Sample Collection Form ▪ 1+ extras field Form: Riparian "Legacy" Trees and Invasive Alien Plants ▪ 1+ extras field Form: Channel Constraint ▪ 1+ extras field Form: Torrent Evidence Form ▪ 1+ extras field Form: Fish Gear and Voucher/Tissue Information Form ▪ 1+ extras field Form: Fish Collection Form ▪ 1+ extras field Form: Slope and Bearing Form ▪ 1+ extras field Form: Visual Assessment Form

6.3 Periphyton

6.3.1 Summary of Method

Collect periphyton from the 11 cross-section transects ("A" through "K") established within the sampling reach. Collect periphyton samples at the same time as sediment enzyme samples (Section 6.1.3) and benthic macroinvertebrate samples (Sections 6.4.1). Prepare one composite "index" sample of periphyton for each site. At the completion of the day's sampling activities, but before leaving the site, prepare four types of laboratory samples (an ID/enumeration sample to determine taxonomic composition and relative abundances, a chlorophyll sample, a biomass sample (for ash-free dry mass [AFDM]), and a acid/alkaline phosphatase activity [APA] sample) from the composite periphyton sample.

6.3.2 Equipment and Supplies

Table 6.3-1 is a checklist of equipment and supplies required to conduct periphyton sample collection and processing activities. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the river.

Table 6.3-1. Equipment and supplies list for periphyton at wadeable sites

For collecting samples	▪ Large Funnel (15-20 cm diameter) ▪ 12-cm^2 area delimiter (3.8 cm diameter pipe, 3 cm tall) ▪ Stiff-bristle toothbrush with handle bent at 90˚ angle ▪ 1-L wash bottle for stream water ▪ 500-mL plastic bottle for the composite sample ▪ 60-mL plastic syringe with 3/8" hole bored into the end ▪ Field Operations Manual or laminated Quick Reference Guide
For recording measurements	▪ Sample Collection Form ▪ Soft (#2) lead pencils for recording data on field forms ▪ Fine-tipped indelible markers for filling out sample labels ▪ Sample labels (4 per set) with the same Sample ID Number ▪ Clear tape strips for covering labels

6.3.3 Sampling Procedure

At each of the 11 transects, collect samples from the sampling station assigned during the layout of the reach. Collect the substrate selected for sampling from a depth no deeper than 0.5 m. If a sample cannot be collected because the location is too deep, skip the transect. The procedure for collecting samples and preparing a composite sample is presented in Table 6.3-2. Collect one sample from each of the transects and composite in one bottle to produce one composite sample for each site. Record the volume of the sample on the Sample Collection Form as shown in Figure 6.1-3.

Table 6.3-2. Procedure for collecting composite index samples of periphyton at wadeable sites

1. Starting with Transect "A", collect a single sample from the assigned sampling station using the procedure below.
   a) Collect a sample of substrate (rock or wood) that is small enough (< 15 cm diameter) and can be easily removed from the river. Place the substrate in a plastic funnel which drains into a 500-mL plastic bottle with volume graduations marked on it.
   b) Use the area delimiter to define a 12-cm^2 area on the upper surface of the substrate. Dislodge attached periphyton from the substrate within the delimiter into the funnel by brushing with a stiff-bristled toothbrush for 30 seconds. Take care to ensure that the upper surface of the substrate is the surface that is being scrubbed, and that the entire surface within the delimiter is scrubbed.
   c) Fill a wash bottle with river water. Using a minimal volume of water from this bottle, wash the dislodged periphyton from the funnel into the 500-mL bottle. If no coarse sediment (cobbles or larger) are present:
      - Use the area delimiter to confine a 12-cm^2 area of soft sediments.
      - Vacuum the top 1 cm of sediments from within the delimited area into a de-tipped 60-mL syringe.
      - Empty the syringe into the same 500-mL plastic bottle as above.
   d) Put the bottle in a cooler on ice while you travel between transects and collect the subsequent samples. (The samples need to be kept cool and dark because a chlorophyll sample will be filtered from the composite.)
2. Repeat Step 1 for transects "B" through "K". Place the sample collected at each sampling site into the single 500-mL bottle to produce the composite index sample.
3. If all 11 samples are not collected, record the number of transects collected and reason for any missed collection on the field forms.
4. After samples have been collected from all 11 transects, thoroughly mix the 500-mL bottle regardless of substrate type. Record the total estimated volume of the composite sample in the periphyton section of the Sample Collection Form.

6.3.4 Sample Processing in the Field

You will prepare four different types of laboratory samples from the composite index samples: an **ID/enumeration sample** (to determine taxonomic composition and relative abundances), a **chlorophyll sample**, a **biomass sample** (for ash-free dry mass [AFDM]), and an **acid/alkaline phosphatase activity** (APA) sample. All the sample containers required for an individual site should be sealed in plastic bags until use to avoid external sources of contamination (e.g., dust, dirt, or mud) that are present at site shorelines. Please refer to Sections 7.2.5 and 7.2.6 processing the periphyton samples.

6.4 Benthic Macroinvertebrates

6.4.1 Summary of Method

Collect benthic macroinvertebrate composite samples using a D-frame net with 500 µm mesh openings. Take the samples from the sampling stations at the 11 transects equally distributed along the targeted reach. You will proportionally sample multiple habitats at sampling stations randomly assigned on each transect. Multiple habitats will include bottom substrate as well as woody debris, macrophytes, and leaf packs. Composite all sample material and field-preserve with ~95% ethanol.

High gradient streams

- Primary samples are taken at each transect at either 25%, 50%, or 75% transect distance (according to the initial randomized pattern). Primary samples will be collected from a 1 square foot quadrat.

Low gradient streams

- Primary samples are taken at each transect at either 25%, 50%, or 75% transect distance (according to the initial randomized pattern). Primary samples will be collected from a 1 square foot quadrat.
- **additional**, separate samples taken at either 0%, 50%, or 100% transect distance to include edge samples (snags, undercut banks, root wads, macrophyte beds, etc.). Low gradient samples will be collected from a 1 linear meter sweep.

6.4.2 Equipment and Supplies

Table 6.4-1 shows the checklist of equipment and supplies required to complete the collection of benthic macroinvertebrates. This checklist is similar to the checklist presented in Appendix A, which is used at the base location to ensure that all of the required equipment is brought to the site. Record collection data on the Sample Collection Form (Fig. 6.1-2).

Table 6.4-1. Equipment and supplies list for benthic macroinvertebrate collection at wadeable sites

For collecting samples	▪ Modified kick net (D-frame with 500 µm mesh) and 4-ft handle ▪ Watch with timer or stopwatch ▪ Buckets, plastic, 8- to 10-qt ▪ Sieve bucket with 500 µm mesh openings (U.S. std No. 35) ▪ Watchmakers' forceps ▪ Wash bottle, 1-L capacity labeled "STREAM WATER" ▪ Funnel, with large bore spout	▪ Small spatula, spoon, or scoop to transfer sample ▪ Sample jars, 1-L HDPE plastic suitable for use with ethanol ▪ 95% ethanol, in a proper container ▪ Cooler (with absorbent material) for transporting ethanol & samples ▪ Plastic electrical tape ▪ Scissors ▪ Field Operations Manual or laminated Quick Reference Guide
For recording measurements	▪ Composite benthic sample labels with & without preprinted ID numbers	▪ Soft (#2) lead pencils ▪ Fine-tip indelible markers

	▪ Blank labels on waterproof paper for inside of jars	▪ Clear tape strips ▪ Sample Collection Form

6.4.3 Sampling Procedure

Figure 6.4-1 summarizes how samples will be collected from wadeable sites. The transect sample design for collecting benthic macroinvertebrates is shown in Figure 6.4-2. This design was used in the EPA's Wadeable Streams Assessment, which provides continuity for a nationwide assessment. Collect a sample from **1-m downstream** of each of the 11 cross-section transects at the assigned sampling station. The process for selecting the sample stations is described in the Initial Site Procedures Section (Section 4). At transects assigned a "Center" sampling point where the stream width is between one and two net widths wide, pick either the "Left" or "Right" sampling point instead. If the stream is only one net wide at a transect, place the net across the entire stream width and consider the sampling point to be "Center". If a sampling point is located in water that is too deep or unsafe to wade, select an alternate sampling point on the transect at random.

The procedure for collecting a sample at each transect is described in Table 6.4-2. At each sampling point, determine if the habitat is a "riffle/run" or a "pool/glide" (any area where there is not sufficient current to extend the net is operationally defined as a pool/glide habitat). Record the dominant substrate type (fine/sand, gravel, coarse substrate (coarse gravel or larger) or other (e.g., bedrock, hardpan, wood, aquatic vegetation, etc.) and the habitat type (pool, glide, riffle, or rapid) for each sample collected on the Sample Collection Form as shown in Figure 6.1-2. As you proceed upstream from transect to transect, combine all samples into a bucket. An **additional separate sample will be taken at low gradient streams** to include edge habitat (leaf litter, organic deposits, undercut banks, root wads, macrophyte beds, etc.)

6.4.4 Sample Processing in Field

Use a 500 μm mesh sieve bucket placed inside a larger bucket full of site water while sampling to carry the composite sample as you travel around the site. It is recommended that teams carry a sample bottle containing a small amount of ethanol with them to enable them to immediately preserve larger predaceous invertebrates such as helgramites and water beetles. Doing so will help reduce the chance that other specimens will be consumed or damaged prior to the end of the field day. Once the composite sample from all stations is sieved and reduced in volume, store in a 1-liter jar and preserve with 95% ethanol. Do not fill jars more than 1/3 full of material. Multiple jars may be required if detritus is heavy (Table 6.4-3). Try to use no more than 5 jars per site. If more than one jar is used for a composite sample, use the "extra jar" label provided; record the SAME sample ID number on this "extra jar" label. **DO NOT use two different sample numbers on two jars containing one single sample**. Cover the labels with clear tape. The sample ID number is also recorded with a No. 2 lead pencil on a waterproof label that is placed inside each jar. Be sure the inside label and outside label describe the same sample.

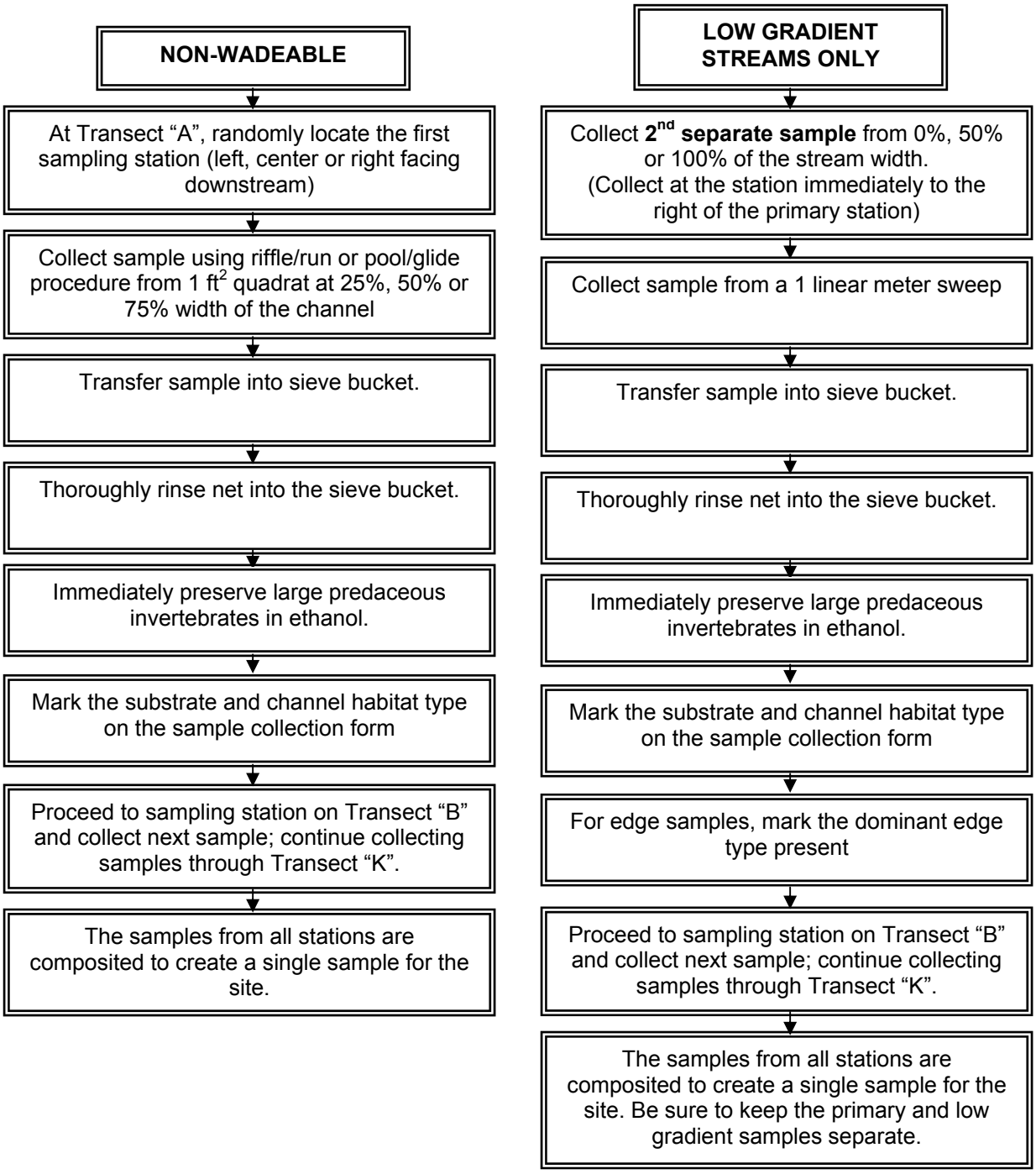


Figure 6.4-1. Benthic macroinvertebrate collection at wadeable sites.

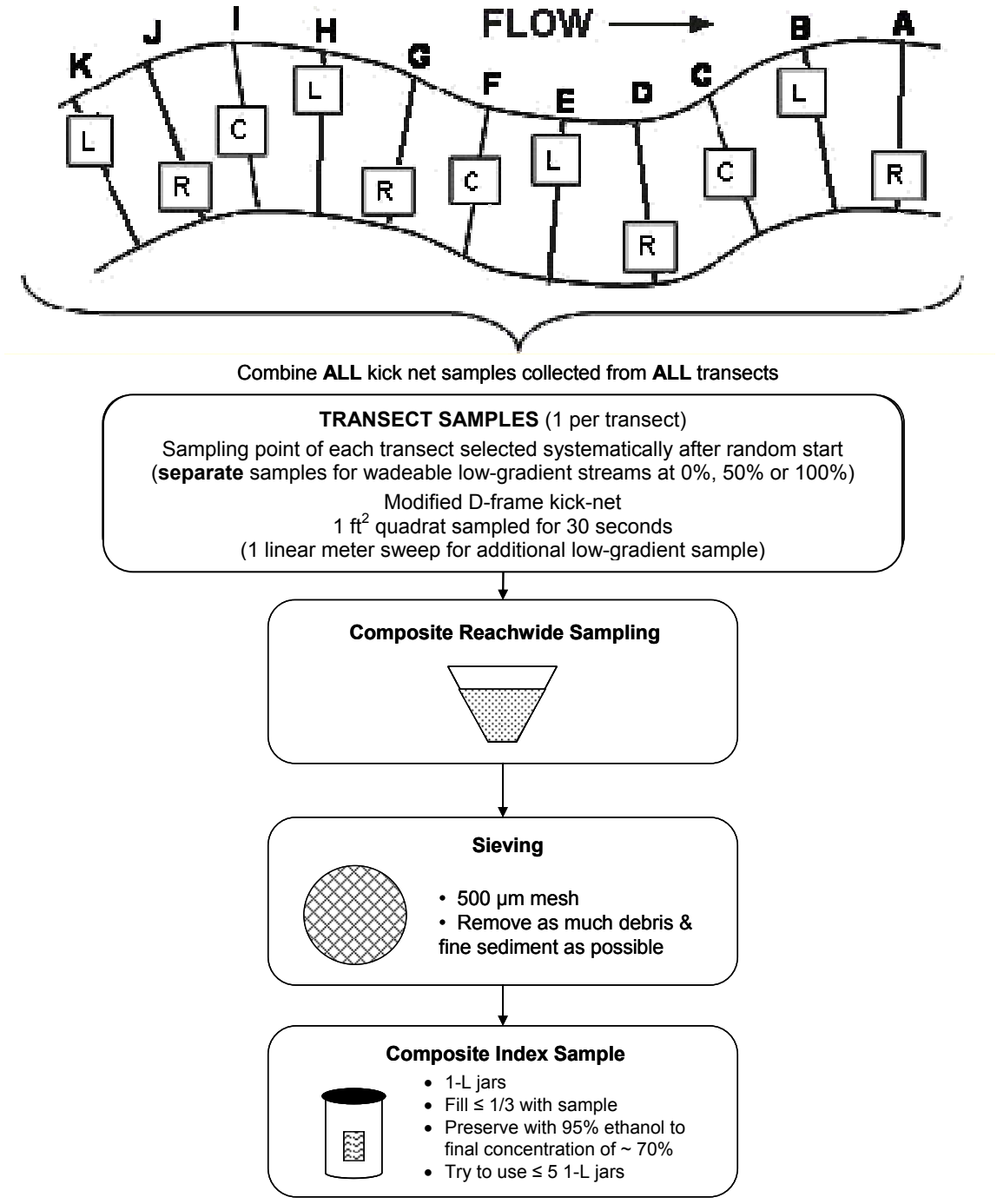


Figure 6.4-2. Transect sample design for collecting benthic macroinvertebrates at wadeable sites.

Table 6.4-2. Procedure for benthic macroinvertebrate sampling at wadeable sites

1. At **1 m downstream** of each transect, beginning with Transect "A", randomly locate the first sampling station (Left, Center, or Right as you face downstream) as 25%, 50%, and 75% of the wetted width, respectively. If you cannot collect a sample at the designated point because of deep water or unsafe conditions, relocate to another random point on the same transect.

2. Determine if there is sufficient current in the area at the sampling station to fully extend the net. If so, classify the habitat as "riffle/run" and proceed to Step 3. If not, use the sampling procedure described for "pool/glide" habitats starting at Step 9.

   NOTE: If the net cannot be used, hand pick a sample for 30 seconds from about 1 ft^2 of substrate at the sampling point. For vegetation-choked sampling points, sweep the net through the vegetation within a 1 ft^2 quadrat for 30 seconds. Place this hand-picked sample directly into the sample container. Assign a "U" flag (non-standard sample) to the sample and indicate which transect(s) required the modified collection procedure in the comments section. Go to Step 13.

Riffle/Run Habitats:

3. With the net opening facing upstream, quickly position the net securely on the stream bottom to eliminate gaps under the frame. Avoid large rocks that prevent the net from seating properly on the stream bottom.

   NOTE: If there is too little water to collect the sample with the D-net, randomly pick up 10 rocks from the riffle and pick and wash the organisms off them into a bucket which is half full of water.

4. Holding the net in position on the substrate, visually define a quadrat that is one net width wide and long upstream of the net opening. The area within this quadrat is 1 ft^2

5. Check the quadrat for heavy organisms, such as mussels and snails. Remove these organisms by hand and place them into the net. Pick up loose rocks or other larger substrate particles in the quadrat. Use your hands or a scrub brush to dislodge organisms and wash them into the net. Scrub all rocks that are golf ball-sized or larger and which are halfway into the quadrat. After scrubbing, place the substrate particles outside of the quadrat.

6. Hold the D-net securely in position. Starting at the upstream end of the quadrat, vigorously kick the remaining finer substrate within the quadrat for 30 seconds (use a stopwatch).

   NOTE: For samples located within dense beds of long, filamentous aquatic vegetation (e.g., algae or moss), kicking within the quadrat may not be sufficient to dislodge organisms in the vegetation. Usually these types of vegetation are lying flat against the substrate due to current. Use a knife or scissors to remove ***only the vegetation that lies within the quadrat*** *(i.e., not entire strands that are rooted within the quadrat) and place it into the net.*

7. Pull the net up out of the water. Immerse the net in the stream several times to remove fine sediments and to concentrate organisms at the end of the net. Avoid having any water or material enter the mouth of the net during this operation.

8. Go to Step 13.

Pool/Glide Habitats:

9. Visually define a quadrat that is one net width wide and long at the sampling point. The area within this quadrat is 1 ft^2.

10. Check the quadrat for heavy organisms, such as mussels and snails. Remove these organisms by

hand and place them into the net. Pick up loose rocks or other larger substrate particles in the quadrat. Use your hands or a scrub brush to dislodge organisms and wash them into the net. Scrub all rocks that are golf ball-sized or larger and which are halfway into the quadrat. After scrubbing, place the substrate particles outside of the quadrat.

11. Vigorously kick the remaining finer substrate within the quadrat with your feet while dragging the net repeatedly through the disturbed area just above the bottom. Keep moving the net all the time so that the organisms trapped in the net will not escape. Continue kicking the substrate and moving the net for 30 seconds.

    NOTE: If there is too little water to use the kick net, stir up the substrate with your gloved hands and use a sieve with 500 µm mesh size to collect the organisms from the water in the same way the net is used in larger pools.

12. After 30 seconds, remove the net from the water with a quick upstream motion to wash the organisms to the bottom of the net.

All samples:

13. Invert the net into a sieve bucket and transfer the sample. Remove as much gravel as possible so that the organisms do not get damaged. Inspect the net for any residual organisms clinging to the net and deposit them into the bucket. Use forceps if necessary to remove organisms from the net. Carefully inspect any large objects (such as rocks, sticks, and leaves) in the bucket and wash any organisms found off of the objects and into the bucket before discarding the object. Remove as much detritus as possible without losing organisms.

14. Determine the **predominant** substrate size/type you within the sampling quadrat. Fill in the appropriate circle for the dominant substrate type for the transect on the Sample Collection Form.

    NOTE: If there are co-dominant substrate types, you may fill in more than one circle; note the co-dominants in the comments section of the form.

    - **F**ine/sand: not gritty (silt/clay/muck <0.06 mm diam.) to gritty, up to ladybug sized (2 mm)
    - **G**ravel: fine to coarse gravel (ladybug to tennis ball sized; 2 mm to 64 mm)
    - **C**oarse: Cobble to boulder (tennis ball to car sized; 64 mm to 4000 mm)
    - **O**ther: bedrock (larger than car sized; > 4000 mm), hardpan (firm, consolidated fine substrate), wood of any size, aquatic vegetation, etc.). Note type of "other" substrate in comments on field form.

15. Identify the habitat type where the sampling quadrat was located. Fill in the appropriate circle for channel habitat type for the transect on the Sample collection Form.

    - **P**ool; Still water; low velocity; smooth, glassy surface; usually deep compared to other parts of the channel
    - **GL**ide: Water moving slowly, with smooth, unbroken surface; low turbulence
    - **RI**ffle: Water moving, with small ripples, waves, and eddies; waves not breaking, and surface tension is not broken; "babbling" or "gurgling" sound.
    - **RA**pid: Water movement is rapid and turbulent; surface with intermittent "white water" with breaking waves; continuous rushing sound.

16. Thoroughly rinse the net before proceeding to the next sampling station. Proceed upstream to the

next transect (through Transect K, the upstream end of the reach) and repeat steps 1 - 16. Combine all kick net samples from riffle/run and pool/glide habitats into the bucket.

Additional Sample for low gradient streams:

17. At low gradient stream sites, an additional separate composite sample will be taken. The sample will be collected with the same methods above, with the following modifications:
18. Collect the samples at 0, 50, or 100% transect distance to include edge samples (collected from leaf litter, snags, organic deposits, undercut banks, root wads, macrophyte beds, etc.).
19. If the primary sample was collected at the Left at Transect A, collect the additional sample at the Center of Transect A, then continue with Right at Transect B, Left at Transect C, until you collect at every transect rotating through Left, Center, and Right.
20. Collect the samples over 1 linear meter. Vigorously disturb the bank or bottom habitat and quickly sweep the net to collect the loosened material.
21. Composite and label this sample separately from the first sample collected. This will be identified in the lab as two separate samples.
22. Write in the appropriate abbreviation for substrate & channel habitat type on the Sample Collection Form. For samples taken at the left or right edge of the transect, write in the appropriate abbreviation for the dominant edge type present.

Record information for each composite sample on the Sample Collection Form as shown in Figure 6.1-2(a). If a sample requires more than one jar, make sure the correct number of jars for the sample is recorded on the Sample Collection Form. **Do not fill out the collection form until you have collected (or confirmed at the site that you will collect) samples.** If forms are filled out before you arrive at the site, and then no samples are collected, a lot of time is wasted by others later trying to find samples that do not exist. Place the samples in a cooler or other secure container for transporting and/or shipping to the laboratory (see Appendix C).

Table 6.4-3. Procedure for preparing composite samples for benthic macroinvertebrates at wadeable sites

1. Pour the entire contents of the bucket into a sieve bucket with 500 µm mesh size. Remove any large objects and wash off any clinging organisms back into the sieve before discarding. Remove any inorganic material, such as cobble or rocks.
2. Using a wash bottle filled with river water, rinse all the organisms from the bucket into the sieve. This is the composite sample for the reach.
3. Estimate the total volume of the sample in the sieve and determine how large a jar will be needed for the sample (500-mL or 1-L) and how many jars will be required. Try to use no more than 5 jars per site.
4. Fill in a sample label with the Sample ID and date of collection. Attach the completed label to the jar and cover it with a strip of clear tape. Record the sample ID number for the composite sample on the Sample Collection Form. For each composite sample, make sure the number on the form matches the number on the label.
5. Wash the contents of the sieve to one side by gently agitating the sieve in the water. Wash the sample into a jar using as little water from the wash bottle as possible. Use a large-bore funnel if necessary. If

Table 6.4-3. Procedure for preparing composite samples for benthic macroinvertebrates at wadeable sites

the jar is too full pour off some water through the sieve until the jar is not more than 1/3 full, or use a second jar if a larger one is not available. Carefully examine the sieve for any remaining organisms and use watchmakers' forceps to place them into the sample jar.

- *If a second jar is needed, fill in a sample label that does not have a pre-printed ID number on it. Record the ID number from the pre-printed label prepared in Step 4 in the "SAMPLE ID" field of the label. Attach the label to the second jar and cover it with a strip of clear tape. Record the number of jars required for the sample on the Sample Collection Form.* ***Make sure the number you record matches the actual number of jars used.*** *Write "Jar N of X" on each sample label using a waterproof marker ("N" is the individual jar number, and "X" is the total number of jars for the sample).*

6. Place a waterproof label inside each jar with the following information written with a number 2 lead pencil:

- Site ID
- Type of sampler and mesh size used
- Name of site
- Date of collection
- Collectors initials
- Number of stations sampled
- Jar "N" of "X"

7. Completely fill the jar with 95% ethanol (no headspace). It is very important that sufficient ethanol be used, or the organisms will not be properly preserved. Existing water in the jar should not dilute the concentration of ethanol below 70%.

   NOTE: Composite samples can be transported back to the vehicle before adding ethanol if necessary. In this case, fill the jar with stream water, which is then drained using the net (or sieve) across the opening to prevent loss of organisms, and replace with ethanol.

8. Replace the cap on each jar. Slowly tip the jar to a horizontal position, then gently rotate the jar to mix the preservative. Do not invert or shake the jar. After mixing, seal each jar with plastic tape.

9. Store labeled composite samples in a container with absorbent material that is suitable for use with 70% ethanol until transport or shipment to the laboratory.

6.5 Fish

6.5.1 Summary of Method

The fish sampling method is designed to provide a representative sample of the fish community, collecting all but the rarest fish taxa inhabiting the site. It is assumed to accurately represent species richness, species guilds, relative abundance, and anomalies. The goal is to collect fish community data that will allow the calculation of an Index of Biotic Integrity (IBI) and Observed/Expected (O/E) models. Backpack or barge electrofishing is the preferred method. If electrofishing is not possible due to safety concerns, high turbidity, or extremes in conductivity, complete the "Not Fished" section of the field form and comment why.

Streams with mean wetted widths less than 12.5 m will be electrofished in their entirety, covering all available habitats. However, the time and effort necessary to sample reaches greater than or equal to 12.5 m wide is prohibitive in the context of the survey, thus sub-sampling is required. Sub-sampling is defined by 5-10 sampling zones, each starting at a transect. In all instances electrofishing in wadeable systems should proceed in an upstream

direction using a single anode. Identification and processing of fish should occur at the completion of each subreach.

6.5.2 Equipment and Supplies

Table 6.5-1 shows the checklist of equipment and supplies required to complete the fish assessment. This checklist is similar to the one presented in Appendix A, which is used at the base location to ensure that all of the required equipment is brought to the site. Record fish collection data on the Fish Collection Form, Side 1 (Fig. 6.5-1).

Table 6.5-1. Equipment and supplies — fish assessment at wadeable sites.

For collecting samples	▪ Electrofishing equipment (including variable voltage pulsator unit, wiring cables, generator, electrodes, dip nets, protective linesman gloves, boots, and necessary safety equipment) ▪ Extra electrofishing unit batteries ▪ Scientific collection permit ▪ Digital camera with extra memory card & battery ▪ 1 Laser rangefinder (optional) ▪ Linesman gloves	▪ 1 Scalpel for slitting open large fish before preservation. ▪ 1 container of 10% buffered formalin ▪ Several Leak-proof HDPE jars for fish voucher specimens (various sizes from 250 mL - 4 L) ▪ 2 non-conducting dip nets with 1/4” mesh 1 Minnow net for dipping small fish from live well ▪ 2 measuring boards (3 cm size classes) ▪ 1 set Fish ID keys ▪ Field Operations Manual and/or laminated Quick Reference Guide
For recording measurements	▪ Sample labels ▪ Sample Collection Form ▪ Clear tape strips	▪ Soft (#2) lead pencils ▪ Fine-tip indelible markers

FISH COLLECTION FORM - WADEABLE (FRONT)

Reviewed by (Initials): ________

SITE ID: FW08XX000   DATE: 07/01/2008   PAGE: 1 of 1

Tag No.	Common Name	Tally	Total Count	Vouch. Count	LENGTH (mm)* Min	LENGTH (mm)* Max	Anom. Count	Mortality Count	Photo(s) Taken	Final Count	Flag	Subreaches (Optional)
01	MOTTLED SCULPIN	卌 卌 卌 卌 卌 卌 卌 ||	37	20	70	95	0	0	○ Y			A, D, F
02	CUTTHROAT TROUT	卌 卌 卌 |	16	3	60	200	0	0	○ Y			A, B, C, D, E, F, G, H, I, J
03	SPECKLED DACE	卌 |||	8	8	60	70	0	0	○ Y			B, C, D, E, F
04	UNKNOWN SUCKER 1	|||	3	3	40	40	1	3	○ Y		F1	C
05	UNKNOWN SUCKER 2	卌 卌 ||	12	12	45	50	0	1	○ Y		F2	C, E
06	SPECKLED DACE	卌 卌 卌	15	15	80	85	0	0	○ Y			G, H
07	TUI CHUB	||	2	0	80	90	0	0	● Y			G
08	NORTHERN PIKEMINNOW	卌 卌 |||	13	5	90	190	2	2	○ Y			F, I, J
09	MOTTLED SCULPIN	卌 ||	7	0	40	85	0	0	○ Y			H, I
10	UNKNOWN LAMPREY	|	1	1	50		0	0	○ Y		F3	J
									○ Y			
									○ Y			

Flag	Comment
F1	MAY BE A MINNOW
F2	TOO SMALL TO ID IN FIELD
F3	MAY BE RIVER LAMPREY

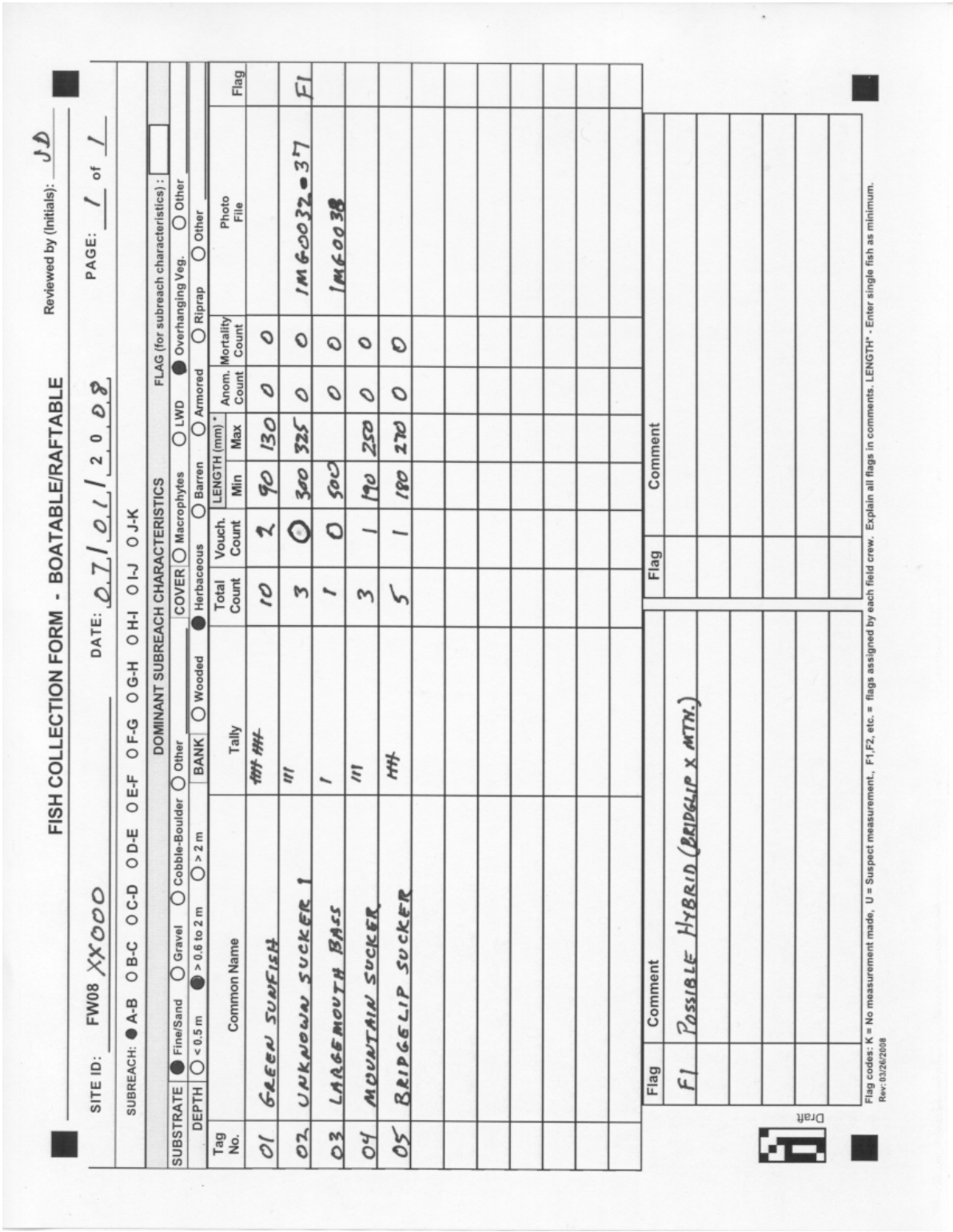
Tag	Photo File
07	IMG-0039
07	IMG-0040
07	IMG-0041
07	IMG-0042

Draft

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = flags assigned by each field crew. Explain all flags in comments. LENGTH* - Enter single fish as minimum.

4/21/2008 Fish Collection Form -Wadeable

Figure 6.5-1. Fish Collection Form for Small Wadeable Streams, Side 1.



FISH COLLECTION FORM - BOATABLE/RAFTABLE

Reviewed by (Initials): JD

SITE ID: FW08 XX000 DATE: 07/01/2008 PAGE: 1 of 1

SUBREACH: ● A-B ○ B-C ○ C-D ○ D-E ○ E-F ○ F-G ○ G-H ○ H-I ○ I-J ○ J-K

DOMINANT SUBREACH CHARACTERISTICS FLAG (for subreach characteristics):

SUBSTRATE ● Fine/Sand ○ Gravel ○ Cobble-Boulder ○ Other COVER ○ Macrophytes ○ LWD ● Overhanging Veg. ○ Other

DEPTH ○ < 0.5 m ● > 0.6 to 2 m ○ > 2 m BANK ○ Wooded ● Herbaceous ○ Barren ○ Armored ○ Riprap ○ Other

Tag No.	Common Name	Tally	Total Count	Vouch. Count	LENGTH (mm)* Min	LENGTH (mm)* Max	Anom. Count	Mortality Count	Photo File	Flag
01	GREEN SUNFISH	////// ///	10	2	90	130	0	0		
02	UNKNOWN SUCKER 1	///	3	0	300	325	0	0	IMG0032-37	F1
03	LARGEMOUTH BASS	/	1	0	500		0	0	IMG0038	
04	MOUNTAIN SUCKER	///	3	1	190	250	0	0		
05	BRIDGELIP SUCKER	////	5	1	180	270	0	0		

Flag	Comment
F1	POSSIBLE HYBRID (BRIDGLIP x MTN.)

Flag	Comment

Draft

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = flags assigned by each field crew. Explain all flags in comments. LENGTH* - Enter single fish as minimum.

Rev: 03/26/2008

Figure 6.5-2. Fish Collection Form for Large Wadeable Streams (Subreach A-B).

6.5.3 Sampling Procedure

At sites with a total reach length <500m, fishing will occur continuously for all habitats along the entire sample reach (40 times the average stream width), regardless of catch. At sites with a total reach length >500 m, sampling is accomplished using subreaches so that effort is distributed along the entire reach. In these streams, electrofishing will occur in sample zones beginning the zero mark at each transect on alternating banks (Figure 6.5-3). Determination of the initial stream bank sampling location at transect A (i.e., right or left bank) is determined at random. The crew should consist of one electrofishing operator, and one dip netter and an optional bucket carrier (who may also have a net to aid in transferring fish to the livewell). Sampling will proceed in an upstream direction from transect to transect.

The total reach extent fished in large wadeable streams (≥12.5 m) is a minimum reach length of 20 times the average stream width (20X) and a maximum reach length of 40 times the average stream width (40X). The subsampling routine is similar to boatables. Fish each subreach for a maximum of 700 seconds or until the next transect is reached. Begin sampling at a randomly determined bank at the beginning of the subreach and fish an area approximately 8m wide in an upstream direction. Fish the subreach thoroughly, covering bank habitat as well as midstream habitat for a maximum of 700 seconds. When 700 seconds are reached, stop electrofishing unless you are "pushing" a large school of fish, in which case continue fishing until you capture them (typically at some form of structure or physical barrier). At a minimum, 5 subreaches or 20 times the mean channel width is sampled. If 500 individuals are caught within this 20X, you may stop sampling. If not, continue sampling subreaches on alternating banks until 500 individuals are captured. Crews must complete each of the additional subreaches as described above, do not stop in the middle of any subreach, even if the 500 fish minimum is attained before the end of the subreach. To reduce stress and mortality, immobilized fish should be netted immediately and deposited into a live-well for processing. For safety, all crew members are required to wear non-breathable waders and insulated gloves. Polarized sunglasses and caps to aid vision are also required. Table 6.5-2 presents the procedure for electrofishing in wadeable streams.

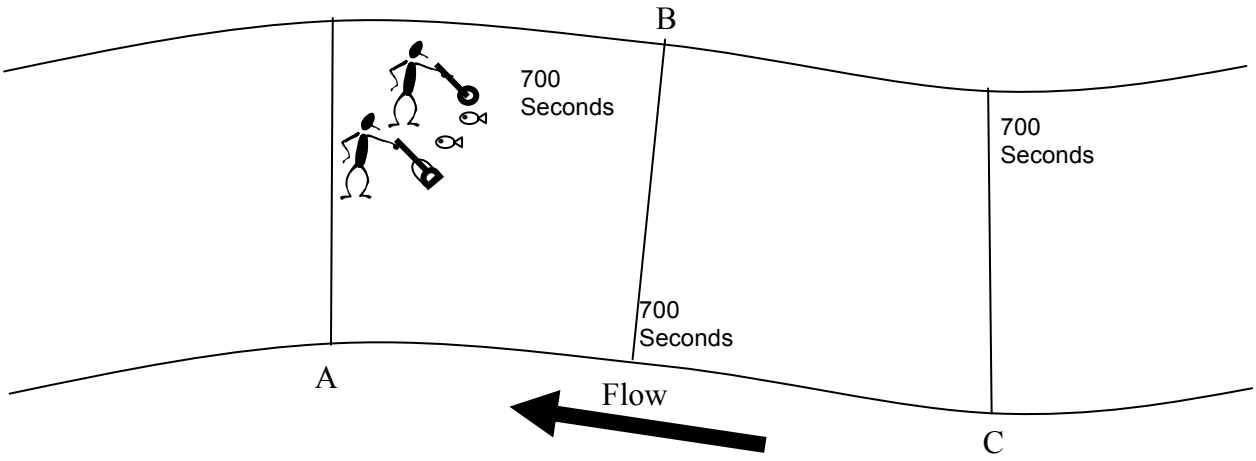


Figure 6.5-3. Transect sample design for fish sampling at wadeable sites ≥500 m (≥12.5m width).

Table 6.5-2. Procedure for electrofishing at wadeable sites <500 m

1. Review all collecting permits to determine if any sampling restrictions are in effect for the site. In some cases, you may have to cease sampling if you encounter certain listed species.
2. Search for fish even if the stream is extremely small, and it appears that sampling may produce no specimens. If none are collected, check the "NONE COLLECTED" circle on the Fish Collection Form. Explain why in comments section. Although not required, you may note amphibians and reptiles captured in the Comments.
3. Backpack and barge tote electrofishing will be used in wadeable streams, and direction of fishing will be in an upstream manner. If you do not sample, complete the "NOT FISHED" field on the Fish Collection Form and comment why.
4. At sites with a total reach length **<500 m**, fishing will occur continuously for all habitats along the entire sample reach. No subsampling.
5. Set unit to pulsed DC. Select initial voltage setting (150-400 V for high conductivity [>300 S/cm]; 500-800 V for medium conductivity [100 to 300 S/cm]; 900-1100 V for low conductivity [< 100 S/cm] waters). In waters with strong-swimming fish (length >200 mm), use a pulse rate of 30 Hz with a pulse width of 2 m/sec. If mostly small fish are expected, use a pulse rate of 60-70 Hz. Start the electrofisher, set the timer, and depress the switch to begin fishing. If fishing success is poor, increase the pulse width first and then the voltage. Increase the pulse rate last to minimize mortality or injury to large fish. If mortalities occur, first decrease pulse rate, then voltage, then pulse width. Start cleared clocks. Note, some electrofishers do not meter all the requested header data; provide what you can. If button time is not metered, estimate it with a stop watch and flag the data.
6. Once the settings on the electrofisher are adjusted properly to sample effectively and minimize injury and mortality, begin sampling at the downstream end of the reach (Transect A) and fish in an upstream direction. Depress the switch and slowly sweep the electrode from side to side. Sample all habitats and available cut-bank and snag habitat as well. Move the anode wand into cover with the current off, turn the anode on when in the cover, and then remove the wand quickly to draw fish out. In fast, shallow water, sweep the anode and fish downstream into a net. Be sure that deep, shallow, fast, slow, complex, and simple habitats are all sampled. In stretches with deep pools, fish the margins of the pool as much as possible, being extremely careful not to step or slide into deep water. Keep the cathode near the anode if fish catch is low.
7. Depending upon crew size, there may be from 2 to 3 people fishing small wadeable sites. Crews may choose to have more than one person holding a net, but ***no more than one person should be netting at any one time***. For example, in a wide stream there may be a netter on both sides of an operator. As the operator moves the probe from the left bank to the right bank the netters will remain on one side or the other and only one netter will be actively netting at any one time. The same fishing effort can be accomplished with 1 netter moving from side to side with the probe.
8. The netter, with the net 1 to 2 ft from the anode, follows the operator, nets stunned individuals, and places them in a bucket.
9. Continue upstream until the next transect is reached. Process fish and/or change water after each subreach to reduce mortality and track sampling effort.
10. Complete header information on the Fish Collection Form Small Wadeable.
11. Repeat Steps 6 through 9 until the last subreach is finished.

Table 6.5-3. Procedure for electrofishing at wadeable sites >500 m

1. Review all collecting permits to determine if any sampling restrictions are in effect for the site. In some cases, you may have to cease sampling if you encounter certain listed species.
2. Search for fish even if the stream is extremely small, and it appears that sampling may produce no specimens. If none are collected, check the "NONE COLLECTED" circle on the Fish Collection Form. Explain why in comments section. Although not required, you may note amphibians and reptiles captured in the Comments.
3. Backpack and barge tote electrofishing will be used in wadeable streams, and direction of fishing will be in an upstream manner. If you do not sample, complete the "NOT FISHED" field on the Fish Collection Form and comment why.
4. Fishing will occur in sample zones of approximately 8M in width with the zero mark at each transect on alternating banks.
5. Set unit to pulsed DC. Select initial voltage setting (150-400 V for high conductivity [>300 S/cm]; 500-800 V for medium conductivity [100 to 300 S/cm]; 900-1100 V for low conductivity [< 100 S/cm] waters). In waters with strong-swimming fish (length >200 mm), use a pulse rate of 30 Hz with a pulse width of 2 m/sec. If mostly small fish are expected, use a pulse rate of 60-70 Hz. Start the electrofisher, set the timer, and depress the switch to begin fishing. If fishing success is poor, increase the pulse width first and then the voltage. Increase the pulse rate last to minimize mortality or injury to large fish. If mortalities occur, first decrease pulse rate, then voltage, then pulse width. Start cleared clocks. Note, some electrofishers do not meter all the requested header data; provide what you can. If button time is not metered, estimate it with a stop watch and flag the data.
6. Once the settings on the electrofisher are adjusted properly to sample effectively and minimize injury and mortality, begin sampling at the downstream end of the reach (Transect A). Randomly choose a bank on which to start and fish in an upstream direction within 8 M of the chosen bank. Depress the switch and slowly sweep the electrode from side to side sampling all habitats thoroughly and available cut-bank and snag habitat as well. Move the anode wand into cover with the current off, turn the anode on when in the cover, and then remove the wand quickly to draw fish out. In fast, shallow water, sweep the anode and fish downstream into a net. Be sure that deep, shallow, fast, slow, complex, and simple habitats are all sampled. In stretches with deep pools, fish the margins of the pool as much as possible, being extremely careful not to step or slide into deep water. Keep the cathode near the anode if fish catch is low.
7. ***When using a barge or pram, the minimum crew size for electrofishing is three***. The barge operator must remain actively at the control box and navigate the barge. The probe operator will use one probe. Depending upon crew size, there may be from 1 to 2 people additional crew members. Crews may choose to have more than one person holding a net, but ***no more than one person should be netting at any one time***. For example, in a wide stream there may be a netter on both sides of an operator. As the operator moves the probe from the left bank to the right bank the netters will remain on one side or the other and only one netter will be actively netting at any one time. The idle netter can assist the active netter by depositing fish into the live well. The same fishing effort can be accomplished with one netter moving from side to side with the probe.
8. Continue upstream for a maximum of 700 seconds. Process fish ***after each transect*** to reduce mortality and track sampling effort by transect.
9. Continue sampling subreaches at alternating banks until Transect F is reached. If less than 500 fish have been collected from the first five subreaches, continue sampling additional subreaches along alternating banks until 500 individuals are captured, or at a maximum, subreach J-K is finished. Crews must complete each of the additional subreaches as described above, do not stop in the middle of any subreach, even if the 500 fish minimum is attained before the end of the subreach.
10. Complete header information on the Fish Collection Form Large Wadeable/Boatable/Raftable.

6.5.4 Processing Fish

Processing of fish must be completed at the end of each transect; however, if fish show signs of stress (e.g., loss of righting response, gaping, gulping air, excessive mucus), change water or stop fishing and initiate processing. Similarly, State- and Federally-listed threatened or endangered species or large game fish should be processed and released as they are captured. If periodic processing is required, fish should be released in a location that prevents the likelihood of their recapture.

For streams <12.5 m wide, use the Fish Collection Form Small Wadeable. For streams ≥12.5 m wide, use the Fish Collection Form – Large Wadeable/Boatable/Raftable. Taxonomic identification and processing should only be completed on specimens greater than 25 mm total length and by crew members designated as "fish taxonomic specialists" by EPA regional coordinators. Fish are tallied by species, evaluated for maximum and minimum length, and examined for the presence of DELT (Deformities, Eroded Fins, Lesions and Tumors) anomalies. Common names of species should follow those established under the American Fisheries Society's publication, "Common and Scientific Names of Fishes from the United States, Canada and Mexico" (Nelson, et al. 2004). A list of species common to freshwater systems of the United States is presented in Appendix D.

Species not positively identified in the field should be separately retained (up to 20 individuals per species) for laboratory identification. Common names for retained species should be assigned as "unknown", followed by its common family name and sequential lettering to designate separate species (e.g., UNKNOWN SCULPIN A). For large wadeable streams, each transect has its own form. Following positive laboratory identification, field form information should be updated to reflect the actual species count and number in the Final Count field. For example, if a sample of 20 specimens of species A is later identified as 15 individuals of species A and 5 of species B, the Final Count of species A should be corrected by assigning 25% to species B and 75% to species A. Table 6.5-4 presents the procedure for processing fish.

Table 6.5-4. Procedure for processing fish at wadeable sites

1. Complete all header information accurately and completely. If no fish were collected, complete the "NONE COLLECTED" field on the Fish Collection Form.
2. Complete the information on the Fish Gear and Voucher/Tissue Sample Information Form.
3. For small wadeable streams (<12.5 m) use the Fish Collection Form - Small Wadeable. For large wadeable streams (≥12.5 m) use the Fish Collection Form – Large Wadeable/Boatable/ Raftable.
4. For small wadeables, use one form for the entire reach.
5. For large wadeables, use one form per subreach and indicate Subreach on form in "SUBREACH" Field.
6. Only identify and process individuals > 25mm in total length, ideally handling specimens only once. Record the common name on the first blank line in the "COMMON NAME" Field of the Fish Collection Form.
7. Fill in the Tag Number. The tag number is a number starting with 01 and continuing sequentially to a number equal to the total number of species collected within the entire sample reach. Each reoccurrence of a species within the entire reach should be assigned the same tag number as it was

assigned initially. For example, if a bluegill is assigned tag number 01 when processing fish from the first subreach, all bluegills from the other subreaches will also be assigned tag number 01. The purpose of the tag number is to connect species identifications with subsequent verification and voucher collections.

8. If a species cannot be positively identified, assign it a sequential tag number in the Tag Number Field and leave the "COMMON NAME" Field Blank. Flag this line and indicate in the "COMMENT" field its common family name (e.g., UNKNOWN SCULPIN A). Retain a maximum subsample of 20 individuals for in-house laboratory identification of Unknowns. Do not include the number of each species retained solely for in-house lab verification in the Voucher Count column of the fish collection form. This column is reserved only for those fish that are to be sent in for independent re-identification as part of a complete voucher collection.
9. Process species listed as threatened and endangered first and return individuals immediately to the stream. Photograph specimens for verification purposes if conditions permit and stress to individuals will be minimal. Indicate if photographed on Fish Collection Form. If individuals are killed, prepare them as verification specimens and preserve noting them in the "MORTALITY COUNT" field.
10. Tally the number of individuals of each species collected in the "TALLY" box on the Fish Collection Form and record the total number in the "COUNT" field on the form.
11. Measure the total length of the largest and smallest individual to provide a size range for the species. Record these values in the "LENGTH" area of the Fish Collection Form. For small wadeables, this is done for the entire reach. For large wadeables, this is recorded by transect.
12. Examine each individual for external anomalies and tally those observed. Identify external anomalies including missing organs (eye, fin), skeletal deformities, shortened operculum, eroded fins, irregular fin rays or scales, tumors, lesions, ulcerous sores, blisters, cysts, blackening, white spots, bleeding or reddening, excessive mucus, and fungus. After all of the individuals of a species have been processed, record the total number of individuals affected in the "ANOMALIES" area of the Fish Collection Form. For small wadeables, this is done for the entire reach. For large wadeables, this is recorded by transect
13. Record total number of mortalities in the "MORTALITY COUNT" field due to electrofishing or handling on the Fish Collection Form.
14. Follow the appropriate procedure to prepare voucher specimens and/or to select specimens for tissue samples. Release all remaining individuals so as to avoid their recapture.
15. For any line with a fish name on the Fish Collection Form, ensure that all spaces on that line are filled in with a number, even if it is zero.
16. Repeat Steps 1 through 10 for all other species and subreaches.

6.5.5 Taxonomic Quality Assurance/Quality Control

6.5.5.1 Sample Preservation

Fish retained for laboratory identification/verification or voucher purposes should be placed in a large sample jar containing a 10% buffered formalin solution in a volume equal to or greater than the total volume of specimens. Individuals larger than 200 mm in total length should be slit along the right side of the fish in the lower abdominal cavity to allow penetration of the solution.

Fish retained for laboratory identification or as vouchers should be preserved in the field following the precautions outlined in the MSDS. All personnel handling 10% buffered formalin must read the MSDS (Appendix D). **Formalin is a potential carcinogen and should be used with extreme caution, as vapors and solution are highly caustic and may cause severe irritation on contact with skin, eyes, or mucus membranes. Wear vinyl or nitrile gloves and safety glasses, and always work in a well-ventilated area.**

6.5.5.2 Laboratory Identification

Fish that are difficult to identify in the field should be kept for laboratory identification or to verify difficult field identifications. Table 6.5-5 outlines the laboratory identification process and completing the Fish Collection Form. Field crews may use a supplemental Fish Identification Lab sheet such as that shown in Figure 6.5-4 for internal laboratory use only. Crews should retain the Fish verification sample – contact your regional EPA coordinator if you cannot store the samples at your facility.

Do not include the number of each species retained solely for in-house lab verification in the Voucher Count column of the fish collection form. This column is reserved only for those fish that are to be sent in for independent re-identification as part of a complete voucher collection.

Field crews should not retain the Fish Collection Form(s) if the laboratory identification process cannot be completed within a short period of time. If the time needed to complete the identification/verification is expected to exceed two weeks, make copies of the Fish Collection Form(s) and send the entire pack of original data forms to the Information Management Coordinator. When the identification/verification process is complete, make the necessary changes to the copied Fish Collection Form(s) and send them as soon as possible to the Information Management Coordinator as well.

Table 6.5-5. Procedure for laboratory identification of fish samples.

1. Fish may be retained for routine laboratory identification and verification purposes. Fish tags are provided with each site kit. Crews may use these tags at their discretion in order to identify fish at their laboratory.
2. Retained fish should be placed in a large sample jar containing a 10% buffered formalin solution in a volume equal to or greater than the total volume of specimens. Individuals larger than 200mm in total length should be slit along the right side of the fish in the lower abdominal cavity to allow penetration of the solution.
3. Following fixation for 5 to 7 days, the volume of formalin should be properly discarded and replaced with tap water for soaking specimens over a 4-5 day period. Soaking may require periodic water changes and should continue until the odor of formalin is barely detectable. Final storage of specimens is done in 45%-50% isopropyl alcohol or 70% ethanol. Formalin is a potential carcinogen and should be used with extreme caution, as vapors and solution are highly caustic and may cause severe irritation on contact with skin, eyes, or mucus membranes. Wear vinyl or nitrile gloves and safety glasses, and always work in a well-ventilated area.
4. Formalin must be disposed of properly. Contact your regional EPA coordinator if your laboratory does not have the capability of handling waste formalin.
5. Unknown fish are identified to species in the laboratory. You may use a Fish Identification Lab Sheet such as the one presented in Figure 6.5-4.

6. Fill in the Unknown species name in the "COMMON NAME" field of the Fish Collection Form and make certain the "FINAL COUNT" field is correct.
7. If species field identifications were incorrect, correct the "COMMON NAME" Field by completely erasing the Common Name and replacing the correct name. Add an additional Common Name if needed. Make certain the "FINAL COUNT" field is correct. If the "COMMON NAME" Field was incorrect or cannot be cleanly erased, cross out the line of data and fill out a new line with the correct "COMMON NAME" and "FINAL COUNT".

6.5.5.3 Voucher Specimens

Approximately 10% of each field crews' sites will be randomly pre-selected for re-identification by an independent taxonomist. A minimum of one complete voucher is required for each person performing field taxonomy and will consist of either preserved specimen(s) or digital images representative of all species in the sample, including common species. Multiple specimens per species can be used as vouchers, if necessary (i.e., to document different life or growth stages, or sexes). Note that a complete sample voucher does not mean that all individuals of each species will be vouchered, only enough so that independent verification can be achieved.

Digital images should be taken as voucher documentation for species that are recognized as Rare, Threatened, or Endangered – they should not be killed. Digital images should also be taken of fish specimens too large for preservation.

Certain states or regions may require that more fish vouchers are taken. Check with your state/regional coordinators to determine if your team will be required to collect complete vouchers at more than 10% or your sites.

For the sample voucher, specimen containers should be labeled with the sample number, site ID number, site name, and collection date. There should be no taxonomic identification labels in or on the container, or in any of the digital photos.

Choose individual specimens that are intact and in good condition, such that re-identification will be possible. Fish that are damaged, have significant scale loss or those that have been dead for a significant amount of time prior to preservation should be avoided if possible. Fish in pristine condition and those possessing clear identification characteristics are preferred. Additionally, fish that are preserved while still live will typically flare their fins and gills and will allow for easier re-identification in the laboratory.

Place one or more representative specimens of each species in plastic mesh sleeves along with one of the corresponding tag number labels provided in your site kit. (Several fish may be placed in a single mesh sleeve, as long as they are of the same species). Ensure that the tag numbers in the voucher collection match the tag numbers on the fish collection data forms. Seal both ends of the mesh sleeve with zip ties and place it inside the voucher collection jar with the appropriate preservative. Unknown fish may be identified in the laboratory as described in section 5.5.5.2 and subsequently included in the voucher collection.

Record the total number of each fish species retained for voucher purposes in each subreach on the fish collection form. Record the voucher sample ID number on the fish gear /

voucher / fish tissue collection form. If no voucher is prepared for the site, fill in the "no vouchers preserved" circle on the fish gear form.

Table 6.5-6. Procedure for vouchering fish samples.

1. Approximately 10% of each field crews' sites will be randomly pre-selected for re-identification by an independent taxonomist. A minimum of one complete voucher is required for each person performing field taxonomy and will consist of either preserved specimen(s) and/or digital images representative of all species in the sample, even common species.
2. Take digital images as voucher documentation for species that are recognized as Rare, Threatened, or Endangered; or when fish specimens are too large for preservation.
3. For the sample voucher, label the specimen containers with the sample number, site ID number, site name, and collection date. Do not put taxonomic identification labels in or on the container.
4. Place one or more representative specimens of each species in plastic mesh sleeves along with one of the corresponding tag number labels provided in your site kit. (Several fish may be placed in a single mesh sleeve, as long as they are of the same species).
5. Ensure that the tag numbers in the voucher collection match the tag numbers on the fish collection data forms.
6. Seal both ends of the mesh sleeve with zip ties and place it inside the voucher collection jar with the appropriate preservative.
7. Unknown fish may be identified in the laboratory as described in section 5.5.5.2 and subsequently included in the voucher collection.
8. Record the total number of each fish species retained for voucher purposes in each subreach on the fish collection form.
9. Record the voucher sample ID number on the fish gear / voucher / fish tissue collection form.
10. If no voucher is prepared for the site, fill in the "no vouchers preserved" circle on the fish gear form.

6.5.5.4 Photovouchering

Digital imagery should be used for fish species that cannot be retained as preserved specimens (e.g., RTE species; very large bodied; or very common). Views appropriate and necessary for an independent taxonomist to accurately identify the specimen should be the primary goal of the photography. Additional detail for these guidelines is provided in Stauffer et al. (2001), and is provided to all field crews as a handout.

The recommended specifications for digital images to be used for photovouchering include: 16-bit color at a minimum resolution of 1024x768 pixels; macro lens capability allowing for images to be recorded at a distance of less than 4 cm; and built-in or external flash for use in low-light conditions. Specimens should occupy as much of the field of view as possible, and the use of a fish board is recommended to provide a reference to scale (i.e., ruler or some calibrated device) and an adequate background color for photographs. Information on Station ID, Site Name, Date and a unique species ID (i.e., A, B, C, etc.) should also be captured in the photograph, so that photos can be identified if file names become corrupted. All photovouchered species should have at least a full-body photo (preferably of the left side of the fish) and other zoom images as necessary for individual species, such as lateral line, ocular/oral orientation, fin rays, gill arches, or others. It may also be necessary to photograph males, females, or juveniles.

Images should be saved in medium- to high-quality jpeg format, with the resulting file name of each picture noted one the Fish Collection Form. It is important that time and date stamps are accurate as this information can also be useful in tracking the origin of photographs. It is recommended that images stored in the camera be transferred to a PC or storage device at the first available opportunity. At this time the original file should be renamed to follow the logic presented below:

F01_CT003_20080326_A.jpg

Where:
F = fish
01 = tag number
CT003 = state (Connecticut) and site number
20080326 = date (yyyymmdd)
A = first of several pictures of same fish (e.g., A, B, C)

Field crews should maintain files for the duration of the sampling season. Notification regarding the transfer of all images to the existing database will be provided at the conclusion of the sampling.

Fish Identification Lab Sheet

Site ID_____Date Collected____/____/_____Date(s) Identified____/____/_____ ID'd by_____
Preservative(Field/Lab)_____ Keys Used_____
Date Data Corrected on Field Sheet_____/_____/_____ Initials_____

Tag no.	Photo (P) or Specimen (S)	Common Name (Field)	Common Name (Lab)	Count	Transect (if known)	PhotoFile (Field)	PhotoFile (Final)

Figure 6.5-4. Fish Identification Lab Sheet.

6.6 Fecal Indicator (Enterococci)

6.6.1 Summary of Method

You will collect a fecal indicator sample at the last transect (Transect K) after all other sampling is completed. Use a pre-sterilized, 250 ml bottle and collect the sample approximately 1 m off the bank at about 0.3 meter (12 inches) below the water. Following collection, place the sample in a cooler, chill for at least 15 minutes, and maintain on ice prior to filtration of four 50 mL volumes. (Samples must be filtered and frozen on dry ice within 6 hours of collection). In addition to collecting the sample, look for signs of disturbance throughout the reach that would contribute to the presence of fecal contamination to the waterbody. Record these disturbances on the Site Assessment Form (Figure 7-2).

6.6.2 Equipment and Supplies

Table 6.6-1 provides the equipment and supplies needed for field crews to collect the fecal indicator sample. Record the fecal indicator sample data on the Sample Collection Form (Figure 6.1-3).

Table 6.6-1. Equipment and supplies list for fecal indicator sampling at wadeable sites

For collecting samples	▪ nitrile gloves ▪ pre-sterilized, 250 ml sample bottle ▪ sodium thiosulfate tablet ▪ Wet ice ▪ cooler
For recording measurements	▪ Sample Collection Form ▪ Site Assessment Form ▪ Fecal Indicator sample labels (4 vial labels and 1 bag label) ▪ Pencils (for data forms) ▪ Fine-tipped indelible markers (for labels) ▪ Clear tape strips

6.6.3 Sampling Procedure

Table 6.6-2 provides the procedure for collecting fecal indicator (i.e., Enterococci) samples at wadeable sites.

Table 6.6-2. Procedure for fecal indicator (Enterococci) sample collection at wadeable sites

Collect the Enterococci Sample

1. Put on nitrile gloves.
2. Select a sampling location at transect K that is approximately 1 m from the bank and approximately 1 m deep. Approach the sampling location slowly from downstream or downwind.
3. Lower the un-capped, inverted 250 ml sample bottle to a depth of 1 foot below the water surface, avoiding surface scum, vegetation, and substrates. Point the mouth of the container away from the

body or boat. Right the bottle and raise it through the water column, allowing bottle to fill completely. If the depth does not reach 1 foot along the transect at 1 m from the bank, take the sample and flag it on the field form.

4. After removing the container from the water, discard a small portion of the sample to allow for proper mixing before analyses.
5. Add the sodium thiosulfate tablet, cap, and shake bottle 25 times.
6. Store the sample in a cooler on ice to chill (not freeze). Chill for at least 15 minutes and do not hold samples longer than 6 hours before filtration and freezing.

7.0 FINAL SITE ACTIVITIES

The activities described in this section apply to both wadeable and non-wadeable sites. Prior to leaving the site, make a general visual assessment of the site and its surrounding catchment. The objective of the site assessment is to record observations of catchment and site characteristics that are useful for future data interpretation, ecological value assessment, development of associations, and verification of stressor data. Your observations and impressions are extremely valuable.

You will filter and process the fecal indicator, *chlorophyll a*, and periphyton samples. Conduct a final check of the data forms, labels and samples. The purpose of the second check of data forms, labels and samples is to assure completeness of all sampling activities. Finally, clean and pack all equipment and supplies, and clean the launch site and staging areas. After you leave the site, report the sampling event to the Information Management Coordinator, and ship or store the samples. Activities described in this section are summarized in Figure 7-1.

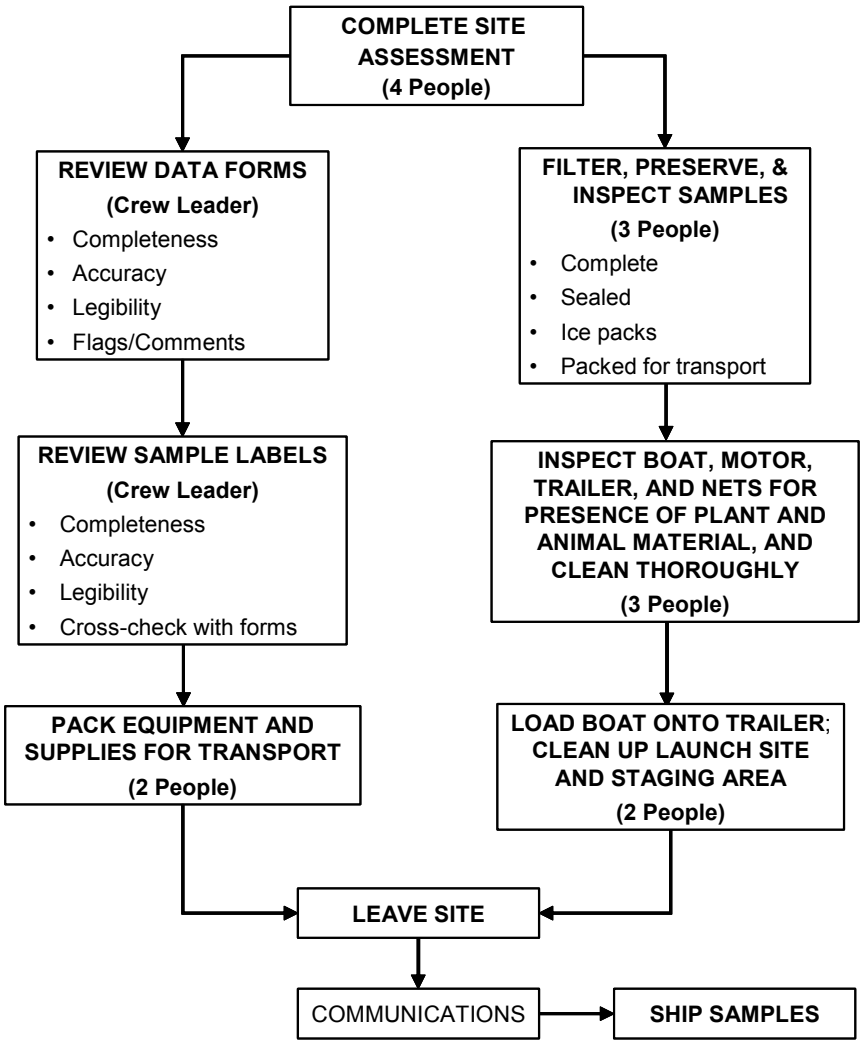


Figure 7.1. Final site activities summary.

7.1 General Site Assessment

Complete the Site Assessment Form (Figure 7-2) after sampling, recording all observations from the site that were noted during the course of the visit. This Site Assessment Form is designed as a template for recording pertinent field observations. It is by no means comprehensive, and any additional observations should be recorded in the General Assessment section.

7.1.1 Watershed Activities and Disturbances Observed

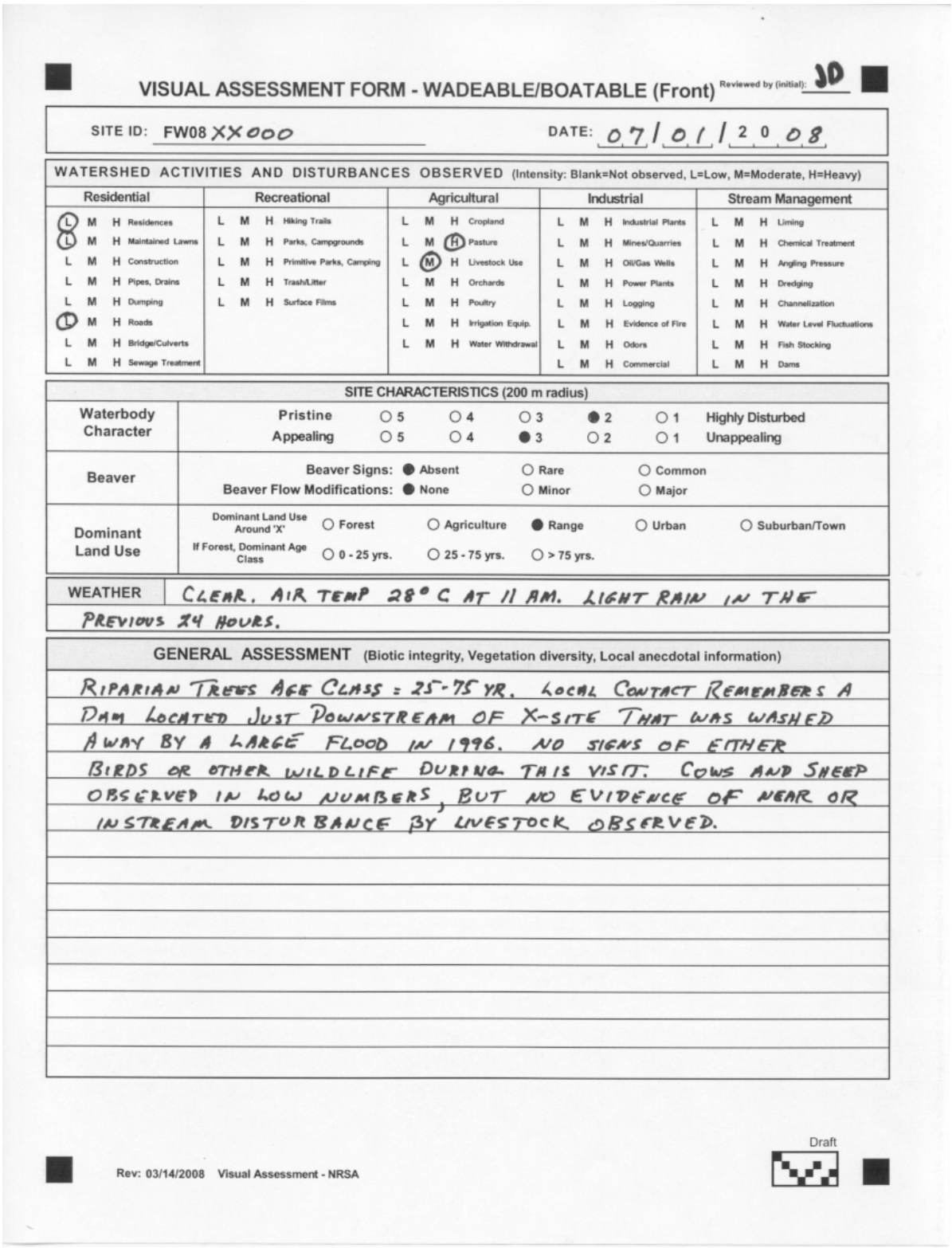
Record any of the sources of potential stressors listed in the "Watershed Activities and Disturbances Observed" section on the Site Assessment Form (Figure 7-2). Include those that were observed while on the site, while driving or walking through the site and catchment, or while flying over the site and catchment. For activities and stressors that you observe, rate their abundance or influence as low (L), moderate (M), or heavy (H) on the line next to the listed disturbance. Leave the line blank for any disturbance not observed. The distinction between low, moderate, and heavy will be subjective. For example, if there are two to three houses on a site, circle "L" for low next to "Houses." If the site is ringed with houses, rate it as heavy (H). Similarly, a small patch of clear-cut logging on a hill overlooking the site would rate a low ranking. Logging activity right on the site shore, however, would get a heavy disturbance ranking. This section includes residential, recreational, agricultural, industrial, and stream management categories.

7.1.2 Site Characteristics

Record observations regarding the general characteristics of the site on the Site Assessment Form (Figure 7-2). When assessing these characteristics, look at a 200 m riparian distance on both banks. Rank the site between "pristine" and "highly disturbed", and between "appealing" and "unappealing." Document any signs of beaver activity and flow modifications. Record the dominant land use and forest age class. Document the weather conditions on the day of sampling, and any extreme weather conditions just prior to sampling.

7.1.3 General Assessment

Record any additional information and observations in this narrative section. Information to include could be observations on biotic integrity, vegetation diversity, presence of wildlife, local anecdotal information, or any other pertinent information about the site or its catchment. Record any observations that may be useful for future data interpretation.



VISUAL ASSESSMENT FORM - WADEABLE/BOATABLE (Front) Reviewed by (initial): JD

SITE ID: FW08XX000 DATE: 07/01/2008

WATERSHED ACTIVITIES AND DISTURBANCES OBSERVED (Intensity: Blank=Not observed, L=Low, M=Moderate, H=Heavy)

Residential	Recreational	Agricultural	Industrial	Stream Management
(L) M H Residences	L M H Hiking Trails	L M H Cropland	L M H Industrial Plants	L M H Liming
(L) M H Maintained Lawns	L M H Parks, Campgrounds	L M (H) Pasture	L M H Mines/Quarries	L M H Chemical Treatment
L M H Construction	L M H Primitive Parks, Camping	L (M) H Livestock Use	L M H Oil/Gas Wells	L M H Angling Pressure
L M H Pipes, Drains	L M H Trash/Litter	L M H Orchards	L M H Power Plants	L M H Dredging
L M H Dumping	L M H Surface Films	L M H Poultry	L M H Logging	L M H Channelization
(L) M H Roads		L M H Irrigation Equip.	L M H Evidence of Fire	L M H Water Level Fluctuations
L M H Bridge/Culverts		L M H Water Withdrawal	L M H Odors	L M H Fish Stocking
L M H Sewage Treatment			L M H Commercial	L M H Dams

SITE CHARACTERISTICS (200 m radius)

Waterbody Character	Pristine	○ 5	○ 4	○ 3	● 2	○ 1	Highly Disturbed
	Appealing	○ 5	○ 4	● 3	○ 2	○ 1	Unappealing
Beaver	Beaver Signs:	● Absent	○ Rare	○ Common			
	Beaver Flow Modifications:	● None	○ Minor	○ Major			
Dominant Land Use	Dominant Land Use Around "X"	○ Forest	○ Agriculture	● Range	○ Urban	○ Suburban/Town	
	If Forest, Dominant Age Class	○ 0 - 25 yrs.	○ 25 - 75 yrs.	○ > 75 yrs.			

WEATHER: CLEAR. AIR TEMP 28° C AT 11 AM. LIGHT RAIN IN THE PREVIOUS 24 HOURS.

GENERAL ASSESSMENT (Biotic integrity, Vegetation diversity, Local anecdotal information)

RIPARIAN TREES AGE CLASS = 25-75 YR. LOCAL CONTACT REMEMBERS A DAM LOCATED JUST DOWNSTREAM OF X-SITE THAT WAS WASHED AWAY BY A LARGE FLOOD IN 1996. NO SIGNS OF EITHER BIRDS OR OTHER WILDLIFE DURING THIS VISIT. COWS AND SHEEP OBSERVED IN LOW NUMBERS, BUT NO EVIDENCE OF NEAR OR INSTREAM DISTURBANCE BY LIVESTOCK OBSERVED.

Draft

Rev: 03/14/2008 Visual Assessment - NRSA

Figure 7.2. Site Assessment Form.

7.2 Processing the Fecal Indicator, *Chlorophyll a*, and Periphyton Samples

7.2.1 Equipment and Supplies (Fecal Indicator)

Table 7-1 provides the equipment and supplies needed for field crews to collect the fecal indicator sample.

Table 7.1. Equipment and supplies list for fecal indicator sample

For processing samples	▪ Nitrile gloves ▪ sterile screw-cap 50-mL centrifuge tube ▪ Sterile filter holder, Nalgene 145/147 ▪ Vacuum pump (electric pump may be used if available) ▪ Sterile phosphate buffered saline (PBS) ▪ Osmotics 47 mm polycarbonate 0.4 µm sterile filters ▪ Sterile disposable forceps ▪ 4 sterile microcentrifuge tubes containing sterile glass beads ▪ Dry ice ▪ Cooler ▪ Field Operations Manual or laminated Quick Reference Guide
For recording measurements	▪ Sample Collection Form ▪ Soft (#2) lead pencils for recording data on field forms ▪ Fine-tipped indelible markers for filling out sample labels ▪ Fecal Indicator sample labels (4 vial labels and 1 bag label) ▪ Clear tape strips for covering labels

7.2.2 Procedures for Processing the Fecal Indicator Sample

The fecal indicator sample **must** be filtered **before** the *chlorophyll a* and periphyton samples, since the filtering apparatus needs to be sterile for this sample. The procedures for processing the fecal indicator sample are presented in Table 7-2. The sample must be filtered and frozen within 6 hours of collection.

Table 7.2. Processing procedure—fecal indicator sample

Processing procedure—fecal indicator <u>filter blank</u> (to be done at Revisit sites only)

Enterococci filter blanks will be prepared at all revisit sites during the first visit (see Fig. 8-1). Prepare the filter blanks **<u>before</u>** filtering the river sample.

1. Set up sample filtration apparatus using same procedure as used for the river sample. Chill Filter Extraction tubes with beads on dry ice.
2. Aseptically transfer 4 polycarbonate filters from filter box to base of opened Petri dish. Close filter box and set aside.
3. Remove cellulose nitrate (CN) filter (the filter with grid design on it) from funnel and discard. Be sure to leave the support pad in the filter funnel.
4. Load filtration funnel with sterile polycarbonate filter on support pad (shiny side up).

Table 7.2. Processing procedure—fecal indicator sample

5. Measure 10-mL of the chilled phosphate buffered saline (PBS) in the sterile graduated centrifuge tube and pour into filter funnel.
6. Replace cover on filter funnel and pump to generate a vacuum (do not generate more than 7 inches of Hg of pressure). Keep pumping until all liquid is in filtrate collection flask.
7. Remove filter funnel from base without disturbing filter. Using sterile disposable forceps remove the filter (touching only the filter edges) and fold it in half, in quarters, in eighths, and then in sixteenths (filter will be folded 4 times).
8. Insert filter into chilled filter extraction tube (with beads) open end down. Replace and tighten the screw cap, insert tube(s) into bubble wrap bag on dry ice for preservation during transport and shipping.
9. Label the samples as "blank" on the label and field form, and package and submit these samples to the lab with the standard samples.
10. Repeat steps 4 to 9 for the remaining three 10-mL volumes of PBS to be filtered.

Processing procedure—fecal indicator samples (All sites)

1. Put on nitrile gloves.
2. Set up sample filtration apparatus on flat surface and attach vacuum pump. Set-out 50-mL sterile centrifuge tube, sterile 60-mm Petri dish, 2 bottles of chilled phosphate buffered saline (PBS), Osmotics 47 mm polycarbonate sterile filter box, and 2 filter forceps.
3. Chill Filter Extraction tubes with beads on dry ice.
4. Aseptically transfer 4 polycarbonate filters from filter box to base of opened Petri dish. Close filter box and set aside.
5. Remove cellulose nitrate (CN) filter (the filter with grid design on it) from funnel and discard. Be sure to leave the support pad in the filter funnel.
6. Load filtration funnel with sterile polycarbonate filter on support pad (shiny side up).
7. Shake sample bottle(s) 25 times to mix well.
8. Measure 25-mL of the mixed water sample in the sterile graduated centrifuge tube and pour into filter funnel.
9. Replace cover on filter funnel and pump to generate a vacuum (do not generate more than 7 inches of Hg of pressure). Keep pumping until all liquid is in filtrate collection flask.
10. If the first 25 mL volume passes readily through the filter, add another 25 mL and continue filtration. If the filter clogs before completely filtering the first or second 25 mL volume, discard the filter and repeat the filtration using a lesser volume.
11. Pour approx. 10-mL of the chilled phosphate buffered saline (PBS) into the graduated PP tube used for the sample. Cap the tube and shake 5 times. Remove the cap and pour rinsate into filter funnel to rinse filter.
12. Filter the rinsate and repeat with another 10 mL of phosphate buffered saline (PBS).

Table 7.2. Processing procedure—fecal indicator sample

13. Remove filter funnel from base without disturbing filter. Using sterile disposable forceps remove the filter (touching only the filter edges) and fold it in half, in quarters, in eighths, and then in sixteenths (filter will be folded 4 times).
14. Insert filter into chilled filter extraction tube (with beads) open end down. Replace and tighten the screw cap, insert tube(s) into bubble wrap bag on dry ice for preservation during transport and shipping.
15. Record the volume of water sample filtered through each filter and the volume of buffer rinsate each filter was rinsed with on the Sample Collection Form, Side 2. Record the filtration start time and finish time for each sample.
16. Repeat steps 6 to 15 for the remaining three 50-mL sub-sample volumes to be filtered.

7.2.3 Equipment and Supplies (*Chlorophyll a* from Water Sample)

Table 7-3 provides the equipment and supplies needed to process the *chlorophyll a* water sample.

Table 7.3. Equipment and supplies list for *chlorophyll a* processing

For filtering *chlorophyll a* sample	▪ Whatman GF/F 0.7 µm glass fiber filter ▪ Filtration apparatus with graduated filter holder ▪ Vacuum pump (electric pump may be used if available) ▪ 50-mL screw-top centrifuge tube ▪ Aluminum foil square ▪ DI water ▪ Nitrile gloves ▪ Forceps
For recording measurements	▪ Sample Collection Form ▪ Sample labels ▪ #2 pencils ▪ Fine-tipped indelible markers ▪ Clear tape strips

7.2.4 Procedures for Processing the *Chlorophyll a* Water Sample

The procedures for processing *chlorophyll a* water samples are presented in Table 7-4. Whenever possible, sample processing should be done in subdued light, out of direct sunlight.

Table 7.4. Processing procedure—*chlorophyll a* sample

1. Put on nitrile gloves.
2. Use clean forceps to place a Whatman GF/F 0.7 µm glass fiber filter in the graduated filter holder apparatus with the gridded side of the filter facing down.
3. Pour 250 mL of water into the filter holder, replace the cap, and use the vacuum pump to draw the sample through the filter. If 250 mL of site water will not pass through the filter, change the filter, rinse the apparatus with DI water, and repeat the procedures using 100-mL of site water. *NOTE: IF the water is green or turbid, use a smaller volume to start with.*
4. Rinse the upper portion of the filtration apparatus thoroughly with DI water to include any remaining cells adhering to the sides and pump through the filter (do not exceed 7 inches of Hg). Monitor the level of water in the lower chamber to ensure that it does not contact the filter or flow into the pump.
5. Observe the filter for visible color. If there is visible color, proceed; if not, repeat steps 3 & 4 until color is visible on the filter or until a maximum of 2,000 mL have been filtered. Record the actual sample volume filtered on the Sample Collection Form.
6. Remove the bottom portion of the apparatus and pour off the water from the bottom.
7. Remove the filter from the holder with clean forceps. Avoid touching the colored portion of the filter. Fold the filter in half, with the colored side folded in on itself.
8. Place the folded filter into a 50-mL screw-top centrifuge tube and cap. Record the sample volume filtered on a chlorophyll label and attach it to the centrifuge tube. Ensure that all written information is complete and legible. Cover with a strip of clear tape. Wrap the tube in aluminum foil and place in a self-sealing plastic bag. Place this bag between two small bags of ice in a cooler.

7.2.5 Equipment and Supplies (Periphyton Sample)

Table 7-5 lists the equipment and supplies needed to process the periphyton sample.

Table 7.5. Equipment and supplies list for periphyton sample processing

For filtering periphyton samples	▪ Whatman 47 mm 0.7 micron GF/F glass fiber filter ▪ Whatman 47 mm 1.2 micron GF/C glass fiber filter ▪ Filtration apparatus with graduated filter holder ▪ Vacuum pump (electric pump may be used) ▪ 25 or 50-mL graduated cylinder ▪ 4 50 mL screw-top centrifuge tubes ▪ 60-mL syringe	▪ Aluminum foil squares ▪ Forceps ▪ deionized water in wash bottle ▪ plastic electrical tape ▪ dry ice ▪ wet ice ▪ coolers
For data recording	▪ Sample Collection Form ▪ Sample labels ▪ Pencils	▪ Fine-tipped indelible markers ▪ Clear tape strips

7.2.6 Procedures for Processing the Periphyton Samples

Four different types of laboratory samples are prepared from the composite index samples: an **ID/enumeration sample** (to determine taxonomic composition and relative abundances), a **chlorophyll sample**, a **biomass sample** (for ash-free dry mass [AFDM]), and an **acid/alkaline phosphatase activity** (APA) sample. All the sample containers required for an individual site should be sealed in plastic bags until use to avoid external sources of contamination (e.g., dust, dirt, or mud) that are present at site shorelines.

7.2.6.1 ID/Enumeration Sample

Prepare the ID/Enumeration sample as a 50-mL aliquot from the composite index sample, following the procedure presented in Table 7-6. Preserve each sample with Lugol's. Record the sample ID number from the container label and the total volume of the sample in the appropriate fields on the Sample Collection Form as shown in Figure 5.1-2 and 6.1-2. Store the preserved samples upright in a container containing absorbent material.

Table 7.6. Procedure for ID/enumeration samples of periphyton

1. Prepare a sample label (with a sample ID number) for the Periphyton ID sample. Record the volume of the subsample (typically 50 mL) and the volume of the composite index sample on the label. Attach completed label to a 50-mL centrifuge tube; avoid covering the volume graduations and markings. Cover the label completely with a clear tape strip.
2. Record the sample ID number of the label and the total volume of the composite index sample on the form.
3. Rinse a 60-mL syringe with deionized water.
4. Thoroughly mix the bottle containing the composite sample.
5. Withdraw 50 mL of the mixed sample into the syringe. Right after mixing, place the contents of syringe sample into the labeled 50-mL centrifuge tube.
6. Use a syringe or bulb pipette to add 1 mL Lugol's to the tube. Cap the tube tightly and seal with plastic electrical tape. Shake gently to distribute preservative.
7. Record the volume of the sample in the centrifuge tube (excluding the volume of preservative) in "Assemblage ID Subsample Vol." field of the Sample Collection Form.

7.2.6.2 Chlorophyll Sample

Prepare the chlorophyll sample by filtering a 25-mL aliquot of the composite index sample through a 47 mm 0.7 micron GF/F glass fiber filter. The procedure for preparing chlorophyll samples is presented in Table 7-7. Chlorophyll can degrade rapidly when exposed to bright light. If possible, prepare the samples in subdued light (or shade), filtering as quickly as possible after collection to minimize degradation. Keep the glass fiber filters in a dispenser inside a sealed plastic bag until use.

It is important to measure the volume of the sample being filtered accurately (±1 mL) with a graduated cylinder. During filtration, do not exceed 7 inches of Hg to avoid rupturing cells. If the vacuum pressure exceeds 7 inches of Hg, prepare a new sample. If the filter clogs completely before all the sample in the chamber has been filtered, discard the sample and filter, and prepare a new sample using a smaller volume of sample.

Table 7.7. Procedure for preparing chlorophyll samples of periphyton

1. Using clean forceps, place a Whatman GF/F 0.7 µm glass fiber filter on the filter holder gridded side down. Use a small amount of deionized water from a wash bottle to help settle the filter properly. Attach the filter funnel to the filter holder and filter chamber, then attach the hand vacuum pump to the chamber.
2. Rinse the sides of the filter funnel and the filter with a small volume of deionized water.
3. Rinse a 25-mL or 50-mL graduated cylinder three times with small volumes of deionized water.
4. Mix the composite sample bottle thoroughly.
5. Measure 25 mL (±1 mL) of sample into the graduated cylinder. • *NOTE: For a composite sample containing fine sediment, allow grit to settle for 10 - 20 seconds before pouring the sample into the graduated cylinder.*
6. Pour the 25-mL aliquot into the filter funnel, replace the cap, and pull the sample through the filter using the hand pump. Vacuum pressure from the pump should not exceed 7 inches of Hg to avoid rupture of fragile algal cells. • *NOTE: If 25 mL of sample will not pass through the filter, discard the filter and rinse the chamber thoroughly with deionized water. Collect a new sample using a smaller volume of sample, measured to ±1 mL. Be sure to record the actual volume sampled on the sample label and the Sample Collection Form.*
7. Remove both plugs from the filtration chamber and pour out the filtered water in the chamber. Remove the filter funnel from the filter holder. Remove the filter from the holder with clean forceps. Avoid touching the colored portion of the filter. Fold the filter in half, with the colored sample (filtrate) side folded in on itself. Place the folded filter in a 50 mL centrifuge tube. Discard filtered water.
8. Complete a periphyton sample label for chlorophyll, including the volume filtered, and attach it to the centrifuge tube. Cover the label completely with a strip of clear tape. Place the centrifuge tube into a self-sealing plastic bag.
9. Record the sample ID number of the label and the total volume of the composite index sample on the form. Record the volume filtered in the “Chlorophyll” field on the Sample Collection Form. Double check that the volume recorded on the collection form matches the total volume recorded on the sample label.
10. Place the centrifuge tube containing the filter on dry ice.

7.2.6.3 Biomass Sample

Prepare the ash-free dry mass (AFDM) sample by filtering a 25-mL aliquot of the composite index sample through a 47 mm 1.2 micron GF/C glass fiber filter. The procedure for preparing AFDM samples is presented in Table 7-8. Keep the glass fiber filters in a dispenser inside a sealed plastic bag until use.

It is important to measure the volume of the sample being filtered accurately (±1 mL) with a graduated cylinder. During filtration, do not exceed 7 inches of Hg to avoid rupturing cells. If the vacuum pressure exceeds 7 inches of Hg prepare a new sample. If the filter clogs completely before all the sample in the chamber has been filtered, discard the sample and filter, and prepare a new sample using a smaller volume of sample.

Table 7.8. Procedure for preparing ash-free dry mass (AFDM) samples of periphyton

1. Using clean forceps, place a Whatman 47 mm 1.2 micron GF/C glass fiber filters on the filter holder gridded side down. Use a small amount of deionized water from a wash bottle to help settle the filter properly. Attach the filter funnel to the filter holder and filter chamber, then attach the hand vacuum pump to the chamber.
2. Rinse the sides of the filter funnel and the filter with a small volume of deionized water.
3. Rinse a 25-mL or 50-mL graduated cylinder three times with small volumes of deionized water.
4. Mix the composite sample bottle thoroughly.
5. Measure 25 mL (±1 mL) of sample into the graduated cylinder. *NOTE: For a composite sample containing fine sediment, allow grit to settle for 10 - 20 seconds before pouring the sample into the graduated cylinder.*
6. Pour the 25-mL aliquot into the filter funnel, replace the cap, and pull the sample through the filter using the hand pump. Vacuum pressure from the pump should not exceed 7 inches of Hg to avoid rupture of fragile algal cells.

NOTE: If 25 mL of sample will not pass through the filter, discard the filter and rinse the chamber thoroughly with deionized water. Collect a new sample using a smaller volume of sample, measured to ±1 mL. Be sure to record the actual volume sampled on the sample label and the Sample Collection Form.

7. Remove both plugs from the filtration chamber and pour out the filtered water in the chamber. Remove the filter funnel from the filter holder. Remove the filter from the holder with clean forceps. Avoid touching the colored portion of the filter. Fold the filter in half, with the colored sample (filtrate) side folded in on itself. Place the folded filter in a 50 mL centrifuge tube. Discard filtered water.
8. Complete a periphyton sample label for biomass, including the volume filtered, and attach it to the centrifuge tube. Cover the label completely with a strip of clear tape. Place the centrifuge tube into a self-sealing plastic bag.
9. Record the sample ID number of the label and the total volume of the composite index sample on the form. Record the volume filtered in the “Biomass” field on the Sample Collection Form. Double check that the volume recorded on the collection form matches the total volume recorded on the sample label.
10. Place the centrifuge tube containing the filter on dry ice.

7.2.6.4 Acid/Alkaline Phosphatase Activity Sample

Prepare the Acid/Alkaline phosphatase activity (APA) sample from a 50-mL subsample of the composite index sample. Table 7-9 presents the procedure for preparing APA samples. No field treatment (i.e., filtration, preservation) of the APA sample is necessary. Complete a label for the sample and affix it to a 50-mL centrifuge tube. Record the sample ID number, and the volume of the subsample on the Sample Collection Form (Figure 6.1-3). Check to ensure that the information recorded on the Sample Collection Form matches the corresponding information recorded on the sample label. Store APA samples frozen until shipment to the laboratory.

Table 7.9. Procedure for preparing acid alkaline phosphatase activity samples for periphyton

1. Prepare a sample label (with a sample number) for the APA sample. Record the volume of the sample (typically 50 mL) and the volume of the composite index sample on the label. Attach the completed label to a 50-mL centrifuge tube; avoid covering the volume graduations and markings. Cover the label completely with a clear tape strip.
2. Rinse a 60-mL syringe with deionized water.
3. Thoroughly mix the bottle containing the composite sample.
4. Withdraw 50 mL of the mixed sample into the syringe. Place the contents of the syringe sample into the labeled 50-mL centrifuge tube. Cap the tube tightly and seal with plastic electrical tape.
5. Record the sample ID number of the label and the total volume of the composite index sample on the form.
6. Record the volume of the sample in the centrifuge tube in the "APA Sample" field of the Sample Collection Form.
7. Freeze the sample immediately and keep frozen until shipping.

7.3 Data Forms and Sample Inspection

After the Site Assessment Form is completed, the Field Team Leader reviews all of the data forms and sample labels for accuracy, completeness, and legibility. The other team members inspect all sample containers and package them in preparation for transport, storage, or shipment. Refer to Appendix C for details on preparing samples for shipping.

Ensure that all required data forms for the site have been completed. Confirm that the SITE-ID, the visit number, and date of visit are correct on all forms. On each form, verify that all information has been recorded accurately, the recorded information is legible, and any flags are explained in the comments section. Ensure that written comments are legible, with no "shorthand" or abbreviations. Make sure there are no marking s in the scan code boxes. Make sure the header information is completed on all pages of each form. After reviewing each form initial the upper right corner of each page of the form.

Ensure that all samples are labeled, all labels are completely filled in, and each label is covered with clear plastic tape. Compare sample label information with the information recorded on the corresponding field data forms (e.g., the Sample Collection Form) to ensure accuracy. Make sure that all sample containers are properly sealed.

7.4 Launch Site Cleanup

Load the boat on the trailer and inspect the boat, motor, and trailer for evidence of weeds and other macrophytes. Clean the boat, motor, and trailer as completely as possible before leaving the launch site. Inspect all nets for pieces of macrophyte or other organisms and remove as much as possible before packing the nets for transport. Pack all equipment and supplies in the vehicle and trailer for transport. Keep equipment and supplies organized so they can be inventoried using the equipment and supply checklists presented in Appendix A. Lastly,

be sure to clean up all waste material at the launch site and dispose of or transport it out of the site if a trash can is not available.

8.0 FIELD QUALITY CONTROL

Standardized training and data forms provide the foundation to help assure that data quality standards for field sampling are met. These Standard Operating Procedures for field sampling and data collection are the primary guidelines for all cooperators and field teams. In addition, repeat sampling, duplicate sampling, and field evaluation and assistance visits will address specific aspects of the data quality standards for the National Rivers and Streams Assessment.

8.1 Repeat and Duplicate Sampling

Repeat and duplicate sampling will provide data to make variance estimates (for measurement variation and index period variation) that can be used to evaluate the NRSA design for its potential to estimate status and detect trends in the target population of sites. A summary of the repeat and duplicate sampling design is provided in Figure 8-1.

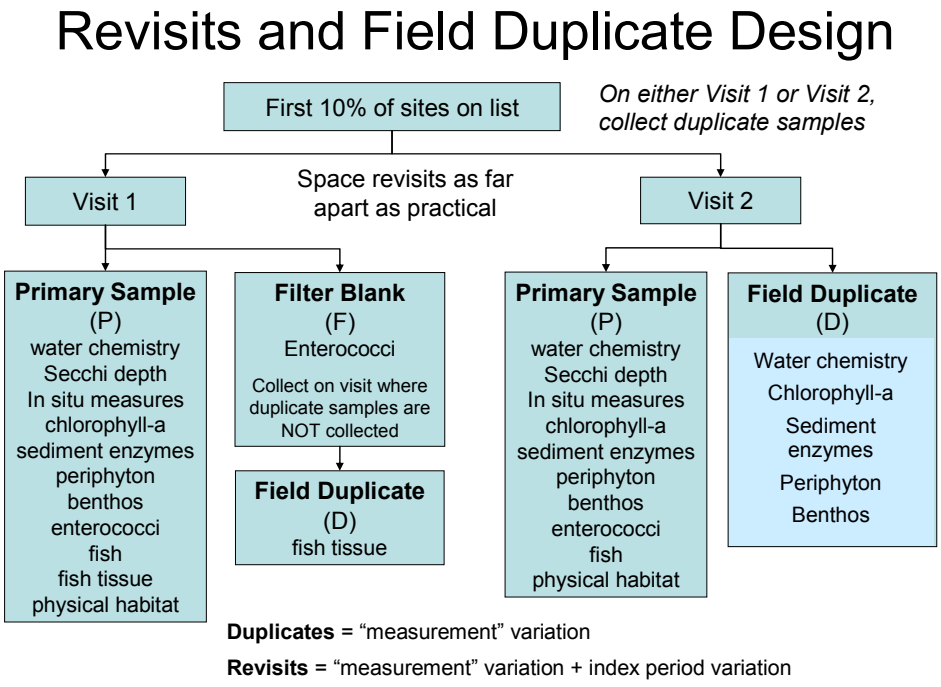


Figure 8.1. Summary of the repeat and duplicate sampling design.

8.1.1 Repeat Sampling

A total of 10% of the target sites visited will be revisited during the same field season by the same field team that initially sampled the site. Repeated samples and measurements are taken from the same reach as the first visit. Each state has four repeat sites; the first two wadeable and the first two non-wadeable sites in their list. If a site selected for repeat sampling is dropped, then the alternate assigned to replace it should be revisited. If a non-wadeable site is sampled with wadeable methods, the next non-wadeable site should be selected as the repeat site. The primary purpose of this "revisit" set of sites is to collect temporal replicate samples to provide variance estimates for both measurement variation and index period variation. The revisit will include the full set of indicators and associated parameters. The time period between the initial and repeat visit to a site should be as long as possible, but not less than 2 weeks. Fish tissue and PPCP water samples will only be collected on the first visit (see Section 8.1.2).

8.1.2 Duplicate Sampling

Duplicate samples will be collected for certain indicators from the sites that are revisited. They will be collected at one of the visits, not both. These duplicate samples will be collected for water chemistry, *chlorophyll a*, sediment enzymes, periphyton, benthos, enterococci, and fish tissue (not for fish community data or physical habitat). These samples and measurements are taken from the same reach as the primary sample. The samples should be taken by the same field crew and ≤2 days later. These spatial replicates will provide measurement variance and spatial variance estimates. Label the samples as (*primary site ID#*)-D to indicate that they are duplicate samples. Duplicates for fish tissue should be taken on the first visit, no fish tissue needs to be collected during the second visit. Duplicate PPCP water samples should also be collected during the first visit at the designated urban river sites.

In addition, a filter blank will be collected for enterococci. The teams will filter a small amount (10 mL) of sterile buffer through 4 filters, label them and write "blank" on the label and field form, and package and submit these samples to the lab. The filter blanks should be run before the sample is filtered. The filter blanks should be collected on the field visit that duplicate samples are not collected (Figure 8-1). A detailed description of the filter blanks is found in table 7-2.

8.1.3 Taking Field Duplicates

On the visit crew are taking duplicates samples, ensure that there are two site kits for supplies and materials. If you are taking duplicates on a subsequent field day follow standard sample procedures for collecting the duplicate samples. If you are collecting duplicates on the same day as the primary sample follow the modified protocols in this section. Fish tissue, both a primary and duplicate, is collected on the first visit only.

After you take the first water chemistry sample, rinse the beaker three times with stream water, replace any torn gloves, and collect a second sample with a new cubitainer following the procedures in the water chemistry sections. The water chemistry *chlorophyll a* sample can be filtered from the same container as the primary sample. If there is not sufficient water for both filters, process the primary sample, then collect a second water sample from the index site for the duplicate sample.

For transect sample duplicates (sediment enzymes, benthic macroinvertebrates, and periphyton) move 1 meter upstream of the primary sample location. At this new location upstream of the transect, take a duplicate sample following the same procedures that are used to collect the primary sample. You do not need to collect a duplicate for the low gradient samples.

8.2 Field Evaluation and Assistance Visits

A rigorous program of field and laboratory evaluation and assistance visits has been developed to support the National Rivers and Streams Assessment Program. These evaluation and assistance visits are explained in detail in the Quality Assurance Project Plan (QAPP) for the NRSA. The following sections will focus only on the field evaluation and assistance visits.

These visits provide a QA/QC check for the uniform evaluation of the data collection methods, and an opportunity to conduct procedural reviews as required to minimize data loss due to improper technique or interpretation of field procedures and guidance. Through uniform training of field teams and review cycles conducted early in the data collection process, sampling variability associated with specific implementation or interpretation of the protocols will be significantly reduced. The field evaluations will be based on the Field Evaluation Plan and Checklists. This evaluation will be conducted for each unique team collecting and contributing data under this program (EPA will make a concerted effort to evaluate every team, but will rely on the data review and validation process to identify unacceptable data that will not be included in the final database).

8.2.1 Specifications for QC Assurance

Field evaluation and assistance personnel are trained in the specific data collection methods detailed in this Field Operations Manual. A plan and checklist for field evaluation and assistance visits have been developed to detail the methods and procedures. The plan and checklist are included in the QAPP. Table 8-1 summarizes the plan, the checklist, and corrective action procedures.

Table 8.1. General information noted during field evaluation

Field Evaluation Plan	▪ Regional Coordinators will arrange the field evaluation visit with each Field Team, ideally within the first two weeks of sampling. ▪ The Evaluator will observe the performance of a team through one complete set of sampling activities. ▪ If the Team misses or incorrectly performs a procedure, the Evaluator will note it on the checklist and immediately point it out so the mistake can be corrected on the spot. ▪ The Evaluator will review the results of the evaluation with the Field Team before leaving the site, noting positive practices and problems.
Field Evaluation Checklist	▪ The Evaluator observes all pre-sampling activities and verifies that equipment is properly calibrated and in good working order, and NRSA protocols are followed. ▪ The Evaluator checks the sample containers to verify that they are the correct type and size, and checks the labels to be sure they are correctly and completely filled out. ▪ The Evaluator confirms that the Field Team has followed NRSA protocols for locating the site.

Table 8.1. General information noted during field evaluation

	▪ The Evaluator observes the complete set of sampling activities, confirming that all protocols are followed. ▪ The Evaluator will record responses or concerns, if any, on the Field Evaluation and Assistance Check List.
Corrective Action Procedures	▪ If the Evaluator's findings indicate that the Field Team is not performing the procedures correctly, safely, or thoroughly, the Evaluator must continue working with this Field Team until certain of the Team's ability to conduct the sampling properly so that data quality is not adversely affected. ▪ If the Evaluator finds major deficiencies in the Field Team operations the Evaluator must contact a NRSA QA official.

It is anticipated that evaluation and assistance visits will be conducted with each Field Team early in the sampling and data collection process, and that corrective actions will be conducted in real time. If the Field Team misses or incorrectly performs a procedure, the Evaluator will note this on the checklist and immediately point this out so the mistake can be corrected on the spot. The role of the Evaluator is to provide additional training and guidance so that the procedures are being performed consistent with the Field Operations Manual, all data are recorded correctly, and paperwork is properly completed at the site.

8.2.2 Reporting

When the sampling operation has been completed, the Evaluator will review the results of the evaluation with the Field Team before leaving the site (if practicable), noting positive practices and problems (i.e., weaknesses [might affect data quality] or deficiencies [would adversely affect data quality]). The Evaluator will ensure that the Team understands the findings and will be able to perform the procedures properly in the future. The Evaluator will record responses or concerns, if any, on the Field Evaluation and Assistance Check List. After the Evaluator completes the Field Evaluation and Assistance Check List, including a brief summary of findings, all Field Team members must read and sign off on the evaluation.

If the Evaluator's findings indicate that the Field Team is not performing the procedures correctly, safely, or thoroughly, the Evaluator must continue working with this Field Team until certain of the Team's ability to conduct the sampling properly so that data quality is not adversely affected. If the Evaluator finds major deficiencies in the Field Team operations (e.g., major misinterpretation of protocols, equipment or performance problems) the Evaluator must contact the following QA official:

- *Sarah Lehmann*, EPA National Rivers and Streams Assessment Project QA Officer

The QA official will contact the Project Manager to determine the appropriate course of action. Data records from sampling sites previously visited by this Field Team will be checked to determine whether any sampling sites must be redone.

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Web Pages:

US EPA Aquatic Monitoring Research: http://www.epa.gov/nheerl/arm
NHD Plus: http://www.horizon-systems.com/nhdplus

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APPENDIX A

List of Equipment and Supplies

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EQUIPMENT & SUPPLY LISTS

General Equipment

- Field Operations Manual and/or laminated Quick Reference Guide
- Laminated invasive species guide
- Covered clipboards
- Filed forms and sample labels
- Clear tape strips for covering labels
- Pencils (#2)
- Fine-tipped indelible markers
- Digital camera with extra memory card & battery
- Maps and access instructions
- Sampling permits and/or permission letters
- GPS unit with manual and reference card
- 50 m or 100 m measuring tape with reel
- Surveyor's flagging tape
- Laser rangefinder (400 ft. distance range) and clear waterproof bag
- Batteries
- 1% - 10% Bleach
- Barometer or elevation chart to use for calibration
- Calibration cups and standards for multi-probe unit
- Electrical tape
- Scissors
- Plastic storage tub
- Cell phone, 2-way radios, and/or walkie-talkies
- 2 pair chest waders
- 1 or 2 fisherman's vest with lots of pockets and snap fittings.

Boat Equipment List

- Motor
- Gas Can
- Lifejackets (1/person)
- Type IV PFD (Throwable Life Saving device)
- Bow/Stern lights
- Anchor with 75m line or sufficient to anchor in 50m depth
- Float to attach to anchor
- Sonar Unit
- Oars or Paddles
- First Aid Kit
- Extra Boat Plug
- Spare Prop Shear Pin
- Emergency Tool kit
- Hand Bilge pump
- Fire Extinguisher
- Boat horn
- Spare prop

Sample/Data Collection

- Multi-parameter water quality meter with pH, DO, temperature, and conductivity probes
- 20 cm diameter Secchi disk and calibrated sounding line, marked in 0.5 m intervals
- 3 L Nalgene beaker
- 1-2L Amber Nalgene bottle
- Tape measure (in centimeters)
- Nitrile gloves
- Calibrated PVC sounding rod, 3-m length, marked in 0.1 m increments
- Convex spherical canopy densiometer (Lemmon Model B), modified with taped "V"
- Clinometer
- Bearing compass (Backpacking type)
- Binoculars
- Surveyor's telescoping leveling rod (round profile, metric scale, 7.5m extended)
- Meter stick for bank angle measurements
- Current velocity meter, probe, and operating manual
- Top-set wading rod for use with current velocity meter
- Neutrally buoyant object (e.g., plastic golf ball with holes, small rubber ball, stick)
- Portable Weir with 60° "V" notch (optional) and plastic sheeting to use with weir
- Plastic bucket (or similar container) with volume graduations
- Petite Ponar sampler with plastic tub, drop line, and spare pinch pin. (*Standard Ponar may substitute*)
- 60-mL plastic syringe with 3/8" hole bored into the end
- Large stainless steel spoon for mixing sediment composite
- Large Funnel (15-20 cm diameter)
- 12-cm^2 area delimiter (3.8 cm diameter pipe, 3 cm tall)
- Stiff-bristle toothbrush with handle bent at 90˚ angle
- Modified kick net (D-frame, 500 µm mesh, 4-ft handle)
- Sieve-bucket, 500 µm mesh (U.S. std No. 35)
- Watch with timer or stopwatch
- Watchmakers' forceps
- Buckets, plastic, 8- to 10-qt capacity
- Plastic electrical tape
- Electrofishing equipment (boat, barge, and/or backpack units, including variable voltage pulsator unit, wiring cables, generator, electrodes, dip nets, and all safety equipment)
- Linesman gloves
- Livewell and/or buckets
- 2 Non-conducting dip nets with 1/4" mesh
- 1 Minnow net for dipping small fish from live well
- Measuring board (millimeter scale)
- Pre-sterilized, 250 ml sample bottle
- Sodium thiosulfate tablet
- 500-mL plastic bottles for the periphyton composite sample
- 25-mL or 50-mL graduated cylinder
- 1-L wash bottle for stream water
- 1-L wash bottle containing deionized water

Sample Processing/Preservation

- Coolers
- Wet ice
- Dry ice
- 95% ethanol
- 10% buffered formalin
- Lugol's solution
- Sterile filtration unit (Nalgene 145/147), including filter funnel, cap, filter holder, and receiving chamber
- Vacuum hand pump and clear plastic tubing
- Sterile disposable forceps
- Whatman 47 mm polycarbonate 0.4 micron filters
- Whatman 47 mm 0.7 micron GF/F glass fiber filters
- Whatman 47 mm 1.2 micron GF/C glass fiber filters
- 60 x 15 disposable Petri dishes
- Phosphate buffered saline solution
- Aluminum foil squares (3" x 6")
- DI water
- Small spatula, spoon, or scoop to transfer sample
- Aluminum foil (solvent-rinsed and baked)
- Heavy-duty food grade polyethylene tubing
- Large plastic (composite) bags
- Knife or scissors
- Plastic cable ties
- Scalpel for slitting open large fish before preservation

Sample Storage

- One 4-L cube container
- Three 1-L Nalgene bottles
- Several Leak-proof HDPE jars for fish voucher specimens (various sizes from 250 mL - 4L)
- 500-mL plastic bottle for sediment sample
- Sample jars, 1-L HDPE plastic suitable for use with ethanol (benthic samples)
- 50-mL screw-top centrifuge tube
- sterile microcentrifuge tubes containing sterile glass beads
- Coolers

Packaging/Shipping

- Coolers
- Cooler liners (30-gal garbage bags)
- Dry ice (~60 lbs per site)
- Wet ice (~50 lbs per site; additional for shipping)
- 1-gallon self-sealing bags
- Packing/strapping tape
- FedEx airbills
- Class 9 Dangerous Goods label (for dry ice shipments)

A **site kit** will be provided to the field crews for each sampling site. Site kits will be shipped out based on the schedule that each field crew provides prior to the start of the sampling season. **Field crew leaders MUST provide a schedule in order to receive the site kits.** If your schedule changes, please report the change as soon as possible to the Field Logistics Coordinator (Jennifer Pitt; 410-356-8993). Prior to sampling, inspect each site kit to ensure all supplies are included.

Supplies provided in each Site Kit:

- Field Data Forms
- Sample Labels
- National Rivers and Streams Assessment Fact Sheets
- 1 4-L cube container
- 1 1-L Nalgene bottle
- 500-mL plastic bottle for sediment sample
- 1 sterile 250 mL fecal indicator bottle
- 1 Zip tie
- 2 1-L HDPE plastic sample jars suitable for use with ethanol (benthic samples)
- 5 50-mL screw-top centrifuge tubes (4 for periphyton, 1 for measuring enterococci sample for filtering and then for storing the *chlorophyll a* filter)
- 4 sterile microcentrifuge tubes containing sterile glass beads
- Funnel analytical test filter 250 mL
- Sterile disposable forceps (2)
- Sterile phosphate buffered saline (PBS)
- Large Plastic Bags
- Foam envelope
- FedEx airbills for all labs
- Dry ice box will be included in approximately every 4th site kit
- Dry ice shipping label

Supplies Provided in Each Fish Tissue Sampling Kit:

- Aluminum foil (solvent-rinsed and baked)
- Heavy-duty food grade polyethylene tubing
- Large plastic (composite) bags
- Plastic cable ties

Supplies Provided in Each Base Kit:

- Nitrile Gloves
- Clinometer
- Spherical Densiometer
- Bottle of 50 Sodium Thiosulfate Tablets
- Aluminum foil 3x6”
- 15” stainless steel spoon
- (2) D-frame Kick Net – 500 µm mesh, 52” handle
- (2) Sieve bucket - 500 µm
- Weighted Secchi disk
- Rectangular fiberglass surveying rod – metric
- CST Berger SAL 20 Automatic Level
- Level tripod
- (2) 1 Liter Nalgene wash bottles
- 3 gallon Rubbermaid Roughneck tote
- Graduated cylinder 250 mL
- 2 Liter amber Nalgene rectangular bottle
- 500-mL plastic bottle for periphyton sample collection
- Nalgene filtering flask
- #8 silicone stopper
- Filter funnel adapter
- Whatman 47 mm polycarbonate 0.4 µ filters
- Whatman 47 mm glass fiber GF/F 0.7 µ filters
- Whatman 47 mm glass fiber GF/C 1.2 µ filters
- Disposable petri dishes 60x15
- 3 Liter Nalgene beaker
- Utility funnel 15cm diameter
- Centrifuge tube stand
- Hand vacuum pump
- 500 mL Lugol’s solution
- 4 Liters of QC check solution
- Tape dispenser
- Tape strips
- ½ gallon bucket
- 60 cc syringe with 3/8” hole and tubing
- 12 cm^2 area delimiter
- (2) 2 mL pipet and pipet bulb
- Toothbrush bent to 90º
- 24 ct of 1 Liter Nalgene bottles

Note: Lugol’s solution, calibration QC check solution, filters, 1 Liter Nalgene bottles, aluminum foil squares, and disposable nitrile gloves will be provided in the base kit; you may order more throughout the field season if needed.

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APPENDIX B

Field Forms

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BOATABLE FORMS PACKET

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STREAM VERIFICATION FORM - WADEABLE/BOATABLE (Front)

Reviewed by (initial): _____

SITE NAME: _____ DATE: ___/___/ 2 0 ___ VISIT: ○ 1 ○ 2 ○ 3

SITE ID: FW08 State of Site Location: _____ Don't forget to record Reach Length on back. TEAM: _____

STREAM/RIVER VERIFICATION INFORMATION

Stream/River Verified by (fill in all that apply) ○ GPS ○ Local Contact ○ Signs ○ Roads ○ Topo. Map
○ Other (Describe Here): ○ Not Verified (Explain in Comments)

Coordinates		Latitude North	Longitude West	# of Satellites	Are GPS Coordinates w/i 10 Sec. of map?
MAP	Degrees, Minutes, and Seconds OR Decimal Degrees	___ ___ ___ . ___	___ ___ ___ . ___	○ ≤3 ○ ≥4	○ Yes ○ No GPS Datum Used (e.g. NAD27): _____
GPS	Degrees, Minutes, and Seconds OR Decimal Degrees	___ ___ ___ . ___	___ ___ ___ . ___		

DID YOU SAMPLE THIS SITE?

○ YES If YES, check one below

SAMPLEABLE (Choose method used)
- ○ Wadeable - Continuous water, greater than 50% wadeable
- ○ Boatable
- ○ Partial - Sampled by wading (>50% of reach sampled). Explain below
- ○ Partial - Sampled by boat (>50% of reach sampled). Explain below.
- ○ Wadeable Interrupted - Not continuous water along reach
- ○ Boatable Interrupted - Not continuous water along reach
- ○ Altered - Stream/River Channel Present but differs from Map

○ NO If NO, check one below

NON-SAMPLEABLE-PERMANENT
- ○ Dry - Visited
- ○ Dry - Not visited
- ○ Wetland (No Definable Channel)
- ○ Map Error - No evidence channel/waterbody ever present
- ○ Impounded (Underneath Lake or Pond)
- ○ Other (explain in comments)

NON-SAMPLEABLE-TEMPORARY
- ○ Not boatable - Need a different crew - Reschedule for this year
- ○ Not wadeable - Need a different crew - Reschedule for this year
- ○ Other (Explain in comments)

NO ACCESS
- ○ Access Permission Denied
- ○ Permanently Inaccessible (Unable/Unsafe to Reach Site)
- ○ Temporarily Inaccessible-Fire, etc. - Reschedule for next year

GENERAL COMMENTS:

DIRECTIONS TO STREAM/RIVER SITE:

Record information used to define length of reach, and sketch general features of reach on reverse side.

STREAM VERIFICATION FORM - WADEABLE/BOATABLE (Back)

Reviewed by (initial): _____

SITE NAME: _____ DATE: ___ / ___ / 2 0 ___ VISIT: ○ 1 ○ 2 ○ 3

SITE ID: FW08 TEAM: _____

STREAM/RIVER REACH DETERMINATION

Channel Width Used to Define Reach (m)	DISTANCE (m) FROM X-SITE		Total Reach Length Intended (m)	Comment
	Upstream Length	Downstream Length		

SKETCH MAP - Arrow Indicates North; Mark site L=Launch X=Index T= Take Out
NOTE: If an outline map is attached here, use a continuous strip of clear tape across the top edge.
You can also attach a separate sheet with the outline map on it.
For boatable sites you can attach topo map with reach, X-site and transect locations marked.

PERSONNEL

NAME	Bio/Chem Sampling	Habitat	Forms Review
_____	○	○	○
_____	○	○	○
_____	○	○	○
_____	○	○	○
_____	○	○	○

FIELD MEASUREMENT FORM - BOATABLE

Reviewed by (initial):_____

SITE ID: FW08 DATE: ___/___/ 2 0___

CALIBRATION INFORMATION

Instrument manufacturer and model:

Instrument ID number: Operator:

TEMPERATURE

Thermometer Reading (℃)	Sensor Reading (℃)	Flag	Comments
___.___	___.___		

DO

Elevation	OR	Barometric Pressure (mm Hg)	Calibration Value	Displayed Value	Flag
○ ft ○ m			○ mg/L ○ %	○ mg/L ○ %	

pH

Cal. STD 1 Description	Cal. STD 1 Value	Cal. STD 2 Description	Cal. STD 2 Value
	___.___		___.___

Calibration Verified with Quality Control Sample (QCS)

QCS Description	QCS True	QCS Measured	Flag
	___.___	___.___	

CONDUCTIVITY

Cal. STD 1 Description	Cal. STD 1 Value	Cal. STD 2 Description	Cal. STD 2 Value

Calibration Verified with Quality Control Sample (QCS)

QCS Description	QCS True (µS/cm @25℃)	QCS Measured (µS/cm @25℃)	Flag

Flag	Comments

FIELD MEASURMENTS

TRANSECT: ☐

	○ Left ○ Right	○ Left Ctr ○ Right Ctr	○ CENTER	○ Right Ctr ○ Left Ctr	○ Right ○ Left
Time of Day (HH:MM)	___:___	___:___	___:___	___:___	___:___
DO (mg/L) XX.X	___.___	___.___	___.___	___.___	___.___
Temp. (℃) XX.X	___.___	___.___	___.___	___.___	___.___
pH XX.XX	___.___	___.___	___.___	___.___	___.___
Cond. (µS/cm@25℃) XX.X					
Corrected to 25℃ ?	○ Y ○ N	○ Y ○ N	○ Y ○ N	○ Y ○ N	○ Y ○ N
Depth Disk Disappears (m)	___.___	___.___	___.___	___.___	___.___
Depth Disk Reappears (m)	___.___	___.___	___.___	___.___	___.___
Clear to Bottom?	○	○	○	○	○
Flag					

Flag	Comments

26769

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = misc. flags assigned by field crew. Explain all flags in comment sections.

04/07/2009 NRSA Field Measurement Boatable

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Reviewed by (Initials): _____

SAMPLE COLLECTION FORM - BOATABLE (Front)

SITE ID: FW08 DATE: ____ / ____ / 2 0 ____

WATER CHEMISTRY (4-L CUBITAINER) No Sample Collected ○

Sample ID	Sample Category *	Chilled	Comments
	○ P ○ D	○	
	○ P ○ D	○	

WATER COLUMN CHLOROPHYLL (Target Volume = 1000 mL; max vol = 2000 mL) No Sample Collected ○

Sample ID	Sample Category *	Volume Filtered (mL)	Frozen	Comments
	○ P ○ D		○	
	○ P ○ D		○	

WATER CHEMISTRY PPCP (AMBER GLASS CUBITAINER) No Sample Collected ○

Sample ID	Sample Category *	Chilled	Comments
	○ P ○ D	○	
	○ P ○ D	○	

TRANSECT BENTHOS No Sample Collected ○

TRANSECT	A	B	C	D	E	F	G	H	I	J	K
Location (L/R):	○ L ○ R	○ L ○ R	○ L ○ R	○ L ○ R	○ L ○ R	○ L ○ R	○ L ○ R	○ L ○ R	○ L ○ R	○ L ○ R	○ L ○ R
Dominant Habitat: (ONE PER TRANSECT)	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT
Secondary Habitat: (ONE PER TRANSECT)	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT	○ C ○ L ○ F ○ M ○ OT
Substrate: (ONE PER TRANSECT)	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT
Channel: (ONE PER TRANSECT)	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT

Habitat:
C = Coarse Substrate / LWD L = Leaf Pack
F = Organic Fine Muds / Sand M = Macrophyte beds
OT = Other (Explain in comment section below)

Substrate:
F = Fine / Sand G = Gravel
C = Coarse substrate OT = Other (Explain in comment section below)

Channel:
P = Pool GL = Glide
RI = Riffle RA = Rapid
OT = Other (Explain in comment section below)

Sample ID	Sample Category *	No. Jars	Pre-served	Comments
	○ P ○ D		○	
	○ P ○ D		○	

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = misc. flags assigned by field crew. Explain all flags in comment sections.

*Sample Categories: P = Primary, D = Field Duplicate

SAMPLE COLLECTION FORM - BOATABLE - (Back)

Reviewed by (initial): _____

SITE ID: FW08 DATE: ___/___/2 0___

COMPOSITE PERIPHYTON SAMPLE - Primary — No Sample Collected ○

Sample ID	Sample Category *	Composite Volume (mL)	Number of transects sampled (1-11):	Flag
	○ P ○ D			

Assemblage ID (.1) (50-mL tube)			Chlorophyll (.2) (GF/F Filter)			Biomass (.3) (GF/C Filter)			APA (.4) (50-mL tube)		
Sample Vol. (mL)	Flag	Preserved	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen
		○									○

COMPOSITE PERIPHYTON SAMPLE — No Sample Collected ○

Sample ID	Sample Category *	Composite Volume (mL)	Number of transects sampled (1-11):	Flag
	○ P ○ D			

Assemblage ID (.1) (50-mL tube)			Chlorophyll (.2) (GF/F Filter)			Biomass (.3) (GF/C Filter)			APA (.4) (50-mL tube)		
Sample Vol. (mL)	Flag	Preserved	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen
		○									○

Flag	Comments

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = misc. flags assigned by field crew. Explain all flags in comment sections

SEDIMENT CHEMISTRY / ENZYMES — No Sample Collected ○

Sample ID	Sample Category *	Composite Volume	# of Transects	Chilled	Comments
	○ P ○ D			○	
	○ P ○ D			○	

ENTEROCOCCI (Target Volume = 250 mL) — No Sample Collected ○

Sample ID One unique ID per line	Sample Category *	Time Collected (hhmm)	Depth Collected (m)	Sample Volume (mL)	Filt. Start Time (hhmm)	Volume Filtered (Target = 50 mL) **				Filt. End Time (hhmm)	Time Frozen (hhmm)	Flag
						Filt. 1	Filt. 2	Filt. 3	Filt. 4			
	○ P ○ D											
	○ P ○ D											
	○ P ○ D											
	○ F											

Flag	Comments

* Sample Categories: P = Primary; D = Field Duplicate; F = Filter Blank (Enterococci sample only) Filter blank is collected at visit where field duplicate sample is NOT taken.
** If <25 ml of buffer solution was used to rinse filter, indicate with an F flag and note in comment section which filter(s) were affected along with the approximate volume(s) of buffer solution used.

PHAB: CHANNEL/RIPARIAN TRANSECT FORM - BOATABLE (FRONT)

Rev'd by (init.): _____

SITE ID: FW08 DATE: ___/___/2 0___ Arrival Time ___:___ Leave Time ___:___

TRANSECT: ○ A ○ B ○ C ○ D ○ E ○ F ○ G ○ H ○ I ○ J ○ K ○ X Chosen bank side: (Facing down stream) ○ Left ○ Right

	GPS Latitude - dd mm ss.s	GPS Longitude - ddd mm ss.s
Transect Midstream	___ ___ ___ . ___	___ ___ ___ . ___
Transect Bank	___ ___ ___ . ___	___ ___ ___ . ___

"LITTORAL" SUBSTRATE INFORMATION

SHORE DOM	SHORE SEC	BOTTOM DOM	BOTTOM SEC	CLASS
RS	RS	RS	RS	RS = Bedrock (Smooth) - (Larger than a car)
RR	RR	RR	RR	RR = Bedrock (Rough) - (Larger than a car)
XB	XB	XB	XB	XB = Large Boulder (1000 to 4000 mm) - (Meterstick to car)
SB	SB	SB	SB	SB = Small Boulder (250 to 1000 mm) - (Basketball to Meterstick)
CB	CB	CB	CB	CB = Cobble (64 to 250 mm) - (Tennis ball to Basketball)
GC	GC	GC	GC	GC = Coarse Gravel (16 to 64 mm) - (Marble to Tennis ball)
GF	GF	GF	GF	GF = Fine Gravel (2 to 16 mm) - (Ladybug to marble)
SA	SA	SA	SA	SA = Sand (0.06 to 2 mm) - (Gritty - up to Ladybug size)
FN	FN	FN	FN	FN = Silt / Clay / Muck - (Not Gritty)
HP	HP	HP	HP	HP = Hardpan - (Firm, Consolidated Fine Substrate)
WD	WD	WD	WD	WD = Wood - (Any Size)
OT	OT	OT	OT	OT = Other (Write comment below)

BOTTOM SUBSTRATE FROM (X ONE): ○ Judgement -or- ○ OBS. @ 5 Littoral Depths Flag ____

DEPTH ○ ft ○ m

SONAR XX.X	POLE X.XX	FLAG

BANK CHARACTERISTICS

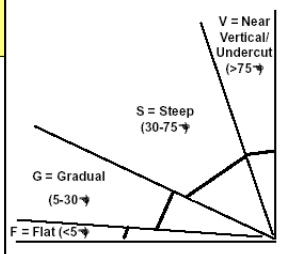
	X.XX (m)	FLAG
Wetted Width		
Bar Width		
Bankfull Width		
Bankfull Height		
Incised Height		

LARGE WOODY DEBRIS (10x20m Plot) TALLY EACH PIECE Flag ____ CHECK IF UNMARKED ARE ZERO ○

DIAMETER LARGE END	Wood All/Part in Wetted Channel: LENGTH 5-15 m	15-30 m	> 30 m	Dry but All/Part in Bankfull Channel: LENGTH 5-15 m	15-30 m	> 30 m
0.3 - 0.6 m						
0.6 - 0.8 m						
0.8 - 1.0 m						
> 1.0 m						

BANK ANGLES CIRCLE ONE

V S G F



SLOPE/BEARING/DISTANCE (Optional): Determine slope if feasible in terms of time and distances. Record GPS coordinates if practical.

Slope and Bearing not determined (use map) ○

INTENDED transect spacing xxx (m): ____ ACTUAL transect spacing xxx (m): ____

Supplemental Waypoints	Slope XX.X %	Backsite Bearing 0 - 359	Distance (m)	Way Point #	GPS Latitude - dd mm ss.s	GPS Longitude - ddd mm ss.s	Flag
MAIN					___ ___ ___ . ___	___ ___ ___ . ___	
1ST					___ ___ ___ . ___	___ ___ ___ . ___	
2ND					___ ___ ___ . ___	___ ___ ___ . ___	
3RD					___ ___ ___ . ___	___ ___ ___ . ___	

Flag	Comments

Flag Codes: K = no measurement made; U = suspect measurement; F1, F2, etc. = flags assigned by each field crew. Explain all flags in comments section on this side or on Side 2 of this form.

24732

PHAB: CHANNEL/RIPARIAN TRANSECT FORM - BOATABLE (Back)

Rev'd by (init.): ____

SITE ID: FW08 DATE: ____ / ____ / 2 0 ____

TRANSECT: ○ A ○ B ○ C ○ D ○ E ○ F ○ G ○ H ○ I ○ J ○ K ○ X

Chosen bank side: (Facing down stream) ○ Left ○ Right

VISUAL RIPARIAN ESTIMATES

0 = Absent (0%) 1 = Spare (<10%) 2 = Moderate (10-40%) 3 = Heavy (40-75%) 4 = Very Heavy (>75%)

D = Deciduous C = Coniferous E = Broadleaf Evergreen M = Mixed N = None

RIPARIAN VEGETATION COVER (10m x 20m Plot)	Left Bank	Right Bank	Flag
Canopy (>5 m high)			
Woody Vegetation Type	D C E M N	D C E M N	
BIG Trees (Trunk >0.3 m DBH)	0 1 2 3 4	0 1 2 3 4	
SMALL Trees (Trunk <0.3 m DBH)	0 1 2 3 4	0 1 2 3 4	
Understory (0.5 to 5 m high)			
Woody Vegetation Type	D C E M N	D C E M N	
Woody Shrubs & Saplings	0 1 2 3 4	0 1 2 3 4	
Non-Woody Herbs, Grasses, & Forbs	0 1 2 3 4	0 1 2 3 4	
Ground Cover (<0.5 m high)			
Woody Shrubs & Saplings	0 1 2 3 4	0 1 2 3 4	
Non-Woody Herbs, Grasses and Forbs	0 1 2 3 4	0 1 2 3 4	
Barren, Bare Dirt or Duff	0 1 2 3 4	0 1 2 3 4	
HUMAN INFLUENCE	0 = Not Present P = >10 m C = Within 10 m B = On Bank Left Bank	Right Bank	
Wall/Dike/Revetment /Riprap/Dam	0 P C B	0 P C B	
Buildings	0 P C B	0 P C B	
Pavement/Cleared Lot	0 P C B	0 P C B	
Road/Railroad	0 P C B	0 P C B	
Pipes (Inlet/Outlet)	0 P C B	0 P C B	
Landfill/Trash	0 P C B	0 P C B	
Park/Lawn	0 P C B	0 P C B	
Row Crops	0 P C B	0 P C B	
Pasture/Range/Hay Field	0 P C B	0 P C B	
Logging Operations	0 P C B	0 P C B	
Mining Activity	0 P C B	0 P C B	

FISH COVER/ OTHER (10m x 20m Plot)

COVER CATEGORIES
0 = Absent (0%) 1 = Spare (<10%) 2 = Moderate (10-40%) 3 = Heavy (40-75%) 4 = Very Heavy (>75%)

	In-Channel Cover (circle one)	Flag
Filamentous Algae	0 1 2 3 4	
Macrophytes	0 1 2 3 4	
Woody Debris >0.3 m (BIG)	0 1 2 3 4	
Brush/Woody Debris <0.3 m (SMALL)	0 1 2 3 4	
Live Trees in Stream	0 1 2 3 4	
Overhanging Veg. =<1 m of Surface	0 1 2 3 4	
Undercut Banks	0 1 2 3 4	
Boulders/Ledges	0 1 2 3 4	
Artificial Structures	0 1 2 3 4	

CHANNEL CONSTRAINT

DISTANCE FROM SHORE TO RIPARIAN VEGETATION (M) XXX ____

CIRCLE ONE

C	Channel is Constrained.
B	Channel is in Broad Valley but Constrained by Incision.
N	Channel is in Narrow Valley but NOT very constrained.
U	Channel is Unconstrained in Broad Valley.

CHECK ONE

○ YES	I COULD READILY SEE OVER THE BANK.
○ NO	I COULD NOT READILY SEE OVER THE BANK.

FLAG ____

Flag	Comments

CANOPY DENSITY @ BANK DENSIOMETER (0 TO 17 MAX)

UP	
DOWN	
LEFT	
RIGHT	
FLAG	

Flag Codes: K = no measurement made; U = suspect or non-standard measurement; F1, F2, etc. = flags assigned by each field crew. Explain all flags in comments section on this side.

22399

PHAB: THALWEG PROFILE FORM - BOATABLE

Reviewed by (initial): _____

SITE ID: FW08 DATE: ___/___/2 0___

TRANSECT: ○ A-B ○ B-C ○ C-D ○ D-E ○ E-F ○ F-G ○ G-H ○ H-I ○ I-J ○ J-K

SUBSTRATE CODES	CHANNEL HABITAT CODES	OTHER
BH = BEDROCK/HARDPAN (SMOOTH OR ROUGH) - (LARGER THAN A CAR) BL = BOULDER (250 TO 4000 mm) - BASKETBALL TO CAR) CB = COBBLE (64 TO 250 mm) - (TENNIS BALL TO BASKETBALL) GR = COARSE TO FINE GRAVEL (2 TO 64 mm) - (LADYBUG TO TENNIS BALL) SA = SAND (0.06 TO 2 mm) - (GRITTY - UP TO LADYBUG SIZE) FN = SILT/ CLAY / MUCK - (NOT GRITTY) OT = OTHER (COMMENT ON OTHER SIDE)	PO = Pool GL = Glide RI = Riffle RA = Rapid CA = Cascade FA = Falls DR = Dry Channel	Off Channel = Off Channel or Backwater

REMEMBER: A = Upstream end of Reach and K = Downstream end of Reach.

THALWEG PROFILE

STATION	SNAG (circle one)	DEPTH (Either) UNITS: ○ ft ○ m SONAR XX	DEPTH (Either) UNITS: ○ ft ○ m POLE X.X	SUBSTRATE Circle one Substrate Code for each station	CHANNEL HABITAT Circle one Channel Habitat Code for each station	OFF CHAN. (circle one)	FLAG
0	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
1	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
2	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
3	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
4	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
5	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
6	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
7	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
8	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
9	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
10	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	
11	Y N			BH BL CB GR SA FN OT	PO GL RI RA CA FA DR	Y N	

FLAG	COMMENT

Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, etc. = flag assigned by field crew. Explain all flags in comment sections.

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FISH GEAR AND VOUCHER/TISSUE SAMPLE INFORMATION - WADEABLE/BOATABLE

Reviewed by (Initials): _____

SITE ID: FW08 DATE: ___ / ___ / 2 0 PAGE: ___ of ___

38513

O Not Fished - No Permit
O Not Fished - Permit Restriction
O Not Fished - Equipment Failure
O Not Fished - Site Conditions Prohibit Sampling
O Not Fished - Other (Explain Below)
O Fished - None Collected

O Fished All 10 Subreaches O Fished 5-9 Subreaches O Fished 1-4 Subreaches

COMMENT

Water Visibility: O Good O Poor Water Temp (°C): _____ Cond (uS) _____ O More than 1 method used to collect fish?

ELECTROFISH O BOAT O RAFT O BP O BANK/TOW No. of Netters: _____ Anodes: Number _____ Diameters _____ O in. O cm Wave Form: O AC O DC O Pulsed DC

Volts: (50-1000) _____ Watts: likely 400 (bp), 2500 or 5000 (boat/raft) _____ Pulse Rate: pps or Hz _____ Amps: (may not be provided for bp) _____ Pulse Width (ms) _____

Total Shock (button) Time (s) _____ Total Fishing Time (min) _____ Reach Length Sampled (m) _____ Avg. Subreach Length (m) _____ Electrofish Flag: ☐

VOUCHER SAMPLE INFORMATION O NO VOUCHERS PRESERVED

Sample ID	Sample Category *	Preserved	Comments
	O P O D	O	
	O P O D	O	

FISH TISSUE SAMPLES O NO SAMPLE COLLECTED

	SAMPLE ID	Common Name	Total Length(mm)	Subreach	Frozen	Comments
.1					O	
.2					O	
.3					O	
.4					O	
.5					O	

FLAG	COMMENT

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = flags assigned by each field crew. Explain all flags in comments. LENGTH* - Enter single fish as minimum.
*Sample Category P = Primary D = Field Duplicate
04/07/2009 NRSA Fish Gear

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FISH COLLECTION FORM - LG. WADEABLE OR BOATABLE/RAFTABLE

Reviewed by (Initials): _____

SITE ID: FW08 DATE: ___/___/2 0___ PAGE: ___ of ___

SUBREACH: ○ A-B ○ B-C ○ C-D ○ D-E ○ E-F ○ F-G ○ G-H ○ H-I ○ I-J ○ J-K | Shock Time _____ (s) | Distance Sampled _____ (m) | ○ FISHED NONE COLLECTED

DOMINANT SUBREACH CHARACTERISTICS FLAG (for subreach characteristics) : ____

SUBSTRATE ○ Fine/Sand ○ Gravel ○ Cobble-Boulder ○ Other _____ | COVER ○ Macrophytes ○ LWD ○ Overhanging Veg. ○ Other _____

DEPTH ○ < 0.5 m ○ > 0.6 to 2 m ○ > 2 m | BANK ○ Wooded ○ Herbaceous ○ Barren ○ Armored ○ Riprap ○ Other _____

Tag No.	Common Name	Tally	Total Count	Vouch. Count	LENGTH (mm) * Min	LENGTH (mm) * Max	Anom. Count	Mortality Count	Photo(s) Taken	Final Count	Flag
									○ Y		
									○ Y		
									○ Y		
									○ Y		
									○ Y		
									○ Y		
									○ Y		
									○ Y		
									○ Y		
									○ Y		
									○ Y		

Flag	Comment

Tag	Photo File

35580

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = flags assigned by each field crew. Explain all flags in comments. LENGTH* - Enter single fish as minimum.

04/07/2009 NRSA Fish Collection Boatable

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RIPARIAN "LEGACY" TREES AND INVASIVE ALIEN PLANTS

Reviewed by (initial): _____

SITE ID: FW08 DATE: ____ / ____ / 2 0 ____

41234

Tran	LARGEST POTENTIAL LEGACY TREE VISIBLE FROM THIS STATION						ALIEN PLANT SPECIES PRESENT IN LEFT AND RIGHT RIPARIAN PLOTS, AND INSTREAM FISH COVER PLOT
	Trees not Visible	DBH (m)	Height (m)	Dist. from wetted margin (m)	Type	Taxonomic Category	Check all that are present
A	◯	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		◯ NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
B	◯	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		◯ NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
C	◯	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		◯ NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced

INSTRUCTIONS

Potential Legacy trees are defined as the largest tree within your search area, which is as far as you can see, but within maximum limits as follows:

Wadeable Streams: Confine search to no more than 50 m from left and right bank and extending upstream to next transect (for 'K' look upstream 4 channel widths)

Non-wadeable Rivers: Confine search to no more than 100 m from left and right bank and extending both upstream and downstream as far as you can see confidently.

Alien Plants: Confine search to riparian plots on left and right bank

Wadeable Streams: 10 m x 10 m

Non-wadeable Rivers: 10 m x 20 m

Not all aliens are to be identified in all states. See Field Manual and Plant Identification Guide.

TAXONOMIC CATEGORIES

Acacia/Mesquite
Alder/Birch
Ash
Maple/Boxelder
Oak
Poplar/Cottonwood
Sycamore
Willow
Unknown or Other Deciduous

Cedar/Cypress/Sequoia
Fir (including Douglas fir and hemlock)
Juniper
Pine
Spruce
Unknown or Other Conifer

Unknown or Other Broadleaf Evergreen

Snag (Dead tree of any species)

ALIEN SPECIES

E Wtrmilf	Eurasian water milfoil	Myriophyllum spicatum
Hydrilla	Hydrilla	Hydrilla verticillata
E Wtrchest	European water chestnut	Trapa natans
W Hyacinth	Water Hyacinth	Eichhornia crassipes
Ylw Fltheart	Yellow Floating Heart	Nymphoides peltata
P Lstrife	Purple loosestrife	Lythrum salicaria
G Reed	Giant Reed	Arundo donax
Flwr Rush	Flowering Rush	Butomus umbellatus
Salt Ced	Salt Cedar	Tamarix spp.
MF Rose	Multi-flora rose	Rosa multiflora
Spurge	Leafy Spurge	Euphorbia esula

COMMENTS

Transects D to K continued on other side

RIPARIAN "LEGACY" TREES AND INVASIVE ALIEN PLANTS

Reviewed by (initial): _____

SITE ID: FW08 DATE: ___ / ___ / 2 0

41234

Tran	LARGEST POTENTIAL LEGACY TREE VISIBLE FROM THIS STATION						ALIEN PLANT SPECIES PRESENT IN LEFT AND RIGHT RIPARIAN PLOTS, AND INSTREAM FISH COVER PLOT	
	Trees not Visible	DBH (m)	Height (m)	Dist. from wetted margin (m)	Type	Taxonomic Category		Check all that are present
D	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
E	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
F	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
G	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
H	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
I	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
J	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
K	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf ○ Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced

CHANNEL CONSTRAINT FORM - WADEABLE/BOATABLE

Reviewed by (initial): _____

SITE ID: FW08 DATE: ____ / ____ / ____

CHANNEL CONSTRAINT

CHANNEL PATTERN (Fill in one)

- ○ **One channel**
- ○ **Anastomosing (complex) channel** - (Relatively long major and minor channels branching and rejoining.)
- ○ **Braided channel** - (Multiple short channels branching and rejoining - mainly one channel broken up by numerous mid-channel bars.)

CHANNEL CONSTRAINT(Fill in one)

- ○ **Channel very constrained in V-shaped valley** (i.e. it is very unlikely to spread out over valley or erode a new channel during flood)
- ○ **Channel is in Broad Valley** but channel movement by erosion during floods is **constrained by Incision** (Flood flows do not commonly spread over valley floor or into multiple channels.)
- ○ **Channel is in Narrow Valley but is not very constrained**, but limited in movement by relatively narrow valley floor (< ~10 x bankfull width)
- ○ **Channel is Unconstrained in Broad Valley** (i.e. during flood it can fill off-channel areas and side channels, spread out over flood plain, or easily cut new channels by erosion)

CONSTRAINING FEATURES (Fill in one)

- ○ **Bedrock** (i.e. channel is a bedrock-dominated gorge)
- ○ **Hillslope** (i.e. channel constrained in narrow V-shaped valley)
- ○ **Terrace** (i.e. channel is constrained by its own incision into river/stream gravel/soil deposits)
- ○ **Human Bank Alterations** (i.e. constrained by rip-rap, landfill, dike, road, etc.)
- ○ **No constraining features**

Percent of channel length with margin in contact with constraining feature: ____ % ---> (0-100%)

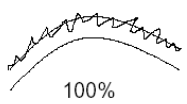
Bankfull width: ____ (m)

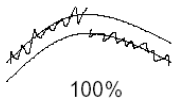
Valley width (Visual Estimated Average): ____ (m)

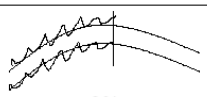
Note: Be sure to include distances between both sides of valley border for valley width.

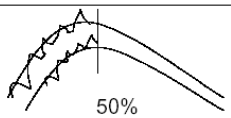
If you cannot see the valley borders, record the distance you can see and mark this box. ○

Percent of Channel Margin Examples









Comments

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Reviewed by (Initials): _____

TORRENT EVIDENCE ASSESSMENT FORM

SITE ID: FW08 DATE: ___/___/2 0___

TORRENT EVIDENCE	
Please fill in any of the following that are evident.	
EVIDENCE OF TORRENT SCOURING:	
○	01 - Stream channel has a recently devegetated corridor two or more times the width of the low flow channel. This corridor lacks riparian vegetation with possible exception of fireweed, even-aged alder or cottonwood seedlings, grasses, or other herbaceous plants.
○	02 - Stream substrate cobbles or large gravel particles are NOT IMBRICATED. (Imbricated means that they lie with flat sides horizontal and that they are stacked like roof shingles -- imagine the upstream direction as the top of the "roof." a torrent scour or deposition channel, the stones are laying in unorganized patterns, lying "every which way." In addit many of the substrate particles are angular (not "water-worn.")
○	03 - Channel has little evidence of pool-riffle structure. (For example, could you ride a mountain bike down the channe
○	04 - The stream channel is scoured down to bedrock for substantial portion of reach.
○	05 - There are gravel or cobble berms (little levees) above bankfull level.
○	06 - Downstream of the scoured reach (possibly several miles), there are massive deposits of sediment, logs, and othe debris.
○	07 - Riparian trees have fresh bark scars at many points along the stream at seemingly unbelievable heights above the channel bed.
○	08 - Riparian trees have fallen into the channel as a result of scouring near their roots.
EVIDENCE OF TORRENT DEPOSITS:	
○	09 - There are massive deposits of sediment, logs, and other debris in the reach. They may contain wood and boulder that, in your judgement, could not have been moved by the stream at even extreme flood stage.
○	10 - If the stream has begun to erode newly laid deposits, it is evident that these deposits are "MATRIX SUPPORTED." This means that the large particles, like boulders and cobbles, are often not touching each other, but have silt, sand, a other fine particles between them (their weight is supported by these fine particles -- in contrast to a normal stream deposit, where fines, if present, normally "fill-in" the interstices between coarser particles.)
NO EVIDENCE:	
○	11 - No evidence of torrent scouring or torrent deposits

COMMENTS

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VISUAL ASSESSMENT FORM - WADEABLE/BOATABLE (Front)

Reviewed by (initial): _____

SITE ID: FW08 DATE: ___ / ___ / 2 0 ___

WATERSHED ACTIVITIES AND DISTURBANCES OBSERVED (Intensity: Blank=Not observed, L=Low, M=Moderate, H=Heavy)

Residential	Recreational	Agricultural	Industrial	Stream Management
L M H Residences	L M H Hiking Trails	L M H Cropland	L M H Industrial Plants	L M H Liming
L M H Maintained Lawns	L M H Parks, Campgrounds	L M H Pasture	L M H Mines/Quarries	L M H Chemical Treatment
L M H Construction	L M H Primitive Parks, Camping	L M H Livestock Use	L M H Oil/Gas Wells	L M H Angling Pressure
L M H Pipes, Drains	L M H Trash/Litter	L M H Orchards	L M H Power Plants	L M H Dredging
L M H Dumping	L M H Surface Films	L M H Poultry	L M H Logging	L M H Channelization
L M H Roads		L M H Irrigation Equip.	L M H Evidence of Fire	L M H Water Level Fluctuations
L M H Bridge/Culverts		L M H Water Withdrawal	L M H Odors	L M H Fish Stocking
L M H Sewage Treatment			L M H Commercial	L M H Dams

SITE CHARACTERISTICS (200 m radius)

Waterbody Character	Pristine	○ 5	○ 4	○ 3	○ 2	○ 1	Highly Disturbed
	Appealing	○ 5	○ 4	○ 3	○ 2	○ 1	Unappealing
Beaver	Beaver Signs:	○ Absent	○ Rare	○ Common			
	Beaver Flow Modifications:	○ None	○ Minor	○ Major			
Dominant Land Use	Dominant Land Use Around 'X'	○ Forest	○ Agriculture	○ Range	○ Urban	○ Suburban/Town	
	If Forest, Dominant Age Class	○ 0 - 25 yrs.	○ 25 - 75 yrs	○ > 75 yrs.			

WEATHER

GENERAL ASSESSMENT (Biotic in[illegible], Vegetation diversity, Local anecdotal information)

VISUAL ASSESSMENT FORM - WADEABLE/BOATABLE (Back)

Reviewed by (initial): ____

SITE ID: FW08

DATE: ___ / ___ / 2 0 ___

GENERAL ASSESSMENT (continued)

INVASIVE OR NUISANCE SPECIES OF LOCAL INTEREST

Record species of plants and animals that were observed but are not on the invasive plant form. Examples would be Zebra Mussel or New Zealand Mud Snail, or invasive plants or animals of concern to a particular state. Indicate your level of confidence in your identification, and provide some idea of how prevelant it is in the sampling reach or adjacent riparian area.

Species (Common Name)	Confidence	Prevalence	Comments
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	

37922

04/07/2009 NRSA Visual Assessment

TRACKING AND SAMPLE STATUS - WRS

SITE ID: FW08 **Visit #:** ○ 1 ○ 2 Date Collected: ____ / ____ / 2 0 ____

SENT BY: _____ SENDER PHONE: _____

State of Site Location: _____ TEAM: _____ DATE SENT: ____ / ____ / 2 0 ____

SHIPPED BY: ○ FedEx ○ UPS ○ Hand Delivery ○ Other: _____ AIRBILL/TRACKING NUMBER: _____

Site Status Report

SAMPLEABLE	NOT SAMPLEABLE	Temporarily Not Sampleable
○ Wadeable	○ Dry - Visited	○ Not Boatable
○ Boatable	○ Dry - Not Visited	○ Not Wadeable
○ Partial Wadeable	○ Wetland	○ Other
○ Partial Boatable	○ Map Error	NO ACCESS
○ Wadeable Interrupted	○ Impounded	○ Access Denied
○ Boatable Interrupted	○ Other	○ Inaccessible
○ Altered		○ Temp Inaccessible

SAMPLE STATUS

○ **No Samples Collected**

Mark the samples that were collected during this site visit:

○ Water Chem (CHEM)	○ Enterococci (ENTE)
○ Water Chl (WCHL)	○ Sediment (SEDE)
○ Water Chem (PPCP)	○ Fish Tissue (FTIS)
○ Periphyton Chl (PCHL)	○ Fish Voucher (VERT)
○ Periphyton Bio (PBIO)	○ Bent Reachwide (BERW)
○ Periphyton ID (PERI)	○ Bent Low Gradient (BELG)
○ Periphyton APA (PAPA)	

Status Comments

Sample ID	Sample Type	Comments
_____	C H E M	
_____	W C H L	
_____ . 2	P C H L	
_____ . 3	P B I O	
_____	C H E M	
_____	W C H L	
_____ . 2	P C H L	
_____ . 3	P B I O	

Sample Types	Condition Codes	Chain of Custody	Contact Information
CHEM - Water chemistry WCHL -Water Column Chlorophyll PCHL - Periphyton Chlorophyll PBIO - Periphyton Biomass	Filled in by recipient C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK = Sample OK T = Thawed	Filled in by recipient Date Received: ____/____/ _____ Received by: _____	Tracking Help: Marlys Cappaert PH: 541-754-4467 Lab: Attn: Phil Monaco, Dynamac c/o U.S. EPA 1350 Goodnight Ave Corvallis, OR 97333 PH: 541-754-4787 monaco.phil@epamail.epa.gov

FAX THIS FORM TO 541-754-4637
OR READ TRACKING INFO TO VOICE MESSAGE CENTER:
541-754-4663

52109

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TRACKING - NERL Cincinnati

SITE ID: FW08 **Visit #:** ○ 1 ○ 2 Date Collected: __/__/20__

SENT BY: _____ SENDER PHONE: _____

State of Site Location: ____ TEAM: _____ DATE SENT: __/__/20__

SHIPPED BY: ○ FedEx ○ UPS ○ Hand Delivery ○ Other: _____ AIRBILL/TRACKING NUMBER: _____

Sample ID	Sample Type	Comments	Condition Code
	P P C P		

Sample Types	Condition Codes	Chain of Custody	Contact Information
PPCP - Water chemistry	Filled in by recipient C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK = Sample OK T = Thawed	Filled in by recipient Date Received: ____/____/____ Received by: _____	Tracking Help: Marlys Cappaert PH: 541-754-4467 Lab: NERL -Cincinnati Attn: Dr. Angela Batt 26 W. Martin Luther King Drive MS 642 Cincinnati, OH 45268 513-569-7284 batt.angela@epa.gov

FAX THIS FORM TO 541-754-4637
OR READ TRACKING INFO TO VOICE MESSAGE CENTER:
541-754-4663

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TRACKING (BATCHED OR RETAINED SAMPLES) National Rivers and Streams Assessment

Include only all BATCHED or RETAINED samples on one form.

SENT BY: _____ SENDER PHONE: _____ STATE OF SITE LOCATION: _____ TEAM: _____

BATCHED SAMPLES - UNPRESERVED samples that will be batched and shipped within 2 weeks.

SHIPPED BY: ○ FedEx ○ UPS ○ Hand Delivery DATE SHIPPED: ____ / ____ / 2 0 ____

AIRBILL/TRACKING NUMBER: _____

RETAINED SAMPLES - PRESERVED samples that will be stored longer than a month at a holding facility.

○ Held at address: _____

Site ID	Date Sample Collected MM/DD/YYYY	Visit	Sample ID	Sample Type	# of Containers	Comments	Cond. Code
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					

Lab	Chain of Custody	Sample Types	Condition Codes
○ ACADEMY OF NATURAL SCIENCES - PHIL, PA ○ BENTHIC LAB ○ GLEC ○ MED - DULUTH, MN ○ MICHIGAN STATE UNIV. ○ NERL - N. CHELMSFORD, MA ○ NERL - CINCINNATI, OH ○ OTHER _____	**Filled in by recipient** Date Received: _____/_____/_____ Received by: _____ **Tracking Help:** Marlys Cappaert p) 541-754-4467	**PRESERVED - RETAINED:** BERW - Benthos Reach Wide BELG - Benthos Low Gradient VERT - Fish Vouchers PERI - Periphyton ID (.1) **UNPRESERVED - BATCHED:** SEDE - Sediment Enzyme FTIS - Fish Tissue PAPA - Periphyton APA (.4) ENTE - Enterococci	**Filled in by recipient** C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK = Sample OK T = Thawed

42504

FAX THIS FORM TO 541-754-4637 OR READ TRACKING INFO TO VOICE MESSAGE CENTER: 541-754-4663

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WADEABLE FORMS PACKET

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STREAM VERIFICATION FORM - WADEABLE/BOATABLE (Front)

Reviewed by (initial): _____

SITE NAME: _____ DATE: ___/___/ 2 0 ___ VISIT: ○ 1 ○ 2 ○ 3

SITE ID: FW08 State of Site Location: _____ Don't forget to record Reach Length on back. TEAM: _____

STREAM/RIVER VERIFICATION INFORMATION

Stream/River Verified by (fill in all that apply) ○ GPS ○ Local Contact ○ Signs ○ Roads ○ Topo. Map
○ Other (Describe Here): ○ Not Verified (Explain in Comments)

Coordinates		Latitude North	Longitude West	# of Satellites	Are GPS Coordinates w/i 10 Sec. of map?
MAP	Degrees, Minutes, and Seconds OR Decimal Degrees	___ ___ ___ . ___	___ ___ ___ . ___	○ ≤3	○ Yes ○ No
GPS	Degrees, Minutes, and Seconds OR Decimal Degrees	___ ___ ___ . ___	___ ___ ___ . ___	○ ≥4	GPS Datum Used (e.g. NAD27): _____

DID YOU SAMPLE THIS SITE?

○ YES If YES, check one below

SAMPLEABLE (Choose method used)
- ○ Wadeable - Continuous water, greater than 50% wadeable
- ○ Boatable
- ○ Partial - Sampled by wading (>50% of reach sampled). Explain below
- ○ Partial - Sampled by boat (>50% of reach sampled). Explain below.
- ○ Wadeable Interrupted - Not continuous water along reach
- ○ Boatable Interrupted - Not continuous water along reach
- ○ Altered - Stream/River Channel Present but differs from Map

○ NO If NO, check one below

NON-SAMPLEABLE-PERMANENT
- ○ Dry - Visited
- ○ Dry - Not visited
- ○ Wetland (No Definable Channel)
- ○ Map Error - No evidence channel/waterbody ever present
- ○ Impounded (Underneath Lake or Pond)
- ○ Other (explain in comments)

NON-SAMPLEABLE-TEMPORARY
- ○ Not boatable - Need a different crew - Reschedule for this year
- ○ Not wadeable - Need a different crew - Reschedule for this year
- ○ Other (Explain in comments)

NO ACCESS
- ○ Access Permission Denied
- ○ Permanently Inaccessible (Unable/Unsafe to Reach Site)
- ○ Temporarily Inaccessible-Fire, etc. - Reschedule for next year

GENERAL COMMENTS:

DIRECTIONS TO STREAM/RIVER SITE:

Record information used to define length of reach, and sketch general features of reach on reverse side.

STREAM VERIFICATION FORM - WADEABLE/BOATABLE (Back)

Reviewed by (initial): _____

SITE NAME: _____ DATE: ____ / ____ / 2 0 ____ VISIT: ○ 1 ○ 2 ○ 3

SITE ID: FW08 TEAM: _____

STREAM/RIVER REACH DETERMINATION

Channel Width Used to Define Reach (m)	DISTANCE (m) FROM X-SITE		Total Reach Length Intended (m)	Comment
	Upstream Length	Downstream Length		

SKETCH MAP - Arrow Indicates North; Mark site L=Launch X=Index T= Take Out
NOTE: If an outline map is attached here, use a continuous strip of clear tape across the top edge.
You can also attach a separate sheet with the outline map on it.
For boatable sites you can attach topo map with reach, X-site and transect locations marked.

PERSONNEL

NAME	Bio/Chem Sampling	Habitat	Forms Review
_____	○	○	○
_____	○	○	○
_____	○	○	○
_____	○	○	○
_____	○	○	○

FIELD MEASUREMENT FORM - WADEABLE

Reviewed by (initial): _____

SITE ID: FW08 DATE: ___/___/___

CALIBRATION INFORMATION

Instrument manufacturer and model: _____

Instrument ID number: _____ Operator: _____

TEMPERATURE	Thermometer Reading (℃)	Sensor Reading (℃)	Flag	Comments		
	___.___	___.___				
DO	Elevation	OR Barometric Pressure(mm Hg)	Calibration Value		Displayed Value	Flag
	○ ft ○ m		___.___ ○ mg/L ○ %		___.___ ○ mg/L ○ %	
pH	Cal. STD 1 Description	Cal. STD 1 Value	Cal. STD 2 Description		Cal. STD 2 Value	
		___.___			___.___	
	Calibration Verified with Quality Control Sample (QCS)					
	QCS Description		QCS True	QCS Measured		Flag
			___.___	___.___		
CONDUCTIVITY	Cal. STD 1 Description	Cal. STD 1 Value	Cal. STD 2 Description		Cal. STD 2 Value	
	Calibration Verified with Quality Control Sample (QCS)					
	QCS Description		QCS True (µS/cm @25℃)	QCS Measured (µS/cm @25℃)		Flag

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = flags assigned by field crew. Explain all flags in comment sections.

Flag	Comments

Field Measurements ○ MID CHANNEL ○ OTHER		Comments
TRANSECT: ☐ Time of Day (hh:mm)	___:___	
DO(mg/L) XX.X	___.___	
Temp. (℃) XX.X	___.___	
pH XX.XX	___.___	
Cond. (µS/cm)XX.X	_____	
Corrected to 25℃ ?	○ Y ○ N	

52151

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DISCHARGE FORM - WADEABLE

Reviewed by (Initials): _____

SITE ID: FW08 DATE: ___/___/20___

○ Velocity Area

Distance Units	Depth Units	Velocity Units
○ ft ○ cm	○ ft ○ cm	○ ft/s XX.X ○ m/s X.XX

	Dist. from Bank	Depth	Velocity	Flag
1	0			
2				
3				
4				
5				
6				
7				
8				
9				
10				
11				
12				
13				
14				
15				
16				
17				
18				
19				
20				

○ Timed Filling

Repeat	Volume (L)	Time (s)	Flag
1	___.___	___.___	
2	___.___	___.___	
3	___.___	___.___	
4	___.___	___.___	
5	___.___	___.___	

○ Neutral Bouyant Object

	Float 1	Float 2	Float 3
Float Dist. ○ ft ○ m			
Float Time (s)			
Flag			

Cross Sections on Float Reach

	Upper Section	Middle Section	Lower Section
Width ○ ft ○ m	___.___	___.___	___.___
Depth 1 ○ ft ○ cm			
Depth 2			
Depth 3			
Depth 4			
Depth 5			

○ Q Value

If discharge is determined directly in field, record value here: Q = _____ ○ cfs ○ m^3/s FLAG ____

Flag	Comments

Flag Codes: K = No measurement or observation made; U = Suspect measurement or observation; Q = Unacceptable QC check associated with measurement; Z = Last station measured (if not Station 20); F1, F2, etc. = Miscellaneous flags assigned by each field crew. Explain all flags in comments section.

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46387

Reviewed by (initial): ______

SAMPLE COLLECTION FORM - WADEABLE (Front)

SITE ID: FW08

DATE: ___ / ___ / 2 0 ___

WATER CHEMISTRY (4-L CUBITAINER)

No Sample Collected ○

Sample ID	Sample Category *	Chilled	Comments
	○ P ○ D	○	
	○ P ○ D	○	

WATER COLUMN CHLOROPHYLL (Target Volume = 1000 mL; max vol = 2000 mL)

No Sample Collected ○

Sample ID	Sample Category *	Volume Filtered (mL)	Frozen	Comments
	○ P ○ D		○	
	○ P ○ D		○	

WATER CHEMISTRY PPCP (Amber Glass Bottle)

No Sample Collected ○

Sample ID	Sample Category *	Chilled	Comments
	○ P ○ D	○	
	○ P ○ D	○	

REACH WIDE BENTHOS SAMPLE

No Sample Collected ○

TRANSECT		A		B		C		D		E		F		G		H		I		J		K	
SUBSTRATE	CHAN.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.
Fine/Sand	Pool	○ F	○ P	○ F	○ P	○ F	○ P	○ F	○ P	○ F	○ P	○ F	○ P	○ F	○ P	○ F	○ P	○ F	○ P	○ F	○ P	○ F	○ P
Gravel	Glide	○ G	○ GL	○ G	○ GL	○ G	○ GL	○ G	○ GL	○ G	○ GL	○ G	○ GL	○ G	○ GL	○ G	○ GL	○ G	○ GL	○ G	○ GL	○ G	○ GL
Coarse	Riffle	○ C	○ RI	○ C	○ RI	○ C	○ RI	○ C	○ RI	○ C	○ RI	○ C	○ RI	○ C	○ RI	○ C	○ RI	○ C	○ RI	○ C	○ RI	○ C	○ RI
Other:	Rapid	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA	○ O	○ RA

Sample ID	Sample Category *	NO. Jars	Pre-served	Comments
	○ P ○ D		○	
	○ P ○ D		○	

LOW - GRADIENT BENTHOS SAMPLE

No Sample Collected ○

Transect	A	B	C	D	E	F	G	H	I	J	K
Location (LCR):	L ○ C ○ R ○	L ○ C ○ R ○	L ○ C ○ R ○	L ○ C ○ R ○	L ○ C ○ R ○	L ○ C ○ R ○	L ○ C ○ R ○	L ○ C ○ R ○	L ○ C ○ R ○	L ○ C ○ R ○	L ○ C ○ R ○
Dominant Substrate: (ONE PER TRANSECT)	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT	○ F ○ C ○ G ○ OT
Channel: (ONE PER TRANSECT)	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT	○ P ○ RA ○ RI ○ GL ○ OT
Dominant Edge: (L and R) (ONE PER TRANSECT)	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT	○ U ○ S ○ R ○ M ○ L ○ OG ○ OT

Edge:
U = Undercut S = Snag R = Rootwad M = Macrophyte bed
L = Leaf Litter OG = Organic deposits OT = Other or Co-Dominant
(Explain in comment section below)

Substrate:
F = Fine/Sand C = Coarse substrate
G = Gravel OT = Other (Explain in comment section below)

Channel:
P = Pool RI = Riffle GL = Glide
RA = Rapid OT = Other (Explain in comment section below)

Sample ID	Sample Category *	No. Jars	Pre-served	Comments
	○ P ○ D		○	
	○ P ○ D		○	

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = misc. flags assigned by field crew. Explain all flags in comment sections. ***Sample Categories: P = Primary, D = Field Duplicate**

04/07/2009 NRSA Sample Collection Wadeable 2009

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SAMPLE COLLECTION FORM - WADEABLE (Back)

Reviewed by (initial): _____

SITE ID: FW08 DATE: ___/___/2 0___

COMPOSITE PERIPHYTON SAMPLE - Primary

No Sample Collected ○

Sample ID	Sample Category *	Composite Volume (mL)	Number of transects sampled (0-11):	Flag
	○ P ○ D			

Assemblage ID (.1) (50-mL tube)			Chlorophyll (.2) (GF/F Filter)			Biomass (.3) (GF/C Filter)			APA (.4) (50-mL tube)		
Sample Vol. (mL)	Flag	Preserved	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen
		○			○			○			○

COMPOSITE PERIPHYTON SAMPLE

No Sample Collected ○

Sample ID	Sample Category *	Composite Volume (mL)	Number of transects sampled (0-11):	Flag
	○ P ○ D			

Assemblage ID (.1) (50-mL tube)			Chlorophyll (.2) (GF/F Filter)			Biomass (.3) (GF/C Filter)			APA (.4) (50-mL tube)		
Sample Vol. (mL)	Flag	Preserved	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen	Sample Vol. (mL)	Flag	Frozen
		○						○			○

Flag	Comments

Flag codes: K = No measurement or observation made; U = Suspect measurement or observation; F1, F2, etc. = flags assigned by field crew. Explain all flags in comment sections.

SEDIMENT / CHEMISTRY / ENZYMES

No Sample Collected ○

Sample ID	Sample Category *	Composite Volume	No. Transects	Chilled	Comments
	○ P ○ D			○	
	○ P ○ D			○	

ENTEROCOCCI (Target Volume = 250 mL)

No Sample Collected ○

Sample ID One unique ID per line	Sample Category *	Time Collected (hhmm)	Depth Collected (m)	Sample Volume (mL)	Filt. Start Time (hhmm)	Volume Filtered (Target = 50 mL) **				Filt. End Time (hhmm)	Time Frozen (hhmm)	Flag
						Filt. 1	Filt. 2	Filt. 3	Filt. 4			
	○ P ○ D											
	○ P ○ D											
	○ P ○ D											
	○ F											

Flag	Comment

* **Sample Categories: P = Primary; D = Field Duplicate; F = Filter Blank (Enterococci sample only)** Filter blank is collected at visit where field duplicate sample is NOT taken.
** If <25 ml of buffer solution was used to rinse filter, indicate with an F flag and note in comment section which filter(s) were affected along with the approximate volume(s) of buffer solution used.

PHab: CHANNEL/RIPARIAN CROSS-SECTION FORM - WADEABLE

Reviewed by (Initials):_____

SITE ID: FW08 DATE: ___/___/20___ TRANSECT: ○A ○B ○C ○D ○E ○F X-tra Side Channel ○G ○H ○I ○J ○K ○

SUBSTRATE CROSS-SECTIONAL INFORMATION

	Dist LB XX.XX m	Depth XXX cm	Size Class Code	Embed. 0-100%	Flag
Left					
LCtr					
Ctr					
RCtr					
Right					

SUBSTRATE SIZE CLASS CODES	Embed. (%)
RS = Bedrock (Smooth) - (Larger than a car)	0
RR = Bedrock (Rough) - (Larger than a car)	0
RC = Concrete/Asphalt	
XB = Large Boulder (1000 to 4000 mm) - (Meterstick to car)	
SB = Small Boulder (250 to 1000 mm) - (Basketball to meterstick)	
CB = Cobble (64 to 250 mm) - (Tennis ball to Basketball)	
GC = Coarse Gravel (16 to 64 mm) - (Marble to Tennis ball)	
GF = Fine Gravel (2 to 16 mm) - (Ladybug to marble)	
SA = Sand (0.06 to 2 mm) - (Gritty - up to Ladybug size)	100
FN = Silt / Clay / Muck - (Not Gritty)	100
HP = Hardpan - (Firm, Consolidated Fine Substrate)	0
WD = Wood - (Any Size)	
OT = Other (Write comment below)	

FISH COVER/ OTHER

0 = Absent (0%) 1 = Sparse (<10%) 2 = Moderate (10-40%) 3 = Heavy (40-75%) 4 = Very Heavy (>75%) (circle one)

	Cover in Channel	Flag
Filamentous Algae	0 1 2 3 4	
Macrophytes	0 1 2 3 4	
Woody Debris >0.3 m (BIG)	0 1 2 3 4	
Brush/Woody Debris <0.3 m (SMALL)	0 1 2 3 4	
Live Trees or Roots	0 1 2 3 4	
Overhanging Veg. =<1 m of Surface	0 1 2 3 4	
Undercut Banks	0 1 2 3 4	
Boulders	0 1 2 3 4	
Artificial Structures	0 1 2 3 4	

VISUAL RIPARIAN ESTIMATES

0 = Absent (0%) 1 = Sparse (<10%) 2 = Moderate (10-40%) 3 = Heavy (40-75%) 4 = Very Heavy (>75%)

D = Deciduous C = Coniferous E = Broadleaf Evergreen M = Mixed N = None

RIPARIAN VEGETATION COVER	Left Bank	Right Bank	Flag
Canopy (>5 m high)			
Woody Vegetation Type	D C E M N	D C E M N	
BIG Trees (Trunk >0.3 m DBH)	0 1 2 3 4	0 1 2 3 4	
SMALL Trees (Trunk <0.3 m DBH)	0 1 2 3 4	0 1 2 3 4	
Understory (0.5 to 5 m high)			
Woody Vegetation Type	D C E M N	D C E M N	
Woody Shrubs & Saplings	0 1 2 3 4	0 1 2 3 4	
Non-Woody Herbs, Grasses, & Forbs	0 1 2 3 4	0 1 2 3 4	
Ground Cover (<0.5 m high)			
Woody Shrubs & Saplings	0 1 2 3 4	0 1 2 3 4	
Non-Woody Herbs, Grasses and Forbs	0 1 2 3 4	0 1 2 3 4	
Barren, Bare Dirt or Duff	0 1 2 3 4	0 1 2 3 4	

HUMAN INFLUENCE	0 = Not Present P = >10 m C = Within 10 m B = On Bank		
	Left Bank	Right Bank	Flag
Wall/Dike/Revetment /Riprap/Dam	0 P C B	0 P C B	
Buildings	0 P C B	0 P C B	
Pavement/Cleared Lot	0 P C B	0 P C B	
Road/Railroad	0 P C B	0 P C B	
Pipes (Inlet/Outlet)	0 P C B	0 P C B	
Landfill/Trash	0 P C B	0 P C B	
Park/Lawn	0 P C B	0 P C B	
Row Crops	0 P C B	0 P C B	
Pasture/Range/Hay Field	0 P C B	0 P C B	
Logging Operations	0 P C B	0 P C B	
Mining Activity	0 P C B	0 P C B	

BANK MEASUREMENTS

	Bank Angle 0 - 360	Undercut Dist. (m)	Flag
Left			
Right			
Wetted Width XXX.X m			
Bar Width XX.X m			
Bankfull Width XXX.X m			
Bankfull Height XX.X m			
Incised Height XX.X m			

CANOPY COVER MEASUREMENTS

DENSIOMETER (0-17Max)

		Flag			Flag
CenUp			CenR		
CenL			Left		
CenDwn			Right		

Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, etc. = flag assigned by field crew. Explain all flags in comment sections.

Flag	Comments

6374

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PHAB: THALWEG PROFILE & WOODY DEBRIS FORM - WADEABLE

Reviewed by (initial):_____

SITE ID: FW08 DATE: ___ / ___ / 2 0 ___ TRANSECT: ○ A-B ○ B-C ○ C-D ○ D-E ○ E-F ○ F-G ○ G-H ○ H-I ○ I-J ○ J-K

THALWEG PROFILE

For Transect A-B ONLY: Increment (m) X.X: Total Reach Length (m):

STA-TION	THALWEG DEPTH (cm) (XXX)	WETTED WIDTH (m) (XXX.X)	BAR WIDTH[1] Present	BAR WIDTH[1] XX.X	SOFT /SMALL SEDI-MENT	CHANNEL UNIT CODE	POOL FORM CODE	SIDE CHANNEL	BACK WATER	THALWEG PROFILE COMMENTS
0			Y N		Y N			Y N	Y N	
1			Y N		Y N			Y N	Y N	
2			Y N		Y N			Y N	Y N	
3			Y N		Y N			Y N	Y N	
4			Y N		Y N			Y N	Y N	
*5			Y N		Y N			Y N	Y N	
6			Y N		Y N			Y N	Y N	
*7			Y N		Y N			Y N	Y N	
8			Y N		Y N			Y N	Y N	
9			Y N		Y N			Y N	Y N	
10			Y N		Y N			Y N	Y N	
11			Y N		Y N			Y N	Y N	
12			Y N		Y N			Y N	Y N	
13			Y N		Y N			Y N	Y N	
14			Y N		Y N			Y N	Y N	

SUBSTRATE	Station (5 or 7)	LFT	LCTR	CTR	RCTR	RGT	FLAG

FLAG	COMMENTS (for SUBSTRATE and LWD)

LARGE WOODY DEBRIS

(≥10 cm small end diameter; ≥1.5 m length)

FILL IN IF UNMARKED BOXES ARE ZERO ○

FLAG

DIAMETER LARGE END	PIECES ALL/PART IN BANKFULL CHANNEL Length 1.5-5m	5-15m	>15m	PIECES BRIDGE ABOVE BANKFULL CHANNEL Length 1.5-5m	5-15m	>15m
0.1-<0.3 m						
0.3-0.6 m						
0.6-0.8 m						
>0.8 m						

SUBSTRATE SIZE CLASS CODES	POOL FORM CODES	CHANNEL UNIT CODES
RS = BEDROCK (SMOOTH) - (LARGER THAN A CAR) RR = BEDROCK (ROUGH) - (LARGER THAN A CAR) RC = CONCRETE/ASPHALT XB = LG. BOULDER (1000 TO 4000 mm) - METERSTICK TO CAR) SB = SM. BOULDER (250 TO 1000 mm) - BASKETBALL TO METERSTICK) CB = COBBLE (64 TO 250 mm) - (TENNIS BALL TO BASKETBALL) GC = COARSE GRAVEL (16 TO 64 mm) - (MARBLE TO TENNIS BALL) GF = FINE GRAVEL (2 TO 16 mm) - (LADYBUG TO MARBLE) SA = SAND (0.06 TO 2 mm) - (GRITTY - UP TO LADYBUG SIZE) FN = SILT/ CLAY / MUCK - (NOT GRITTY) HP = HARDPAN - (FIRM, CONSOLIDATED FINE SUBSTRATE) WD = WOOD - (ANY SIZE) OT = OTHER (COMMENT ON OTHER SIDE)	N = Not a pool W = Large Woody Debris R = Rootwad B = Boulder or Bedrock F = Unknown, fluvial COMBINATIONS: eg. WR, BR, WRB	PP = Pool, Plunge PT = Pool, Trench PL = Pool, Lateral Scour PB = Pool, Backwater PD = Pool, Impoundment GL = Glide RI = Riffle RA = Rapid CA = Cascade FA = Falls DR = Dry Channel

56113

Flag Codes: K = no measurement made; U = suspect measurement; F1, F2, etc. = flags assigned by each field crew; G1, G2, etc. for flags not specific to one station. Explain all flags in comments.

1 = Measure Bar Width at Station 0 and Mid-Station (5 or 7).

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FISH GEAR AND VOUCHER/TISSUE SAMPLE INFORMATION - WADEABLE/BOATABLE

Reviewed by (Initials): _____

SITE ID: FW08 DATE: ___ / ___ / 2 0 PAGE: ___ of ___

38513

○ Not Fished - No Permit ○ Not Fished - Equipment Failure ○ Not Fished - Other (Explain Below)
○ Not Fished - Permit Restriction ○ Not Fished - Site Conditions Prohibit Sampling
○ Fished - None Collected

○ Fished All 10 Subreaches ○ Fished 5-9 Subreaches ○ Fished 1-4 Subreaches

COMMENT

Water Visibility: ○ Good ○ Poor Water Temp (℃): _____ Cond (uS) _____ ○ More than 1 method used to collect fish?

ELECTROFISH ○ BOAT ○ RAFT ○ BP ○ BANK/TOW No. of Netters: _____ Anodes: Number _____ Diameters _____ ○ in. ○ cm Wave Form: ○ AC ○ DC ○ Pulsed DC

Volts: (50-1000) _____ Watts: likely 400 (bp), 2500 or 5000 (boat/raft) _____ Pulse Rate: pps or Hz _____ Amps: (may not be provided for bp) _____ Pulse Width (ms) _____

Total Shock (button) Time (s) _____ Total Fishing Time (min) _____ Reach Length Sampled (m) _____ Avg. Subreach Length (m) _____ Electrofish Flag: ☐

VOUCHER SAMPLE INFORMATION ○ NO VOUCHERS PRESERVED

Sample ID	Sample Category *	Preserved	Comments
	○ P ○ D	○	
	○ P ○ D	○	

FISH TISSUE SAMPLES ○ NO SAMPLE COLLECTED

SAMPLE ID	Common Name	Total Length(mm)	Subreach	Frozen	Comments
.1				○	
.2				○	
.3				○	
.4				○	
.5				○	

FLAG	COMMENT

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = flags assigned by each field crew. Explain all flags in comments. LENGTH* - Enter single fish as minimum.
*Sample Category P = Primary D = Field Duplicate
04/07/2009 NRSA Fish Gear

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FISH COLLECTION FORM - WADEABLE

Reviewed by (Initials):______

SITE ID: FW08

DATE: ___ / ___ / 2 0 ___

PAGE: ___ of ___

Tag No.	Common Name	Tally	Total Count	Vouch. Count	LENGTH (mm)*		Anom. Count	Mortality Count	Photo(s) Taken	Final Count	Flag	Subreaches (Optional)
					Min	Max						
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J
									○ Y			○ A ○ C ○ E ○ G ○ I ○ B ○ D ○ F ○ H ○ J

Flag	Comment

Tag	Photo File

1133311205

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = flags assigned by each field crew. Explain all flags in comments. LENGTH* - Enter single fish as minimum.

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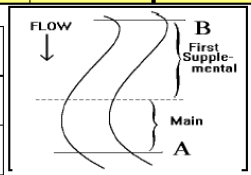
PHab: SLOPE AND BEARING FORM - WADEABLE

Reviewed by (initial): _____

SITE ID: FW08 DATE: ___/___/_____

TRANSECT & METHOD (Mark method for every Transect)	MAIN (always used)			FIRST SUPPLEMENTAL			SECOND SUPPLEMENTAL			FLAG
	Slope(%) or Elev. Diff. (cm) (Mark Units for every Transect)	BEARING 0 - 359	PROPOR-TION %	Slope(%) or Elev. Diff. (cm)	BEARING 0 - 359	PROPOR-TION %	Slope(%) or Elev. Diff. (cm)	BEARING 0 - 359	PROPOR-TION %	
A < B ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	. ○ % ○ cm			.			.			
B < C ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	. ○ % ○ cm			.			.			
C < D ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	. ○ % ○ cm			.			.			
D < E ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	. ○ % ○ cm			.			.			
E < F ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	. ○ % ○ cm			.			.			
F < G ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	. ○ % ○ cm			.			.			
G < H ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	. ○ % ○ cm			.			.			
H < I ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	. ○ % ○ cm			.			.			
I < J ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	. ○ % ○ cm			.			.			
J < K ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	. ○ % ○ cm			.			.			

FLAG	COMMENT



2104

Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, M (M = Method - used for method comment only) = flag assigned by field crew. Explain all flags in comment sections.

 CL=Clinometer; HL=Hand Level; LA=Laser rangefinder with electronic clinometer; TR=Transit, surveyors level or total station; WT=Water Tubing.

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RIPARIAN "LEGACY" TREES AND INVASIVE ALIEN PLANTS

Reviewed by (initial): _____

SITE ID: FW08 DATE: ____ / ____ / 2 0 ____

41234

Tran	LARGEST POTENTIAL LEGACY TREE VISIBLE FROM THIS STATION						ALIEN PLANT SPECIES PRESENT IN LEFT AND RIGHT RIPARIAN PLOTS, AND INSTREAM FISH COVER PLOT
	Trees not Visible	DBH (m)	Height (m)	Dist. from wetted margin (m)	Type	Taxonomic Category	Check all that are present
A	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		○ NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
B	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		○ NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
C	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		○ NONE ○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced

INSTRUCTIONS

Potential Legacy trees are defined as the largest tree within your search area, which is as far as you can see, but within maximum limits as follows:

Wadeable Streams: Confine search to no more than 50 m from left and right bank and extending upstream to next transect (for 'K' look upstream 4 channel widths)

Non-wadeable Rivers: Confine search to no more than 100 m from left and right bank and extending both upstream and downstream as far as you can see confidently.

Alien Plants: Confine search to riparian plots on left and right bank

Wadeable Streams: 10 m x 10 m
Non-wadeable Rivers: 10 m x 20 m

Not all aliens are to be identified in all states. See Field Manual and Plant Identification Guide.

TAXONOMIC CATEGORIES

Acacia/Mesquite
Alder/Birch
Ash
Maple/Boxelder
Oak
Poplar/Cottonwood
Sycamore
Willow
Unknown or Other Deciducous

Cedar/Cypress/Sequoia
Fir (including Douglas fir and hemlock)
Juniper
Pine
Spruce
Unknown or Other Conifer

Unknown or Other Broadleaf Evergreen

Snag (Dead tree of any species)

ALIEN SPECIES

E Wtrmilf	Eurasian water milfoil	Myriophyllum spicatum
Hydrilla	Hydrilla	Hydrilla verticillata
E Wtrchest	European water chestnut	Trapa natans
W Hyacinth	Water Hyacinth	Eichhornia crassipes
Ylw Fltheart	Yellow Floating Heart	Nymphoides peltata
P Lstrife	Purple loosestrife	Lythrum salicaria
G Reed	Giant Reed	Arundo donax
Flwr Rush	Flowering Rush	Butomus umbellatus
Salt Ced	Salt Cedar	Tamarix spp.
MF Rose	Multi-flora rose	Rosa multiflora
Spurge	Leafy Spurge	Euphorbia esula

COMMENTS

Transects D to K continued on other side

RIPARIAN "LEGACY" TREES AND INVASIVE ALIEN PLANTS

Reviewed by (initial): _____

SITE ID: FW08 DATE: ___/___/20__

41234

Tran	LARGEST POTENTIAL LEGACY TREE VISIBLE FROM THIS STATION						ALIEN PLANT SPECIES PRESENT IN LEFT AND RIGHT RIPARIAN PLOTS, AND INSTREAM FISH COVER PLOT	
	Trees not Visible	DBH (m)	Height (m)	Dist. from wetted margin (m)	Type	Taxonomic Category		Check all that are present
D	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
E	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
F	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
G	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
H	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
I	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
J	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced
K	○	○ 0-0.1 ○ .75-2 ○ .1-.3 ○ >2 ○ .3-.75	○ <5 ○ 5-15 ○ 15-30 ○ >30		○ Deciduous ○ Coniferous ○ Broadleaf Evergreen		○ NONE	○ E Wtrmilf ○ W Hyacinth ○ G Reed ○ MF Rose ○ Hydrilla ○ Ylw Fltheart ○ Flwr Rush ○ Spurge ○ E Wtrchest ○ P Lstrife ○ Salt Ced

CHANNEL CONSTRAINT FORM - WADEABLE/BOATABLE

Reviewed by (initial): _____

SITE ID: FW08 DATE: ____ / ____ / ____

CHANNEL CONSTRAINT

CHANNEL PATTERN (Fill in one)

- ○ **One channel**
- ○ **Anastomosing (complex) channel** - (Relatively long major and minor channels branching and rejoining.)
- ○ **Braided channel** - (Multiple short channels branching and rejoining - mainly one channel broken up by numerous mid-channel bars.)

CHANNEL CONSTRAINT(Fill in one)

- ○ **Channel very constrained in V-shaped valley** (i.e. it is very unlikely to spread out over valley or erode a new channel during flood)
- ○ **Channel is in Broad Valley** but channel movement by erosion during floods is **constrained by Incision** (Flood flows do not commonly spread over valley floor or into multiple channels.)
- ○ **Channel is in Narrow Valley but is not very constrained**, but limited in movement by relatively narrow valley floor (< ~10 x bankfull width)
- ○ **Channel is Unconstrained in Broad Valley** (i.e. during flood it can fill off-channel areas and side channels, spread out over flood plain, or easily cut new channels by erosion)

CONSTRAINING FEATURES (Fill in one)

- ○ **Bedrock** (i.e. channel is a bedrock-dominated gorge)
- ○ **Hillslope** (i.e. channel constrained in narrow V-shaped valley)
- ○ **Terrace** (i.e. channel is constrained by its own incision into river/stream gravel/soil deposits)
- ○ **Human Bank Alterations** (i.e. constrained by rip-rap, landfill, dike, road, etc.)
- ○ **No constraining features**

Percent of channel length with margin in contact with constraining feature: ____ % ---> (0-100%)

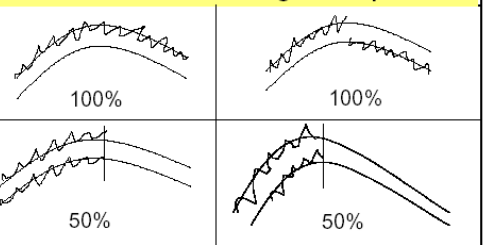
Bankfull width: ____ (m)

Valley width (Visual Estimated Average): ____ (m)

Note: Be sure to include distances between both sides of valley border for valley width.

If you cannot see the valley borders, record the distance you can see and mark this box. ○

Percent of Channel Margin Examples



Comments

15186

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Reviewed by (Initials): _____

TORRENT EVIDENCE ASSESSMENT FORM

SITE ID: FW08 DATE: ___/___/20___

TORRENT EVIDENCE	
Please fill in any of the following that are evident.	
EVIDENCE OF TORRENT SCOURING:	
○	01 - Stream channel has a recently devegetated corridor two or more times the width of the low flow channel. This corridor lacks riparian vegetation with possible exception of fireweed, even-aged alder or cottonwood seedlings, grasses, or other herbaceous plants.
○	02 - Stream substrate cobbles or large gravel particles are NOT IMBRICATED. (Imbricated means that they lie with flat sides horizontal and that they are stacked like roof shingles -- imagine the upstream direction as the top of the "roof." a torrent scour or deposition channel, the stones are laying in unorganized patterns, lying "every which way." In addit many of the substrate particles are angular (not "water-worn.")
○	03 - Channel has little evidence of pool-riffle structure. (For example, could you ride a mountain bike down the channe
○	04 - The stream channel is scoured down to bedrock for substantial portion of reach.
○	05 - There are gravel or cobble berms (little levees) above bankfull level.
○	06 - Downstream of the scoured reach (possibly several miles), there are massive deposits of sediment, logs, and othe debris.
○	07 - Riparian trees have fresh bark scars at many points along the stream at seemingly unbelievable heights above the channel bed.
○	08 - Riparian trees have fallen into the channel as a result of scouring near their roots.
EVIDENCE OF TORRENT DEPOSITS:	
○	09 - There are massive deposits of sediment, logs, and other debris in the reach. They may contain wood and boulder that, in your judgement, could not have been moved by the stream at even extreme flood stage.
○	10 - If the stream has begun to erode newly laid deposits, it is evident that these deposits are "MATRIX SUPPORTED." This means that the large particles, like boulders and cobbles, are often not touching each other, but have silt, sand, a other fine particles between them (their weight is supported by these fine particles -- in contrast to a normal stream deposit, where fines, if present, normally "fill-in" the interstices between coarser particles.)
NO EVIDENCE:	
○	11 - No evidence of torrent scouring or torrent deposits

COMMENTS

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VISUAL ASSESSMENT FORM - WADEABLE/BOATABLE (Front)

Reviewed by (initial): _____

SITE ID: FW08 DATE: ___/___/20___

WATERSHED ACTIVITIES AND DISTURBANCES OBSERVED (Intensity: Blank=Not observed, L=Low, M=Moderate, H=Heavy)

Residential	Recreational	Agricultural	Industrial	Stream Management
L M H Residences	L M H Hiking Trails	L M H Cropland	L M H Industrial Plants	L M H Liming
L M H Maintained Lawns	L M H Parks, Campgrounds	L M H Pasture	L M H Mines/Quarries	L M H Chemical Treatment
L M H Construction	L M H Primitive Parks, Camping	L M H Livestock Use	L M H Oil/Gas Wells	L M H Angling Pressure
L M H Pipes, Drains	L M H Trash/Litter	L M H Orchards	L M H Power Plants	L M H Dredging
L M H Dumping	L M H Surface Films	L M H Poultry	L M H Logging	L M H Channelization
L M H Roads		L M H Irrigation Equip.	L M H Evidence of Fire	L M H Water Level Fluctuations
L M H Bridge/Culverts		L M H Water Withdrawal	L M H Odors	L M H Fish Stocking
L M H Sewage Treatment			L M H Commercial	L M H Dams

SITE CHARACTERISTICS (200 m radius)

Waterbody Character	Pristine	○ 5	○ 4	○ 3	○ 2	○ 1	Highly Disturbed
	Appealing	○ 5	○ 4	○ 3	○ 2	○ 1	Unappealing
Beaver	Beaver Signs:	○ Absent	○ Rare	○ Common			
	Beaver Flow Modifications:	○ None	○ Minor	○ Major			
Dominant Land Use	Dominant Land Use Around 'X'	○ Forest	○ Agriculture	○ Range	○ Urban	○ Suburban/Town	
	If Forest, Dominant Age Class	○ 0 - 25 yrs.	○ 25 - 75 yrs.	○ > 75 yrs.			

WEATHER

GENERAL ASSESSMENT (Biotic integrity, Vegetation diversity, Local anecdotal information)

VISUAL ASSESSMENT FORM - WADEABLE/BOATABLE (Back)

Reviewed by (initial): ______

SITE ID: FW08

DATE: ____/____/2 0____

GENERAL ASSESSMENT (continued)

INVASIVE OR NUISANCE SPECIES OF LOCAL INTEREST

Record species of plants and animals that were observed but are not on the invasive plant form. Examples would be Zebra Mussel or New Zealand Mud Snail, or invasive plants or animals of concern to a particular state. Indicate your level of confidence in your identification, and provide some idea of how prevelant it is in the sampling reach or adjacent riparian area.

Species (Common Name)	Confidence	Prevalence	Comments
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	
	○ LOW ○ HIGH	○ DOMINANT ○ SPARSE ○ COMMON	

TRACKING AND SAMPLE STATUS - WRS

SITE ID: FW08 **Visit #:** ○ 1 ○ 2 Date Collected: ____ / ____ / 2 0 ____

SENT BY: _____ SENDER PHONE: _____

State of Site Location: _____ TEAM: _____ DATE SENT: ____ / ____ / 2 0 ____

SHIPPED BY: ○ FedEx ○ UPS ○ Hand Delivery ○ Other: _____ AIRBILL/TRACKING NUMBER: _____

Site Status Report

SAMPLEABLE	NOT SAMPLEABLE	Temporarily Not Sampleable
○ Wadeable	○ Dry - Visited	○ Not Boatable
○ Boatable	○ Dry - Not Visited	○ Not Wadeable
○ Partial Wadeable	○ Wetland	○ Other
○ Partial Boatable	○ Map Error	NO ACCESS
○ Wadeable Interrupted	○ Impounded	○ Access Denied
○ Boatable Interrupted	○ Other	○ Inaccessible
○ Altered		○ Temp Inaccessible

SAMPLE STATUS

○ **No Samples Collected**

Mark the samples that were collected during this site visit:

○ Water Chem (CHEM)	○ Enterococci (ENTE)
○ Water Chl (WCHL)	○ Sediment (SEDE)
○ Water Chem (PPCP)	○ Fish Tissue (FTIS)
○ Periphyton Chl (PCHL)	○ Fish Voucher (VERT)
○ Periphyton Bio (PBIO)	○ Bent Reachwide (BERW)
○ Periphyton ID (PERI)	○ Bent Low Gradient (BELG)
○ Periphyton APA (PAPA)	

Status Comments

Sample ID	Sample Type	Comments
_____	CHEM	
_____	WCHL	
_____.2	PCHL	
_____.3	PBIO	
_____	CHEM	
_____	WCHL	
_____.2	PCHL	
_____.3	PBIO	

Sample Types	Condition Codes	Chain of Custody	Contact Information
CHEM - Water chemistry WCHL - Water Column Chlorophyll PCHL - Periphyton Chlorophyll PBIO - Periphyton Biomass	Filled in by recipient C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK = Sample OK T = Thawed	Filled in by recipient Date Received: ____/____/ _____ Received by: _____	Tracking Help: Marlys Cappaert PH: 541-754-4467 Lab: Attn: Phil Monaco, Dynamac c/o U.S. EPA 1350 Goodnight Ave Corvallis, OR 97333 PH: 541-754-4787 monaco.phil@epamail.epa.gov

FAX THIS FORM TO 541-754-4637
OR READ TRACKING INFO TO VOICE MESSAGE CENTER:
541-754-4663

52109

04/07/2009 NRSA Tracking - WRS

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TRACKING - NERL Cincinnati

SITE ID: FW08 **Visit #:** ○ 1 ○ 2 Date Collected: ____ / ____ / 2 0 ____

SENT BY: _____ SENDER PHONE: _____

State of Site Location: _____ TEAM: _____ DATE SENT: ____ / ____ / 2 0 ____

SHIPPED BY: ○ FedEx ○ UPS ○ Hand Delivery ○ Other: _____ AIRBILL/TRACKING NUMBER: _____

Sample ID	Sample Type	Comments	Condition Code
	P P C P		

Sample Types	Condition Codes	Chain of Custody	Contact Information
PPCP - Water chemistry	Filled in by recipient C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK = Sample OK T = Thawed	Filled in by recipient Date Received: ____/____/____ Received by: _____	Tracking Help: Marlys Cappaert PH: 541-754-4467 Lab: NERL -Cincinnati Attn: Dr. Angela Batt 26 W. Martin Luther King Drive MS 642 Cincinnati, OH 45268 513-569-7284 batt.angela@epa.gov

FAX THIS FORM TO 541-754-4637
OR READ TRACKING INFO TO VOICE MESSAGE CENTER:
541-754-4663

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TRACKING (BATCHED OR RETAINED SAMPLES) National Rivers and Streams Assessment
Include only all BATCHED or RETAINED samples on one form.

SENT BY: _____ SENDER PHONE: _____ STATE OF SITE LOCATION: _____ TEAM: _____

BATCHED SAMPLES - UNPRESERVED samples that will be batched and shipped within 2 weeks.
SHIPPED BY: ○ FedEx ○ UPS ○ Hand Delivery DATE SHIPPED: ____ / ____ / 2 0 ____
AIRBILL/TRACKING NUMBER: _____

RETAINED SAMPLES - PRESERVED samples that will be stored longer than a month at a holding facility.
○ Held at address: _____

Site ID	Date Sample Collected MM/DD/YYYY	Visit	Sample ID	Sample Type	# of Containers	Comments	Cond. Code
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					
FW08		○ 1 ○ 2					

Lab	Chain of Custody	Sample Types	Condition Codes
○ ACADEMY OF NATURAL SCIENCES - PHIL, PA ○ BENTHIC LAB ○ GLEC ○ MED - DULUTH, MN ○ MICHIGAN STATE UNIV. ○ NERL - N. CHELMSFORD, MA ○ NERL - CINCINNATI, OH ○ OTHER _____	**Filled in by recipient** Date Received: ____/____/____ Received by: _____ **Tracking Help:** Marlys Cappaert p) 541-754-4467	**PRESERVED - RETAINED:** BERW - Benthos Reach Wide BELG - Benthos Low Gradient VERT - Fish Vouchers PERI - Periphyton ID (.1) **UNPRESERVED - BATCHED:** SEDE - Sediment Enzyme FTIS - Fish Tissue PAPA - Periphyton APA (.4) ENTE - Enterococci	**Filled in by recipient** C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK = Sample OK T = Thawed

42504

FAX THIS FORM TO 541-754-4637 OR READ TRACKING INFO TO VOICE MESSAGE CENTER: 541-754-4663

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APPENDIX C

Shipping and Tracking Guidelines

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Tracking Forms

If you have access to a computer, fill out the **electronic tracking forms**

- Be careful to fill out all information accurately and completely
- If you do not have a printer, you will need to include the paper form in the cooler

3 Forms

1 - Tracking and Sample Status – WRS

- This form is filled out for the samples that are shipped immediately after each sampling event (water chemistry, AFDM, and both chlorophyll samples)
- All of these samples will go together in one cooler to the EPA Corvallis lab
- Save form according to the file naming convention on the bottom of form
- Email to address on bottom of form and print form to include in the shipping cooler

**Emailing the electronic WRS form serves as the "status report" for that sampling event*

2 - Tracking (Batched and Retained Samples)

- BATCHED samples are held & shipped within 2 weeks. Send form when SHIPPED.
- RETAINED samples are stored over a month at a holding facility. Send form when COLLECTED **and** when SHIPPED
- Do not combine both BATCHED and RETAINED samples on the same form
- Use one tracking form for each laboratory
- Save form according to the file naming convention on the bottom of form
- Email to address on bottom of form and print form to include in the shipping cooler

3 - Tracking – NERL – Cincinnati

- A subset of urban sites that are 5th order or greater will be sampled for PPCP contaminants.
- Both of the PPCP samples (water and fish tissue) will go to the EPA NERL Cincinnati lab
- Save form according to the file naming convention on the bottom of form
- Email to address on bottom of form and print form to include in the shipping cooler

If you cannot use a computer before shipping:

- Fill out the paper version of the tracking form
- Notify the Information Management Center (contact info on bottom of form) – FAX form or leave voice message with ALL info from the form
- Include the form in the shipping cooler
- Make sure to FAX or leave a voice message BEFORE the form is sealed in the cooler!

Status Report

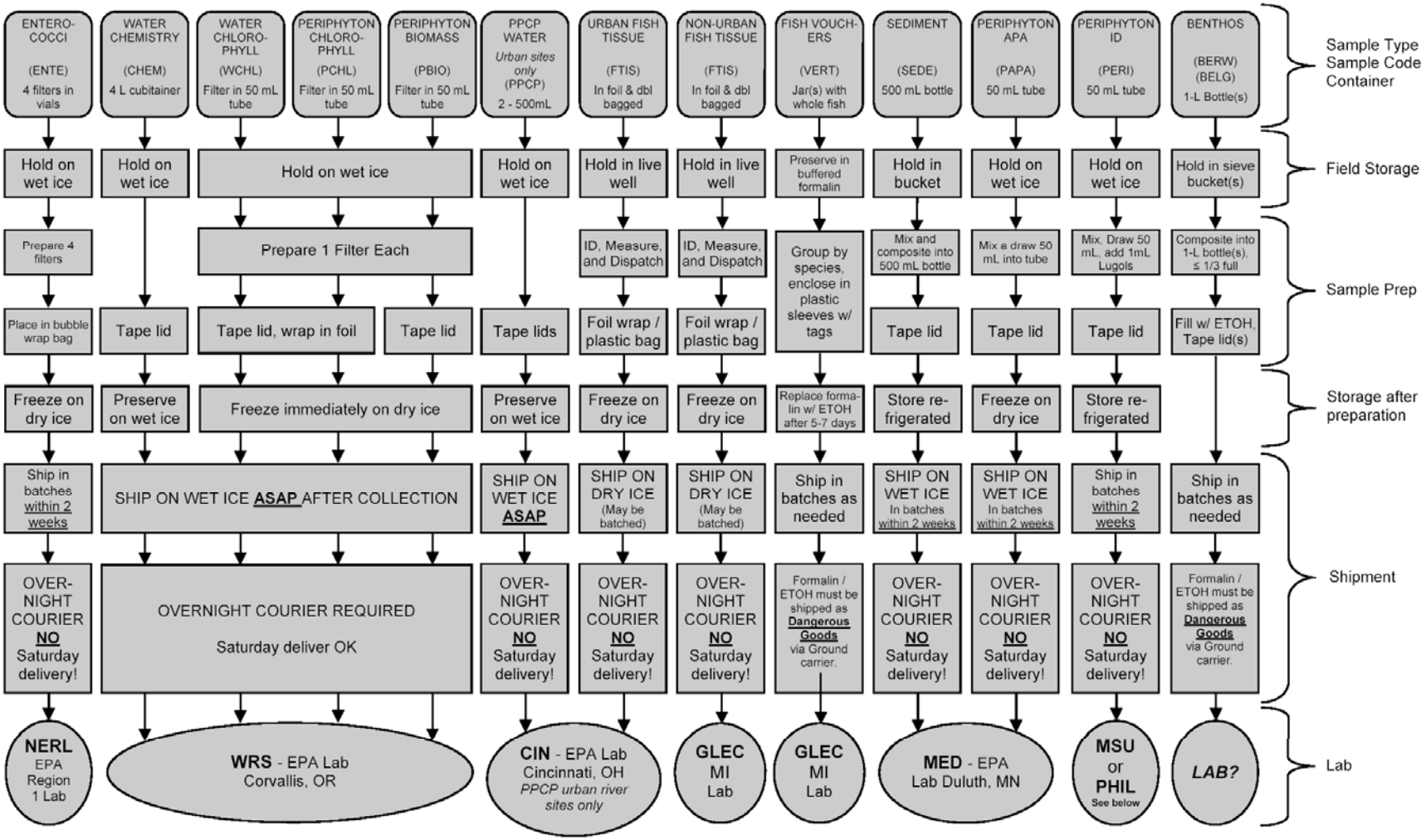
- After each site, the Field Team Leader must file a status report with the Information Management Center and the Field Logistics Coordinator to track visits/samples and to describe activities, problems, and requests
- Emailing the electronic WRS form serves as the status report!

- If the form cannot be emailed, faxing or phoning the information serves as the status report

SHIPPING GUIDELINES

Before shipping, it is very important to preserve each sample as directed in the sample collection portion of this Field Operations Manual.

- Preserve the samples as specified for each indicator before shipping (Fig. C-1).
- Be aware of the holding times for each type of sample (Table C-1):
- Enterococci samples must be filtered and frozen on dry ice within 6 hours of collection
- Fish tissue samples must be frozen on dry ice as soon as possible (hold on wet ice until freezing on dry ice).
- Fish voucher specimens are held on wet ice until being preserved in formalin in the laboratory.
- Water chemistry samples (including PPCP water samples) must be shipped within 24 hours of collection.
- *Chlorophyll a* has a longer holding time, but will be sent with the water chemistry samples since they are going to the same laboratory.
- The remaining samples must be preserved immediately upon collection; they may then be sent in batches to the appropriate laboratory.
- The sediment enzyme sample has a two week holding time.



Periphyton ID Sample (PERI) will be shipped to one of two labs depending on the state from which the sample was obtained.
Find your state below and ship the sample to corresponding lab:

MSU (Michigan State University in East Lansing, MI):	AL, AR, FL, GA, IL, IN, IA, KS, KY, LA, MI, MN, MS, MO, NE, NC, OH, OK, SC, TN, TX, WI
PHIL (Academy of Natural Sciences in Philadelphia, PA)	AZ, CA, CO, CT, DE, ID, MA, MD, ME, MT, ND, NH, NJ, NM, NV, NY, OR, PA, RI, SD, UT, VA, VT, WA, WV, WY

Field Forms: All field forms should be reviewed and sent in to the Information Management Coordinator every 2 weeks

Figure C-1. Sample packaging and shipping summary

SAMPLE	PRESERVATIVE	PACKAGING FOR SHIPMENT	HOLDING TIME
Water Chemistry	Wet ice	Ship in cooler with wet ice	24 hours; ship these samples together (Corvallis lab)
Chlorophyll a	Dry ice in field		
Periphyton – *chlorophyll a*	Dry ice in field		
Periphyton Biomass - AFDM	Dry ice in field		
Sediment enzymes	Wet ice in field; refrigerate to hold	Ship in cooler with wet ice	Batch; ship these samples together every 2 weeks (Duluth lab)[1]
Periphyton - APA	Wet ice in field; hold in freezer		
Periphyton - ID	1 mL Lugol's	Ship in cooler or sturdy container	Batch; ship every 2 weeks
Benthic macroinvertebrates	95% Ethanol		
Fish Vouchers	Formalin		
Fecal Indicator	Dry ice in field; hold in freezer; MUST be filtered & frozen within 6 hours of collection	Ship in cooler with DRY ICE	Batch; ship every 2 weeks (Region 1 lab)
Fish Tissue (non urban sites)	Dry ice in field; hold in freezer	Ship in cooler with DRY ICE	Batch; ship every 2 weeks to GLEC lab
*PPCP Fish Tissue (urban sites)	Dry ice in field; hold in freezer	Ship in cooler with DRY ICE	Batch; ship every 2 weeks to EPA Cincinnati lab
*PPCP Water (urban sites only)	Wet ice	Ship in cooler with wet ice	24 hours; ship to EPA Cincinnati lab

*Urban fish tissue and PPCP water samples are only collected at pre-selected urban 5th order or greater sites
[1]Sediment enzyme samples should not be frozen and must be shipped within two weeks of sampling

When ice is used for shipment (water chemistry, *chlorophyll a*, sediment enzymes, APA, AFDM):

- Ensure that the ice is fresh before shipment; pack the entire cooler full with ice.
- Line the cooler with a large, 30-gallon plastic bag.
- Contain the ice separately within numerous 1-gallon self-sealing plastic bags. Double-bag the ice.
- Use white or clear bags and label with a dark indelible marker. Label all bags of ice as "ICE" to prevent misidentification by couriers of any water leakage as a possible hazardous material spill.
- Place bagged samples and bags of ice inside the cooler liner and seal the liner.
- Secure the cooler with strapping tape.

When dry ice is used for shipping (fish tissue and fecal indicator samples):

- Indicate dry ice on shipping airbill.
- Label cooler with a Class 9 Dangerous Goods label.
- Securely tape the cooler drainage open to prevent pressure build-up in the cooler.
- Secure the cooler with strapping tape
- See "Dry Ice Shipping Protocols" at the end of this Appendix.

WATER CHEMISTRY and CHLOROPHYLL-*a (from water sample and periphyton sample)*

- **Water Chemistry**
  Stored in a 4-L cube container
  - Confirm that the cube container is labeled and covered with clear tape.
  - Place the cube container in a second bag inside the cooler liner.
- ***Chlorophyll* a**
  Two filters each stored in a 50-mL steam-top centrifuge tube wrapped with aluminum foil
  - Confirm that the labels with sample IDs are completed and covered with clear tape.
  - Place the centrifuge tubes in a 1-qt self-sealing plastic bag.
  - Place the bag in a1-gal self-sealing plastic bag and place inside cooler liner with water chemistry sample.

SEDIMENT ENZYMES SAMPLES

Stored in 500 mL jars
- Confirm that the label with sample ID is completed and covered with clear tape.
- Place the 500 mL jar in a 1-gal self-sealing plastic bag and place inside cooler liner.

PERIPHYTON SAMPLES

ID samples preserved with Lugol's solution and sealed at the site.
- Confirm that the label with sample ID is completed and covered with clear tape.
- Verify that the bottle is sealed with electrical tape.
- Place the sealed bottles in a gallon-size self-sealing plastic bag.
- Place the bagged samples in the appropriate shipping container.
- Surround the jars with crumpled newspaper, vermiculite, or other absorbent material.
- Samples can be held and shipped in batches to the laboratory for analysis.

AFDM and APA samples held frozen until shipment
- Confirm that the label with sample ID is completed and covered with clear tape.
- Place the frozen samples in a 1-gal self-sealing plastic bag and place inside cooler liner.

BENTHIC INVERTEBRATE SAMPLES

Preserved in 95% ethanol and sealed at the site.
- Confirm that the label with sample ID is completed and covered with clear tape.
- Check to make sure jars are sealed with electrical tape.
- Place up to twenty 500-mL or ten 1-L jars in each cooler.
- Surround the jars with crumpled newspaper, vermiculite, or other absorbent material.
- Samples can be held and shipped in batches to the laboratory for analysis.

NOTE: These samples must be shipped as "DANGEROUS GOODS" and should be packaged and labeled in accordance with the requirements of the chosen courier.
Alternatively, the ethanol may be decanted from the benthic invertebrate samples so that they may be shipped using standard overnight shipping:

- Allow the samples to sit for at least 1 week to adequately preserve the organisms.
- Immediately before shipping, decant the ethanol from the samples jars, leaving enough liquid to keep the samples moist.
- Make sure to use an overnight delivery so that the lab can immediately restore the ethanol to the sample jars.
- Alert the laboratory so that they are aware they will need to refill the jars immediately upon receipt.

FISH TISSUE SAMPLES

The samples need to be frozen as soon as possible after collection (within 6 hours).

- Pack the cooler with 50 lbs of dry ice.
- Refer to the DRY ICE SHIPPING PROTOCOLS at the end of this Appendix.
- Samples may be stored on dry ice for a maximum of 24 hours. Sampling teams have the option, depending on site logistics, of:
  - shipping the samples packed on dry ice (50 pounds), via priority overnight delivery so that they arrive at the sample preparation laboratory within 24 hours of sample collection, or
  - freezing the samples within 24 hours of collection at ≤-20°C, and storing the frozen samples until shipment within 2 weeks of sample collection (frozen samples will be packed on dry ice and shipped to the sample preparation laboratory via priority overnight delivery service).

FISH VOUCHER SAMPLES

Preserved in a laboratory with formalin

- Confirm that the label with sample ID is completed and covered with clear tape.
- Check to make sure jars are sealed with electrical tape.
- Surround the jars with crumpled newspaper, vermiculite, or other absorbent material.
- Samples can be held and shipped in batches to the laboratory for analysis.

NOTE: These samples must be shipped as "DANGEROUS GOODS" and should be packaged and labeled in accordance with the requirements of the chosen courier.

FECAL INDICATOR SAMPLES

The sample needs to be filtered and frozen as soon as possible after collection (within 6 hours).

- Confirm that the container is labeled and properly sealed.
- Confirm that the bottle is labeled with the appropriate sample ID and covered with clear plastic tape.
- Place the container in the cooler and close.
- Pack the cooler with 10-15 lbs of dry ice (10 lbs if using dry ice blocks or slices, 15 lbs if using dry ice pellets).
- Refer to the DRY ICE SHIPPING PROTOCOLS at the end of this Appendix.
- Samples can be held frozen and shipped in batches to the laboratory for analysis.

DRY ICE SHIPPING PROTOCOLS

1. Indicate dry ice on shipping airbill

   - Fill out Section 1 and Section 3 of the Fed Ex airbill with your Sender and Recipient address and phone number.
   - In Section 4, check "FedEx Priority Overnight."
   - In Section 5, check "Other."
   - In Section 6, under "Does this shipment contain dangerous goods?":
     - Check "Yes/Shipper's Declaration not required."
     - Check "Dry Ice," and fill out "_1_ x _(*amt. of dry ice in kg*)_ kg"
   - In Section 7, fill out weight and declared value of package.

2. Label cooler with a Class 9 Dangerous Goods label (available from FedEx) (Fig. C-2).

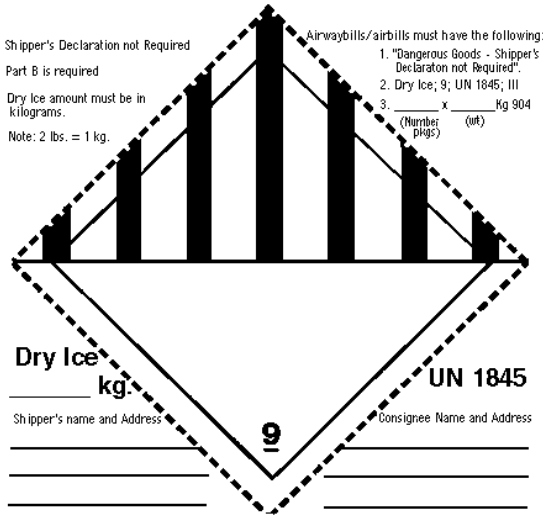


Figure C-2. Class 9 Dangerous Goods label.

- Place the label on the front side of the cooler, not the top of the cooler.
- Fill out #3 in the top right hand corner of the label with the same information as in Section 6 of the FedEx airbill.
- Declare the weight of the dry ice again in the lower left hand corner.
- Fill out the Sender ("Shipper") and Recipient ("Consignee") address on the bottom of the label.

3. Securely tape the cooler drainage open to prevent pressure build-up in the cooler. This is critical to ensure proper venting of the dry ice.
4. Secure the cooler with strapping tape.
5. Place the completed airbill on the top of the cooler.

NOTE: *Not all FedEx locations will accept shipments containing dry ice. Dry ice shipments can be shipped from "FedEx staffed" locations. You can also arrange for a pick-up from your lab or hotel. Dry ice shipments usually cannot be shipped from FedEx Kinko's Office and Print Centers® or FedEx Authorized ShipCenter® locations. These types of locations are differentiated on FedEX.com in the "Find FedEx Locations" feature. Please be sure to call in advance to ensure your location will accept the package for shipment.*

TRACKING FORMS

A Tracking Form must be filled out to accompany each sample shipment. Please refer to Figures 3.2 and 3.3 for examples of Tracking Forms completed for both unpreserved and preserved samples. Be very careful to fill in the information correctly and legibly, especially the airbill number, Site ID, and Sample ID numbers. Use the codes on the bottom of the form to indicate sample type. The Tracking Form is to be placed in a self-sealing plastic bag and included inside the shipping container. Before sealing the container, remember to submit the status report (via email) to sampletracking@epa.gov (see Section 3.2.6); you will need the information on the tracking form to fill out the status report form. For preserved samples, submit a status report both when the samples are brought to the holding facility AND when they are shipped to the appropriate laboratory. For each shipment, you must fill out a scanable tracking form to include in the cooler and submit the electronic status report.

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APPENDIX D

Common and Scientific Names of Fishes of the United States

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Table D-1. Common and Scientific Names of Fishes of the United States

(From: Nelson, J.S., E.J. Crossman, H. Espinosa-Perez, L.T. Findley, C.R. Gilbert, R.N. Lea, and J.D. Williams. 2004. Common and Scientific Names of Fishes from the United States, Canada, and Mexico. American Fisheries Society, Special Publication 29, Bethesda, Maryland.)

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Petromyzontiformes	Petromyzontidae	*Ichthyomyzon bdellium*	Ohio lamprey
Petromyzontiformes	Petromyzontidae	*Ichthyomyzon castaneus*	chestnut lamprey
Petromyzontiformes	Petromyzontidae	*Ichthyomyzon fossor*	northern brook lamprey
Petromyzontiformes	Petromyzontidae	*Ichthyomyzon gagei*	southern brook lamprey
Petromyzontiformes	Petromyzontidae	*Ichthyomyzon greeleyi*	mountain brook lamprey
Petromyzontiformes	Petromyzontidae	*Ichthyomyzon unicuspis*	silver lamprey
Petromyzontiformes	Petromyzontidae	*Lampetra aepyptera*	least brook lamprey
Petromyzontiformes	Petromyzontidae	*Lampetra appendix*	American brook lamprey
Petromyzontiformes	Petromyzontidae	*Lampetra ayresii*	river lamprey
Petromyzontiformes	Petromyzontidae	*Lampetra camtschatica*	Arctic lamprey
Petromyzontiformes	Petromyzontidae	*Lampetra hubbsi*	Kern brook lamprey
Petromyzontiformes	Petromyzontidae	*Lampetra lethophaga*	Pit-Klamath brook lamprey
Petromyzontiformes	Petromyzontidae	*Lampetra minima*	Miller Lake lamprey
Petromyzontiformes	Petromyzontidae	*Lampetra richardsoni*	western brook lamprey
Petromyzontiformes	Petromyzontidae	*Lampetra similis*	Klamath lamprey
Petromyzontiformes	Petromyzontidae	*Lampetra tridentata*	Pacific lamprey
Petromyzontiformes	Petromyzontidae	*Petromyzon marinus*	sea lamprey
Carcharhiniformes	Carcharhinidae	*Carcharhinus leucas*	bull shark
Pristiformes	Pristidae	*Pristis pectinata*	smalltooth sawfish
Myliobatiformes	Dasyatidae	*Dasyatis sabina*	Atlantic stingray
Acipenseriformes	Acipenseridae	*Acipenser brevirostrum*	shortnose sturgeon
Acipenseriformes	Acipenseridae	*Acipenser fulvescens*	lake sturgeon
Acipenseriformes	Acipenseridae	*Acipenser medirostris*	green sturgeon
Acipenseriformes	Acipenseridae	*Acipenser oxyrinchus*	Atlantic sturgeon
Acipenseriformes	Acipenseridae	*Acipenser transmontanus*	white sturgeon
Acipenseriformes	Acipenseridae	*Scaphirhynchus albus*	pallid sturgeon
Acipenseriformes	Acipenseridae	*Scaphirhynchus platorynchus*	shovelnose sturgeon
Acipenseriformes	Acipenseridae	*Scaphirhynchus suttkusi*	Alabama sturgeon
Acipenseriformes	Polyodontidae	*Polyodon spathula*	paddlefish
Lepisosteiformes	Lepisosteidae	*Atractosteus spatula*	alligator gar
Lepisosteiformes	Lepisosteidae	*Lepisosteus oculatus*	spotted gar
Lepisosteiformes	Lepisosteidae	*Lepisosteus osseus*	longnose gar
Lepisosteiformes	Lepisosteidae	*Lepisosteus platostomus*	shortnose gar
Lepisosteiformes	Lepisosteidae	*Lepisosteus platyrhincus*	Florida gar
Amiiformes	Amiidae	*Amia calva*	bowfin
Hiodontiformes	Hiodontidae	*Hiodon alosoides*	goldeye
Hiodontiformes	Hiodontidae	*Hiodon tergisus*	mooneye
Osteoglossiformes	Notopteridae	*Chitala ornata*	clown knifefish
Elopiformes	Elopidae	*Elops affinis*	machete
Elopiformes	Elopidae	*Elops saurus*	ladyfish
Elopiformes	Megalopidae	*Megalops atlanticus*	tarpon
Anguilliformes	Anguillidae	*Anguilla rostrata*	American eel
Clupeiformes	Engraulidae	*Anchoa mitchilli*	bay anchovy

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Clupeiformes	Clupeidae	*Alosa aestivalis*	blueback herring
Clupeiformes	Clupeidae	*Alosa alabamae*	Alabama shad
Clupeiformes	Clupeidae	*Alosa chrysochloris*	skipjack herring
Clupeiformes	Clupeidae	*Alosa mediocris*	hickory shad
Clupeiformes	Clupeidae	*Alosa pseudoharengus*	alewife
Clupeiformes	Clupeidae	*Alosa sapidissima*	American shad
Clupeiformes	Clupeidae	*Dorosoma cepedianum*	gizzard shad
Clupeiformes	Clupeidae	*Dorosoma petenense*	threadfin shad
Clupeiformes	Clupeidae	*Harengula jaguana*	scaled sardine
Clupeiformes	Clupeidae	*Opisthonema oglinum*	Atlantic thread herring
Cypriniformes	Cyprinidae	*Acrocheilus alutaceus*	chiselmouth
Cypriniformes	Cyprinidae	*Agosia chrysogaster*	longfin dace
Cypriniformes	Cyprinidae	*Campostoma anomalum*	central stoneroller
Cypriniformes	Cyprinidae	*Campostoma oligolepis*	largescale stoneroller
Cypriniformes	Cyprinidae	*Campostoma ornatum*	Mexican stoneroller
Cypriniformes	Cyprinidae	*Campostoma pauciradii*	bluefin stoneroller
Cypriniformes	Cyprinidae	*Carassius auratus*	goldfish
Cypriniformes	Cyprinidae	*Clinostomus elongatus*	redside dace
Cypriniformes	Cyprinidae	*Clinostomus funduloides*	rosyside dace
Cypriniformes	Cyprinidae	*Couesius plumbeus*	lake chub
Cypriniformes	Cyprinidae	*Ctenopharyngodon idella*	grass carp
Cypriniformes	Cyprinidae	*Cyprinella analostana*	satinfin shiner
Cypriniformes	Cyprinidae	*Cyprinella caerulea*	blue shiner
Cypriniformes	Cyprinidae	*Cyprinella callisema*	Ocmulgee shiner
Cypriniformes	Cyprinidae	*Cyprinella callistia*	Alabama shiner
Cypriniformes	Cyprinidae	*Cyprinella callitaenia*	bluestripe shiner
Cypriniformes	Cyprinidae	*Cyprinella camura*	bluntface shiner
Cypriniformes	Cyprinidae	*Cyprinella chloristia*	greenfin shiner
Cypriniformes	Cyprinidae	*Cyprinella formosa*	beautiful shiner
Cypriniformes	Cyprinidae	*Cyprinella galactura*	whitetail shiner
Cypriniformes	Cyprinidae	*Cyprinella gibbsi*	Tallapoosa shiner
Cypriniformes	Cyprinidae	*Cyprinella labrosa*	thicklip chub
Cypriniformes	Cyprinidae	*Cyprinella leedsi*	bannerfin shiner
Cypriniformes	Cyprinidae	*Cyprinella lepida*	plateau shiner
Cypriniformes	Cyprinidae	*Cyprinella lutrensis*	red shiner
Cypriniformes	Cyprinidae	*Cyprinella nivea*	whitefin shiner
Cypriniformes	Cyprinidae	*Cyprinella proserpina*	proserpine shiner
Cypriniformes	Cyprinidae	*Cyprinella pyrrhomelas*	fieryblack shiner
Cypriniformes	Cyprinidae	*Cyprinella spiloptera*	spotfin shiner
Cypriniformes	Cyprinidae	*Cyprinella trichroistia*	tricolor shiner
Cypriniformes	Cyprinidae	*Cyprinella venusta*	blacktail shiner
Cypriniformes	Cyprinidae	*Cyprinella whipplei*	steelcolor shiner
Cypriniformes	Cyprinidae	*Cyprinella xaenura*	Altamaha shiner
Cypriniformes	Cyprinidae	*Cyprinella zanema*	Santee chub
Cypriniformes	Cyprinidae	*Cyprinus carpio*	common carp
Cypriniformes	Cyprinidae	*Dionda argentosa*	Manantial roundnose minnow
Cypriniformes	Cyprinidae	*Dionda diaboli*	Devils River minnow

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Cypriniformes	Cyprinidae	*Dionda episcopa*	roundnose minnow
Cypriniformes	Cyprinidae	*Dionda nigrotaeniata*	Guadalupe roundnose minnow
Cypriniformes	Cyprinidae	*Dionda serena*	Nueces roundnose minnow
Cypriniformes	Cyprinidae	*Eremichthys acros*	desert dace
Cypriniformes	Cyprinidae	*Erimonax monachus*	spotfin chub
Cypriniformes	Cyprinidae	*Erimystax cahni*	slender chub
Cypriniformes	Cyprinidae	*Erimystax dissimilis*	streamline chub
Cypriniformes	Cyprinidae	*Erimystax harryi*	Ozark chub
Cypriniformes	Cyprinidae	*Erimystax insignis*	blotched chub
Cypriniformes	Cyprinidae	*Erimystax x-punctatus*	gravel chub
Cypriniformes	Cyprinidae	*Exoglossum laurae*	tonguetied minnow
Cypriniformes	Cyprinidae	*Exoglossum maxillingua*	cutlip minnow
Cypriniformes	Cyprinidae	*Gila alvordensis*	Alvord chub
Cypriniformes	Cyprinidae	*Gila atraria*	Utah chub
Cypriniformes	Cyprinidae	*Gila bicolor*	tui chub
Cypriniformes	Cyprinidae	*Gila boraxobius*	Borax Lake chub
Cypriniformes	Cyprinidae	*Gila coerulea*	blue chub
Cypriniformes	Cyprinidae	*Gila crassicauda*	thicktail chub
Cypriniformes	Cyprinidae	*Gila cypha*	humpback chub
Cypriniformes	Cyprinidae	*Gila ditaenia*	Sonora chub
Cypriniformes	Cyprinidae	*Gila elegans*	bonytail
Cypriniformes	Cyprinidae	*Gila intermedia*	Gila chub
Cypriniformes	Cyprinidae	*Gila nigra*	headwater chub
Cypriniformes	Cyprinidae	*Gila nigrescens*	Chihuahua chub
Cypriniformes	Cyprinidae	*Gila orcuttii*	arroyo chub
Cypriniformes	Cyprinidae	*Gila pandora*	Rio Grande chub
Cypriniformes	Cyprinidae	*Gila purpurea*	Yaqui chub
Cypriniformes	Cyprinidae	*Gila robusta*	roundtail chub
Cypriniformes	Cyprinidae	*Gila seminuda*	Virgin chub
Cypriniformes	Cyprinidae	*Hemitremia flammea*	flame chub
Cypriniformes	Cyprinidae	*Hesperoleucus symmetricus*	California roach
Cypriniformes	Cyprinidae	*Hybognathus amarus*	Rio Grande silvery minnow
Cypriniformes	Cyprinidae	*Hybognathus argyritis*	western silvery minnow
Cypriniformes	Cyprinidae	*Hybognathus hankinsoni*	brassy minnow
Cypriniformes	Cyprinidae	*Hybognathus hayi*	cypress minnow
Cypriniformes	Cyprinidae	*Hybognathus nuchalis*	Mississippi silvery minnow
Cypriniformes	Cyprinidae	*Hybognathus placitus*	plains minnow
Cypriniformes	Cyprinidae	*Hybognathus regius*	eastern silvery minnow
Cypriniformes	Cyprinidae	*Hybopsis amblops*	bigeye chub
Cypriniformes	Cyprinidae	*Hybopsis amnis*	pallid shiner
Cypriniformes	Cyprinidae	*Hybopsis hypsinotus*	highback chub
Cypriniformes	Cyprinidae	*Hybopsis lineapunctata*	lined chub
Cypriniformes	Cyprinidae	*Hybopsis rubrifrons*	rosyface chub
Cypriniformes	Cyprinidae	*Hybopsis winchelli*	clear chub
Cypriniformes	Cyprinidae	*Hypophthalmichthys molitrix*	silver carp
Cypriniformes	Cyprinidae	*Hypophthalmichthys nobilis*	bighead carp
Cypriniformes	Cyprinidae	*Iotichthys phlegethontis*	least chub

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Cypriniformes	Cyprinidae	*Lavinia exilicauda*	hitch
Cypriniformes	Cyprinidae	*Lepidomeda albivallis*	White River spinedace
Cypriniformes	Cyprinidae	*Lepidomeda altivelis*	Pahranagat spinedace
Cypriniformes	Cyprinidae	*Lepidomeda mollispinis*	Virgin spinedace
Cypriniformes	Cyprinidae	*Lepidomeda vittata*	Little Colorado spinedace
Cypriniformes	Cyprinidae	*Leuciscus idus*	ide
Cypriniformes	Cyprinidae	*Luxilus albeolus*	white shiner
Cypriniformes	Cyprinidae	*Luxilus cardinalis*	cardinal shiner
Cypriniformes	Cyprinidae	*Luxilus cerasinus*	crescent shiner
Cypriniformes	Cyprinidae	*Luxilus chrysocephalus*	striped shiner
Cypriniformes	Cyprinidae	*Luxilus coccogenis*	warpaint shiner
Cypriniformes	Cyprinidae	*Luxilus cornutus*	common shiner
Cypriniformes	Cyprinidae	*Luxilus pilsbryi*	duskystripe shiner
Cypriniformes	Cyprinidae	*Luxilus zonatus*	bleeding shiner
Cypriniformes	Cyprinidae	*Luxilus zonistius*	bandfin shiner
Cypriniformes	Cyprinidae	*Lythrurus alegnotus*	Warrior shiner
Cypriniformes	Cyprinidae	*Lythrurus ardens*	rosefin shiner
Cypriniformes	Cyprinidae	*Lythrurus atrapiculus*	blacktip shiner
Cypriniformes	Cyprinidae	*Lythrurus bellus*	pretty shiner
Cypriniformes	Cyprinidae	*Lythrurus fasciolaris*	scarlet shiner
Cypriniformes	Cyprinidae	*Lythrurus fumeus*	ribbon shiner
Cypriniformes	Cyprinidae	*Lythrurus lirus*	mountain shiner
Cypriniformes	Cyprinidae	*Lythrurus matutinus*	pinewoods shiner
Cypriniformes	Cyprinidae	*Lythrurus roseipinnis*	cherryfin shiner
Cypriniformes	Cyprinidae	*Lythrurus snelsoni*	Ouachita shiner
Cypriniformes	Cyprinidae	*Lythrurus umbratilis*	redfin shiner
Cypriniformes	Cyprinidae	*Macrhybopsis aestivalis*	speckled chub
Cypriniformes	Cyprinidae	*Macrhybopsis australis*	prairie chub
Cypriniformes	Cyprinidae	*Macrhybopsis gelida*	sturgeon chub
Cypriniformes	Cyprinidae	*Macrhybopsis hyostoma*	shoal chub
Cypriniformes	Cyprinidae	*Macrhybopsis marconis*	burrhead chub
Cypriniformes	Cyprinidae	*Macrhybopsis meeki*	sicklefin chub
Cypriniformes	Cyprinidae	*Macrhybopsis storeriana*	silver chub
Cypriniformes	Cyprinidae	*Macrhybopsis tetranema*	peppered chub
Cypriniformes	Cyprinidae	*Margariscus margarita*	pearl dace
Cypriniformes	Cyprinidae	*Meda fulgida*	spikedace
Cypriniformes	Cyprinidae	*Moapa coriacea*	Moapa dace
Cypriniformes	Cyprinidae	*Mylocheilus caurinus*	peamouth
Cypriniformes	Cyprinidae	*Mylopharodon conocephalus*	hardhead
Cypriniformes	Cyprinidae	*Nocomis asper*	redspot chub
Cypriniformes	Cyprinidae	*Nocomis biguttatus*	hornyhead chub
Cypriniformes	Cyprinidae	*Nocomis effusus*	redtail chub
Cypriniformes	Cyprinidae	*Nocomis leptocephalus*	bluehead chub
Cypriniformes	Cyprinidae	*Nocomis micropogon*	river chub
Cypriniformes	Cyprinidae	*Nocomis platyrhynchus*	bigmouth chub
Cypriniformes	Cyprinidae	*Nocomis raneyi*	bull chub
Cypriniformes	Cyprinidae	*Notemigonus crysoleucas*	golden shiner
Cypriniformes	Cyprinidae	*Notropis albizonatus*	palezone shiner

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Cypriniformes	Cyprinidae	*Notropis alborus*	whitemouth shiner
Cypriniformes	Cyprinidae	*Notropis altipinnis*	highfin shiner
Cypriniformes	Cyprinidae	*Notropis amabilis*	Texas shiner
Cypriniformes	Cyprinidae	*Notropis ammophilus*	orangefin shiner
Cypriniformes	Cyprinidae	*Notropis amoenus*	comely shiner
Cypriniformes	Cyprinidae	*Notropis anogenus*	pugnose shiner
Cypriniformes	Cyprinidae	*Notropis ariommus*	popeye shiner
Cypriniformes	Cyprinidae	*Notropis asperifrons*	burrhead shiner
Cypriniformes	Cyprinidae	*Notropis atherinoides*	emerald shiner
Cypriniformes	Cyprinidae	*Notropis atrocaudalis*	blackspot shiner
Cypriniformes	Cyprinidae	*Notropis baileyi*	rough shiner
Cypriniformes	Cyprinidae	*Notropis bairdi*	Red River shiner
Cypriniformes	Cyprinidae	*Notropis bifrenatus*	bridle shiner
Cypriniformes	Cyprinidae	*Notropis blennius*	river shiner
Cypriniformes	Cyprinidae	*Notropis boops*	bigeye shiner
Cypriniformes	Cyprinidae	*Notropis braytoni*	Tamaulipas shiner
Cypriniformes	Cyprinidae	*Notropis buccatus*	silverjaw minnow
Cypriniformes	Cyprinidae	*Notropis buccula*	smalleye shiner
Cypriniformes	Cyprinidae	*Notropis buchanani*	ghost shiner
Cypriniformes	Cyprinidae	*Notropis cahabae*	Cahaba shiner
Cypriniformes	Cyprinidae	*Notropis candidus*	silverside shiner
Cypriniformes	Cyprinidae	*Notropis chalybaeus*	ironcolor shiner
Cypriniformes	Cyprinidae	*Notropis chihuahua*	Chihuahua shiner
Cypriniformes	Cyprinidae	*Notropis chiliticus*	redlip shiner
Cypriniformes	Cyprinidae	*Notropis chlorocephalus*	greenhead shiner
Cypriniformes	Cyprinidae	*Notropis chrosomus*	rainbow shiner
Cypriniformes	Cyprinidae	*Notropis cummingsae*	dusky shiner
Cypriniformes	Cyprinidae	*Notropis dorsalis*	bigmouth shiner
Cypriniformes	Cyprinidae	*Notropis edwardraneyi*	fluvial shiner
Cypriniformes	Cyprinidae	*Notropis girardi*	Arkansas River shiner
Cypriniformes	Cyprinidae	*Notropis greenei*	wedgespot shiner
Cypriniformes	Cyprinidae	*Notropis harperi*	redeye chub
Cypriniformes	Cyprinidae	*Notropis heterodon*	blackchin shiner
Cypriniformes	Cyprinidae	*Notropis heterolepis*	blacknose shiner
Cypriniformes	Cyprinidae	*Notropis hudsonius*	spottail shiner
Cypriniformes	Cyprinidae	*Notropis hypsilepis*	highscale shiner
Cypriniformes	Cyprinidae	*Notropis jemezanus*	Rio Grande shiner
Cypriniformes	Cyprinidae	*Notropis leuciodus*	Tennessee shiner
Cypriniformes	Cyprinidae	*Notropis longirostris*	longnose shiner
Cypriniformes	Cyprinidae	*Notropis lutipinnis*	yellowfin shiner
Cypriniformes	Cyprinidae	*Notropis maculatus*	taillight shiner
Cypriniformes	Cyprinidae	*Notropis mekistocholas*	Cape Fear shiner
Cypriniformes	Cyprinidae	*Notropis melanostomus*	blackmouth shiner
Cypriniformes	Cyprinidae	*Notropis micropteryx*	highland shiner
Cypriniformes	Cyprinidae	*Notropis nubilus*	Ozark minnow
Cypriniformes	Cyprinidae	*Notropis orca*	phantom shiner
Cypriniformes	Cyprinidae	*Notropis ortenburgeri*	Kiamichi shiner
Cypriniformes	Cyprinidae	*Notropis oxyrhynchus*	sharpnose shiner

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Cypriniformes	Cyprinidae	*Notropis ozarcanus*	Ozark shiner
Cypriniformes	Cyprinidae	*Notropis percobromus*	carmine shiner
Cypriniformes	Cyprinidae	*Notropis perpallidus*	peppered shiner
Cypriniformes	Cyprinidae	*Notropis petersoni*	coastal shiner
Cypriniformes	Cyprinidae	*Notropis photogenis*	silver shiner
Cypriniformes	Cyprinidae	*Notropis potteri*	chub shiner
Cypriniformes	Cyprinidae	*Notropis procne*	swallowtail shiner
Cypriniformes	Cyprinidae	*Notropis rafinesquei*	Yazoo shiner
Cypriniformes	Cyprinidae	*Notropis rubellus*	rosyface shiner
Cypriniformes	Cyprinidae	*Notropis rubricroceus*	saffron shiner
Cypriniformes	Cyprinidae	*Notropis rupestris*	bedrock shiner
Cypriniformes	Cyprinidae	*Notropis sabinae*	Sabine shiner
Cypriniformes	Cyprinidae	*Notropis scabriceps*	New River shiner
Cypriniformes	Cyprinidae	*Notropis scepticus*	sandbar shiner
Cypriniformes	Cyprinidae	*Notropis semperasper*	roughhead shiner
Cypriniformes	Cyprinidae	*Notropis shumardi*	silverband shiner
Cypriniformes	Cyprinidae	*Notropis simus*	bluntnose shiner
Cypriniformes	Cyprinidae	*Notropis spectrunculus*	mirror shiner
Cypriniformes	Cyprinidae	*Notropis stilbius*	silverstripe shiner
Cypriniformes	Cyprinidae	*Notropis stramineus*	sand shiner
Cypriniformes	Cyprinidae	*Notropis suttkusi*	rocky shiner
Cypriniformes	Cyprinidae	*Notropis telescopus*	telescope shiner
Cypriniformes	Cyprinidae	*Notropis texanus*	weed shiner
Cypriniformes	Cyprinidae	*Notropis topeka*	Topeka shiner
Cypriniformes	Cyprinidae	*Notropis uranoscopus*	skygazer shiner
Cypriniformes	Cyprinidae	*Notropis volucellus*	mimic shiner
Cypriniformes	Cyprinidae	*Notropis wickliffi*	channel shiner
Cypriniformes	Cyprinidae	*Notropis xaenocephalus*	Coosa shiner
Cypriniformes	Cyprinidae	*Opsopoeodus emiliae*	pugnose minnow
Cypriniformes	Cyprinidae	*Oregonichthys crameri*	Oregon chub
Cypriniformes	Cyprinidae	*Oregonichthys kalawatseti*	Umpqua chub
Cypriniformes	Cyprinidae	*Orthodon microlepidotus*	Sacramento blackfish
Cypriniformes	Cyprinidae	*Phenacobius catostomus*	riffle minnow
Cypriniformes	Cyprinidae	*Phenacobius crassilabrum*	fatlips minnow
Cypriniformes	Cyprinidae	*Phenacobius mirabilis*	suckermouth minnow
Cypriniformes	Cyprinidae	*Phenacobius teretulus*	Kanawha minnow
Cypriniformes	Cyprinidae	*Phenacobius uranops*	stargazing minnow
Cypriniformes	Cyprinidae	*Phoxinus cumberlandensis*	blackside dace
Cypriniformes	Cyprinidae	*Phoxinus eos*	northern redbelly dace
Cypriniformes	Cyprinidae	*Phoxinus erythrogaster*	southern redbelly dace
Cypriniformes	Cyprinidae	*Phoxinus neogaeus*	finescale dace
Cypriniformes	Cyprinidae	*Phoxinus oreas*	mountain redbelly dace
Cypriniformes	Cyprinidae	*Phoxinus saylori*	laurel dace
Cypriniformes	Cyprinidae	*Phoxinus tennesseensis*	Tennessee dace
Cypriniformes	Cyprinidae	*Pimephales notatus*	bluntnose minnow
Cypriniformes	Cyprinidae	*Pimephales promelas*	fathead minnow
Cypriniformes	Cyprinidae	*Pimephales tenellus*	slim minnow
Cypriniformes	Cyprinidae	*Pimephales vigilax*	bullhead minnow

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Cypriniformes	Cyprinidae	*Plagopterus argentissimus*	woundfin
Cypriniformes	Cyprinidae	*Platygobio gracilis*	flathead chub
Cypriniformes	Cyprinidae	*Pogonichthys ciscoides*	Clear Lake splittail
Cypriniformes	Cyprinidae	*Pogonichthys macrolepidotus*	splittail
Cypriniformes	Cyprinidae	*Pteronotropis euryzonus*	broadstripe shiner
Cypriniformes	Cyprinidae	*Pteronotropis grandipinnis*	Apalachee shiner
Cypriniformes	Cyprinidae	*Pteronotropis hubbsi*	bluehead shiner
Cypriniformes	Cyprinidae	*Pteronotropis hypselopterus*	sailfin shiner
Cypriniformes	Cyprinidae	*Pteronotropis merlini*	orangetail shiner
Cypriniformes	Cyprinidae	*Pteronotropis signipinnis*	flagfin shiner
Cypriniformes	Cyprinidae	*Pteronotropis welaka*	bluenose shiner
Cypriniformes	Cyprinidae	*Ptychocheilus grandis*	Sacramento pikeminnow
Cypriniformes	Cyprinidae	*Ptychocheilus lucius*	Colorado pikeminnow
Cypriniformes	Cyprinidae	*Ptychocheilus oregonensis*	northern pikeminnow
Cypriniformes	Cyprinidae	*Ptychocheilus umpquae*	Umpqua pikeminnow
Cypriniformes	Cyprinidae	*Relictus solitarius*	relict dace
Cypriniformes	Cyprinidae	*Rhinichthys atratulus*	eastern blacknose dace
Cypriniformes	Cyprinidae	*Rhinichthys cataractae*	longnose dace
Cypriniformes	Cyprinidae	*Rhinichthys cobitis*	loach minnow
Cypriniformes	Cyprinidae	*Rhinichthys deaconi*	Las Vegas dace
Cypriniformes	Cyprinidae	*Rhinichthys evermanni*	Umpqua dace
Cypriniformes	Cyprinidae	*Rhinichthys falcatus*	leopard dace
Cypriniformes	Cyprinidae	*Rhinichthys obtusus*	western blacknose dace
Cypriniformes	Cyprinidae	*Rhinichthys osculus*	speckled dace
Cypriniformes	Cyprinidae	*Rhinichthys umatilla*	Umatilla dace
Cypriniformes	Cyprinidae	*Rhodeus sericeus*	bitterling
Cypriniformes	Cyprinidae	*Richardsonius balteatus*	redside shiner
Cypriniformes	Cyprinidae	*Richardsonius egregius*	Lahontan redside
Cypriniformes	Cyprinidae	*Scardinius erythrophthalmus*	rudd
Cypriniformes	Cyprinidae	*Semotilus atromaculatus*	creek chub
Cypriniformes	Cyprinidae	*Semotilus corporalis*	fallfish
Cypriniformes	Cyprinidae	*Semotilus lumbee*	sandhills chub
Cypriniformes	Cyprinidae	*Semotilus thoreauianus*	Dixie chub
Cypriniformes	Cyprinidae	*Snyderichthys copei*	leatherside chub
Cypriniformes	Cyprinidae	*Tinca tinca*	tench
Cypriniformes	Catostomidae	*Carpiodes carpio*	river carpsucker
Cypriniformes	Catostomidae	*Carpiodes cyprinus*	quillback
Cypriniformes	Catostomidae	*Carpiodes velifer*	highfin carpsucker
Cypriniformes	Catostomidae	*Catostomus ardens*	Utah sucker
Cypriniformes	Catostomidae	*Catostomus bernardini*	Yaqui sucker
Cypriniformes	Catostomidae	*Catostomus catostomus*	longnose sucker
Cypriniformes	Catostomidae	*Catostomus clarkii*	desert sucker
Cypriniformes	Catostomidae	*Catostomus columbianus*	bridgelip sucker
Cypriniformes	Catostomidae	*Catostomus commersonii*	white sucker
Cypriniformes	Catostomidae	*Catostomus discobolus*	bluehead sucker
Cypriniformes	Catostomidae	*Catostomus fumeiventris*	Owens sucker
Cypriniformes	Catostomidae	*Catostomus insignis*	Sonora sucker
Cypriniformes	Catostomidae	*Catostomus latipinnis*	flannelmouth sucker

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Cypriniformes	Catostomidae	*Catostomus macrocheilus*	largescale sucker
Cypriniformes	Catostomidae	*Catostomus microps*	Modoc sucker
Cypriniformes	Catostomidae	*Catostomus occidentalis*	Sacramento sucker
Cypriniformes	Catostomidae	*Catostomus platyrhynchus*	mountain sucker
Cypriniformes	Catostomidae	*Catostomus plebeius*	Rio Grande sucker
Cypriniformes	Catostomidae	*Catostomus rimiculus*	Klamath smallscale sucker
Cypriniformes	Catostomidae	*Catostomus santaanae*	Santa Ana sucker
Cypriniformes	Catostomidae	*Catostomus snyderi*	Klamath largescale sucker
Cypriniformes	Catostomidae	*Catostomus tahoensis*	Tahoe sucker
Cypriniformes	Catostomidae	*Catostomus warnerensis*	Warner sucker
Cypriniformes	Catostomidae	*Chasmistes brevirostris*	shortnose sucker
Cypriniformes	Catostomidae	*Chasmistes cujus*	cui-ui
Cypriniformes	Catostomidae	*Chasmistes liorus*	June sucker
Cypriniformes	Catostomidae	*Chasmistes muriei*	Snake River sucker
Cypriniformes	Catostomidae	*Cycleptus elongatus*	blue sucker
Cypriniformes	Catostomidae	*Cycleptus meridionalis*	southeastern blue sucker
Cypriniformes	Catostomidae	*Deltistes luxatus*	Lost River sucker
Cypriniformes	Catostomidae	*Erimyzon oblongus*	creek chubsucker
Cypriniformes	Catostomidae	*Erimyzon sucetta*	lake chubsucker
Cypriniformes	Catostomidae	*Erimyzon tenuis*	sharpfin chubsucker
Cypriniformes	Catostomidae	*Hypentelium etowanum*	Alabama hog sucker
Cypriniformes	Catostomidae	*Hypentelium nigricans*	northern hog sucker
Cypriniformes	Catostomidae	*Hypentelium roanokense*	Roanoke hog sucker
Cypriniformes	Catostomidae	*Ictiobus bubalus*	smallmouth buffalo
Cypriniformes	Catostomidae	*Ictiobus cyprinellus*	bigmouth buffalo
Cypriniformes	Catostomidae	*Ictiobus niger*	black buffalo
Cypriniformes	Catostomidae	*Minytrema melanops*	spotted sucker
Cypriniformes	Catostomidae	*Moxostoma anisurum*	silver redhorse
Cypriniformes	Catostomidae	*Moxostoma ariommum*	bigeye jumprock
Cypriniformes	Catostomidae	*Moxostoma austrinum*	Mexican redhorse
Cypriniformes	Catostomidae	*Moxostoma breviceps*	smallmouth redhorse
Cypriniformes	Catostomidae	*Moxostoma carinatum*	river redhorse
Cypriniformes	Catostomidae	*Moxostoma cervinum*	blacktip jumprock
Cypriniformes	Catostomidae	*Moxostoma collapsum*	notchlip redhorse
Cypriniformes	Catostomidae	*Moxostoma congestum*	gray redhorse
Cypriniformes	Catostomidae	*Moxostoma duquesnei*	black redhorse
Cypriniformes	Catostomidae	*Moxostoma erythrurum*	golden redhorse
Cypriniformes	Catostomidae	*Moxostoma lacerum*	harelip sucker
Cypriniformes	Catostomidae	*Moxostoma lachneri*	greater jumprock
Cypriniformes	Catostomidae	*Moxostoma macrolepidotum*	shorthead redhorse
Cypriniformes	Catostomidae	*Moxostoma pappillosum*	V-lip redhorse
Cypriniformes	Catostomidae	*Moxostoma pisolabrum*	pealip redhorse
Cypriniformes	Catostomidae	*Moxostoma poecilurum*	blacktail redhorse
Cypriniformes	Catostomidae	*Moxostoma robustum*	robust redhorse
Cypriniformes	Catostomidae	*Moxostoma rupiscartes*	striped jumprock
Cypriniformes	Catostomidae	*Moxostoma valenciennesi*	greater redhorse
Cypriniformes	Catostomidae	*Thoburnia atripinnis*	blackfin sucker
Cypriniformes	Catostomidae	*Thoburnia hamiltoni*	rustyside sucker

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Cypriniformes	Catostomidae	*Thoburnia rhothoeca*	torrent sucker
Cypriniformes	Catostomidae	*Xyrauchen texanus*	razorback sucker
Cypriniformes	Cobitidae	*Misgurnus anguillicaudatus*	oriental weatherfish
Characiformes	Characidae	*Astyanax mexicanus*	Mexican tetra
Siluriformes	Ictaluridae	*Ameiurus brunneus*	snail bullhead
Siluriformes	Ictaluridae	*Ameiurus catus*	white catfish
Siluriformes	Ictaluridae	*Ameiurus melas*	black bullhead
Siluriformes	Ictaluridae	*Ameiurus natalis*	yellow bullhead
Siluriformes	Ictaluridae	*Ameiurus nebulosus*	brown bullhead
Siluriformes	Ictaluridae	*Ameiurus platycephalus*	flat bullhead
Siluriformes	Ictaluridae	*Ameiurus serracanthus*	spotted bullhead
Siluriformes	Ictaluridae	*Ictalurus furcatus*	blue catfish
Siluriformes	Ictaluridae	*Ictalurus lupus*	headwater catfish
Siluriformes	Ictaluridae	*Ictalurus pricei*	Yaqui catfish
Siluriformes	Ictaluridae	*Ictalurus punctatus*	channel catfish
Siluriformes	Ictaluridae	*Noturus albater*	Ozark madtom
Siluriformes	Ictaluridae	*Noturus baileyi*	smoky madtom
Siluriformes	Ictaluridae	*Noturus elegans*	elegant madtom
Siluriformes	Ictaluridae	*Noturus eleutherus*	mountain madtom
Siluriformes	Ictaluridae	*Noturus exilis*	slender madtom
Siluriformes	Ictaluridae	*Noturus flavater*	checkered madtom
Siluriformes	Ictaluridae	*Noturus flavipinnis*	yellowfin madtom
Siluriformes	Ictaluridae	*Noturus flavus*	stonecat
Siluriformes	Ictaluridae	*Noturus funebris*	black madtom
Siluriformes	Ictaluridae	*Noturus furiosus*	Carolina madtom
Siluriformes	Ictaluridae	*Noturus gilberti*	orangefin madtom
Siluriformes	Ictaluridae	*Noturus gyrinus*	tadpole madtom
Siluriformes	Ictaluridae	*Noturus hildebrandi*	least madtom
Siluriformes	Ictaluridae	*Noturus insignis*	margined madtom
Siluriformes	Ictaluridae	*Noturus lachneri*	Ouachita madtom
Siluriformes	Ictaluridae	*Noturus leptacanthus*	speckled madtom
Siluriformes	Ictaluridae	*Noturus miurus*	brindled madtom
Siluriformes	Ictaluridae	*Noturus munitus*	frecklebelly madtom
Siluriformes	Ictaluridae	*Noturus nocturnus*	freckled madtom
Siluriformes	Ictaluridae	*Noturus phaeus*	brown madtom
Siluriformes	Ictaluridae	*Noturus placidus*	Neosho madtom
Siluriformes	Ictaluridae	*Noturus stanauli*	pygmy madtom
Siluriformes	Ictaluridae	*Noturus stigmosus*	northern madtom
Siluriformes	Ictaluridae	*Noturus taylori*	Caddo madtom
Siluriformes	Ictaluridae	*Noturus trautmani*	Scioto madtom
Siluriformes	Ictaluridae	*Pylodictis olivaris*	flathead catfish
Siluriformes	Ictaluridae	*Satan eurystomus*	widemouth blindcat
Siluriformes	Ictaluridae	*Trogloglanis pattersoni*	toothless blindcat
Siluriformes	Clariidae	*Clarias batrachus*	walking catfish
Siluriformes	Ariidae	*Ariopsis felis*	hardhead catfish
Siluriformes	Doradidae	*Platydoras armatulus*	southern striped Raphael
Siluriformes	Callichthyidae	*Hoplosternum littorale*	brown hoplo
Siluriformes	Loricariidae	*Hypostomus plecostomus*	suckermouth catfish

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Siluriformes	Loricariidae	*Pterygoplichthys anisitsi*	southern sailfin catfish
Siluriformes	Loricariidae	*Pterygoplichthys disjunctivus*	vermiculated sailfin catfish
Siluriformes	Loricariidae	*Pterygoplichthys multiradiatus*	Orinoco sailfin catfish
Siluriformes	Loricariidae	*Pterygoplichthys pardalis*	Amazon sailfin catfish
Esociformes	Esocidae	*Esox americanus*	redfin pickerel
Esociformes	Esocidae	*Esox lucius*	northern pike
Esociformes	Esocidae	*Esox masquinongy*	muskellunge
Esociformes	Esocidae	*Esox niger*	chain pickerel
Esociformes	Umbridae	*Dallia pectoralis*	Alaska blackfish
Esociformes	Umbridae	*Novumbra hubbsi*	Olympic mudminnow
Esociformes	Umbridae	*Umbra limi*	central mudminnow
Esociformes	Umbridae	*Umbra pygmaea*	eastern mudminnow
Salmoniformes	Osmeridae	*Hypomesus nipponensis*	wakasagi
Salmoniformes	Osmeridae	*Hypomesus olidus*	pond smelt
Salmoniformes	Osmeridae	*Hypomesus pretiosus*	surf smelt
Salmoniformes	Osmeridae	*Hypomesus transpacificus*	delta smelt
Salmoniformes	Osmeridae	*Osmerus mordax*	rainbow smelt
Salmoniformes	Osmeridae	*Spirinchus thaleichthys*	longfin smelt
Salmoniformes	Osmeridae	*Thaleichthys pacificus*	eulachon
Salmoniformes	Salmonidae	*Coregonus artedi*	cisco
Salmoniformes	Salmonidae	*Coregonus autumnalis*	Arctic cisco
Salmoniformes	Salmonidae	*Coregonus clupeaformis*	lake whitefish
Salmoniformes	Salmonidae	*Coregonus hoyi*	bloater
Salmoniformes	Salmonidae	*Coregonus johannae*	deepwater cisco
Salmoniformes	Salmonidae	*Coregonus kiyi*	kiyi
Salmoniformes	Salmonidae	*Coregonus laurettae*	Bering cisco
Salmoniformes	Salmonidae	*Coregonus nasus*	broad whitefish
Salmoniformes	Salmonidae	*Coregonus nigripinnis*	blackfin cisco
Salmoniformes	Salmonidae	*Coregonus pidschian*	humpback whitefish
Salmoniformes	Salmonidae	*Coregonus reighardi*	shortnose cisco
Salmoniformes	Salmonidae	*Coregonus sardinella*	least cisco
Salmoniformes	Salmonidae	*Coregonus zenithicus*	shortjaw cisco
Salmoniformes	Salmonidae	*Oncorhynchus clarkii*	cutthroat trout
Salmoniformes	Salmonidae	*Oncorhynchus gilae*	Gila trout
Salmoniformes	Salmonidae	*Oncorhynchus gorbuscha*	pink salmon
Salmoniformes	Salmonidae	*Oncorhynchus keta*	chum salmon
Salmoniformes	Salmonidae	*Oncorhynchus kisutch*	coho salmon
Salmoniformes	Salmonidae	*Oncorhynchus mykiss*	rainbow trout
Salmoniformes	Salmonidae	*Oncorhynchus nerka*	sockeye salmon
Salmoniformes	Salmonidae	*Oncorhynchus tshawytscha*	Chinook salmon
Salmoniformes	Salmonidae	*Prosopium abyssicola*	Bear Lake whitefish
Salmoniformes	Salmonidae	*Prosopium coulterii*	pygmy whitefish
Salmoniformes	Salmonidae	*Prosopium cylindraceum*	round whitefish
Salmoniformes	Salmonidae	*Prosopium gemmifer*	Bonneville cisco
Salmoniformes	Salmonidae	*Prosopium spilonotus*	Bonneville whitefish
Salmoniformes	Salmonidae	*Prosopium williamsoni*	mountain whitefish
Salmoniformes	Salmonidae	*Salmo salar*	Atlantic salmon
Salmoniformes	Salmonidae	*Salmo trutta*	brown trout

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Salmoniformes	Salmonidae	*Salvelinus alpinus*	Arctic char
Salmoniformes	Salmonidae	*Salvelinus confluentus*	bull trout
Salmoniformes	Salmonidae	*Salvelinus fontinalis*	brook trout
Salmoniformes	Salmonidae	*Salvelinus malma*	Dolly Varden
Salmoniformes	Salmonidae	*Salvelinus namaycush*	lake trout
Salmoniformes	Salmonidae	*Stenodus leucichthys*	inconnu
Salmoniformes	Salmonidae	*Thymallus arcticus*	Arctic grayling
Percopsiformes	Percopsidae	*Percopsis omiscomaycus*	trout-perch
Percopsiformes	Percopsidae	*Percopsis transmontana*	sand roller
Percopsiformes	Aphredoderidae	*Aphredoderus sayanus*	pirate perch
Percopsiformes	Amblyopsidae	*Amblyopsis rosae*	Ozark cavefish
Percopsiformes	Amblyopsidae	*Amblyopsis spelaea*	northern cavefish
Percopsiformes	Amblyopsidae	*Chologaster cornuta*	swampfish
Percopsiformes	Amblyopsidae	*Forbesichthys agassizii*	spring cavefish
Percopsiformes	Amblyopsidae	*Speoplatyrhinus poulsoni*	Alabama cavefish
Percopsiformes	Amblyopsidae	*Typhlichthys subterraneus*	southern cavefish
Gadiformes	Gadidae	*Lota lota*	burbot
Gadiformes	Gadidae	*Microgadus tomcod*	Atlantic tomcod
Mugiliformes	Mugilidae	*Agonostomus monticola*	mountain mullet
Mugiliformes	Mugilidae	*Mugil cephalus*	striped mullet
Mugiliformes	Mugilidae	*Mugil curema*	white mullet
Atheriniformes	Atherinopsidae	*Labidesthes sicculus*	brook silverside
Atheriniformes	Atherinopsidae	*Membras martinica*	rough silverside
Atheriniformes	Atherinopsidae	*Menidia audens*	Mississippi silverside
Atheriniformes	Atherinopsidae	*Menidia beryllina*	inland silverside
Atheriniformes	Atherinopsidae	*Menidia extensa*	Waccamaw silverside
Beloniformes	Belonidae	*Strongylura marina*	Atlantic needlefish
Cyprinodontiformes	Aplocheilidae	*Rivulus hartii*	giant rivulus
Cyprinodontiformes	Aplocheilidae	*Rivulus marmoratus*	mangrove rivulus
Cyprinodontiformes	Fundulidae	*Fundulus albolineatus*	whiteline topminnow
Cyprinodontiformes	Fundulidae	*Fundulus bifax*	stippled studfish
Cyprinodontiformes	Fundulidae	*Fundulus blairae*	western starhead topminnow
Cyprinodontiformes	Fundulidae	*Fundulus catenatus*	northern studfish
Cyprinodontiformes	Fundulidae	*Fundulus chrysotus*	golden topminnow
Cyprinodontiformes	Fundulidae	*Fundulus cingulatus*	banded topminnow
Cyprinodontiformes	Fundulidae	*Fundulus confluentus*	marsh killifish
Cyprinodontiformes	Fundulidae	*Fundulus diaphanus*	banded killifish
Cyprinodontiformes	Fundulidae	*Fundulus dispar*	starhead topminnow
Cyprinodontiformes	Fundulidae	*Fundulus escambiae*	russetfin topminnow
Cyprinodontiformes	Fundulidae	*Fundulus euryzonus*	broadstripe topminnow
Cyprinodontiformes	Fundulidae	*Fundulus grandis*	Gulf killifish
Cyprinodontiformes	Fundulidae	*Fundulus heteroclitus*	mummichog
Cyprinodontiformes	Fundulidae	*Fundulus jenkinsi*	saltmarsh topminnow
Cyprinodontiformes	Fundulidae	*Fundulus julisia*	Barrens topminnow
Cyprinodontiformes	Fundulidae	*Fundulus kansae*	northern plains killifish
Cyprinodontiformes	Fundulidae	*Fundulus lineolatus*	lined topminnow
Cyprinodontiformes	Fundulidae	*Fundulus luciae*	spotfin killifish

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Cyprinodontiformes	Fundulidae	*Fundulus notatus*	blackstripe topminnow
Cyprinodontiformes	Fundulidae	*Fundulus nottii*	bayou topminnow
Cyprinodontiformes	Fundulidae	*Fundulus olivaceus*	blackspotted topminnow
Cyprinodontiformes	Fundulidae	*Fundulus parvipinnis*	Guadalupe cardinalfish
Cyprinodontiformes	Fundulidae	*Fundulus pulvereus*	bayou killifish
Cyprinodontiformes	Fundulidae	*Fundulus rathbuni*	speckled killifish
Cyprinodontiformes	Fundulidae	*Fundulus rubrifrons*	redface topminnow
Cyprinodontiformes	Fundulidae	*Fundulus sciadicus*	plains topminnow
Cyprinodontiformes	Fundulidae	*Fundulus seminolis*	Seminole killifish
Cyprinodontiformes	Fundulidae	*Fundulus stellifer*	southern studfish
Cyprinodontiformes	Fundulidae	*Fundulus waccamensis*	Waccamaw killifish
Cyprinodontiformes	Fundulidae	*Fundulus zebrinus*	plains killifish
Cyprinodontiformes	Fundulidae	*Leptolucania ommata*	pygmy killifish
Cyprinodontiformes	Fundulidae	*Lucania goodei*	bluefin killifish
Cyprinodontiformes	Fundulidae	*Lucania parva*	rainwater killifish
Cyprinodontiformes	Poeciliidae	*Belonesox belizanus*	pike killifish
Cyprinodontiformes	Poeciliidae	*Gambusia affinis*	western mosquitofish
Cyprinodontiformes	Poeciliidae	*Gambusia amistadensis*	Amistad gambusia
Cyprinodontiformes	Poeciliidae	*Gambusia gaigei*	Big Bend gambusia
Cyprinodontiformes	Poeciliidae	*Gambusia geiseri*	largespring gambusia
Cyprinodontiformes	Poeciliidae	*Gambusia georgei*	San Marcos gambusia
Cyprinodontiformes	Poeciliidae	*Gambusia heterochir*	Clear Creek gambusia
Cyprinodontiformes	Poeciliidae	*Gambusia holbrooki*	eastern mosquitofish
Cyprinodontiformes	Poeciliidae	*Gambusia nobilis*	Pecos gambusia
Cyprinodontiformes	Poeciliidae	*Gambusia rhizophorae*	mangrove gambusia
Cyprinodontiformes	Poeciliidae	*Gambusia senilis*	blotched gambusia
Cyprinodontiformes	Poeciliidae	*Gambusia speciosa*	Tex-Mex gambusia
Cyprinodontiformes	Poeciliidae	*Heterandria formosa*	least killifish
Cyprinodontiformes	Poeciliidae	*Poecilia formosa*	Amazon molly
Cyprinodontiformes	Poeciliidae	*Poecilia latipinna*	sailfin molly
Cyprinodontiformes	Poeciliidae	*Poecilia mexicana*	shortfin molly
Cyprinodontiformes	Poeciliidae	*Poecilia reticulata*	guppy
Cyprinodontiformes	Poeciliidae	*Poecilia sphenops*	Mexican molly
Cyprinodontiformes	Poeciliidae	*Poeciliopsis gracilis*	porthole livebearer
Cyprinodontiformes	Poeciliidae	*Poeciliopsis occidentalis*	Gila topminnow
Cyprinodontiformes	Poeciliidae	*Xiphophorus hellerii*	green swordtail
Cyprinodontiformes	Poeciliidae	*Xiphophorus maculatus*	southern platyfish
Cyprinodontiformes	Poeciliidae	*Xiphophorus variatus*	variable platyfish
Cyprinodontiformes	Goodeidae	*Crenichthys baileyi*	White River springfish
Cyprinodontiformes	Goodeidae	*Crenichthys nevadae*	Railroad Valley springfish
Cyprinodontiformes	Goodeidae	*Empetrichthys latos*	Pahrump poolfish
Cyprinodontiformes	Goodeidae	*Empetrichthys merriami*	Ash Meadows poolfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon arcuatus*	Santa Cruz pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon bovinus*	Leon Springs pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon diabolis*	Devils Hole pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon elegans*	Comanche Springs pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon eremus*	Sonoyta pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon eximius*	Conchos pupfish

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon macularius*	desert pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon nevadensis*	Amargosa pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon pecosensis*	Pecos pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon radiosus*	Owens pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon rubrofluviatilis*	Red River pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon salinus*	Salt Creek pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon tularosa*	White Sands pupfish
Cyprinodontiformes	Cyprinodontidae	*Cyprinodon variegatus*	sheepshead minnow
Cyprinodontiformes	Cyprinodontidae	*Jordanella floridae*	flagfish
Gasterosteiformes	Gasterosteidae	*Apeltes quadracus*	fourspine stickleback
Gasterosteiformes	Gasterosteidae	*Culaea inconstans*	brook stickleback
Gasterosteiformes	Gasterosteidae	*Gasterosteus aculeatus*	espinocho
Gasterosteiformes	Gasterosteidae	*Pungitius pungitius*	ninespine stickleback
Gasterosteiformes	Syngnathidae	*Microphis brachyurus*	opossum pipefish
Gasterosteiformes	Syngnathidae	*Syngnathus scovelli*	Gulf pipefish
Synbranchiformes	Synbranchidae	*Monopterus albus*	Asian swamp eel
Scorpaeniformes	Cottidae	*Clinocottus acuticeps*	sharpnose sculpin
Scorpaeniformes	Cottidae	*Cottus aleuticus*	coastrange sculpin
Scorpaeniformes	Cottidae	*Cottus asper*	prickly sculpin
Scorpaeniformes	Cottidae	*Cottus asperrimus*	rough sculpin
Scorpaeniformes	Cottidae	*Cottus baileyi*	black sculpin
Scorpaeniformes	Cottidae	*Cottus bairdii*	mottled sculpin
Scorpaeniformes	Cottidae	*Cottus beldingii*	Paiute sculpin
Scorpaeniformes	Cottidae	*Cottus bendirei*	Malheur sculpin
Scorpaeniformes	Cottidae	*Cottus caeruleomentum*	Blue Ridge sculpin
Scorpaeniformes	Cottidae	*Cottus carolinae*	banded sculpin
Scorpaeniformes	Cottidae	*Cottus cognatus*	slimy sculpin
Scorpaeniformes	Cottidae	*Cottus confusus*	shorthead sculpin
Scorpaeniformes	Cottidae	*Cottus echinatus*	Utah Lake sculpin
Scorpaeniformes	Cottidae	*Cottus extensus*	Bear Lake sculpin
Scorpaeniformes	Cottidae	*Cottus girardi*	Potomac sculpin
Scorpaeniformes	Cottidae	*Cottus greenei*	Shoshone sculpin
Scorpaeniformes	Cottidae	*Cottus gulosus*	riffle sculpin
Scorpaeniformes	Cottidae	*Cottus hubbsi*	Columbia sculpin
Scorpaeniformes	Cottidae	*Cottus hypselurus*	Ozark sculpin
Scorpaeniformes	Cottidae	*Cottus klamathensis*	marbled sculpin
Scorpaeniformes	Cottidae	*Cottus leiopomus*	Wood River sculpin
Scorpaeniformes	Cottidae	*Cottus marginatus*	margined sculpin
Scorpaeniformes	Cottidae	*Cottus paulus*	pygmy sculpin
Scorpaeniformes	Cottidae	*Cottus perplexus*	reticulate sculpin
Scorpaeniformes	Cottidae	*Cottus pitensis*	Pit sculpin
Scorpaeniformes	Cottidae	*Cottus princeps*	Klamath Lake sculpin
Scorpaeniformes	Cottidae	*Cottus rhotheus*	torrent sculpin
Scorpaeniformes	Cottidae	*Cottus ricei*	spoonhead sculpin
Scorpaeniformes	Cottidae	*Cottus tenuis*	slender sculpin
Scorpaeniformes	Cottidae	*Leptocottus armatus*	Pacific staghorn sculpin
Scorpaeniformes	Cottidae	*Myoxocephalus quadricornis*	fourhorn sculpin
Scorpaeniformes	Cottidae	*Myoxocephalus thompsonii*	deepwater sculpin

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Perciformes	Centropomidae	*Centropomus ensiferus*	swordspine snook
Perciformes	Centropomidae	*Centropomus parallelus*	smallscale fat snook
Perciformes	Centropomidae	*Centropomus pectinatus*	tarpon snook
Perciformes	Centropomidae	*Centropomus undecimalis*	common snook
Perciformes	Moronidae	*Morone americana*	white perch
Perciformes	Moronidae	*Morone chrysops*	white bass
Perciformes	Moronidae	*Morone mississippiensis*	yellow bass
Perciformes	Moronidae	*Morone saxatilis*	striped bass
Perciformes	Centrarchidae	*Acantharchus pomotis*	mud sunfish
Perciformes	Centrarchidae	*Ambloplites ariommus*	shadow bass
Perciformes	Centrarchidae	*Ambloplites cavifrons*	Roanoke bass
Perciformes	Centrarchidae	*Ambloplites constellatus*	Ozark bass
Perciformes	Centrarchidae	*Ambloplites rupestris*	rock bass
Perciformes	Centrarchidae	*Archoplites interruptus*	Sacramento perch
Perciformes	Centrarchidae	*Centrarchus macropterus*	flier
Perciformes	Centrarchidae	*Enneacanthus chaetodon*	blackbanded sunfish
Perciformes	Centrarchidae	*Enneacanthus gloriosus*	bluespotted sunfish
Perciformes	Centrarchidae	*Enneacanthus obesus*	banded sunfish
Perciformes	Centrarchidae	*Lepomis auritus*	redbreast sunfish
Perciformes	Centrarchidae	*Lepomis cyanellus*	green sunfish
Perciformes	Centrarchidae	*Lepomis gibbosus*	pumpkinseed
Perciformes	Centrarchidae	*Lepomis gulosus*	warmouth
Perciformes	Centrarchidae	*Lepomis humilis*	orangespotted sunfish
Perciformes	Centrarchidae	*Lepomis macrochirus*	bluegill
Perciformes	Centrarchidae	*Lepomis marginatus*	dollar sunfish
Perciformes	Centrarchidae	*Lepomis megalotis*	longear sunfish
Perciformes	Centrarchidae	*Lepomis microlophus*	redear sunfish
Perciformes	Centrarchidae	*Lepomis miniatus*	redspotted sunfish
Perciformes	Centrarchidae	*Lepomis punctatus*	spotted sunfish
Perciformes	Centrarchidae	*Lepomis symmetricus*	bantam sunfish
Perciformes	Centrarchidae	*Micropterus cataractae*	shoal bass
Perciformes	Centrarchidae	*Micropterus coosae*	redeye bass
Perciformes	Centrarchidae	*Micropterus dolomieu*	smallmouth bass
Perciformes	Centrarchidae	*Micropterus notius*	Suwannee bass
Perciformes	Centrarchidae	*Micropterus punctulatus*	spotted bass
Perciformes	Centrarchidae	*Micropterus salmoides*	largemouth bass
Perciformes	Centrarchidae	*Micropterus treculii*	Guadalupe bass
Perciformes	Centrarchidae	*Pomoxis annularis*	white crappie
Perciformes	Centrarchidae	*Pomoxis nigromaculatus*	black crappie
Perciformes	Percidae	*Ammocrypta beanii*	naked sand darter
Perciformes	Percidae	*Ammocrypta bifascia*	Florida sand darter
Perciformes	Percidae	*Ammocrypta clara*	western sand darter
Perciformes	Percidae	*Ammocrypta meridiana*	southern sand darter
Perciformes	Percidae	*Ammocrypta pellucida*	eastern sand darter
Perciformes	Percidae	*Ammocrypta vivax*	scaly sand darter
Perciformes	Percidae	*Crystallaria asprella*	crystal darter
Perciformes	Percidae	*Etheostoma acuticeps*	sharphead darter
Perciformes	Percidae	*Etheostoma aquali*	coppercheek darter

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Perciformes	Percidae	*Etheostoma artesiae*	redspot darter
Perciformes	Percidae	*Etheostoma asprigene*	mud darter
Perciformes	Percidae	*Etheostoma baileyi*	emerald darter
Perciformes	Percidae	*Etheostoma barbouri*	teardrop darter
Perciformes	Percidae	*Etheostoma barrenense*	splendid darter
Perciformes	Percidae	*Etheostoma basilare*	corrugated darter
Perciformes	Percidae	*Etheostoma bellator*	Warrior darter
Perciformes	Percidae	*Etheostoma bellum*	orangefin darter
Perciformes	Percidae	*Etheostoma bison*	Buffalo darter
Perciformes	Percidae	*Etheostoma blennioides*	greenside darter
Perciformes	Percidae	*Etheostoma blennius*	blenny darter
Perciformes	Percidae	*Etheostoma boschungi*	slackwater darter
Perciformes	Percidae	*Etheostoma brevirostrum*	holiday darter
Perciformes	Percidae	*Etheostoma burri*	brook darter
Perciformes	Percidae	*Etheostoma caeruleum*	rainbow darter
Perciformes	Percidae	*Etheostoma camurum*	bluebreast darter
Perciformes	Percidae	*Etheostoma cervus*	Chickasaw darter
Perciformes	Percidae	*Etheostoma chermocki*	vermilion darter
Perciformes	Percidae	*Etheostoma chienense*	relict darter
Perciformes	Percidae	*Etheostoma chlorobranchium*	greenfin darter
Perciformes	Percidae	*Etheostoma chlorosoma*	bluntnose darter
Perciformes	Percidae	*Etheostoma chuckwachatte*	lipstick darter
Perciformes	Percidae	*Etheostoma cinereum*	ashy darter
Perciformes	Percidae	*Etheostoma collettei*	creole darter
Perciformes	Percidae	*Etheostoma collis*	Carolina darter
Perciformes	Percidae	*Etheostoma colorosum*	coastal darter
Perciformes	Percidae	*Etheostoma coosae*	Coosa darter
Perciformes	Percidae	*Etheostoma corona*	crown darter
Perciformes	Percidae	*Etheostoma cragini*	Arkansas darter
Perciformes	Percidae	*Etheostoma crossopterum*	fringed darter
Perciformes	Percidae	*Etheostoma davisoni*	Choctawhatchee darter
Perciformes	Percidae	*Etheostoma denoncourti*	golden darter
Perciformes	Percidae	*Etheostoma derivativum*	stone darter
Perciformes	Percidae	*Etheostoma ditrema*	coldwater darter
Perciformes	Percidae	*Etheostoma douglasi*	Tuskaloosa darter
Perciformes	Percidae	*Etheostoma duryi*	blackside snubnose darter
Perciformes	Percidae	*Etheostoma edwini*	brown darter
Perciformes	Percidae	*Etheostoma etnieri*	cherry darter
Perciformes	Percidae	*Etheostoma etowahae*	Etowah darter
Perciformes	Percidae	*Etheostoma euzonum*	Arkansas saddled darter
Perciformes	Percidae	*Etheostoma exile*	Iowa darter
Perciformes	Percidae	*Etheostoma flabellare*	fantail darter
Perciformes	Percidae	*Etheostoma flavum*	saffron darter
Perciformes	Percidae	*Etheostoma fonticola*	fountain darter
Perciformes	Percidae	*Etheostoma forbesi*	Barrens darter
Perciformes	Percidae	*Etheostoma fragi*	Strawberry darter
Perciformes	Percidae	*Etheostoma fricksium*	Savannah darter
Perciformes	Percidae	*Etheostoma fusiforme*	swamp darter

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Perciformes	Percidae	*Etheostoma gracile*	slough darter
Perciformes	Percidae	*Etheostoma grahami*	Rio Grande darter
Perciformes	Percidae	*Etheostoma gutselli*	Tuckasegee darter
Perciformes	Percidae	*Etheostoma histrio*	harlequin darter
Perciformes	Percidae	*Etheostoma hopkinsi*	Christmas darter
Perciformes	Percidae	*Etheostoma inscriptum*	turquoise darter
Perciformes	Percidae	*Etheostoma jessiae*	blueside darter
Perciformes	Percidae	*Etheostoma jordani*	greenbreast darter
Perciformes	Percidae	*Etheostoma juliae*	yoke darter
Perciformes	Percidae	*Etheostoma kanawhae*	Kanawha darter
Perciformes	Percidae	*Etheostoma kantuckeense*	Highland Rim darter
Perciformes	Percidae	*Etheostoma kennicotti*	stripetail darter
Perciformes	Percidae	*Etheostoma lachneri*	Tombigbee darter
Perciformes	Percidae	*Etheostoma lawrencei*	headwater darter
Perciformes	Percidae	*Etheostoma lepidum*	greenthroat darter
Perciformes	Percidae	*Etheostoma longimanum*	longfin darter
Perciformes	Percidae	*Etheostoma luteovinctum*	redband darter
Perciformes	Percidae	*Etheostoma lynceum*	brighteye darter
Perciformes	Percidae	*Etheostoma maculatum*	spotted darter
Perciformes	Percidae	*Etheostoma mariae*	pinewoods darter
Perciformes	Percidae	*Etheostoma microlepidum*	smallscale darter
Perciformes	Percidae	*Etheostoma microperca*	least darter
Perciformes	Percidae	*Etheostoma moorei*	yellowcheek darter
Perciformes	Percidae	*Etheostoma neopterum*	lollypop darter
Perciformes	Percidae	*Etheostoma nianguae*	Niangua darter
Perciformes	Percidae	*Etheostoma nigripinne*	blackfin darter
Perciformes	Percidae	*Etheostoma nigrum*	johnny darter
Perciformes	Percidae	*Etheostoma nuchale*	watercress darter
Perciformes	Percidae	*Etheostoma obeyense*	barcheek darter
Perciformes	Percidae	*Etheostoma okaloosae*	Okaloosa darter
Perciformes	Percidae	*Etheostoma olivaceum*	sooty darter
Perciformes	Percidae	*Etheostoma olmstedi*	tessellated darter
Perciformes	Percidae	*Etheostoma oophylax*	guardian darter
Perciformes	Percidae	*Etheostoma osburni*	candy darter
Perciformes	Percidae	*Etheostoma pallididorsum*	paleback darter
Perciformes	Percidae	*Etheostoma parvipinne*	goldstripe darter
Perciformes	Percidae	*Etheostoma percnurum*	duskytail darter
Perciformes	Percidae	*Etheostoma perlongum*	Waccamaw darter
Perciformes	Percidae	*Etheostoma phytophilum*	rush darter
Perciformes	Percidae	*Etheostoma podostemone*	riverweed darter
Perciformes	Percidae	*Etheostoma proeliare*	cypress darter
Perciformes	Percidae	*Etheostoma pseudovulatum*	egg-mimic darter
Perciformes	Percidae	*Etheostoma punctulatum*	stippled darter
Perciformes	Percidae	*Etheostoma pyrrhogaster*	firebelly darter
Perciformes	Percidae	*Etheostoma radiosum*	orangebelly darter
Perciformes	Percidae	*Etheostoma rafinesquei*	Kentucky darter
Perciformes	Percidae	*Etheostoma ramseyi*	Alabama darter
Perciformes	Percidae	*Etheostoma raneyi*	Yazoo darter

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Perciformes	Percidae	*Etheostoma rubrum*	bayou darter
Perciformes	Percidae	*Etheostoma rufilineatum*	redline darter
Perciformes	Percidae	*Etheostoma rupestre*	rock darter
Perciformes	Percidae	*Etheostoma sagitta*	arrow darter
Perciformes	Percidae	*Etheostoma sanguifluum*	bloodfin darter
Perciformes	Percidae	*Etheostoma scotti*	Cherokee darter
Perciformes	Percidae	*Etheostoma sellare*	Maryland darter
Perciformes	Percidae	*Etheostoma serrifer*	sawcheek darter
Perciformes	Percidae	*Etheostoma simoterum*	snubnose darter
Perciformes	Percidae	*Etheostoma smithi*	slabrock darter
Perciformes	Percidae	*Etheostoma spectabile*	orangethroat darter
Perciformes	Percidae	*Etheostoma squamiceps*	spottail darter
Perciformes	Percidae	*Etheostoma stigmaeum*	speckled darter
Perciformes	Percidae	*Etheostoma striatulum*	striated darter
Perciformes	Percidae	*Etheostoma susanae*	Cumberland darter
Perciformes	Percidae	*Etheostoma swaini*	Gulf darter
Perciformes	Percidae	*Etheostoma swannanoa*	Swannanoa darter
Perciformes	Percidae	*Etheostoma tallapoosae*	Tallapoosa darter
Perciformes	Percidae	*Etheostoma tecumsehi*	Shawnee darter
Perciformes	Percidae	*Etheostoma tetrazonum*	Missouri saddled darter
Perciformes	Percidae	*Etheostoma thalassinum*	seagreen darter
Perciformes	Percidae	*Etheostoma tippecanoe*	Tippecanoe darter
Perciformes	Percidae	*Etheostoma trisella*	trispot darter
Perciformes	Percidae	*Etheostoma tuscumbia*	Tuscumbia darter
Perciformes	Percidae	*Etheostoma uniporum*	current darter
Perciformes	Percidae	*Etheostoma variatum*	variegate darter
Perciformes	Percidae	*Etheostoma virgatum*	striped darter
Perciformes	Percidae	*Etheostoma vitreum*	glassy darter
Perciformes	Percidae	*Etheostoma vulneratum*	wounded darter
Perciformes	Percidae	*Etheostoma wapiti*	boulder darter
Perciformes	Percidae	*Etheostoma whipplei*	redfin darter
Perciformes	Percidae	*Etheostoma zonale*	banded darter
Perciformes	Percidae	*Etheostoma zonifer*	backwater darter
Perciformes	Percidae	*Etheostoma zonistium*	bandfin darter
Perciformes	Percidae	*Gymnocephalus cernuus*	ruffe
Perciformes	Percidae	*Perca flavescens*	yellow perch
Perciformes	Percidae	*Percina antesella*	amber darter
Perciformes	Percidae	*Percina aurantiaca*	tangerine darter
Perciformes	Percidae	*Percina aurolineata*	goldline darter
Perciformes	Percidae	*Percina aurora*	pearl darter
Perciformes	Percidae	*Percina austroperca*	southern logperch
Perciformes	Percidae	*Percina brevicauda*	coal darter
Perciformes	Percidae	*Percina burtoni*	blotchside logperch
Perciformes	Percidae	*Percina caprodes*	logperch
Perciformes	Percidae	*Percina carbonaria*	Texas logperch
Perciformes	Percidae	*Percina copelandi*	channel darter
Perciformes	Percidae	*Percina crassa*	Piedmont darter
Perciformes	Percidae	*Percina cymatotaenia*	bluestripe darter

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Perciformes	Percidae	*Percina evides*	gilt darter
Perciformes	Percidae	*Percina fulvitaenia*	Ozark logperch
Perciformes	Percidae	*Percina gymnocephala*	Appalachia darter
Perciformes	Percidae	*Percina jenkinsi*	Conasauga logperch
Perciformes	Percidae	*Percina kathae*	Mobile logperch
Perciformes	Percidae	*Percina lenticula*	freckled darter
Perciformes	Percidae	*Percina macrocephala*	longhead darter
Perciformes	Percidae	*Percina macrolepida*	bigscale logperch
Perciformes	Percidae	*Percina maculata*	blackside darter
Perciformes	Percidae	*Percina nasuta*	longnose darter
Perciformes	Percidae	*Percina nevisense*	chainback darter
Perciformes	Percidae	*Percina nigrofasciata*	blackbanded darter
Perciformes	Percidae	*Percina notogramma*	stripeback darter
Perciformes	Percidae	*Percina oxyrhynchus*	sharpnose darter
Perciformes	Percidae	*Percina palmaris*	bronze darter
Perciformes	Percidae	*Percina pantherina*	leopard darter
Perciformes	Percidae	*Percina peltata*	shield darter
Perciformes	Percidae	*Percina phoxocephala*	slenderhead darter
Perciformes	Percidae	*Percina rex*	Roanoke logperch
Perciformes	Percidae	*Percina roanoka*	Roanoke darter
Perciformes	Percidae	*Percina sciera*	dusky darter
Perciformes	Percidae	*Percina shumardi*	river darter
Perciformes	Percidae	*Percina squamata*	olive darter
Perciformes	Percidae	*Percina stictogaster*	frecklebelly darter
Perciformes	Percidae	*Percina suttkusi*	Gulf logperch
Perciformes	Percidae	*Percina tanasi*	snail darter
Perciformes	Percidae	*Percina uranidea*	stargazing darter
Perciformes	Percidae	*Percina vigil*	saddleback darter
Perciformes	Percidae	*Sander canadensis*	sauger
Perciformes	Percidae	*Sander lucioperca*	zander
Perciformes	Percidae	*Sander vitreus*	walleye
Perciformes	Lutjanidae	*Lutjanus griseus*	gray snapper
Perciformes	Gerreidae	*Diapterus auratus*	Irish pompano
Perciformes	Gerreidae	*Eucinostomus harengulus*	tidewater mojarra
Perciformes	Gerreidae	*Eugerres plumieri*	striped mojarra
Perciformes	Haemulidae	*Orthopristis chrysoptera*	pigfish
Perciformes	Sparidae	*Archosargus probatocephalus*	sheepshead
Perciformes	Sparidae	*Lagodon rhomboides*	pinfish
Perciformes	Sciaenidae	*Aplodinotus grunniens*	freshwater drum
Perciformes	Sciaenidae	*Bairdiella chrysoura*	silver perch
Perciformes	Sciaenidae	*Bairdiella icistia*	bairdiella
Perciformes	Sciaenidae	*Cynoscion nebulosus*	spotted seatrout
Perciformes	Sciaenidae	*Cynoscion xanthulus*	orangemouth corvina
Perciformes	Sciaenidae	*Leiostomus xanthurus*	spot
Perciformes	Sciaenidae	*Micropogonias undulatus*	Atlantic croaker
Perciformes	Sciaenidae	*Sciaenops ocellatus*	red drum
Perciformes	Elassomatidae	*Elassoma alabamae*	spring pygmy sunfish
Perciformes	Elassomatidae	*Elassoma boehlkei*	Carolina pygmy sunfish

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Perciformes	Elassomatidae	*Elassoma evergladei*	Everglades pygmy sunfish
Perciformes	Elassomatidae	*Elassoma okatie*	bluebarred pygmy sunfish
Perciformes	Elassomatidae	*Elassoma okefenokee*	Okefenokee pygmy sunfish
Perciformes	Elassomatidae	*Elassoma zonatum*	banded pygmy sunfish
Perciformes	Cichlidae	*Astronotus ocellatus*	oscar
Perciformes	Cichlidae	*Cichla ocellaris*	butterfly peacock bass
Perciformes	Cichlidae	*Cichlasoma bimaculatum*	black acara
Perciformes	Cichlidae	*Cichlasoma citrinellum*	midas cichlid
Perciformes	Cichlidae	*Cichlasoma cyanoguttatum*	Rio Grande cichlid
Perciformes	Cichlidae	*Cichlasoma managuense*	jaguar guapote
Perciformes	Cichlidae	*Cichlasoma meeki*	firemouth cichlid
Perciformes	Cichlidae	*Cichlasoma nigrofasciatum*	convict cichlid
Perciformes	Cichlidae	*Cichlasoma octofasciatum*	Jack Dempsey
Perciformes	Cichlidae	*Cichlasoma salvini*	yellowbelly cichlid
Perciformes	Cichlidae	*Cichlasoma urophthalmus*	Mayan cichlid
Perciformes	Cichlidae	*Geophagus surinamensis*	redstriped eartheater
Perciformes	Cichlidae	*Hemichromis letourneuxi*	African jewelfish
Perciformes	Cichlidae	*Heros severus*	banded cichlid
Perciformes	Cichlidae	*Oreochromis aureus*	blue tilapia
Perciformes	Cichlidae	*Oreochromis mossambicus*	Mozambique tilapia
Perciformes	Cichlidae	*Oreochromis niloticus*	Nile tilapia
Perciformes	Cichlidae	*Oreochromis urolepis*	Wami tilapia
Perciformes	Cichlidae	*Sarotherodon melanotheron*	blackchin tilapia
Perciformes	Cichlidae	*Tilapia mariae*	spotted tilapia
Perciformes	Cichlidae	*Tilapia zillii*	redbelly tilapia
Perciformes	Embiotocidae	*Cymatogaster aggregata*	shiner perch
Perciformes	Embiotocidae	*Hysterocarpus traskii*	tule perch
Perciformes	Eleotridae	*Dormitator maculatus*	fat sleeper
Perciformes	Eleotridae	*Eleotris amblyopsis*	largescaled spinycheek
Perciformes	Eleotridae	*Eleotris perniger*	smallscaled spinycheek
Perciformes	Eleotridae	*Eleotris picta*	spotted sleeper
Perciformes	Eleotridae	*Gobiomorus dormitor*	bigmouth sleeper
Perciformes	Eleotridae	*Guavina guavina*	guavina
Perciformes	Gobiidae	*Acanthogobius flavimanus*	yellowfin goby
Perciformes	Gobiidae	*Awaous banana*	river goby
Perciformes	Gobiidae	*Clevelandia ios*	arrow goby
Perciformes	Gobiidae	*Ctenogobius boleosoma*	darter goby
Perciformes	Gobiidae	*Ctenogobius claytonii*	Mexican goby
Perciformes	Gobiidae	*Ctenogobius fasciatus*	blotchcheek goby
Perciformes	Gobiidae	*Ctenogobius pseudofasciatus*	slashcheek goby
Perciformes	Gobiidae	*Ctenogobius shufeldti*	freshwater goby
Perciformes	Gobiidae	*Eucyclogobius newberryi*	tidewater goby
Perciformes	Gobiidae	*Gillichthys mirabilis*	longjaw mudsucker
Perciformes	Gobiidae	*Gobioides broussonetii*	violet goby
Perciformes	Gobiidae	*Gobiosoma bosc*	naked goby
Perciformes	Gobiidae	*Lophogobius cyprinoides*	crested goby
Perciformes	Gobiidae	*Microgobius gulosus*	clown goby
Perciformes	Gobiidae	*Neogobius melanostomus*	round goby

ORDER	FAMILY	SCIENTIFIC NAME	COMMON NAME
Perciformes	Gobiidae	*Proterorhinus marmoratus*	tubenose goby
Perciformes	Gobiidae	*Tridentiger barbatus*	Shokihaze goby
Perciformes	Gobiidae	*Tridentiger bifasciatus*	shimofuri goby
Perciformes	Belontiidae	*Trichopsis vittata*	croaking gourami
Perciformes	Channidae	*Channa marulius*	bullseye snakehead
Pleuronectiformes	Paralichthyidae	*Citharichthys spilopterus*	bay whiff
Pleuronectiformes	Paralichthyidae	*Paralichthys lethostigma*	southern flounder
Pleuronectiformes	Pleuronectidae	*Platichthys stellatus*	starry flounder
Pleuronectiformes	Achiridae	*Trinectes maculatus*	hogchoker

APPENDIX E

PPCP and PFC Samples at Selected Urban Sites

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EPA's Office of Science and Technology (OST) within the Office of Water is collaborating with the Office of Research and Development's National Exposure Research Laboratory in Cincinnati, Ohio to conduct a study of contaminants of emerging concern (CECs) within the framework of the National Rivers and Streams Assessment (NRSA). These CECs include pharmaceuticals and personal care products (PPCPs), along with perfluorinated compounds (PFCs). This study involves collection of ambient water (water chemistry) samples and fish tissue samples at about 150 urban river sites. These sites comprise a statistical subset within the 1800 sites selected for NRSA sampling. The urban river sites were assigned to the PPCP and PFC Study based on 5th order or greater Strahler stream order. **The majority of these sites will be boatable, but a few of them will be wadeable. PPCP and PFC water and fish tissue samples need to be collected at the boatable and wadeable sites in this subset of urban river locations to maintain the statistical integrity of the data.**

PPCP Water Chemistry Samples

The water chemistry protocols for collection of PPCP water samples are identical to the general water chemistry sample collection protocols for the NRSA water quality indicators. OST will provide field teams with coolers and 500 ml (0.5 L) amber glass bottles for the PPCP water samples. Water for the PPCP samples will be collected using the beaker provided for collection of other water chemistry samples. Field teams will use river water from the beaker to rinse the sample bottles and caps before filling each of the sample bottles completely with water from the beaker to eliminate air from the bottle. After fastening the caps tightly on the sample bottles, the field crews will place the samples in the cooler on wet ice. **Field teams will collect two 500 ml PPCP water samples at all the urban river sites (boatable and wadeable) except the repeat urban river sites. At the repeat urban river sites, field teams will collect four 500 ml PPCP water samples during the first site visit only.**

1. Collect the PPCP water samples mid-channel at the X-site (located via GPS). Samples are taken mid-channel, at a depth of 0.5 meters or at mid-depth if the site is less than 1 meter deep.
2. Put on nitrile gloves. Avoid touching the inside of the container to prevent contamination. Make sure not to handle sunscreen or other chemical contaminants until after the sample is collected.
3. Pre-rinse the beaker with river water 3 times, discarding rinse water downstream. Hold the container so the opening faces upstream. Collect the sample at a depth of 0.5 meters below the surface, with the beaker slightly angled as you pull it to the surface.
4. Rinse each PPCP sample bottle with a small amount of the sample water before filling the sample bottle with water from the beaker.
5. Fill the two 500 ml amber glass bottles (or four 500 ml amber glass bottles during the first visit at urban river repeat sites) using water from the beaker. After filling each sample bottle completely to eliminate air from the bottle, fasten the cap firmly on the bottle. Make sure that the label is complete and taped over with clear tape, and then place the sample bottles in the PPCP water sample cooler on wet ice.
6. **Water samples collected at the pre-selected urban PPCP sites must be shipped to the EPA CINCINNATI lab ON MONDAYS THROUGH THURSDAYS. Do not send PPCP water samples to the EPA Corvallis lab. Please follow the instructions provided in the PPCP urban site water sample supply cooler.**

Please use the following special instructions for shipping PPCP water samples:

- PPCP water samples collected from the pre-selected urban river sites must be shipped on wet ice to the EPA CINCINNATI LAB within 3 days of collection (for delivery at the lab on the fourth day) using the pre-addressed FedEx airbill provided in the PPCP water sample cooler.

- There is **No Saturday, Sunday, or Federal Holiday Delivery** at the EPA CINCINNATI LAB, so PPCP water coolers must be shipped on Monday through Thursday.

- **IMPORTANT NOTE:** PPCP water samples have a holding time of 4 days. Therefore, PPCP water samples cannot be collected on Friday, held on wet ice over the weekend, and shipped on Monday or they will exceed the sample holding time.

PFC Water Chemistry Samples

The first 4 steps of the procedures for collecting PFC water samples are identical to the PPCP water sample collection procedures except that the PFC sample bottles are rinsed 3 times with water from the sampling beaker before filling them. However, there are four important differences in the remaining procedures for collecting PFC water samples: PFC samples contain 1 L of water (twice the volume of PPCP samples); water collected for PFC analysis requires HDPE bottles; PFC samples are preserved with a nitric acid solution; and PFC sample bottles are shipped in coolers at ambient temperatures with no ice. OST will provide field teams with coolers and 1 L HDPE bottles for the PFC samples, along with the labels, stickers, pre-addressed airbills, and other forms necessary for shipping the coolers. As for the PPCP water samples, water for the PFC samples will be collected using the beaker provided for collection of other water chemistry samples. Field teams will use river water from the beaker to rinse the HDPE sample bottles 3 times before filling them almost to the top. Space is left at the top of the bottle to add 5 ml of a nitric acid solution to preserve the samples. The filled HDPE water bottles are placed in the cooler with no ice and shipped at the ambient temperature within 3 days to the laboratory designated for PFC analysis. **Field teams will collect two 1 L PFC water samples at all the urban river sites that are sampled in 2009 (both boatable and wadeable urban sites that are 5th order or greater) except the repeat urban river sites. At the repeat urban river sites, field teams will collect four 1 L PFC water samples during the first site visit only.**

1. Collect the PFC water samples mid-channel at the X-site (located via GPS). Samples are taken mid-channel, at a depth of 0.5 meters or at mid-depth if the site is less than 1 meter deep.
2. Put on nitrile gloves. Avoid touching the inside of the container to prevent contamination. Make sure not to handle sunscreen or other chemical contaminants until after the sample is collected.
3. Pre-rinse the beaker with river water 3 times, discarding rinse water downstream. Hold the container so the opening faces upstream. Collect the sample at a depth of 0.5 meters below the surface, with the beaker slightly angled as you pull it to the surface.
4. Rinse each PFC sample bottle 3 times with sample water before filling the sample bottle with water from the beaker.
5. Fill the two 1 L HDPE bottles (or four 1 L HDPE bottles during the first visit at urban river repeat sites) using water from the beaker. All sample bottles should only be filled to the

top of the cylindrical portion of the bottle, leaving the shoulder and the neck empty to allow room for the preservative (5 ml of 35% nitric acid) to be added.

6. Add 5 ml of 35% nitric acid, supplied in the premeasured ampoules, into the sample, cap tightly, place an orange EP HNO_3 sticker onto the water collection bottles to indicate that the preservation agent has been added, and mix well. **Only the contents of the ampoule should be added to the sample – the opened ampoule should not be placed into the sample bottles.**
7. Make sure that the labels are complete and taped over with clear tape, and then place the sample bottles in the PFC water sample cooler. Return sample bottles to the original shipping container (coolers) and maintain at ambient temperature. **Do not cool with wet or dry ice.**
8. **Water samples collected at the pre-selected urban PFC sites must be shipped to the designated lab ON MONDAYS THROUGH THURSDAYS. Do not send PFC water samples to the EPA Corvallis lab. Please follow the instructions provided in the PFC urban site water sample supply cooler.**

Please use the following special instructions for shipping PFC water samples:

- PFC water samples collected from the pre-selected urban river sites must be shipped at ambient temperature (**without wet or dry ice**) to the designated lab within 3 days of collection (for delivery at the lab on the fourth day) using the pre-addressed FedEx airbill provided in the PFC water sample cooler.

- There is **No Saturday, Sunday, or Federal Holiday Delivery** at the designated lab, so PFC water coolers must be shipped on Monday through Thursday.

PPCP Fish Tissue

A single fish tissue composite sample will be collected at the approximately 150 designated urban river sites, **except at the repeat urban river sites where two duplicate fish tissue samples will be collected during the first site visit**. The urban river fish composite samples will provide tissue for analysis of PPCP chemicals and for analysis of the list of EMAP chemicals. **An important exception is that fish tissue samples will be collected at all urban sites that are ≥5th order and wadeable.** Field crews will use the protocols outlined in Section 5.6 of the Field Operations Manual to collect the fish tissue samples at both the boatable and wadeable urban river sites. These protocols are summarized below. Please note in step 15 that fish tissue samples collected at urban river sites are shipped directly to the EPA CINCINNATI LAB.

1. Put on clean nitrile gloves before handling the fish. Do not handle any food, drink, sunscreen, or insect repellant until after the composite sample has been collected, measured, and wrapped.
2. Rinse potential target species/individuals in ambient water to remove any foreign material from the external surface and place in clean holding containers (e.g., livewells, buckets). Return non-target fishes or small specimens to the river or stream.
3. Retain one predator species composite from each site. The composite must consist of five fish of adequate size to provide a total of 500 grams of edible tissue for analysis (refer to Table 5.6-2 for minimum species length guidelines). Select fish for each composite based on the following criteria:

- all are of the same species,
- all satisfy legal requirements of harvestable size (or weight) for the sampled river, or at least be of consumable size if no legal harvest requirements are in effect,
- all are of similar size, so that the smallest individual in a composite is no less than 75% of the total length of the largest individual, and
- all are collected at the same time, i.e., collected as close to the same time as possible, but no more than one week apart (Note: Individual fish may have to be frozen until all fish to be included in the composite are available for delivery to the designated laboratory).

Accurate taxonomic identification is essential in assuring and defining the organisms that have been composited and submitted for analysis. Under no circumstances should individuals from different species be used in a single composite sample.

4. Measure each individual fish to determine total body length. Measure total length of each specimen in millimeters, from the anterior-most part of the fish to the tip of the longest caudal fin ray (when the lobes of the caudal fin are depressed dorsoventrally).
5. Record sample number, species retained, specimen length, location collected, and sampling date and time on the Fish Collection Form (Figure 5.5-1) in black ink. Mark "URBAN" next to the site identification number at the top left of the fish form, and write primary or duplicate in the comment section. Make sure the sample identification numbers recorded on the collection form match those on the sample labels.
6. Sign and date the Fish Collection Form.
7. Remove each fish retained for analysis from the clean holding container(s) (e.g., livewell) using clean nitrile gloves. Dispatch each fish using a clean wooden bat (or equivalent wooden device).
8. Wrap each fish in extra heavy-duty aluminum foil, with the dull side in (foil provided by EPA as solvent-rinsed, oven-baked sheets).
9. Prepare a Sample Identification Label for each sample, ensuring that the label information matches the information recorded on the Fish Collection Form. **Be sure to include fish species and specimen length on each label.**
10. Cut a length of food grade tubing (provided by EPA) that is long enough to contain each individual fish and to allow extra length on each end to secure with cable ties. Place each foil-wrapped specimen into the appropriate length of tubing. Seal each end of the tubing with a plastic cable tie. Attach the fish sample label to the outside of the food-grade tubing with clear tape and secure the label by taping around the entire fish (so that tape sticks to tape).
11. Place all the wrapped fish in the composite from each site in a large plastic bag and seal with another cable tie.
12. After each sample is packaged, place it immediately on dry ice for shipment. If samples will be carried back to a laboratory or other facility to be frozen before shipment, wet ice can be used to transport wrapped and bagged fish samples in the coolers to a laboratory or other interim facility.
13. If possible, keep all (five) specimens designated for a particular composite in the same shipping container (ice chest) for transport.
14. Samples may be stored temporarily on dry ice (replenishing the dry ice daily). You have

the option, depending on site logistics, of:

- shipping the samples packed on dry ice in sufficient quantities to keep samples frozen for up to 48 hours (50 pounds are recommended), via priority overnight delivery service (e.g., Federal Express), so that they arrive at the sample preparation laboratory within less than 24 hours from the time of sample collection, or
- freezing the samples within 24 hours of collection at ≤-20°C, and storing the frozen samples until shipment within 3 weeks of sample collection (frozen samples will subsequently be packed on dry ice and shipped to the sample preparation laboratory via priority overnight delivery service).

15. **Fish Tissue samples collected at the pre-selected urban PPCP sites must be shipped to the EPA CINCINNATI lab. Do not send PPCP fish tissue samples to the GLEC lab. Please follow the instructions provided in the PPCP site fish tissue supply cooler. Be sure to include fish species and specimen lengths for all fish tissue samples on the Sample Tracking Form (Figure E-1).**

PPCP Contacts

For any questions about collecting, handling, or shipping PPCP water or fish tissue samples, please contact Leanne Stahl in the Office of Science and Technology at EPA or Blaine Snyder of Tetra Tech, Inc. using the information below.

Leanne Stahl
USEPA/OST (4305T)
1200 Pennsylvania Avenue, NW
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(202) 566-0404 (phone)
(202) 566-0409 (fax)
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Blaine Snyder
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400 Red Brook Blvd., Suite 200
Owings Mills, MD 21117
(410) 356-8993 (phone)
(410) 356-9005 (fax)
Blaine.Snyder@tetratech.com

Please use the tab button to navigate through this form.

TRACKING (BATCHED AND RETAINED SAMPLES)
National Rivers and Streams Assessment

Choose One: ☒ **BATCHED* SAMPLES** ☐ **RETAINED** SAMPLES**
Do not combine both BATCHED and RETAINED samples on the same form – please complete a separate form.

Sender:	DAVID ALTFATER	
Sender Phone:	614-836-8786	XXX-XXX-XXXX
State of Site Location:	OH	2 digit state code
Team:	1	
Date Shipped:	07/16/2008	MM/DD/YYYY
Shipped By:	☒FedEx ☐UPS ☐ Hand Delivery ☐ Other	
	If other please specify:	
Airbill:	861012765368	
For Retained Samples enter Holding Facility and Address:		Street address, City, State & Zip Code

Site ID (FW08XXXXX)	Date Collected MM/DD/YYYY	Visit	Sample ID XXXXXX.X	Sample Type	# of Jars	Comments
FW08OH033	07/07/2008	1	524309.1	FTIS	1	SMALLMOUTH BASS 400 MM
FW08OH033	07/07/2008	1	524309.2	FTIS	1	SMALLMOUTH BASS 330 MM
FW08OH033	07/07/2008	1	524309.3	FTIS	1	SMALLMOUTH BASS 298 MM
FW08OH012	07/15/2008	1	522039.1	FTIS	1	CHANNEL CATFISH 482 MM
FW08OH012	07/15/2008	1	522039.2	FTIS	1	CHANNEL CATFISH 458 MM
FW08OH012	07/15/2008	1	522039.3	FTIS	1	CHANNEL CATFISH 460 MM
FW08OH012	07/15/2008	1	522039.4	FTIS	1	CHANNEL CATFISH 498 MM
FW08OH012	07/15/2008	1	522039.5	FTIS	1	CHANNEL CATFISH 430 MM
FW08		1		BERW		
FW08		1		BERW		
FW08		1		BERW		
FW08		1		BERW		
FW08		1		BERW		
FW08		1		BERW		
FW08		1		BERW		
FW08		1		BERW		

Shipped to Lab	Sample Types	Tracking Assistance	Completed Forms
☐ MED - DULUTH	**PRESERVED - RETAINED**	Tracking Help:	Send completed electronic tracking forms to:
☒ NERL	BERW – Benthos Reach Wide	Marlys Cappaert	sampletracking@epa.gov
☐ FISH TISSUE LAB	BELG – Benthos Low Gradient	Ph: 541-754-4467	
☐ PERIPHYTON LAB	VERT – Fish Vouchers		OR you can fax a printed copy to 541-754-4637
☐ BENTHIC LAB	PERI - Periphyton ID (.1)	Electronic Form Help:	OR call info to 541-754-4663
☐ FISH MUSEUM	**UNPRESERVED - BATCHED**	Michelle Gover	
☐ OTHER (list below)	SEDE – Sediment Enzyme	Ph: 541-754-4793	
	FTIS – Fish Tissue		
	PAPA – Periphyton APA (.4)		
	ENTE - Enterococci		

Save file as BR_SITE ID_Date Collected. For Site ID & Date Collected use the first Site listed. For example, if you are batching samples for multiple sites with the first ID listed as FW08OR123 collected on 05/05/2008, then the file name would be BR_FW08OR123_05_05_08.

*BATCHED - samples that will be batched and shipped within 2 weeks. Send sample information when SHIPPED.
**RETAINED - samples that will be stored longer than a month at a holding facility. Send sample information when COLLECTED, and then when shipped to designated lab.

NRSA – Batch/Retain Form – 5/08/2008

Figure E-1. Example Sample Tracking Form showing fish tissue samples, fish species, and specimen lengths