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Surface Waters

Western Pilot Study:

Field Operations Manual for Wadeable Streams

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Assessment Program

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ENVIRONMENTAL MONITORING AND ASSESSMENT PROGRAM-SURFACE WATERS:

WESTERN PILOT STUDY
FIELD OPERATIONS MANUAL FOR
WADEABLE STREAMS

Edited by

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FOREWORD

The National Exposure Research Laboratory (NERL) and the National Health and Environmental Effects Research Laboratory (NHEERL) provide scientific understanding, information and assessment tools that will reduce and quantify the uncertainty in the Agency's exposure and risk assessments for all environmental stressors. Stressors include chemicals, biologicals, radiation, climate, and land and water use changes.

Research at NERL focuses on: (1) characterizing the sources of environmental stressors and the compartments of the environment in which they reside or move; (2) studying the pathways through environmental compartments that lead to exposure of receptors to stressors; (3) investigating intra- and inter compartmental stressor transfers and their transformations; and (4) studying and characterizing receptors and their activities as required to predict or measure stressor exposure. Research products from NERL provide effects researchers and risk assessors with information on stressor sources, pollutant transport and transformations and exposure, and state-of-the-science source-to-receptor predictive exposure models applicable at the appropriate temporal scales and site, watershed/regional and global scales. It also provides risk managers with receptor-back-to-source and stressor-back-to-cause analyses and evaluations of alternative mitigation, management or restoration strategies from an exposure perspective.

Ecological research at NHEERL contribute to improving hazard identification, dose-response assessments, and risk characterization at multiple spatial and temporal scales. Research products from NHEERL include improved assessment methods and improved approaches to interpreting the data acquired by these methods. Major uncertainties in assessing the effects on ecosystems resulting from exposure to environmental stressors are addressed through the development of the tools necessary for effective monitoring of ecosystems and their components, by mechanistic studies, and through modeling.

To accomplish its mission, NERL conducts fundamental and applied research designed to:

1. Characterize air, soil, surface water, sediment, and subsurface systems to evaluate spatial and temporal patterns, exposure to environmental stressors/ pollutants;
2. Identify, quantify, and predict the physical, chemical, biological and biochemical behavior of stressors, including characterization of their sources, transformations pathways and other factors that determine stressor exposure to humans and ecosystems across multiple media
3. Characterize the ecological and human receptors potentially impacted by stressors and pollutants;
4. Measure, predict, and apply data on environmental stressors to characterize exposure to humans and ecosystems;
5. Incorporate scientific understanding of environmental processes and ecosystem behavior, along with environmental exposure data, into predictive multimedia models to estimate exposure and to evaluate mitigation, restoration, prevention and management options;
6. Develop and implement receptor level exposure and dose models to provide risk assessors with better and more refined estimates of exposure and dose.
7. Develop chemical, physical, and biological measurement methods to identify and quantify environmental stressors and to characterize the environment;
8. Develop quality assurance methodologies for chemical, physical, radiological, and biological analyses;
9. Develop and apply geographical informational systems, remote sensing, photographic interpretation, information management technologies, software engineering technologies, computational chemistry, expert systems, and high performance computing to support the application of exposure and risk assessment tools;
10. Demonstrate, field test/evaluate, and transfer scientific information, measurement and quality assurance protocols, data bases, predictive exposure and risk assessment tools, and other innovative exposure assessment technologies, and provide environmental education materials to support Program Offices, Regions, State/Municipal/Tribal governments, and other Federal Agencies;
11. Provide technical support to Program Offices, Regions, State/Municipal/Tribal governments and other Federal Agencies to help in performing state-of-the-science exposure assessments of known certainty.

Research activities at NHEERL related to improving ecosystem risk assessment are designed to:

1. Develop and evaluate appropriate and meaningful indicators of ecological condition and develop associated criteria to characterize condition.
2. Develop and test approaches for monitoring frameworks that are integrated over multiple spatial and temporal scales to provide representative information about spatial extent of ecosystem resources, their current status (i.e., baseline condition) and how condition is changing through time.
3. Develop approaches to demonstrate relationships between effects on ecological condition and the relative magnitude of current stressors at multiple scales.

This field operations and methods manual represents a collaborative effort among principal investigators at NERL and NHEERL. The manual describes guidelines and standardized procedures for evaluating the biological integrity of surface waters of streams. It was developed to provide the Environmental Monitoring and Assessment Program (EMAP) with bioassessment methods for determining the status and monitoring trends of the environmental condition of freshwater streams. These bioassessment studies are carried out to assess biological criteria for the recognized beneficial uses of water, to monitor surface water quality, and to evaluate the health of the aquatic environment.

PREFACE

The Ecosystems Research Branch (ERB), Ecological Exposure Research Division, National Exposure Research Laboratory, U.S. Environmental Protection Agency - Cincinnati is responsible for field and laboratory exposure methods and ecological indicators that are used in assessing aquatic ecosystems. Research areas include the development, evaluation, validation, and standardization of Agency methods for the collection of biological field and laboratory data. These methods can be used by USEPA regional, enforcement, and research programs engaged in inland, estuarine, and marine water quality and permit compliance monitoring, and status and/or trends monitoring for the effects of impacts on aquatic organisms, including phytoplankton, zooplankton, periphyton, macrophyton, macroinvertebrates, and fish. The program addresses methods and techniques for sample collection; sample preparation; processing of structural and functional measures by using organism identification and enumeration; the measurement of biomass and benthic metabolism; the bioaccumulation and pathology of toxic substances; acute, chronic, and sediment toxicity; the computerization, analysis, and interpretation of biological data; and ecological assessments. ERB also includes field and laboratory support of the ecological biomarker research program and transfer of monitoring technology to the regions and state programs.

This document contains the EMAP-Surface Water field operations and bioassessment methods for evaluating the health and biological integrity of wadeable freshwater streams in the Western Pilot Study.

ABSTRACT

The methods and instructions for field operations presented in this manual for surveys of wadeable streams were initially developed and tested during 5 years of pilot and demonstration projects (1993 through 1997). These projects were conducted under the sponsorship of the U.S. Environmental Protection Agency and its collaborators through the Environmental Monitoring and Assessment Program (EMAP). This program focuses on evaluating ecological conditions on regional and national scales. This document describes procedures for collecting data, samples, and information about biotic assemblages, environmental measures, or attributes of indicators of stream ecosystem condition. The procedures presented in this manual were developed based on standard or accepted methods, modified as necessary to adapt them to EMAP sampling requirements for the Western Pilot Study. They are intended for use in field studies sponsored by EMAP, and related projects such as the USEPA Regional Environmental Monitoring and Assessment Program (R-EMAP), and the Temporally Integrated Monitoring of Ecosystems study (TIME). In addition to methodology, additional information on data management, safety and health, and other logistical aspects is integrated into the procedures and overall operational scenario. Procedures are described for collecting field measurement data and/or acceptable index samples for several response and stressor indicators, including water chemistry, physical habitat, benthic macroinvertebrate assemblages, aquatic vertebrate assemblages, fish tissue contaminants, and periphyton assemblages. The manual describes field implementation of these methods and the logistical foundation constructed during field projects. Flowcharts and other graphic aids provide overall summaries of specific field activities required to visit a stream site and collect data for these indicators. Tables give step-by-step protocol instructions. These figures and tables can be extracted and bound separately to make a convenient quick field reference for field teams. The manual also includes example field data forms for recording measurements and observations made in the field and sample tracking information. Checklists of all supplies and equipment needed for each field task are included to help ensure that these materials are available when required.

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ACRONYMS, ABBREVIATIONS, AND MEASUREMENT UNITS

Acronyms and Abbreviations

AFDM	Ash-free dry mass
APA	Acid/Alkaline Phosphatase Activity
BPJ	Best Professional Judgment
BOD	Biological Oxygen Demand
CENR	(White House) Committee on the Environment and Natural Resources
CFR	Code of Federal Regulations
dbh	Diameter at breast height
DC	Direct Current
DIC	Dissolved Inorganic Carbon
DLGs	Digital Line Graphs
DO	Dissolved oxygen
EERD	Ecological Exposure Research Division
EMAP	Environmental Monitoring and Assessment Program
EMAP-SW	Environmental Monitoring and Assessment Program-Surface Waters Resource Group
EMAP-WP	Environmental Monitoring and Assessment Program- Western Pilot study
EPA	U.S. Environmental Protection Agency
ERB	Ecosystems Research Branch
GPS	Global Positioning System
ID	identification
LWD	Large Woody Debris
MAHA	Mid-Atlantic Highlands Assessment
MAIA	Mid-Atlantic Integrated Assessment
NAWQA	National Water-Quality Assessment Program
NERL	National Exposure Research Laboratory
NHEERL	National Health and Environmental Effects Research Laboratory
ORD	Office of Research and Development
OSHA	Occupational Safety and Health Administration
P-Hab	physical habitat
PVC	polyvinyl chloride
QA	quality assurance
QC	quality control
RBP	(EPA) Rapid Bioassessment Protocol
R-EMAP	Regional Environmental Monitoring and Assessment Program
SL	Standard length
SOP	Standard Operating Procedure

ACRONYMS, ABBREVIATIONS, AND MEASUREMENT UNITS (CONTINUED)

Acronyms and Abbreviations (continued)

TIME	Temporally Integrated Monitoring of Ecosystems
TL	Total length
USGS	United States Geological Survey
WED	Western Ecology Division
YOY	young of year
YSI	Yellow Springs Instrument system

Measurement Units

amps	amperes
cm	centimeter
ft	foot
gal	gallon
ha	hectare
Hz	Hertz
in	inches
L	liter
m	meter
m^2	square meters
mg/L	milligram per liter
mm	millimeter
µm	micrometer
µS/cm	microsiemens per centimeter
mS/cm	millisiemens per centimeter
msec	millisecond
ppm	parts per million
psi	pounds per square inch
V	volts
VA	volt-ampere

SECTION 1
INTRODUCTION

by
James M. Lazorchak[1], Alan T. Herlihy[2], Donald J. Klemm[1], and Steven G. Paulsen[3]

This manual contains procedures for collecting samples and measurement data from various biotic and abiotic components of streams in the western United States. These procedures were initially developed and used between 1993 and 1998 in research studies of the U.S. Environmental Protection Agency's (EPA) Environmental Monitoring and Assessment Program (EMAP), and published in Lazorchak et al. (1998). The purposes of this manual are to: (1) Document the procedures used in the collection of field data and various types of samples for the EMAP Western Pilot Study (EMAP-WP) and (2) provide these procedures for use by other groups participating in EMAP-WP or implementing stream monitoring programs similar to EMAP.

These procedures are designed for use during a one-day visit by a crew of four persons to sampling sites located on smaller, wadeable streams (stream order 1 through 3, or higher for semi-arid and arid regions of the western U.S.). They were initially developed based on information gained from a workshop of academic, State, and Federal experts (Hughes, 1993), and subsequent discussions between aquatic biologists and ecologists within EMAP, with scientists of the U.S. Geological Survey National Water Quality Assessment Program (NAWQA), with biologists from the U.S. Fish & Wildlife Service, and with State and Regional biologists within EPA Region 3. EMAP staff has also sought information from various Federal and State scientists in the western U.S.

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EMAP initiated additional research activities in 1997 to develop field procedures for use in nonwadeable riverine systems. These procedures are currently still under development and will be published separately.

1.1 OVERVIEW OF EMAP-SURFACE WATERS

The U.S. EPA has designated EMAP to develop the necessary monitoring tools to determine the current status, extent, changes and trends in the condition of our nation's ecological resources on regional and national scales (U.S. EPA, 1998). The nation's ecological resources are a national heritage, as essential to the country now and in the future as they have been in the past. Data indicate that regional and international environmental problems may be endangering these essential resources. The potential threats include acid rain, ozone depletion, point and nonpoint sources of pollution, and climate change.

The tools being developed by EMAP include appropriate indicators of ecological condition, and statistical sampling designs to determine the status and extent of condition, and to detect regional-scale trends in condition. When fully implemented in a national monitoring framework, such as that being developed by the White House Committee on Environment and Natural Resources (CENR; Committee on Environment and Natural Resources, 1997), these tools will provide environmental decision makers with statistically valid interpretive reports describing the health of our nation's ecosystems (Whittier and Paulsen, 1992). Knowledge of the health of our ecosystems will give decision makers and resource managers the ability to make informed decisions, set rational priorities, and make known to the public costs, benefits, and risks of proceeding or refraining from implementing specific environmental regulatory actions. Ecological status and trend data will allow decision makers to objectively assess whether or not the nation's ecological resources are responding positively, negatively, or not at all, to existing or future regulatory programs.

The following three objectives guide EMAP research activities (U.S. EPA, 1998):

- Estimate the current status, extent, changes and trends in indicators of the condition of the nation's ecological resources on a regional basis with known confidence.
- Monitor indicators of pollutant exposure and habitat condition and seek associations between human-induced stresses and ecological condition.
- Provide periodic statistical summaries and interpretive reports on ecological status and trends to resource managers and the public.

The EMAP Surface Waters Resource Group (EMAP-SW) is charged with developing the appropriate tools to assess the health of lakes, streams, and wetlands in the United States. The first phase of the program started with a study of northeastern lakes between 1991 and 1996 (Larsen and Christie, 1993; Baker et al., 1997). In 1992 and 1993, a pilot study of wetland ecosystems was conducted in the Prairie Pothole region of the northern plains region of the U.S. (Peterson et al., 1997). The specific research studies dealing with streams are described in more detail in the following section.

1.2 STREAM SAMPLING COMPONENTS OF EMAP-SURFACE WATERS

The procedures presented in this manual were developed and refined during several different research projects conducted between 1993 and 1997. These projects represent two types of field activities to be performed prior to full-scale implementation of a monitoring program that addresses EMAP objectives. *Pilot projects* are intended to answer questions about proposed ecological indicators, such as plot design (how to obtain representative samples and data from each stream site), responsiveness to various stressors, evaluation of alternative methods, and logistical constraints. Pilot studies are not primarily intended to provide regional estimates of condition, but may provide these estimates for a few indicators.

Demonstration projects are conducted at larger geographic scales, and may be designed to answer many of the same questions as pilot studies. Additional objectives of these larger studies are related to characterizing spatial and temporal variability of ecological indicators, and to demonstrating the ability of a suite of ecological indicators to estimate the condition of regional populations of aquatic resources.

1.2.1 Mid-Atlantic Highlands Assessment Project

The stream sampling component of EMAP-SW was initiated in 1993 in the mid-Appalachian region of the eastern United States, in conjunction with a Regional-EMAP (R-EMAP) project being conducted by EPA Region 3. This R-EMAP study was known as the Mid-Atlantic Highlands Assessment study (MAHA), and was carried out over a 4-year period. The MAHA project was designed to test the EMAP approach in a few of the most heavily impacted ecoregions of Region 3, the mid-Appalachians, the Ridge and Valley, the Central Appalachians, the Piedmont and some of the Coastal Plain.

The Region 3 R-EMAP project was designed to answer the following questions:

- What are biological reference conditions for the Central Appalachian Ridge and Valley Ecoregion?
- Do biological communities differ between subregions?
- What is the status of mid-Atlantic Highlands stream biota?
- Can linkages be established between impairment and possible causes of impairment?
- How can an EMAP-like approach be used to design programs to restore and manage stream resources on a regional scale?

During the MAHA study, 577 wadeable stream sites throughout EPA Region 3 (DE, MD, VA, WV, PA) and the Catskill Mts. of New York were visited and sampled using the field protocols being developed by EMAP. Streams were sampled each year during a 10-week index period from April to July by field crews from EPA, the U.S. Fish and Wildlife Service, State, and contract personnel.

1.2.2 Mid-Atlantic Integrated Assessment Program

In 1997 and 1998 the EMAP Surface Waters Program became a collaborator in the Mid-Atlantic Integrated Assessment (MAIA) project, which is attempting to produce an assessment of the condition of surface water and estuarine resources. The MAIA project represented a follow-up to the MAHA study, with an expanded geographic scope (southern New York to northern North Carolina, with more sites located in the Piedmont and Coastal Plain ecoregions) and a different index period (July-September). The first year of the MAIA study, approximately 200 sites (150 wadeable sites, 13 repeated wadeable sites, and approximately 30 riverine sites) were visited for sampling.

1.2.3 Temporal Integrated Monitoring of Ecosystems Project

A special interest component of EMAP-SW is the Temporal Integrated Monitoring of Ecosystems Project (TIME). The purpose of the TIME project is to assess the changes and trends in chemical condition in acid-sensitive surface waters (lakes and streams) of the northeastern and eastern U.S. resulting from changes in acidic deposition caused by the 1990 Clean Air Act Amendments. The TIME project has three goals (Stoddard, 1990):

- Monitor current status and trends in chemical indicators of acidification in acid-sensitive regions of the U.S.
- Relate changes in deposition to changes in surface water conditions.

- Assess the effectiveness of the Clean Air Act emissions reductions in improving the acid/base status of surface waters.

1.2.4 Other Projects

The basic procedures and methods presented in this manual have also been used in other areas of the U.S. as part of R-EMAP projects being conducted by other EPA Regions. These include Regions 7 (central U.S.), 8 (Colorado), 9 (California), and 10 (Oregon and Washington). Each of these projects have modified the basic procedures to be compatible with the geographic region or other project-specific requirements.

1.2.5 Western Pilot Study

The second major geographic study within EMAP is targeted for the states and tribal nations in the western conterminous U.S. Details regarding this research initiative can be found in the peer-reviewed research plan (U.S. EPA, 2000). The purpose for this western study is to further advance the science of monitoring and to demonstrate the application of core tools from EMAP in monitoring and assessment across the West. The Western Geographic Study will serve to advance both the science of monitoring and the application of monitoring to policy, provide an opportunity to push the science and its application to new levels, both in terms of the type of systems addressed (mountainous and arid systems) and the size of the region covered (essentially one third of the conterminous U.S), and demonstrate the application of EMAP designs in answering the urgent and practical assessment questions facing the western EPA Regional Offices, while framing these unique studies in a methodology that can be extended to the entire nation.

The primary objectives of the Western Pilot Study (EMAP-WP), the surface waters component of the Western Geographic Study are to:

- Develop the monitoring tools (biological indicators, stream survey design, estimates of reference condition) necessary to produce unbiased estimates of the ecological condition of surface waters across a large geographic area (or areas) of the West; and
- Demonstrate those tools in a large-scale assessment.

The goal of EMAP-WP is to provide answers to three general assessment questions:

1. What proportion of stream and river miles in the western U.S. are in acceptable (or poor) biological condition?

2. What is the relative importance of potential stressors (habitat modification, sedimentation, nutrients, temperature, grazing, timber harvest, etc.) in streams and rivers across the West?; and
3. With what stressors are streams and rivers in poor biological condition associated?

The resource population of interest for EMAP-WP are all perennial streams and rivers as represented in EPA's River Reach File (RF3), with the exception of the "Great Rivers" (the Columbia, Snake, Colorado and Missouri Rivers). The pilot study will utilize an EMAP probability design to select sites which are statistically representative of the resource population of interest. This will allow one to extrapolate ecological results from the sites sampled to the entire population. A comprehensive set of ecological indicators (see below) will be implemented in a coarse survey of streams and rivers across all of the West (the conterminous portions of EPA Regions 8, 9 and 10), as well as in several more spatially-intensive "focus areas" in each Region (see Figure 1-1). Sample sizes (i.e., numbers of stream sites) have been chosen to allow eventual estimates of condition to be made for each state, each Regional focus area, numerous aggregated ecological regions (e.g., mountainous areas of the Pacific states, the Southern Basin and Range, etc.), major river basins, and many other potential geographic classifications.

1.3 SUMMARY OF ECOLOGICAL INDICATORS

The following sections describe the rationale for each of the ecological indicators currently included in the stream sampling procedures presented in this manual. Evaluation activities to determine the suitability of individual indicators to robustly determine ecological condition are ongoing at this time. This information is presented to help users understand the various field procedures and the significance of certain aspects of the methodologies.

Currently, EMAP considers two principal types of indicators, condition and stressor (U.S. EPA, 1998). Condition indicators are biotic or abiotic characteristics of an ecosystem that can provide an estimate of the condition of an ecological resource with respect to some environmental value, such as biotic integrity. Stressor indicators are characteristics that are expected to change the condition of a resource if the intensity or magnitude is altered.

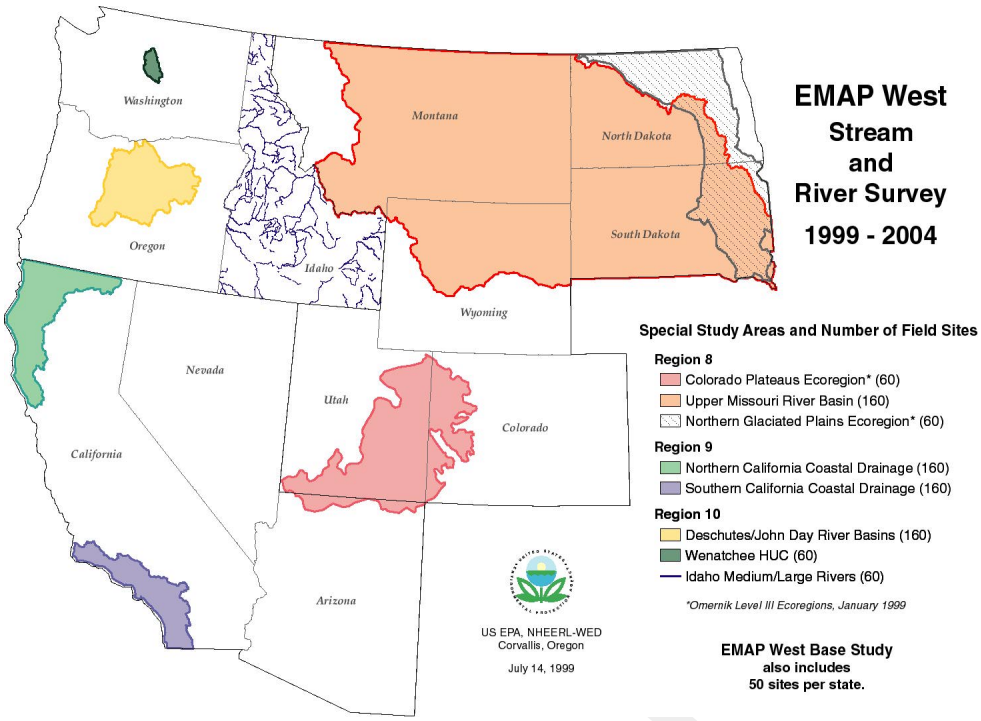

Figure 1-1. The geographic scope of the EMAP-Surface Waters Western Pilot Study, including the "special interest" study areas within each EPA Region.

1.3.1 Water Chemistry

Data are collected from each stream for a variety of physical and chemical constituents. Information from these analyses is used to evaluate stream condition with respect to stressors such as acidic deposition (of importance to the TIME project), nutrient enrichment, and other inorganic contaminants. In addition, streams can be classified with respect to water chemistry type, water clarity, mass balance budgets of constituents, temperature regime, and presence of anoxic conditions.

1.3.2 Physical Habitat

Naturally occurring differences among surface waters in physical habitat structure and associated hydraulic characteristics contributes to much of the observed variation in species composition and abundance within a zoogeographic province. The structural complexity of aquatic habitats provides the variety of physical and chemical conditions to

support diverse biotic assemblages and maintain long-term stability. Anthropogenic alterations of riparian areas and stream channels, wetland drainage, grazing and agricultural practices, and stream bank modifications such as revetments or development, generally act to reduce the complexity of aquatic habitat and result in a loss of species and ecosystem degradation.

Stressor indicators derived from data collected about physical habitat quality will be used to help explain or diagnose stream condition relative to various condition indicators. Important attributes of physical habitat in streams are channel dimensions, gradient, substrate characteristics; habitat complexity and cover; riparian vegetation cover and structure; disturbance due to human activity, and channel-riparian interaction (Kaufmann, 1993). Overall objectives for this indicator are to develop quantitative and reproducible indices, using both multivariate and multimetric approaches, to classify streams and to monitor biologically relevant changes in habitat quality and intensity of disturbance. Kaufmann et al. (1998) discuss procedures for reducing EMAP field habitat measurements and observations to metrics that describe channel and riparian habitat at the reach scale.

1.3.3 Periphyton Assemblage

Periphyton are the algae, fungi, bacteria, and protozoa associated with substrates in aquatic habitats. These organisms exhibit high diversity and are a major component in energy flow and nutrient cycling in aquatic ecosystems. Many characteristics of periphyton community structure and function can be used to develop indicators of ecological conditions in streams (Hill et al., 1999). Periphyton are sensitive to many environmental conditions, which can be detected by changes in species composition, cell density, ash free dry mass (AFDM), chlorophyll, and enzyme activity (e.g., alkaline and acid phosphatase). Each of these characteristics may be used, singly or in concert, to assess condition with respect to societal values such as biological integrity and trophic condition.

A hierarchical framework is being used in the development of the periphyton indices of stream condition. The framework involves the calculation of composite indices for biotic integrity, ecological sustainability, and trophic condition. The composite indices will be calculated from measured or derived first-order and second-order indices. The first-order indices include species composition (richness, diversity), cell density, AFDM, chlorophyll, and enzyme activity (e.g., Saylor et al., 1979), which individually are indicators of ecological condition in streams. Second-order indices will be calculated from periphyton characteristics, such as the autotrophic index (Weber, 1973), community similarity compared to refer-

ence sites, and autecological indices (e.g., Lowe, 1974; Lange-Bertalot, 1979; Charles, 1985; Dixit et al, 1992).

1.3.4 Benthic Macroinvertebrate Assemblage

Benthic macroinvertebrates inhabit the sediment or live on the bottom substrates of streams. The macroinvertebrate assemblages in streams reflect overall biological integrity of the benthic community , and monitoring these assemblages is useful in assessing the status of the water body and discerning trends. Benthic communities respond differently to a wide array of stressors. As a result of this, it is often possible to determine the type of stress that has affected a benthic macroinvertebrate community (Plafkin et al., 1989; Klemm et al., 1990; Barbour et al. 1999). Because many macroinvertebrates have relatively long life cycles of a year or more and are relatively immobile, macroinvertebrate community structure is a function of past conditions.

Two different approaches are currently being evaluated to developing ecological indicators based on benthic invertebrate assemblages. The first is a multimetric approach, where different structural and functional attributes of the assemblage are characterized as "metrics". Individual metrics that respond to different types of stressors are scored against expectations under conditions of minimal human disturbance. The individual metric scores are then summed into an overall index value that is used to judge the overall level of impairment of an individual stream reach. Examples of multimetric indices based on benthic invertebrate assemblages include Kerans and Karr (1993), Fore et al. (1996) and Barbour et al. (1995; 1996).

The second approach being investigated is to develop indicators of condition based on multivariate analysis of benthic assemblages and associated abiotic variables. Examples of this type of approach as applied to benthic invertebrate assemblages include RIVPACS (Wright, 1995), and BEAST (Reynoldson et al., 1995). Rosenberg and Resh (1993) present various approaches to biological monitoring using benthic invertebrates, and Norris (1995) briefly summarizes and discusses approaches to analyzing benthic macroinvertebrate community data.

1.3.5 Aquatic Vertebrate Assemblages

Aquatic vertebrate assemblages of interest to EMAP include fish and amphibians. The fish assemblage represents a critical component of biological integrity from both an ecosystem function and a public interest perspective. Historically, fish assemblages have

been used for biological monitoring in streams more often than in lakes (e.g., Plafkin et al., 1989; Karr, 1991). Fish assemblages can serve as good indicators of ecological conditions because fish are long-lived and mobile, forage at different trophic levels, integrate effects of lower trophic levels, and are reasonably easy to identify in the field (Plafkin et al., 1989). Amphibians comprise a substantial portion of vertebrate biomass in streams of many areas of the U.S. (Hairston, 1987; Bury et al., 1991). Reports of dramatic declines in amphibian biodiversity (e.g., Blaustein and Wake, 1990; Phillips, 1990) has increased the level of interest in monitoring these assemblages. Amphibians may also provide more information about ecosystem condition in headwater or intermittent streams in certain areas of the country than other biological response indicators (Hughes, 1993). The objective of field sampling is to collect a representative sample of the aquatic vertebrate assemblage by methods designed to 1) collect all except very rare species in the assemblage and 2) provide a measure of the abundance of species in the assemblages (McCormick, 1993). Information collected for EMAP that is related to vertebrate assemblages in streams includes assemblage attributes (e.g., species composition and relative abundance) and the incidence of external pathological conditions.

Indicators based on vertebrate assemblages are being developed primarily using the multimetric approach described in Section 1.3.5 for benthic macroinvertebrates, and originally conceived by Karr and others (Karr et al., 1986). Simon and Lyons (1995) provide a recent review of multimetric indicators as applied to stream fish assemblages. (McCormick et al. (In press) provide an example of a multimetric indicator developed for the Mid-Atlantic region using EMAP data, based on an evaluation process described by Hughes et al. (1998).

1.3.6 Fish Tissue Contaminants

Indicators of fish tissue contaminants attempt to provide measures of bioaccumulation of toxic chemicals in fish. The primary purpose of determining contaminant levels in fish tissue is to provide a measure of the potential exposure of stream systems to toxic compounds. It is also meant to be used in conjunction with the other stressor indicators (physical habitat, water chemistry, land use, population density, other records of relevant anthropogenic stresses) and condition indicators (fish, macroinvertebrates, periphyton) to help diagnose whether the probable cause of stream degradation, when it is shown by the condition indicators to occur, is water quality, physical habitat, or both.

The various studies that have been done on fish tissue contaminants have focused on different parts of the fish: whole fish, fillets, livers. For EMAP-SW, the focus is on

whole fish because of the emphasis on the ecological health of the whole stream (as opposed to a focus on human health concerns). Whole fish are a better indicator of risk to piscivorous wildlife than fillets. It is hoped to also be able to say something about risks to human health by analyzing whole fish. Whole fish also present fewer logistical problems for field crews (no gutting required in the field) and the analytical lab (no filleting necessary).

Samples are prepared for two major categories of fish species. One sample is prepared using a species whose adults are small (e.g., small minnows, sculpins, or darters). The second sample is prepared using a species whose adults are of larger size (e.g., suckers, bass, trout, sunfish, carp). In addition to being more ubiquitous than the larger fish (and therefore more likely to be present in sufficient numbers to composite), small fish have other advantages over large fish. Most importantly, it may be possible to get a more representative sample of the contaminant load in that stream segment (although it could be at a lower level of bioaccumulation) by creating a composite sample from a larger number of small individuals than by compositing a few individuals of larger species. The major advantage that larger fish could potentially offer, whether predators (piscivores) or bottom feeders, is a higher level of bioaccumulation and thus greater sensitivity to detect contaminants. The relative bioaccumulation of contaminants by large and small stream fish is not known, thus the reason for preparing two samples in this study.

In addition, specimens are collected for determination of the presence of various internal pathogens..

1.4 OBJECTIVES AND SCOPE OF THE FIELD OPERATIONS MANUAL

Only field-related sampling and data collection activities are presented in this manual. Laboratory procedures and methods (including sample processing and analytical methods) associated with each ecological indicator are summarized in Chaloud and Peck (1994); detailed procedures will be published as a separate document.

This manual is organized to follow the sequence of field activities during the 1-day site visit. Section 2 presents a general overview of all field activities. Section 3 presents those procedures that are conducted at a “base” location before and after a stream site visit. Section 4 presents the procedures for verifying the site location and defining a reach of the stream where subsequent sampling and data collection activities are conducted. Sections 5 through 14 describes the procedures for collecting samples and field measurement data for various condition and stressor indicators. Specific procedures associated with each indicator are presented in standalone tables that can be copied, laminated, and taken

into the field for quick reference. Section 15 describes the final activities that are conducted before leaving a stream site. Appendix A contains a list of all equipment and supplies required by a crew to complete all field activities at a stream.

Depending on the specific project and approach to information management, field teams may also be provided with an information management handbook that contains instructions for tracking samples and generating sampling status reports as well as using the computers and associated hardware and software. Field teams are also required to keep the field operations and methods manual available in the field for reference and to address questions pertaining to protocols that might arise.

1.5 QUALITY ASSURANCE

Large-scale and/or long-term monitoring programs such as those envisioned for EMAP require a rigorous quality assurance (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 (Stanley and Verner, 1986). Field teams should be provided a copy of the QA project plan (e.g., Chaloud and Peck, 1994 for EMAP-SW activities). The QA plan 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. A QA project plan will be prepared for the Western Pilot Study and distributed to all participants.

Quality control (QC) activities associated with field operations are integrated into the field procedures. Important QA activities associated with field operations include a comprehensive training program that includes practice sampling visits, and the use of a qualified museum facility or laboratory to confirm any field identifications of biological specimens. The overall sampling design for EMAP-SW related studies usually includes a subset of sites (10 to 15 percent) that are revisited within a single sampling period and/or across years (e.g., Larsen, 1997; Urquhart et al., 1998). Information from these repeat visits is used in part to describe overall sampling and measurement precision for the various ecological indicators.

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NOTES

NOTES

SECTION 2
OVERVIEW OF FIELD OPERATIONS

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This section presents a general overview of the activities a 4-person field team conducts during a typical one-day sampling visit to a stream site. General guidelines for recording data and using standardized field data forms and sample labels are also presented. Finally, safety and health considerations and guidelines related to field operations are provided.

2.1 DAILY OPERATIONAL SCENARIO

The field team is divided into two groups, termed the "Geomorphs" and the "Biomorphs", that reflect their initial responsibilities more than their expertise. The geomorphs are primarily responsible for conducting the intensive physical habitat characterization. The biomorphs are primarily responsible for collecting biological samples. Table 2-1 provides the estimated time required to conduct various field activities. Figure 2-1 presents one scenario of the general sequence of activities conducted at each stream reach. For some wide, shallow streams, the required reach length and/or the larger area requiring sampling effort may necessitate two days be allocated for completing all required activities.

Upon arrival at a stream site, the geomorphs are responsible for verifying and documenting the site location, determining the length of stream reach to be sampled, and establishing the required transects (Section 4). The biomorphs collect samples and field measurements for water chemistry (Section 5) and determine stream discharge (Section 6). The biomorphs also collect periphyton and benthos samples (Sections 8 and 11, respectively). The geomorphs conduct the intensive physical habitat characterization (Section 7).

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TABLE 2-1. ESTIMATED TIMES AND DIVISION OF LABOR FOR FIELD ACTIVITIES

Activity	Group	Est. Time Required
Site verification and establishing sampling reach and transects	Geomorphs (2 persons)	2 hr
Water chemistry sampling and stream discharge determination	Biomorphs (2 persons)	1 hr
Collecting and processing benthos and periphyton samples	Biomorphs (2 persons)	3 hr
Intensive physical habitat characterization, including legacy tree identification and presence of invasive plant taxa	Geomorphs (2 persons)	2 to 3 hr
Aquatic vertebrate sampling and processing	Geomorphs and Biomorphs (4 persons)	2 to 5 hr
Rapid habitat assessment Visual stream assessment	Biomorphs (2 persons)	0.5 hr
Sample tracking and packing	Geomorphs (2 persons)	1 hr
SUMMARY	28 to 32 person-hours	7 to 8 hrs per team[a]

[a] For wider wadeable streams (e.g., > 20 m, it may require more than 1 day to complete all required activities.

Both groups are involved with collecting aquatic vertebrates (Section 12) and preparing fish samples for analysis of toxic contaminants (Section 13). Finally, the biomorphs conduct a habitat characterization based on the Rapid Bioassessment Protocols (RBP; Plafkin et al., 1989; Barbour et al., 1999) and a visual stream assessment (Section 14), while the geomorphs prepare samples for transport and shipment (Section 3).

2.2 GUIDELINES FOR RECORDING DATA AND INFORMATION

During the one-day visit to a stream, a field team is required to obtain and record a substantial amount of data and other information for all of the various ecological indicators described in Section 1.3. In addition, all the associated information for each sample collected must be recorded on labels and field data forms to ensure accurate tracking and subsequent linkage of other data with the results of sample analyses.

The field data forms to be used for the Western Pilot Study are designed to be compatible with an optical scanner system to allow rapid conversion of the printed form into

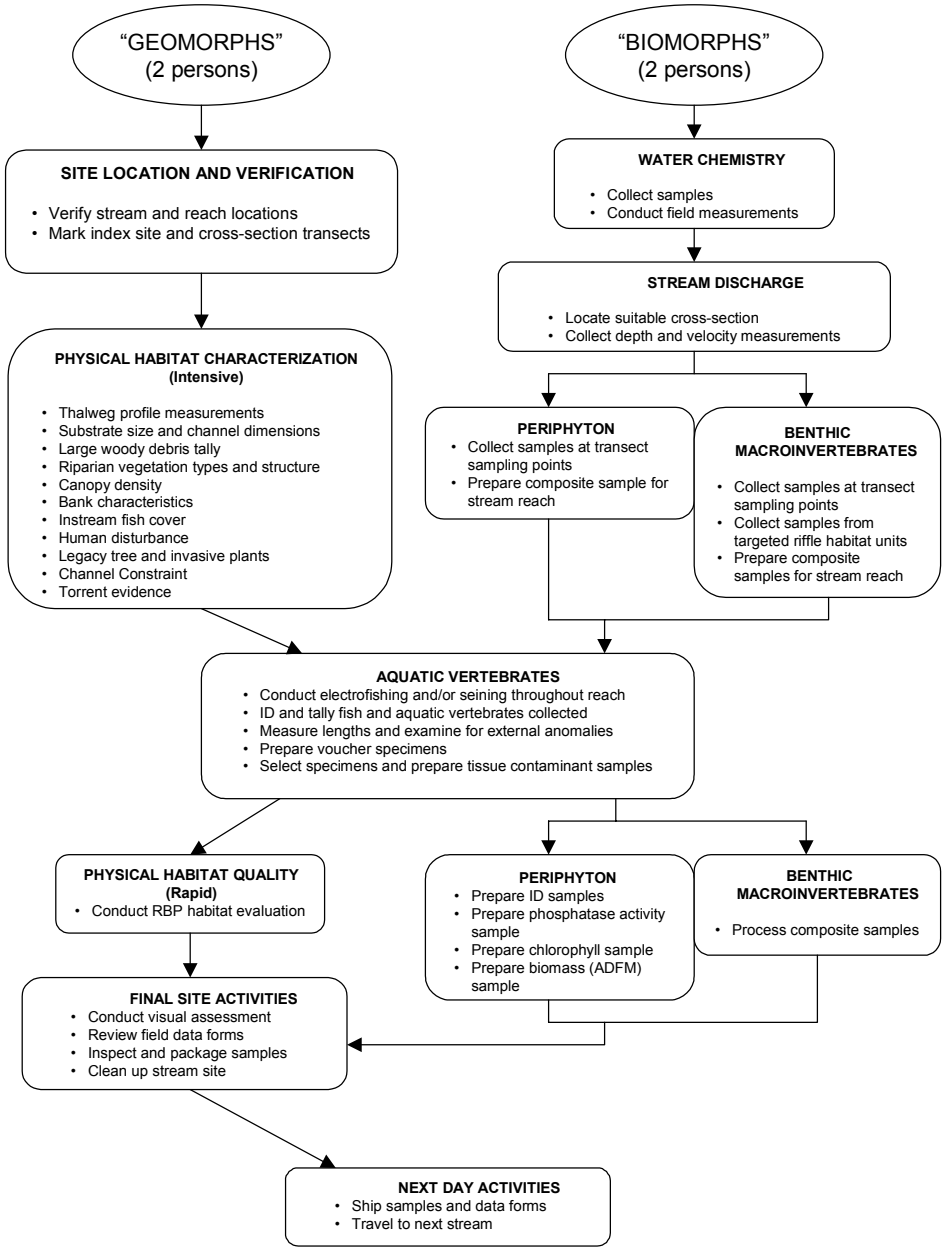


Figure 2-1. General sequence of stream sampling activities (modified from Chaloud and Peck, 1994).

one or more electronic files and reduce the need for manual data entry. While these forms should facilitate data recording by the field crew, it is imperative that field and sample information be recorded accurately, consistently, and legibly. Measurement data that cannot be accurately interpreted by others besides the field teams, and/or samples with incorrect or illegible information associated with them, are lost to the program. The cost of a sampling visit coupled with the short index period severely limits the ability to re-sample a stream because the initial information recorded was inaccurate or illegible. Some guidelines to assist field personnel with recording information are presented in Table 2-2. Examples of completed data forms and labels are presented in the sections describing field sampling and measurement procedures for different indicators, and a complete set of blank field data forms are included as Appendix C.

2.3 SAFETY AND HEALTH

Collection and analysis of samples (e.g., benthic invertebrates, fish, periphyton, sediment) can involve significant risks to personal safety and health (drowning, electrical shock, pathogens, etc.). While safety is often not considered an integral part of field sampling routines, personnel must be aware of unsafe working conditions, hazards connected with the operation of sampling gear, boats, and other risks (Berry et al., 1983). Personnel safety and health are of the highest priority for all investigative activities and must be emphasized in safety and health plans for field, laboratory, and materials handling operations. Preventive safety measures and emergency actions must be emphasized. Management should assign health and safety responsibilities and establish a program for training in safety, accident reporting, and medical and first aid treatment. Safety documents and standard operating procedures (SOPs) containing necessary and specific safety precautions should be available to all field personnel. Additional sources of information regarding field and laboratory safety related to biomonitoring studies include Berry et al. (1983), U.S. EPA (1986) and Ohio EPA (1990).

2.3.1 General Considerations

Important considerations related to field safety are presented in Table 2-3. It is the responsibility of the group safety officer or project leader to ensure that the necessary safety courses are taken by all field personnel and that all safety policies and procedures are followed. Sources of information regarding safety-related training include the American Red Cross (1989), the National Institute for Occupational Safety and Health (1981), U.S. Coast Guard (1987) and Ohio EPA (1990).

TABLE 2-2. GUIDELINES FOR RECORDING FIELD DATA AND OTHER INFORMATION

Field Measurements:

Data Recording:

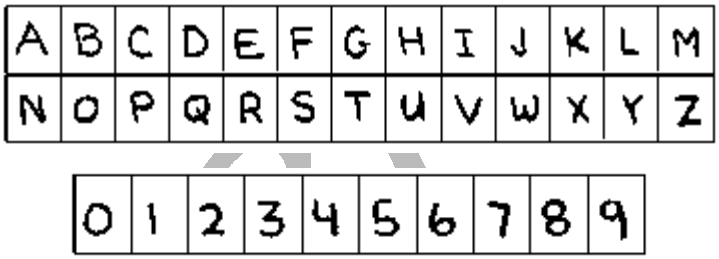
Record measurement values and/or observations on data forms preprinted on water-resistant paper.

Headers on the second pages of all forms link the data. Fill in all headers of all pages or data will be lost or linked to the wrong site record(this is a good one to review at the end of the day).

NEVER EVER mark on or around the cornerblocks or ID Box (the squares in the corners and the funky box with the number over it.) These markings are crucial to the scanning software and changing them in any way will affect performance.

Write legibly. Use a dark pencil lead that is at least a No. 2 for softness (HB), or use a dark pen. Your writing must be dark enough to be picked up by the scanner. Erase mistakes completely and write the correct value whenever you can. If you must line out an incorrect value, place the correct value nearby in the appropriate box so the data entry operator can easily find it.

Use all caps when filling in the name fields on the forms. Clearly distinguish letters from numbers (e.g., 0 versus O, 2 versus Z, 7 versus T or F, etc.). Do not put lines through 7's, 0's, or Z's. Do not use slashes. Below Here are examples of lettering that are readable by the scanning software:



It is not necessary to write in all caps in the long comments sections on the Stream Verification and Stream Assessment forms, but write legibly (because the data entry operators still need to read it to type it in.) Avoid marginal notes, etc. Be concise, but avoid using abbreviations and/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.

When you need to circle a choice, make a medium-sized circle around your choice.

(Yes) **No** **1** (2) **3** **4**

(continued)

TABLE 2-2 (continued)

Record data and information so that all entries are obvious. Enter data completely in every field that you use. Follow the "comb" guidelines--print each number or letter in the individual space provided. Keep letters and numerals from overlapping. Record data to the number of decimal places provided on the forms. Illegible information is equivalent to no information.

If the measurement for a field is zero, enter zero. If left blank, it will be recorded as missing data. (There are parts of forms that are left blank when they are not being used. A typical example is page two of the field measurement form. Usually only one type of velocity and discharge information is taken and the unused areas of the form are left blank).

If the field calls for meters, write the answer in meters. Do not fill in a number and put (cm) for units. The same goes for adding decimal places (we just end up doing the rounding for you. If you have a negative reading for velocity on the Stream Discharge section, write the number and flag it as negative in the comments section.

	Dist. from Bank	Velocity	Depth	Flag
1	0	0	0	
2	10	0.1	0.6	F1
3	20	0.8	1.0	
4	30	1.3	1.3	

F1 Stream velocity negative

Record information on each line, even if it has to be recorded repeatedly on a series of lines (e.g., fish names or species codes, physical habitat characteristics). "Ditto" marks (") can be used if necessary and if they are clearly distinguishable from letters or numbers. DO NOT USE a straight vertical line to indicate repeated entries.

Data Qualifiers (Flags):

Use only defined flag codes from the list below and record on data form in appropriate field. If the information is important enough to write on the page, use an "Fn" flag and put it in the comment section. If you have been instructed to collect a piece of information for which there is no space on the form, choose a flag and comment section, and use them consistently.

FLAG	COMMENT
F1, F2, etc.	Miscellaneous comments assigned by Field crew (e.g. Fish dead)
K	Sample not collected; No measurement or observation made
U	Suspect sample, measurement or observation
Q	Unacceptable QC check associated with measurement
Z	Last station sampled before next transect

(continued)

TABLE 2-2 (continued)

If you cannot take a measurement, leave the measurement field blank and put the K flag in the Flag column.

	Dist. from Bank	Velocity	Depth	Flag
1	0	0	0	
2	10	0.1	0.6	F1
3	20	0.8	1.0	
4	30		1.3	K

F1 Stream velocity negative

Review of Data Forms:

Have someone who did not fill in the forms review them at the end of the day. Some information is duplicated. Sometimes, however, when one measurement is missing, as many as 100 other metrics based on that measurement are lost. Be thorough.

Example: Site_ID
Visit Date
Missing Data
Increment (on the back of the Thalweg form)

Returning the Forms

Return the originals
If you want a copy of the data, make a Xerox and keep it.
Try to keep the forms in their original order.
Do not staple the forms together.

Include a list of sites visited. Please include a list with Site ID and Visit Date for forms being returned.

Sample Labels and Tracking

Sample Labels:

Sample Labels– Use adhesive labels with preprinted ID numbers and a standard recording format for each type of sample.

Record information on labels using a fine-point indelible marker. Cover completed labels with clear tape.

(continued)

TABLE 2-2 (continued)

Sample Tracking Information:

Record sample ID number from the label and associated collection information on sample collection form. Use a dark pencil or pen.

Complete any sample tracking forms required. Include tracking forms with all sample shipments.

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; re-sampling not possible.
- U Suspect sample (e.g., possible contamination, does not meet minimum acceptability requirements, or collected using a nonstandard procedure)
- F*n* Miscellaneous flags (*n*=1, 2, etc.) assigned by a field team for a particular sample shipment.

Explain all flags in comments section on sample collection form.

Review of Labels and Collection Forms:

The field team compares information recorded on labels, sample collection forms, and tracking forms for accuracy before leaving a stream. Make sure Sample ID numbers match on all forms.

Persons using sampling devices should become familiar with the hazards involved and establish appropriate safety practices prior to using them. Individuals involved in electrofishing must be trained by a person experienced in this method or by attending a certified electrofishing training course. Reynolds (1983) and Ohio EPA (1990) provide additional information regarding electrofishing safety procedures and practices.

If boats are used to access sampling sites, personnel must consider and prepare for hazards associated with the operation of motor vehicles, boats, winches, tools, and other incidental equipment. Boat operators should 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). All boats with motors must have fire extinguishers, boat horns, life jackets or flotation cushions, and flares or communication devices.

TABLE 2-3. GENERAL HEALTH AND SAFETY CONSIDERATIONS

Training:

- First aid
- Cardiopulmonary resuscitation (CPR)
- Vehicle safety (e.g., operation of 4-wheel drive vehicles)
- Boating and water safety (if boats are required to access sites)
- Field safety (e.g., weather conditions, personal safety, orienteering, reconnaissance of sites prior to sampling
- Equipment design, operation, and maintenance
- Electrofishing safety
- Handling of chemicals and other hazardous materials

Communications

- Check-in schedule
- Sampling itinerary (vehicle used and its description, time of departure, travel route, estimated time of return)
- Contacts for police, ambulance, fire departments, search and rescue personnel
- Emergency services available near each sampling site and base location

Personal Safety

- Field clothing and other protective gear
- 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, and search and rescue personnel.

Proper field clothing should be worn to prevent hypothermia, heat exhaustion, sunstroke, drowning, or other dangers. Field personnel should be able to swim. Chest waders made of rubberized or neoprene material and suitable footwear must always be worn with a belt to prevent them from filling with water in case of a fall. The use of a life jacket is advisable at dangerous wading stations if one is not a strong swimmer because of the possibility of sliding into deep water.

Many hazards lie out of sight in the bottoms of lakes, 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.

Prior to a sampling trip, personnel should determine that all necessary equipment is in safe working condition. Good housekeeping practice should be followed in the field. These practices protect staff from injury, prevent or reduce exposure to hazardous or toxic substances, and prevent damage to equipment and subsequent down time and/or loss of valid data.

2.3.2 Safety Equipment and Facilities

Appropriate safety apparel such as waders, lab coats, gloves, safety glasses, etc. must be available and used when necessary. Bright colored caps (e.g., orange) must be available and worn during field activities. First aid kits, fire extinguishers, and blankets must be readily available in the field. A properly installed and operating fume hood must be provided in the laboratory for use when working with carcinogenic chemicals (e.g., formaldehyde, formalin) that may produce dangerous fumes. Cellular telephones or portable radios should be provided to field teams working in remote areas for use in case of an emergency. Facilities and supplies must be available for cleaning of exposed body parts that may have been contaminated by pollutants in the water. Soap and an adequate supply of clean water or ethyl alcohol, or equivalent, should be suitable for this purpose.

2.3.3 Safety Guidelines for Field Operations

General safety guidelines for field operations are presented in Table 2-4. Personnel participating in field activities on a regular or infrequent basis should be in sound physical condition and have a physical exam annually or in accordance with Regional, State, or organizational requirements. All surface waters and sediments should be considered potential health hazards due to toxic substances or pathogens. Persons must become familiar with the health hazards associated with using chemical fixing and/or preserving agents. Formaldehyde (or formalin) is highly allergenic, toxic, and dangerous to human

TABLE 2-4. GENERAL SAFETY GUIDELINES FOR FIELD OPERATIONS

- Two persons (three to four persons for electrofishing) must be present during all sample collection activities, and no one should be left alone while in the field.
- Exposure to stream 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 and when turning over rocks during hand picking.
- Use appropriate protective equipment (e.g., gloves, safety glasses) when handling and using hazardous chemicals
- Persons working in areas where poisonous snakes may be encountered 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.

  If local advice is not available and medical assistance is more than an hour away, carry a snake bite kit and be familiar with its use.
- Any person allergic to bee stings, other insect bites, or plants must take proper precautions and have any needed medications handy.
- Field personnel should also protect themselves against the bite of deer or wood ticks because of the potential risk of acquiring pathogens that cause Rocky Mountain spotted fever and Lyme disease.
- All 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°C or 50°F) if he or she is exposed to wind or becomes wet.
- Handle and dispose of chemical wastes properly. Do not dispose any chemicals in the field.

health (carcinogenic) if utilized improperly. Chemical wastes can cause various hazards 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 during any environmental incident, it is extremely important to protect the health and safety of all personnel. Take any 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 error on the side of personal safety

- Field personnel should never disturb, or even worse, retrieve improperly disposed hazardous materials from the field and bring them back to a facility for "disposal". To do so may worsen the impact to the area of the incident, may incur personal liability, may incur liability for the team members and their respective organizations, may cause personal injury, or my cause unbudgeted expenditure of time and money for proper treatment and disposal of the material. However, it is important not to ignore environmental incidents. There is a requirement to notify the proper authorities of any incident of this type. The appropriate authorities may then take the necessary actions to 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.

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NOTES

SECTION 3
BASE LOCATION ACTIVITIES

Donald J. Klemm[1], Brian H. Hill[1], Frank H. McCormick[1], David V. Peck[2], and Marlys Cappaert[3]

Field teams conduct a number of activities at a "base" location before and after visiting each stream site. These activities are generally conducted on the same day as the sampling visit. Close attention to these activities is required to ensure that the field teams know where they are going, that access to the stream site is possible and permissible, that all the necessary equipment and supplies are in good order to complete the sampling effort, and that samples are packaged and shipped correctly and promptly.

Modifications to base location procedures described in the previous EMAP-SW field operations manual for wadeable streams (Klemm et al., 1998) are summarized in Table 3-1. Conductivity pens are not used in the Western Pilot Study. Sediment samples for metabolism and sediment toxicity are not being collected for the Western Pilot Study. Performance evaluation procedures for field meters have been modified to reflect new types of instrumentation. Beginning in 2001, field measurements of conductivity and dissolved oxygen are optional, and the frequency of inspection and evaluation of field meters is reduced. In some situations, field teams may have personnel available who are certified to ship preserved biological samples that constitute dangerous goods. Such samples must be transported and presented for shipment in accordance with State, Federal, and international regulations. Because of the large geographic area being sampled for the Western Pilot, it is critical to minimize the potential for transferring exotic or nuisance species of plants and animals or waterborne pathogens such as salmonid Whirling Disease.

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TABLE 3-1. SUMMARY OF CHANGES IN BASE LOCATION ACTIVITIES FOR THE EMAP-SW WESTERN PILOT STUDY

Changes from Klemm et al. (1998):
7. Reference to conductivity pens has been removed
8. Procedures and information related to sediment metabolism and sediment toxicity sampling have been removed
9. Performance evaluation procedures for field instrumentation have been modified or added
10. Added procedures for preparing dangerous goods samples for shipment
11. Cleaning procedures and solutions to prevent interstream transfer of Whirling Disease spores have been included.
Changes from EMAP-Western Pilot Study Year 2000 activities:
1. The frequency of performance evaluation checks for field conductivity and dissolved oxygen meters is reduced from before each stream site to before and after the field season.
2. The use of ice substitute packs whenever possible to ship samples is strongly recommended to avoid problems associated with melted ice during shipment.

Figure 3-1 illustrates operations and activities that are conducted before and after each visit to a stream site. Activities that are conducted after a stream visit include equipment cleanup and maintenance, packing and shipping samples, and communications with project management to report the status of the visit.

3.1 ACTIVITIES BEFORE EACH STREAM VISIT

Before each stream visit, each field team should confirm access to the stream site, develop a sampling itinerary, inspect and repair equipment, check to make sure all supplies required for the visit are available, and prepare sample containers. Procedures to accomplish these activities are described in the following sections.

3.1.1 Confirming Site Access

Field crews should be provided with dossiers containing important locational and access information for each stream they are scheduled to visit. Before visiting a stream, the crew should review the contents of the specific stream dossier. The landowner(s) listed in the dossier should be contacted to confirm permission to sample and identify any revisions to the information contained in the dossier.

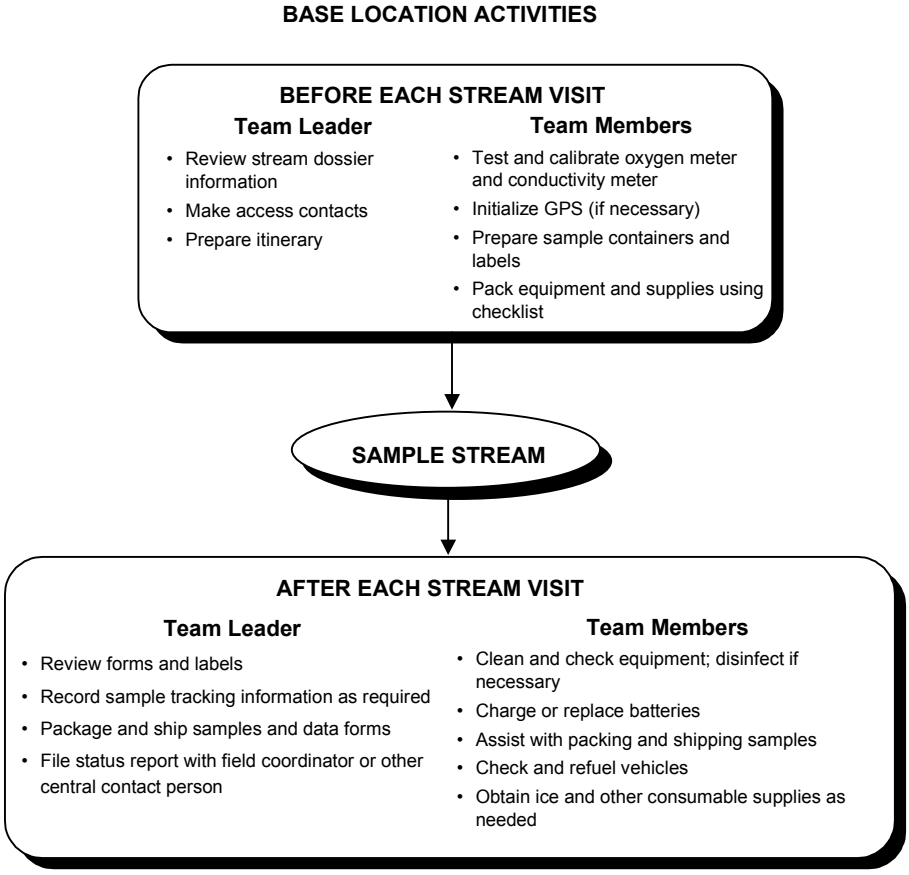

Figure 3-1. Activities conducted at base locations.

3.1.2 Daily Sampling Itinerary

Based upon the sampling schedule provided to each team, team leaders are responsible for developing daily itineraries. The team leader reviews each stream dossier to ensure that it contains the appropriate maps, contact information, copies of permission letters, and access instructions. Additional activities include determining the best access routes, calling the landowners or local contacts to confirm permission, confirming lodging plans for the upcoming evening, and coordinating rendezvous locations with individuals who must meet with field teams prior to accessing a site. This information is used to develop an itinerary for the stream. The itinerary should include anticipated departure time, routes of travel, location of any intermediate stops (e.g., to drop off samples, pick up supplies, etc.) and estimated time of arrival at the final destination after completing the stream visit. This information (and any changes that occur due to unforeseen circumstances), should be provided to the field coordinator or other central contact person identified for the specific field study. Failure to adhere to the reported itinerary can result in the initiation of expensive search and rescue procedures and disruption of carefully planned schedules. In addition, each team should carry individual emergency medical and personal information with them, possibly in the form of a "safety log" that remains in the vehicle (see Section 2).

3.1.3 Instrument Inspections and Performance Tests

Each field team is required to test and calibrate some instruments prior to departure for the stream site**.** Field instruments include a global positioning system (GPS) receiver, a current velocity meter, a conductivity meter, and a dissolved oxygen meter. **NOTE: Conductivity and dissolved oxygen are optional measurements beginning in 2001**. Backup instruments should be available if instruments fail the performance tests or calibrations described in the following subsections.

3.1.3.1 Global Positioning System Receiver–

Specific performance checks will vary among different brands of GPS receivers. Follow the instructions in the receiver's operating manual to make sure the unit is functioning properly. Turn on the receiver and check the batteries. Replace batteries immediately if a battery warning is displayed. Make sure extra batteries are stored with the receiver and will be available in the field if necessary. Follow the manufacturer's instructions for initializing the receiver when it becomes necessary (e.g., before first use, after replacing batteries, or if a new positional reference is required).

3.1.3.2 Dissolved Oxygen Meter–

NOTE: Dissolved oxygen is an optional measurement beginning in 2001.

As an initial performance test before use each year, dissolved oxygen (DO) meters should be tested for accuracy against the Winkler titration method, In addition, inspect and test the dissolved oxygen meters periodically during the course of field sampling operations. At a minimum, check the instruments before and after field sampling has been completed. The inspection and testing procedure, based on the use of a Yellow Springs Instruments (YSI) Model 53 oxygen meter, is summarized in Figure 3-2. Some modification to the procedure may be necessary for other models or types of dissolved oxygen meters. The procedure to use for newer models of DO meters (e.g., the YSI Model 85 or 95), is presented in Table 3-2.

Inspect the meter by checking the status of the batteries, and the functioning of the electronics. Confirm the meter is adjusted correctly for measurements in fresh water. Inspect the membrane of the probe. If bubbles are present, if the membrane is discolored, or if the membrane is torn, use a backup probe and/or replace the membrane on the original probe. (NOTE: For older models of meters, new membranes may require conditioning for 24 hours before use).

After inspecting the meter and probe, attempt to calibrate it using the procedure presented in Table 3-2 (for YSI Models 85 or 95), or by following the instructions in the instrument operating manual. Do not record the calibration information obtained during the performance test. The meter is calibrated again at each stream site. If the meter cannot be successfully calibrated, check the temperature probe reading against a thermometer and/or replace the membrane, probe, or meter (if spare units are available). After the test, turn the meter off, and store the probe according to the manufacturer's instructions.

3.1.3.3 Conductivity Meters–

NOTE: Conductivity is an optional measurement beginning in 2001.

Follow the operating manual provided with the meter to check the batteries, the electronics, and to inspect the probe. New probes or probes that have been stored dry may require conditioning before use.

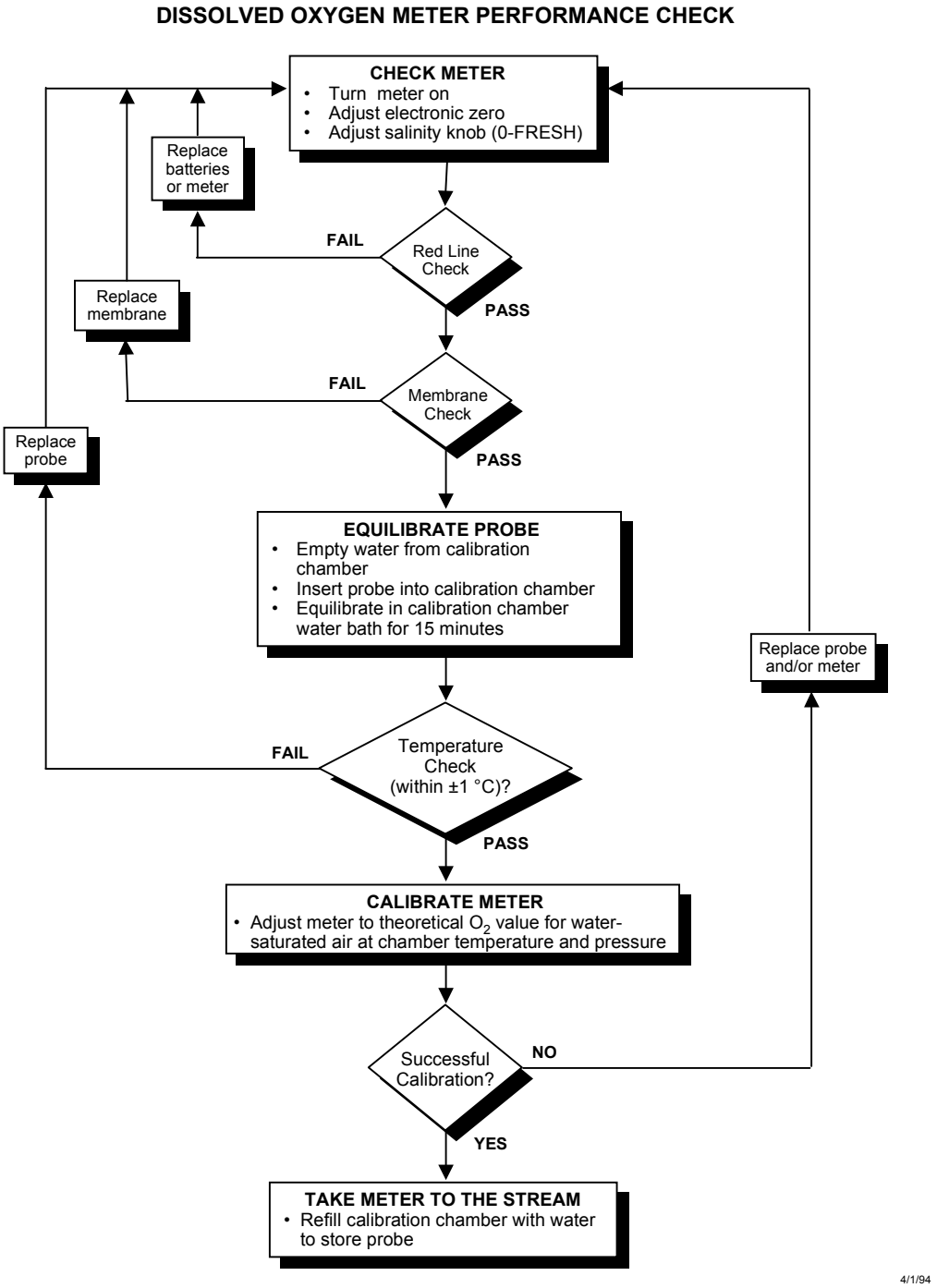

Figure 3-2. Performance test procedure for a dissolved oxygen meter.

TABLE 3-2. CHECKING THE CALIBRATION OF THE DISSOLVED OXYGEN METER[a]

Note: Beginning in 2001, dissolved oxygen is an optional measurement.

1. Periodically, check the temperature probe of the meter against a field thermometer. This can be done in a bucket of water at a base location or at a stream site. The displayed temperature should be within ± 1 EC of the thermometer reading.

2. At each location, obtain the approximate local altitude from a topographic map or other source (e.g., local airport).

3. Inspect the DO probe membrane for wrinkles, cracks, bubbles, etc. Replace the membrane cap assembly if necessary.

4. Check the calibration chamber and fill it with cold tap water to dampen the sponge. Drain the chamber and insert the probe into the chamber.

5. Turn the meter on and make sure the meter passes all the internal electronics checks.

6. Press the MODE key until the dissolved oxygen reading inside the chamber is displayed in mg/L. Allow approximately 15 minutes for the readings to stabilize (i.e., a change of < 0.02 mg/L over a 1-minute period).

7. Press the UP ARROW and DOWN ARROW keys simultaneously to enter calibration mode.

8. Use the UP ARROW or DOWN ARROW key to enter the local altitude [to the nearest 100 feet (e.g., “15" equals 1500 ft)]. After the correct altitude is displayed, press the ENTER button.

9. In the lower part of the display, “CAL” should appear along with the theoretical value based on temperature and altitude.

10. Once the actual value displayed is stable, compare the actual and theoretical values. They should agree ± 0.5 mg/L. If not, check the temperature probe against a thermometer (Step 1), or install a new membrane cap assembly, then repeat the calibration procedure.

[a] For use with YSI Models 85 and 95. Modified from YSI Incorporated. 1986. *Model 85 Handheld Oxygen, Conductivity, Salinity, and Temperature System Operations Manual.* YSI Incorporated, Yellow Springs, OH.

The operation of the conductivity meter is checked periodically at a base location using a standard solution of known conductivity. A quality control check sample (QCCS) is prepared as described in Table 3-3. The QCCS can be prepared as either of two dilutions of the stock standard, depending on the theoretical conductivity desired based on the anticipated range of conductivities in the field. A 1:100 dilution of the stock provides a QCCS with a conductivity of 75.3 µS/cm at 25 °C (Metcalf and Peck, 1993). A 1:200 dilution results in a QCCS with a conductivity of 37.8 µS/cm at 25 °C (Peck and Metcalf, 1991). A fresh lot of the QCCS should be prepared every two weeks from the stock standard solution. For higher conductivity systems, a 0.01 N potassium chloride solution is used as a QCCS (theoretical value = 1,413 µS/cm at 25 °C).

If a YSI Model 85 meter is being used, check the performance of the conductivity pen or conductivity meter by following the procedure presented in Table 3-4. Make sure the correct mode (temperature compensated conductivity) is used for the check. The displayed value of the QCCS should be compared directly to the theoretical value of the QCCS at 25 EC (75.3 µS/cm or 37.8 µS/cm).

If another model of conductivity meter is used, refer to the procedure presented in Table 3-5. If the meter cannot display temperature compensated conductivity, the team should be provided with a table showing theoretical values of the QCCS solution at different temperatures.

3.1.3.4 Current Velocity Meters--

Field teams may be using one of three types of current velocity meters, a photo-optical impeller type meter (e.g., Swoffer Model 2100) a vertical axis meter (e.g., Price type AA), or an electromagnetic type meter (e.g., Marsh McBirney Model 201D). General guidelines regarding performance checks and inspection of current meters are presented in Table 3-6. Consult the operating manual for the specific meter and modify this information as necessary.

3.1.4 Preparation of Equipment and Supplies

To ensure that all activities at a stream can be conducted completely and efficiently, field teams should check all equipment and supplies before traveling to a stream site. In addition, they should prepare sample containers and labels for use to the extent possible.

TABLE 3-3. STOCK SOLUTIONS, USES, AND INSTRUCTIONS FOR PREPARATION

SOLUTION	USE	PREPARATION
Bleach (10%)	Clean seines, dip nets, kick nets, or other equipment that is immersed in the stream	Dilute 400 mL chlorine bleach solution to 4 L with tap water.
Bleach (90%)	To disinfect gear from spores of whirling disease	Dilute 3.6-L bleach with 400 mL tap water.
"Sparquat"	To disinfect gear from spores of whirling disease	Dissolve 120 mL (2 oz) in 5 gal (19 L) tap water
Conductivity Standard Stock Solution[a]	To prepare conductivity quality control check sample solution	Dissolve 3.4022 g KH_2PO_4 and 3.5490 g Na_2HPO_4 (analytical grade; dried at 120 °C for 3 h and stored desiccated) in 1000.0 g (1.0018 L at 20 °C, 1.0029 L at 25 °C) reagent water.
Quality Control Check Sample	To check operation of conductivity meter	1:100 dilution of standard stock solution with reagent water (theoretical conductivity = 75.3 µS/cm at 25 °C)[a] 1:200 dilution of standard stock solution with reagent water (theoretical conductivity = 37.6 µS/cm at 25 °C)[b]
Formalin, borax buffered[c] (pH 7-8)	Preservative for fish specimens and periphyton samples	Add 400 g borax detergent (e.g., Twenty Mule Team®) to each 20-L container of 100% formalin. Test with pH paper.
Ethanol (95%)	Preservative for benthic macroinvertebrate samples.	None.

[a] Metcalf and Peck (1993)
[b] Peck and Metcalf (1991)
[c] Handle formalin according to 29 CFR 1910.1048.

TABLE 3-4. PERFORMANCE CHECK OF NEWER CONDUCTIVITY METERS[a]

1. If using a combination DO/conductivity meter (e.g., the YSI Model 85), check the conductivity probe after completing the calibration check for the DO probe.

2. Inspect the probe for deposits or fouling.

3. Turn the meter on and make sure all internal electronics checks are completed successfully.
4. Use the MODE key to display “temperature compensated” conductivity (The “EC” symbol on the display will be flashing).

5. Swirl the conductivity probe for 3-5 seconds in a 250-mL bottle containing the daily QCCS solution labeled "RINSE".

6. Transfer the probe from the “RINSE” bottle to a second 250-mL bottle of QCCS labeled “TEST”. Let stabilize for 20 seconds.

7. If the measured value of the QCCS is within ±10% or ±10 µS/cm of the theoretical value (whichever is greater at the theoretical value), rinse the probe in deionized water. Store as described in the operating manual and package the meter for transport to the stream site.
   If the measured value of the QCCS is not within ±10% or ±10 uS/cm of theoretical value, repeat Steps 5 through 7.

8. If the value is still unacceptable, replace the QCCS in both the “rinse” and “test” bottles and repeat the measurement process.
   If the measured value is still not acceptable, clean the conductivity probe as described in the manual, check the batteries, soak in deionized water for 24 hours, and repeat Steps 1 through 7.
   If the measured value is still unacceptable, replace the meter.

[a] For use with YSI Models 85 and 95. Modified from YSI Incorporated. 1986. *Model 85 Handheld Oxygen, Conductivity, Salinity, and Temperature System Operations Manual.* YSI Incorporated, Yellow Springs, OH.

TABLE 3-5. PERFORMANCE CHECK OF OLDER CONDUCTIVITY METERS[a]

1. Check the functioning of the meter according to the manufacturer's operating manual (e.g., zero and "red line" of the meter).

2. Swirl the conductivity probe for 3-5 seconds in a 250-mL bottle containing the daily QCCS solution labeled "RINSE".

3. Transfer the probe from the "RINSE" bottle to a second 250-mL bottle of QCCS labeled "TEST". Let stabilize for 20 seconds.

4. If the measured value of the QCCS is within ±10% or ±10 µS/cm of the theoretical value (whichever is greater at the theoretical value), rinse the probe in deionized water. Store as described in the operating manual and package the meter for transport to the stream site.

   If the measured value of the QCCS is not within ±10% or ±10 uS/cm of theoretical value, repeat Steps 1 through 3.

   If the value is still unacceptable, replace the QCCS in both the "rinse" and "test" bottles and repeat the measurement process.

   If the measured value is still not acceptable, clean the conductivity probe as described in the manual, check the batteries, soak in deionized water for 24 hours, and repeat Steps 1 through 3.

   If the measured value is still unacceptable, replace the meter.

[a] For use with older models of conductivity meters (e.g., YSI Model 33 S-C-T).

TABLE 3-6. GENERAL PERFORMANCE CHECKS FOR CURRENT VELOCITY METERS

Photoelectric Impeller Meters (e.g., Swoffer Model 2100)
● Check that the calibration adjustment cover screws are tightly fitted on the display case.
● Periodically check the condition of the connector fitting between the display unit and the sensor.
● Connect the sensor to the display unit and check the calibration value stored in memory. If this value is less than the correct value for the display unit-sensor rotor combination, replace the batteries.
● Periodically perform a spin test of the rotor assembly, following the instructions in the meter's operating manual. A displayed count value of 300 or greater is indicative of satisfactory performance at low current velocities.
● If a buzzing sound occurs when the rotor assembly is spun by hand, or if the shaft shows visible wear, replace the rotor assembly.
● Periodically examine the thrust-bearing nut on the rotor assembly. If a "cup" begins to form on the bottom surface of the nut, it should be replaced.
Vertical-axis Meters (from Smoot and Novak, 1968)
● Inspect the bucket and wheel hub assembly, yoke, cups, tailpiece, and the pivot point each day before use.
● Inspect the bearings and check the contact chamber for proper adjustment.
● Periodically conduct a spin test of the meter. The minimum spin time is 1.5 minutes, while the recommended time is between 3 and 4 minutes.
Electromagnetic Meters
● Check the meter calibration daily as part of morning routine. Calibration value should be 2.00 $\pm$ 0.05.
● Once per week, check the zero value using a bucket of quiescent water. Place the probe in the bucket and allow to sit for 30 minutes with no disturbance. The velocity value obtained should be 0.0 $\pm$ 0.1. Adjust the meter zero if the value is outside this range.

Check the inventory of equipment and supplies prior to departure using the stream-visit checklists presented in Appendix A. Pack meters, probes, and sampling gear in such a way as to minimize physical shock and vibration during transport. If necessary, prepare stock preservative solutions as described in Table 3-3. Follow the regulations of the Occupational Safety and Health Administration (OSHA) for handling and transporting hazardous materials such as formalin and ethanol. Regulations pertaining to formalin are in the Code of Federal Regulations (CFR; specifically 29 CFR 1910.1048). These requirements should be summarized for all hazardous materials being used for the project and provided to field personnel. Transport formalin and ethanol in appropriate containers with absorbent material.

Inspect the vehicles every morning before departure. Refuel vehicles and conduct maintenance activities the night before a sampling trip. Check vehicle lights, turn signals, brake lights, and air pressure in the tires.

Some sample containers can be labeled before departing from the base location. Figure 3-3 illustrates the preprinted labels. A set of three water chemistry sample containers all having the same ID number (one for the 4-L cubitainer and two for the 60-mL syringes) can be pre-labeled with the appropriate information (described in Section 5). After labeling, place the syringes in their plastic container, and place the cubitainer and beakers in a clean self-sealing plastic bag to prevent contamination. Sample containers for biological and sediment samples should **NOT** be pre-labeled before reaching the stream site. Problems in sample tracking can result if jars are labeled and then are not used at a stream.

3.2 ACTIVITIES AFTER EACH STREAM VISIT

Upon reaching a lodging location after sampling a stream, the team reviews all completed data forms and sample labels for accuracy, completeness, and legibility, and makes a final inspection of samples. If information is missing from the forms or labels, the team leader should fill in the missing information as accurately as possible. The team leader initials all data forms after review. The other team member should inspect and clean sampling equipment, check the inventory of supplies, and prepare samples for shipment. Other activities include shipping samples and communicating with the field coordinator or other central contact person.

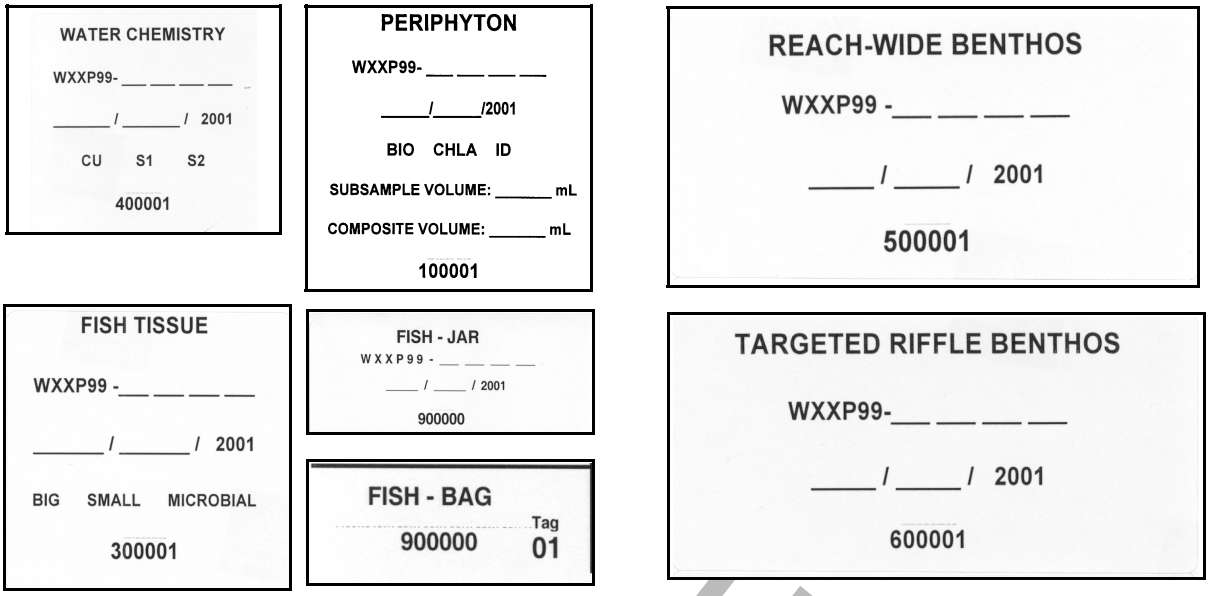

Figure 3-3. Sample container labels.

3.2.1 Equipment Care

Equipment cleaning procedures are given in Table 3-7. Inspect all equipment, including nets, and clean off any plant and animal material. This effort ensures that introductions of nuisance species do not occur between streams, and prevents possible cross-contamination of samples. If nets cannot be cleaned thoroughly using water and detergent, clean and disinfect them with a 10 percent chlorine bleach solution (Table 3-3). Use bleach only as a last resort, as repeated use will destroy the net material. Take care to avoid damage to lawns or other property.

3.2.1.1 Special Precautions Related to Salmonid Whirling Disease–

Salmonid Whirling Disease is caused by a sporozoan parasite (*Myxobolus cerebralis*), and is a serious threat to salmonid populations in several western states. The life cycle of the parasite includes both a "hard spore" and a "fragile spore" stage. The hard spores reside in mud and are very resistant to environmental conditions, remaining dormant for 30 yrs or more. The fragile spores reside in fish and fish parts and the density is very high and concentrated.

TABLE 3-7. EQUIPMENT CARE AFTER EACH STREAM VISIT

1. General cleaning for biological contaminants (e.g., plant and animal material).
   - Prior to departing a stream, drain all water from live wells and buckets used to hold and process fish.
   - Inspect sampling gear and waders, boots, etc. for evidence of plant fragments or animal remains and remove them.
   - At the base location, inspect seines, dip nets, kick nets, waders, and boots with water and dry. If there appears to be the potential for contamination, disinfect gear with a 10 percent bleach solution.

2. Additional precautions to prevent transfer of Whirling Disease spores
   - Consult the site dossier and determine if the stream has been classified as whirling disease positive or negative
   - If the stream is listed as “positive” or no information is available, chemically treat ALL fish and benthos sampling gear and other equipment that has come into contact with water (i.e., waders, boots, etc.) or sediments should be treated by either:
     - A 10-minute soak in a 90% bleach solution, followed by copious rinsing. or
     - A 10-minute soak is Sparquat solution, followed by copious rinsing

3. Clean and dry other equipment prior to storage.

   - Rinse chlorophyll filtration chamber three times with distilled water after each use.
   - Rinse periphyton sampling equipment with tap water at the base location.
   - Rinse coolers with water to clean off any dirt or debris on the outside and inside.
   - Make sure conductivity meter probes are rinsed with deionized water and are stored moist.
   - Rinse all beakers used to collect water chemistry samples three times with deionized water to prevent contamination of the next stream sample. Place the beakers in a 1-gallon self-sealing plastic bag with a cubitainer for use at the next stream.

3. Check fish nets for holes and repair, if possible; otherwise, set damaged gear aside and locate replacements.

4. Inventory equipment and supply needs and relay orders to the Field Coordinator through the Communications Center.

5. Remove DO meters and GPS receivers from carrying cases and set up for pre-visit inspections and performance tests. Examine the DO membrane for cracks, wrinkles, or bubbles; replace if necessary.

6. Recharge all batteries overnight if possible (e.g., electrofishing batteries, 12-V wet cells), computer battery). Replace others (GPS, DO meter, current meter) as necessary.

7. Recheck field forms from the day's sampling activities. Make corrections and completions where possible, and initial each form after review.

8. Replenish fuel in vehicles and/or electrofishing generator (if necessary).

It is extremely important to wash all gear thoroughly with water and remove all mud, debris, etc. to eliminate the possibility of transferring hard spores from one stream to another during the course of a field season. Of higher concern is if infected fish or fish parts (containing fragile spores) are inadvertently transferred from one stream to another and then released or otherwise introduced into a stream.

Field teams should be provided with the latest information (as part of the site dossier) regarding those streams, drainages, etc. that are believed to be infested with Whirling Disease. This information is available for State fishery biologists or pathologists, of from organizations such as the Whirling Disease Foundation (Bozeman, MT). If a team has completed sampling at an infested site and is scheduled to sample a non-infested site next, all gear and sampling equipment must be treated with either a strong bleach solution (90%) or a solution containing “Sparquat” (see Table 3-3), as described in Table 3-7. Pay particular attention to felt soles on wading boots, as the hard spores may embed in this material.

3.2.2 Sample Tracking, Packing, and Shipment

Each field team packs and ships samples from each stream visit as soon as possible after collection, normally the day following a stream visit. Field teams must be provided with specific information for the shipping destinations, contact persons, and the required shipping schedule for each type of sample. Sample tracking information (including sample types, sample ID numbers, and other field-related information that is required by the laboratory to conduct analyses and associate results to a specific sample and stream site) is recorded during the packing process. This information is recorded onto paper forms. The tracking form must be filled out for all samples taken. One form will be filled out on a daily basis and will remain with the site packet. A copy of this form (Figure 3-4) , either xerox or filled in by hand, will be included with unpreserved samples (water chemistry, fish tissue, and periphyton except for ID) shipped to the EPA analytical laboratory facility in Corvallis (Willamette Research Station [WRS]). Another tracking form (Figure 3-5) will include all preserved samples, which will likely be transported to intermediate storage "depots" where they will accumulate prior to shipment to appropriate support laboratories. This form is expected to track samples from multiple sites. The tracking form can be returned to the Information Management staff in Corvallis once it is complete and a copy, either photocopied or filled in by hand, will accompany each shipment of the samples.

FIELD SAMPLE SHIPMENT PACKING/TRACKING FORM

Destination:
Willamette Research Station ☒
1350 Goodnight Ave.
Corvallis, OR 97333
OR: ☐

Date Sent: 07/02/2001
Contact:
Date Received: /__/2001

Airbill Number: 808117727040
IM Contact: MARLYS CAPPAERT (541)754-4467
Lab Contact: KATHY MOTTER (541)754-4877

Site ID	Sample ID	Sample Type	Condition	Comments (List fish tissue species and # small fish here)
WXXP99-9999	100000	CHEM	OK	CU, SI, SZ
WXXP99-9999	300000	FISH	T	20 SMALL FISH - REDSIDE SHINER
WXXP99-9999	300001	FISH	OK	BIG - CUTTHROAT TROUT
WXXP99-9999	300002	FISH	OK	BIG - CUTTHROAT TROUT
WXXP99-9999	300003	FISH	OK	BIG - CUTTHROAT TROAT
WXXP99-9999	300004	FISH	OK	BIG - NORTHERN PIKEMINNOW
WXXP99-9999	300005	FISH	OK	BIG - NORTHERN PIKEMINNOW
WXXP99-9999	300006	FISH	ML	BIG - NORTHERN PIKEMINNOW
WXXP99-9999	300007	FISH	OK	MICROBIAL - MOTTLED SCULPIN
WXXP99-9999	300008	FISH	T	MICROBIAL - MOTTLED SCULPIN
WXXP99-9999	300009	FISH	W	MICROBIAL - CUTTHROAT TROUT
WXXP99-9999	200000	PERI	OK	CHLA, BIO

SAMPLE TYPES	CONDITION CODES
BENT = Benthos	B = Broken Syringe Tip
CHEM = Water Chemistry	C = Cracked Jar
FISH = Fish Tissue	F = Frozen
PERI = Periphyton	L = Leaking
VERT = Fish Museum	ML = Missing Label
	NP = Not Preserved
	OK = Seems Fine
	T = Thawed but Still Cold
	W = Warm

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Figure 3-4. Sample tracking form for unpreserved samples.

FIELD SAMPLE SHIPMENT PACKING/TRACKING FORM

Destination:
Willamette Research Station ☐
1350 Goodnight Ave.
Corvallis, OR 97333
OR: POISON DEPOT ☒
123 HAZMAT RD.
ANYTOWN, XX

Date Sent: 07/12/2001
Contact: H. OLDEMFORME
Date Received: 07/12/2001

Airbill Number: HAND DELIVERED
IM Contact: MARLYS CAPPAERT (541)754-4467
Lab Contact: KATHY MOTTER (541)754-4877

Site ID	Sample ID	Sample Type	Condition	Comments (List fish tissue species and # small fish here)
WXXP99-9999	500000	BENT	NP	RW
WXXP99-9999	500001	BENT	OK	2 jars, TR
WXXP99-9999	200000	PERI	OK	ID
WXYP99-9999	259000	VERT	OK	

SAMPLE TYPES			CONDITION CODES		
BENT	=	Benthos	B	=	Broken Syringe Tip
CHEM	=	Water Chemistry	C	=	Cracked Jar
FISH	=	Fish Tissue	F	=	Frozen
PERI	=	Periphyton	L	=	Leaking
VERT	=	Fish Museum	ML	=	Missing Label
			NP	=	Not Preserved
			OK	=	Seems Fine
			T	=	Thawed but Still Cold
			W	=	Warm

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Figure 3-5. Sample tracking form for preserved samples.

General guidelines for packing and shipping the various types of samples described in this manual are presented in Table 3-8. Use ice substitute packs whenever possible to avoid leakage due to melting ice. When shipping samples using ice, use fresh ice. Use block ice when available; it should be sealed in a large plastic bags. If block ice is not available, contain the ice in several self-sealing plastic bags. Label each bag of ice as "ICE" with an indelible marker to prevent any leakage of meltwater from being misidentified by couriers as a possible hazardous material spill. If ice substitute packs are used, place each pack into a self-sealing plastic bag before use.

Water chemistry samples must be shipped as soon as possible after collection in order to meet holding time requirements for some laboratory analyses (especially nutrients). To ship water chemistry samples, place a large (30-gallon) plastic bag in an insulated shipping container (e.g., a plastic or metal cooler). The sample labels on the cubitainer and syringes should be completely covered with clear tape to prevent damage from water or condensation during shipment. Place the syringes into a separate plastic container for shipment. Place the cubitainer and syringe container into a second large plastic bag and close. Place the bag containing the samples inside the plastic bag lining the shipping container. Place bags of ice around the bag of samples, but inside the plastic bag lining the shipping container.

Then close the outer plastic bag. Seal the cooler with clear tape. Place the required sample tracking forms in the shipping container and close it. Seal the container with shipping tape and affix any required shipping-related labels to the outside of the container. Attach an adhesive plastic sleeve to the lid of the container and insert any required shipping forms.

Samples requiring freezing (Table 3-8) may be stored in the field in a portable freezer or on dry ice for a short period (e.g., one week). If only ice (or ice substitute packs) is available for field storage, ship the samples to the laboratory as soon as possible after collection, using fresh ice (or ice substitute packs) to keep them as cold as possible. When using ice, double bag the ice and tape the last bag shut to prevent contamination of samples by melting ice. If ice substitute packs are used, place each pack into a self-sealing plastic bag. If possible, place samples into a sealed plastic container to protect them from meltwater. Dry ice may also be used for shipping. Note that dry ice is considered a hazardous material, and requires special shipping containers, shipping labels, and shipping forms for ground or air transport. If dry ice is used, the requirements and directions for packing and shipping samples should be provided to each field team.

TABLE 3-8. GENERAL GUIDELINES FOR PACKING AND SHIPPING UNPRESERVED SAMPLES

Sample Type (container)	Guidelines
Samples requiring refrigeration (4 °C)	
Water Chemistry (4-L cubitainer and 60-mL syringes)	Ship on day of collection or within 24 hr by overnight courier. Use fresh ice in labeled plastic bags for shipping. Line each shipping container with a large plastic bag. Place syringes in a plastic container. Place syringe container and cubitainer inside of a second plastic bag. Cover labels completely with clear tape. The cubitainer and syringes should have same sample ID number assigned. Confirm the sample ID assigned on the labels matches the ID number recorded on the field collection form and the sample tracking form.
Samples requiring freezing (-20 °C) within 24 hours of collection	
Periphyton chlorophyll (filter in aluminum foil)	If samples cannot be kept frozen in the field, ship on day of collection or within 24 h by overnight courier. Cover the label completely with clear tape. Protect samples from meltwater if ice is used by double bagging ice and placing samples in a plastic container. Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form (or other sample tracking report). If dry ice is used to transport or ship samples, special shipping containers, outside labeling, and shipping forms may be required.
Periphyton biomass (filter in a numbered container)	
Periphyton activity (50-mL centrifuge tube)	
Fish Tissue (aluminum foil; two 30-gal plastic bags)	If samples cannot be kept frozen in the field, ship on day of collection or within 24 h by overnight courier. Cover labels completely with clear tape. Label on each bag should have identical Sample ID number assigned. Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form (or other sample tracking report). Protect samples from meltwater if ice is used by double bagging ice. Special shipping containers, outside labeling, and shipping forms may be required for shipments containing dry ice.

Each team leader should contact the field coordinator or other central contact person after each stream visit to provide a brief update of each sampling visit, and to request replenishment of supplies if necessary. For each shipment, provide the stream identification number, date sampled, date that samples are being shipped, and the airbill number from the courier's shipping form. If the shipment date is on a Friday, call the contact person or leave a message that a Saturday delivery is coming. Teams should inventory their supplies after each stream visit and submit requests for replenishment well in advance of exhausting on-hand stocks.

3.2.2.1 Packing, Transport and Shipment of Preserved Samples–

Samples that are preserved in buffered formalin (periphyton ID samples and fish voucher specimens) or ethanol (benthic macroinvertebrate samples) should be transported in appropriate containers and surrounded with some type of acceptable absorbent material (e.g., vermiculite). The total volume of formalin in the periphyton ID samples (2 mL per 50-mL centrifuge tube) may be small enough that they may be transported or shipped without designating them as a hazardous material. Guidelines for packing, labeling, transporting, and shipping samples containing formalin or ethanol are presented in Table 3-8, and it may be necessary to provide additional guidance to each field team. Alternatively, these samples may be transported to a central repository for later shipment.

Table 3-9 presents additional guidelines for dealing with preserved samples. In order for field personnel to ship dangerous goods from field locations, they must be trained by a person who has been certified to prepare dangerous goods for shipment. Most of this training deals with packaging, forms and labels that must be used. Dangerous goods shipments must always be presented to the shipper directly as either a pick-up by a driver or a drop-off at a shipping facility.

3.3 EQUIPMENT AND SUPPLIES

A checklist of equipment and supplies required to conduct the activities described in Section 3 is presented in Figure 3-6. This checklist is similar to the checklist in Appendix A, which is used at the base location to ensure that all of the required equipment is brought to the stream. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

TABLE 3-9. GENERAL GUIDELINES FOR PACKING AND SHIPPING PRESERVED SAMPLES

Sample Type (container)	Preservative	Guidelines
Samples requiring preservation in formalin		
Periphyton ID (50-mL centrifuge tube)	10% buffered formalin (2 mL per 50-mL sample)	Labels or tags placed inside of the jar must be of water-resistant paper or 100% rag content paper. The label on outside of the container should be completely covered with clear tape. Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form and sample tracking form.
Fish Specimens (1-L and/or 4-L jars)	10 % buffered formalin	
	Packaging and Shipping Guidance	
Inside packaging		
Outside packaging		
Absorbent material		
Labeling		
Shipping forms		
Samples requiring preservation in ethanol		
Benthic Macro-invertebrates (500-mL or 1-L jars)	70 % ethanol	Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form and sample tracking from.
Packaging and shipping guidance		
Inside packaging	HDPE bottles with leakproof screw-top cap (must meet UN specification IP2). Apply a strip of plastic tape around the cap to seal each bottle securely. Place bottles in upright position in outer package and surround with sufficient absorbent material to prevent tipping.	
Outside packaging	Screw-top plastic pail (5-gal size) with ratcheted lid is recommended. Must meet UN specification 1H2. Each pail can hold no more than 5.0 L total liquid (= 8 500-mL bottles or 5 1-L bottles).	
Absorbent material	Sufficient volume of absorbent material (vermiculite or equivalent) to absorb contents of all inner packaging.	
Labeling	Outside package marked with UN ID no. and name (“1170-Ethanol”), “Flammable Liquid” label, and package orientation label	
Shipping Forms		

BASE LOCATION ACTIVITIES

QTY.	ITEM	
Before Departure for Stream		
1	Dossier of access information for scheduled stream site	
1	Sampling itinerary form or notebook	
1	Safety log and/or personal safety information for each team member	
1	GPS receiver with extra batteries	
1	Dissolved oxygen/temperature meter with probe	
1	Conductivity meter with probe	
1	500-mL plastic bottle containing deionized water	
2	500-mL plastic bottles containing conductivity QCCS, labeled “Rinse” and “Test”	
1	Current velocity meter with probe and wading rod	
	Assorted extra batteries for dissolved, conductivity, and current velocity meters	
1 set	Completed water chemistry sample labels (3 labels with same barcode)	
1 set	Water chemistry sample containers (one 4-L Cubitainer and two 60-mL syringes with a plastic storage container	
1 box	Clear tape strips to cover completed sample labels	
1	Checklist of all equipment and supplies required for a stream visit	
Packing and Shipping Samples		
	Ice (also dry ice if it is used to ship frozen samples)	
1 box	1-gal heavy-duty sealable plastic bags	
1-box	30-gal plastic garbage bags	
2	Insulated shipping containers for frozen samples (special containers may be needed if dry ice is used)	
2	Containers, absorbent material, labels, and shipping forms required to transport and/or ship samples preserved in formalin and ethanol	
2-4	Sample tracking forms (can xerox completed originals or complete two sets of forms per shipment)	
	Shipping airbills and adhesive plastic sleeves	

Figure 3-6. Equipment and supply checklist for base location activities.

3.4 LITERATURE CITED

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NOTES

SECTION 4
INITIAL SITE PROCEDURES

by
Alan T. Herlihy[1]

When a field team first arrives at a stream site, they must first confirm they are at the correct site. Then they determine if the stream meets certain criteria for sampling and data collection activities to occur. They must decide whether the stream is unduly influenced by rain events which could affect the representativeness of field data and samples. Certain conditions at the time of the visit may warrant the collection of only a subset of field measurements and samples. Finally, if it is determined that the stream is to be sampled, the team lays out a defined reach of the stream within which all subsequent sampling and measurement activities are conducted.

Table 4-1 summarizes modifications to procedures from those published previously for EMAP-SW by Herlihy (1998), and from EMAP-WP field activities in 2000. Modifications from Herlihy 91998) include providing guidance for sampling streams that are partially wadeable (Section 4.3.2), and for wide streams with braided channels (Section 4.3.3). Changes from EMAP-WP 2000 activities include not collecting any field data at stream sites having completely dry reaches, and modifying the field data form for use with streams that are either determined to be non-target before a field visit, or that are non-target when visited.

4.1 SITE VERIFICATION ACTIVITIES

4.1.1 Locating the Index Site

Stream sampling points were chosen from the “blue line” stream network represented on 1:100,000- scale USGS maps, following a systematic randomized selection process developed for EMAP stream sampling. Sample sites were then marked with an "X" on finer-resolution 1:24,000-scale USGS maps. This spot is referred to as the “index site”

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TABLE 4-1. SUMMARY OF CHANGES IN INITIAL SITE PROCEDURES FOR THE WESTERN PILOT STUDY

Changes from Herlihy (1998)
4. Developed guidance for sampling streams that are partially wadeable.
5. Developed guidance for how to sample streams that have braided channels..
Changes from Year 2000 Western Pilot Study Activities
1. Field data are non longer collected at sites where the entire sampling reach is dry when visited. They are now classified as non-sampleable
2. The field data form has been revised to deal more clearly with sites that are either determined to be non-target before a field visit or at the time of the visit, ncluding those that are temporarily inaccessible and can be visited again in a future year.
3. Site coordinates can be recorded in DMS or decimal degree format to accomodate different types of GPS units or other data recording requirements of EMAP-WP participants..

or "**X-site**". The latitude/longitude of the X-site will be listed on a stream information sheet that is part of the dossier compiled for each stream (see Section 3).

Complete a verification form for each stream visited (regardless of whether you end up sampling it), following the procedures described in Table 4-2. 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 4-1). This information will allow others to find the site again in the future. Upon reaching the X-site for a stream, confirm its location and that the team is at the correct stream. Use all available means to accomplish this, and record the information on page 1 of the Verification Form (Figure 4-1).

4.1.2 Determining the Sampling Status of a Stream

Not all chosen stream sites will turn out to be streams. On the basis of previous synoptic surveys, it was found that the maps are far from perfect representations of the stream network. A significant part of EMAP is the estimation of the actual extent of stream length in the area. After the stream and location of the X-site are confirmed, evaluate the stream reach surrounding the X-site and classify the stream into one of three major sampling status categories (Table 4-2). The primary distinction between "Sampleable" and

TABLE 4-2. SITE VERIFICATION PROCEDURES

1. Find the stream location in the field corresponding to the "X" marked on a 7.5" topographic map (X-site) that is provided with the dossier for each site. Record the routes taken and other directions on the Verification Form so that someone can visit the same location in the future.

2. If available, use a GPS receiver to confirm the latitude and longitude at the X-site against the coordinates provided in the dossier for the site. Record these on the Verification Form.

3. Use all available means to insure that you are at the correct stream as marked on the map, including: 1:24,000 USGS map orienteering, topographic landmarks, county road maps, local contacts, etc.

4. Scan the stream channel upstream and downstream **from the X-site**, decide if the site is sampleable and mark the appropriate box on the verification form. Assign one of the following sampling status categories to the stream. Record the category on the Verification Form.

Sampleable Categories

Boatable: The site can be sampled by boat following non-wadeable river protocols
Partial Boatable/ Wadeable: Over half the reach cannot be safely sampled by wadeable protocols AND the reach is inaccessible to boat sampling due to barriers or water velocity/depth. Sample using modified procedures.
Wadeable: The stream can be sampled with wadeable stream protocols, continuous water flow and > 50% of the sample reach is wadeable.
Interrupted: The flow of water is not continual, but there is water in the sample reach (e.g. isolated pools). Sample using modified procedures. Record as Wadeable Interrupted or Boatable Interrupted.
Altered Channel: There is a stream at the location marked with the X-site on the map, but the stream channel does not appear the way it is drawn on the map. An example would be a channel rerouting following a flood event that cut off a loop of the stream. Establish a new X-site at the same relative position in the altered channel. Make careful notes and sketches of the changes on the Verification Form.

Non-Sampleable Categories

Dry Channel: A discernible stream channel is present but there is no water in the sample reach. If determined in the field, record on the field form as "Dry-Visited"; if site was determined to be dry from some other source and not field verified, record as "Dry-Not visited".
Wetland (No definable stream channel): There is standing water present, but no definable stream channel. In cases of wetlands surrounding a stream channel, define the site as Target but restrict sampling to the stream channel.
Map Error: No water body or stream channel is present at the coordinates provided for the X-site.
Impounded stream: The stream is submerged under a lake or pond due to man-made or natural (e.g., beaver dam) impoundments. If the impounded stream, however, is still wadeable, record the stream as Altered and sample.
NON-SAMPLEABLE-TEMPORARY: A site that should be sampled but wasn't because the crew did not have the right equipment. Examples are a boatable river visited by a wadeable stream crew without rafts (or vice versa).

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 at the present time due to barriers that may not be present at some future date (e.g. forest fire, high water, road temporarily closed)

5. Do not sample non-target or "Non-sampleable" or "No Access" sites. Place an "X" in the "NO" box for "Did you sample this site?" and check the appropriate box in the "NON-SAMPLEABLE" or "NO ACCESS" section of the Verification Form; provide detailed explanation in comments section.

STREAM VERIFICATION FORM - STREAMS/RIVERS

Reviewed by (initial): DP

SITE NAME: PILOT CREEK DATE: 07/01/2001 VISIT: 0 (1) 2 3

SITE ID: WXXP99-9999 TEAM: XX-1

STREAM/RIVER VERIFICATION INFORMATION

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

Coordinates		Latitude North	Longitude West	Type of GPS Fix	Are GPS Coordinates w/i 10 Sec. of map?
MAP	Degrees, Minutes, and Seconds OR Decimal Degrees	38 10 25	114 25 10	☐ 2D ☒ 3D	☒ Yes ☐ No
GPS	Degrees, Minutes, and Seconds OR Decimal Degrees	38 10 26	114 25 15		

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 (Explain in comments)
- ☐ Partial - Sampled by boat (Explain in comments)
- ☐ Wadeable Interrupted - Not continuous water along reach
- ☐ Boatable Interrupted - Not continuous water along reach
- ☐ Altered - Stream/River Present but not as on 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
- ☐ Not wadeable - Need a different crew
- ☐ Other (Explain in comments)

NO ACCESS
- ☐ Access Permission Denied
- ☐ Permanently Inaccessible (Unable/Unsafe to Reach Site)
- ☐ Temporarily Inaccessible-Fire, etc. (Explain in comments)

GENERAL COMMENTS:

DIRECTIONS TO STREAM/RIVER SITE: From Barnesville, go East on County Road 996 ~5 miles to Smithtown Road. Turn South and travel 0.6 miles to gravel road on left. Turn onto gravel road and drive 0.5 miles to house on right side of road. Owner will unlock gate to road leading to stream near the X-site location.

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

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

"Non-Sampleable" streams is based on the presence of a defined stream channel and water content.

Record the site class and pertinent site verification information on the Verification Form (Figure 4-1). If the site is non-sampleable or inaccessible, the site visit is completed, and no further sampling activities are conducted.

4.1.3 Sampling During or After Rain Events

Avoid sampling during high flow rainstorm events. For one, it is often unsafe to be in the water during such times. In addition, biological and chemical conditions during 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 4-3. The major indicator of the influence of storm events will be the condition of the stream itself. If a field team decides a site is unduly influenced by a storm event, do not sample the site that day. Notify the field coordinator or other central contact person to reschedule the stream for another visit.

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 any photographs at a stream, start the sequence with one photograph of an 8.5 × 11 inch piece of paper with the stream ID, stream name, and date printed in large letters. After the photo of the stream 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. For pictures of aquatic vertebrates (see Section 12) or other small objects, place the paper with the stream ID and date in each snapshot.

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 represen

TABLE 4-3. 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 if it is unsafe to wade in the majority of the stream reach.
- 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, go ahead and sample it, even if it has recently rained or is raining.

tative picture of the ecological community. Previous EMAP pilot studies have suggested that a length of 40 times the channel width is necessary to collect at least 90% of the fish species occurring in the stream reach. Thus, a support reach that is 40 channel widths long around the X-site is required to characterize the community and habitat associated with the sampling point. Establish the sampling reach about the X-site using the procedures described in Table 4-4. 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 4-2. Figure 4-3 illustrates the principal features of the established sampling reach, including the location of 11 cross-section transects used for physical habitat characterization (Section 7), and specific sampling points on each cross-section transect for later collection of periphyton samples (Section 8) and benthic macroinvertebrate samples (Section 11).

There are some conditions that may require adjusting the reach about the X-site (i.e., the X-site no longer is located at the midpoint of the reach) to avoid features we do not wish to sample across. 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). If such a confluence is reached, note the distance and flag the confluence as the endpoint of the reach. Make up for the loss of reach length by moving ("sliding") the other end of the reach an equivalent distance away from the X-site. Similarly, if you run into a lake, reservoir, or pond while laying out the reach, stop, flag the lake/stream confluence as the reach end, and make up for the loss of reach length by moving the other end of the reach an equivalent distance from the X-site. Do not “slide” the reach so that the X-site falls outside of the reach boundaries. Also, do not “slide” a reach to avoid man-made obstacles

TABLE 4-4. LAYING OUT THE SAMPLING REACH

1. Use a surveyor's rod or tape measure to determine the wetted width of the channel at five places considered to be of "typical" width within approximately 5 channel widths upstream and downstream from the X-site. Average the five readings together and round to the nearest 1 m. If the average width is less than 4 m, use 150 m as a minimum sample reach length. Record this width on page 2 of the Verification Form.

   For dry or intermittent channels, estimate the width based on the unvegetated width of the channel.

2. Check the condition of the stream upstream and downstream of 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 from the X-site.

   For example, if the reach length is determined to be 150 m, each person would proceed 75 m from the X-site to lay out the reach boundaries.

3. Determine if the reach needs to be adjusted about the X-site due to confluences with higher order streams (downstream), lower order streams (upstream), or lakes, reservoirs, or ponds.

   If such a confluence is reached, note the distance and flag the confluence as the end-point of the reach. Move the other endpoint of the reach an equivalent distance away from the X-site.

   NOTE: Do not slide the reach to avoid man-made obstacles such as bridges, culverts, rip-rap, or channelization.

4. Starting back 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 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. Using the tape measure, measure 1/10 (4 channel widths in big streams or 15 m in small streams) of the required stream length upstream from the start point (transect A). Flag this spot as the next cross-section or transect (transect B). For transect B, roll the dice to determine if it is a left (L), center (C), or right (R) sampling point for collecting periphyton and benthic macroinvertebrate samples. A roll of 1 or 2 indicates L, 3 or 4 indicates C, and 5 or 6 indicates R (or use a digital wristwatch and glance at the last digit (1-3=L, 4-6=C, 7-9=R). Mark L, C, or R on the transect flagging.

6. Proceed upstream with the tape measure and flag the positions of 9 additional transects (labeled “C” through “J” as you move upstream) at intervals equal to 1/10 of the reach length. Assign sampling spots to each transect in order as L, C, R after the first random selection.

   For example, if the sampling spot assigned to transect “B” was C, transect “C” is assigned R, transect “D” is L, transect “E” is C, etc.

STREAM VERIFICATION FORM - STREAMS/RIVERS (cont.) Reviewed by (initial): DP

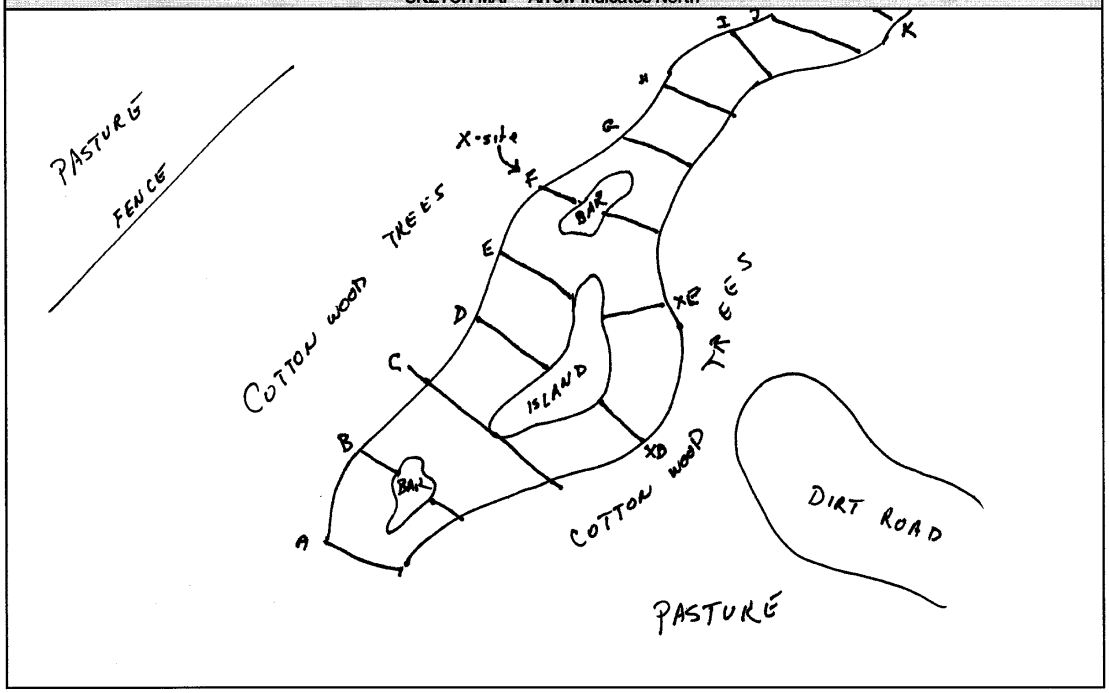
SITE NAME: PILOT CREEK DATE: 07/01/2001 VISIT: 0 (1) 2 3

SITE ID: WXXP99- TEAM: XX-1

STREAM/RIVER REACH DETERMINATION

Channel Width Used to Define Reach (m)	DISTANCE (m) FROM X-SITE		Comment
	Upstream Length	Downstream Length	
3	7.5	7.5	

SKETCH MAP - Arrow Indicates North



PERSONNEL Team Number: XX-1

NAME	DUTIES Biomorph	Geomorph	Forms
J. SHMOE	☒	☐	☐
G. WHIZ	☒	☐	☐
E. MAPPER	☐	☒	☒
S. TREAM	☐	☒	☐
	☐	☐	☐

23755

03/26/2001 2001 Stream Verification

Figure 4-2. Verification Form (page 2).

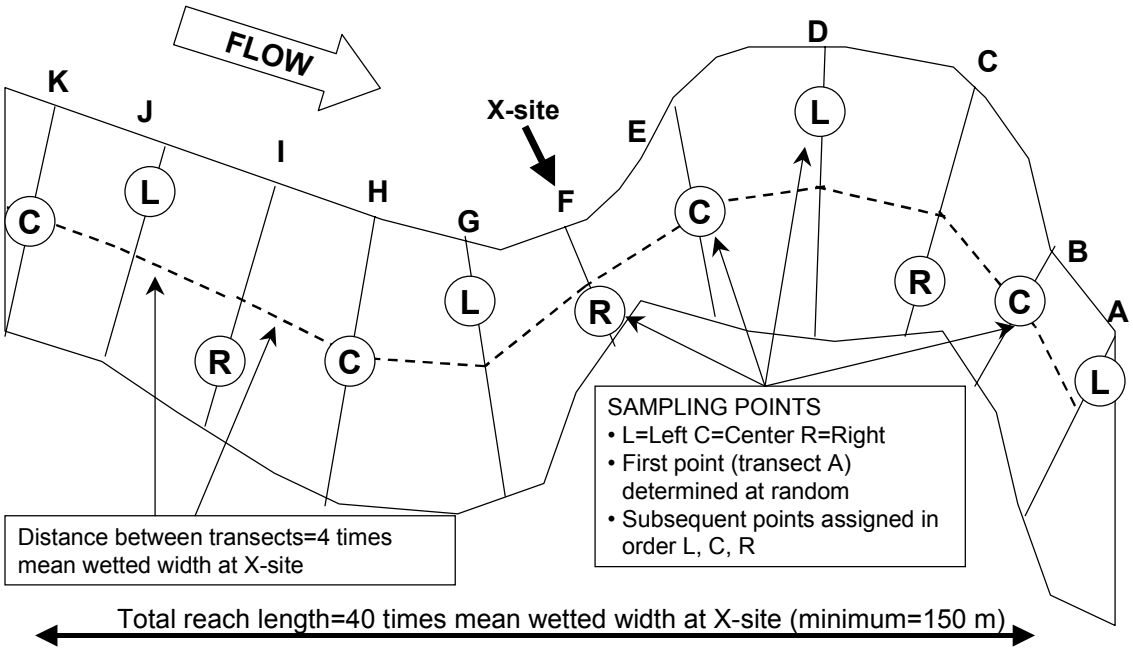


Figure 4-3. Sampling reach features.

such as bridges, culverts, rip-rap, or channelization. These represent features and effects that EMAP is attempting to study.

Before leaving the stream, complete a rough sketch map of the stream reach you sampled on the page 2 of the Verification Form (Figure 4-2). 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.

4.3 MODIFYING SAMPLE PROTOCOLS FOR HIGH OR LOW FLOWS

4.3.1 Dry and Intermittent Streams

The full complement of field data and samples cannot be collected from streams that are categorized as “Interrupted” (Table 4-1). Note that no data should be collected from streams that are completely “Dry” as defined in Table 4-1. Interrupted streams will have some cross-sections with biological and habitat measurements and some with none. Modified procedures for interrupted streams are presented in Table 4-5. Samples and measurements for water chemistry (Section 5) should 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, the sample and chemical measurements are taken from a location having water with a surface area greater than 1 m^2 and a depth greater than 10 cm.

Data for the physical habitat indicator (Section 7) are collected along the entire sample reach from interrupted streams, regardless of the amount of water present at the transects. Depth measurements along the deepest part of the channel (the “thalweg”) are obtained along the entire sampling reach providing 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.

4.3.2 Partial Boatable/Wadeable Sites

Some sites are too deep or swift to safely wade or float the majority or all of the sample reach yet they are also impossible to sample by boat or wading due to shallowness, barriers or current velocity. In these reaches, it will be impossible to do all of either the wadeable or non-wadeable sample protocols. In these sites, keeping safety in mind, the crews should try to do as much of the indicator sampling as they can. It will be impossible to do thalweg depth profiles and flow measurements but it should be possible to do the various assessments that don’t require getting in the water (stream/river assessment, RBP form, riparian condition). It is also usually possible to collect a water sample for chemistry and perhaps to do the transect sampling near the bank for benthos and periphyton. The amount of sampling that can actually be done will be dependent on observed conditions. Be sure to only sample what can be done **safely**. Be sure to make detailed comments on the Verification Form describing what the conditions were like and how much sampling could actually be done. Use the sketch map on the back of the Verification Form to indicate

TABLE 4-5. MODIFICATIONS FOR INTERUPTED STREAMS

Water Chemistry

- If the X-site is dry but there is flowing water or a pool of water having a surface area greater than 1 m^2 and a depth greater than 10 cm somewhere along the defined sampling reach, take the water sample and water chemistry measurements at the pool or flowing water location that is nearest to the X-site. Note that the sample wasn't 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 Characterization, Periphyton, Sediment, 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 macroinvertebrates, sediment or periphyton from this transect.

  DAMP CHANNEL: No flowing water at transect, only puddles of water < 10 cm deep;
  Collect all physical habitat data.
  Collect periphyton samples from the wet spots; these are great places for algae.
  Do not collect a benthic macroinvertebrate or sediment sample.

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

- If at the end of sampling, there were more than 2 transects Dry or Damp so that there are missing macroinvertebrate or periphyton transects in the composite (< 10 transects in composite), then take additional samples from other places in the stream reach that had sufficient water. Preferably, these samples would be taken at the mid-point between transects but may be taken anywhere in streams with only a small amount of water. Try to get an equivalent amount of material as you would from an 11 transect composite. Make detailed notes on the sample collection form for how and where you did sample.

Aquatic Vertebrates

- In interrupted streams, sample any wet areas within the sampling reach that are potential habitat for aquatic vertebrates. **Do not sample downstream of Transect "A" or upstream of Transect "K", even if there appears to be good habitat present.**

problem areas and where samples were collected if you had to go off transect. If barriers to the site prohibit physically reaching the X-site, then the site is not a Sampleable site but should be coded as "No Access - Inaccessible" on the Verification Form.

4.3.3 Braided Systems

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 are generally small and short, often with no obvious dominant channel (See Section 7.6.1). If you encounter a braided stream, establish the sampling reach using the procedures presented in Table 4-6. Figuring the mean width of extensively braided systems for purposes of setting up the sample reach length is a bit of a challenge. For braided systems, calculate the mean width as the bankfull channel width as defined in the physical habitat protocol (Section 7). For relatively small streams (mean bankfull width # 15 m) the sampling reach is defined as 40 times the mean bankfull widths. For larger streams, (> 15 m), sum up the actual wetted width of all the braids and use that as the width for calculating the 40 channel width reach length. If that seems too short for the system in question, by all means set up a longer sample reach. Make detailed notes and sketches on the Verification Form (Figure 4-2) about what you did. It's 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.

4.4 EQUIPMENT AND SUPPLIES

A list of the equipment and supplies required to conduct the stream verification and to lay out the sampling reach is presented in Figure 4-4. 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. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

TABLE 4-6. MODIFICATIONS FOR BRAIDED STREAMS

1. Estimate the mean width as the bankfull channel width as defined in the physical habitat protocol.

   1A. If the mean width is less than or equal to 15 m, set up a 40 channel width sample reach in the normal manner.

   1B. If more than 15 m, sum up the actual wetted width of all the braids and use that as the width for calculating the 40 channel width reach length. Remember the minimum reach length is always 150 m.

   1C. If the reach length determined in 1B seems too short for the system in question, set up a longer sample reach, taking into consideration 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.

4.5 LITERATURE CITED

Herlihy, A.T. 1998. Initial site procedures. pp. 45-56 **IN**: J.M. Lazorchak, D.J. Klemm, and D.V. Peck (Eds.). *Environmental Monitoring and Assessment Program-Surface Waters: Field Operations and Methods for Measuring the Ecological Condition of Wadeable Streams*. EPA/620/R-94/004F. U.S. Environmental Protection Agency, Washington, D.C.

EQUIPMENT AND SUPPLIES FOR INITIAL SITE ACTIVITIES

QTY.	Item	
1	Dossier of site and access information	
1	Topographic map with "X-site" marked	
1	Site information sheet with map coordinates and elevation of X-site	
1	GPS receiver and operating manual	
	Extra batteries for GPS receiver	
1	Verification Form	
	Soft lead (#2) pencils	
1	Surveyor's telescoping leveling rod	
1	50-m fiberglass measuring tape with reel	
2 rolls	Surveyor's flagging tape (2 colors)	
	Fine-tipped indelible markers to write on flagging	
1	Waterproof camera and film (or digital camera)	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for initial site activities	

Figure 4-4. Equipment and supplies checklist for initial site activities.

NOTES

NOTES

SECTION 5
WATER CHEMISTRY

by
Alan T. Herlihy[2]

There are two components to collecting water chemistry information: Collecting samples of stream water to ship to the analytical laboratory, and obtaining *in situ* or streamside measurements of specific conductance, dissolved oxygen, and temperature. At each stream, teams fill one 4-L container and two-four 60 mL syringes (depending on lab analytes to be measured) with streamwater. These samples are stored in a cooler packed with plastic bags filled with ice and are shipped or driven to the analytical laboratory within 24 hours of collection (see Section 3). The primary purposes of the water samples and the field chemical measurements are to determine:

- Acid-base status
- Trophic condition (nutrient enrichment)
- Chemical Stressors
- Classification of water chemistry type.

Water from the 4-L bulk sample is used to measure the major cations and anions, nutrients, total iron and manganese, turbidity and color. The syringe samples are analyzed for pH, dissolved inorganic carbon, and monomeric aluminum species. Syringes are used to seal off the samples from the atmosphere because the pH, dissolved inorganic carbon (DIC), and aluminum concentrations will all change if the streamwater equilibrates with atmospheric CO_2. Overnight express mail for these samples is required because the syringe samples need to be analyzed, and the 4-L bulk sample needs to be stabilized (by filtration and/or acidification) within a short period of time (72 hours) after collection.

In situ and streamside measurements are made using field meters and recorded on standard data forms. Specific conductance (or conductivity) is a measure of the ability of the water to pass an electrical current which is related to the ionic strength of a solution.

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Dissolved oxygen (DO) is a measure of the amount of oxygen dissolved in solution. In natural waters, minimal concentrations of oxygen are essential for survival of most aquatic organisms. Measures of DO and temperature are used to assess water quality and the potential for healthy aerobic organism populations. Most of the procedures outlined in this section are similar to the ones utilized by the EPA in streams for the National Surface Water Survey (Kaufmann et al., 1988) and have been adapted from the Survey's field operations handbook (U.S. EPA, 1989).

Changes in procedures from Herlihy (1998) and from year 2000 EMAP-WP field operations are summarized in Table 5-1. Activities and procedures presented here are essentially unchanged from those previously published for EMAP-SW (Herlihy, 1998). The volume of the bulk water sample has been reduced from approximately 4 L to approximately 3 L. Procedures for measuring in situ DO and conductivity using a combination oxygen/ conductivity/temperature meter are now included. Beginning in 2001, field measurements of dissolved oxygen and conductivity are optional. If field measurements are done, the frequency of QCCS checks of the conductivity meter is reduced. Also, the time of field measurements is recorded.

5.1 SAMPLE COLLECTION

Before leaving the base location, package the sample containers (one 4-L cubitainer and 60 mL syringes) and the stream sample beaker to prevent contamination (see Section 3). Fill out a set of water chemistry sample labels as shown in Figure 5-1. Attach a completed label to the cubitainer and each syringe and cover with clear tape strips as described in Section 3. Make sure the syringe labels do not cover the volume gradations on the syringe. In the field, make sure that the labels all have the same sample ID number (barcode), and that the labels are securely attached.

The procedure to collect a water chemistry sample is described in Table 5-2. The sample is collected from the middle of the stream channel at the X-site, unless no water is present at that location (see Section 4). Throughout the sampling process, it is important to take precautions to avoid contaminating the sample. Rinse all sample containers three times with portions of stream water before filling them with the sample. Many streams have a very low ionic strength and can be contaminated quite easily by perspiration from hands, sneezing, smoking, insect repellent, or other chemicals used when collecting other types of samples. Thus, make sure that none of the water sample contacts your hands before going into the cubitainer. All of the chemical analyses conducted using the syringe samples are

TABLE 5-1. SUMMARY OF CHANGES IN WATER CHEMISTRY PROCEDURES FOR THE WESTERN PILOT STUDY

Changes from Herlihy (1998)
3. The volume of bulk water sample is reduced from 4-L to 3-L.
4. Procedures for using combination oxygen/conductivity/temperature meters are included.
Changes from Year 2000 Western Pilot Study Activities
1. Dissolved oxygen and conductivity measurements are now optional.
2. The frequency of performance evaluation checks for field conductivity meters is reduced from before each field measurement to less frequent checks at base sites or home offices/ laboratories. If used, meters should be subjected to QCCS checks at a minimum frequency of before and after the field season.
3. The field form where D.O. and temperature measurements are recorded also has the Channel constraint information and is not on the reverse of the discharge form.
4. If field measurements are taken, the time of the measurements is recorded on the filed data form..
.

affected by equilibration with atmospheric carbon dioxide; thus, it is essential that no outside air contact the syringe samples during or after collection.

Record the information from the sample label on the Sample Collection Form as shown in Figure 5-2. Note any problems related to possible contamination in the comments section of the form.

5.2 FIELD MEASUREMENTS

Table 5-3 presents the procedures for obtaining field measurement data for the water chemistry indicator. The conductivity and dissolved oxygen meters (if used) are checked in the field using the same procedures as those used at a base location (Section 3). The quality control check sample solution (QCCS) for conductivity is prepared according to directions presented in Section 3. The results of field checks of these meters, the transect where the measurement was made (usually the X-site), as well as the measured values for specific conductance, dissolved oxygen, collection time, and stream temperature, are recorded on the Field Measurement Form as shown in Figure 5-3.

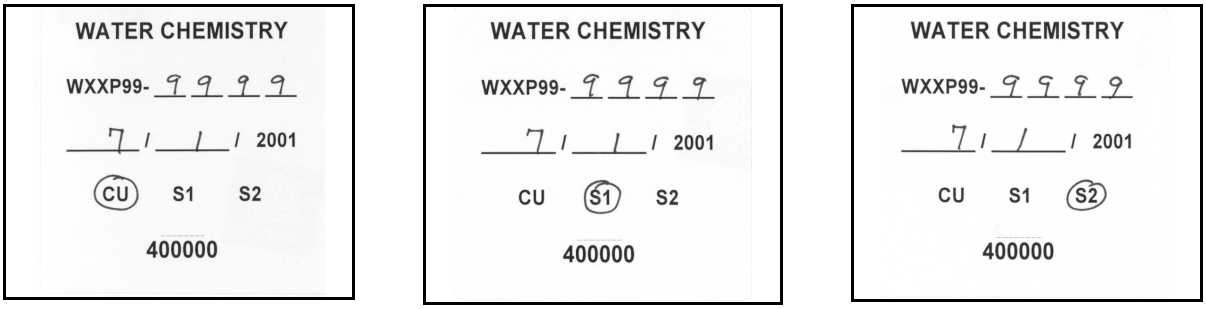

Figure 5-1. Completed sample labels for water chemistry.

If a combination dissolved oxygen/conductivity/temperature meter is being used to determine in situ conditions, the procedure presented in Table 5-4 may be more appropriate to use.

5.3 EQUIPMENT AND SUPPLIES

A list of equipment and supplies required to collect samples and field data for the water chemistry indicator is presented in Figure 5-4. 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. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

5.4 LITERATURE CITED

Herlihy, A.T. 1998. Water chemistry. pp. 57-65 **IN**: J.M. Lazorchak, D.J. Klemm, and D.V. Peck (Eds.). *Environmental Monitoring and Assessment Program-Surface Waters: Field Operations and Methods for Measuring the Ecological Condition of Wadeable Streams*. EPA/620/R-94/004F. U.S. Environmental Protection Agency, Washington, D.C.

TABLE 5-2. SAMPLE COLLECTION PROCEDURES FOR WATER CHEMISTRY

Collect the water samples from the X-site in a flowing portion near the middle of the stream.

1. Rinse the 500 mL sample beaker three times with streamwater, Discard the rinse downstream.

2. Remove the cubitainer lid and expand the cubitainer by pulling out the sides. **NOTE: DO NOT BLOW into the cubitainers to expand them, this will cause contamination.**

3. Fill the beaker with streamwater and slowly pour 30-50 mL into the cubitainer. Cap the cubitainer and rotate it so that the water contacts all the surfaces. Discard the water downstream. Repeat the above rinsing procedure two more times.

4. Collect additional portions of streamwater with the beaker and pour them into the cubitainer. Let the weight of the water expand the cubitainer. The first two portions will have to be poured slowly as the cubitainer expands. Fill the cubitainer to at least three-fourths of its maximum volume. Rinse the cubitainer lid with streamwater. Eliminate any air space from the cubitainer, and cap it tightly. Make sure the cap is tightly sealed and not on at an angle.

5. Place the cubitainer in a cooler (on ice or streamwater) and shut the lid. If a cooler is not available, place the cubitainer in an opaque garbage bag and immerse it in the stream.

6. Submerge a 60-mL syringe halfway into the stream and withdraw a 15-20 mL aliquot. Pull the plunger to its maximum extension and shake the syringe so the water contacts all surfaces. Point the syringe downstream and discard the water by depressing the plunger. Repeat the rinsing procedure two more times.

7. Submerge the syringe into the stream again and **slowly** fill the syringe with a fresh sample. Try not to get any air bubbles in the syringe. If more than 1-2 tiny bubbles are present, discard the sample and draw another one.

8. Invert the syringe (tip pointing up), and cap it with a syringe valve. Tap the syringe lightly to detach any trapped air bubbles. With the valve open, expel the air bubbles and a small volume of water, leaving between 50 and 60 mL of sample in the syringe. Close the syringe valve. If any air bubbles were drawn into the syringe during this process, discard the sample and fill the syringe again (step 8).

9. Repeat Steps 6 through 8 with a second syringe. Place the syringes together in the cooler or in the streamwater with the cubitainer.

10. Record the barcode number (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 the sample was collected at the X-site, record an "X" in the "STATION COLLECTED" field. If you had to move to another part of the reach to collect the sample, place the letter of the nearest transect in the "STATION COLLECTED" field. Record more detailed reasons and/or information in the Comments section.

11. After carrying the samples out to the vehicles, place the cubitainer and syringes in a cooler and surround with 1 gallon self-sealing plastic bags filled with ice.

SAMPLE COLLECTION FORM - STREAMS

Reviewed by (initial): BW

SITE ID: WXXP99-9999 DATE: 07/01/2001

WATER CHEMISTRY

Sample ID	Transect	Comments
229015	X	

REACH-WIDE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999001	2	FOR TRANSECT K OTHER

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	☐ G	☐ G	☒ G	☐ G	☐ G	☒ G	☒ G	☒ G	☐ G	☐ G	☐ G	☒ G	☐ G	☐ G	☒ G	☐ G	☐ G	☐ G	☐ G	☐ G	☐ G
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: Note in Comments	Rapid	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☒ O	☐ RA

TARGETED RIFFLE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999002	1	

NEAREST TRANSECT	A	A	E	E	F	F	G	G
Dom. Substrate: Fine/Sand	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S
Gravel	☒ G	☒ G	☐ G	☒ G	☐ G	☐ G	☒ G	☒ G
Coarse	☐ C	☐ C	☒ C	☐ C	☒ C	☒ C	☐ C	☐ C
Other: Note in Comments	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O

SUBSTRATE SIZE CLASSES
F/S - ladybug or smaller (<2 mm)
G - ladybug to tennis ball (2 to 64 mm)
C - tennis ball to car sized (64 to 4000 mm)
O - bedrock, hardpan, wood, etc

Additional Benthos Comments

COMPOSITE PERIPHYTON SAMPLE

Sample ID	Composite Volume (mL)	Number of transects sampled (0-11):
800990	500	11

Assemblage ID (50-mL tube, preserved)		Chlorophyll (GF/F filter)		Biomass (GF/F Filter)	
Sample Vol. (mL)	Flag	Sample Vol. (mL)	Flag	Sample Vol. (mL)	Flag
50		25		25	

Flag	Comments

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

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03/26/2001 2001 Sample Collection

Figure 5-2. Sample Collection Form, showing data recorded for water chemistry samples.

TABLE 5-3. PROCEDURES FOR STREAMSIDE AND IN SITU CHEMISTRY MEASUREMENTS

Specific Conductance

NOTE: Beginning in 2001, streamside and in situ chemistry measurements are optional.

1. Check the batteries and electronic functions (e.g., zero, “red line") of the conductivity meter as instructed by the operating manual.

2. If you haven’t tested the meter at a base location recently, insert the probe into the “RINSE” container of the quality control check sample (QCCS) and swirl for 3 to 5 seconds. Remove the probe, shake it off gently, transfer it to the "TEST" container of QCCS, and let it stabilize for 20 seconds.

   If the measured conductivity is not within 10% or 10 µS/cm of theoretical value, repeat the measurement process. If the value is still unacceptable, do not use the meter until it can be troubleshooted and repaired.

3. Submerge the probe in and area of flowing water near the middle of the channel at the same location where the water chemistry sample is collected. Record the measured conductivity and any pertinent data comments about the measurement on the Field Measurement Form.

Dissolved Oxygen and Temperature

1. Inspect the probe for outward signs of fouling and for an intact membrane. Do not touch the electrodes inside the probe with any object. Always keep the probe moist by keeping it inside its calibration chamber.

2. Check the batteries and electronic functions of the meter as described in the operating manual.

2. Calibrate the oxygen probe in water-saturated air as described in the operating manual. Allow at least 15 minutes for the probe to equilibrate before attempting to calibrate. Try to perform the calibration as close to stream temperature as possible (not air temperature) by using stream water to fill the calibration chamber prior to equilibration.

3. After the calibration, submerge the probe in midstream at mid-depth at the same location where the water chemistry sample is collected. Face the membrane of the probe upstream, and allow the probe to equilibrate. Record the measured DO and stream temperature on the Field Measurement Form. Record the time the DO and temperature measurement was made in 24 hour units (e.g. 14:23) on the field form. If the DO meter is not functioning, measure the stream temperature with a field thermometer and record the reading on the Field Measurement Form along with any pertinent data comments.

   NOTE: Older model dissolved oxygen probes require a continuous movement of water (0.3 to 0.5 m/s) across the probe to provide accurate measurements. If the velocity of the stream is appreciably less than that, jiggle the probe in the water as you are taking the measurement.

CHANNEL CONSTRAINT AND FIELD CHEMISTRY - STREAMS/RIVERS

Reviewed by (initial): SP

SITE ID: WXXI99-9999 DATE: 07/01/2001

IN SITU MEASUREMENTS		Station ID: (Assume X-site unless marked)
		Comments
STREAM/RIVER DO mg/l: (optional)	5.3	
STREAM RIVER TEMP. (°C):	20.5	
TIME OF DAY:	11:25	

CHANNEL CONSTRAINT

CHANNEL PATTERN (Check 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 (Check 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 (Check 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 % ---> (0-100%)

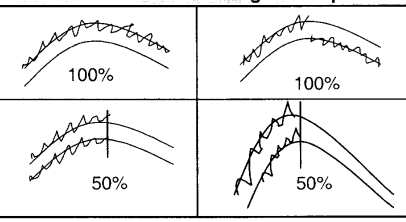
Bankfull width: 6 (m)

Valley width (Visual Estimated Average): 2000 (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: VALLEY WIDTH > 2000 meters

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03/26/2001 2001 Chan Con/Fld Chem

Figure 5-3. Channel Constraint and Field Measurement Form, showing data recorded for water chemistry.

TABLE 5-4. PROCEDURES FOR IN SITU MEASUREMENTS OF DISSOLVED OXYGEN, CONDUCTIVITY, AND TEMPERATURE USING A MULTI-FUNCTION METER[a]

NOTE: Beginning in 2001, field conductivity measurements are optional.

Conductivity QCCS check (no longer required at every site):

5. Check the probe for fouling, intact and unwrinkled oxygen membrane, and bubble behind the membrane. Replace the electrolyte solution and membrane cap assembly if necessary.

6. Turn the meter on and allow the self-test sequence to finish (approx. 15 seconds).

7. Use the MODE key to display "temperature compensated" conductivity (The "EC" symbol on the display will be flashing).

8. Swirl the conductivity probe for 3-5 seconds in a 250-mL bottle containing the daily QCCS solution labeled "RINSE".

9. Transfer the probe from the "RINSE" bottle to a second 250-mL bottle of QCCS labeled "TEST". Let stabilize for 20 seconds.

10. If the measured value of the QCCS is within ±10% or ±10 µS/cm of the theoretical value (whichever is greater at the theoretical value), rinse the probe in deionized water and proceed to Step 8.
    If the measured value of the QCCS is not within ±10% or ±10 µS/cm of theoretical value, repeat Steps 4 through 6.

11. If the value is still unacceptable, replace the QCCS in both the "Rinse" and "Test" bottles and repeat the measurement process.
    If the measured value is still not acceptable, clean the conductivity probe as described in the manual, check the batteries, and repeat Steps 1 through 6.
    If the measured value is still unacceptable, do not make any conductivity measurements. Note problems in the comments section of the field measurement form.

Dissolved oxygen calibration:

12. Check the calibration chamber and fill it with stream water to dampen the sponge and get the chamber temperature as close to stream temperature as possible. Drain the chamber and insert the probe into the chamber.

13. Press the MODE key until the dissolved oxygen reading inside the chamber is displayed in mg/L. Allow approximately 15 minutes for the readings to stabilize (i.e., a change of < 0.02 mg/L over a 1-minute period).

(Continued)

[a] For use with YSI Models 85 and 95. Modified from YSI Incorporated. 1986. *Model 85 Handheld Oxygen, Conductivity, Salinity, and Temperature System Operations Manual.* YSI Incorporated, Yellow Springs, OH.

TABLE 5-4 (continued)

Dissolved oxygen calibration (cont.):

14. Press the UP ARROW and DOWN ARROW keys simultaneously to enter calibration mode.

15. Obtain the approximate local altitude from either the site dossier or from a topographic map. Use the UP ARROW or DOWN ARROW key to enter the local altitude [to the nearest 100 feet (e.g., "15" equals 1500 ft)]. After the correct altitude is displayed, press the ENTER button.

16. In the lower part of the display, "CAL" should appear along with the theoretical value based on temperature and altitude.

17. Once the actual value displayed is stable for 10 seconds, press the ENTER button to save the calibration. **NOTE: make sure the display says "SAVE"**. Do not turn the meter off after saving the calibration.

In situ measurements:

18. Remove the probe from the calibration chamber and hold it in mid-channel and mid-depth at the X-site. Press the MODE button to cycle the display to DO in mg/L. Face the probe up-stream and/or jiggle the probe up and down to ensure a continuous movement of water across the membrane surface. Unstable and inaccurate measurements will result if the flow of water across the membrane is < 0.1 m/s.

19. Wait at least 1 minute for the displayed readings to stabilize, and record the DO value and stream temperature on the Field Measurement Form.

20. Press the MODE button to cycle the display to specific conductance (the "EC" symbol will flash). Record the displayed conductivity value in FS/cm on the Field Measurement Form.
    NOTE: If the conductivity is high (> 999 FS/cm), the display will convert from FS/cm to mS/cm. Be sure to check the units indicated on the display. If mS/cm are displayed, multiply the value by 1000 to convert it to FS/cm before recording it on the data form (e.g., 9 mS/cm would be recorded as 9000 FS/cm). Extremely low values (< 10) are likely to be in mS/cm in most streams sampled in the Western Pilot Study.

21. After completing all in situ measurements, rinse the probe with deionized water and store in the calibration chamber. Be sure to keep the sponge in the chamber moist at all times.

[a] For use with YSI Model 85 or equivalent. Modified from YSI Incorporated. 1986. *Model 85 Handheld Oxygen, Conductivity, Salinity, and Temperature System Operations Manual.* YSI Incorporated, Yellow Springs, OH.

EQUIPMENT AND SUPPLIES FOR WATER CHEMISTRY

QTY.	Item	
1	Dissolved oxygen/Temperature meter with probe	
1	DO repair kit containing additional membranes and probe filling solution	
1	Conductivity meter with probe	
1	500-mL plastic bottle of conductivity QCCS labeled “Rinse” (in plastic bag)	
1	500-mL plastic bottle of conductivity QCCS labeled “Test” (in plastic bag)	
1	Field thermometer	
1	500 mL plastic beaker with handle (in clean plastic bag)	
1	4-L cubitainer with completed sample label attached (in clean plastic bag)	
2-4	60 mL plastic syringes (with Luer type tip) with completed sample labels attached	
1	Plastic container with snap-on lid to hold filled syringes	
2-4	Syringe valves (Mininert® with Luer type adapter, or equivalent, available from a chromatography supply company)	
1	Cooler with 4 to 6 plastic bags (1-gal) of ice OR a medium or large opaque garbage bag to store the water sample at streamside	
1	Sample Collection From	
1	Field Measurement Form	
	Soft-lead pencils for filling out field data forms	
	Fine-tipped indelible markers for filling out labels	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for water chemistry	

Figure 5-4. Checklist of equipment and supplies for water chemistry.

Kaufmann, P., A. Herlihy, J. Elwood, M. Mitch, S. Overton, M. Sale, J. Messer, K. Reckhow, K. Cougan, D. Peck, J. Coe, A. Kinney, S. Christie, D. Brown, C. Hagley, and Y. Jager. 1988. *Chemical Characteristics of Streams in the Mid-Atlantic and South-eastern United States. Volume I: Population Descriptions and Physico-Chemical Relationships*. EPA 600/3-88/021a. U.S. Environmental Protection Agency, Washington, D.C.

U.S. EPA. 1989. *Handbook of Methods for Acid Deposition Studies: Field Operations for Surface Water Chemistry*. EPA 600/4-89/020. U.S. Environmental Protection Agency, Office of Research and Development, Washington, D.C.

NOTES

SECTION 6
STREAM DISCHARGE

by
Philip R. Kaufmann[1]

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 (typically the X-site; see Section 5), so that these data correspond.

Discharge is usually determined after collecting water chemistry samples. Although discharge is part of the physical habitat indicator (Section 7), it is presented as a separate section because the "biomorphs" measure it while the "geomorphs" conduct the other habitat characterization procedures (see Section 2).

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.

The procedures and activities presented here for the EMAP-WP are unchanged from those previously published for EMAP-SW (Kaufmann, 1998). Beginning in 2001, the

[1] U.S. EPA, National Health and Environmental Effects Research Laboratory, Western Ecology Division, 200 SW 35th St., Corvallis, OR 97333.

field data forms have been modified to allow field crews to record a calculated value for discharge, and to record data for more than 20 intervals (using an additional form).

6.1 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 6-1). 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 6-1. Record the data from each measurement on the Stream Discharge Form as shown in Figure 6-2. To reduce redundancy and to conserve space, Figure 6-2 shows measurement data recorded for all procedures. In the field, data will be recorded using only one of the available procedures.

6.2 TIMED FILLING PROCEDURE

In channels too "small" for the velocity-area method, discharge can sometimes be determined directly by measuring the time it takes to fill a container of known volume. “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 free-falling 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.

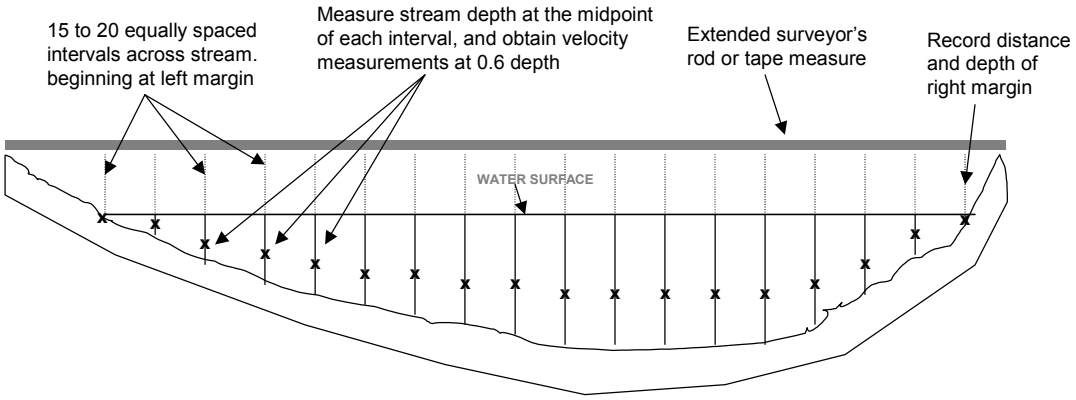

Figure 6-1. Layout of channel cross-section for obtaining discharge data by the velocity-area procedure.

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.

TABLE 6-1. 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 meter 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. Place an "X" in the "VELOCITY AREA" box in the "STREAM DISCHARGE" section of the Field Measurement 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. Record the distance from the left bank (in centimeters) and the depth indicated on the wading rod (in centimeters) on the Field Measurement 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.

(continued)

TABLE 6-1 (continued)

8. Wait 20 seconds to allow the meter to equilibrate, then measure the velocity. Record the value on the Field Measurement Form. Note for the first interval, velocity may equal 0 m/s because depth will equal 0 cm.

    - 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

STREAM DISCHARGE FORM

Reviewed by (Initials): JP

SITE ID: WXXP99-9999 DATE: 07/01/2001

☐ Velocity Area

Distance Units ☐ ft ☒ cm	Depth Units ☐ ft ☒ cm	Velocity Units ☐ ft/s XX.X ☒ m/s X.XX

(Final measurement should be left bank.)

	Dist. from Bank (cm)	Depth	Velocity	Flag
1	0	0	0	F1
2	10	6	0.30	
3	20	6	0.59	
4	30	12	0.37	
5	40	15	0.34	
6	50	15	0.34	
7	60	24	0.43	
8	70	27	0.37	
9	80	40	0.43	
10	90	40	0.37	
11	100	46	0.30	
12	110	37	0.27	
13	120	30	0.27	
14	130	24	0.30	
15	140	15	0.30	
16	150	0	0	Z
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	9
Flag	F1		

Cross Sections on Float Reach

	Upper Section	Middle Section	Lower Section
Width ☐ ft ☒ m	2.5	1.8	1.5
Depth 1 (cm)	10	5	12
Depth 2 (cm)	9	15	20
Depth 3 (cm)	9	20	15
Depth 4 (cm)	8	7	6
Depth 5 (cm)	5	2	5

If discharge is determined directly in field, record value here: Q = 0.12 ☐ cfs ☒ m^3/s

Flag	Comments
F1	MEASUREMENTS FOR ALL 4 DISCHARGE PROCEDURES ARE SHOWN
Z	LAST INTERVAL MEASURED

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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03/26/2001 2001 Stream Discharge

Figure 6-2. Stream Discharge Form, showing data recorded for all discharge measurement procedures.

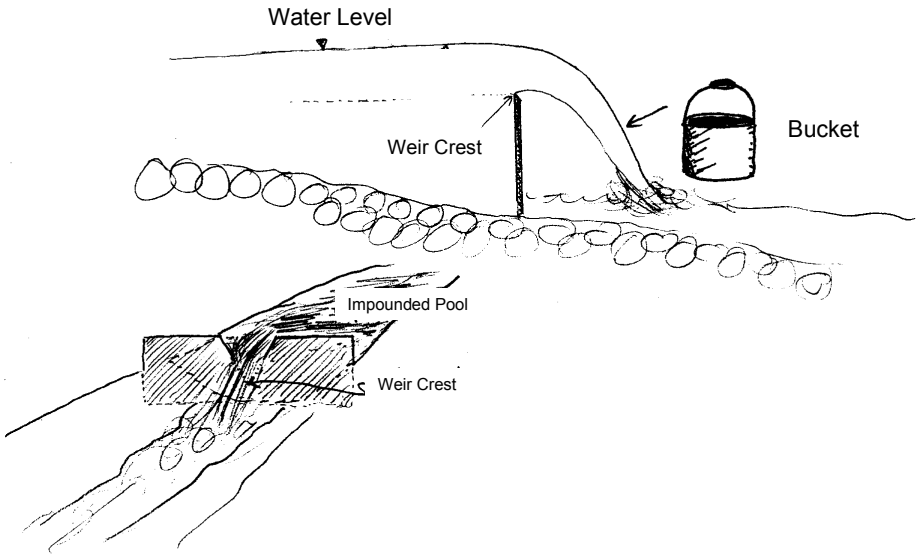


Figure 6-3. Use of a portable weir in conjunction with a calibrated bucket to obtain an estimate of stream discharge.

The timed filling procedure is presented in Table 6-2. 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 field measurement forms if necessary.

TABLE 6-2. 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. Place an "X" in the "TIMED FILLING" box in the stream discharge section of the Field Measurement 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 Field Measurement 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

6.3 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 6-3. Examples of suitable objects include oranges, 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 Field Measurement Form as shown in Figure 6-2.

6.4 EQUIPMENT AND SUPPLIES

Figure 6-4 shows the list of equipment and supplies necessary to measure stream discharge. 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. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

TABLE 6-3. NEUTRALLY BUOYANT OBJECT PROCEDURE FOR DETERMINING STREAM DISCHARGE

1. Place an "X" in the "NEUTRALLY BUOYANT OBJECT" box on the Field Measurement 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. Record the length of the segment in the "FLOAT DISTANCE" field of the Field Measurement 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 (m) using a surveyor's rod or tape measure, and record on the Field Measurement 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 depths (not the distances) in centimeters on the Field Measurement 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 Field Measurement Form.

7. Repeat Step 6 two more times. The float distance may differ somewhat for the three trials

EQUIPMENT AND SUPPLIES FOR STREAM DISCHARGE

QTY.	ITEM	
1	Surveyor's telescoping leveling rod	
1	50-m fiberglass measuring tape and reel	
1	Current velocity meter, probe, and operating manual	
1	Top-set wading rod (metric scale) for use with current velocity meter	
1	Portable Weir with 60° "V" notch (optional)	
1	Plastic sheeting to use with weir	
1	Plastic bucket (or similar container) with volume graduations	
1	Stopwatch	
1	Neutrally buoyant object (e.g., orange, small rubber ball, stick)	
1	Covered clipboard	
	Soft (#2) lead pencils	
	Field Measurement Forms (1 per stream plus extras if needed for timed filling procedure)	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for stream discharge	

Figure 6-4. Equipment and supply checklist for stream discharge.

6.5 LITERATURE CITED

Kaufmann, P.R. 1998. Stream Discharge. pp. 67-76 **IN**: J.M. Lazorchak, D.J. Klemm, and D.V. Peck (Eds.). *Environmental Monitoring and Assessment Program-Surface Waters: Field Operations and Methods for Measuring the Ecological Condition of Wadeable Streams*. EPA/620/R-94/004F. U.S. Environmental Protection Agency, Washington, D.C.

Linsley, R.K., M.A. Kohler, and J.L.H. Paulhus. 1982. *Hydrology for Engineers*. McGraw-Hill Book Co. New York.

Rantz, S.E. and others. 1982. *Measurement and Computation of Streamflow: Volume 1. Measurement of Stage and Discharge.* U.S. Geological Survey Water-Supply Paper 2175.

NOTES

SECTION 7
PHYSICAL HABITAT CHARACTERIZATION (Rev 3/12/01)

(a modification of Kaufmann and Robison, 1998)

Philip R. Kaufmann[1]

In the broad sense, physical habitat in streams includes all those physical attributes that influence or provide sustenance to organisms within the stream. Stream physical habitat varies naturally, as do biological characteristics; 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, because of their influence on discharge, flood stage, and stream power (the product of discharge times gradient). Summarizing the habitat results of a workshop conducted by EMAP on stream monitoring design, 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

All of these attributes may be directly or indirectly altered by anthropogenic activities. Nevertheless, their expected values tend to vary systematically with stream size (drainage area) and overall gradient (as measured from topographic maps). The relationships of specific physical habitat measurements described in this section to these seven attributes are discussed by Kaufmann (1993). Aquatic macrophytes, riparian vegetation, and large woody debris are included in this and other physical habitat assessments because of their

[1] U.S. EPA, Office of Research and Development, National Health and Environmental Effects Laboratory, Western Ecology Division, 200 SW 35th St., Corvallis, OR 97333.

role in modifying habitat structure and light inputs, even though they are actually biological measures. The field physical habitat measurements from this field habitat characterization are used in the context of water chemistry, temperature, and other data sources (e.g., remote sensing of basin land use and land cover). The combined data analyses will more comprehensively describe additional habitat attributes and larger scales of physical habitat or human disturbance than are evaluated by the field assessment alone. A comprehensive data analysis guide (Kaufmann et al., 1999) discusses the detailed procedures used to calculate metrics related to stream reach and riparian habitat quality from filed data collected using the EMAP field protocols. This guide also discusses the precision associated with these measurements and metrics.

These procedures are intended for evaluating physical habitat in wadeable streams. The EMAP field procedures are most efficiently applied during low flow conditions and during times when terrestrial vegetation is active, but may be applied during other seasons and higher flows except as limited by safety considerations. This collection of procedures is designed for monitoring applications where robust, quantitative descriptions of reach-scale habitat are desired, but time is limited. The qualitative nature of the habitat quality rank scores produced by many currently available rapid habitat assessment methods (e.g., those described in Section 14) have not been demonstrated, as yet, to meet the objectives of EMAP, where more quantitative assessment is needed for site classification, trend interpretation, and analysis of possible causes of biotic impairment.

The habitat characterization protocol developed for EMAP differs from other rapid habitat assessment approaches (e.g., Plafkin et al., 1989; Rankin, 1995) by employing a randomized, systematic spatial sampling design that minimizes bias in the placement and positioning of measurements. Measures are taken over defined channel areas and these sampling areas or points are placed systematically at spacings that are proportional to baseflow channel width. This systematic sampling design scales the sampling reach length and resolution in proportion to stream size. It also allows statistical and series analyses of the data that are not possible under other designs. We strive to make the protocol objective and repeatable by using easily learned, repeatable measures of physical habitat in place of estimation techniques wherever possible. Where estimation is employed, we direct the sampling team to estimate attributes that are otherwise measurable, rather than estimating the quality or importance of the attribute to the biota or its importance as an indicator of disturbance. We have included the more traditional visual classification of channel unit scale habitat types because they have been useful in past studies and enhance comparability with other work.

The time commitment to gain repeatability and precision is greater than that required for more qualitative methods. The additional substrate measurements (pebble count of 105 vs 55 particles) adds 20 to 30 minutes to the protocol described by Kaufmann and Robison (1998). In our field trials, two people typically complete the specified channel, riparian, and discharge measurements in about 3.5 hours of field time (see Section 2, Table 2-1). However, the time required can vary considerably with channel characteristics. On streams up to about 4 meters wide with sparse woody debris, measurements can be completed in about two hours. The current protocol, requiring 21 wetted width measurements, will require less than 4.5 hours for a well-practiced crew of two, even in large (>10 m wide), complex streams with abundant woody debris and deep water.

The procedures are employed on a sampling reach length 40 times its low flow 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 spacings. Woody debris is tallied along the full length of the sampling reach, and discharge is measured at one location (see Section 6). 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.

For EMAP-WP, there several modifications to various procedures previously published for EMAP-SW by Kaufmann and Robison (1998). These are summarized in Table 7-1. Four procedures (substrate particle size, instream fish cover, human influence, and thalweg habitat classification) are modified slightly from previous versions., The increase in the number of particles to be included in the systematic pebble count (from 55 particles to 105) increases the precision of substrate characterizations such as %fines. To obtain the additional particles, 10 “supplemental” cross-sections are located mid-way between successive “regular” transects. Procedures for locating and estimating the size of particles on each cross-section remain unchanged, for “regular” and “supplemental” cross-sections, except that only the substrate size class and the wetted width data are recorded at the 10 supplemental cross-sections. Logistically, the supplemental substrate cross-section procedures are accomplished as part of the thalweg profile that is undertaken between regular transects (Section 7.4.1). However, the details of the actual measurements and observations are described in Section 7.5.2. The instream fish cover (Section 7.5.6) and human influence procedures (Section 7.5.7) now include additional or modified features.

TABLE 7-1. SUMMARY OF PHYSICAL HABITAT PROTOCOL CHANGES FOR THE EMAP-SW WESTERN PILOT STUDY

Modifications from Kaufmann and Robison (1998):

11. Substrate: The systematic pebble count is augmented from 55 particles (5 particles in each of 11 cross-sections) to 105 particles (5 particles in each of 21 cross-sections). Ten additional cross-sections are located mid-way between each regular transects. Only the substrate size class and the wetted width data are recorded at each supplemental cross-section.

12. Instream Fish Cover: Fish concealment features now include in-channel live trees or roots. In ephemeral streams these are assessed within the bankfull channel.

13. Human Influence: The human influence category "Pavement" is modified to include cleared barren areas and renamed "Pavement/cleared lot."

14. Riparian "Legacy" Trees and Invasive Alien Plants: New protocol to obtain information on the size and proximity of large, old riparian trees and on the occurrence of non-native invasive tree, shrub and grass species.

15. Channel Constraint: New protocol to classify the general degree of geomorphic channel constraint. This is an overall assessment of reach characteristics that is done after completing the thalweg profile and other measurements at the 11 Cross-section Transects.

16. Debris torrents: New protocol to identify evidence of major floods or debris torrents (lahars). This is an overall assessment for the reach as a whole, and is done after completing the other measurements.

Modifications from Year 2000 Western Pilot Study Activities:

1. Dry Streams: Physical habitat data are no longer collected at streams reaches that are completely dry at the time of the field visit.

2. Off-Channel Backwater Habitat: The thalweg habitat classification now includes the tallying of presence/absence of off-channel backwater habitats, (e.g., sloughs, alcoves, backwater pools). If a backwater pool dominates the main channel habitat, PB is also entered as the channel unit classification code, as in previous versions of this field protocol.

3. Riparian "Legacy" Trees and Invasive Alien Plants: Additional details regarding these procedures is included. Target species of non-native invasive tree, shrub and grass species is modified for some areas of the western U.S.

4. Channel Constraint: Additional detail regarding procedure is included; the number of constraint classes is reduced

In ephemeral streams, fish cover is assessed within the bankfull channel. The thalweg habitat classification (Section 7.4.1) now includes the tallying of presence/absence of off-channel backwater habitats, (e.g., sloughs, alcoves, backwater pools). Backwater pools are included in this tally, but if they are the dominant channel habitat classification, they are also identified by a channel unit classification, as in previous versions of this field protocol.

Three new procedures are included for EMAP-WP. The first (Section 7.5.8) is added to provide additional data on the size and proximity of large, old riparian trees and on the occurrence of non-native invasive tree, shrub and grass species. The second (Section 7.6.1), is added to classify the general degree of geomorphic channel constraint. This is an overall assessment of reach characteristics that is done after completing the thalweg profile and other measurements at the 11 cross-section Transects. Finally, a procedure is added (Section 7.6.2) to identify evidence of major floods or debris torrents (lahars). This is an overall assessment for the reach as a whole, and is done after completing the other measurements. The field form and procedures for assessing debris torrent evidence have been applied in Oregon and Washington research and R-EMAP surveys since 1994.

7.1 COMPONENTS OF THE HABITAT CHARACTERIZATION

There are five different components of the EMAP physical habitat characterization (Table 7-2), including stream discharge, which is described in Section 6. Measurements for the remaining four components are recorded on 11 copies of a two-sided field form, plus separate forms for recording slope and bearing measurements, recording observations concerning riparian legacy (large) trees and alien invasive 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 soft/small sediment deposits, and off-channel habitat at 100 equally spaced intervals (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 plus 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

TABLE 7-2. COMPONENTS OF PHYSICAL HABITAT CHARACTERIZATION

Component	Description
Thalweg Profile: (Section 7.4.1)	• Measure maximum depth, classify habitat and pool-forming features, check presence of backwaters, side channels and deposits of soft, small sediment at 10-15 equally spaced intervals between each of 11 channel cross-section transects (100 or 150 individual measurements along entire reach). • Measure wetted width and evaluate substrate size classes at 11 regular channel cross-section transects and midway between them (21 width measurements and substrate cross-sections).
Woody Debris Tally: (Section 7.4.2)	• Between each of the channel cross sections, tally large woody debris numbers within and above the bankfull channel according to length and diameter classes (10 separate tallies).
Channel and Riparian Characterization: (Section 7.5)	• At 11 cross-section transects (21 for substrate size) placed at equal intervals along reach length: - 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 and large trees; presence of alien plants
Assessment of Channel Constraint, Debris Torrents, and Major Floods (Section 7.6)	• After completing Thalweg and Transect measurements and observations, identify features causing channel constraint, estimate the percentage of constrained channel margin 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: (see Section 6)	• In medium and large streams (defined in Section 6) 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 filling of a bucket or timing the passage of a neutral buoyant object through a segment whose cross-sectional area has been estimated.

[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.

structure, presence of large (legacy) riparian trees, non-native (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.

7.2 HABITAT SAMPLING LOCATIONS WITHIN THE SAMPLING 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 7-1 illustrates the locations within the sampling reach where data for the different components of the physical habitat characterization are obtained. We assess habitat over stream reach lengths that are approximately 40 times their average wetted width at baseflow, but not less than 150 m long. This allows us to adjust the sample reach length to accommodate varying sizes of streams (see Section 2). Many of the 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 sampling reach. In addition, they must be sufficiently close together that they do not "miss" deep areas and habitat units that are in a size range of about ⅓ to ½ of the average channel width. Follow these guidelines for choosing the interval between thalweg profile measurements:

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

Following these guidelines, you will 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. We specify width measurements only at the 11 regular transect cross-sections and 10 supplemental cross-sections at the thalweg measurement points midway between each pair of regular transects (a total of 21 wetted widths). If more resolution is desired, width measurements may be made at all 100 or 150 thalweg profile locations. In contrast with a

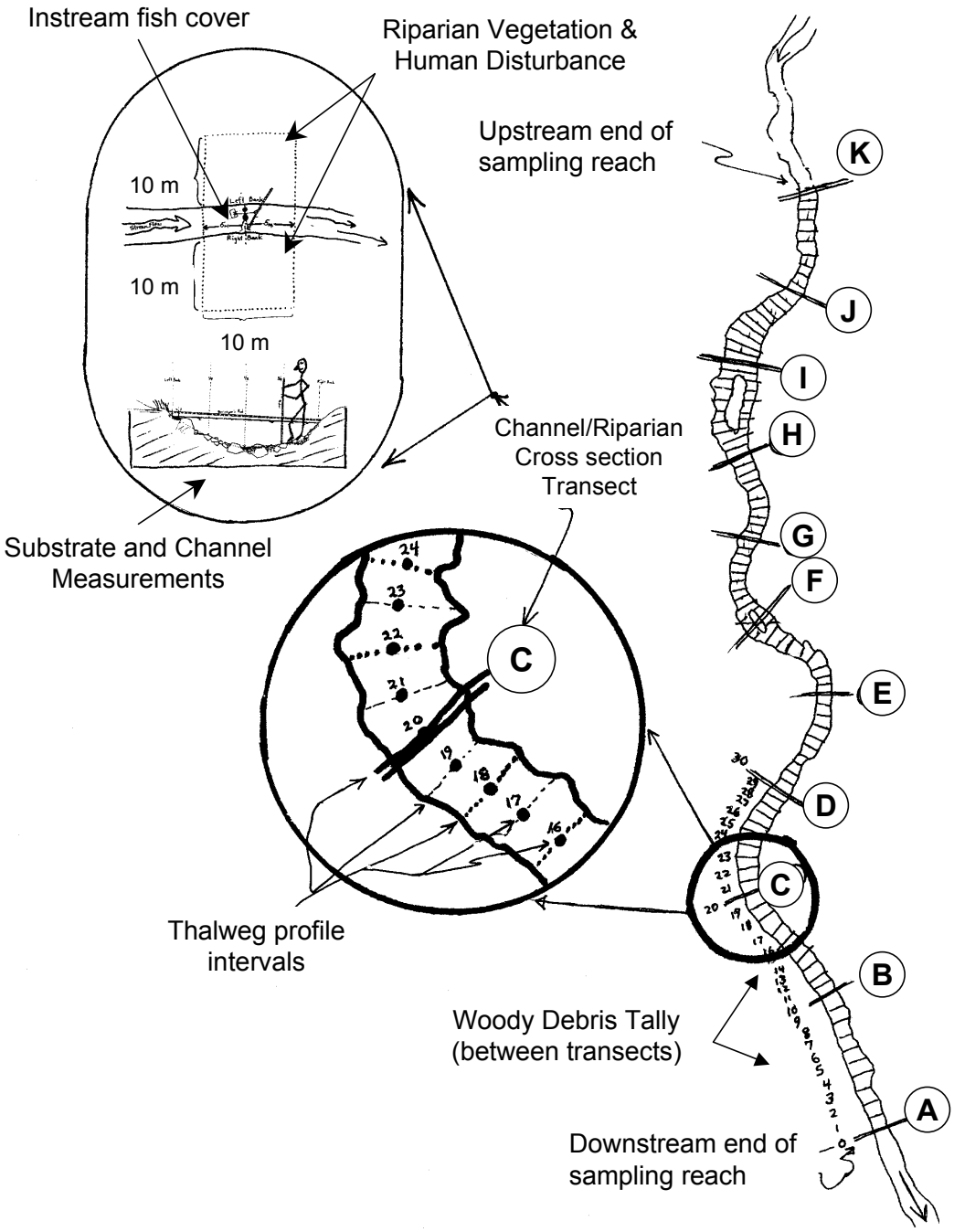

Instream fish cover
Riparian Vegetation &
Human Disturbance
Upstream end of
sampling reach
10 m
10 m
10 m
Channel/Riparian
Cross section
Transect
Substrate and Channel
Measurements
Thalweg profile
intervals
Woody Debris Tally
(between transects)
Downstream end of
sampling reach
K
J
I
H
G
F
E
D
C
B
A

Figure 7-1. Sampling reach layout for physical habitat measurements (plan view).

previous publication of these methods (Kaufmann and Robison, 1998), where substrate particles are evaluated at 5 cross-section locations at 11 transects, we specify substrate measurements at the 10 supplemental cross-sections in addition to those at the 11 regular transects, for a systematic "pebble count" of 105 (rather than 55) particles.

7.3 LOGISTICS AND WORK FLOW

The five components (Table 7-2) of the habitat characterization are organized into four grouped activities:

1. Thalweg Profile and Large Woody Debris Tally (Section 7.4). Two people (the "geomorphs") proceed upstream from the downstream end of the sampling reach (see Figure 7-1) 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), s/he also locates and estimates the size class of the substrate articles 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.

2. Channel/Riparian Cross-Sections (Section 7.5). 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 and Invasive "Alien" Plant field form. Slope and bearing are determined together by backsiting to the previous transect. Intermediate flagging (of a different color)

may have to be used if the stream is extremely brushy, sinuous, or steep to the point that you cannot site for slope and bearing measures between two adjacent transects. (Note that the crews could tally woody debris while doing the back-sight, rather than during the thalweg profile measurements.)

3. Channel Constraint and Torrent Evidence (Section 7.6). 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. Discharge (Section 6). Discharge measurements are made after collecting the 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.

7.4 THALWEG PROFILE AND LARGE WOODY DEBRIS MEASUREMENTS

7.4.1 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 depth and several other selected characteristics at 100 or 150 equally spaced points along the centerline of the stream between the two ends of the stream reach. 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. The EMAP-SW habitat assessment modifies traditional methods by proceeding upstream in the middle of the channel, rather than along the thalweg itself (though each thalweg depth measurement is taken at the deepest point at each incremental position). One field person walks upstream (wearing felt-soled waders) 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). 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 7-3. Record data on the Thalweg Profile and Woody Debris Data Form as shown in Figure 7-2. Use the surveyor's rod and a metric ruler or calibrated rod or pole to make the required

TABLE 7-3. THALWEG PROFILE PROCEDURE

1. Determine the interval between measurement stations based on the wetted width used to determine the length of the sampling reach.

   For widths < 2.5 m, establish stations every 1 m.
   For widths between 2.5 and 3.5 m, establish stations every 1.5 m
   For widths > 3.5 m, establish stations at increments equal to 0.01 times the sampling reach length.

2. Complete the header information on the Thalweg Profile and Woody Debris Form, noting the transect pair (downstream to upstream). Record the interval distance determined in Step 1 in the "INCREMENT" field on the field data form.

   NOTE: If a side channel is present, and contains between 16 and 49% of the total flow, establish secondary cross-section transects as necessary. Use separate field data forms to record data for the side channel, designating each secondary transect by checking both "X" and the associated primary transect letter (e.g., XA, XB, etc.). Collect all channel and riparian cross-section measurements from the side channel.

3. Begin at the downstream end (station "0") of the first transect (Transect "A").

4. Measure the wetted width if you are at station "0", 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 widths up to about 3 meters, and to the nearest 5% for widths > 3 m. This is 0.2 m for widths of 4 to 6 m, 0.3 m for widths of 7 to 8 m, and 0.5 m for widths of 9 or 10 m, and so on. For dry and intermittent streams, where no water is in the channel, record zeros for wetted width.

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

5. At station 5 or 7 (see above) classify the substrate particle size 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 A through K, except that for these mid-way supplemental cross-sections, substrate size is entered on the Thalweg Profile side of the field form.

6. At each thalweg profile station, use a meter ruler or a calibrated pole or rod to locate the deepest point (the "thalweg"), which may not always be located at mid-channel. Measure the thalweg depth to the nearest cm, and record it on the thalweg profile form. Read the depth on the side of the ruler, rod, or pole to avoid inaccuracies due to the wave formed by the rod in moving water.

   NOTE: For dry and intermittent streams, where no water is in the channel, record zeros for depth.

(continued)

TABLE 7-3 (Continued)

NOTE: **At stations where the thalweg is too deep to measure directly**, stand in shallower water and extend the surveyor's rod or calibrated rod or pole at an angle to reach the thalweg. Determine the rod 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. Record the water level on the rod and the rod angle in the comments section of the field data form. For even deeper depths, it is possible to 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 then 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.

7. At the point where the thalweg depth is determined, observe whether 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 staff. 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 for this assessment. 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.

11. 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.

12. Proceed upstream to the next station, and repeat Steps 4 through 11.

13. Repeat Steps 4 through 12 until you reach the next transect. At this point complete Channel/Riparian measurements at the new transect (Section 7.5). 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").

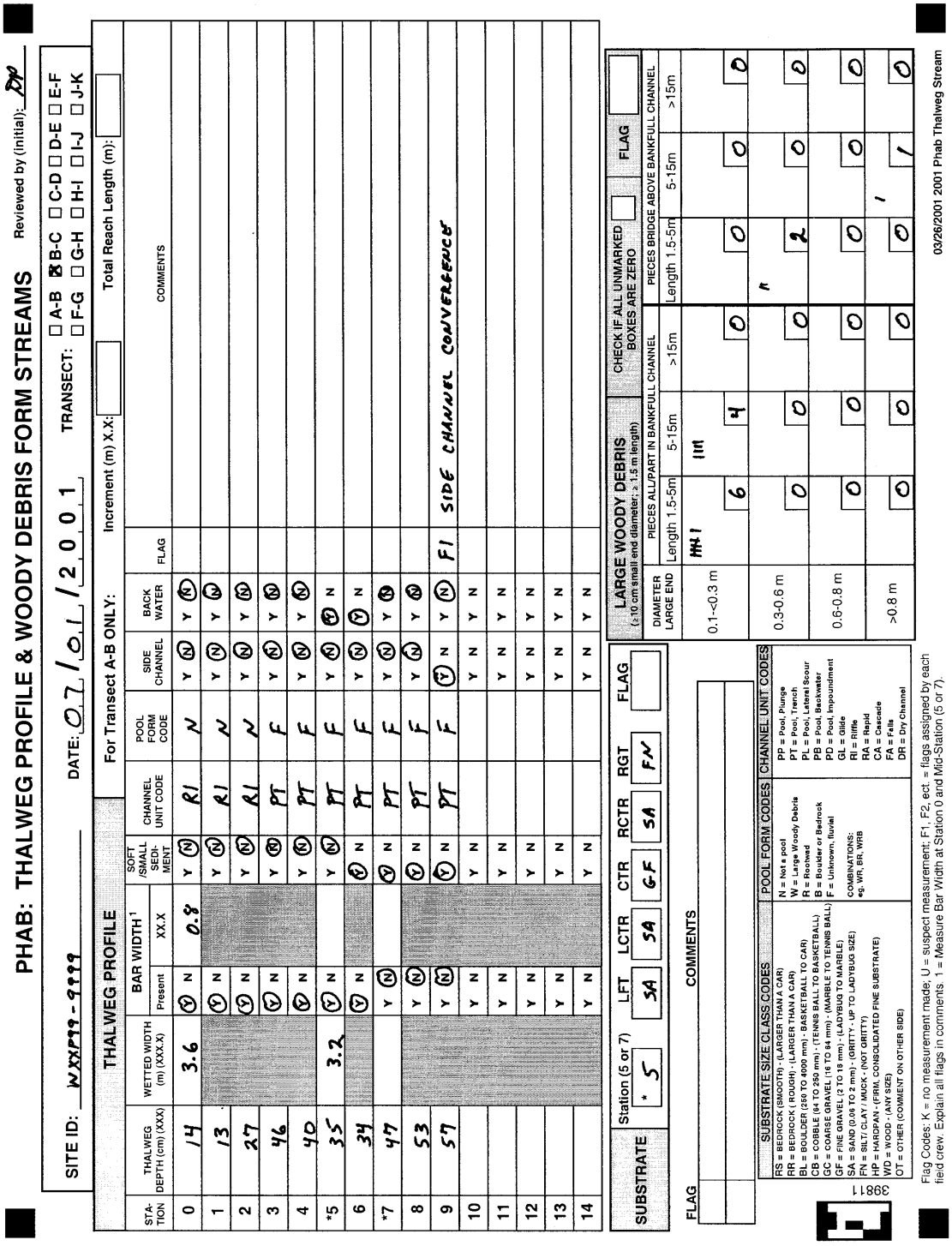


Figure 7-2. Thalweg Profile and Woody Debris Form.

depth and width measurements, and to measure off the distance between measurement points as you proceed upstream. Ideally, every tenth thalweg measurement will bring you within one increment spacing from the flag marking a new cross-section profile. The flag will have been set previously by carefully taping along the channel, making the same bends that you do while measuring the thalweg profile (refer to Figure 7-1). However, you may still need to make minor adjustments to align each 10th measurement to be one thalweg increment short of the cross section. In streams with average widths smaller than 2.5m, you will be making thalweg measurements at 1-meter increments. Because the minimum reach length is set at 150 meters, there will be 15 measurements between each cross section. Use the 5 extra lines on the thalweg profile portion of the data form (Figure 7-2) to record these measurements.

It is very important that thalweg depths are obtained from all measurement points. Missing depths at the ends of the sampling 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 occurring in the middle of the sampling reach are more difficult to deal with. Flag these missing measurements using a "K" code and explain the reason for the missing measurements in the comments section of the field data form. At points where a direct depth measurement cannot be obtained, make your best estimate of the depth, record it on the field form, and flag the value using a "U" code (for suspect 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°). This procedure can also be done with a taut string or fishing line (see Table 7-3). In analyzing this data we calculate the thalweg depth as the length of rod (or string) under water multiplied by the trigonometric *sin* of the rod angle. (For example, if 3 meters of the rod are under water when the rod held at 30 degrees (*sin*=0.5), the actual thalweg depth is 6 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, and a comment to the right saying "depth taken at an angle of xx degrees."

At every thalweg measurement increment, determine by sight or feel whether deposits of soft/small sediment is 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 NOT the same as "Fines"** described when determining the substrate particle sizes at the cross-section transects (Section 7.5.2). For the thalweg

profile, determine if soft/small sediment deposits are readily obvious by feeling the bottom with your boot, the surveyor's rod, or the calibrated rod or pole. (Note that **a very thin coating of silt or algae on cobble bottom substrate does not qualify as "soft/small" sediment** for this purpose.)

Wetted width is measured 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. Substrate size is estimated for 5 particles evenly spaced across each midway cross-section using procedures identical to those described for substrate at regular cross-sections (Section 7.5.2), but at the supplemental cross-sections, only the size class (not the distance and depth) data are recorded in spaces provided on the Thalweg Profile side of the field form.

While recording the width and depth measurements and the presence of soft/small sediments, the second person chooses and records the habitat class and the pool forming element codes (Table 7-4) applicable to each of the 100 (or 150) measurement points along the length of the reach. These channel unit habitat classifications and pool-forming elements are modified from those of Bisson et al. (1982) and Frissell et al. (1986). The resulting database of traditional visual habitat classifications will provide a bridge of common understanding with other studies. Channel unit scale habitat classifications are to be made 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 7-4. 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, don't record it as a pool unless it occupies an area about as wide or long as the channel is wide. <u>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, 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.</u>

Mid-channel bars, islands, and side channels pose some problems for the sampler conducting a thalweg profile and necessitate some guidance. Bars are defined here as

TABLE 7-4. CHANNEL UNIT AND POOL FORMING ELEMENT CATEGORIES

Channel Unit Habitat Classes[a]	
Class (Code)	**Description**
Pools:	Still water, low velocity, 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.
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 white-water with breaking waves. Sound: continuous rushing, but not as loud as cascade.
Cascade (CA)	Water movement rapid and 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 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

(continued)

[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.

TABLE 7-4 (Continued)

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] 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.

mid-channel features below the bankfull flow mark that are dry during baseflow conditions (see Section 7.5.3 for the definition of bankfull channel). Islands are mid-channel features that are dry even when the stream is experiencing a bankfull flow. Both bars and islands cause the stream to split into side channels. When a mid-channel bar is encountered along the thalweg profile, it is noted on the field form and the active channel is considered to include the bar. Therefore, the wetted width is measured as the distance between wetted left and right banks. It is measured across and over mid-channel bars and boulders. If mid-channel bars are present, record the bar width in the space provided.

If a mid-channel feature is as high as the surrounding flood plain, it is considered an island. Treat side channels resulting from islands different from mid-channel bars. Handle the ensuing side channel based on visual estimates of the percent of total flow within the side channel as follows:

Less than 15% Indicate the presence of a side channel on the field data form.

16 to 49% Indicate the presence of a side channel on the field data form. Establish a secondary transect across the side channel designated as "X" plus the primary transect letter; (e.g., XA), by checking boxes for both "X" and the appropriate transect letter (e.g., A through K) on a separate copy of the field data form. Complete the detailed channel and riparian cross-section measurements for the side channel on this form.

When a side channel occurs due to an island, reflect its presence with continuous entries in the "Side Channel" field on the Thalweg Profile and Woody Debris Form (Figure 7-2). In addition, note the points of divergence and confluence of the side channel in the comments section of the thalweg profile form. Begin entries at the point where the side channel converges with the main channel; note the side channel presence continuously until the upstream point where it diverges. When doing width measures with a side channel separated by an island, include only the width of the main channel in the measures at the time and then measure the side channel width separately.

For dry and intermittent streams, where no water is in the channel at a thalweg station, record zeros for depth and wetted width. Record the habitat type as dry channel (DR).

7.4.2 Large Woody Debris Tally

Methods for large woody debris (LWD) measurement are a simplified adaptation of those described by Robison and Beschta (1990). This component of the EMAP physical habitat characterization allows quantitative estimates of the number, size, total volume and distribution of wood within the stream reach. LWD 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 7-5. The tally includes all pieces of LWD that are at least partially in the baseflow channel, the "active channel" (flood channel up to bankfull stage), or spanning above the active channel (Figure 7-3). The active (or “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 the active channel is tallied over the entire length of the reach, including the area between the channel cross-section transects. As in the thalweg profile, LWD measurements in the LWD piece is tallied in only one box. Pieces of LWD that are not at least partially within Zones 1, 2, or 3 are not tallied.

For each LWD piece, first visually estimate its length and its large and small end diameters in order to place it in one of the diameter and length categories. The diameter class on the field form (Figure 7-2) refers 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 a circular cross section that would have the same volume. When evaluating length, include only the part of the LWD piece that has a diameter greater than 10 cm (4 in). Count each of the LWD pieces as one tally entry and include the whole piece when assessing dimensions, even if part of it is in Zone 4 (outside of the bankfull channel). For both the Zone 1-2 wood and the Zone 3 LWD, the field form (Figure 7-2) provides 12 entry boxes for tallying debris pieces visually estimated within three length and four diameter class combinations. Each LWD piece is tallied in only one box. There are 12 size classes for wood at least partially in Zones 1 and 2, and 12 for wood partially within Zone 3. Wood that is not at least partially within those zones is not tallied.

7.5 CHANNEL AND RIPARIAN MEASUREMENTS AT CROSS-SECTION TRANSECTS

7.5.1 Slope and Bearing

The slope, or gradient, of the stream reach is useful in three different ways. First, the overall stream gradient is one of the major stream classification variables, giving an

TABLE 7-5. PROCEDURE FOR TALLYING LARGE WOODY DEBRIS

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

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.]; 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 greater than 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.

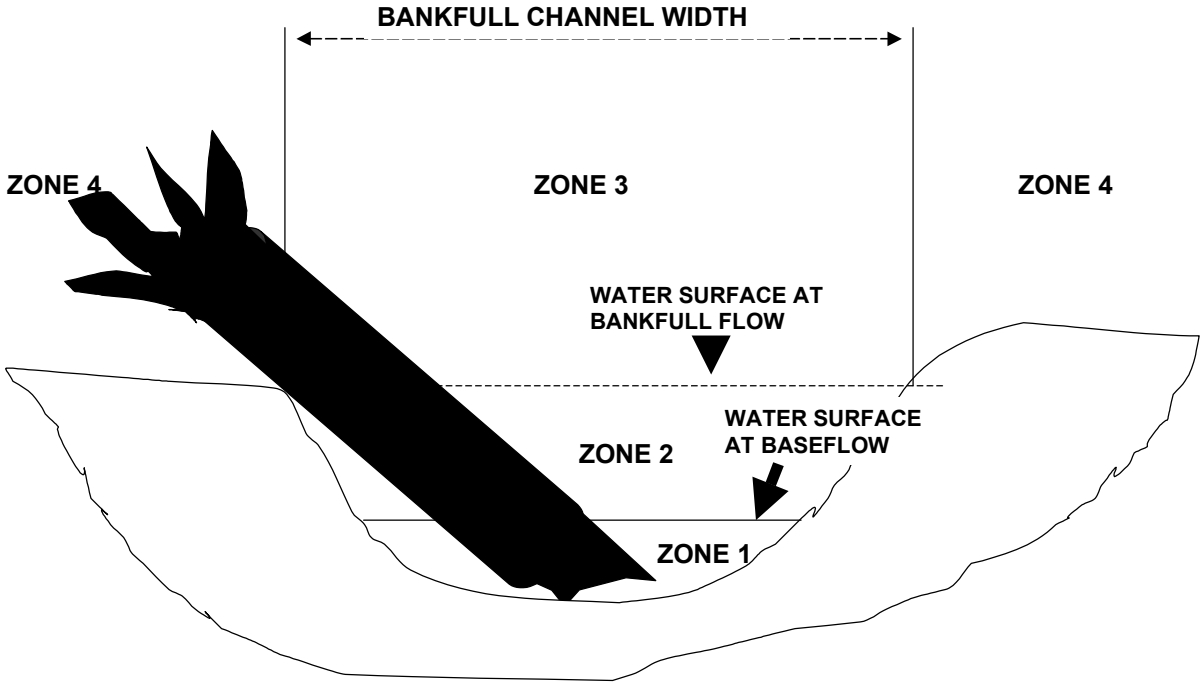


Figure 7-3. Large woody debris influence zones (modified from Robison and Beschta, 1990)

indication of potential water velocities and stream power, which are in turn important controls on aquatic habitat and sediment transport within the reach. Second, the spatial variability of stream gradient is a measure of habitat complexity, as reflected in the diversity of water velocities and sediment sizes within the stream reach. Lastly, using methods described by Stack (1989) and Robison and Kaufmann (1994), the water surface slope will allow us to compute residual pool depths and volumes from the multiple depth and width measurements taken in the thalweg profile (Section 7.4.1). Compass bearings between cross section stations, along with the distance between stations, will allow us to estimate the sinuosity of the channel (ratio of the length of the reach divided by the straight line distance between the two reach ends).

Measure slope and bearing by "backsighting" downstream between transects (e.g., transect “B” to “A”, “C” to “B”, etc.) as shown in Figure 7-4. To measure the slope and bearing between adjacent stations, use a clinometer, bearing compass, tripod, tripod extension, and flagging, following the procedure presented in Table 7-6. Record slope and bearing data on the Slope and Bearing Form as shown in Figure 7-5.

Slope can also be measured by two people, each having a pole that is marked at the same height. Alternatively, the second person can be “flagged” at the eye level of the person doing the backsiting. Be sure that you mark your eye level on the other person or on a separate pole beforehand while standing on level ground. Site to **your eye level** when backsiting on your co-worker. **Particularly in streams with slopes less than 3%, we recommend that field crews use poles marked at exactly the same height for sighting slope. When two poles are used, site from the mark on one pole to the mark on the other. Also, be sure that the second person is standing (or holding the marked pole) at the water’s edge or in the same depth of water as you are.** The intent is to get a measure of the **water surface slope**, which may not necessarily be the same as the bottom slope.

The clinometer reads both percent slope and degrees of the slope angle; be careful to read and record percent slope. Percent slope is the scale on the right-hand side as you look through most clinometers. If using an Abney Level, insure that you are reading the scale marked “PERCENT.” With the clinometer or the Abney level, verify this by comparing the two scales. Percent slope is always a higher number than degrees of slope angle (e.g., 100% slope=45° angle). For slopes > 2%, read the clinometer to the nearest 0.5%. For slopes < 2%, read to the nearest 0.25%. If the clinometer reading is 0%, but water is

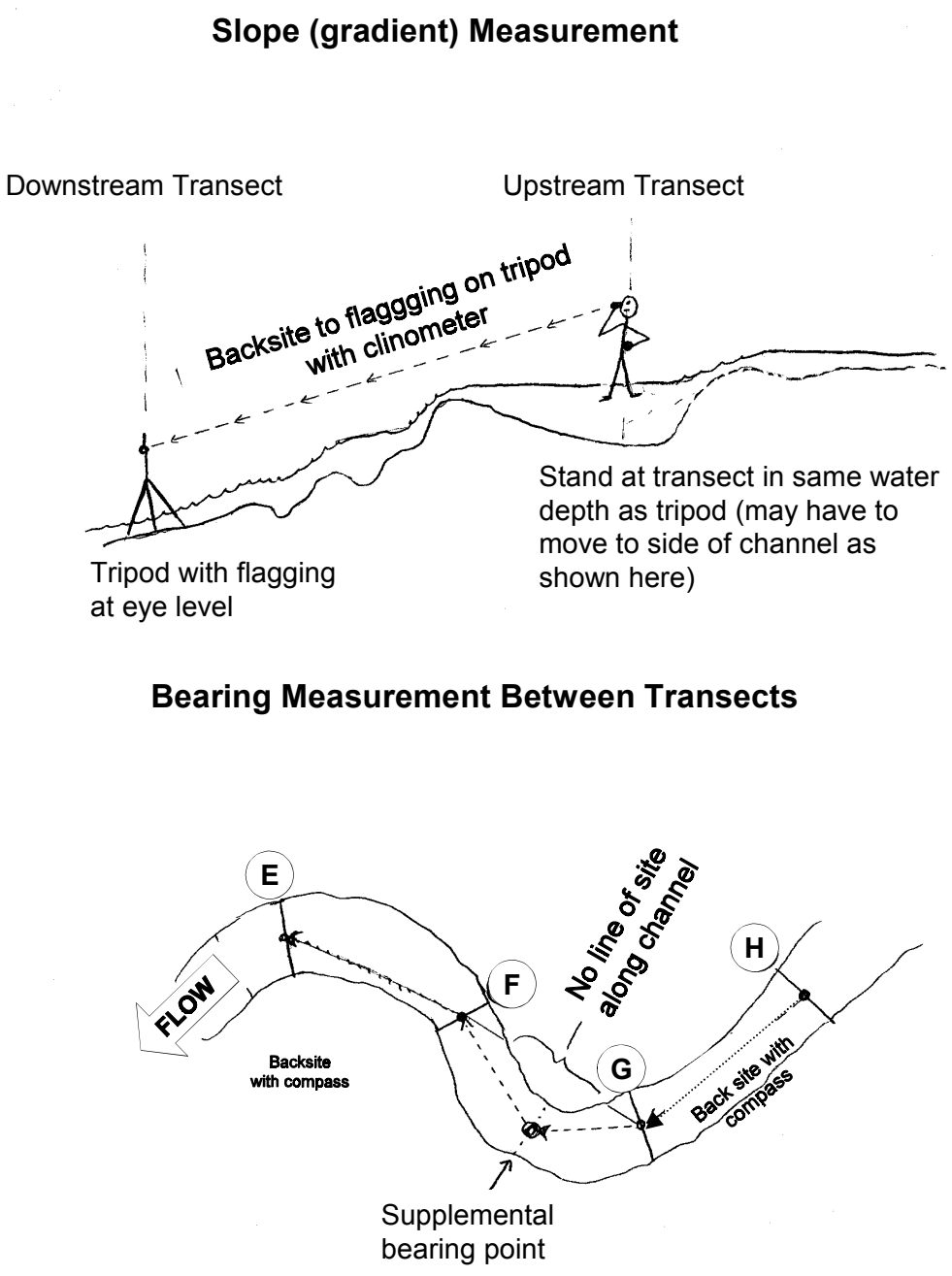


Figure 7-4. Channel slope and bearing measurements.

TABLE 7-6. PROCEDURE FOR OBTAINING SLOPE AND BEARING DATA

1. Stand in the center of the channel at the downstream cross-section transect. Determine if you can see the center of the channel at the next cross-section transect upstream 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.

2. Set up the tripod in shallow water or at the water's edge at the downstream cross-section transect (or at a supplemental point). Standing tall in a position with your feet as near as possible to the water surface elevation, set the tripod extension and mark it with a piece of flagging at your eye level. Remember the depth of water in which you are standing when you adjust the flagging to eye level.

   - On gradually sloped streams, it is advisable to use two people, each holding a pole marked with flagging at the same height on both poles.

3. Walk upstream to the next cross-section transect. Find a place to stand at the upstream transect (or at a supplemental point) that is at the same depth as where you stood at the downstream transect when you set up the eye-level flagging.

   - If you have determined in Step 1 that supplemental measurements are required for this segment, walk upstream to the furthest point where you can still see the center of the channel at the downstream cross-section transect from the center of the channel. Mark this location with a different color flagging than that marking the cross-section transects.

4. With the clinometer, site back downstream on your flagging at the downstream transect (or at the supplementary point). Read and record the **percent** slope in the "MAIN" section on the Slope and Bearing Form. Record the "PROPORTION" as 100%.

   - If two people are involved, place the base of each pole at the water level (or at the same depth at each transect). Then site with the clinometer (or Abney level) from the flagged height on upstream pole to the flagged height on the downstream pole.

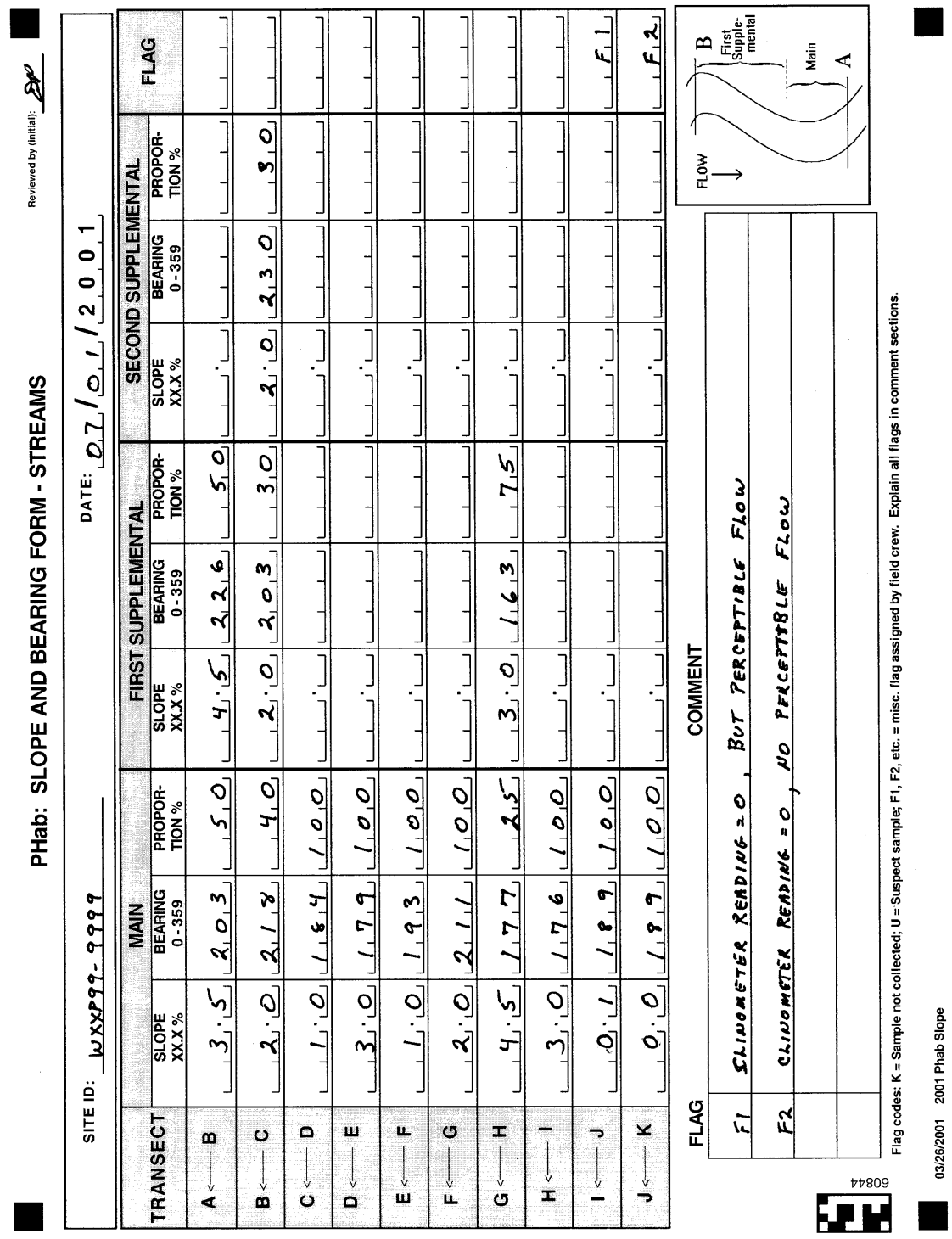
   - If you are backsiting from a supplemental point, record the slope (%) and proportion (%) of the stream segment that is included in the measurement in the appropriate "SUPPLEMENTAL" section of the Slope and Bearing Form.

5. Stand in the middle of the channel at upstream transect (or at a supplemental point), and site back with your compass to the middle of the channel at the downstream transect (or at a supplemental point). Record the bearing (degrees) in the "MAIN" section of the Slope and Bearing Form.

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

6. Retrieve the tripod from the downstream cross section station (or from the supplemental point) and set it up at the next upstream transect (or at a supplemental point) as described in Step 2.

7. When you get to each new cross-section transect (or to a supplementary point), backsight on the previous transect (or the supplementary point), repeat Steps 2 through 6 above.



PHab: SLOPE AND BEARING FORM - STREAMS

Reviewed by (initial): BP

SITE ID: WXWP99-9999 DATE: 07/01/2001

TRANSECT	MAIN			FIRST SUPPLEMENTAL			SECOND SUPPLEMENTAL			FLAG
	SLOPE XX.X %	BEARING 0-359	PROPORTION %	SLOPE XX.X %	BEARING 0-359	PROPORTION %	SLOPE XX.X %	BEARING 0-359	PROPORTION %	
A ⟵ B	3.5	203	50	4.5	226	50				
B ⟵ C	2.0	218	40	2.0	203	30	2.0	230	30	
C ⟵ D	1.0	184	100							
D ⟵ E	3.0	179	100							
E ⟵ F	1.0	193	100							
F ⟵ G	2.0	211	100							
G ⟵ H	4.5	177	25	3.0	163	75				
H ⟵ I	3.0	176	100							
I ⟵ J	0.1	189	100							F1
J ⟵ K	0.0	189	100							F2

COMMENT

FLAG

F1 SLIDOMETER READING = 0, BUT PERCEPTIBLE FLOW

F2 CLINOMETER READING = 0, NO PERCEPTIBLE FLOW

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

60844

03/26/2001 2001 Phab Slope

Figure 7-5. Slope and Bearing Form.

moving, record the slope as 0.1%. If the clinometer reading is 0% and water is not moving, record the slope as 0%.

For 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. 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 ("supplementary") slope and bearing points between a pair of cross-section transects if you do not have direct line-of-site along (and within) the channel between stations (see Figure 7-4). 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 supplementary measurements (i.e., do not "short-circuit" a meander bend). Mark these intermediate station 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 7-5). Note that the main slope and bearing observations are always downstream of supplemental observations. Similarly, first supplemental observations are always downstream of second supplemental observations.

7.5.2 Substrate Size and Channel Dimensions

Substrate size is one of the most important determinants of habitat character for fish and macroinvertebrates in streams. Along with bedform (e.g., riffles and pools), substrate influences the hydraulic roughness and consequently the range of water velocities in the channel. It also influences the size range of interstices that provide living space and cover for macroinvertebrates, salamanders, and sculpins. Substrate characteristics are often sensitive indicators of the effects of human activities on streams. Decreases in the mean substrate size and increases in the percentage of fine sediments, for example, may destabilize channels and indicate changes in the rates of upland erosion and sediment supply (Dietrich et al, 1989; Wilcock, 1998).

In the EMAP protocol, substrate size and embeddedness are evaluated at each of the 11 cross-section transects (refer to Figure 7-1) using a combination of methods adapted from those described by Wolman (1954), Bain et al. (1985), Platts et al. (1983), and Plafkin et al. (1989). Substrate size is evaluated also at 10 additional cross-sections located mid-way between each of the 11 regular transects (A-K). The basis of the protocol is a systematic selection of 5 substrate particles from each of 21 cross-section transects (Figure 7-6). In the process of measuring substrate particle sizes at each channel cross section, you also measure the wetted width of the channel and the water depth at each substrate sample point (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 7.4.1), 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 with dry channels, make measurements across the unvegetated portion of the channel.

The distance you record to the right bank is the same as the wetted channel width. (NOTE: this is the same value that is also recorded under "BANK MEASUREMENTS" on the same form [Section 7.5.3]). 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 7-7. Record these measurements on the Channel/Riparian Cross-section side of the field form, as shown in Figure 7-7. For the supplemental cross-sections midway between regular transects, 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 7-7) based on the middle dimension of its length, width, and depth. This “median” dimension determines the sieve size through which the particle can pass. Always distinguish “hardpan” from “fines”, coding hardpan as

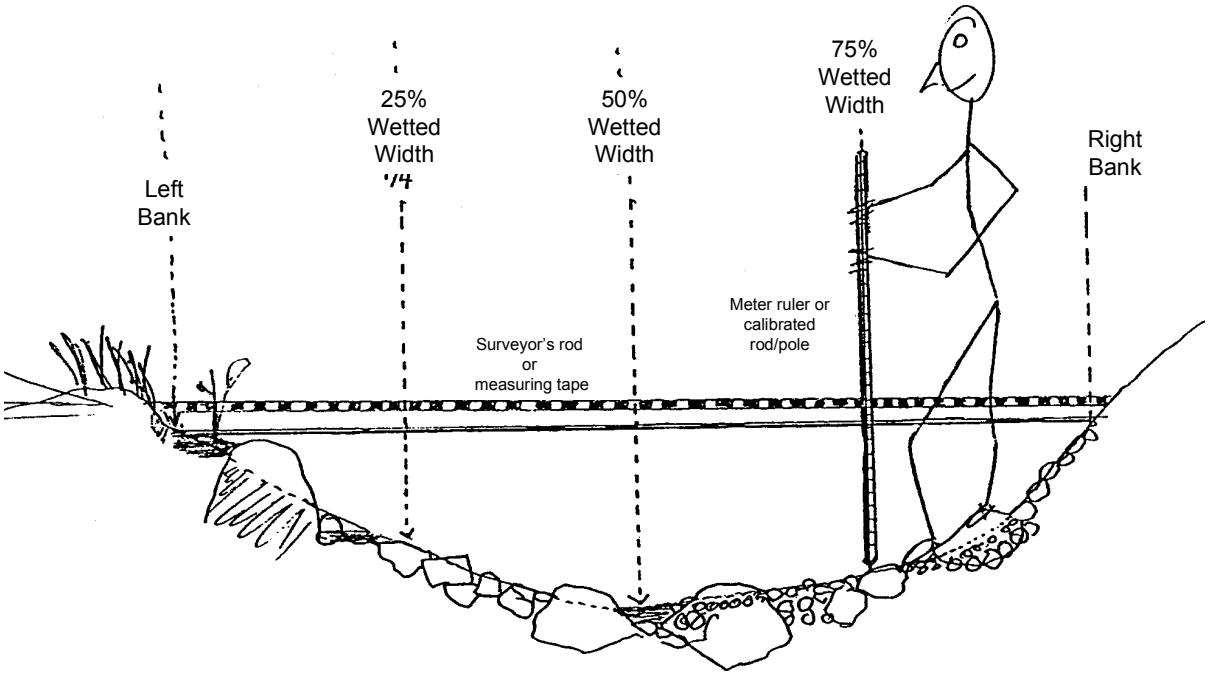


Figure 7-6. Substrate sampling cross-section.

"HP". Similarly, always distinguish concrete or asphalt from bedrock; denote these artificial substrates as "other" ("OT") and describe them in the comments section of the field data form. Code and describe other artificial substrates (including metal, tires, car bodies, etc.) in the same manner. When you record the size class as "OT" (other), assign an "F"-series flag on the field data form (Figure 7-7) and describe the substrate type in the comments section of the field form, as shown in Figure 7-2.

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 surface that is surrounded by (embedded in) sand or finer sediments on the stream bottom. By definition, the embeddedness of sand,

TABLE 7-7. 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 across the channel perpendicular to the flow, with the "zero" end at the left bank (facing downstream). If the channel is too wide for the rod, stretch the metric tape in the same manner.

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, 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		Firm, consolidated fine substrate
BL	Boulders	>250 to 4000	Basketball to car 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, but visible as particles - gritty between fingers
FN	Fines	<0.06	Silt Clay Muck (not gritty between fingers)
WD	Wood	Regardless of Size	Wood & other organic particles
OT	Other	Regardless of Size	Concrete, metal, tires, car bodies etc. (describe in comments)

5. Evaluate substrate embeddedness as follows at 11 transects A-K. For particles larger than sand, examine the surface for stains, markings, and algae. Estimate the average percentage embeddedness of particles in the 10 cm circle around the measuring rod. Record this value on the field data form. By definition, sand and fines are embedded 100 percent; bedrock and hardpan are embedded 0 percent.

6. Move successively to the next location along the cross section. Repeat steps 4 through 6 at each location. Repeat Steps 1 through 6 at each new cross section transect.

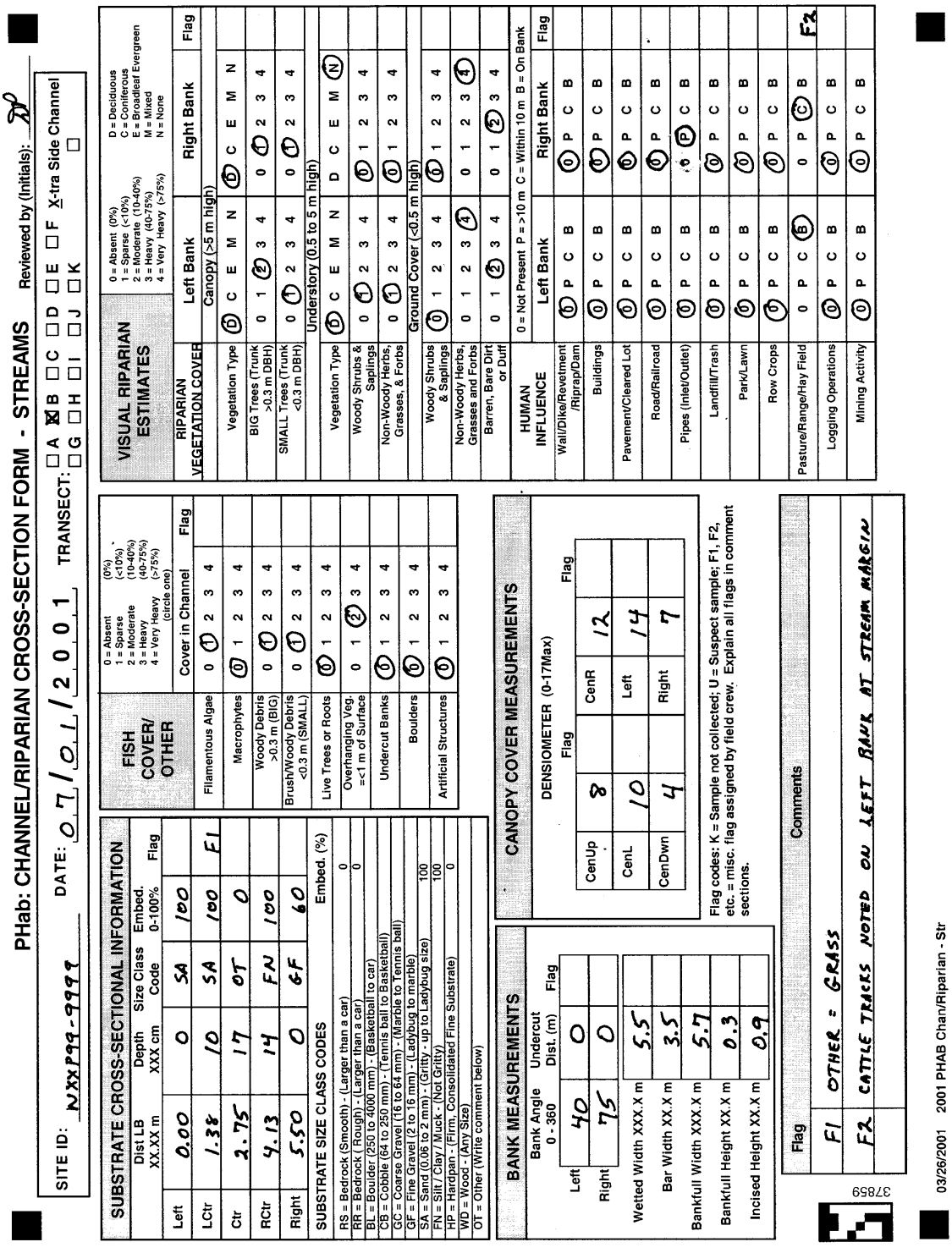


Figure 7-7. Channel/Riparian Cross-section Form.

silt, clay, and muck is 100 percent, and the embeddedness of hardpan and bedrock is 0 percent.

7.5.3 Bank Characteristics

The procedure for obtaining bank and channel dimension measurements is presented in Table 7-8. Data are recorded in the “Bank Measurements” section of the Channel/Riparian Cross-section Form as shown in Figure 7-7. Bank angle and bank undercut distance are determined on the left and right banks at each cross section transect. Other features include the wetted width of the channel (as determined in Section 7.5.2), 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 7-8. The “bankfull” or “active” channel is defined as the channel that is filled by moderate-sized flood events that typically occur every one or two years. Such flows do not generally overtop the channel banks to inundate the valley floodplain, and are believed to control channel dimensions in most streams.

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 channel height smaller than incision height (Figure 7-8). 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" is not necessarily an indication of recent incision. Examine both banks to more accurately determine channel downcutting.

Spotting the level of bankfull flow during baseflow conditions requires judgement and practice; even then it remains somewhat subjective. In many cases there is an obvious slope break that differentiates the channel from a relatively flat floodplain terrace higher than the channel. Because scouring and inundation from bankfull flows are often frequent enough to inhibit the growth of terrestrial vegetation, the bankfull channel may be evident by a transition from exposed stream sediments to terrestrial vegetation. Similarly, it may be identified by noting moss growth on rocks along the banks. Bankfull flow level may also be seen by the presence of drift material caught on overhanging vegetation. However, in years with large floods, this material may be much higher than other bankfull indicators. In these cases, record the lower value, flag it, and also record the height of drift material in the comments section of the field data form.

TABLE 7-8. PROCEDURE FOR MEASURING BANK CHARACTERISTICS

1. To measure bank angle, lay the surveyor's rod or your meter ruler down against the left bank (determined as you face downstream), with one end at the water's edge. Lay the clinometer on the rod, 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 degrees; undercut banks have angles >90 degrees approaching 180 degrees, and more gradually sloped banks have angles <90 degrees. To measure bank angles >90 degrees, turn the clinometer (which only reads 0 to 90 degrees) over and subtract the angle reading from 180 degrees.

2. If the bank is undercut, measure the horizontal distance of the undercutting to the nearest 0.01 m. Record the distance on the field data form. 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.

    - 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. Using the surveyor's rod as a guide while examining both banks, estimate (by eye) 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). Record this value in the "INCISED HEIGHT" field of the bank measurement section on the field data form.

5. Still holding the surveyor's rod as a guide, examine both banks to estimate 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.
    - 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. Record data for each transect on a separate field data form.

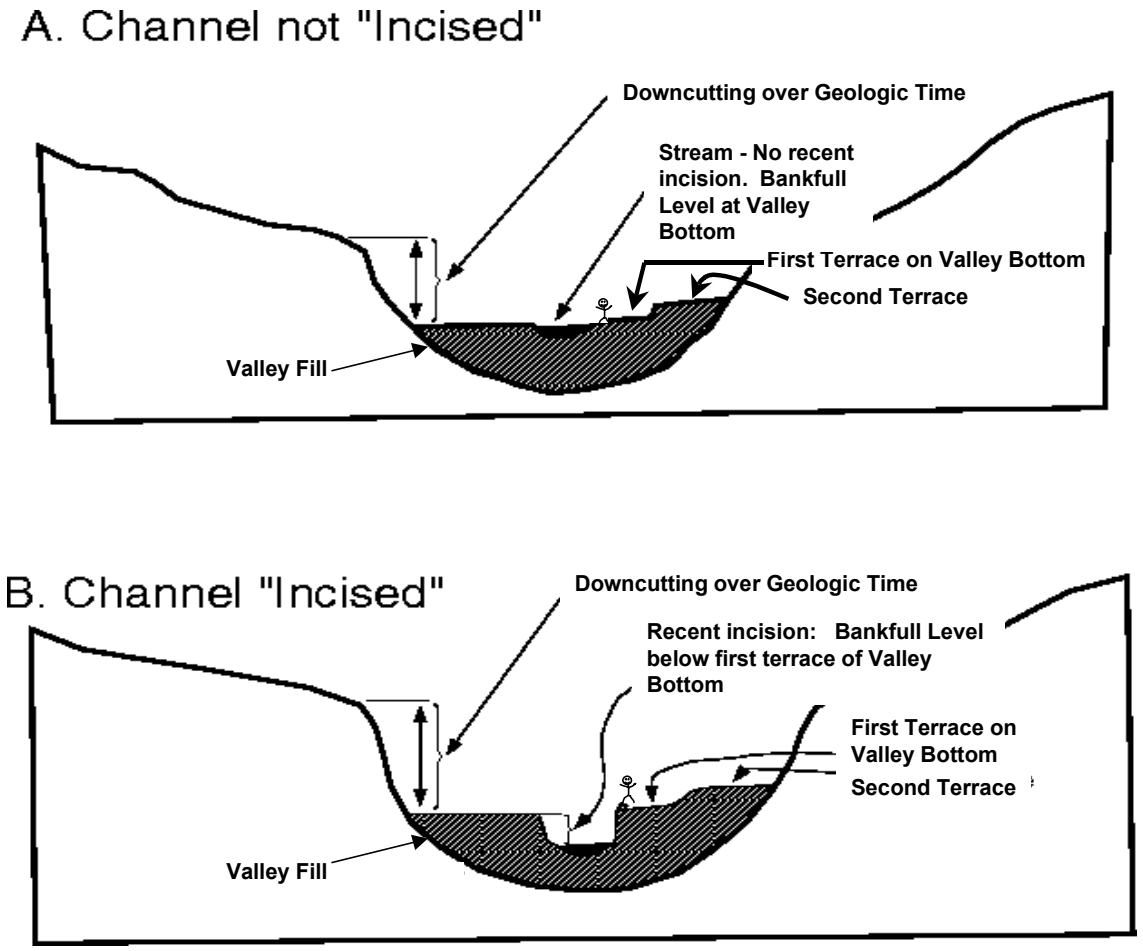


Figure 7-8. Schematic showing bankfull channel and incision for channels. (A) not recently incised, and (B) recently incised into valley bottom. Note level of bankfull stage relative to elevation of first terrace on valley bottom (Stick figure included for scale).

7.5.4 Canopy Cover Measurements

Riparian canopy cover over a stream is important not only in its role in moderating stream temperatures through shading, but also as an indicator of conditions that control bank stability and the potential for inputs of coarse and fine particulate organic material. Organic inputs from riparian vegetation become food for stream organisms and structure to create and maintain complex channel habitat.

Canopy cover over the stream is determined at each of the 11 cross-section transects. A Convex Spherical Densiometer (model B) is used (Lemmon, 1957). The densiometer must be taped exactly as shown in Figure 7-9 to limit the number of square grid intersections 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 four directions at mid-channel and one at each bank). The mid-channel measurements are used to estimate canopy cover over the channel. The two bank measurements complement your visual estimates of vegetation structure and cover within the riparian zone itself (Section 7.5.5), and are particularly important in wide streams, where riparian canopy may not be detected by the densiometer when standing midstream.

The procedure for obtaining canopy cover data is presented in Table 7-9. Densiometer measurements are taken at 0.3 m (1 ft) above the water surface, rather than at waist level, to (1) avoid errors because people differ in height; (2) avoid errors from standing in water of varying depths; and (3) include low overhanging vegetation more consistently in the estimates of cover. 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 7-9. 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 7-7).

7.5.5 Riparian Vegetation Structure

The previous section (7.5.4) 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

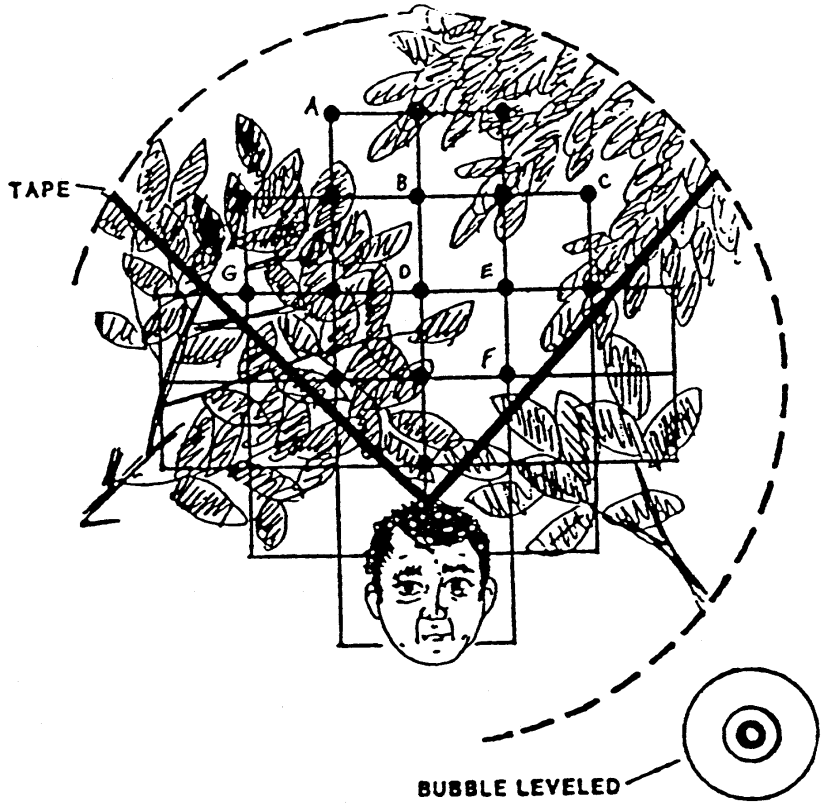


Figure 7-9. Schematic of modified convex spherical canopy densiometer (From Mulvey et al., 1992). In this example, 10 of the 17 intersections show canopy cover, giving a densiometer reading of 10. Note proper positioning with the bubble leveled and face reflected at the apex of the "V."

TABLE 7-9. 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. Hold the densiometer level 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. Repeat Steps 2 and 3 again, this time facing the bank while standing first at the left bank, then the right bank. Record the values in the "LFT" and "RGT" fields of the field data form.

7. Repeat Steps 1 through 6 at each cross-section transect. Record data for each transect on a separate field data form.

of riparian vegetation. These data are used to evaluate the health and level of disturbance of the stream corridor. They also provide an indication of the present and future potential for various types of organic inputs and shading.

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 7-1). 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 × 10 m riparian plot on each side of the stream (Figure 7-10). The riparian plot dimensions are estimated, not measured. On steeply sloping channel margins, the 10 m × 10 m plot boundaries are defined as if they were projected down from an aerial view. If the wetted channel is split by a mid-channel bar, the bank and riparian measurements are made at each side of the channel, not the bar.

Table 7-10 presents the procedure for characterizing riparian vegetation structure and composition. Figure 7-7 illustrates how measurement data are recorded in the "VISUAL RIPARIAN ESTIMATES" section of the Channel/Riparian Cross-section Form. Conceptually divide the riparian vegetation into three layers: a CANOPY LAYER (> 5 m high), an UNDERSTORY (0.5 to 5 m high), and a 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).

Before estimating the areal coverage of the vegetation layers, record the type of vegetation (Deciduous, Coniferous, broadleaf Evergreen, Mixed, or None) 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.

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 7-7). When rating vegetation cover types, 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"

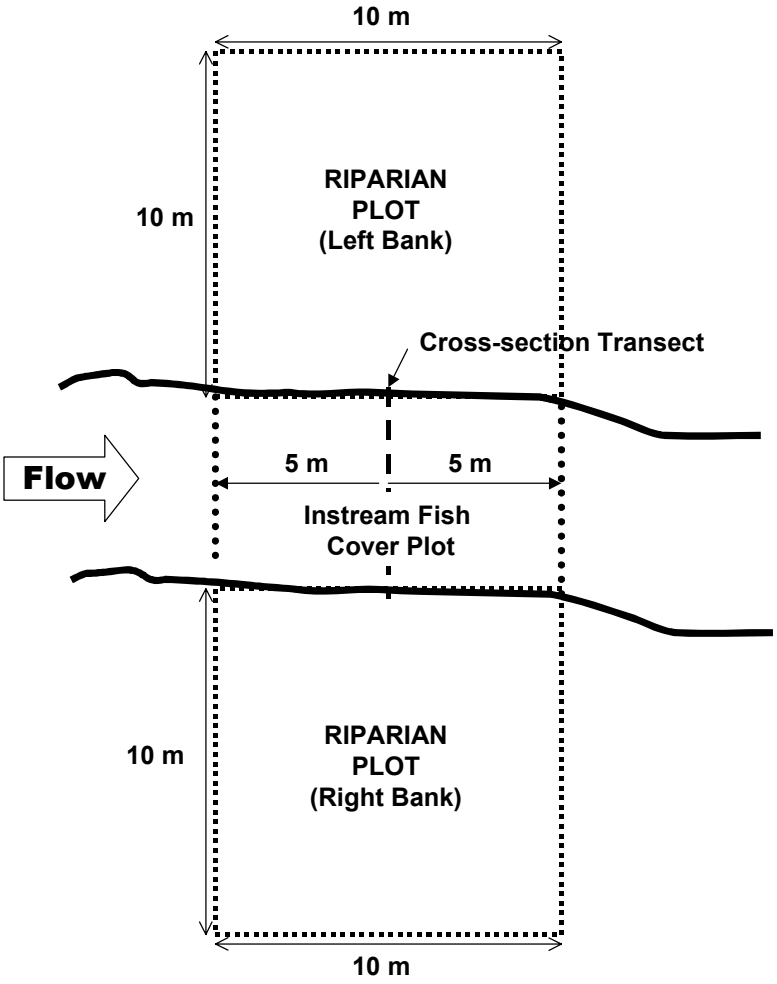


Figure 7-10. Boundaries for visual estimation of riparian vegetation, fish cover, and human influences.

TABLE 7-10. 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 three layers: a CANOPY LAYER (>5m 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 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. 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 vegetation type for the understory layer as described in Step 4 for the canopy layer.

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, using a separate field data form for each transect.

with all the remaining classes rated as either moderate ("2"), sparse ("1") or absent ("0"). Two heavy classes with 40-75% cover can both be rated "3".

7.5.6 Instream Fish Cover, Algae, and Aquatic Macrophytes

This portion of the EMAP physical habitat protocol is a visual estimation procedure that semi-quantitatively evaluates the type and amount of important types of cover for fish and macroinvertebrates. Alone and in combination with other metrics, this information is used to assess habitat complexity, fish cover, and channel disturbance.

The procedure to estimate the types and amounts of instream fish cover is outlined in Table 7-11. Data are recorded in the "Fish Cover/Other" section of the Channel /Riparian Cross-section Form as shown in Figure 7-7. Estimate the areal cover of all of the fish cover and other listed features that are in the water and on the banks 5 meters upstream and downstream of the cross-section (see Figure 7-10). The areal cover classes of fish concealment and other features are the same as those described for riparian vegetation (Section 7.5.5).

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 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 7.4]). "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. For ephemeral channels, estimate the proportional cover of these trees that is inundated during bankfull flows. "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 discarded (e.g., cars or tires) or purposefully placed for diversion, impoundment, channel stabilization, or other purposes.

TABLE 7-11. 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 the 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, recording data from each transect on a separate field data form.

7.5.7 Human Influence

The field evaluation of the presence and proximity of various important types of human land use activities in the stream riparian area is used in combination with mapped watershed land use information to assess the potential degree of disturbance of the sample stream reaches.

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 7-12. Relate your observations and proximity evaluations to the stream and riparian area within 5 m upstream and 5 m downstream from the station (Figure 7-10). 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 7-7. 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 site through another transect or its 10 m × 10 m riparian plot.

7.5.8 Riparian “Legacy” Trees and Invasive Alien Plants

The Riparian “Legacy” Tree protocol contributes to the assessment of “old growth” characteristics of riparian vegetation, and aids the determination of possible historic conditions and the potential for riparian tree growth. Follow the procedures presented in Table 7-13 to locate a legacy tree associated with each transect. Note that only one tree is identified at each transect. and that 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 7-13). Record this information, along with the estimated height, diameter at breast height (dbh), and distance from the wetted margin of the stream on the left hand column of the field form for Riparian “Legacy” Trees and Invasive Alien Plants (Figure 7-11).

TABLE 7-12. 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 lot (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 site 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.
O ("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, recording data for each transect on a separate field form.

TABLE 7-13. PROCEDURE FOR IDENTIFYING RIPARIAN LEGACY TREES AND ALIEN INVASIVE PLANT SPECIES

Legacy Trees:

- Beginning at Transect A, look upstream. Search both sides of the stream upstream to the next transect. Locate the largest riparian tree visible within 50m (or as far as you can see, if less) from the wetted bank.

- Classify this tree as 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 or Other Broadleaf Evergreen
16. Unknown or Other Conifer
17. Unknown or Other Deciduous

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

- Estimate the height of the 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.

(Continued)

TABLE 7-13 (Continued)

Alien Invasive Plants:

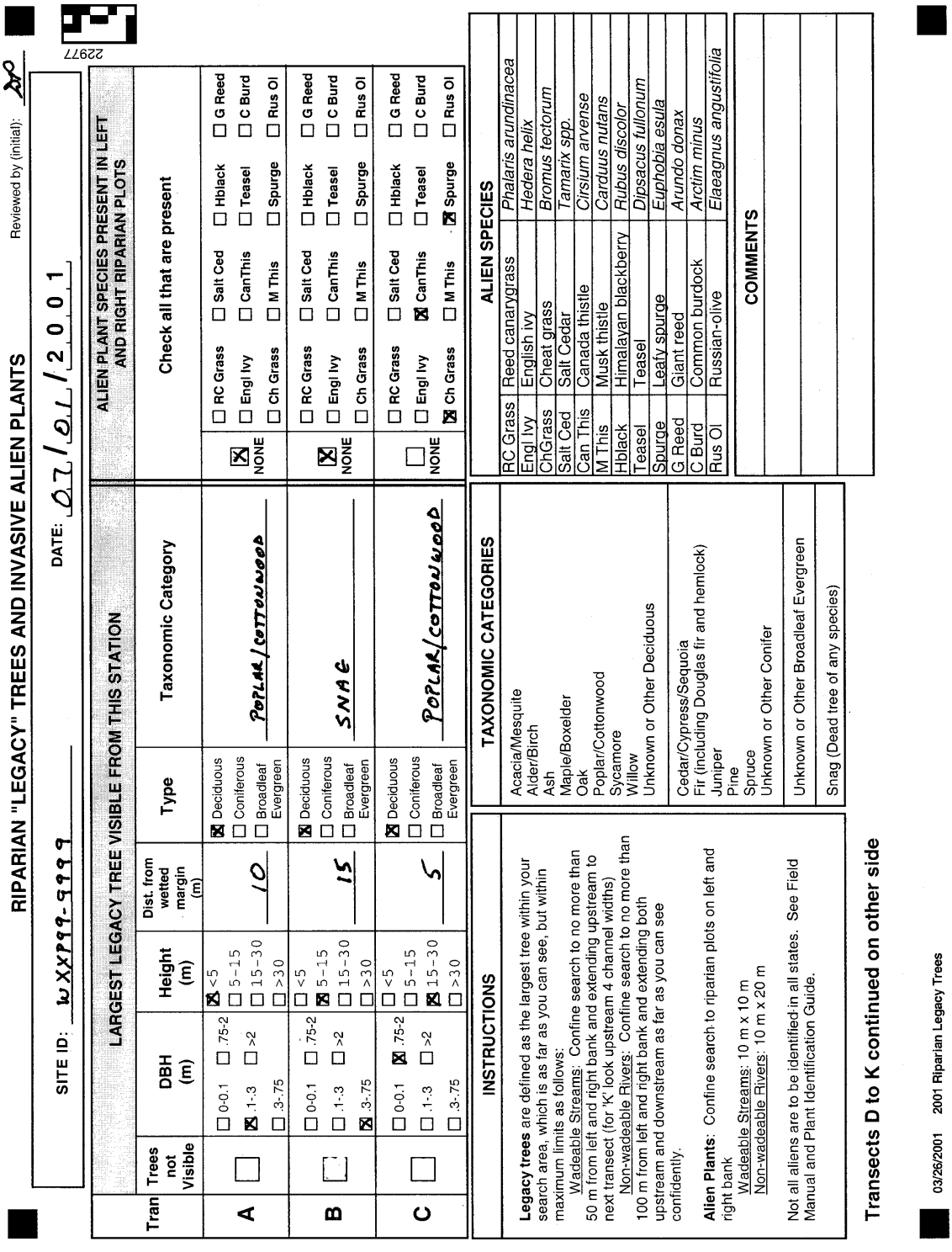
- Examine the 10m x 10m riparian plots on both banks for the presence of alien plant species. Look for those species from the following table that are listed as "target" species for your State.

Name to Check on Form	Common Name	Binomial: Genus species	CA	OR	WA	ID	ND	SD	WY	CO	AZ	UT	MT	NV
Can This	Canada Thistle	*Cirsium arvense*	X	X	X	X	X	X	X	X			X	
G Reed	Giant Reed	*Arundo donax*	X								X	X		X
Hblack	Himalayan Blackberry	*Rubus discolor*	X	X	X	X								
Spurge	Leafy Spurge	*Euphorbia esula*					X	X	X	X			X	
M This	Musk Thistle	*Carduus nutans*	X	X	X	X	X	X	X	X			X	
EngIvy	English Ivy	*Hedera helix*	X	X	X	X	X	X	X	X	X	X	X	X
RCGrass	Reed Canarygrass	*Phalaris arundinacea*	X	X	X	X								
Rus Ol	Russian-olive	*Elaeagnus angustifolia*	X				X	X	X	X	X	X	X	X
SaltCed	Salt Cedar	*Tamarix spp.*	X				X	X	X	X	X	X	X	X
ChGrass	Cheatgrass	*Bromus tectorum*	X	X	X	X	X	X	X	X	X	X	X	X
Teasel	Teasel	*Dipsacus fullonum*	X	X	X					X			X	
C Burd	Common Burdock	*Arctium minus*	X	X	X	X	X	X	X	X	X	X	X	X

X On the list for this state
Not on the list for this state

- Record the presence of any species listed for your State within the plot on either the left or right bank by marking the appropriate box(es) on the right hand column of the Riparian "Legacy" Trees and Invasive Alien Plants field form. If none of the species listed for your state is present in either of the plots at a given transect check the box labeled "None" for this transect.

- Repeat Steps 1 through 5 for each remaining transect (B through K). At transect "K", look upstream a distance of 4 channel widths) when locating the legacy tree.



RIPARIAN "LEGACY" TREES AND INVASIVE ALIEN PLANTS

Reviewed by (initial):

22977

SITE ID: WXPP9-9999 DATE: 07/01/2001

LARGEST LEGACY TREE VISIBLE FROM THIS STATION

Tran	Trees not Visible	DBH (m)	Height (m)	Dist. from wetted margin (m)	Type	Taxonomic Category
A	☐	☐ 0-0.1 ☐ .75-2 ☒ .1-.3 ☐ >2 ☐ .3-.75	☒ <5 ☐ 5-15 ☐ 15-30 ☐ >30	10	☒ Deciduous ☐ Coniferous ☐ Broadleaf ☐ Evergreen	POPLAR/COTTONWOOD
B	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☒ .3-.75	☐ <5 ☒ 5-15 ☐ 15-30 ☐ >30	15	☒ Deciduous ☐ Coniferous ☐ Broadleaf ☐ Evergreen	SNAG
C	☐	☐ 0-0.1 ☒ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☒ 15-30 ☐ >30	5	☒ Deciduous ☐ Coniferous ☐ Broadleaf ☐ Evergreen	POPLAR/COTTONWOOD

ALIEN PLANT SPECIES PRESENT IN LEFT AND RIGHT RIPARIAN PLOTS

Check all that are present

☒ NONE	☐ RC Grass	☐ Salt Ced	☐ Hblack	☐ G Reed
	☐ Engl Ivy	☐ CanThis	☐ Teasel	☐ C Burd
	☐ Ch Grass	☐ M This	☐ Spurge	☐ Rus Ol
☒ NONE	☐ RC Grass	☐ Salt Ced	☐ Hblack	☐ G Reed
	☐ Engl Ivy	☐ CanThis	☐ Teasel	☐ C Burd
	☐ Ch Grass	☐ M This	☐ Spurge	☐ Rus Ol
☐ NONE	☐ RC Grass	☐ Salt Ced	☐ Hblack	☐ G Reed
	☐ Engl Ivy	☒ CanThis	☐ Teasel	☐ C Burd
	☒ Ch Grass	☐ M This	☒ Spurge	☐ Rus Ol

INSTRUCTIONS

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

RC Grass	Reed canarygrass	*Phalaris arundinacea*
Engl Ivy	English ivy	*Hedera helix*
ChGrass	Cheat grass	*Bromus tectorum*
Salt Ced	Salt Cedar	*Tamarix spp.*
Can This	Canada thistle	*Cirsium arvense*
M This	Musk thistle	*Carduus nutans*
Hblack	Himalayan blackberry	*Rubus discolor*
Teasel	Teasel	*Dipsacus fullonum*
Spurge	Leafy spurge	*Euphobia esula*
G Reed	Giant reed	*Arundo donax*
C Burd	Common burdock	*Arctim minus*
Rus Ol	Russian-olive	*Elaeagnus angustifolia*

COMMENTS

Transects D to K continued on other side

03/26/2001 2001 Riparian Legacy Trees

Figure 7-11. Riparian "Legacy" Tree and Invasive Alien Plant Form (Page 1).

A trend of increasing concern along streams in many parts of the Western U.S. is the invasion of alien (non-native) tree, shrub, and grass species. A list of "target" invasive species has been prepared for each individual State, and is summarized as part of the procedure presented in Table 7-13. At each transect, the presence of listed invasive plant species within the 10 m x 10 m riparian plots on either bank is recorded on the Riparian "Legacy" Trees and Invasive Alien Plants field form (Figure 7-11). Note that the list of target plants varies from State to State. Record only the presence of plants which are targets in your state, even though you may observe other alien species in stream reaches within your state. Record an observation for each transect, even if none of the species listed for your state is present.

7.6 CHANNEL CONSTRAINT, DEBRIS TORRENTS, AND RECENT FLOODS

7.6.1 Channel Constraint

Whether natural or the result of human activities, the presence of immovable or difficult-to-move river margins constrains the degree to which the stream can form its own channel and banks through scour and deposition. The degree of channel constraint can strongly influence the quantity and quality of habitat for aquatic organisms. Constraint also influences the type and degree of stream channel adjustment to anthropogenic alterations in flow and sediment supply, or to direct channel manipulations (e.g., dredging, revetment, impoundment). To assess overall reach channel constraint, we have modified methods used by Oregon Department of Fish and Wildlife in their Aquatic Inventories (Moore et al., 1993).

After completing the thalweg profile and littoral-riparian 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 7-14. Record data on the Channel Constraint Assessment Form (Figure 7-12). First, classify the stream reach channel pattern as predominantly a **single** channel, an **anastomosing** channel, or a **braided** channel.

- **Anastomosing channels have relatively long major and minor channels** branching and rejoining in a complex network.

TABLE 7-14. 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 is 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.

  Anastomosing channels have relatively long major and minor channels branching and rejoining in a complex network.

  Braided channels also have multiple branching and rejoining channels, but these sub-channels are generally smaller, shorter, and more numerous, often with no obvious dominant channel.

- After classifying 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., road, 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 box on the field form.

CHANNEL CONSTRAINT AND FIELD CHEMISTRY - STREAMS/RIVERS

Reviewed by (initial): DP

SITE ID: WXXP99-9999 DATE: 07/01/2001

IN SITU MEASUREMENTS		Station ID: (Assume X-site unless marked)
		Comments
STREAM/RIVER DO mg/l: (optional)	5.3	
STREAM RIVER TEMP. (°C):	20.5	
TIME OF DAY:	11:25	

CHANNEL CONSTRAINT

CHANNEL PATTERN (Check 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 (Check 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 (Check 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 % ---> (0-100%)

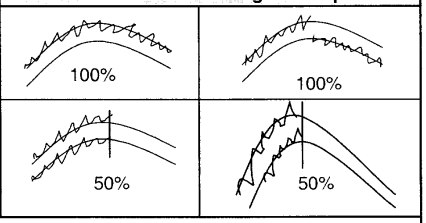
Bankfull width: 6 (m)

Valley width (Visual Estimated Average): 2000 (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: VALLEY WIDTH > 2000 Meters

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03/26/2001 2001 Chan Con/Fld Chem

Figure 7-12. Channel Constraint and Field Chemistry Form, showing data for channel constraint.

- **Braided channels also have multiple branching and rejoining channels**, but these sub-channels are generally smaller, shorter, and more numerous, often with no obvious dominant channel.

After classifying 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., road, dike, landfill, riprap, 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 visually estimate the average width of the valley floor. 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 box on the field form.

7.6.2 Debris Torrents and Recent Major Floods

Major floods are those that substantially overtop the banks of streams and occur with an average frequency of less than once every 5 years. Major floods may scour away or damage riparian vegetation on banks and gravel bars that are not frequently inundated. They typically cause movement of large woody debris, transport of bedload sediment, and changes in the streambed and banks through scouring and deposition. While they may kill aquatic organisms and temporarily suppress their populations, floods are an important natural resetting mechanism that maintains habitat volume, clean substrates, and riparian productivity.

Debris torrents, or lahars, differ from "conventional" floods in that they are flood waves of higher magnitude and shorter duration, and their flow is comprised 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, debris torrent flood waves can exceed 5 meters deep in small streams normally 3 meters wide and 15 cm deep. These torrents move boulders in excess of 1m diameter and logs >1m diameter and >10m 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 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 7-13). 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): DP

TORRENT EVIDENCE ASSESSMENT FORM - STREAMS

SITE ID: WXXP99-9999 DATE: 07/01/2001

TORRENT EVIDENCE	
Please X 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.
☐	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

5118

03/26/2001 2001 Torrrent Evidence

Figure 7-13. Torrent Evidence Assessment Form.

7.7 EQUIPMENT AND SUPPLIES

Figure 7-14 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. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

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EQUIPMENT AND SUPPLIES FOR PHYSICAL HABITAT

QTY.	Item	
1	Surveyor's telescoping leveling rod (round profile, metric scale, 7.5m extended)	
1	50-m fiberglass measuring tape & reel	
1	Hip chain (metric) for measuring reach lengths (Optional)	
1	Clinometer (or Abney level) with percent and degree scales.	
1	Lightweight telescoping camera tripod (necessary only if slope measurements are being determined by one person)	
2	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)	
1	Meter stick. Alternatively, a short (1-2 m) rod or pole (e.g., a ski pole) with cm markings for thalweg measurements, or the PVC pipe described for slope determinations can be marked in cm and used.	
1 roll ea.	Colored surveyor's plastic flagging (2 colors)	
1	Convex spherical canopy densiometer (Lemmon Mod.B), modified with taped "V"	
1	Bearing compass (Backpacking type)	
1 or 2	Fisherman's vest with lots of pockets and snap fittings. Used at least by person conducting the in-channel measurements to hold the various measurement equipment (densiometer, clinometer, compass, etc.). Useful for both team members involved with physical habitat characterization.	
2 pair	Chest waders with felt-soled boots for safety and speed if waders are the neoprene "stocking" type. Hip waders can be used in shallower streams.	
	Covered clipboards (lightweight, with strap or lanyard to hang around neck)	
	Soft (#2) lead pencils (mechanical are acceptable)	
11 plus extras	Channel/Riparian Cross-section & Thalweg Profile and Woody Forms	
1 plus extras	Slope and Bearing Form; Riparian Legacy Tree and Invasive Alien Plant Form; Channel Constraint Assessment Form; Torrent Evidence Form.	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for physical habitat characterization	

Figure 7-14. Checklist of equipment and supplies for physical habitat.

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NOTES

SECTION 8
PERIPHYTON

by
Brian H. Hill[1]

Periphyton are algae, fungi, bacteria, protozoa, and associated organic matter associated with channel substrates. 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 (e.g., Hill et al., 2000).

Modifications to the periphyton sampling procedures from the published EMAP-SW field operations manual (Hill, 1998) are summarized in Table 8-1. These modifications include increasing the number of transects where samples are collected, and reducing the number of composite samples from two to one per site. Also, pre-leached and pre-weighed glass-fiber filters are no longer required. Beginning in 2001, modifications include changing the containers used for chlorophyll and biomass samples, and eliminating the collection of the acid/alkaline phosphatase activity (APA) sample.

The "biomorphs" (refer to Figure 2-1) collect periphyton samples are collected at each transect at the same time as benthic macroinvertebrate samples (Section 11). Periphyton samples are collected from the dominant habitat type (erosional or depositional) located at each of the eleven cross-section transects (transects "A" through "K") established within the sampling reach (Section 4). At each stream, a single composite "index" sample of periphyton is prepared by combining individual transect samples. At the completion of the day's sampling activities, but before leaving the stream, four types of laboratory samples are prepared from each composite index sample.

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TABLE 8-1. SUMMARY OF CHANGES IN PERIPHYTON PROCEDURES FOR THE WESTERN PILOT STUDY

Changes from Hill (1998)
1. The number of transects where periphyton samples are collected is increased from nine to eleven.
2. A single composite sample is prepared from the 11 cross-section samples, rather than preparing separating samples for erosional and depositional transect samples.
3. The same glass-fiber filters are now used for both chlorophyll and biomass samples. Previously a pre-treated and pre-weighed filter was provided to use for the biomass sample.
Changes from Year 2000 Western Pilot Study Activities
1. Filters for chlorophyll and biomass are no longer wrapped in foil, but are folded and placed in separate 50-mL centrifuge tubes, which are labeled and then placed in a black plastic bag.
2. Samples for acid/alkaline phosphatase activity (APA) will not be collected in 2001.

8.1 SAMPLE COLLECTION

The general scheme for collecting periphyton samples from the sampling reach at each stream is illustrated in Figure 8-1. The procedure for collecting periphyton samples is presented in Table 8-2. At each transect, samples are collected from an assigned sampling point (left, center, or right). Sampling points at each transect may have been assigned when the sampling reach was laid out (Figure 8-1; refer also to Section 4; Table 4-3). If not, the sampling point at Transect "A" is assigned at random using a die or other suitable means (e.g., digital watch). Once the first sampling point is determined, either an erosional or depositional sample is collected, depending on whether the dominant habitat at the sampling point is flowing water (e.g., a riffle or run) or slack water (e.g., a pool). A composite sample for the reach is prepared by combining the individual transect samples as they are collected into a single plastic bottle. The volume of the composite sample are recorded on the Sample Collection Form as shown in Figure 8-2.

8.2 PREPARATION OF LABORATORY SAMPLES

Four different types of laboratory samples are prepared from the composite index sample: an ID/enumeration sample (to determine taxonomic composition and relative

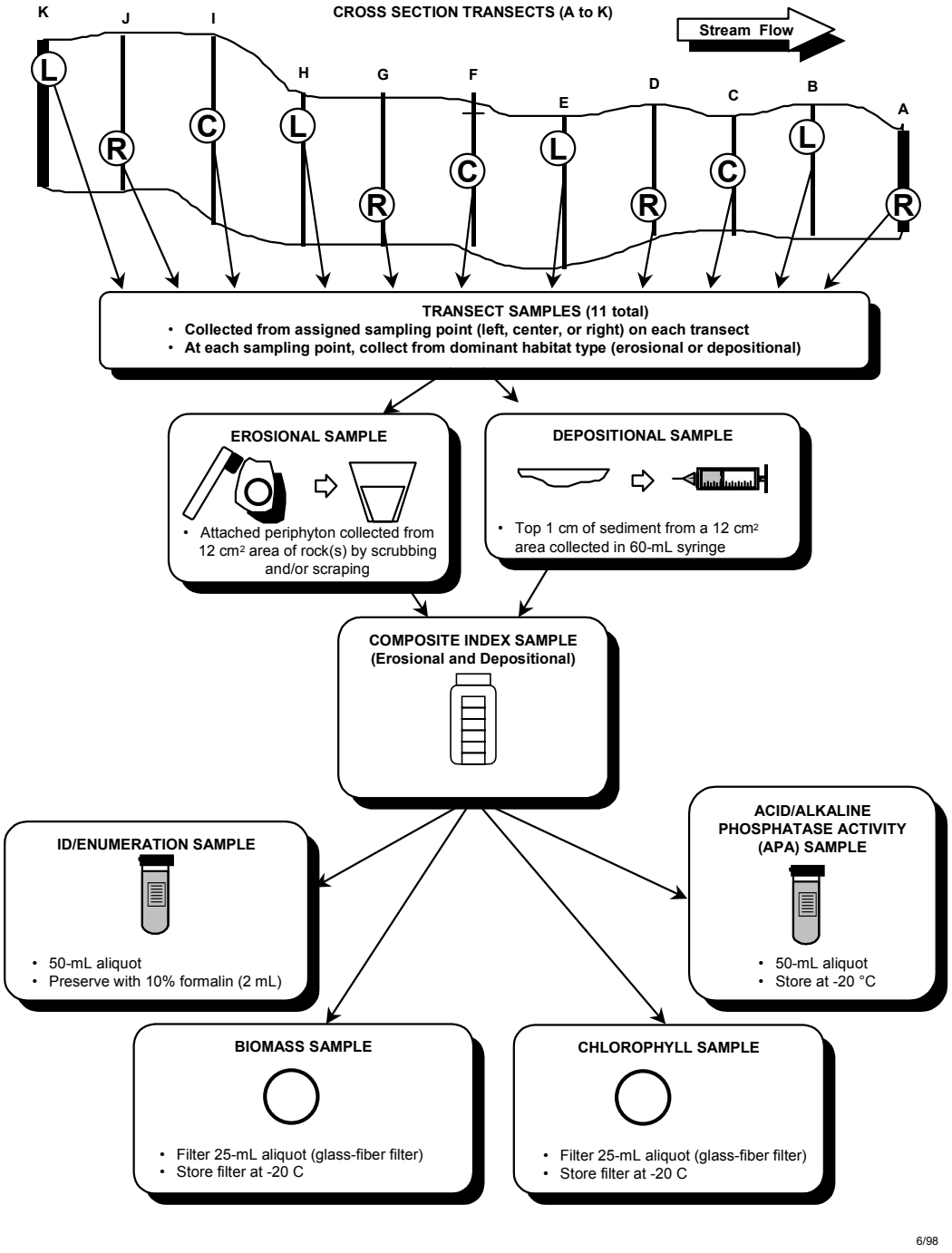


Figure 8-1. Index sampling design for periphyton.

TABLE 8-2. PROCEDURE FOR COLLECTING COMPOSITE INDEX SAMPLES OF PERIPHYTON

1. Starting with Transect "A", determine if the assigned sampling point (Left, Center, or Right) is located in an erosional (riffle) habitat or a slack water (pool) habitat. Collect a single sample at the point using the appropriate procedure in Step 2 below.

   If the sampling points were not assigned previously when laying out the sampling reach, proceed to Transect "A". Roll a die to determine if it is a left (L), center (C), or right (R) sampling point for collecting periphyton and benthic macroinvertebrate samples. A roll of 1 or 2 indicates L, 3 or 4 indicates C, and 5 or 6 indicates R (or use a digital wristwatch and glance at the last digit (1-3=L, 4-6=C, 7-9=R). Mark L, C, or R on the transect flagging. Assign sampling points at each successive transect in order as L, C, R after the first random selection.

2A. Erosional habitats:

   (1) Collect a sample of substrate (rock or wood) that is small enough (< 15 cm diameter) and can be easily removed from the stream. Place the substrate in a plastic funnel which drains into a 500-mL plastic bottle with volume graduations marked on it and labeled "PERIPHYTON."

   (2) 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.**

   (3) Fill a wash bottle with stream water. Using a minimal volume of water from this bottle, wash the dislodged periphyton from the rock, delimiter, and funnel into the 500-mL bottle.

2B. Depositional habitats:

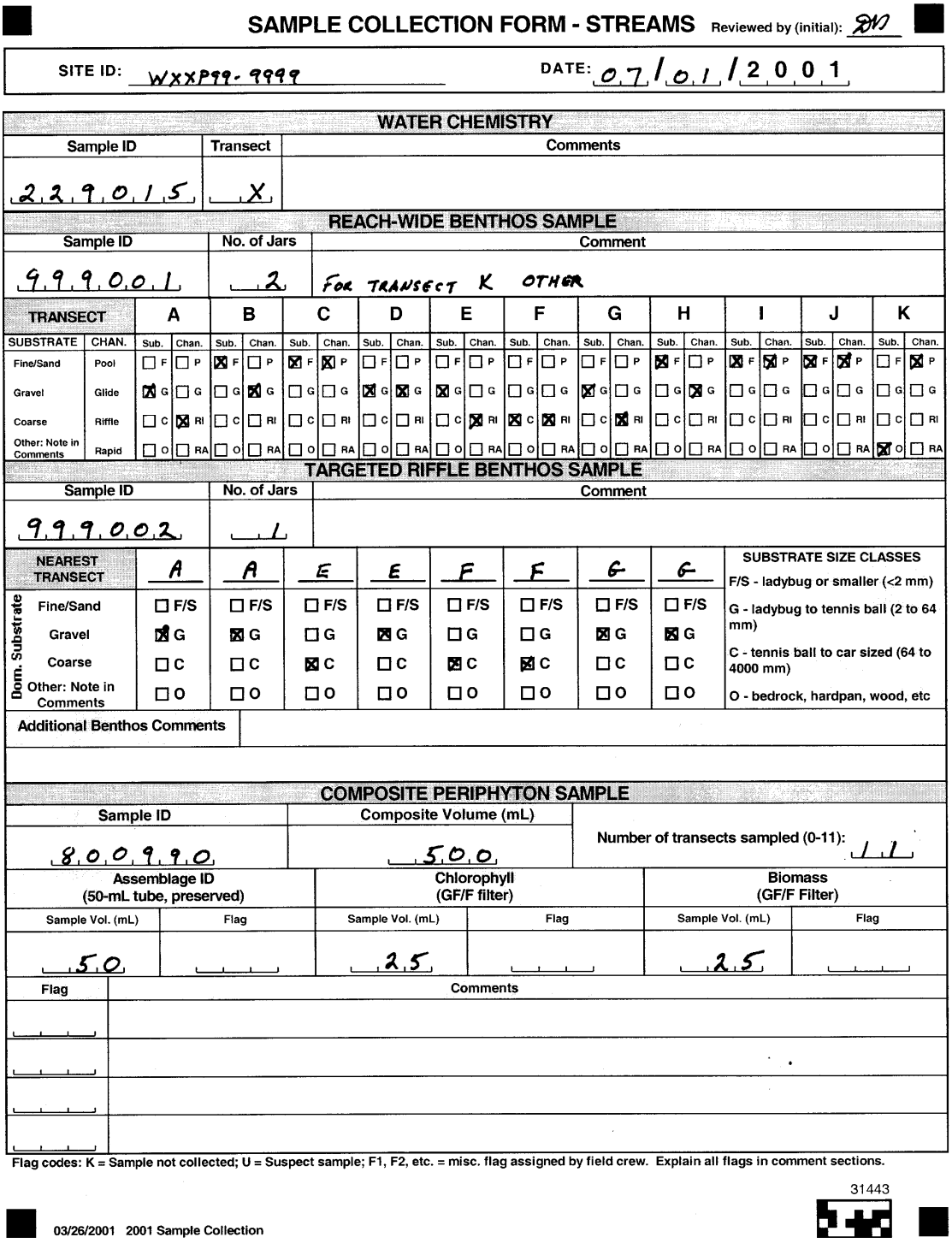
   (1) Use the area delimiter to confine a 12-cm^2 area of soft sediments.

   (2) Vacuum the top 1 cm of sediments from within the delimited area into a 60-mL syringe.

   (3) Empty the syringe into the 500-mL 'PERIPHYTON" bottle (combining it with samples collected from erosional habitats).

3. Repeat Steps 1and 2 for transects "B" through "K" to produce the composite index sample for the stream reach. Keep the collection bottle out of direct sunlight as much as possible to minimize degradation of chlorophyll.

4. After samples have been collected from all eleven transects, mix the 500-mL bottle thoroughly. Record the total estimated volume of the composite sample in the periphyton section of the Sample Collection Form. Also record the number of transects at which you obtained a periphyton sample.



SAMPLE COLLECTION FORM - STREAMS Reviewed by (initial): DW

SITE ID: WXXP99-9999 DATE: 07/01/2001

WATER CHEMISTRY

Sample ID	Transect	Comments
229015	X	

REACH-WIDE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999001	2	FOR TRANSECT K OTHER

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	☐ G	☐ G	☒ G	☐ G	☐ G	☒ G	☒ G	☒ G	☐ G	☐ G	☐ G	☒ G	☐ G	☐ G	☒ G	☐ G	☐ G	☐ G	☐ G	☐ G	☐ G
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: Note in Comments	Rapid	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☒ O	☐ RA

TARGETED RIFFLE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999002	1	

NEAREST TRANSECT	A	A	E	E	F	F	G	G
Dom. Substrate: Fine/Sand	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S
Gravel	☒ G	☒ G	☐ G	☒ G	☐ G	☐ G	☒ G	☒ G
Coarse	☐ C	☐ C	☒ C	☐ C	☒ C	☒ C	☐ C	☐ C
Other: Note in Comments	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O

SUBSTRATE SIZE CLASSES
F/S - ladybug or smaller (<2 mm)
G - ladybug to tennis ball (2 to 64 mm)
C - tennis ball to car sized (64 to 4000 mm)
O - bedrock, hardpan, wood, etc

Additional Benthos Comments

COMPOSITE PERIPHYTON SAMPLE

Sample ID	Composite Volume (mL)	Number of transects sampled (0-11):
800990	500	11

Assemblage ID (50-mL tube, preserved)		Chlorophyll (GF/F filter)		Biomass (GF/F Filter)	
Sample Vol. (mL)	Flag	Sample Vol. (mL)	Flag	Sample Vol. (mL)	Flag
50		25		25	

Flag	Comments

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

31443

03/26/2001 2001 Sample Collection

Figure 8-2. Sample Collection Form, showing data recorded for periphyton samples.

abundances), an acid/alkaline phosphatase activity (APA) sample, a chlorophyll sample, and a biomass sample (for ash-free dry mass). All the sample containers required for an individual stream should be sealed in plastic bags until use (see Section 3) to avoid external sources of contamination (e.g., dust, dirt, or mud) that are present at streamside.

A set of completed periphyton sample labels is shown in Figure 8-3. All labels in a set have the same sample ID number. Circle the appropriate type of sample (chlorophyll, biomass, etc.) on each label. Attach completed labels to the appropriate containers and cover with clear tape. When attaching the completed labels, do not cover any volume graduations and markings on the container.

8.2.1 ID/Enumeration Sample

Prepare the ID/Enumeration sample as a 50-mL aliquot from the composite index sample, following the procedure presented in Table 8-3. Preserve each sample with 2 mL of 10% formalin., observing all safety precautions associated with handling formalin solution. Record the ID number (barcode) from the sample container label and the total volume of the sample (50 mL) in the appropriate fields on the Sample Collection Form as shown in Figure 8-2. Explain any deviations from the 50 mL target volume in the comments field of the collection form. Store the preserved samples upright in a container containing absorbent material, according to the guidelines provided for handling formalin-preserved samples.

8.2.2 Acid/Alkaline Phosphatase Activity Sample

NOTE: The Acid/Alkaline Phosphatase Activity Sample will not be prepared in 2001.

The Acid/alkaline phosphatase activity (APA) sample is prepared as a 50-mL subsample of the composite index in the same manner as the ID/enumeration sample (Table 8-3). No field treatment (i.e., filtration, preservation) of the APA sample is necessary. Complete a label for each sample as shown in Figure 8-3 and affix it to a 50-mL centrifuge tube. Record the ID number (barcode), and the volume of the subsample on the Sample Collection Form (Figure 8-2). 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 (Section 3).

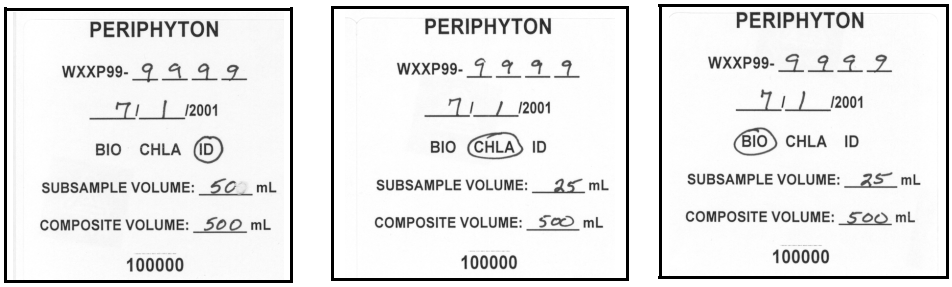


Figure 8-3. Completed set of periphyton sample labels.

8.2.3 Chlorophyll Sample

Prepare a chlorophyll sample by filtering a 25-mL aliquot of the composite index sample through a glass fiber filter (Whatman GF/F or equivalent). The procedure for preparing chlorophyll samples is presented in Table 8-4. 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. The filtration apparatus is illustrated in Figure 8-4. Rinse the filtration chamber with deionized water each day before use at the base site and then seal in a plastic bag until use at the stream (see Section 3). 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 no exceed 7 pounds per square inch (psi) to avoid rupturing cells. If the vacuum pressure exceeds 7 psi, 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.

After filtering the sample, fold the filter paper in half and place it in a 50-mL centrifuge tube. Complete a sample label (Figure 8-3) and check it to ensure that all written information is complete and legible. Affix the label to the centrifuge tube and cover it completely with a strip of clear tape. Record the sample ID number printed on the label on the Sample Collection Form (Figure 8-2). Make sure the volume recorded on each sample label matches the corresponding volume recorded on the Sample Collection Form. Record a flag and provide comments on the Sample Collection Form if there are any problems in collecting the sample or if conditions occur that may affect sample integrity. Store each

TABLE 8-3. PREPARATION OF ID/ENUMERATION AND ACID/ALKALINE PHOSPHATASE ACTIVITY SAMPLES FOR PERIPHYTON

NOTE: THE APA sample is not prepared in 2001.

1. Thoroughly mix the bottle containing the composite index sample.

2. Prepare a barcoded sample label. Circle the sample type ("ID" or "APA") on the label. Record the volume of the subsample (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.

3. Record the sample ID number (barcode) of the label and the total volume of the composite index sample on the Sample Collection Form. Explain any deviations from the target volume in the comments section of the form.

4. Rinse a 60-mL syringe with deionized water.

5. Withdraw 50 mL of the composite index sample into the syringe. Place the contents of the syringe sample into the labeled 50-mL centrifuge tube.

6. Repeat Steps 1 through 5 for the acid/alkaline phosphatase activity (APA) sample. **Note that in 2001, the APA sample is not collected.**

7. A. For the ID sample (wearing gloves and safety glasses), use a syringe or bulb pipette to add 2 mL of 10% formalin solution to the ID sample tube. Cap the tube tightly and seal with plastic electrical tape. Shake gently to distribute the preservative.

   B. Do NOT add preservative to the APA sample. Cap the tube tightly and seal with plastic electrical tape. Place in a cooler

8. Record the volume of each sample (typically 50 mL; exclude the volume of preservative added to the ID sample) on the Sample Collection Form. Double check that the volumes recorded on the collection form matches the total volume recorded on the corresponding sample labels.

TABLE 8-4. PROCEDURE FOR PREPARING CHLOROPHYLL AND BIOMASS SAMPLES FOR PERIPHYTON

1. Mix the composite index sample bottle thoroughly.

2. Using clean forceps, place a glass fiber filter on the filter holder. 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.

4. Rinse the sides of the filter funnel and the filter with a small volume of deionized water.

5. Rinse a 25-mL or 50-mL graduated cylinder three times with small volumes of deionized water. Measure 25 mL (±1 mL) of sample into the graduated cylinder.

    - NOTE: For composite samples containing fine sediment, allow grit to settle before pouring the sample into the graduated cylinder.

6. Pour the 25-mL aliquot into the filter funnel, replace the cap, and pump the sample through the filter using the hand pump. **NOTE: Vacuum pressure from the pump should not exceed 7 psi to avoid rupture of fragile algal cells.**

    - 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 side folded in on itself. Place the folded filter paper into a 50-mL centrifuge tube.

9. 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 and store in darkness.

10. Record the sample ID number (barcode) of the label and the total volume of the composite index sample on the Sample Collection 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.

11. Place the plastic bag containing the centrifuge tube into a portable freezer, a cooler containing dry ice, or between two sealed plastic bags of ice in a cooler.

12. Rinse the filter funnel, filter holder, filter chamber, and graduated cylinder thoroughly with deionized water.

13. Repeat Steps 1 through 12 to prepare the biomass sample, completing a periphyton sample label for biomass and recording sample information in the biomass section of the Sample Collection Form.

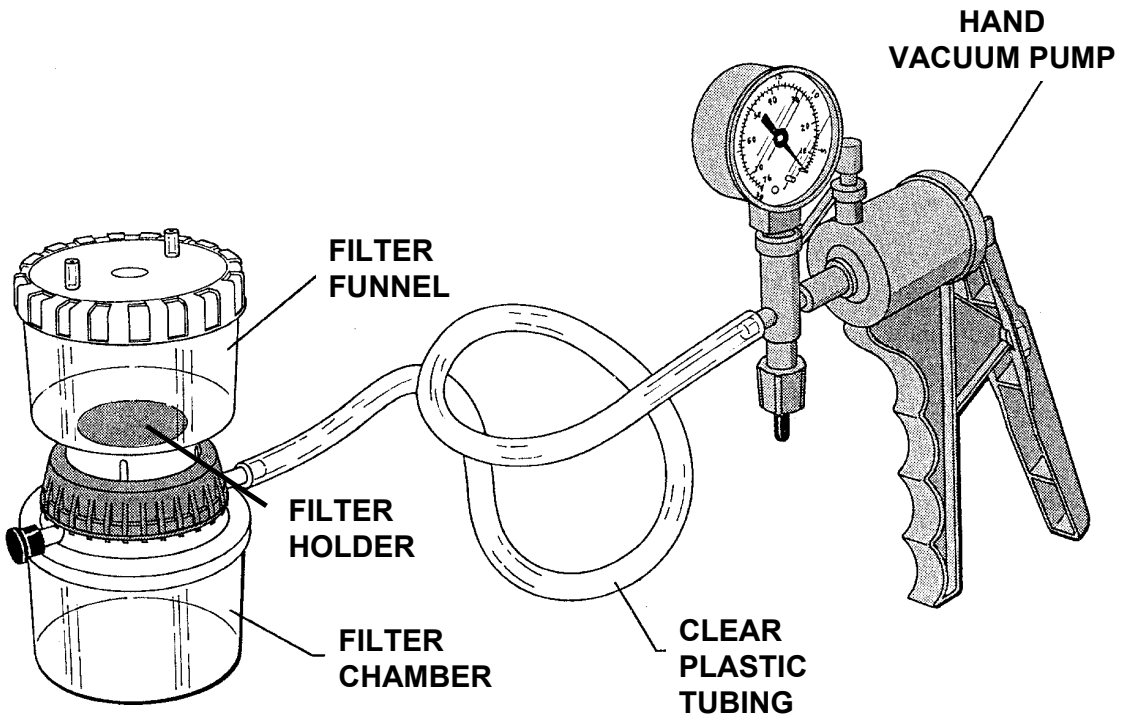


Figure 8-4. Filtration apparatus for preparing chlorophyll and biomass subsamples for periphyton. Modified from Chaloud et al. (1989).

centrifuge tube in a self-sealing plastic bag in darkness. Store the sample frozen until shipment to the laboratory (Section 3).

8.2.4 Biomass Sample

Prepare the biomass sample from a 25-mL aliquot of the composite index sample. Prepare the sample according to the procedure presented in Table 8-4. As with the chlorophyll sample, it is important to measure the volume to be filtered accurately (±1 mL). Rinse the filter chamber components (Figure 8-4) and the graduated cylinder thoroughly between the chlorophyll and biomass samples with deionized water.

After filtering the sample, complete a biomass sample label as shown in Figure 8-3. Check the sample label to ensure that all written information is complete and legible. Affix the label to the 50-mL centrifuge tube and cover it completely with clear tape. Record the sample ID number printed on the label and the volume filtered on the Sample Collection Form as shown in Figure 8-2. Make sure the information recorded on each sample label matches the corresponding values recorded on the Sample Collection Form. Record a flag and provide comments on the Sample Collection Form if there are any problems in collecting the sample or if conditions occur that may affect sample integrity. Store each labeled sample container frozen until shipment to the laboratory (Section 3).

8.3 EQUIPMENT AND SUPPLIES

Figure 8-5 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 stream. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

8.4 LITERATURE CITED

Chaloud, D.J., J.M. Nicholson, B.P. Baldigo, C.A. Hagley, and D.W. Sutton. 1989. *Handbook of Methods for Acid Deposition Studies: Field Methods for Surface Water Chemistry*. EPA 600/4-89-020. U.S. Environmental Protection Agency, Washington, D.C.

Hill, B.H. 1998. Periphyton. Pp. 199-132 IN: J.M. Lazorchak, D.J. Klemm, and D.V. Peck (eds.). *Environmental Monitoring and Assessment Program-Surface Waters: Field Operations and Methods for Measuring the Ecological Condition of Wadeable Streams.* EPA/620/R-94/004F. U.S. Environmental Protection Agency, Washington, DC.

Hill, B.A., A.T. Herlihy, P.R. Kaufmann, R.J. Stevenson, F.H. McCormick, and C. Burch-Johnson. 2000. Use of periphyton assemblage data as an index of biotic integrity. *Journal of the North American Benthological Society* 19(1):50-67.

EQUIPMENT AND SUPPLIES FOR PERIPHYTON

QTY.	Item	
1	Large funnel (15-20 cm diameter)	
1	12-cm^2 area delimiter (3.8 cm diameter PVC pipe, 3 cm tall)	
1	Stiff-bristle toothbrush with handle bent at 90^o angle	
1	1-L wash bottle labeled “STREAM WATER”	
1	1-L wash bottle labeled for and containing deionized water	
1	500-mL plastic bottle (with volume markings) for composite index samples, labeled “PERIPHYTON COMPOSITE SAMPLE”	
1	35-60 mL catheter-tipped plastic syringe	
4	50-mL screw-top centrifuge tubes	
1 box	Glass-fiber filters for chlorophyll and biomass samples	
1 pair	Forceps for filter handling.	
1	25-mL or 50-mL graduated cylinder	
1	Filtration unit, including filter funnel, cap, filter holder, and receiving chamber	
1	Hand-operated vacuum pump and clear plastic tubing	
1	Small lightproof plastic bags for storing chlorophyll and biomass samples	
2	Self-sealing plastic bags for chlorophyll and biomass samples	
4 mL	10% formalin solution for ID/Enumeration samples	
1	Small syringe or bulb pipette for dispensing formalin	
1 pair	Chemical-resistant gloves for handling formalin	
1 pair	Safety glasses for use when handling formalin	
2 sets	Sample labels (4 per set) with the same barcode ID number	
1	Sample Collection Form for stream	
	Soft (#2) lead pencils for recording data on field forms	
	Fine-tipped indelible markers for filling out sample labels	
1 pkg.	Clear tape strips for covering labels	
1	Portable freezer, cooler with dry ice, or cooler with bags of ice to store frozen samples	
1 copy	Field operations and method manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for peri-phyton	

Figure 8-5. Checklist of equipment and supplies for periphyton.

NOTES

NOTES

SECTION 9
SEDIMENT COMMUNITY METABOLISM

Sediment community metabolism is not being considered for the Western Pilot Study.

SECTION 10
SEDIMENT TOXICITY

Sediment toxicity is not being considered for the Western Pilot Study.

SECTION 11
BENTHIC MACROINVERTEBRATES

by
Donald J. Klemm[1], James M. Lazorchak[1], and Philip A. Lewis[1, 2]

Benthic invertebrates inhabit the sediment or live on the bottom substrates of streams. Benthic macroinvertebrate assemblages in streams reflect overall biological integrity of the benthic community. Monitoring these assemblages is useful in assessing the status of the water body and detecting trend in ecological condition. Benthic communities 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 community (e.g., Klemm et al., 1990). Because many macroinvertebrates have relatively long life cycles of a year or more and are relatively immobile, macroinvertebrate community structure is a function of present or past conditions.

The EMAP-SW benthic macroinvertebrate protocol is intended to evaluate the biological integrity of wadeable streams in the United States for the purpose of detecting stresses on community structure and assessing the relative severity of these stresses. It is based on the "Rapid Bioassessment Protocol III - Benthic Macroinvertebrates" published by the U.S. Environmental Protection Agency (Plafkin et al., 1989; Barbour et al., 1999) and adopted for use by many states. Modifications to the previously published protocol for EMAP-Surface Waters (Klemm et al., 1998) for the EMAP-SW Western Pilot Study are summarized in Table 11-1. The two man kick net procedure of the Rapid Bioassessment Protocol (RBP) is replaced in the EMAP-SW protocol with a D-frame kick net modified for use by one person (Figure 11-1). Note this net is modified from that used in previous EMAP and R-EMAP projects (Klemm et al., 1998), in terms of frame type, mesh size, and dimensions. The modified protocol still requires only one person and is the preferred macroinvertebrate collecting method for streams with flowing water (a second person is often used for water safety and to keep time and record information on the field forms).

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[2] Current address: 1037 Wylie Road, RR #2, Seaman, OH 45679.

TABLE 11-1. SUMMARY OF BENTHIC MACROINVERTEBRATE PROTOCOL CHANGES FOR THE EMAP-SW WESTERN PILOT STUDY

Modifications from Klemm et al. (1998)

14. Two types of samples are collected, a "targeted riffle" sample and a "reach-wide" sample, replacing the "riffle/run" and "pool/glide" samples. The targeted riffle sample is focused on riffle areas only (i.e., if no riffle areas are present, the sample is not collected). The reach-wide sample is collected from transects spaced throughout the reach, as was described in the previous published protocol.

15. The number of kick samples in the targeted riffle sample is 8. The number of kick samples in the reach-wide sample is increased from 9 (transects B through J) to 11 (Transects A through K).

16. Each sample type is prepared as a single composite sample. For the reach-wide sample, all kick samples are combined into a single composite sample, replacing the "RIFFLE" composite and the "POOL" composite samples.

17. The sampling device is changed from a rectangular kick net to a D-Frame design. Mesh size is decreased from 595 Fm to 500 Fm. Net width is decreased from 18 in to 12 in (50 cm to 30 cm).

18. The area of each kick sample is reduced from 0.5 m^2 to 0.09 m^2 (1 ft^2).

19. The time for each kick sample is increased from 20 seconds to 30 seconds.

Modifications from Western Pilot Study Year 2000 Activities:

1. Clarified procedure for collecting at sampling points choked with vegetation.

2. Field form has been modified to record the microhabitat type (pool, glide, riffle, rapid) for each reachwide kicknet sample.

The "biomorphs" (refer to Figure 2-1) collect kick net samples for benthic macroinvertebrates at sampling points located on each cross-section transect (termed the "reach-wide" sample) and from riffle habitats located within the sampling reach (termed the "targeted riffle" sample). Kick net samples are collected at the same time as periphyton samples (Section 8). Samples collected as part of the "reach-wide" sample are combined into a single composite for the stream reach, while those collected for the "targeted riffle" sample are combined into a separate composite.

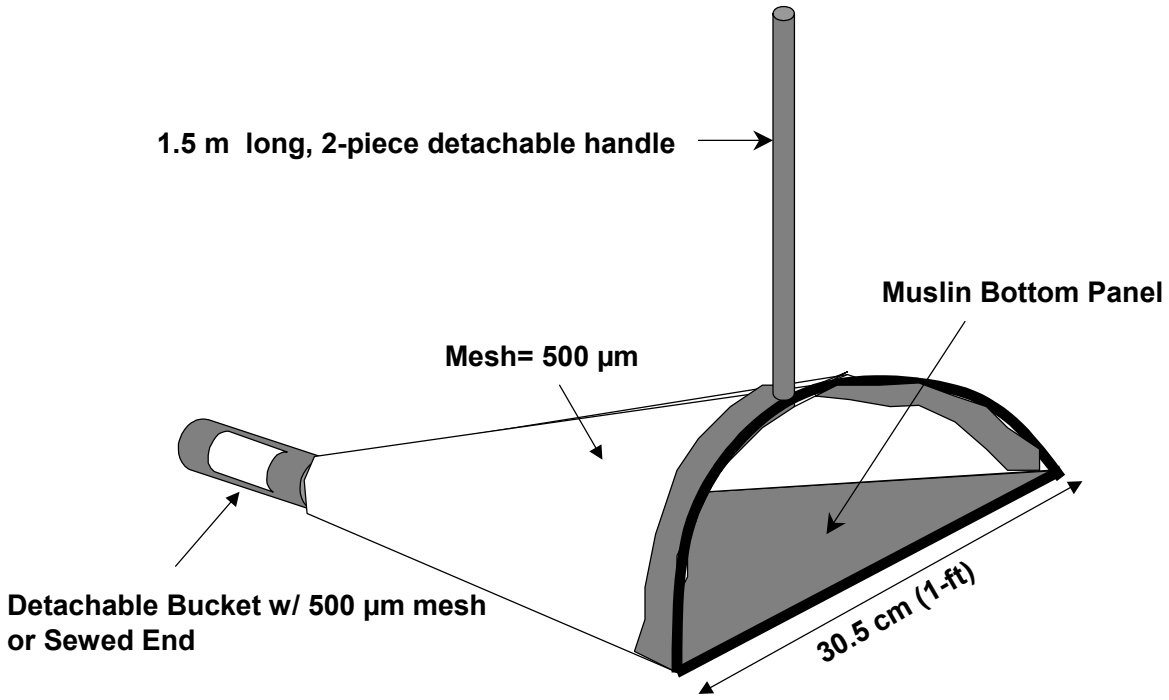


Figure 11-1. Modified D-frame kick net. (Not drawn to scale.)

11.1 SAMPLE COLLECTION

11.1.1 Reach-Wide Sample

The index sample design for collecting the reach-wide sample for benthic macroinvertebrates is shown in Figure 11-2. This design was used in the EMAP and R–EMAP stream studies in the mid-Atlantic region (refer to Section 1 for project descriptions).

A kick net sample is collected from each of the eleven cross-section transects (Transects “A” through “K”) at an assigned sampling point (Left, Center, or Right). These

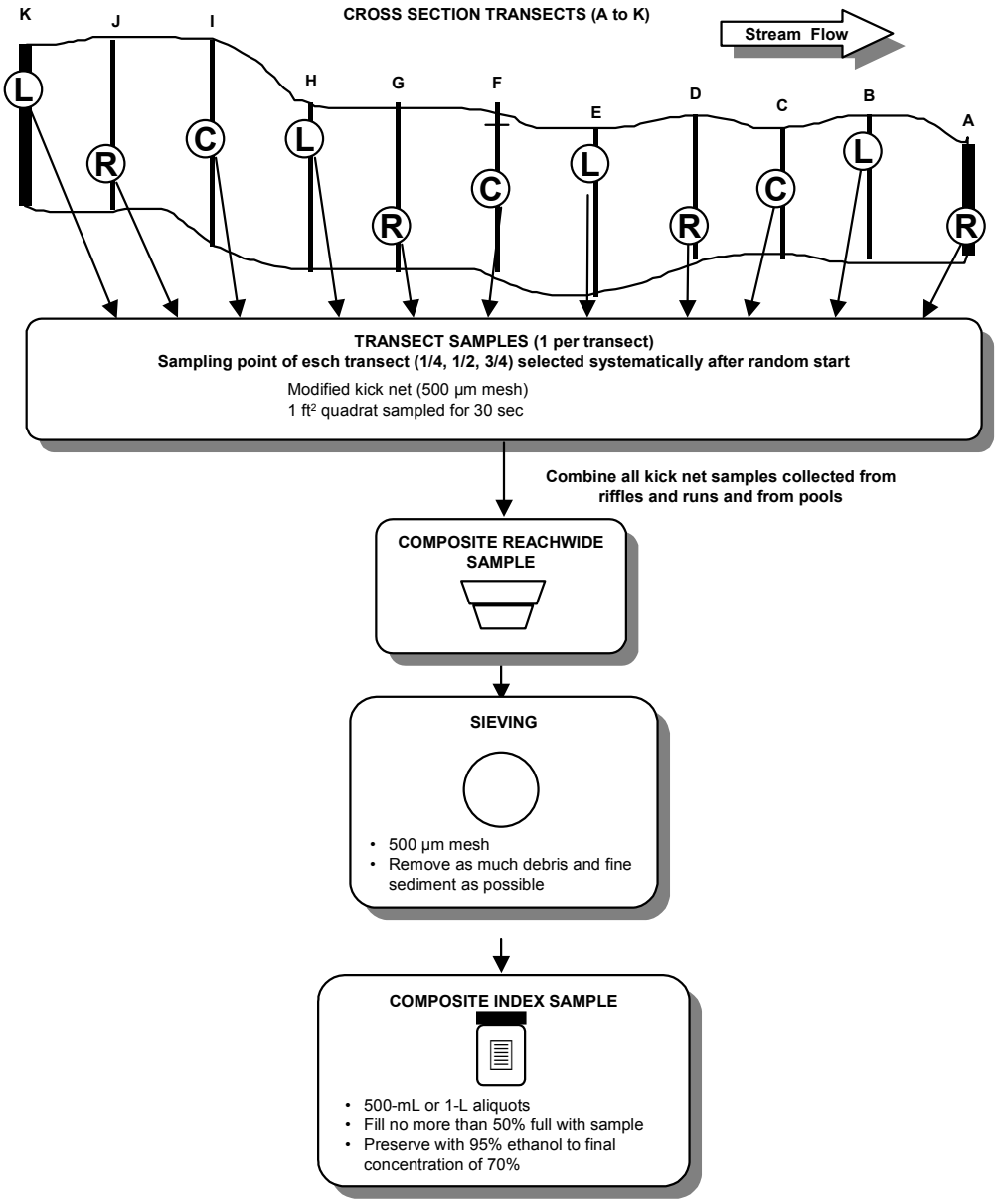


Figure 11-2. Index sampling design for benthic macroinvertebrate reachwide sample.

points may have been assigned when the sampling reach was laid out (Figure 11-2; refer also to Section 4; Table 4-3). If not, the sampling point at Transect "A" is assigned at random using a die or other suitable means (e.g., digital watch). Once the first sampling point is determined, points at successive transects are assigned in order (Left, Center, Right). These are the same sampling points as those used for periphyton samples (Section 8). 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 otherwise unsafe to wade, select an alternate sampling point on the transect at random.

The procedure for collecting a kick net sample at each transect is described in Table 11-2. 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 kick net sample collected on the Sample Collection Form as shown in Figure 11-3. As you proceed upstream from transect to transect, combine all kick net samples into a bucket or similar container labeled "REACH-WIDE", regardless of whether they were collected using the "riffle/run" or "pool/glide" procedure.

If it is impossible to sample at the sampling point with the modified kick net following either procedure, spend about 30 seconds hand picking a sample from about 0.09 m^2 (1 ft^2) of substrate at the sampling point. For vegetation-choked sampling points, sweep the net through the vegetation for 30 seconds. Place the contents of this hand-picked sample into the "REACH-WIDE" sampling container.

11.1.2 Targeted Riffle Sample

Figure 11-4 illustrates the sampling design for the targeted riffle sample. Table 11-3 presents the procedure for selecting individual sampling points within the available riffle macrohabitat units located within the sampling reach. Note that if the total available area of riffle habitat is less than 8 ft^2 (i.e., such that 8 non-overlapping kick net samples cannot be collected), do not collect a targeted riffle sample. There may be stream reaches where more than one 1 ft^2 kick net sample is collected from a single riffle unit. The objective for

TABLE 11-2. PROCEDURE TO COLLECT KICK NET SAMPLES FOR THE REACH-WIDE COMPOSITE SAMPLE

1. At each cross-section transect, beginning with Transect "A", locate the assigned sampling point (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 the point on the transect nearby.

2. Attach the 4-ft handle to the kick net. Make sure that the handle is on tight or the net may become twisted in a strong current, causing the loss of part of the sample.

3. Determine if there is sufficient current in the area at the sampling point to fully extend the net. If so, classify the habitat as "riffle/run" and proceed to Step 4. If not, use the sampling procedure described for "pool/glide" habitats (Step 9).

   NOTE: If the net cannot be used, spend 30 seconds hand picking a sample from about 0.09 m^2 (1 ft^2) of substrate at the sampling point. For vegetation-choked sampling points, sweep the net through the vegetation within a 0.09 m^2 (1 ft^2) quadrat for 30 seconds. Place the contents of this hand-picked sample into the "REACH-WIDE" sampling container. Go to Step 15.

Riffle/Run Habitats:

4. With the net opening facing upstream, position the net quickly and securely on the stream bottom to eliminate gaps under the frame. Avoid large rocks that prevent the sampler from seating properly on the stream bottom.

   NOTE: If there is too little water to collect the sample with the kick net, randomly pick up 10 rocks from the riffle and pick and wash the organisms off them into a bucket labeled "REACH-WIDE" which is half-full of water.

5. Holding the net in position on the substrate, visually define a rectangular quadrat that is one net width wide and one net width long upstream of the net opening. The area within this quadrat is ~0.09 m^2 (1 ft^2). Alternatively, place a wire frame of the correct dimensions in front of the net to help delineate the quadrat to be sampled.

6. Hold the net in place with your knees. Check the quadrat for heavy organisms, such as mussels and snails. Remove these organisms from the substrate by hand and place them into the net. Pick up any loose rocks or other larger substrate particles in the quadrat. Use your hands or a small scrub brush to dislodge organisms so that they are washed into the net. Scrub all rocks that are golf ball-sized or larger and which are over halfway into the quadrat. Large rocks that are less than halfway into the sampling area are pushed aside. After scrubbing, place the substrate particles outside of the quadrat.

7. Keep holding the sampler securely in position. Start at the upstream end of the quadrat, vigorously kick the remaining finer substrate within the quadrat for 30 seconds (use a stopwatch).

8. 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.

9. Go to Step 14.

(continued)

TABLE 11-2. (Continued)

Pool/Glide habitats:

10. Visually define a rectangular quadrat that is one net width wide and one net width long at the sampling point. The area within this quadrat is ~0.09 m^2 (1 ft^2). Alternatively, lay a wire frame of the correct dimensions in front of the net at the sampling point to help delineate the quadrat.

11. Inspect the stream bottom within the quadrat for any heavy organisms, such as mussels and snails. Remove these organisms by hand and place them into the net or into a bucket labeled "REACH-WIDE". Pick up any loose rocks or other larger substrate particles within the quadrat and hold them in front of the net. Use your hands (or a scrub brush) to rub any clinging organisms off of rocks or other pieces of larger substrate (especially those covered with algae or other debris) into the net. After scrubbing, place the larger substrate particles outside of the quadrat.

12. 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 Fm mesh size to collect the organisms from the water in the same way the net is used in larger pools.

13. After 30 seconds, remove the net from the water with a quick upstream motion to wash the organisms to the bottom of the net.

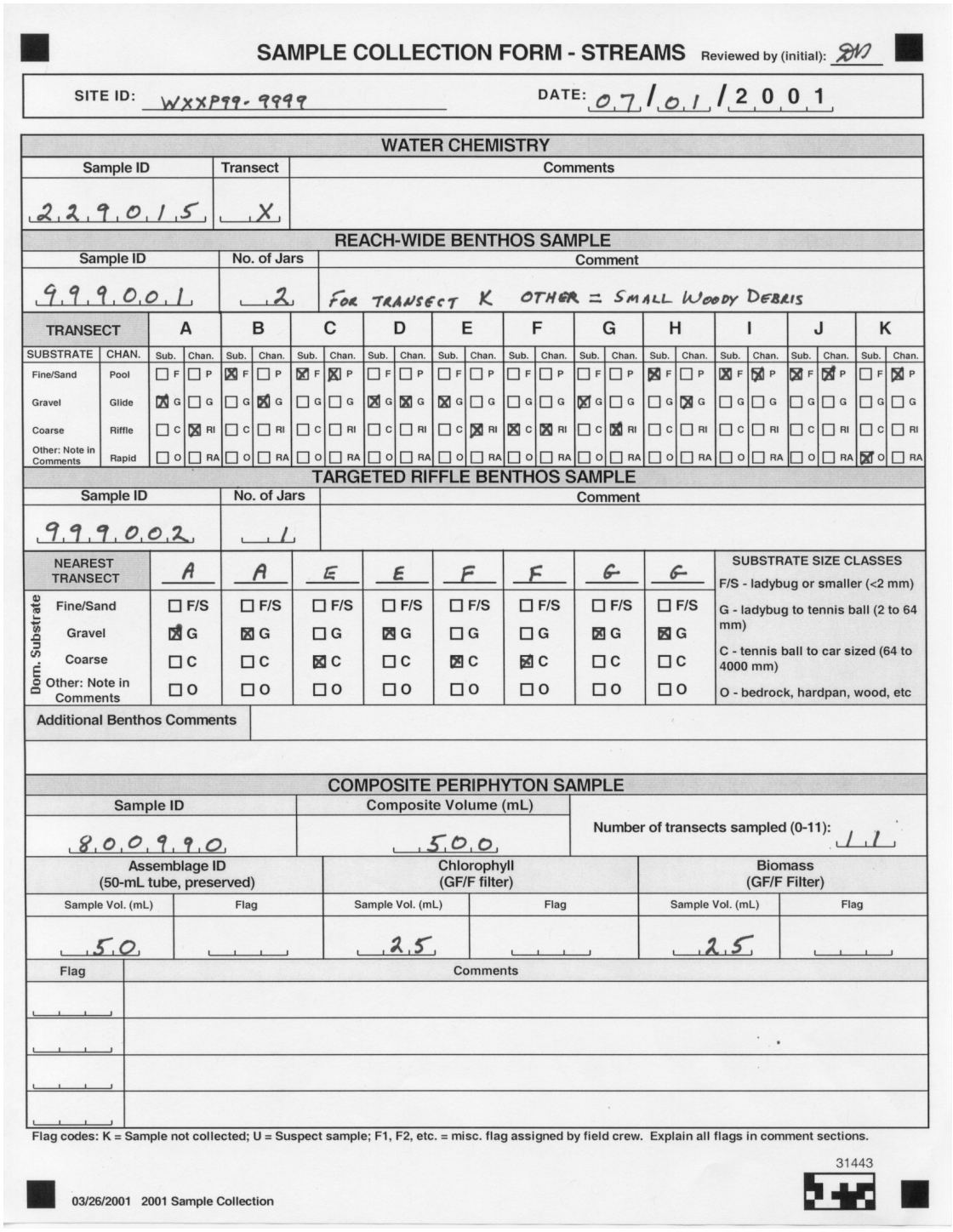
All samples:

14. Invert the net into a plastic bucket marked "REACH-WIDE" and transfer the sample. Inspect the net for any residual organisms clinging to the net and deposit them into the "REACH-WIDE" bucket. Use watchmakers' 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 any organisms.

15. Place an "X" in the appropriate substrate type box for the transect on the Sample Collection Form.

    Fine/sand: not gritty (silt/clay/muck < 0.06 mm diam.) to gritty, up to ladybug sized (2 mm diam.)
    Gravel: fine to coarse gravel (ladybug to tennis ball sized; 2 mm to 64 mm diam.)
    Coarse: Cobble to boulder (tennis ball to car sized; 64 mm to 4000 mm)
    Other: 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.

16. Thoroughly rinse the net before proceeding to the next sampling location. Proceed upstream to the next transect (including Transect K, the upstream end of the sampling reach) and repeat Steps 1 through 9. Combine all kick net samples from riffle/run and pool/glide habitats into the "REACH-WIDE" bucket.



SAMPLE COLLECTION FORM - STREAMS Reviewed by (initial): BW

SITE ID: WXXP99-9999 DATE: 07/01/2001

WATER CHEMISTRY

Sample ID	Transect	Comments
229015	X	

REACH-WIDE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999001	2	FOR TRANSECT K OTHER = SMALL WOODY DEBRIS

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	☐ G	☐ G	☒ G	☐ G	☐ G	☒ G	☒ G	☒ G	☐ G	☐ G	☐ G	☒ G	☐ G	☐ G	☒ G	☐ G	☐ G	☐ G	☐ G	☐ G	☐ G
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: Note in Comments	Rapid	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☒ O	☐ RA

TARGETED RIFFLE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999002	1	

NEAREST TRANSECT	A	A	E	E	F	F	G	G
Dom. Substrate: Fine/Sand	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S
Gravel	☒ G	☒ G	☐ G	☒ G	☐ G	☐ G	☒ G	☒ G
Coarse	☐ C	☐ C	☒ C	☐ C	☒ C	☒ C	☐ C	☐ C
Other: Note in Comments	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O

SUBSTRATE SIZE CLASSES
F/S - ladybug or smaller (<2 mm)
G - ladybug to tennis ball (2 to 64 mm)
C - tennis ball to car sized (64 to 4000 mm)
O - bedrock, hardpan, wood, etc

Additional Benthos Comments

COMPOSITE PERIPHYTON SAMPLE

Sample ID	Composite Volume (mL)	Number of transects sampled (0-11):
800990	500	11

Assemblage ID (50-mL tube, preserved)		Chlorophyll (GF/F filter)		Biomass (GF/F Filter)	
Sample Vol. (mL)	Flag	Sample Vol. (mL)	Flag	Sample Vol. (mL)	Flag
50		25		25	

Flag	Comments

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

31443

03/26/2001 2001 Sample Collection

Figure 11-3. Sample Collection Form (page 1), showing information for the reach-wide and targeted riffle benthic macroinvertebrate samples.

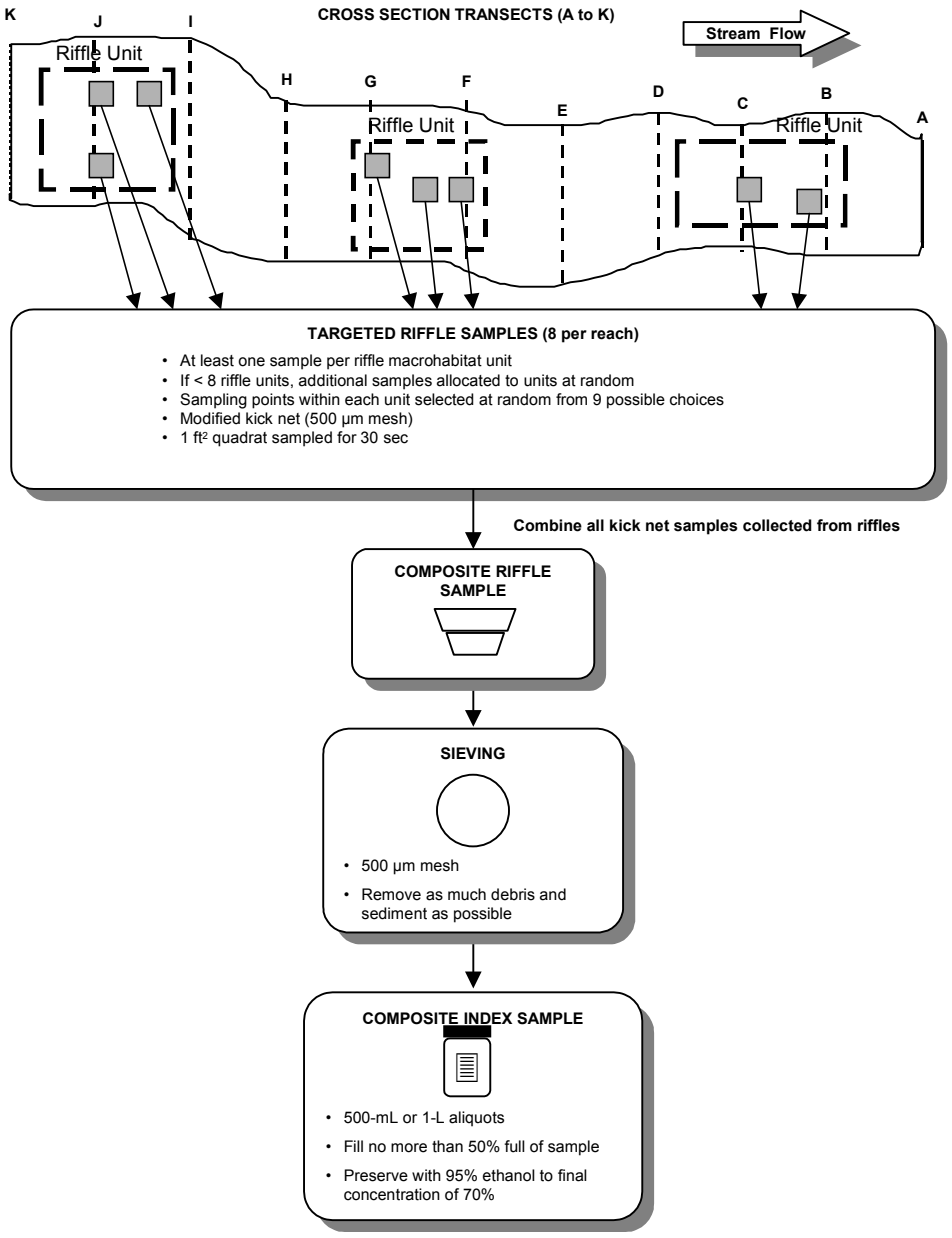


Figure 11-4. Index sampling design for benthic macroinvertebrate targeted riffle sample.

TABLE 11-3. LOCATING SAMPLING POINTS FOR KICK NET SAMPLES: TARGETED RIFFLE SAMPLE

1. Before sampling, survey the stream reach to estimate visually the total number (and area) of riffle macrohabitat "units" contained in the defined stream reach. To be considered as a unit, the area of the riffle must be greater than 1 ft^2.

   A. Do not sample poorly represented habitats. If the reach contains less than 8 ft^2 of riffle macrohabitat, then do not collect a targeted riffle sample.

   B. If the reach contains more than one distinct riffle macrohabitat units but less than eight, allocate the eight sampling points among the units so as to spread the effort throughout the reach as much as possible. You may need to collect more than one kick sample from a given riffle unit.

   C. If the number of riffle macrohabitat units is greater than eight, skip one or more habitat units at random as you work upstream, again attempting to spread the sampling points throughout the reach.

2. Begin sampling at the most downstream riffle unit, and sample units as they are encountered to minimize instream disturbance.

3. At each unit, exclude "margin" habitats by constraining the potential sampling area. Margin habitats are edges, along the channel margins or upstream or downstream edges of the riffle macrohabitat unit. Define a core area for each riffle unit as the central portion, visually estimating a "buffer" strip circumscribing the identified unit. In some cases, the macrohabitat unit may be so small that it will not be feasible to define a core area and avoid an edge.

4. Visually lay out the core area of the unit sampled into 9 equal quadrats (i.e., a 3 × 3 grid). For each macrohabitat type, select a quadrat for sampling at random from the following list of locations (right and left are determined as you look downstream):

   Lower right quadrat
   Lower center quadrat
   Lower left quadrat
   Right center quadrat
   Center quadrat
   Left center quadrat
   Upper right quadrat.
   Upper center quadrat.
   Upper left quadrat

5. Collect the kick sample in the center of the selected quadrat as described in Table 11-4.

6. If a second sample is required from a single macrohabitat unit, select additional quadrats at random from the list in Step 4.

TABLE 11-4. COLLECTING A KICK NET SAMPLE FROM WADEABLE STREAMS FOR THE TARGETED RIFFLE COMPOSITE SAMPLE

1. Beginning at the most downstream riffle unit within the sampling reach, locate the sampling point within the macrohabitat unit as described in Table 11-3.

2. Position the kick net quickly and securely on the stream bottom so as to eliminate gaps between the frame and the stream bottom. If necessary, rotate the net so the narrower side is against the bottom.

3. Hold the sampler firmly in position on the substrate. Define a quadrat immediately upstream from the mouth of the net having a width equal to the width of the net frame (total area = 0.09 $m^{2)}$.

4 Hold the net in place with your knees and pick up any loose rocks or other larger substrate particles in the quadrat. Use your hands or a small scrub brush to dislodge organisms so that they are washed into the net. Scrub all rocks that are golf ball-sized or larger and which are over halfway into the quadrat. Large rocks that are less than halfway into the sampling area are pushed aside. After scrubbing, place the substrate particles outside of the quadrat.

6. Keep holding the sampler securely in position. Start at the upstream end of the quadrat, vigorously kick the remaining finer substrate within quadrat for 30 seconds (use a stopwatch).

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. Invert the net into a plastic bucket marked "TARGETED RIFFLE" and transfer the sample. Inspect the net for any residual organisms clinging to the net and deposit them into the "TARGETED RIFFLE" bucket. Use watchmakers' forceps if necessary to remove organisms from the net.

9. Record the nearest transect location in the box for the sample on the Sample Collection Form. Also note the dominant substrate type by checking the appropriate box on the Sample Collection Form.

   Fine/sand: not gritty (silt/clay/muck < 0.06 mm diam.) to gritty, up to ladybug sized (2 mm diam.)
   Gravel: fine to coarse gravel (ladybug to tennis ball sized; 92 mm to 64 mm diam.)
   Coarse: Cobble to boulder (tennis ball to car sized; 64 mm to 4000 mm)
   Other: 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.

(continued)

TABLE 11-4. (Continued)

10. Thoroughly rinse the net before proceeding to the next sampling location (either the next riffle unit or a different quadrat location within the same riffle unit).

11. Repeat steps 1-10 at subsequent riffle sampling points until 8 kick samples have been collected and placed into the "TARGETED RIFFLE" bucket.

selecting sampling points within the available riffle macrohabitat units is to allocate points throughout the sampling reach as much as possible.

Procedures for collecting a point sample using the kick net from riffle macrohabitat units are presented in Table 11-4. At each sampling point, a quadrat having a total area of 0.09 m^2 (1 ft^2) is sampled. Because the reach-wide and targeted riffle samples are collected in the order they are encountered during a single pass through the reach, it is very important to rinse the kick net thoroughly between samples to avoid carryover and possible cross-contamination of the targeted riffle sample and the reach-wide sample.

11.2 SAMPLE PROCESSING

After collecting kick net samples for both the reach-wide and targeted riffle samples, prepare two composite index samples from the contents of the "REACH-WIDE" and "TARGETED RIFFLE" buckets as described in Table 11-5. Record tracking information for each composite sample on the Sample Collection Form as shown in Figure 11-3. A set of completed sample labels, including the label that is used if more than one jar is required for a single composite sample, is shown in Figure 11-5. Note that each composite sample has a different sample number (barcode). The ID number is also recorded on a waterproof label that is placed inside the jar (Figure 11-5, lower right). If more than one jar is used for a composite sample, a special label (Figure 11-5, lower left) is used to record the ID number assigned to the sample. DO NOT use two different barcode numbers on two jars containing one single sample. Blank labels for use inside of sample jars are presented in Figure 11-6. These can be copied onto waterproof paper.

Check to be sure that the prenumbered adhesive barcoded label is on the jar and covered with clear tape, and that the waterproof label is in the jar and filled in properly. Be sure the inside label and outside label describe the same sample. Replace the cap on each jar and seal them with plastic electrical tape. Check to make sure the cap is properly marked with site number, habitat type (reach-wide or targeted riffle). Place the samples in a cooler or other secure container for transporting and/or shipping the laboratory (see Section 3). The container and absorbent material should both be suitable for transporting ethanol. Check to see that all equipment is in the vehicle.

TABLE 11-5. PROCEDURE FOR PREPARING COMPOSITE SAMPLES FOR BENTHIC MACROINVERTEBRATES

1. Pour the entire contents of the "REACH-WIDE" bucket through a sieve with 500 µm mesh size). Remove any large objects and wash off any clinging organisms back into the sieve before discarding.

2. Using a wash bottle filled with stream water, rinse all the organisms from the bucket into the sieve. This is the composite reach-wide sample for the site.

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). Avoid using more than one jar for each of the composite samples.

4. Fill in a "REACH-WIDE" (or "TARGETED RIFFLE") sample label with the stream ID and date of collection. Attach the completed label to the jar and cover it with a strip of clear tape.

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 the jar is too full pour off some water through the sieve until the jar is not more than ¼ 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.

6. Place a waterproof label with the following information inside each jar:

   - Stream Number
   - Type of sampler and mesh size used
   - Habitat type (riffle or pool)
   - Name of stream
   - Date of collection
   - Collectors initials
   - Number of transect samples composited

7. Completely fill the jar with 95% ethanol (no headspace) so that the final concentration of ethanol is between 75 and 90%. It is very important that sufficient ethanol be used, or the organisms will not be properly preserved.

   - NOTE: Prepared composite samples can be transported back to the vehicle before adding ethanol if necessary.

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. Repeat Steps 1 through 8 for the "TARGETED RIFFLE" bucket.

10. Store labeled composite samples in a container with absorbent material that is suitable for use with 95% ethanol until transport or shipment to the laboratory.

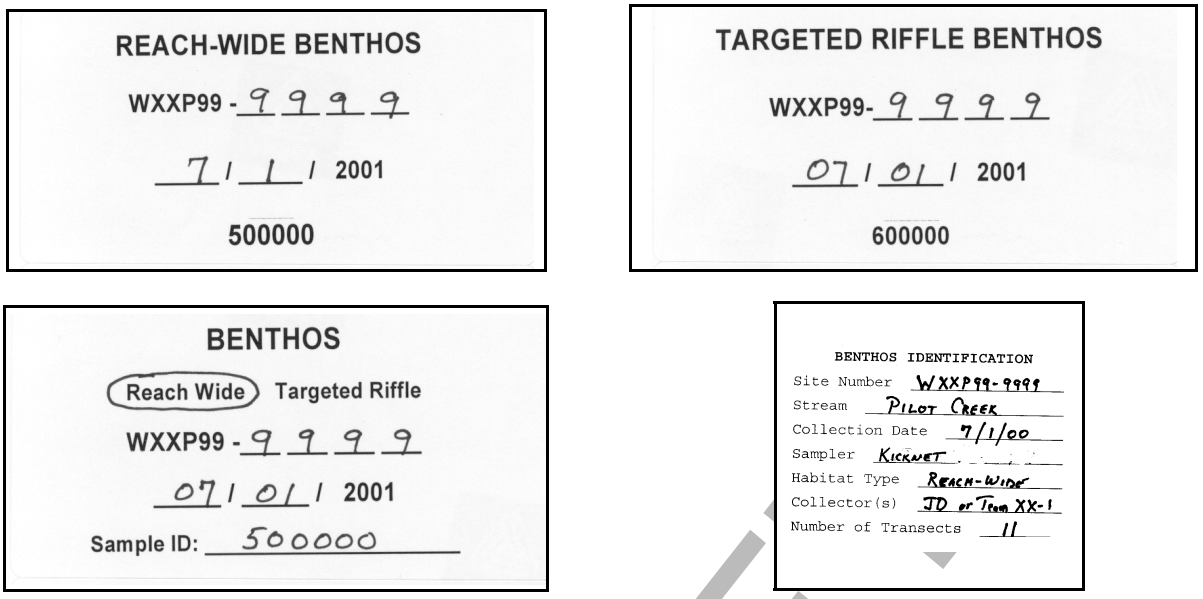


Figure 11-5. Completed labels for benthic macroinvertebrate samples. The label at lower left is used if more than one jar is required for a composite sample. The label at lower right is placed inside the sample container.

11.3 EQUIPMENT AND SUPPLY CHECKLIST

Figure 11-7 shows the checklist of equipment and supplies required to complete the collection of benthic macroinvertebrates from streams. 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. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

11.4 LITERATURE CITED

Barbour, M.T., J. Gerritsen, B.D. Snyder, and J.B. Stribling. 1999. *Rapid Bioassessment Protocols for Use in Streams and Wadeable Rivers: Periphyton, Benthic Macroinvertebrates, and Fish*. Second Edition. EPA/841-B-99-002. U.S. Environmental Protection Agency, Office of Water, Assessment and Watershed Protection Division, Washington, D.C.

BENTHOS IDENTIFICATION

Site Number ____________

Stream ____________

Collection Date ____________

Sampler ____________

Habitat Type ____________

Collector(s) ____________

Number of Transects ______

BENTHOS IDENTIFICATION

Site Number ____________

Stream ____________

Collection Date ____________

Sampler ____________

Habitat Type ____________

Collector(s) ____________

Number of Transects ______

BENTHOS IDENTIFICATION

Site Number ____________

Stream ____________

Collection Date ____________

Sampler ____________

Habitat Type ____________

Collector(s) ____________

Number of Transects ______

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Number of Transects ______

BENTHOS IDENTIFICATION

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Stream ____________

Collection Date ____________

Sampler ____________

Habitat Type ____________

Collector(s) ____________

Number of Transects ______

BENTHOS IDENTIFICATION

Site Number ____________

Stream ____________

Collection Date ____________

Sampler ____________

Habitat Type ____________

Collector(s) ____________

Number of Transects ______

Figure 11-6. Blank labels for benthic invertebrate samples.

EQUIPMENT AND SUPPLIES FOR BENTHIC MACROINVERTEBRATES

QTY.	ITEM	
1	Modified kick net (D-frame with 500 µm mesh) and 4-ft handle (Wildco #425-C50)	
	Spare net(s) and/or spare bucket assembly for end of net	
1	Watch with timer or a stopwatch	
2	Buckets, plastic, 8- to 10-qt capacity, labeled “REACH-WIDE” and “TARGETED RIFFLE”	
1	Sieve with 500 µm mesh openings	
1	Sieve-bottomed bucket, 500 µm mesh openings	
2 pr.	Watchmakers’ forceps	
1	Wash bottle, 1-L capacity labeled “STREAM WATER”	
1	Small spatula, spoon, or scoop to transfer sample	
1	Funnel, with large bore spout	
4 to 6 each	Sample jars, HDPE plastic with screw caps, 500-mL and 1-L capacity, suitable for use with ethanol	
2 gal	95% ethanol, in a proper container	
2 pr.	Rubber gloves, heavy rubber	
1	Cooler (with suitable absorbent material) for transporting ethanol and samples	
2	Composite Benthic sample labels, with preprinted ID numbers (barcodes)	
4	Composite Benthic sample labels without preprinted ID numbers	
6	Blank labels on waterproof paper for inside of jars	
1	Sample Collection Form for site	
	Soft (#2) lead pencils	
	Fine-tip indelible markers	
1 pkg.	Clear tape strips	
4 rolls	Plastic electrical tape	
1	Knife, pocket, with at least two blades	
1	Scissors	
1	Pocket-sized field notebook (optional)	
1 pkg.	Kim wipes in small self-sealing plastic bag	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for benthic macroinvertebrates	

Figure 11-7. Equipment and supply checklist for benthic macroinvertebrates.

Cuffney, T.F, M.E. Gurtz, and M.R. Meador. 1993. *Methods for Collecting Benthic Invertebrate Samples as Part of the National Water-Quality Assessment Program.* U.S. Geological Survey Open-File Report 93-406, Raleigh, North Carolina.

Klemm, D.J., P.A. Lewis, F. Fulk, and J.M. Lazorchak. 1990. *Macroinvertebrate Field and Laboratory Methods for Evaluating the Biological Integrity of Surface Waters.* EPA/600/4-90/030. U.S. Environmental Protection Agency, Environmental Monitoring Systems Laboratory, Cincinnati, Ohio.

Klemm, D.J., J.M. Lazorchak, and P.A. Lewis. 1998. Benthic Macroinvertebrates. pp. 147-182 IN: J.M. Lazorchak, D.J. Klemm, and D.V. Peck (Eds.). *Environmental Monitoring and Assessment Program-Surface Waters: Field Operations and Methods for Measuring the Ecological Condition of Wadeable Streams*. EPA/620/R-94/004F. U.S. Environmental Protection Agency, Washington, D.C.

Plafkin, J.L., M.T. Barbour, K.D. Porter, S.K. Gross, and R.M. Hughes. 1989. *Rapid Bioassessment Protocols for Use in Streams and Rivers: Benthic Macroinvertebrates and Fish.* EPA/440/4-89/001. U.S. Environmental Protection Agency, Assessment and Watershed Protection Division, Washington, D.C.

NOTES

NOTES

SECTION 12
AQUATIC VERTEBRATES

by
Frank H. McCormick[1] and Robert M. Hughes[2]

Sampling amphibian, fish, and crayfish (no reptiles) species to determine their proportionate abundances and the presence of external anomalies is conducted after all other field sampling and measurement activities are completed. The objective is to collect a representative sample of all except very rare species in the assemblage. Backpack electrofishing equipment is used as the principal sampling gear (Section 12.1.1). Bank or towed electrofishers are recommended for wide but shallow streams (Section 12.1.2), and seining (Section 12.1.3) is used in habitats where high conductivity or turbidity preclude electrofishing. All team personnel are involved in collecting aquatic vertebrates. In addition to gathering data on the assemblage, fish specimens are retained for analysis of tissue contaminants and microbial pathogens (Section 13).

The procedures and activities presented here differ slightly from those previously published for EMAP-SW (McCormick and Hughes, 1998). These changes are summarized in Table 12-1. In 2000, aquatic vertebrates collected between transects were tallied and recorded separately to provide a means to evaluate sampling efficiency. Crayfish collected during aquatic vertebrate sampling are counted and included as part of the aquatic vertebrate sample. Identifying and tallying specific types of external anomalies, and measuring total lengths of 30 individuals of dominant species are no longer included. Beginning in 2001, aquatic vertebrates are tallied and recorded on a single data form for the entire reach. If a wadeable river is too shallow to sample by boat and too wide (> 20 m) to sample efficiently with backpack electrofishers or seines, the stream is probably more effectively sampled using methods similar to those presented in the field operations manual for nonwadeable rivers and streams, and may require more than 1 day to sample completely.

[1] U.S. EPA, National Exposure Research Laboratory, Ecological Exposure Research Division, 26 W. Martin Luther King Dr., Cincinnati, OH 45268.

[2] Dynamac International Corp., 200 SW 35th St., Corvallis, OR 97333

TABLE 12-1. SUMMARY OF CHANGES IN AQUATIC VERTEBRATE PROCEDURES FOR THE WESTERN PILOT STUDY

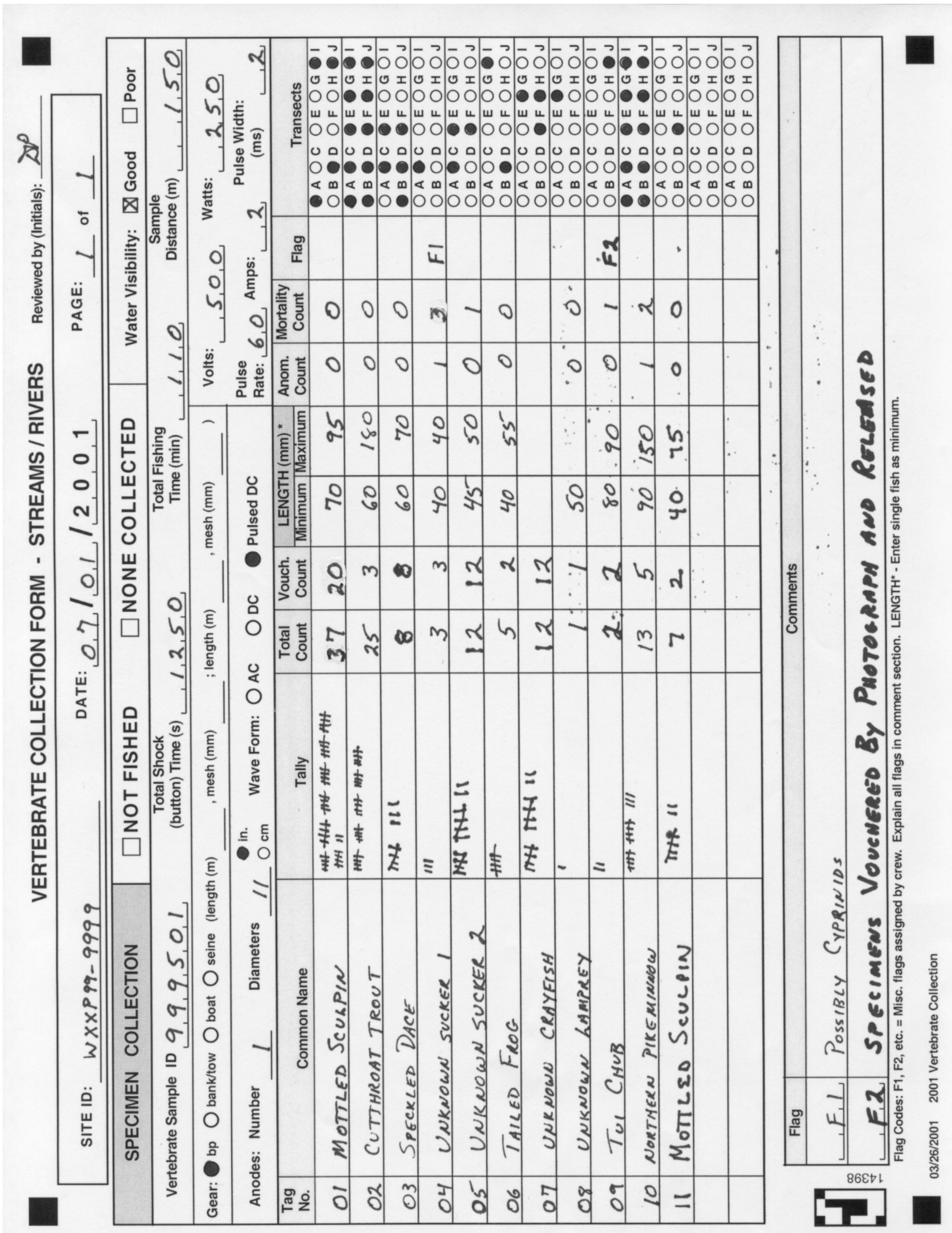
Changes from McCormick and Hughes (1998)
1. Aquatic vertebrates collected between each pair of transects are tallied and recorded on separate field data forms.
2. Crayfish collected during aquatic vertebrate sampling are counted and included as part of the aquatic vertebrate sample.
3. Recording the occurrence of specific types of external anomalies is not required.
4. Determination of total lengths of 30 individual fish of each dominant species collected is not required.
Changes from Year 2000 Western Pilot Study Activities
1. Aquatic vertebrates and crayfish are tallied and recorded on a single data form for each stream; all transects where a species is collected are noted on the form
2. Procedures for dealing with wide (>20 m) yet wadeable streams have been clarified and/or included.

12.1 SAMPLE COLLECTION

The entire channel within the sampling reach is sampled through use of transects (see Section 4) so that effort is distributed along the entire reach. Collection time should be 45 minutes to 3 hours within the reach (Section 4) to obtain a representative sample. If a stream is very wide, however, it may take 2 days to effectively sample it. Sampling data and general comments (perceived fishing efficiency, missed fish, gear operation, suggestions) are recorded on the Vertebrate Collection Form (Figure 12-1).

12.1.1 Electrofishing

Primary responsibility for safety while electrofishing rests with the crew chief (Section 2). Electrofishing units may deliver a fatal electrical shock. While electrofishing, avoid contact with the water unless sufficiently insulated against electrical shock. Use chest waders with nonslip soles and linesman gloves (***NOTE: some types of "breathable" waders do not provide adequate insulation against electric current when wet***). 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



VERTEBRATE COLLECTION FORM - STREAMS / RIVERS   Reviewed by (Initials): DP

SITE ID: WXXP99-9999   DATE: 07/01/2001   PAGE: 1 of 1

SPECIMEN COLLECTION   ☐ NOT FISHED   ☐ NONE COLLECTED   Water Visibility: ☒ Good ☐ Poor

Vertebrate Sample ID 999501   Total Shock (button) Time (s) 1250   Total Fishing Time (min) 110   Sample Distance (m) 150

Gear: ● bp ○ bank/tow ○ boat ○ seine (length (m) ___, mesh (mm) ___; length (m) ___, mesh (mm) ___)   Volts: 500   Watts: 250

Anodes: Number 1   Diameters 11 ● in. ○ cm   Wave Form: ○ AC ○ DC ● Pulsed DC   Pulse Rate: 60   Amps: 2   Pulse Width: (ms) 2

Tag No.	Common Name	Tally	Total Count	Vouch. Count	LENGTH (mm)* Minimum	LENGTH (mm)* Maximum	Anom. Count	Mortality Count	Flag	Transects
01	MOTTLED SCULPIN		37	20	70	95	0	0		
02	CUTTHROAT TROUT		25	3	60	180	0	0		
03	SPECKLED DACE		8	8	60	70	0	0		
04	UNKNOWN SUCKER 1		3	3	40	40	1	3	F1	
05	UNKNOWN SUCKER 2		12	12	45	50	0	1		
06	TAILED FROG		5	2	40	55	0	0		
07	UNKNOWN CRAYFISH		12	12						
08	UNKNOWN LAMPREY		1	1	50		0	0		
09	TUI CHUB		2	2	80	90	0	1	F2	
10	NORTHERN PIKEMINNOW		13	5	90	150	1	2		
11	MOTTLED SCULPIN		7	2	40	75	0	0		

Flag	Comments
F1	POSSIBLY CYPRINIDS
F2	SPECIMENS VOUCHERED BY PHOTOGRAPH AND RELEASED

Flag Codes: F1, F2, etc. = Misc. flags assigned by crew. Explain all flags in comment section. LENGTH* - Enter single fish as minimum.

03/26/2001   2001 Vertebrate Collection   14398

Figure 12-1. Vertebrate Collection Form (page1).

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 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 turning off the electricity. Avoid electrofishing near unprotected people, pets, or livestock. Discontinue activity during thunderstorms or heavy 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.

12.1.1.1 Backpack Electrofishing--

The backpack electrofishing procedure is presented in Table 12-1; record information on the Vertebrate Collection Form (Figure 12-1). If the stream cannot be sampled by either electrofishing or seining, mark the "NOT FISHED" field on the form. Determine that all team members are wearing waders and gloves and are clear of both electrodes. Wear polarized sunglasses and caps to aid vision. The backpack unit is equipped with an audio alarm that sounds when the output voltage exceeds 300 V. It also serves as an input current indicator for pulse cycles greater than 5Hz. It begins as a strong continuous tone and begins to beep slowly at currents of 1.25 amps. It beeps faster as input current increases. In case of an overload (in excess of 3 amps), the beep becomes very rapid and the overload indicator comes on. Release the anode switch, adjust voltage and waveform, and resume fishing.

The anode is fitted with a net and a second netter uses an insulated dip net to retrieve stunned individuals, which are immediately deposited into a bucket for later processing (Section 12.3). If individuals show signs of stress (loss of righting response, gaping, gulping air, excessive mucus), change water or stop fishing and process them. This should only be necessary on very warm days, in long transects, or if large numbers or biomasses of aquatic vertebrates are collected. Cease electrofishing to process and release listed threatened or endangered species or large game fish as they are netted (see Section 12.2). If periodic processing is required, be sure to release individuals downstream to reduce the likelihood of collecting them again.

TABLE 12-2. BACKPACK ELECTROFISHING PROCEDURES

1. Allocate the total fishing time (45-180 min) among all transects based on stream size & complexity. It may be necessary to spend 2 days on extremely wide wadeable streams.

2. 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, or use alternate gear types.

3. Search for aquatic vertebrates and crayfish even if the stream is extremely small, and it appears that sampling may produce no specimens. If none are collected, check the "NONE COLLECTED" box on the Vertebrate Collection Form. Explain why in comments section.

4. If conductivity, turbidity, or depth preclude backpack electrofishing, sample by seining or bank/towed electrofishing if possible, otherwise do not sample. If you do not sample, complete the "NOT FISHED" field on the Vertebrate Collection Form and comment why.

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 msec. 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.

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 in the water in riffles and pools. Sample available cut-bank and snag habitat as well. Move the anode wand into cover with the current on, then remove the wand quickly to draw fish out. In fast, shallow water, sweep the anode and fish downstream into a net. In extremely wide streams, work from the midline of the stream channel to the banks. 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. The netter, with the net 1 to 2 ft from the anode, follows the operator, nets stunned individuals, and places them in a bucket.

8. Continue upstream until the next transect is reached. Process fish and/or change water after each transect to reduce mortality and track sampling effort.

9. Complete header information on the Vertebrate Collection Form.

10. Repeat Steps 6 through 8 until Transect "J-K" is finished.

12.1.1.2 Bank/towed Electrofishing--

Bank/towed electrofishing sampling procedures are presented in Table 12-3. The primary electrofishing gear is a 9 ft. inflatable kayak modified to carry all fishing equipment. Kayak configuration consists of a frame mounted generator and electrofishing control box, port and starboard cathodes, and one or two hand-held anodes fitted with netting. Alternatively, the generator and control box may remain on the riverbank connected to the electrodes by a 100 m long heavily insulated wire. The kayak is maneuvered by one or two persons, and the vertebrates are collected by the others wading in the water. Wear chest waders and linesman gloves to avoid electric shock, and polarized sunglasses and caps to minimize glare. Starting at the bottom of the reach and along the designated shoreline, fish in an upriver direction. Adjust voltage and output according to sampling effectiveness and incidental mortality to specimens.

The netters use a dip net and an insulated anode with a net ring to retrieve stunned individuals, which are then deposited into a livewell in the kayak for later processing (Section 12.2). **Note, to avoid shocking crew members, insure that the thumb switch is off anytime the anode leaves the water.** Change the water in the live well at each transect to minimize mortality. If individuals show signs of stress (loss of righting response, gaping, gulping air, excessive mucus), stop fishing and process them. This should only be necessary on very warm days, in long transects, or if very large biomasses of aquatic vertebrates are collected. Cease electrofishing to immediately process and release specimens (e.g., listed species or large game fish) as they are netted (Section 12.2). If periodic processing is required, be sure to release individuals downriver and away from the shoreline to reduce the likelihood of collecting them again. At the completion of electrofishing each transect, record information on the Vertebrate Collection Form (Figure 12-1). Use ear protection and hand signals to communicate direction and power on or off when using generators.

Gasoline is extremely volatile and flammable. Its vapors readily ignite on contact with heat, spark or flame. Never attempt to refill the generator while it is running. Always allow the generator to cool before refilling. Keep gasoline out of direct sunlight to reduce volatilization and vapor release. Keep gasoline only in approved, tightly closed plastic containers.

TABLE 12-3. BANK/TOWED ELECTROFISHING PROCEDURES

1. Select river bank for initial fishing (left for odd numbered sites [e.g., WXXP99-0503], right for even) unless immediate hazards or obstructions preclude this. Stay along the selected bank for two transects to the degree it is safely wadeable. Switch to the opposite bank for the next two transects, alternating in this manner until the entire site is fished or hazards prevent it. Using a rangefinder, determine a downstream point that is 4 mean channel widths distant (this is the transect length). Record this distance on the Vertebrate Collection Form.

2. Fill tank with gas, check all electrical connections and potential conductors, and place the anodes and cathodes in the water. Fill livewell and put on linesman gloves. Verify that all electrical switches are off, that cathodes are submerged, that all non-target organisms are clear of the water or 20' away, and that barge surfaces are dry.

3. 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, or use alternate gear types.

4. Start generator, switch to pulsed DC, a pulse rate of 30 pps, low range (duty cycle) and 40%. Increase % (voltage) as needed to roll fish. If success is poor, reduce %, switch to high range, and again increase % as needed. If effectiveness is still low, switch to 60 pps and repeat the process. If the current (amperage needle) is reduced, switch back to low range to avoid over-loading the generator. **Switching should occur when power to the control box is off.** If the conductivity of the river is > 1700 &S/cm, use a larger generator or seine. Netters activate thumb switches and insure that when they are off current ceases. Crew members towing the barge activate the generator and pulsator switches. Verify that fish are rolled and relaxed but not rigid before beginning transect. Record settings on the Vertebrate Collection Form and clear clocks.

5. Zero the timer, and depress the thumb switch to begin fishing. With system activated and safety switches on, fish upstream near shore. Maneuver the anode(s) to cover a swath 3-4 meters wide, near cover, and at depths less than 1 meter wherever possible. Do not place yourself or the gear in danger in order to fish particular habitats; cut the generator and stow the gear before negotiating hazards.

6. Place fish directly in livewell; do not hold them in the electrical field. Pay special attention to netting small and benthic fishes as well as fishes that respond differently to the current--not just the big fish that move to the surface. Try to net all fish seen, but in productive areas this is impossible. Do not chase individual fish or place yourselves in unbalanced positions to net them. If benthic fish are not being collected, occasionally hold a net in the current and along the bottom, then sweep the anode downstream into the net. Draw fish from cover by thrusting the anode into the cover with the power on, then quickly removing it.

7. Cease sampling at the end of the transect. Process the fish quickly and carefully, returning them to the water unless they are vouchered or saved for tissue.

8. Return to step 1 for each of the subsequent 9 transects, but begin upstream from where fish were released and alternate banks on every other transect.

12.1.2 Seining

Seining is used when the conductivity of the stream is too high and/or in streams with extremely high turbidity where electrofishing is ineffective. Avoid high mortality rates by employing multiple short seine hauls at each of the 11 transects by using either the "riffle" or "pool" method (Table 12-4). Allocate the total sampling time (240 minutes) among the 11 transects (i.e., from 16 to 22 minutes per transect). If no aquatic vertebrates were collected, indicate this on the form (Figure 12-1). Record the seine length, mesh size, the time spent seining ("SAMPLING TIME") and the length of the seine haul ("SAMPLING DISTANCE") on the Vertebrate Collection Form (Figure 12-1). If more than one size or type of seine is required, record the information for the primary seine used on the collection form, and note the alternative types used in the comments section of the form.

12.2 SAMPLE PROCESSING

Sample processing involves tallying and identifying fish, crayfish and amphibians, examining individual specimens for external anomalies, obtaining length measurements from selected specimens, preparing voucher specimens for taxonomic confirmation and archival at a museum, and selecting specimens to prepare samples for fish tissue contaminants (Section 13). Process collections as quickly as possible to minimize stress to live specimens. One person can process fish from one bucket while the other team members continue to collect fish and deposit them into a second bucket. One person can identify, measure, and examine individuals while another person records information on the field data forms.

12.2.1 Taxonomic Identification and Tally

Table 12-5 presents the procedure for identifying and tallying aquatic vertebrates. Record identification, tally data, and comments for each species on the Vertebrate Collection Form (Figure 12-1). It is important to note all transects where a species is collected, as this is information is needed to develop estimates of sampling efficiency. Use common names from Page and Burr (1991) or similar keys. Taxonomic identification should be performed only by trained ichthyologists familiar with the fish species and other aquatic vertebrate taxa of the region. Use taxonomic reference books and other materials that contain species descriptions, ranges, and identification keys to make species identifications in the field. Where there are many individuals of easily identified species, processing is facilitated by keeping a tally count of the number of individuals of each species and totaling

TABLE 12-4. SEINING PROCEDURES

12. Allocate the sampling effort throughout the reach so that the total fishing time will be between 45 minutes (small stream) and 3 hours (large stream). It may be necessary to spend 2 days on extremely wide wadeable streams.

13. 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.

14. Search for aquatic vertebrates & crayfish even if the stream is extremely small and it appears that sampling will produce no specimens. If none are collected, complete the "NONE COLLECTED" field on the Vertebrate Collection Form. Explain why in the comments section.

15. Begin at the downstream end of the sampling reach (Transect A). Proceed along the reach, sampling available habitats using the appropriate methods below:

    4A. Riffle habitats: Use a seine 2 m long (1.25 m high with 0.6 cm mesh.

        1. Place the seine perpendicular to the current across the downstream end of the riffle. Ensure that the lead line is on the bottom. Tilt the net slightly downstream to form a pocket to trap aquatic vertebrates.
        2. Starting about 2 m upstream, kick the substrate and overturn rocks, proceeding downstream toward the net.
        3. Raise the net quickly and examine it carefully for aquatic vertebrates (and crayfish).

    3B. Pool habitats: Use a seine 3-9 m long × 2 m high with 0.6 cm mesh size.

        1. Two people pull the seine across the pool, using the shore or riffles as barriers.
        2. In areas with current, pull the net downstream and then sweep toward the shore with one or both poles, or keep one end of the seine near shore and sweep the other end in a wide arc from midstream to the same shore.
        3. Pull the net onto the shore and examine it carefully for aquatic vertebrates (and crayfish).

    3C. Snags & undercut banks: Use a seine 2 m long × 1.25 m high; 0.6 cm mesh size.

        1. Jab the seine under the cover and near the stream bottom, then quickly lift it above the stream surface.

16. Place individuals in buckets for processing, and continue upstream to the next habitat area.

17. Complete the header information on the Vertebrate Collection Form.

18. Repeat Steps 4 through 6 for successive habitat areas until Transect "I-K" is finished.

TABLE 12-5. PROCEDURE TO IDENTIFY, TALLY, AND EXAMINE AQUATIC VERTEBRATES

1. Complete all header information accurately and completely. If no vertebrates or crayfish were collected, complete the "NONE COLLECTED" field on the Vertebrate Collection Form.

2. Identify and process each individual completely, ideally handling it only once. Record the common name (PRINT USING CAPITAL LETTERS) on the first blank line in the "SPECIMENS" section of the Vertebrate Collection Form. If a species cannot be positively identified, assign it as "unknown" followed by its common family name (e.g., UNKNOWN SCULPIN A). Note every transect where a species is collected (letters represent the downstream transect).

3. Process species listed as threatened and endangered first and return individuals immediately to the stream. Photograph specimens for voucher purposes if conditions permit and stress to individuals will be minimal. Indicate if photographed on Vertebrate Collection Form. If individuals have died, prepare them as voucher specimens and preserve in formalin.

4. Keep voucher specimens (up to 20) of smaller individuals of each species. If no smaller individuals are collected, photograph each species and indicate so on the data form. Large, questionable species may be placed on ice and then frozen.

5. Tally the number of individuals of each species collected in the "TALLY" box on the Vertebrate Collection Form and record the total number in the "COUNT" field on the form.

6. Measure the total length (body length for amphibians, no lengths for crayfish) of the largest and smallest individual to provide a size range for the species. Record these values in the "LENGTH" area of the Vertebrate Collection Form.

7. Examine each individual for external anomalies and tally those observed. Readily identified external anomalies include 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 Vertebrate Collection Form. Photograph specimens with especially extreme anomalies and save them for fish tissue microbial samples.

8. Record the total number of mortalities due to electrofishing or handling on the Vertebrate Collection Form.

9. 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.

10. 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.

11. Repeat Steps 1 through 10 for all other species.

(Continued)

TABLE 12-5 (continued)

12. After processing the fish from all 10 transects, calculate a Jaccard Coefficient (JC) to assess sample adequacy. To help randomize the calculation, list the species collected from transects A-B, C-D, E-F, G-H, and I-J in group "A" and those from B-C, D-E, F-G, H-I, and J-K in group "B". Calculate the Jaccard coefficient as:

$$\mathbf{JC} = \frac{\mathbf{S}}{\mathbf{S} + \mathbf{A} + \mathbf{B}}$$

where **S** is the number of species shared by both groups, **A** is the number of species unique to group A, and **B** is the number of species unique to group B. Record JC = "n" in the comments section; if JC < 0.7, sample two additional transects. List the species in the appropriate group and recalculate JC. Continue until JC $\geq$ 0.7 or there is insufficient time or space to sample.

EXAMPLE: You have collected 6 different species from the group "A" transects, and 4 different species from the group "B" transects. Of these, three species were shared by both groups.

JC = (3) / (3) + (6-3) + (4-3)
JC = (3) / (3 + 3 + 1)
JC = (3) / (7)
JC = 0.4 In this case, sample two additional transects and recalculate.

the tally once processing is complete. If protected fish have died, voucher them. Notify the appropriate state officials as soon as possible.

After the entire reach has been sampled , sampling adequacy is estimated based on the Jaccard coefficient of similarity, using the presence/absence of species between the downstream and upstream halves of the reach (i.e., the top and bottom rows of transects on the form. If the calculated Jaccard value is < 0.7, sample an additional 2 transects (8 channel widths) upstream of the reach. Continue with additional transects until the calculated Jaccard value is $ 0.7, or until there is insufficient time or space to sample. For the data presented in Figure 12-1, a total of 9 vertebrate species were collected. Six species are shared between the two groups of transects, 2 species were only collected in the "A" group of transects, and 1 species was only collected in the "B" group of transects. The Jaccard coefficient is calculated as:

$$JC = \frac{6}{6+2+1} = 0.67 \approx 0.7$$

For this site, the sampling effort is adequate, and no additional transects are sampled.

12.2.2 External Examination and Length Measurements

During the tallying procedure for each species (Table 12-5), examine each individual for the presence of external anomalies. Record the number of individuals affected on the Vertebrate Collection Form (Figure 12-1). Blackening and exopthalmia (popeye) may occasionally result from electrofishing. Injuries due to sampling are not included in the tally of external anomalies, but should be noted in the comments section of the Vertebrate Collection Form (Figure 12-1). Blackening from electrofishing usually follows the myomeres or looks like a bruise. If fish die due to the effects of sampling or processing, record the number for each species on the Vertebrate Collection Form (Figure 12-1).

For each species, use a measuring board or ruler to determine the length of the largest and smallest individuals collected at a site. Measure total length for fish (nose to distal end of caudal fin) and body length for amphibians (tip of snout to vent) on the Vertebrate Collection Form (Figure 12-1). No length measurements are taken for crayfish.

12.2.3 Preparing Voucher Specimens

With the exception of very large individuals or easily identified species, up to 20 individuals of each species are vouchered from each stream site to provide a permanent, archived, historical record of fish collections. Prepare the voucher sample for a site according to the procedure presented in Table 12-6. Retain additional specimens of the appropriate species for the fish tissue contaminants samples (Section 13). **For each species, voucher specimens take priority over specimens for the tissue contaminants samples.**

The number of voucher specimens and the method of vouchering varies with species. Large, easily identified species, larger species that are difficult to identify in the field, or species that are uncommon in the region require a few specimens of both adults and juveniles, if both were collected. Very large specimens, especially of easily identified game fish, are “vouchered” by photographing them and then releasing them alive. More voucher specimens are required for smaller species, which are typically more difficult to identify in the field. Species of "special concern" (state and federally protected species), are processed first, vouchered by photographs, and released alive. Include any individuals of protected species that die before they can be released as part of the preserved voucher sample for the stream.

For taking photographs, use a film or digital camera with sufficient macro capability to take clear, close-up photographs of small fish or other aquatic vertebrates. The photographs must be of sufficient clarity or resolution that important external identifying characteristics can be distinguished. For each photograph, include a card with the site ID printed on it, and a measuring board, ruler, or some other object to provide a length reference.

Individuals selected as voucher specimens are first anaesthetized in a concentrated solution of carbon dioxide. Voucher specimens for each species are counted and placed in nylon mesh bags, stockings or plastic jars (1 or more bags per species). Each bag contains a numbered tag (Figure 12-2). Single specimens of easily identified and distinct species (e.g., sandroller, smallmouth bass) and crayfish may be placed directly in the jar with the tag. Record the tag number and the number of individuals vouchered for each species on the Vertebrate Collection Form (Figure 12-1). Preserve vouchers of sculpins, minnows, lampreys and other difficult species from throughout the reach. Use multiple bags and tags to do so. **This bagging, tagging, and recording is crucial, as it enables us to estimate**

TABLE 12-6. GUIDELINES AND PROCEDURES FOR PREPARING AQUATIC VERTEBRATE VOUCHER SPECIMENS

1. Determine the voucher class of a species and the number of specimens to include in the voucher sample based on the following guidelines. Process **Class 1 species first.** Voucher samples take priority over tissue contaminant samples.

   Class 1--State or federally listed species. Photograph and **release immediately**. Photographs should include (1) a card with the stream ID and (2) an object of known length with the specimen. If specimens have died, proceed to Step 2 and include them in the voucher sample. Flag the species with an "Fn" on the Vertebrate Collection Form and note it is a listed species in the comments section of the form. Notify the appropriate state officials as soon as possible.

   Class 2--Large easily identified species **OR** adults that are difficult to identify **OR** species that are uncommon in that region (e.g., sunfish, suckers, bullheads, trout, crayfish). Preserve 1-2 small (<150 mm total length) adult individuals per site plus 2-5 juveniles. If only large adults are collected, reserve smallest individuals until voucher procedure is complete and preserve ONLY if space is available. Individuals with a total length > 160 mm should be slit on the lower abdomen of the RIGHT side before placing them into the container. Photograph if considered too large for the jar or place in a bag on ice for freezing **(Do not voucher large gamefish)**. Retain additional individuals for the tissue contaminant sample.

   Class 3--Small to moderate-sized fish **OR** difficult to identify species (e.g., lampreys, juvenile salmonids, minnows, sculpins). Preserve up to 20 adults and juveniles (several per transect). If fewer than 20 individuals are collected, voucher them all. Retain additional individuals for the tissue contaminant sample.

2. Anesthesize voucher specimens in a bucket with two carbon dioxide tablets and a small volume of water, then transfer them to a nylon mesh bag. Tally, then record the number of individuals included in the voucher sample in the "VOUCHERED COUNT" field for the species on the Vertebrate Collection Form.

3. Select a "FISH-BAG" tag with the same ID number as the voucher sample jar (Step 6). Record the tag number in the "TAG NO." field on the corresponding line for the species on the Vertebrate Collection Form. Place the tag into the mesh bag and seal. **This bagging, tagging, and recording is crucial, as it enables us to estimate species proportionate abundances in the assemblage even when 1 suspected species turns out to be multiple species.**

4. Immediately place the bag into a container large enough to hold all voucher specimens loosely and half-filled with 10% formalin. **Use additional jars if necessary to avoid close packing and bending of voucher specimens**.

5. Repeat Steps 1 through 4 for all species collected.

(Continued)

TABLE 12-6 (continued)

6. Prepare two "FISH-JAR" labels (each having the same sample ID number) by filling in the stream ID and the date of collection. Place one label into the sample jar. Cap tightly and seal with plastic electrical tape. Attach the second label to the outside of the sample container by covering it with a strip of clear tape. Record the voucher sample ID number on page 1 of the Vertebrate Collection Form. Record general comments (perceived fishing efficiency, missed fish, gear operation, suggestions) in blank lines of form. NOTE: If more than one jar is required, use labels that have the same ID number printed on them and flag.

7. Place the preserved sample in a suitable container with absorbent material. Store the container in a well-ventilated area during transport. Follow all rules and regulations pertaining to the transport and shipment of samples containing 10% formalin.

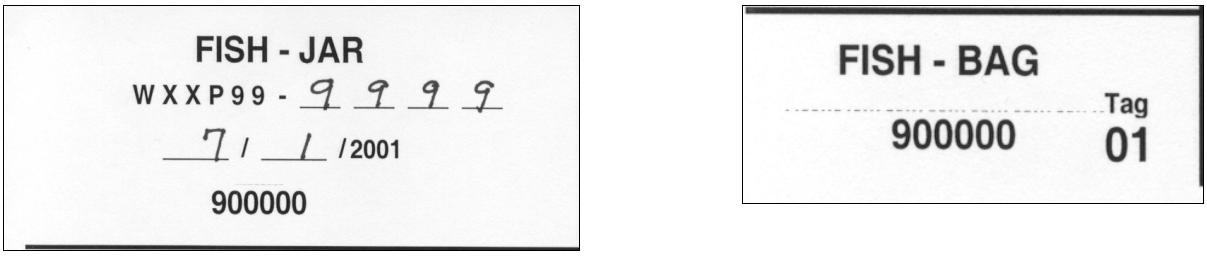


Figure 12-2. Completed voucher sample label and specimen bag tag for aquatic vertebrates. Note that two voucher sample labels (left) are filled out– one is placed inside the jar, the second is taped to the outside.

species proportionate abundances in the assemblage even when 1 suspected species turns out to be multiple species.

Place specimen bags into a large sample jar containing 10 % buffered formalin (Section 3). The final volume of 10% formalin in the sample container should equal to or exceed the total volume of specimens. Use additional containers if necessary and avoid tight packing of specimen bags or bending of specimens. Delays in carrying out the anaesthetization and preservation procedures, overpacking a bag or sample container, or an inadequate volume of preservative will result in unidentifiable (worthless) specimens. Formalin vapors and solution are extremely caustic and may cause severe irritation on contact with skin, eyes or mucus membranes, and it is a potential carcinogen. Contact with it should be avoided. Wear gloves and safety glasses and always work in a well-ventilated area. In case of contact with skin or eyes, rinse immediately with large quantities of water. Store stock solution in sealed containers in a safety cabinet or cooler lined with vermiculite. If possible, transport outside of the passenger compartment of a vehicle. A set of two sample labels is completed for each sample container as shown in Figure 12-2. Place one label inside each sample container, and tape the second label to the outside of the jar. Record the sample ID number on the Vertebrate Collection Form (Figure 12-1).

12.3 EQUIPMENT AND SUPPLIES

Figure 12-3 is a checklist of equipment and supplies required to conduct protocols described in this section. This checklist may differ from the checklists presented in Appendix A, which are used at a base site to ensure that all equipment and supplies are brought to the stream site. Field teams are required to use the checklist presented in this section to

EQUIPMENT AND SUPPLIES FOR AQUATIC VERTEBRATE SAMPLING

QTY.	Item	
1	Gasoline or battery-powered backpack electrofishing unit with netted anode (electrode wand)	
	Extra battery (charged) or gasoline	
4 pr	Heavy-duty rubber gloves	
3 pr	Chest waders with non-slip soles & patch kit	
3 pr	Polarized sunglasses	
2	Long-handled dip nets (0.6 cm mesh) with insulated handles	
1	Watch or stopwatch to track elapsed fishing time	
4	Collapsible buckets for holding and processing aquatic vertebrates	
1	Minnow seine (2m X 1.25 m, 0.6 cm mesh) with poles	
1	Large seine (9 m X 2 m, 0.6 cm mesh) with poles	
1	Aquarium net	
1 set	Taxonomic reference books and keys for fishes and amphibians of the region	
1	Camera and film (or digital camera) with macro capability for photographing vouchers	
1-2	Fish measuring board & small plastic rulers (2)	
5-20	Small nylon mesh bags or stockings for holding voucher specimens	
1	Jackknife for preparing larger voucher specimens for preservation	
1 ea.	1, 2, and/or 4-L screw-top plastic jars (leakproof) for voucher samples	
2 L	10% (buffered) formalin or voucher sample jar half full of 10% formalin	
1	Container with absorbent material to hold formalin solution and preserved voucher sample jars	
1 pr	Safety glasses	
1 pr	Chemical-resistant gloves	
1	Covered clipboard	
1 box	Carbon dioxide tablets	
1	Sheet of pre-printed jar labels (4) and voucher bag tags (36), all with same preprinted sample ID number (barcode)	
1 pr	Scissors for cutting jar labels and tags	
1 roll	Plastic electrical tape	
1 pkg.	Clear tape strips	
2	Soft lead pencils for recording data and completing tags	
2	Extra fine-tipped indelible markers for completing sample labels	
12	Vertebrate Collection Forms	
3	Plastic safety whistles & ear protection if generators are used	
1	Field operations manual	
1 set	Laminated sheets of aquatic vertebrate procedure tables	
1 ea.	Vertebrate collection permits (State, Federal, Tribal)	

Figure 12-3. Equipment and supplies checklist for aquatic vertebrates.

ensure that equipment and supplies are organized and available to conduct the protocols efficiently.

12.4 LITERATURE CITED

Page, L.M., and B.M. Burr. 1991. *A Field Guide to Freshwater Fishes of North America North of Mexico*. Houghton Mifflin Co., Boston, MA.

McCormick, F.H. 1993. Fish. pp. 29-36 IN: R.M. Hughes (ed.). *Stream Indicator Workshop.* EPA/600/R-93/138. U.S. Environmental Protection Agency, Corvallis, Oregon.
McCormick, F.H., and R.M. Hughes. 1998. Aquatic Vertebrates. pp. 161-182 IN: J.M. Lazorchak, D.J. Klemm, and D.V. Peck (eds.). *Environmental Monitoring and Assessment Program-Surface Waters: Field Operations and Methods for Measuring the Ecological Condition of Wadeable Streams.* EPA/620/R-94-004F. U.S. Environmental Protection Agency, Washington, DC.

NOTES

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SECTION 13
FISH TISSUE CONTAMINANTS

by
Roger B. Yeardley[1], Frank H. McCormick[2], Robert M. Hughes[3],
James M. Lazorchak[2], and Spencer A. Peterson[4]

In addition to gathering data on the aquatic vertebrate assemblage (Section 12), fish are retained for analysis of fish tissue contaminants. In general, the focus is on fish species that commonly occur throughout the region of interest, and that are sufficiently abundant within a sampling reach. The fish tissue contaminants indicator is used to evaluate the potential burden of toxic chemicals and fish pathogens at a site. EMAP focuses on whole fish because they present fewer logistical problems and integrate all fish parts. Three types of fish samples are prepared for each site (if possible). The small fish composite sample uses individuals <100 mm long. The big fish sample uses individuals that are >120 mm long. Additional specimens, using a range of fish sizes, are collected for a "Microbial" sample, which are subjected to internal examination for certain types of pathogens.

Only minor modifications have been made to procedures used in EMAP-WP in 2000. These modifications include clarifying the preparation, labeling, and tracking of "big" fish samples, and increasing the number of possible microbial samples to 6 individuals.

13.1 PREPARING SAMPLES FOR TISSUE CONTAMINANTS

Prepare tissue samples as described in Table 13-1.To determine the proper quantity for each sample, weight is used for the small fish sample and individual length is used for the large fish samples. In the small fish composite, use similar sized individuals if possible

1 SoBran Environmental, c/o U.S. EPA, 26 Martin Luther King Dr., Cincinnati, OH 45268.

2 U.S. EPA, National Exposure Research Laboratory, Ecological Exposure Research Division, 26 Marrtin Luther King Dr., Cincinnati, OH 45268.

3 Dynamac, Inc. C/o U.S. EPA, 200 SW 35th St., Corvallis, OR 97333.

4 U.S. EPA, National Health and Environmental Effects Research Laboratory, Western Ecology Division, 200 SW 35th St., Corvallis, OR 97333.

TABLE 13-1. PROCEDURE TO PREPARE FISH TISSUE SAMPLES

NOTE: Use your best judgement to collect some type of fish tissue sample.

SMALL FISH. After voucher specimens have been prepared, choose a small fish species that has enough similarly sized individuals (ideally small-large size difference < 25%) to equal 400 g(14 oz).

BIG FISH. After considering voucher specimens, select 3 individuals >120 mm total length with a wide size range for each of 3 species. Pacific preference order: bass, pikeminnow, trout, catfish, sucker; Atlantic preference order: bass, walleye/sauger, pike, trout, catfish, sucker).

MICROBIAL. After preparing vouchers and the above specimens, select 6 small adults or large juveniles (preferably with anomalies).

8. Anesthesize fish. Keep hands, foil, & bags clean and free of potential contaminants (mud, fuel, formalin, sun screen, insect repellant, soap, etc.)

9. Record standard common name of species (IN CAPITAL LETTERS) on Vertebrate Collection Form.

10. For **small** fish; record number of individuals for each species in comment line.

11. For **big** and **microbial** samples: Record total length of each individual in the appropriate box of the Vertebrate Collection Form.

12. Indicate sample type by placing an “X” in appropriate box on form.

13. Wrap all **small** fish together in a single piece of aluminum foil, with **dull side of foil in contact with fish**. Place sample in a self-sealing plastic bag.

14. **Wrap each big adult and microbial fish sample separately** in aluminum foil, with dull side of the foil in contact with fish. Place each individual in a single plastic bag.

15. Expel excess air and seal bag.

16. Prepare Fish Tissue sample label for each bag by filling in stream ID, sample type (big, small, microbe) and collection date on each label. Record sample ID for each bag on the Vertebrate Collection Form.

17. Attach appropriate label to bag. Cover label with a strip of clear tape. Place labeled bag into second plastic bag, and re-label and re-tape it.

18. Keep the double-bagged samples **on ice (or frozen if possible)** until shipment.

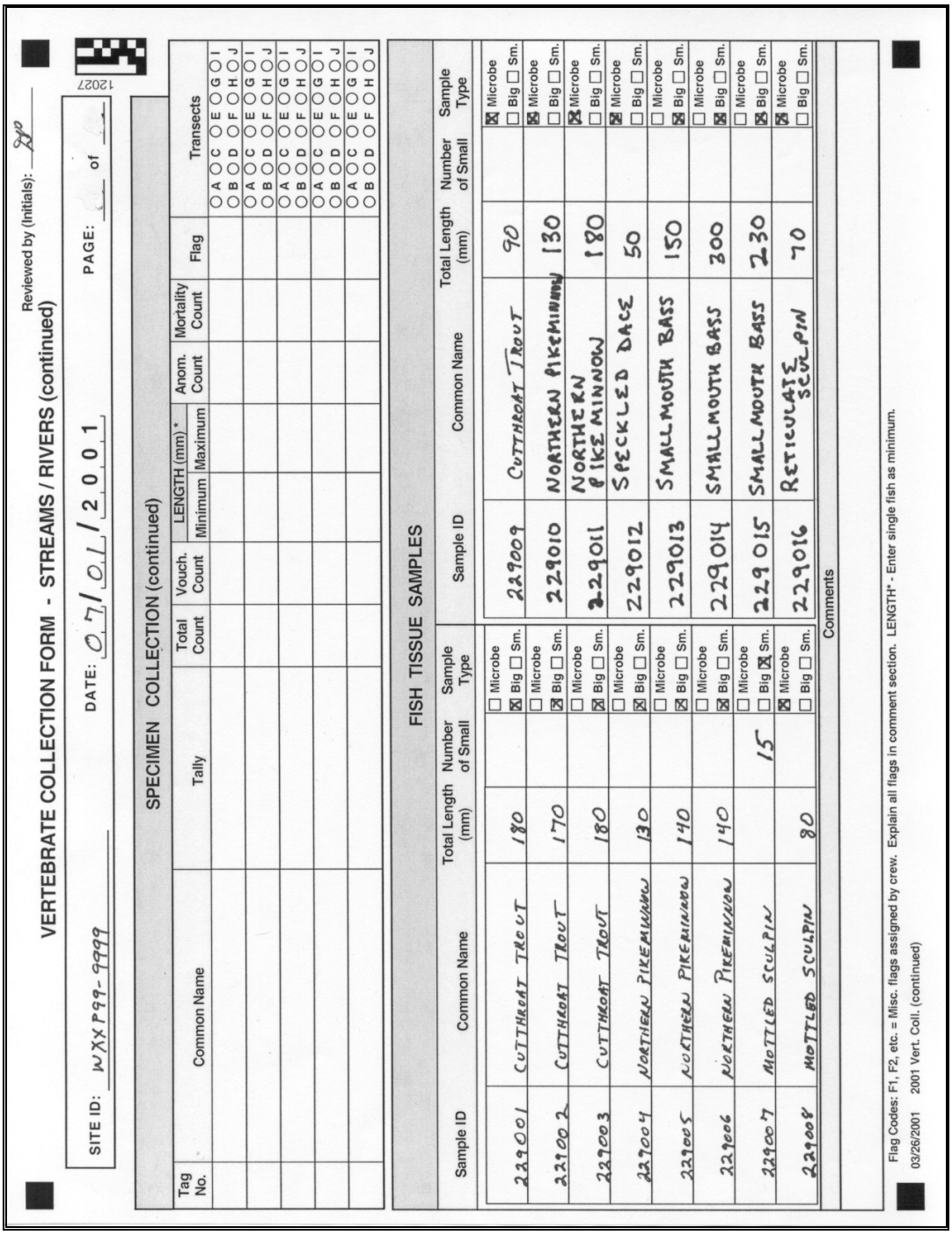
(size difference between smallest to largest < 25%), but getting a sufficient sample is a higher priority than getting similar-sized individuals. For the small fish composite, send as may fish as possible up to 400 g. If there is no single species with enough individuals available, prepare composite samples using individuals of multiple species. For the big fish sample, send as many fish as possible, up to 3 fish for each of 3 species. For the microbial sample, choose any large juveniles or small adults—especially those with external anomalies – and send as many fish as possible up to 6.

Note that voucher specimens have higher priority than tissue samples, and toxic contamination samples have higher priority than the microbial contamination sample.

Record information for the fish tissue and microbial samples on page 2 of the Vertebrate Collection Form (Figure 13-1). Examples of completed sample labels are presented in Figure 13-2. Use a permanent marker to complete labels. Each individual comprising the big adult and microbial samples is wrapped, labeled, and bagged separately, while the small fish composite is wrapped together. Thus, up to 16 different sample labels may be required (9 “big”, 1 “small”, and 6 “microbial”). Each sample is double-bagged. Tissue samples are stored in a cooler with several bags of ice (or ice substitute packs). Double bag the ice and tape the last bag shut to prevent contamination of samples by melting ice. Store tissue samples on ice (freeze them if possible) until they can be shipped (Section 3). Tissue samples can be stored and shipped with other samples requiring icing or freezing (water chemistry and periphyton samples).

13.2 EQUIPMENT AND SUPPLIES

Figure 13-3 is a checklist of equipment and supplies required to conduct protocols described in this section. This checklist may differ from the checklists presented in Appendix A, which are used at a base site to ensure that all equipment and supplies are brought to and are available at the stream site. Field teams are required to use the checklist presented in this section to ensure that equipment and supplies are organized and available to conduct the protocols efficiently.



VERTEBRATE COLLECTION FORM - STREAMS / RIVERS (continued)

Reviewed by (Initials): SJP

SITE ID: WXP99-9999 DATE: 07/01/2001 PAGE: 1 of 1

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SPECIMEN COLLECTION (continued)

Tag No.	Common Name	Tally	Total Count	Vouch. Count	LENGTH (mm) * Minimum	LENGTH (mm) * Maximum	Anom. Count	Mortality Count	Flag	Transects
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J

FISH TISSUE SAMPLES

Sample ID	Common Name	Total Length (mm)	Number of Small	Sample Type	Sample ID	Common Name	Total Length (mm)	Number of Small	Sample Type
229001	CUTTHROAT TROUT	180		☐ Microbe ☒ Big ☐ Sm.	229009	CUTTHROAT TROUT	90		☒ Microbe ☐ Big ☐ Sm.
22900 2	CUTTHROAT TROUT	170		☐ Microbe ☒ Big ☐ Sm.	229010	NORTHERN PIKEMINNOW	130		☒ Microbe ☐ Big ☐ Sm.
229003	CUTTHROAT TROUT	180		☐ Microbe ☒ Big ☐ Sm.	229011	NORTHERN PIKE MINNOW	180		☒ Microbe ☐ Big ☐ Sm.
229004	NORTHERN PIKEMINNOW	130		☐ Microbe ☒ Big ☐ Sm.	229012	SPECKLED DACE	50		☒ Microbe ☐ Big ☐ Sm.
229005	NORTHERN PIKEMINNOW	140		☐ Microbe ☒ Big ☐ Sm.	229013	SMALLMOUTH BASS	150		☐ Microbe ☒ Big ☐ Sm.
229006	NORTHERN PIKEMINNOW	140		☐ Microbe ☒ Big ☐ Sm.	229014	SMALLMOUTH BASS	200		☐ Microbe ☒ Big ☐ Sm.
229007	MOTTLED SCULPIN		15	☐ Microbe ☐ Big ☒ Sm.	229015	SMALLMOUTH BASS	230		☐ Microbe ☒ Big ☐ Sm.
229008	MOTTLED SCULPIN	80		☒ Microbe ☐ Big ☐ Sm.	229016	RETICULATE SCULPIN	70		☒ Microbe ☐ Big ☐ Sm.

Comments

Flag Codes: F1, F2, etc. = Misc. flags assigned by crew. Explain all flags in comment section. LENGTH* - Enter single fish as minimum.

03/26/2001 2001 Vert. Coll. (continued)

Figure 13-1. Vertebrate Collection Form, showing information recorded for fish tissue samples.

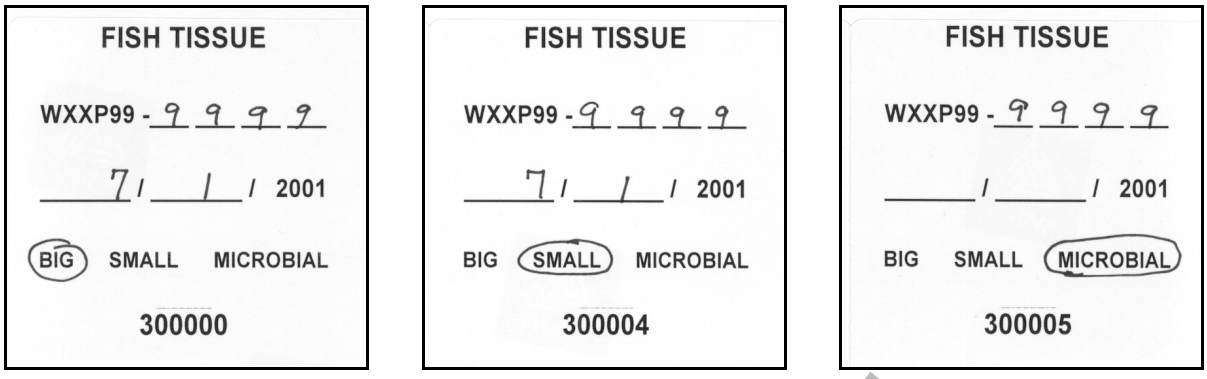

Figure 13-2. Completed sample labels for fish tissue contaminants. Note that a different label (i.e., ID number) is prepared for each individual used for the "big" and "microbial" samples, and up to 16 different ID numbers could be used at a site.

EQUIPMENT AND SUPPLIES FOR FISH TISSUE CONTAMINANTS

QTY.	ITEM	
1	Bucket for anesthetization	
4	Carbon dioxide tablets (Alka-Seltzer® or equivalent)	
1 roll	Aluminum foil (heavy duty) (or 16 18" x 11" rectangles) / (for wrapping fish)	
32	½ - 2 -gallon self-sealing plastic bags, or heavy duty garbage bags (rivers)	
2	Soft (#2) lead pencils or eversharps to record data	
2	Fine-point indelible markers to fill out labels	
1 pkg.	Clear tape strips	
16 pr,	Fish tissue labels (each pair with different ID numbers)	
2	Vertebrate Collection forms	
1 set	Laminated procedure tables for fish tissue contaminants	
1	Cooler with ice (double-bagged and taped)	

Figure 13-3. Equipment and supplies checklist for fish tissue contaminants.

NOTES

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SECTION 14
RAPID HABITAT AND VISUAL STREAM ASSESSMENTS

by
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After all other samples and field data have been collected, the field team conducts an visual-based habitat assessment of the stream reach, makes a general visual assessment of the stream and adjacent area, and performs a final check of the data forms and samples before leaving the stream site (see Section 15). The habitat assessment procedures used are those included in EPA's Rapid Bioassessment Protocols (RBP), originally published by Plafkin et al. (1989), and revised by Barbour et al. (1999). The procedures used for EMAP-WP are modified from those published previously for EMAP-SW (Lazorchak et al., 1998), and the original RBP procedures (Plafkin et al., 1989) to include additional assessment parameters for high gradient streams and a more appropriate parameter set for low gradient streams. These modifications are based on refinements d from various applications across the country. The approach focuses on integrating information from specific parameters on the structure of the physical habitat.

The visual stream assessment is used to record field team observations of catchment and stream characteristics that are useful for data validation, future data interpretation, ecological value assessment, development of associations, and verification of stressor data. The observations and impressions of field teams are extremely valuable. Thus, it is important that these observations about stream characteristics be recorded for future data interpretation and validation.

Beginning in 2001, the rapid habitat assessment is an optional activity. The general description of weather conditions at a site are now included on the field form used for the visual assessment. Evidence of fire has been added as a disturbance type for the visual assessment.

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14.1 RAPID HABITAT ASSESSMENT

NOTE: Beginning in 2001, the rapid habitat assessment is an optional procedure.

The rapid habitat assessment approach based on visual observation is separated into two basic approaches—one designed for high-gradient streams and one designed for low-gradient streams. Based on the perception gained from collecting samples and measurements from throughout the sampling reach, classify the stream as either "Riffle/run prevalent" or "Pool/glide prevalent" based on your visual impression of the dominant habitat type. Choose the prevalent habitat type based on which habitat type occupies the majority of the length of the sampling reach. Landscapes of moderate to high-gradient typically contain "riffle/run prevalent" streams. Under natural conditions, riffle/run prevalent streams contain primarily coarse substrates (i.e., coarse gravel or larger; refer to Section 7) or numerous areas dominated by coarse substrates along a stream reach (Barbour et al, 1998). Landscapes of low to moderate gradient are characterized by glide/pool prevalent streams. These streambeds are dominated by finer substrates (fine gravel or smaller)or occasional areas of coarser sediments along a stream reach (Barbour et al., 1999). The entire sampling reach is evaluated for each parameter.

A different field data form is completed depending upon the prevalent habitat type. For each prevalent stream type, ten "parameters" of habitat are considered and evaluated. These parameters are described in Table 14-1. Most of the parameters are evaluated similarly for both types of prevalent habitats. In three cases, a parameter is evaluated differently, or a different (but ecologically equivalent) parameter is evaluated in riffle/run prevalent versus pool/glide prevalent streams. Substrate embeddedness is evaluated in riffle/run prevalent streams, while pool substrate composition is evaluated in pool/glide prevalent streams. The presence of four potential types of microhabitat types based on combinations of depth and current velocity is evaluated in riffle/run prevalent streams, while the presence of four potential types of pool microhabitat based on depth and area are evaluated in pool/glide prevalent streams. The frequency of riffles is evaluated in riffle/run prevalent streams, while channel sinuosity is evaluated in pool/glide prevalent streams. For three parameters, each bank is evaluated separately and the cumulative score (right and left) is used for the reach.

The procedure for conducting the rapid habitat assessment is presented in Table 14-2. For each of the 10 parameters, rate the overall quality of the sampling reach on a scale of 0 to 20. For riffle/run prevalent streams, record your scores for each parameter on the

TABLE 14-1. DESCRIPTIONS OF PARAMETERS USED IN THE RAPID HABITAT ASSESSMENT OF STREAMS[a]

Habitat Parameter (Prevalent Habitat Type R=Riffle/run P=Pool/glide)	Description and Rationale
Parameters Evaluated within Sampling Reach	
1. Epifaunal Substrate/ Available Cover (R, P)	Includes the relative quantity and variety of natural structures in the stream, such as cobble (riffles), large rocks, fallen trees, logs and branches, and undercut banks, available as refugia, feeding, or sites for spawning and nursery functions of aquatic macrofauna. A wide variety and/or abundance of submerged structures in the stream provides macroinvertebrates and fish with a large number of niches, thus increasing habitat diversity. As variety and abundance of cover decreases, habitat structure becomes monotonous, diversity decreases, and the potential for recovery following disturbance decreases. Riffles and runs are critical for maintaining a variety and abundance of insects in most high-gradient streams and serving as spawning and feeding refugia for certain fish. The extent and quality of the riffle is an important factor in the support of a healthy biological condition in high-gradient streams. Riffles and runs offer a diversity of habitat through variety of particle size, and, in many small high-gradient streams, will provide the most stable habitat. Snags and submerged logs are among the most productive habitat structure for macroinvertebrate colonization and fish refugia in low-gradient streams. However, "new fall" will not yet be suitable for colonization.
2A. Embeddedness (R)	Refers to the extent to which rocks (gravel, cobble, and boulders) and snags are covered or sunken into the silt, sand, or mud of the stream bottom. Generally, as rocks become embedded, the surface area available to macroinvertebrates and fish (shelter, spawning, and egg incubation) is decreased. Embeddedness is a result of large-scale sediment movement and deposition, and is a parameter evaluated in the riffles and runs of high-gradient streams. The rating of this parameter may be variable depending on where the observations are taken. To avoid confusion with sediment deposition (another habitat parameter), observations of embeddedness should be taken in the upstream and central portions of riffles and cobble substrate areas.
2B. Pool Substrate Characterization (P)	Evaluates the type and condition of bottom substrates found in pools. Firmer sediment types (e.g., gravel, sand) and rooted aquatic plants support a wider variety of organisms than a pool substrate dominated by mud or bedrock and no plants. In addition, a stream that has a uniform substrate in its pools will support far fewer types of organisms than a stream that has a variety of substrate types.
3A. Velocity and Depth Regimes (R)	Patterns of velocity and depth are included for high-gradient streams under this parameter as an important feature of habitat diversity. The best streams in most high-gradient regions will have all 4 patterns present: (1) slow-deep, (2) slow-shallow, (3) fast-deep, and (4) fast-shallow. The general guidelines are 0.5 m depth to separate shallow from deep, and 0.3 m/sec to separate fast from slow. The occurrence of these 4 patterns relates to the stream's ability to provide and maintain a stable aquatic environment.
3B. Pool Variability (P)	Rates the overall mixture of pool types found in streams, according to size and depth. The 4 basic types of pools are large-shallow, large-deep, small-shallow, and small-deep. A stream with many pool types will support a wide variety of aquatic species. Rivers with low sinuosity (few bends) and monotonous pool characteristics do not have sufficient quantities and types of habitat to support a diverse aquatic community. General guidelines are any pool dimension (i.e., length, width, oblique) greater than half the cross-section of the stream for separating large from small and 1 m depth separating shallow and deep.

[a] Modified from Barbour et al. (1999)

(continued)

TABLE 14-1[a] (Continued)

Habitat Parameter (Prevalent Habitat Type R=Riffle/ run P=Pool/ glide)	Description and Rationale
4. Sediment Deposition (R, P)	Measures the amount of sediment that has accumulated in pools and the changes that have occurred to the stream bottom as a result of deposition. Deposition occurs from large-scale movement of sediment. Sediment deposition may cause the formation of islands, point bars (areas of increased deposition usually at the beginning of a meander that increase in size as the channel is diverted toward the outer bank) or shoals, or result in the filling of runs and pools. Usually deposition is evident in areas that are obstructed by natural or manmade debris and areas where the stream flow decreases, such as bends. High levels of sediment deposition are symptoms of an unstable and continually changing environment that becomes unsuitable for many organisms.
5. Channel Flow Status (R, P)	The degree to which the channel is filled with water. The flow status will change as the channel enlarges (e.g., aggrading stream beds with actively widening channels) or as flow decreases as a result of dams and other obstructions, diversions for irrigation, or drought. When water does not cover much of the streambed, the amount of suitable substrate for aquatic organisms is limited. In high-gradient streams, riffles and cobble substrate are exposed; in low-gradient streams, the decrease in water level exposes logs and snags, thereby reducing the areas of good habitat. Channel flow is especially useful for interpreting biological condition under abnormal or lowered flow conditions. This parameter becomes important when more than one biological index period is used for surveys or the timing of sampling is inconsistent among sites or annual periodicity.
Parameters Evaluated Broader than the Sampling Reach	
6. Channel Alteration (R, P)	Is a measure of large-scale changes in the shape of the stream channel. Many streams in urban and agricultural areas have been straightened, deepened, or diverted into concrete channels, often for flood control or irrigation purposes. Such streams have far fewer natural habitats for fish, macroinvertebrates, and plants than do naturally meandering streams. Channel alteration is present when artificial embankments, riprap, and other forms of artificial bank stabilization or structures are present; when the stream is very straight for significant distances; when dams and bridges are present; and when other such changes have occurred. Scouring is often associated with channel alteration.
7A. Frequency of Riffles (or Bends) (R)	Is a way to measure the sequence of riffles and thus the heterogeneity occurring in a stream. Riffles are a source of high-quality habitat and diverse fauna, therefore, an increased frequency of occurrence greatly enhances the diversity of the stream community. For high gradient streams where distinct riffles are uncommon, a run/bend ratio can be used as a measure of meandering or sinuosity (see 7b). A high degree of sinuosity provides for diverse habitat and fauna, and the stream is better able to handle surges when the stream fluctuates as a result of storms. The absorption of this energy by bends protects the stream from excessive erosion and flooding and provides refugia for benthic invertebrates and fish during storm events. To gain an appreciation of this parameter in some streams, a longer segment or reach than that designated for sampling should be incorporated into the evaluation. In some situations, this parameter may be rated from viewing accurate topographical maps. The "sequencing" pattern of the stream morphology is important in rating this parameter. In headwaters, riffles are usually continuous and the presence of cascades or boulders provides a form of sinuosity and enhances the structure of the stream. A stable channel is one that does not exhibit progressive changes in slope, shape, or dimensions, although short-term variations may occur during floods (Gordon et al. 1992).

[a] Modified from Barbour et al. (1999) **(continued)**

TABLE 14-1[a] (Continued)

Habitat Parameter (Prevalent Habitat Type R=Riffle/run P=Pool/glide)	Description and Rationale
7B. Channel Sinuosity (P)	Evaluates the meandering or sinuosity of the stream. A high degree of sinuosity provides for diverse habitat and fauna, and the stream is better able to handle surges when the stream fluctuates as a result of storms. The absorption of this energy by bends protects the stream from excessive erosion and flooding and provides refugia for benthic invertebrates and fish during storm events. To gain an appreciation of this parameter in low gradient streams, a longer segment or reach than that designated for sampling may be incorporated into the evaluation. In some situations, this parameter may be rated from viewing accurate topographical maps. The “sequencing” pattern of the stream morphology is important in rating this parameter. In "oxbow" streams of coastal areas and deltas, meanders are highly exaggerated and transient. Natural conditions in these streams are shifting channels and bends, and alteration is usually in the form of flow regulation and diversion. A stable channel is one that does not exhibit progressive changes in slope, shape, or dimensions, although short-term variations may occur during floods (Gordon et al. 1992).
8. Bank Stability (Condition of Banks) (R, P)	Measures whether the stream banks are eroded (or have the potential for erosion). Steep banks are more likely to collapse and suffer from erosion than are gently sloping banks, and are therefore considered to be unstable. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil. Eroded banks indicate a problem of sediment movement and deposition, and suggest a scarcity of cover and organic input to streams. Each bank is evaluated separately and the cumulative score (right and left) is used for this parameter.
9. Bank Vegetative Protection (R, P)	Measures the amount of vegetative protection afforded to the stream bank and the near-stream portion of the riparian zone. The root systems of plants growing on stream banks help hold soil in place, thereby reducing the amount of erosion that is likely to occur. This parameter supplies information on the ability of the bank to resist erosion as well as some additional information on the uptake of nutrients by the plants, the control of instream scouring, and stream shading. Banks that have full, natural plant growth are better for fish and macroinvertebrates than are banks without vegetative protection or those shored up with concrete or riprap. This parameter is made more effective by defining the native vegetation for the region and stream type (i.e., shrubs, trees, etc.). In some regions, the introduction of exotics has virtually replaced all native vegetation. The value of exotic vegetation to the quality of the habitat structure and contribution to the stream ecosystem must be considered in this parameter. In areas of high grazing pressure from livestock or where residential and urban development activities disrupt the riparian zone, the growth of a natural plant community is impeded and can extend to the bank vegetative protection zone. Each bank is evaluated separately and the cumulative score (right and left) is used for this parameter.
10. Riparian Vegetated Zone Width (R, P)	Measures the width of natural vegetation from the edge of the stream bank out through the riparian zone. The vegetative zone serves as a buffer to pollutants entering a stream from runoff, controls erosion, and provides habitat and nutrient input into the stream. A relatively undisturbed riparian zone supports a robust stream system; narrow riparian zones occur when roads, parking lots, fields, lawns, bare soil, rocks, or buildings are near the stream bank. Residential developments, urban centers, golf courses, and rangeland are the common causes of anthropogenic degradation of the riparian zone. Conversely, the presence of "old field" (i.e., a previously developed field not currently in use), paths, and walkways in an otherwise undisturbed riparian zone may be judged to be inconsequential to altering the riparian zone and may be given relatively high scores. For variable size streams, the specified width of a desirable riparian zone may also be variable and may be best determined by some multiple of stream width (e.g., 4 x wetted stream width). Each bank is evaluated separately and the cumulative score (right and left) is used for this parameter.

[a] Modified from Barbour et al. (1999)

TABLE 14-2. PROCEDURE FOR CONDUCTING THE RAPID HABITAT ASSESSMENT

1. Based on observations during previous sample collection and field measurement activities, classify the sampling reach as predominantly flowing water habitat ("Riffle/run") or slow water habitat ("Pool/glide").

2. Select the appropriate version of the Rapid Habitat Assessment Form ("Riffle/Run Prevalence" or "Pool/Glide Prevalence") based on the classification in Step 1.

3. For each of the 10 habitat parameters, determine the general "quality" category ("POOR", "MARGINAL", "SUB-OPTIMAL", or "OPTIMAL") of the entire sampling reach. Assign and circle a score from the values available within each quality category. For Parameters 1 through 7, the sampling reach can be scored from 0 (worst) to 20 (best). For Parameters 8 through 10, each bank is evaluated separately (from 0 to 10), and the cumulative score for both right and left banks are used.

4. After the sampling reach has been scored for all parameters, transfer the score circled for each category to the corresponding "SCORE" box in the "HABITAT PARAMETER" column of the assessment form.

5. Sum the scores recorded in Step 4 over all 10 habitat parameters. Record the total score for the sampling reach in the "TOTAL SCORE" box on page 1 of the assessment form. The total score can range from 0 to 200.

Reviewed by (Initials): RC

RAPID HABITAT ASSESSMENT FORM: RIFFLE/RUN - STREAM

SITE ID: WXXP99-9999 DATE: 07/01/2000

HABITAT PARAMETER	CONDITION CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
1. Epifaunal Substrate/ Available Cover	Greater than 70% of substrate favorable for epifaunal colonization and fish cover; mix of snags, submerged logs, undercut banks, cobble or other stable habitat and at stage to allow full colonization potential; (i.e., logs/snags that are NOT new fall and NOT transient.)	40-70% mix of stable habitat; well-suited for full colonization potential; adequate habitat for maintainance of populations; presence of additional substrate in the form of newfall, but not yet prepared for colonization (may rate at high end of scale).	20-40% mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed.	Less than 20% stable habitat; lack of habitat is obvious; substrate unstable or lacking.
Score: 12	20 19 18 17 16	15 14 13 (12) 11	10 9 8 7 6	5 4 3 2 1 0
2. Embeddedness	Gravel, cobble, and boulder particles are 0-25% surrounded by fine sediment. Layering of cobble provides diversity of niche space.	Gravel, cobble, and boulder particles are 25-50% surrounded by fine sediment.	Gravel, cobble, and boulder particles are 50-75% surrounded by fine sediment.	Gravel, cobble, and boulder particles are more than 75% surrounded by fine sediment.
Score: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
3. Velocity/Depth Regime	All four velocity/depth regimes present (slow-deep, slow-shallow, fast-deep, fast-shallow). (Slow is less than 0.3 m/s, deep is greater than 0.5 m.)	Only 3 of the 4 regimes present (if fast-shallow is missing, score lower than if missing other regimes).	Only 2 of the 4 habitat regimes present (if fast-shallow or slow-shallow are missing, score low).	Dominated by 1 velocity/depth regime (usually slow-deep).
Score: 15	20 19 18 17 16	(15) 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
4. Sediment Deposition	Little or no enlargement of islands or point bars and less than 5% of the bottom affected by sediment deposition.	Some new increases in bar formation, mostly from gravel, sand or fine sediment; 5-30% of the bottom affected; slight deposition in pools.	Moderate deposition of new gravel, sand or fine sediment on old and new bars; 30-50% of the bottom affected; sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools prevalent.	Heavy deposits of fine material; increased bar development; more than 50% of the bottom changing frequently; pools almost absent due to substantial sediment deposition.
Score: 14	20 19 18 17 16	15 (14) 13 12 11	10 9 8 7 6	5 4 3 2 1 0
5. Channel Flow Status	Water reaches base of both lower banks, and minimal amount of channel substrate is exposed.	Water fills over 75% of the available channel; or less than 25% of channel substrate is exposed.	Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed.	Very little water in channel and mostly present as standing pools.
Score: 12	20 19 18 17 16	15 14 13 (12) 11	10 9 8 7 6	5 4 3 2 1 0
6. Channel Alteration	Channelization or dredging absent or minimal; stream with normal pattern.	Some channelization present, usually in areas of bridge abutments; evidence of past channelization, i.e., dredging, (greater than past 20 yr) may be present, but recent channelization is not present.	Channelization may be extensive; embankments or shoring structures present on both banks; and 40 to 80% of stream reach channelized and disrupted.	Banks shored with gabion or cement; over 80% of the stream reach channelized and disrupted. Instream habitat greatly altered or removed entirely.
Score: 18	20 19 (18) 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

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03/31/2000 2000 Riffle Run

Figure 14-1. Rapid Habitat Assessment Form for riffle/run prevalent streams (page 1).

Reviewed by (Initials): RC

RAPID HABITAT ASSESSMENT FORM: RIFFLE/RUN (continued) - STREAM

SITE ID: WXXP99-9999 DATE: 07/01/2000

HABITAT PARAMETER	CONDITION CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
7. Frequency of Riffles (or bends)	Occurrence of riffles relatively frequent; ratio of distance between riffles divided by width of the stream greater than 7:1 (generally 5 to 7); variety of habitat is key. In streams where riffles are continuous, placement of boulders or other large, natural obstruction is important.	Occurrence of riffles infrequent; distance between riffles divided by width of stream is between 7 to 15.	Occasional riffle or bend; bottom contours provide some habitat; distance between riffles divided by width of stream is between 15 to 25.	Generally all flat water or shallow riffles; poor habitat; distance between riffles divided by width of stream is a ratio of over 25.
Score: 13	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
8. Bank Stability (score each bank) NOTE: Determine left or right side by facing downstream.	Banks stable; evidence of erosion or bank failure absent or minimal; little potential for future problems. Less than 5% of bank affected.	Moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion.	Moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; obvious bank sloughing; 60-100% of bank has erosional scars.
Left Bank Score: 7	Left Bank: 10 9	8 7 6	5 4 3	2 1 0
Right Bank Score: 5	Right Bank: 10 9	8 7 6	5 4 3	2 1 0
9. Vegetative Protection (score each bank)	More than 90% of the streambank surfaces and immediate riparian zone covered by native vegetation, including trees, understory shrubs, or nonwoody macrophytes; vegetative disruption through grazing or mowing minimal or not evident; almost all plants allowed to grow naturally.	70-90% if the streambank surfaces covered by native vegetation; but one class of plants is not well represented; disruption evident but not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	50-70% of the streambank surfaces covered by vegetation; disruptions obvious; patches of bare soil or closely cropped vegetation common; less than one-half of the potential plant stubble height remaining.	Less than 50% of the streambank surfaces covered by vegetation; disruption of streambank vegetation is very high; vegetation has been removed to 5 centimeters or less in average stubble height.
Left Bank Score: 8	Left Bank: 10 9	8 7 6	5 4 3	2 1 0
Right Bank Score: 7	Right Bank: 10 9	8 7 6	5 4 3	2 1 0
10. Riparian Vegetative Zone Width (score each bank)	Width of riparian zone greater than 18 meters; human activities (i.e., parking lots, roadbeds, clear-cuts, lawns, or crops) have not impacted the zone.	Width of riparian zone 12-18 meters; human activities have impacted zone only minimally.	Width of riparian zone 6-12 meters; human activities have impacted zone a great deal.	Width of riparian zone less than 6 meters; little or no riparian vegetation due to human activities.
Left Bank Score: 6	Left Bank: 10 9	8 7 6	5 4 3	2 1 0
Right Bank Score: 5	Right Bank: 10 9	8 7 6	5 4 3	2 1 0

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03/15/2000 2000 Riffle Run

Figure 14-2. Rapid Habitat Assessment Form for riffle/run prevalent streams (page 2).

riffle/run version of the Rapid Habitat Assessment Form as shown in Figures 14-1 and 14-2. If the stream is classified as a pool/glide prevalent stream, record your scores for each parameter on the pool/glide version of the Rapid Habitat Assessment Form as shown in Figures 14-3 and 14-4. Transfer the scores assigned for each parameter to the box in the left-hand column of the form. Sum the scores for each parameter and record the total score in the box at the top of page 1 of the form.

14.2 VISUAL STREAM ASSESSMENT

The 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 of the form. Complete the assessment form after all other sampling and measurement activities have been completed. Consider only things at or upstream of the X-site (things that may impact the sample reach). Take into account all observations the sampling team has made while at the site. The assessment includes the following components: watershed activities and observed disturbances, site characteristics, weather during sampling, and a general assessment. The procedure for conducting the visual assessment of the sampling reach is presented in Table 14-3. Record data and observations for each component of the assessment on the Assessment Form as shown in Figure 14-5.

Each watershed activity or disturbance is rated into one of four categories of abundance or influence: not observed, low, medium, or high. Leave the line blank for any activity or disturbance type not observed. The distinction between low, medium, and high will be subjective. For example, if there are 2-3 houses away from the stream, the rating for “Houses” may be low. If the stream is in a suburban housing development, rate it as high. Similarly, a small patch of clear cut logging on a hill overlooking the stream would be rated as low. Logging activity right on the stream shore, however, would be rated as high.

When assessing site characteristics, imagine a circle with a 200 m radius around the x-site (400 m diameter). Consider the land use and other activities within this circle. Water body character is defined as "the physical habitat integrity of the water body, largely a function of riparian and littoral habitat structure, volume change, trash, turbidity, slicks, scums, color, and odor.” Water body character is assessed using two attributes, the degree of human development, and aesthetics. Rate each of these attributes on a scale of 1 to 5. For development, give the stream a "5" rating if it is pristine, with no signs of any human development. A rating of "1" indicates a stream which is totally developed (e.g., the entire

Reviewed by (Initials): Rc

RAPID HABITAT ASSESSMENT FORM: GLIDE/POOL - STREAMS

SITE ID: WXXP99-9999 DATE: 07/01/2000

HABITAT PARAMETER	CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
1. Epifaunal Substrate/ Available Cover	Greater than 50% of substrate favorable for epifaunal colonization and fish cover; mix of snags, submerged logs, undercut banks, cobble or other stable habitat and at stage to allow full colonization potential (i.e. logs/snags that are NOT new fall and NOT transient.)	30-50% mix of stable habitat; well-suited for full colonization potential; adequate habitat for maintenance of populations; presence of additional substrate in the form of newfall, but not yet prepared for colonization (may rate at high end of scale).	10-30% mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed.	Less than 10% stable habitat; lack of habitat is obvious; substrate unstable or lacking.
Score: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
2. Pool Substrate Characterization	Mixture of substrate materials, with gravel and firm sand prevalent; root mats and submerged vegetation common.	Mixture of soft sand, mud, or clay; mud may be dominant; some root mats and submerged vegetation present.	All mud or clay or sand bottom; little or no root mat; no submerged vegetation.	Hard-pan clay or bedrock; no root mat or vegetation.
Score: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
3. Pool Variability	Even mix of large-shallow, large-deep, small shallow, small-deep pools present.	Majority of pools large-deep; very few shallows.	Shallow pools much more prevalent than deep pools.	Majority of pools small-shallow or absent.
Score: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
4. Sediment Deposition	Little or no enlargement of islands or point bars and less than 20% of the bottom affected by sediment deposition.	Some new increases in bar formation, mostly from gravel, sand or fine sediment; 20-50% of the bottom affected; slight deposition in pools.	Moderate deposition of new gravel, sand or fine sediment on old and new bars; 50-80% of the bottom affected; sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools prevalent.	Heavy deposits of fine material; increased bar development; more than 80% of the bottom changing frequently; pools almost absent due to substantial sediment deposition.
Score: 7	20 19 18 17 16	15 14 13 12 11	10 9 8 (7) 6	5 4 3 2 1 0
5. Channel Flow Status	Water reaches base of both lower banks, and minimal amount of channel substrate is exposed.	Water fills over 75% of the available channel; or less than 25% of channel substrate is exposed.	Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed.	Very little water in channel and mostly present as standing pools.
Score: 18	20 19 (18) 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
6. Channel Alteration	Channelization or dredging absent or minimal; stream with normal pattern.	Some channelization present, usually in areas of bridge abutments; evidence of past channelization, i.e., dredging, (greater than past 20 yr) may be present, but recent channelization is not present.	Channelization may be extensive; embankments or shoring structures present on both banks; and 40 to 80% of stream reach channelized and disrupted.	Banks shored with gabion or cement; over 80% of the stream reach channelized and disrupted. Instream habitat greatly altered or removed entirely.
Score: 16	20 19 18 17 (16)	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

03/31/2000 Glide Pool

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Figure 14-3. Rapid Habitat Assessment Form for pool/glide prevalent streams (page 1).

Reviewed by (Initials): RC

RAPID HABITAT ASSESSMENT FORM: GLIDE/POOL (continued) - STREAMS

SITE ID: WXXP99-9999 DATE: 07/01/2000

HABITAT PARAMETER	CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
7. Channel Sinuosity	The bends in the stream increase the stream length 3 to 4 times longer than if it was in a straight line. (Note- channel braiding is considered normal in coastal plains and other low-lying areas. This parameter is not easily rated in these areas.)	The bends in the stream increase the stream length 2 to 3 times longer than if it was in a straight line.	The bends in the stream increase the stream length 1 to 2 times longer than if it was in a straight line.	Channel straight; waterway has been channelized for a long distance.
Score: 13	20 19 18 17 16	15 14 (13) 12 11	10 9 8 7 6	5 4 3 2 1 0
8. Bank Stability (score each bank) NOTE: Determine left or right side by facing downstream.	Banks stable; evidence of erosion or bank failure absent or minimal; little potential for future problems. Less than 5% of bank affected.	Moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion.	Moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; obvious bank sloughing; 60-100% of bank has erosional scars.
Left Bank Score: 9	Left Bank: 10 (9)	8 7 6	5 4 3	2 1 0
Right Bank Score: 10	Right Bank: (10) 9	8 7 6	5 4 3	2 1 0
9. Vegetative Protection (score each bank)	More than 90% of the streambank surfaces and immediate riparian zone covered by native vegetation, including trees, understory shrubs, or nonwoody macrophytes; vegetative disruption through grazing or mowing minimal or not evident; almost all plants allowed to grow naturally.	70-90% if the streambank surfaces covered by native vegetation; but one class of plants is not well represented; disruption evident but not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	50-70% of the streambank surfaces covered by vegetation; disruptions obvious; patches of bare soil or closely cropped vegetation common; less than one-half of the potential plant stubble height remaining.	Less than 50% of the streambank surfaces covered by vegetation; disruption of streambank vegetation is very high; vegetation has been removed to 5 centimeters or less in average stubble height.
Left Bank Score: 4	Left Bank: 10 9	8 7 6	5 (4) 3	2 1 0
Right Bank Score: 6	Right Bank: 10 9	8 7 (6)	5 4 3	2 1 0
10. Riparian Vegetation Zone Width (score each bank)	Width of riparian zone greater than 18 meters; human activities (i.e., parking lots, roadbeds, clear-cuts, lawns, or crops) have not impacted the zone.	Width of riparian zone 12-18 meters; human activities have impacted zone only minimally.	Width of riparian zone 6-12 meters; human activities have impacted zone a great deal.	Width of riparian zone less than 6 meters; little or no riparian vegetation due to human activities.
Left Bank Score: 5	Left Bank: 10 9	8 7 6	(5) 4 3	2 1 0
Right Bank Score: 7	Right Bank: 10 9	8 (7) 6	5 4 3	2 1 0

03/31/2000 Glide Pool

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Figure 14-4. Rapid Habitat Assessment Form for glide/pool prevalent streams (page 2).

TABLE 14-3. PROCEDURE FOR CONDUCTING THE FINAL VISUAL ASSESSMENT OF A STREAM

1. After all other sampling and measurement activities are completed, fill out the header section of an Assessment Form. Use your perceptions obtained during the course of the day, while at the stream or driving/walking through the catchment to complete the remainder of the form. Consider only things at or upstream of the x-site.

2. WATERSHED ACTIVITIES AND DISTURBANCES OBSERVED: Rate each type of activity or disturbance listed on the form as either "Not observed", "Low", "Medium", or "High", and record the rating on the Assessment Form. Keep in mind that ratings will be somewhat subjective and that an extensive effort to quantify the presence and intensity of each type of stressor is not required. General categories of activities and types of disturbance are described below:

   - Residential: The presence of any of the listed disturbances adjacent to or near the stream.
   - Recreational: The presence of organized public or private parks, campgrounds, beaches or other recreation areas around the stream. If there are signs of informal areas of camping, swimming or boating around the stream (e.g., swimming hole), record them as "primitive" parks, camping.
   - Agriculture: The presence of cropland, pasture, range, orchards, poultry, and/or livestock. Also note any evidence of water withdrawals for agriculture.
   - Industrial: Any industrial activity (e.g., canning, chemical, pulp), commercial activity (stores, businesses) or logging/mining activities around the stream or in the catchment. Describe in more detail in the comments section.
   - Management: Any evidence of water treatment, dredging or channelization, flow control structures, fish stocking, dams or other management activities.

   Any oddities, or further elaboration should be recorded in the Comments section.

3. SITE CHARACTERISTICS: (based on a circle with a 200 m radius around the x-site)

   - WATER BODY CHARACTER: Assign a rating of 1 (highly disturbed) to 5 (pristine) based on your general impression of the intensity of impact from human disturbance. Place an "X" in the box next to the assigned rating on the Assessment Form. Assign a rating to the stream based on overall aesthetic quality, based on your opinion of how suitable the stream water is for recreation and aesthetic enjoyment today. Place and "X" in the box next to the assigned rating on the Assessment Form.

     5. Beautiful, could not be any nicer.
     4. Very minor aesthetic problems; excellent for swimming, boating, enjoyment.
     3. Enjoyment impaired.
     2. Level of enjoyment substantially reduced.
     1. Enjoyment nearly impossible.

(continued)

TABLE 14-3 (Continued)

- Beaver: If you noticed any signs of beaver presence in the stream (chewed sticks, trees, dams, lodges) rate the beaver presence as either rare or common. If no beaver signs were present, mark the absent box. Also rate the amount of flow modification caused by any **beaver activity** as none, minor, or major.

- Dominant Land Use: Make one estimate of the dominant land use in the circle around the x-site. Pick just one land use from among Forest, Agriculture, Range, Urban, Suburban/Town. If there are other major land uses, make note of them in the General Assessment section of the form. If forest is the dominant land use, make a guess as to the dominant age class of the forest (0-25, 25-75, or > 75 years).

3. WEATHER: record a very brief description of the weather conditions during stream sampling (e.g., sunny, fair, partly cloudy, overcast, light rain, unseasonably warm, cold, or hot, etc.). Any unusual weather right before sampling (e.g., heavy rain, 6 inches of snow) is also worth noting here.

4. GENERAL ASSESSMENT: record comments on wildlife observed, perceived diversity of terrestrial/riparian vegetation, or overall biotic integrity on the Assessment Form. Record any information regarding the past or present characteristics or condition of the stream provided by local residents here as well.

STREAM ASSESSMENT FORM - STREAMS/RIVERS

Reviewed by (initial): ZP

SITE ID: WXXP99-9999 DATE: 07/01/2001

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, WITH LIGHT RAIN IN THE PREVIOUS 24 HOURS. AIR TEMP 28° AT 11 AM.

GENERAL ASSESSMENT (Biotic integrity, Vegetation diversity, Local anecdotal information)

RIPARIAN TREES AGE CLASS: 25-75 YR. THOUGH TREE DENSITY IS LOW.
LOCAL CONTACT REMEMBERS A DAM LOCATED JUST DOWNSTREAM OF X-SITE THAT WAS WASHED AWAY 10 YR AGO DURING A LARGE FLOOD EVENT.
NO SIGNS OF BIRDS OR WILDLIFE OBSERVED DURING VISIT.

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03/26/2001 2001 Stream Assessment

Figure 14-5. Stream Assessment Form (page 1).

stream is lined with houses, or the riparian zone has been removed). For aesthetics, base your decision on any factor about the stream that bothers you (e.g., trash, algal growth, weed abundance, overcrowding). Also, rate the presence/absence of beaver and the dominant land use within this circle according to the classes listed on the form

The weather and general assessment component includes any observations that will help in data interpretation in the pertinent section. The weather component is just a place to record a brief description of the weather during sampling or just before sampling. General assessment comments can include comments on wildlife observed, diversity of terrestrial/riparian vegetation, overall biotic integrity, or any other observation. Comments from locals about current or past conditions are often useful and should be recorded in this section as well. The back side of the form (Figure 14-6) is available for additional general comments.

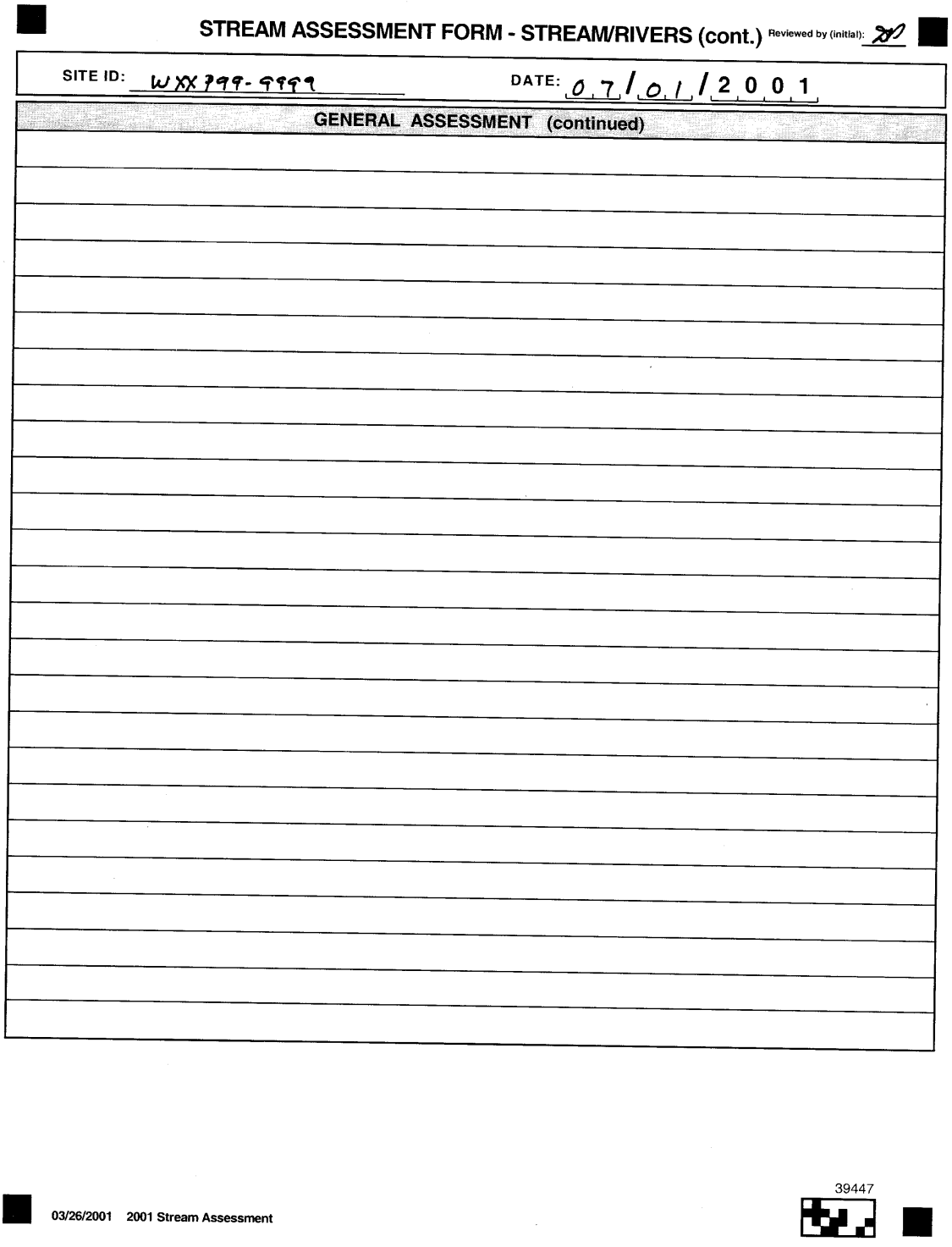
14.3 EQUIPMENT AND SUPPLIES

Figure 14-7 is a checklist of the supplies required to complete the visual stream assessment. This checklist may differ from the checklists presented in Appendix A, which are used at a base site to ensure that all equipment and supplies are brought to and are available at the stream site. Field teams are required to use the checklist presented in this section to ensure that equipment and supplies are organized and available to conduct the protocols efficiently.

14.3 LITERATURE CITED

Barbour, M.T., J. Gerritsen, B.D. Snyder, and J.B. Stribling. 1999. *Rapid Bioassessment Protocols for Use in Streams and Wadeable Rivers: Periphyton, Benthic Macroinvertebrates, and Fish.* Second Edition. EPA/841-B-99-002. U.S. Environmental Protection Agency, Office of Water, Assessment and Watershed Protection Division, Washington, D.C.

Gordon, N.D., T.A. McMahon, and B.L. Finlayson. 1992. *Stream hydrology: an introduction for ecologists.* John Wiley and Sons, Inc., West Sussex, England.



STREAM ASSESSMENT FORM - STREAM/RIVERS (cont.) Reviewed by (initial):

SITE ID: WXX799-9999 DATE: 07/01/2001

GENERAL ASSESSMENT (continued)

03/26/2001 2001 Stream Assessment

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Figure 14-6. Stream Assessment Form (page 2).

EQUIPMENT AND SUPPLIES FOR RAPID HABITAT AND VISUAL STREAM ASSESSMENTS

QTY.	Item	
1	Rapid Habitat Assessment Form for Riffle/run prevalent streams	
1	Rapid Habitat Assessment Form for Pool/glide prevalent streams	
1	Assessment Form for visual stream assessment	
6	Soft (#2) lead pencils	
1	Covered clipboard or forms holder	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for rapid habitat and visual assessments	

Figure 14-7. Checklist of equipment and supplies required for rapid habitat and visual stream assessments.

Lazorchak, J.M., A.T. Herlihy, and J. Green. 1998. Rapid Habitat and Visual Stream Assessments. pp. 193-209 IN: J.M. Lazorchak, D.J. Klemm, and D.V. Peck (Eds.). *Environmental Monitoring and Assessment Program-Surface Waters: Field Operations and Methods for Measuring the Ecological Condition of Wadeable Streams*. EPA/620/R-94/004F. U.S. Environmental Protection Agency, Washington, D.C.

Plafkin, J.L., M.T. Barbour, K.D. Porter, S.K. Gross, and R.M. Hughes. 1989. *Rapid Bioassessment Protocols for Use in Streams and Rivers: Benthic Macroinvertebrates and Fish.* EPA/440/4-89/001. U.S. Environmental Protection Agency, Assessment and Watershed Protection Division, Washington, D.C.

NOTES

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SECTION 15
FINAL SITE ACTIVITIES

by
James M. Lazorchak[3]

Before leaving a stream site, the team leader reviews all of the data forms and sample labels for accuracy, completeness, and legibility. A second team member inspects all sample containers and packages them in preparation for transport, storage, or shipment. Refer to Section 3 for details on preparing and shipping samples.

When reviewing field data forms, ensure that all required data forms for the stream have been completed. Confirm that the stream identification code, the year, the visit number, and the date of the 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 and use no "shorthand" or abbreviations. 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.

When inspecting samples, ensure that each sample is labeled, all labels are completely filled in and legible, 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.

The other team members should return all of the equipment and supplies to the vehicle for transport and clean up the stream site. Pack all equipment and supplies in the vehicle for transport. Keep them organized so they can be inventoried using the equipment and supply checklists presented in Appendix A. Clean up and dispose of all waste material at the stream site. Transport it out of the area if necessary.

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[3] U.S. EPA, National Exposure Research Laboratory, Ecological Exposure Research Division, 26 W. Martin Luther King Dr., Cincinnati, OH 45268.

NOTES

APPENDIX A
EQUIPMENT AND SUPPLY CHECKLISTS

FIELD DATA FORMS AND SAMPLE LABELS

Number per site	Item	
1	Verification Form	
1	Sample Collection Form	
11 + extras	Channel/Riparian Cross-section and Thalweg Profile Forms	
1	Slope and Bearing Form	
1	Legacy Tree/ Invasive Plant form	
1	Field Measurement and Channel Constraint Form	
1	Torrent Evidence Assessment Form	
2-3	Vertebrate Collection Form	
1	Rapid Habitat Assessment Form for Riffle/run prevalent streams (optional)	
1	Rapid Habitat Assessment Form for Pool/glide prevalent streams (optional)	
1	Assessment Form for visual stream assessment	
4 + extras	Sample Tracking Form	
3	Water chemistry labels (same ID number)	
3t	Periphyton labels (same ID number)	
1	Reachwide Benthic sample labels, with preprinted ID numbers	
1	Targeted Riffle Benthic sample labels with preprinted ID numbers	
1 sheet	Benthic labels for extra containers (no preprinted ID number)	
1 sheet	Blank benthic sample labels on waterproof paper for inside of jars	
1 sheet	Pre-printed aquatic vertebrate jar labels (4) and voucher bag tags (36), all with same preprinted sample ID number	
1 sheet	Fish tissue sample labels (up to 16 different sample ID number)	
2 copies	Field operations and methods manual	
2 sets	Laminated sheets of procedure tables and/or quick reference guides	

OFFICE SUPPLIES AND TOOLS

Number per site	Item	
1	Dossier of access information for scheduled stream site	
1	Topographic map with “X-site” marked	
1	Site information sheet with map coordinates and elevation of X-site	
1	Sampling itinerary form or notebook	
1	Safety log and/or personal safety information for each team member	
4	Covered clipboards or forms holders	
1	Field notebook (optional)	
12	Soft (#2) lead pencils	
6	Fine-tip indelible markers	
1 roll	Duct tape	
1 pr	Scissors for cutting labels	
1	Pocket knife or multipurpose tool	
1	Battery charger (if needed for electrofishing unit)	
1	Toolbox with basic tools needed to maintain/repair sampling gear	

PERSONAL EQUIPMENT AND SUPPLIES

Number per site	Item	
1 pair per person	Chest waders with felt-soled boots for safety and speed if waders are the neoprene "stocking" type. Hip waders can be used in shallower streams (except for electrofishing).	
1 per person	Life vests	
3 pair	Polarized sunglasses	
1	First aid kit	
1 per person	Rain gear	
1 or 2	Fisherman's vest for physical habitat characterization.	
1 per person	Safety Whistles	
1 pr. per person	Earplugs (if gas-powered generators are used)	
1 per person	Day packs, backs, fanny packs, and/or dry bags for personal gear	
1 ea.	Insect repellent, sunscreen, Tec-nu (for poison oak), hand sanitizer, water purifier unit	
1	Patch kit for waders	

CHEMICALS

Number per site	Item	
1 pr	Safety glasses	
2 pr	Chemical-resistant gloves	
1	Laboratory apron, resistant to ethanol and formalin	
1	Cooler (with suitable absorbent material) for transporting ethanol and samples	
2 gal	95% ethanol	
1	Cooler (with suitable absorbent material) for transporting formaldehyde/ formalin	
2 gal	10% (buffered) formalin solution OR 0.2 gal buffered formaldehyde solution	
1 gal	Sparquat® disinfectant	
	Gasoline for electrofishing unit in approved container	

PACKING AND SHIPPING SUPPLIES

Number per site	Item	
	Ice (also dry ice if it is used to ship frozen samples) or ice substitute packs	
1 box	1-gal heavy-duty self-sealing (e.g., with a zipper-type closure) plastic bags	
1-box	30-gal plastic garbage bags for lining shipping containers	
1 roll	Clear tape for sealing shipping containers	
2 pkg.	Clear tape strips for covering labels	
4 rolls	Plastic electrical tape	
3	Insulated shipping containers for samples	
1	Portable freezer, cooler with dry ice, or cooler with bags of ice (or substitute ice packs) to store frozen samples (special containers may be needed if dry ice is used)	
2	Containers and absorbent material suitable to transport and/or ship samples preserved in formalin or ethanol	
6	Shipping airbills and adhesive plastic sleeves	

SITE VERIFICATION AND SAMPLING REACH LAYOUT

Number per site	Item	
1	GPS receiver and operating manual	
	Extra batteries for GPS receiver	
1	Surveyor's telescoping leveling rod (round profile, metric scale, 7.5 m extended)	
1	50-m fiberglass measuring tape with reel	
2 rolls	Surveyor's flagging tape (2 colors)	
1	Waterproof camera and film (or digital camera)	

WATER CHEMISTRY

Number per site	Item	
1	Dissolved oxygen/Conductivity/Temperature meter with probe and operating manual (optional)	
1	DO repair kit with additional membranes and probe filling solution (optional)	
1	Conductivity meter, probe, and operating manual (if not integrated with DO/ Temp meter (optional)	
	Extra batteries for dissolved oxygen and conductivity meters (optional)	
1	500-mL plastic bottle of conductivity QCCS labeled "Rinse" (in plastic bag) (optional)	
1	500-mL plastic bottle of conductivity QCCS labeled "Test" (in plastic bag) (optional)	
1	500-mL plastic bottle of deionized water to store conductivity probe (optional)	
1	Field thermometer	
1	500 mL plastic beaker with handle (in clean plastic bag)	
1	4-L cubitainer	
2	60 mL plastic syringes	
1	½ gal. size plastic container with snap-on lid to hold filled syringes	
2	Syringe valves	

STREAM DISCHARGE

Number per site	Item	
1	Current velocity meter and probe, with operating manual (e.g. Marsh-McBirney Model 201, Swoffer Model 2100, or equivalent)	
1	Top-set wading rod (metric scale) for use with current velocity meter	
1	Portable Weir with 60° "V" notch (optional)	
1	Plastic sheeting to use with weir (optional)	
1	Plastic bucket (or similar container) with volume graduations	
1	Stopwatch	
1	Neutrally buoyant object (e.g., orange, small rubber ball, stick, bobber)	

PHYSICAL HABITAT

Number per site	Item	
1	Fisherman's vest with lots of pockets and snap fittings.	
1	50-m tape measure	
1	Clinometer with percent and degree scales.	
1	Lightweight telescoping camera tripod, (necessary only if slope measurements are being determined by only one person)	
1	½-inch diameter PVC pipe, 2-3 m long, each marked at the same height (for use in slope determinations involving two persons)	
1	Spherical convex canopy densiometer, modified with taped "V"	
1	Bearing compass (Backpacking type)	
1	Meter stick. Alternatively, a short (1-2 m) rod or pole (e.g., a ski pole) with cm markings for thalweg measurements	

PERIPHYTON

Number per site	Item	
1	Large funnel (15-20 cm diameter)	
1	12-cm^2 area delimiter (3.8 cm diameter pipe, 3 cm tall)	
1	Stiff-bristle Toothbrush with handle bent at 90^o angle	
1	1-L wash bottle for stream water	
1	1-L wash bottle containing deionized water	
1	500-mL plastic bottle for composite index sample labeled “PERIPHYTON”	
1	35-60 mL plastic syringe (catheter-tip)	
3	50-mL screw-top centrifuge tubes (or similar sample vials)	
1 box	Glass-fiber filters for chlorophyll and biomass samples	
1 pair	Forceps for filter handling.	
1	25-mL or 50-mL graduated cylinder (or use a 50-mL centrifuge tube with volume graduations)	
1	Filtration unit, including filter funnel, cap, filter holder, and receiving chamber	
1	Hand-operated vacuum pump with length of flexible plastic tubing	
1	Small syringe or bulb pipette for dispensing formalin	
1	small, lightproof plastic bag for storing chlorophyll and biomass samples	

BENTHIC MACROINVERTEBRATES

Number per site	Item	
1	Modified kick net (500 µm mesh) and 4-ft handle	
	Spare net(s) for the kick net sampler or extra sampler	
2	Buckets, plastic, 8- to 10-qt capacity, labeled “REACHWIDE” and “TARGETED RIFFLE”	
1	Sieve, U.S. Std. No. 35 (500 µm mesh), or Sieve bucket with 500-µm mesh openings	
2 pr. ea.	Watchmakers’ and curved tip forceps	
1	Small spatula, spoon, or scoop to transfer sample	
1	Funnel, with large bore spout	
4 to 6 ea.	Sample jars, HDPE plastic with leakproof screw caps, 500-mL and 1-L capacity, suitable for use with ethanol	
1 pkg.	Kim wipes in small self-sealing plastic bag	
1	Screw-top pail with absorbent material to transport preserved samples	

AQUATIC VERTEBRATES AND FISH TISSUE CONTAMINANTS

Number per site	Item	
1	Gasoline or battery-powered backpack electrofishing unit with electrode wand	
	Extra battery (or gasoline)	
4 pr	heavy-duty rubber gloves for electrofishing	
2	Long-handled dip nets (0.6 cm mesh) with insulated handles	
1	Minnow seine (2m × 1.25 m, 0.6 cm mesh) with brailles	
1	Large seines (3 m × 2 m, 0.6 cm mesh) with brailles	
4	Collapsible buckets for holding and processing aquatic vertebrates	
1 set	Taxonomic reference books and keys for fishes and amphibians of the region	
1-2	Fish measuring board	
5-20	Small nylon mesh bags or nylon stockings for holding voucher specimens	
1	Jackknife for preparing larger voucher specimens for preservation	
1 ea.	1 and 2-L HDPE plastic jars with leakproof screw-top caps for voucher samples	
4	carbon dioxide tablets (Alka-Seltzer® or equivalent)	
1 roll	Aluminum foil to make 18" x 11" rectangles (10 per site)	
1-2 L	10% (buffered) formalin solution OR 0.2 gal buffered formaldehyde solution	
1	Cooler to hold preserved voucher sample jars	
1	Aquarium net	
1 ea.	All required collection permits (State, Federal, Tribal)	