# ENVIRONMENTAL MONITORING AND ASSESSMENT PROGRAM

# SURFACE WATERS FIELD OPERATIONS MANUAL FOR LAKES

The information in this Adobe Acrobat Reader PDF file is one of several PDF files extracted from this report. The PDF files from the report are:

lake_ove.pdf Overview of EMAP Surface Waters Lake Sampling, daily operations, lake verification and index site location, and general lake assessment (Sections 1, 2, 3, 4, 9)

lake_hab.pdf Protocols for temperature, dissolved oxygen, shoreline physical habitat (Section 5)

lake_fis.pdf Protocols for fish sampling (Section 6)

lake_wat.pdf Protocols for Secchi transparency, water sample collection, chlorophyll a, zooplankton, sediment diatom (Section 7)

lake_ben.pdf Protocols for benthic invertebrate sampling (Section 8)

lake_avi.pdf Protocols for avian assemblages (Appendix A)

lake_vis.pdf Lake-Visit Checklists for all Field Measurements (Appendix B)

field_for.pdf Field Data Forms for all Field Measurements (Appendix C)

The Table of Contents, acknowledgments, notice page, listing of figures, listing of tables, and listing of acronyms for the document appear at the end of each pdf file.

In all cases, the source for this information for citation purposes is:

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# ENVIRONMENTAL MONITORING AND ASSESSMENT PROGRAM SURFACE WATERS

# FIELD OPERATIONS MANUAL FOR LAKES

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## ABSTRACT

The methods and instructions for field operations presented in this manual for lake surveys were developed and tested through 4 years of pilot and demonstration projects from 1991 through 1994. 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 lake 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. In addition to methodology, additional information on data management and other logistical aspects is integrated into the procedures and overall operational scenario. Procedures are described for collecting chlorophyl *a*, water, sedimentary diatoms, and zooplankton data in conjunction with the development of standard methods to obtain acceptable index samples for macrobenthos, fish assemblage, fish tissue contaminants, riparian birds, and physical habitat structure. The manual describes field implementation of these methods and the logistical foundation constructed during field projects. The manual includes flow charts with overall summaries of specific field activities required to visit a lake 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.

# SECTION 8
# BENTHIC INVERTEBRATE SAMPLING

by
Wesley L. Kinney, R. O. Brinkhurst, Thomas R. Whittier, and David V. Peck

There are two separate activities for benthic invertebrate sampling. The first is a quantitative sampling of sublittoral sediments for all benthic invertebrate organisms using the sediment coring device (Figure 7-3). These procedures are detailed in sections 8.1 and 8.2. The second is a qualitative survey for the presence of zebra mussels (*Dreissena sp.*). These procedures are described in Section 8.3.

Benthos sampling is restricted to the sublittoral zones of lakes. Wherever possible, collect samples in weed-free areas. Take single core samples in the soft sediments at 10 sampling sites located at or near the 10 physical habitat stations established for physical habitat characterizations (Section 5). Very rigid quality assurance practices must be observed in the field. Prior to launching the boat, ensure that all sample containers and forms are filled out for lake ID, date, and sample type where required. Criteria for accepting or rejecting a sample are specified in the following procedures. Every attempt should be made to obtain the full number of cores. To ensure completeness, one individual completes the field forms and another checks to verify that all pertinent information is included. Activities described in this section are summarized in Figure 8-1. Activities associated with collecting replicate benthos samples (if required) are described in the regional activities plan.

## 8.1 SITE SELECTION AND SAMPLE COLLECTION

The process for locating the site and collecting benthic samples is described in the following section and is summarized in Table 8-1. The actual site location for benthic sampling is determined from the vertical distribution (depth profile) of temperature and dissolved oxygen (DO). In thermally stratified lakes, samples are taken in well-oxygenated areas (where DO is greater than 5 mg/L and at sites where the upper limits of the metalimnion meet the lake bottom) or within the metalimnion where dissolved oxygen concentration still exceeds 5 mg/L. The dissolved oxygen value of 5 mg/L is operationally defined and is intended to ensure that samples are collected from the sublittoral zone rather than from locations that might be more characteristic of the profundal zone. The depth of the top of the metalimnion will generally vary between 3 and 5 m depending upon such factors as time of year, lake depth, lake shape, and exposure to

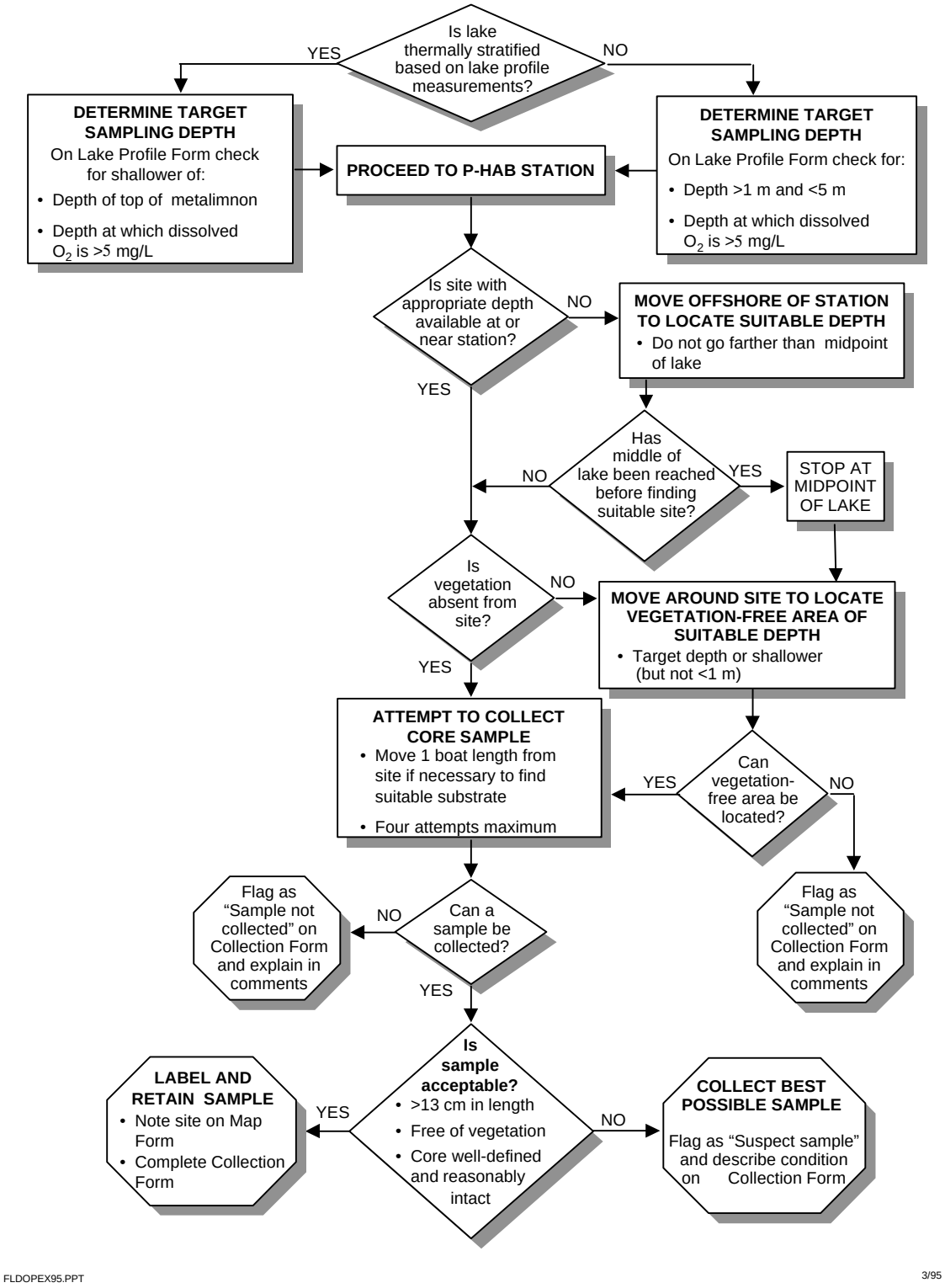



**Figure 8-1. Benthic invertebrate sampling activities summary.**

**TABLE 8-1. COLLECTION PROTOCOL FOR BENTHIC SAMPLING**

1. Note the target depth at which the top of the metalimnion was observed during lake profile activities and record on Side 1 of the Benthos Sample Location and Collection Form (benthos collection form).

2. Proceed to the physical habitat station, record the start time on Side 2, and find a suitable location (well-oxygenated [DO > 5.0 mg/L]) at or near a physical habitat observation point:

   a. where the upper limits of the metalimnion meet the lake bottom, or
   b. near the physical habitat station in a shallow area of the lake where the depth is greater than 1 m and there are very few or no weeds. Try to ensure that 10 cores are obtained from widely separated points if the physical habitat sites are located over sediment that is hard to sample.

3. Note this location on the map on Side 1 of the benthos collection form and record any pertinent comments.

4. Collect a core sample.

5. Determine if the core is acceptable. Discard and resample the core if:

   a. the sampler malfunctions and the core is <13 cm long,
   b. the core contains a large amount of aquatic vegetation, or
   c. the core is disturbed (the sediments are stirred up).

6. Obtain a 13-cm long sample from the top of the core using the extruder and sectioning apparatus. Slide the sample into a 1-gal heavy-duty self-sealing plastic freezer bag. Seal the bag and write the station ID and the core length on the bag with a permanent marker. Rinse the remaining sample from the sectioning apparatus using a wash bottle containing lake water.

7. Remove the ribbon marking the physical habitat station and move to the next station. Record the depth collected and substrate type on the benthos collection form.

wind. Some shallow lakes may be completely mixed from top to bottom. In shallow basins of stratified lakes or in unstratified lakes, collect the samples in weedless areas at or near the physical habitat station where the depth is greater than 1 m.

To locate the upper depth of the metalimnion (see Figure 5-1), refer to the Lake Profile Form (Figure 8-2) which was filled out the previous day. The top of the metalimnion should be noted on this form (if not, refer to Section 5 for directions on determining this depth). On the map portion of the benthos collection form (Benthos Sample Location and Collection Form, Side 1, Figure 8-3), record the depth of the top of the metalimnion (or the deepest depth where DO is greater than 5.0 mg/L, whichever is shallower). Use this depth as a target sampling depth at each of the 10 physical habitat stations. Follow the process identified in Figure 8-4 for locating a suitable sampling site at each station. Use the sonar to locate a suitable sampling site at or near a physical habitat station. Mark the location of each sampling site on the sketch map on Side 1 of the benthos collection form (Figure 8-3). Identify the site on the map with a circled letter corresponding to the nearest physical habitat station.

After the sampling site has been identified, anchor the boat. Wear surgical gloves during the collection process. At the first station, record the "START" time on Side 2 of the benthos collection form (Figure 8-5). Insert the core tube into the sampling apparatus and tighten the hose clamp screws to secure the core tube. Attach the messenger to the sampler line and slowly lower the sampler to the lake bottom so that it contacts the sediments in a vertical position with as little disturbance to the bottom as possible. Maintain some tension on the line to keep the sampler vertical while deploying the messenger. Activate the sampler by sending the messenger down the line, tripping the closing mechanism. Slowly retrieve the sampler to just below the surface. While the sampler tube is still submerged in water, insert a rubber stopper into the bottom of the core tube. Retrieve the sampler into the boat and place it in a vertical position in a large tub to prevent contamination of the boat with sediment. Remove the Plexiglas core tube from the sampler. Have one person hold the sampler in a vertical position while another person dismantles the unit. Examine the sediment sample within the core tube. Retain only undisturbed, intact samples that are essentially free of aquatic plants and debris. An acceptable sample is one that contains fine sediments that fill the core to a depth of at least 13 cm and has an undisturbed surface layer. Unacceptable samples (which are discarded) include cores less than 13 cm in length due to improper functioning of the sampler or due to unsuitable substrate material. It may not be possible to obtain "acceptable" samples at all sites. In such cases, retain the best sample obtainable, record a "U" (suspect sample) on Side 2 of the benthos collection form (Figure 8-5), and explain the flag in the comments section.

## LAKE PROFILE FORM

LAKE NAME: L. WOEBEGUS DATE OF PROFILE: 7/4/94 VISIT #: (1) 2

LAKE ID: NY000L SITE ID (circle): (INDEX X) OTHER: _____

TEAM ID (circle): 1 (2) 3 4 5 6 7 8 9 10 OTHER: ___

PRECIPITATION (circle): (NONE) LIGHT HEAVY

SURFACE CONDITIONS (circle): FLAT (RIPPLES) CHOPPY WHITECAPS

ODOR? X NO ☐ YES Description:

SCUM? X NO ☐ YES Description:

INDEX SITE DEPTH: 18.9 M CHECK (✓) IF SONAR NOT USED: ☐

FLAG: COMMENTS:

### DISSOLVED OXYGEN & TEMPERATURE PROFILE

(Depth of Measurement[a] [m]: Surface, 1.5, 2, 3, 4, 5, 6, 7, 8, 9, 10, 11, 12, 13, 14, 15, 20, 25, 30, 35, 40, 45, and 50 m), Also include readings at 1 m above bottom.

| DEPTH (m) XX.X | $O_2$ (mg/L) XX.X | TEMP. (°C) XX.X | FLAG | META-LIMNION (T, B)[b] | DEPTH (m) XX.X | $O_2$ (mg/L) XX.X | TEMP. (°C) XX.X | FLAG | META-LIMNION (T,B)[b] |
|---|---|---|---|---|---|---|---|---|---|
| SURFACE | 8.8 | 21.1 | | | 11.0 | 4.2 | 12.1 | | |
| 1.5 | 8.8 | 21.0 | | | 12.0 | 3.8 | 12.0 | | |
| 2.0 | 8.8 | 21.0 | | | 13.0 | 3.7 | 11.9 | | |
| 3.0 | 8.8 | 21.0 | | | 14.0 | 3.4 | 11.8 | | |
| 4.0 | 8.8 | 21.0 | | T | 15.0 | 3.4 | 11.8 | | |
| 5.0 | 7.0 | 18.8 | | | 17.9 | 1.9 | 11.3 | | |
| 6.0 | 5.7 | 15.6 | | | 16.0 | 3.0 | 11.2 | | |
| 7.0 | 4.4 | 14.2 | | | 17.0 | 2.1 | 11.3 | | |
| 8.0 | 4.9 | 13.2 | | B | | | | | |
| 9.0 | 4.3 | 12.9 | | | | | | | |
| 10.0 | 4.4 | 12.5 | | | | | | | |
| SURFACE (Dup.) | 8.8 | 21.1 | | | | | | | |

IS THE DUPLICATE $O_2$ READING WITHIN ±0.5 MG/L OF THE INITIAL SURFACE READING? X YES ☐ NO

CHECK HERE IF ADDITIONAL PROFILE MEASUREMENTS ARE RECORDED ON THE REVERSE SIDE: _____

[a] If the site depth is ≤ 3 m, take readings at the surface, every 0.5 m, and 1 m above the bottom.

[b] METALIMNION = The region of the profile where the temperature changes at a rate of 1 °C or greater per meter of depth. Indicate the depth of the top of the metalimnion with a "T," and the bottom of the metalimnion (when the rate of change becomes less than 1 °C per meter) with a "B." After the metalimnion is encountered, take readings every 1 m until bottom of the metalimnion is reached. Record the depth of the top of the metalimnion on the Benthos Sample Location and Collection Form.

FLAG CODES: K = NO MEASUREMENT OR OBSERVATION MADE; U = SUSPECT MEASUREMENT OR OBSERVATION; Q = UNACCEPTABLE QC CHECK ASSOCIATED WITH MEASUREMENT; F1, F2, ETC. = MISCELLANEOUS FLAGS ASSIGNED BY EACH FIELD CREW. EXPLAIN ALL FLAGS IN COMMENTS SECTION ON BACK OF FORM.

REVIEWED BY (INITIAL):_____



Figure 8-2. Lake Profile Form..

## BENTHOS SAMPLE LOCATION AND COLLECTION FORM-LAKES

LAKE NAME: L. WOEBEUS DATE OF COLLECTION: 7/4/94 VISIT #: (1) 2

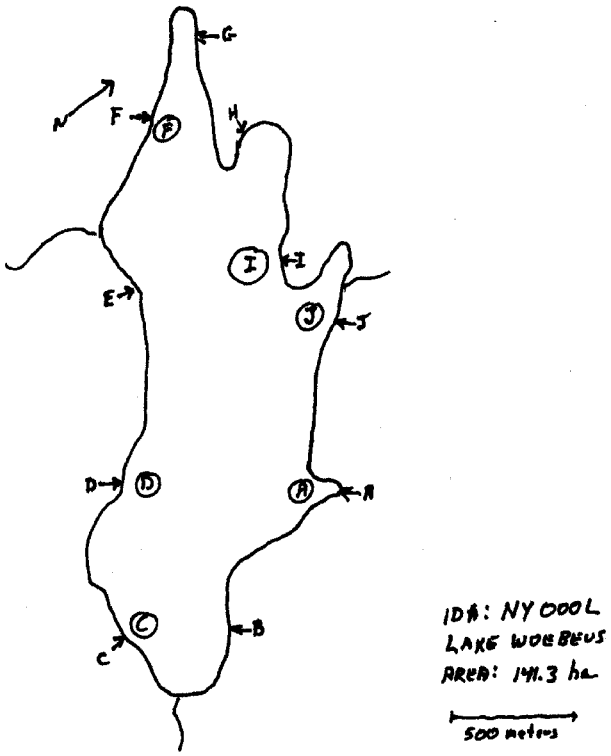
LAKE ID: NY 000L TEAM ID (circle): 1 (2) 3 4 5 6 7 8 9 10 OTHER: ___

### OUTLINE MAP OF LAKE (WITH PHYSICAL HABITAT STATIONS IDENTIFIED)

INDICATE LOCATIONS WHERE BENTHIC CORE SAMPLES ARE COLLECTED WITH THE LETTER OF THE NEAREST PHYSICAL HABITAT SITE (A - J).

ARROW INDICATES NORTH.

RECORD THE **SHALLOWER** OF THE FOLLOWING DEPTHS (FROM LAKE PROFILE FORM)
A) THE DEPTH OF TOP OF METALIMNION OR
B) THE DEEPEST DEPTH AT WHICH DISSOLVED OXYGEN ≥ 5 MG/L. TARGET DEPTH 4 M




COMMENTS:

REVIEWED BY (INITIAL):

Rev. 3/95 FLDFRMS.95

Benthos Sample Location and Collection Form - Lakes - 1

**Figure 8-3. Benthos Sample Location and Collection Form, Side 1.**

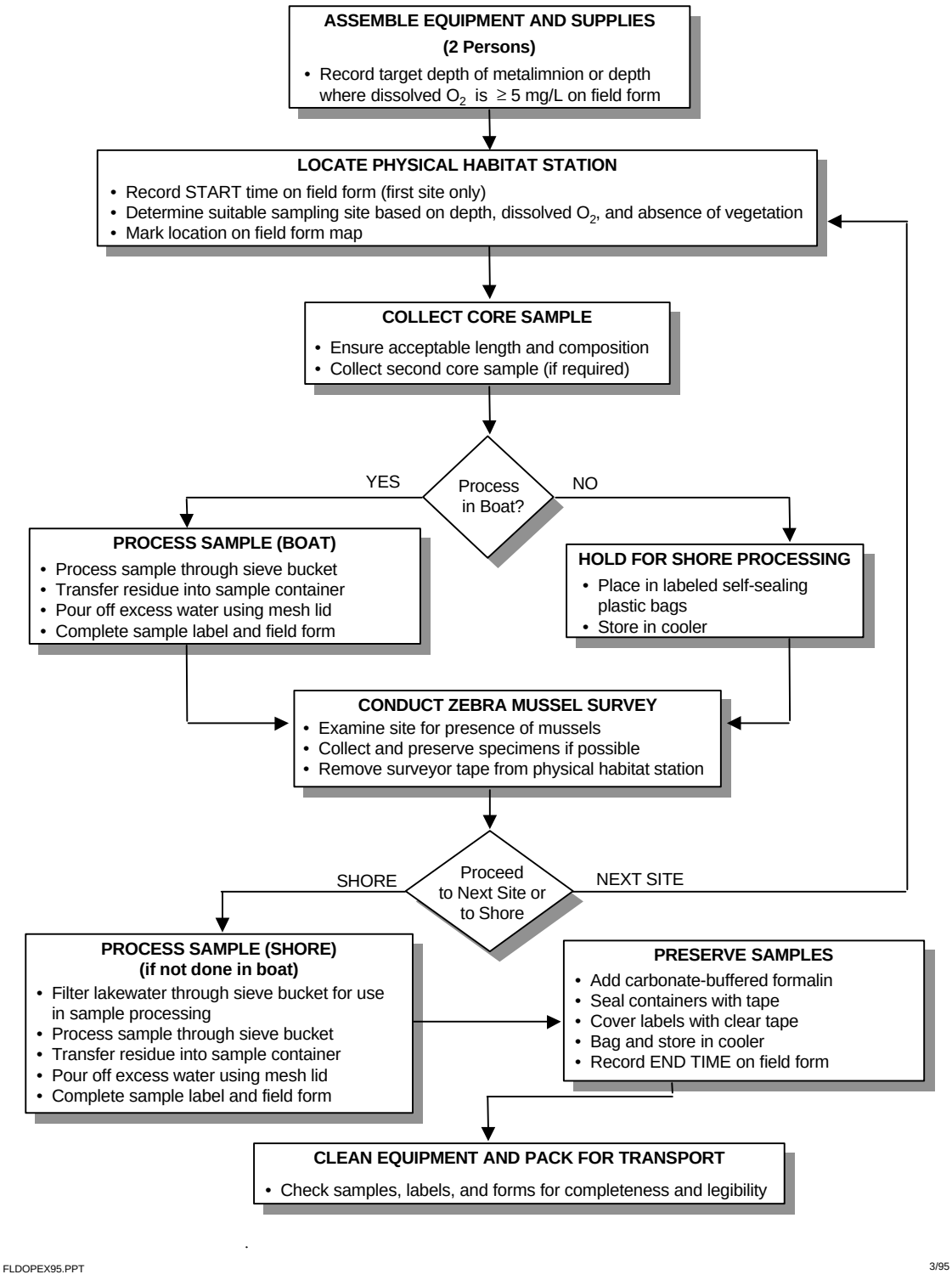



**Figure 8-4. Process for selecting benthic sample sites.**

**BENTHOS SAMPLE LOCATION AND COLLECTION FORM (CONTINUED)** VISIT # : (1) 2

LAKE ID: NY0000L DATE OF COLLECTION: 7/4/94

RECORD SAMPLING START TIME: 14:30 RECORD PROCESSING COMPLETION TIME: 19:00

| SAMPLE ID # (Barcode) | STATION ID | DEPTH COLLECTED | SUBSTRATE TYPE[a] | FLAG[b] | COMMENTS |
|---|---|---|---|---|---|
| 302001 | A | 5.8 M | C | | |
| | B | M | | K | NO SAMPLE COLLECTED - ROCKY BOTTOM |
| 302002 | C | 6.2 M | O | | 50:50 SAND/CLAY |
| 302003 | D | 5.5 M | S | | |
| | E | M | | K | NO SAMPLE COLLECTED - TOO WEEDY |
| 302004 | F | 3.5 M | C | U | BEST SAMPLE OBTAINED WAS IN VEG. |
| | G | M | | K | BEDROCK BOTTOM |
| | H | M | | K | BEDROCK BOTTOM |
| 302005 | I | 5.5 M | G | U | CORE ONLY 5 cm LONG |
| 302006 | J | 6.0 M | O | | SAND, CLAY, WOODY DEBRIS |
| | | M | | | |
| | | M | | | |
| | | M | | | |
| | | M | | | |
| | | M | | | |

[a] SUBSTRATE TYPE CODES: G = GRAVEL; S = SAND; C = SILT CLAY, OR MUCK; W = WOODY DEBRIS; O = OTHER (DESCRIBE IN COMMENTS)

[b] FLAG CODES: K = NO SAMPLE COLLECTED; U = SUSPECT SAMPLE; F1. F2, ETC.= MISC. FLAGS ASSIGNED BY EACH FIELD CREW. EXPLAIN ALL FLAGS IN COMMENTS SECTION.

**ZEBRA MUSSEL OBSERVATION AND COLLECTION**

| STATION | OBSERVED (Y/N) | COLLECTED (Y/N) | COMMENTS |
|---|---|---|---|
| A | N | N | |
| B | N | N | |
| C | N | N | |
| D | N | N | |
| E | N | N | |
| F | N | N | |
| G | N | N | |
| H | N | N | |
| I | N | N | |
| J | N | N | |
| LAUNCH | Y | Y | PRESENT IN LOW NUMBERS ATTACHED TO SMALL COBBLE. |
| OTHER | N | N | |

REVIEWED BY (INITIAL): JA



**Figure 8-5. Benthos Sample Location and Collection Form, Side 2.**

Insert the core extruder through the lower end of the core tube and extrude the sample by forcing the rubber stopper down against the extruder. Water overlaying the core does not need to be removed by a siphon as described for sediment diatom collection in Section 7. Place screen top lid over the core tube and extrude the water overlaying the core. Remove the lid and place the stage and sectioning tube on the core tube. Slowly extrude the core until the top of the core is level with the top of the sectioning tube (~13 cm; see Figure 7-3). Carefully slide the sectioning tube containing the top 13 cm of core into an appropriately labeled 1-gal heavy-duty, self-sealing plastic freezer bag. Use a wash bottle containing lake water to rinse the sample from the storage and sectioning tube into the bag. Store bags in a cooler until processing.

Discard the remainder of the core by extruding it into the lake. Thoroughly rinse the extruding apparatus, core tube, and sectioning apparatus with lake water. Record the dominant substrate type (gravel; sand; silt, clay or muck; woody debris; or other, to be described in the comments section) of the core on Side 2 of the benthos collection form (Figure 8-5). Also record the actual depth from which the sample was collected. If no sample can be collected from a site, enter a "K" flag (for missing sample) on the benthos collection form, and explain in the comments section why no sample was collected. Remove the ribbon marking the physical habitat station and move to the next station.

## 8.2 SAMPLE PROCESSING

Sample processing activities are summarized in Table 8-2. At the option of the field crew, the sample may be processed at the collection site while the boat is still anchored in position or it may be taken to shore for further processing. An advantage of processing the sample at the collection site is that there is no need to filter rinse water as the likelihood of introducing benthic organisms into the sample from open lake water is negligible. Water obtained near shore may contain benthic animals dislodged from weeds or shallow, disturbed substrata and must be filtered through the number 60-mesh screen bottom bucket prior to rinsing the sample. Thoroughly rinse the screen bottom bucket before processing samples.

Transfer the 13-cm portion of core retained for processing from the 1-gal self-sealing bag to a plastic bucket with a number 60-mesh screen bottom. Rinse all material adhering to the sides and bottom of the 1-gal self-sealing bag into the screen bottom bucket with lake water (or filtered lake water). Tap the screen bottom bucket on the surface of the lake to force water through the screen bottom. Continue this process until the fine sediments are rinsed through the screen. Samples are adequately screened when water draining through the screen becomes clear and no "sediment cloud" is visible around the bottom of the bucket. When agitating the bucket in the lake, it is very important that the bucket not be submersed to prevent losing some organisms in the sample over the top of the bucket. If the bucket is submersed, discard

**TABLE 8-2. PROCESSING BENTHIC SAMPLE**

1. For each station sample, complete a sample label with lake ID, date, and station ID and attach it to a 500-mL bottle. Cover the label completely with clear tape. Copy the sample bar code number from the label onto the benthos collection form. Also record the "depth collected" and the "substrate type" on the form. For stations where no sample is collected, enter a K in the flag field and explain it in the comments section.

2. Processing - Do in boat or on shore. **If performed on shore, all lake water used must first be filtered through No. 60 mesh screen bottom bucket.** Transfer sample from collection bucket into 60-mesh sieve bucket. Rinse 1-gal self-sealing bag into sieve bucket with lake water.

3. Tap the screen bottom bucket repeatedly on the lake water surface to force water through the screen bottom until the water draining through the screen is clear. If the sieve bucket becomes totally submerged, the sample is no longer acceptable because organisms may have been lost.

4. Place the sieve bucket containing the sample over a bucket or pan. Concentrate residue in the sieve bucket in one area. Transfer the residue in the sieve bucket into a 500-mL bottle by hand.

5. Rinse the remaining residue into the container using a plastic funnel, using small amounts of lake water.

6. Attach a lid with 60-mesh screening to the container and pour out the excess water. Rinse the residue on the lid back into the container with water from the rinse bottle. Add the filtered water to bring the total volume (residue plus water) to about 400 mL. Complete the information on the benthos collection form before leaving the site.

7. On shore, fill a plastic syringe with 50 mL of 100 percent carbonate buffered (pH 10) formalin solution. Be sure to use formalin of pH 10. The formalin used for the fish samples is pH 7.6 to 7.8 and will dissolve the chitinous exoskeletons and mollusk shells in the sample. Add the pH 10 formalin to the sample bottle. Cap the container tightly. Seal the container by taping the cap clockwise* with plastic tape.

8. Place all of the sample bottles into a 30-gallon clear or white plastic bag and seal with tape or wire ties. Write the Lake ID number on the bag with a permanent marker and place in a cooler for transport.

* If the sample containers have only 1 to 2 threads on the neck, applying the tape in a counterclockwise direction may be better protection against leakage. This should be tested during training to determine the best procedure for taping containers.

the sample and collect a new sample. Also, do **not** mix the sample by hand or with a spatula to speed the sieving process. This practice destroys the small and fragile organisms.

Complete a sample label with the Lake ID, date, and site ID and circle the type of sample (CORE). Attach the label to a 500-mL bottle. Check the labels to ensure that all written information is complete and legible. Place a strip of clear packing tape over the label and bar code, covering the label completely. While holding the labeled sample container over another bucket or tub, transfer the residue from the screen bottom bucket, catching any residue that falls outside the sample container in the second container. The objective is to capture all the residue in the sample container while introducing as little water as possible. Tilt the screen bottom bucket during the final stages of sieving to concentrate the residue into a small area on the bottom of the bucket. Transfer the bulk of this material by hand into the sample container. Rinse the remaining residue in the bucket into the sample container through a plastic funnel using a lake water rinse (filtered through number 60 mesh) contained in a 1,000-mL plastic rinse bottle fitted with a rinse spout. Fit a screen top lid (number 60 mesh) onto the sample container and drain off the excess water in the sample container. Gently rinse the residue retained on the screen top lid back into the sample container with small amounts of lake water in the rinse bottle. Add filtered lake water from the rinse bottle to bring the volume in the sample container to 400 mL. Use a marked bottle as a guide. Record the bar code printed on the label on Side 2 of the benthos collection form (Figure 8-5).

Record a "U" flag (for suspect sample) and provide comments on the benthos collection form if:

a. there are any problems in collecting the sample,

b. conditions occur that may affect sample integrity, or

c. a nonstandard procedure was used to collect a sample.

If there are other observations of note about a sample that do not render it suspect, use a miscellaneous flag (F*n*).

After all 10 sites are sampled, return to shore and add 40 to 50 mL of carbonate-buffered formalin to each container to prepare a 10-percent formalin solution. Cap the containers tightly and wrap electrical tape clockwise around each cap to seal it for transport. Invert and shake bottles to mix the formalin throughout the sample. Record the time sample processing ended on Side 2 of the benthos collection form (Figure 8-5).

## 8.3 QUALITATIVE ZEBRA MUSSEL SURVEY

In the late 1980s at least one species of exotic freshwater mussels (Unionidae: *Dreissena sp.*, known as zebra mussels) became established in the Great Lakes. Since 1990 they have been spreading into other inland surface waters (Ludyanskiy et al. 1993). EMAP is in a position to be able to monitor the rate and extent of zebra mussel invasion into inland lakes (Whittier at al., in press). At this time, the goal is only to detect and document their presence in a lake, not to do quantitative in-lake assessments of abundance. Currently, zebra mussels are not widespread in inland lakes, having been found in a few large lakes and in large rivers. In addition, the zebra mussel appears to require moderately hard water to reproduce successfully.

The general procedure is to actively look for zebra mussels at each of the 10 physical habitat stations, the benthos sampling sites, and at the launch site. Observations of mussels at any other location should also be recorded. If any mussels are observed, example specimens should be collected if possible, and preserved for species verification. Samples need to be collected from only 2 or 3 locations if they are widespread in a lake. Observations and collections may be made during the physical habitat assessment (Section 5) or in conjunction with quantitative benthos sampling. Observations and collection at any other time are also valid. Record any data related to zebra mussels on Side 2 of the benthos collection form (Figure 8-5).

### 8.3.1 Species Characteristics and Probable Habitat

The zebra mussel (Figure 8-6) is a small bivalve (the adults are generally 25 to 30 mm in length) that normally attaches firmly and permanently to solid substrates, in the manner of saltwater mussels. However, there are new reports (only in the Great Lakes so far) of a second zebra mussel species ("quagga" mussel) that will colonize soft substrates. Once established, they usually form large clusters (i.e., you are unlikely to find one lone mussel) on rocks, buoys, pier pilings, woody debris, trash, native freshwater mussels, and each other. In lakes they tend not to survive in locations subject to ice scour or heavy wave action, on soft substrates like sand or mud, or in areas of bright light. They tend to become abundant in water greater than 1 m deep.

### 8.3.2 Collection and Data Recording

Table 8-3 gives the procedures for the zebra mussel survey. At each physical habitat station, each benthic sampling site, and at the launch site of each lake, make a brief visual search of hard substrates for zebra mussels. Conduct observations in water greater than 1 m deep if possible.

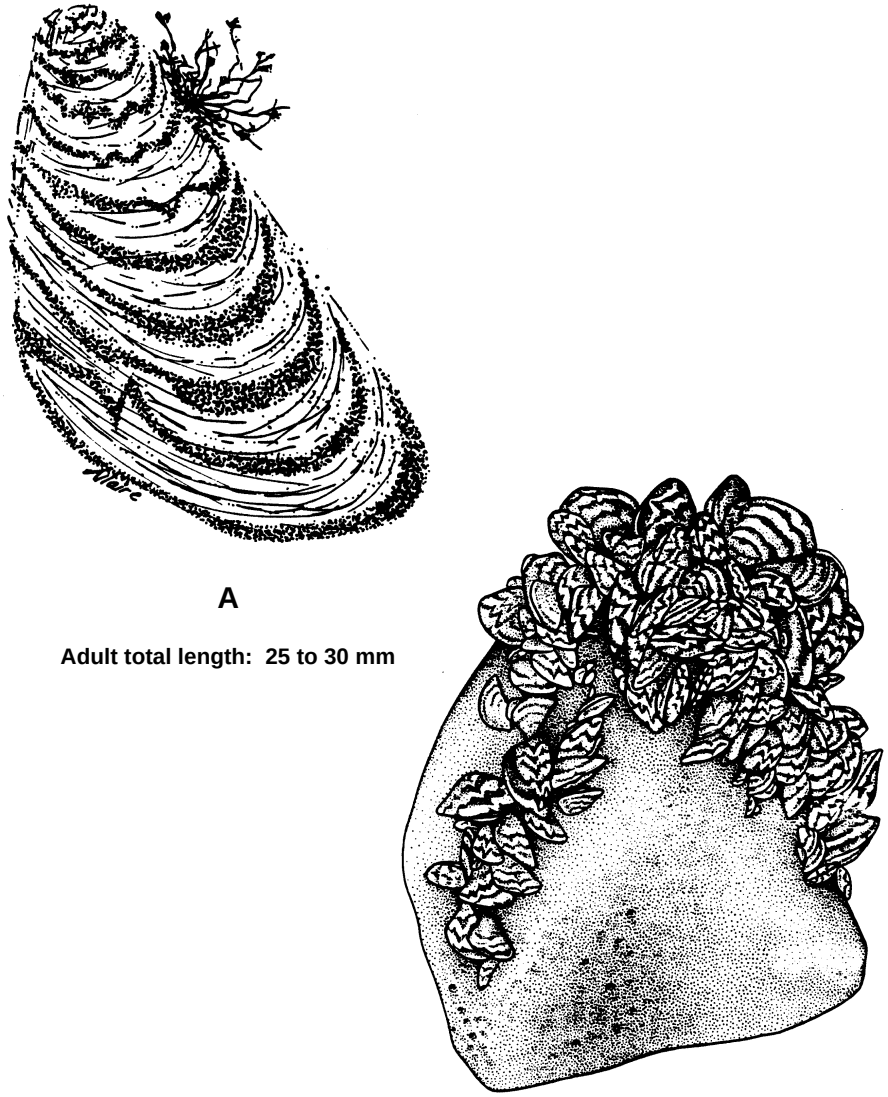



**Figure 8-6. Zebra mussel (*Dreissena polymorpha*). A. Single zebra mussel showing byssal threads (Credit: Carol Allaire); B. Cluster of zebra mussels on a rock (Credit: Margaret Van Bolt). (Illustrations provided by the Michigan Sea Grant Program.)**

**TABLE 8-3. QUALITATIVE ZEBRA MUSSEL SURVEY**

1. At each physical habitat station and benthos sampling site, search for likely locations for zebra mussels based on the following guidelines:

   a. Depths >1 m (not subject to ice scour or heavy wave action).
   b. Harder bottom substrates (although some forms may colonize soft substrates).
   c. Possible attachment sites (e.g., rocks, buoys, peer and dock pilings, woody debris, trash, and native freshwater mussels).

2. Use the viewing box to aid in underwater observations of substrate and potential attachment sites.

3. If no mussels are observed, enter an "N" in the "OBSERVED" box on Side 2 of the benthos collection form. Diagnostic features of zebra mussels include:

   - thin shells,
   - adults approximately 25 to 30 mm (1 inch) long,
   - dark color, with characteristic "zebra" striping, and
   - clusters attached on solid substrates.

4. If mussels are observed (even if they are not believed to be zebra mussels), enter a "Y" in the "OBSERVED" box on Side 2 of the benthos collection form. Make a reasonable effort to collect a sample. Use a knife to slice the attachment threads and gently pull or pry one or two individuals from the substrate. Take care to avoid breaking the knife blade. If possible, collect a cluster of mussels that are attached to a small object (e.g., a rock or shell).

   If mussels are observed but are not collectable at the physical habitat stations, benthos sampling sites, or launch site, enter an "N" in the "COLLECTED" box. Attempt to collect them from another location in the lake. Record the locations as comments for the nearest physical habitat station. If this is possible, enter a "Y" in the "COLLECTED" box.

   If mussels are widespread in a lake, collect specimens from only two or three sites.

5. Place specimens in a self-sealing plastic bag with some lake water until they can be transported to the launch site.

6. Preserve specimens in 10 percent carbonate-buffered formalin, using an extra benthic sample container. Prepare a label from a blank sheet of paper (100 percent rag content or water resistant, if possible) with the following information:

   - Lake ID
   - Visit
   - Nearest physical habitat station
   - Identify as "Zebra mussel sample"

   Attach the label to the container with clear tape that covers the label completely. Seal the container and prepare it for transport using the same techniques as those used for benthic samples.

7. Ship zebra mussel samples with the fish voucher samples, unless otherwise directed by the Communications Center.

If you observe mussels, make a reasonable effort to collect a sample. Native North American freshwater lake bivalves are usually found on soft substrates and are mobile. The regional museums are interested in freshwater mollusks in general, so collect examples of other bivalves and gastropods, if possible. Diving or swimming is not required to obtain such a sample. A better alternative is to collect a cluster of mussels attached to a small object (e.g., a small rock or another mollusk shell). If zebra mussels are widespread, collect samples from only two or three locations. If mussels are present in the lake but are not collectable at any of the designated sites, try to get a sample from some other location. The object is to detect and document their presence in a lake. Place the collected mussels in a self-sealing plastic bag for later preservation in formalin. Preserve and label mussel samples along with other nonfish specimens collected (one or two containers per lake); see the regional activities plan for any additional guidance. Preserve mollusks in the carbonate-buffered formalin used for benthic invertebrates (the alkaline pH will minimize breakdown of the shell and associated diagnostic features).

Use the comments section of the benthos collection form to explain why mussels were seen but not collected, as well as to add comments on observations such as substrate or numbers of mussels.

## 8.4 EQUIPMENT AND SUPPLY LIST

A checklist of equipment and supplies required to conduct the protocols described in this section is provided in Figure 8-7. The field teams are required to use the checklist presented in this section to ensure that equipment and supplies are organized and available on the boat in order to conduct the protocols efficiently.

## 8.5 REFERENCES

Ludyanskiy, M. L., D. McDonald, and D. MacNeill. 1993. Impact of the zebra mussel, a bivalve invader. Bioscience 43:533-544.

Whittier, T. R., A. T. Herlihy, and S. M. Pierson. 1995. Regional susceptibility of Northeast lakes to zebra mussel invasion. Fisheries 20:20-27.

**EQUIPMENT AND SUPPLY CHECKLIST FOR BENTHOS SAMPLING**

| | |
|---|---|
| Completed Lake Profile Form | 1 |
| Benthic Sample Collection Form with preprinted lake outline (from dossier) | 1 |
| Field Operations Manual | 1 |
| Quick Reference Handbook | 1 |
| Sediment core tube | 1 |
| Sectioning stage | 1 |
| Sectioning tube | 1 |
| Plastic funnel | 1 |
| Sieve bucket | 1 |
| Rinse bottle, 500-mL | 1 |
| Screen top lid (No. 60 mesh) for sample containers | 1 |
| Sample containers, 500-mL (marked at 400-mL) | 10 |
| Heavy-duty self-sealing plastic bags, 1-gallon, labeled with station ID | 10 |
| Large plastic tub | 1 |
| Plastic electrical tape | 1 roll |
| Permanent markers | 2-3 |
| Garbage bags, large kitchen size (for storing sample containers) | 2 |
| Cooler | 1 |
| Benthic sample labels with bar codes | 1 sheet |
| Benthic sample labels without bar codes (for extra containers) | 1 sheet |
| Clear tape strips | 1 pkg. |
| 60-cc plastic syringe for dispensing formalin | 1 |
| Carbonate-buffered formalin solution (sodium bicarbonate) | 500 mL |
| Surgical gloves | 2 pair |
| Parts kit | 1 |

**Figure 8-7. Benthic invertebrate sampling checklist.**

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# ACRONYMS AND ABBREVIATIONS

| | |
|---|---|
| BPJ | Best Professional Judgment |
| DLGs | Digital Line Graphs |
| DO | dissolved oxygen |
| EMAP | Environmental Monitoring and Assessment Program |
| EPA | U.S. Environmental Protection Agency |
| GPS | Global Positioning System |
| GQ | geometric quality |
| ID | identification |
| 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 |
| SQ | signal quality |
| STARS | Sample Tracking and Reporting System |
| T | Top |
| TIME | Temporally Integrated Monitoring of Ecosystems |
| USGS | United States Geological Survey |
| YOY | young of year |
| YSI | Yellow Springs Instrument system |

## Measurement Units

| | |
|---|---|
| ha | hectare |
| m | meter |
| ppm | parts per million |