Environmental Monitoring and Assessment Program-Surface Waters:

Field Operations and Methods for Measuring the Ecological Condition of Non-wadeable Rivers And Streams

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Notice

This research described in this report has been funded wholly or in part by the U.S. Environmental Protection Agency. This document has been prepared at the EPA National Exposure Research Laboratory (Ecological Exposure Research Division, Cincinnati, Ohio) and the National Health and Environmental Effects Research Laboratory (Western Ecology Division, Corvallis, Oregon), under the following contracts and cooperative agreements:

Contract 68-C6-0006 to Dynamac International, Inc.
Contract 68-W5-0065 to OAO, Inc.
Cooperative Agreement CR824682 to Oregon State University

This work is in support of the Environmental Monitoring and Assessment Program (EMAP). Mention of trade names or commercial products does not constitute endorsement or recommendation for use.

The correct citation for this document is:

Lazorchak, J.M., Hill, B.H., Averill, D.K., D.V. Peck, and D.J. Klemm (editors). 2000. Environmental Monitoring and Assessment Program -Surface Waters: Field Operations and Methods for Measuring the Ecological Condition of Non-Wadeable Rivers and Streams U.S. Environmental Protection Agency, Cincinnati OH.

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Abstract

The methods and instructions for field operations presented in this manual for surveys of non-wadeable streams and rivers were developed and tested based on 55 sample sites in the Mid-Atlantic region and 53 sites in an Oregon study during two years of pilot and demonstration projects (1997 and 1998). 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 non-wadeable 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. They are intended for use in field studies sponsored by EMAP. 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, periphyton assemblages, and sediment community metabolism. 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 river 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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Acknowledgements

We greatly appreciate the peer review comments of J. David Allan, Richard W. Merritt, and Kelly Wessell from the Department of Entomology, Michigan State Univeristy, Tom Bassita from West Virginia Department of Natural Resources, and Matthew J. Kline from the University of Maryland Center for Enivronmental Science. We also appreciate a review from S.E. Gwin, Dynamac Corp., Corvallis, OR. The efforts and dedication of the following field personnel from 1997 and 1998 in implementing these protocols and providing feedback for clarification and improvement are also recognized: S. Intelmann, A. Cooley, C. Johnson, M. McCrae, J. Shaich, S. Corbett, A. Uber, and R. Curl. M. Hails-Avery (National Asian Pacific Center on Aging, Senior Environmental Employment Program, Corvallis, OR) and F. Beck (Dynamac Corp., Corvallis, OR) assisted with preparing many of the

Acronyms, Abbreviations, and Measurement Units

figures. G. Mosher (OAO Inc., Corvallis, OR) prepared the field data forms.

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

EPA	U.S. Environmental Protection Agency
ERB	Ecosystems Research Branch

Acronyms, Abbreviations, and Measurement Units (continued)

Acronyms and Abbreviations

GPS	Global Positioning System
ID	identification
LWD	Large Woody Debris
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
PFD	Personal Flotation Device
P-Hab	physical habitat
PVC	polyvinyl chloride
QA	quality assurance
QC	quality control
R-EMAP	Regional Environmental Monitoring and Assessment Program
SL	Standard length
SOP	Standard Operating Procedure
TL	Total length
USGS	United States Geological Survey
WED	Western Ecology Division

YOY	young of year
YSI	Yellow Springs Instrument system

Acronyms, Abbreviations, and Measurement Units (continued)

Measurement Units

amps	amperes
cm	centimeter
gal	gallon
ha	hectare
Hz	Hertz
in	inches
L	liter
m	meter
m2	square meters
mg/L	milligram per liter
mm	millimeter
m	micrometer
S/cm	microsiemens per centimeter
msec	millisecond
ppm	parts per million
psi	pounds per square inch
V	volts
VA	volt-ampere

Section 1
Introduction

James M. Lazorchak[1], Alan T. Herlihy[2], and Daniel K. Averill[3]

This manual contains procedures for collecting samples and measurement data from various biotic and abiotic components of non-wadeable streams and rivers in the Mid Atlantic and Pacific Northwest. These procedures were developed and used between 1997 and 1998 in research studies of the U.S. Environmental Protection Agency's (EPA) Environmental Monitoring and Assessment Program (EMAP). 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 various research studies; and (2) provide these procedures for use by other groups implementing river monitoring programs. These procedures are designed for use during a one-day visit by a crew of four or five persons to sampling sites located on larger, non-wadeable streams and rivers (generally stream order 4 or greater in the Mid Atlantic and Northwestern U.S.).

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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, rivers, 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). In 1993 - 1994 the U.S. EPA Office of Research and Development and Region 3 Office, with assistance from the U.S. Fish and Wildlife Service and States in the eastern United States (WV, NY, PA, VA and MD) conducted the first EMAP wadeable stream pilot which was called the Mid-Atlantic Highlands Assessment (MAHA). In 1997 - 1998 the pilot was expanded to additional states (DE, NJ, and NC) and ecoregions and both wadeable and non-wadeable streams were sampled. The 1997 - 1998 study was called the Mid-Atlantic Integrated Assessment (MAIA). Protocols that were used in wadeable streams in the MAHA and MAIA studies are contained in the manual "Environmental Monitoring and Assessment Program - Surface Waters. Field Operations and Methods for Measuring the Ecological Condition of Wadeable Streams. EPA/620/R-94/004F." (Lazorchak, et al., 1998). Many of the protocols used on non-wadeable streams in 1997-1998 in the eastern and western United states were adapted or modified from the 1997 manual. The specific indicators dealing with non-wadeable streams and rivers are described in more detail in the following section.

1.2 Summary of Ecological Indicators

The following sections describe the rationale for each of the ecological indicators

currently included in the non-wadeable river 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.

1.2.1 Water Chemistry

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

1.2.2 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 river channels, wetland drainage, grazing and agricultural practices, and river 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 river condition relative to various condition indicators. Important attributes of physical habitat in rivers 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 rivers and to monitor biologically relevant changes in habitat quality and intensity of disturbance. Kaufmann et al. (1999) discuss procedures for reducing EMAP field habitat measurements and observations to metrics that describe channel and riparian habitat at the reach scale.

1.2.3 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. 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 was used in the development of the periphyton indices of river condition. The framework involved the calculation of composite indices for biotic integrity, ecological sustainability, and trophic condition. The composite indices were calculated from measured or derived first-order and second-order indices. The first-order indices included 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 were calculated from periphyton characteristics, such as the autotrophic index (Weber, 1973), community similarity compared to reference sites, and autecological indices (e.g., Lowe, 1974; Lange-Bertalot, 1979; Charles, 1985; Dixit et al, 1992).

1.2.4 Sediment Community Metabolism

Ecosystems are complex, self-regulating, functional units defined by rates and processes, such as energy flow or material cycling. These processes are mediated by the trophic structure of the ecosystem, and integrate the functioning of the entire community. Energy flow and material cycling are important components of two major concepts in stream ecology: The river continuum concept and resource spiraling. Heterotrophic microorganisms (bacteria and fungi) are responsible for oxygen sags in streams and for much of the decomposition of organic matter deposited in them. Measuring the rate of oxygen consumption within the soft sediments of a river provides a functional indicator of energy flow and material transformation within the ecosystem

1.2.5 Benthic Macroinvertebrate Assemblage

Benthic macroinvertebrates inhabit the sediment or live on the bottom substrates of rivers. The macroinvertebrate assemblages in rivers 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). 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 river reach. Examples of multimetric indices based on benthic invertebrate assemblages include

Kerans and Karr (1994), 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.

1.2.6 Aquatic Vertebrate Assemblages

Aquatic vertebrate assemblages of interest to EMAP include fish and amphibians (more so in the western U.S. where fish taxa richness is less). 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 rivers 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.

1.2.7 Fish Tissue Contaminants

Indicators of fish tissue contaminants attempt to provide measures of bioaccumulation of toxic chemicals in fish. When coupled with study designs such as those being developed by EMAP, these indicators can be used to estimate regional risks of consumption to predators of fish (either wildlife or human), and to track how this risk changes with time in a region. 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 river 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 river (as opposed to a focus on human health concerns). Whole fish are a better indicator of risk to piscivorous wildlife than fillets. We also should be able to address potential 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., sculpins and small minnows). The second sample is prepared using a species whose adults are of larger size (e.g., squawfish, trout, suckers, and sunfish). 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 river 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. Small fish may be a more appropriate indicator for assessing ecological risk, as they might be expected to be prey for a larger number of fish-eating animals (the majority of which will be piscivorous birds and small mammals). 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 river fish is not known, thus the reason for preparing two samples in this study.

1.3 Objectives and Scope of the Field Operations and Methods 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 and Lazorchak et al. 1998); 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 river site visit. Section 4 presents the procedures for verifying the site location and defining a reach of the river where subsequent sampling and data collection activities are conducted. Sections 5 through 12 describe the procedures for collecting samples and field measurement data for various condition and stressor indicators. Specific procedures associated with each indicator are presented in stand alone tables that can be copied, laminated, and taken into the field for quick reference. Section 13 describes the final activities that are conducted before leaving a river site. Appendix A contains a list of all equipment and supplies required by a crew to complete all field activities at a river. Appendix B presents a set of brief

summaries of field procedures and activities that can be laminated, collated into a 3-ring binder, and taken into the field along with the procedure tables. This waterproof handbook can serve as the primary field reference for field teams after they complete an intensive training program. Appendix C contains a list of vertebrate species names and corresponding species codes developed for use in the eastern U.S. and Oregon studies.

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.

1.4 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.

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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Section 2
Overview of Field Operations

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This section presents a general overview of the activities that an EMAP field team conducted during a typical one-day sampling visit to a non-wadeable river site. General guidelines for recording data and using standardized field data forms and sample labels are also presented in this section. Finally, safety and health considerations and guidelines related to field operations are provided. Depending on the survey region, river sampling distances are defined as either 40 or 100 times the wetted width in the vicinity of the point of entry (Figure 2.1). One reason for the length difference is that in the Oregon river pilot, an objective is to determine a reach length that will usually yield 95% of the vertebrate species collected in a full day of electrofishing or from a reach 100 channel widths long. Note, subsequently it was found that this reach length is 85 channel widths for Oregon rivers (Hughes et al. In Review). River reaches of 40 channel widths long were used in the Mid-Atlantic region in order to make this aspect of field methods consistent between wadeable and non-wadeable streams. In eastern rivers, one 14-16 foot john boat and one 11-13 foot inflatable raft, outfitted with a 4 horse powered motor were used. The john boats were used only for electrofishing and drift net retrieval, and were outfitted with 6.6 - 15 horsepower outboard motors. Two 12-14 foot inflatable rafts were used in a large river pilot in Oregon because river access, flow, depth, obstructions and State or Federal restrictions usually made it impractical, dangerous, or impossible to use rigid boats and outboard motors. In both studies, the larger crafts were generally used on the larger or faster rivers. Two trucks were used in each survey, with one pulling a boat trailer. These boats, motors and truck configurations are only examples of what were

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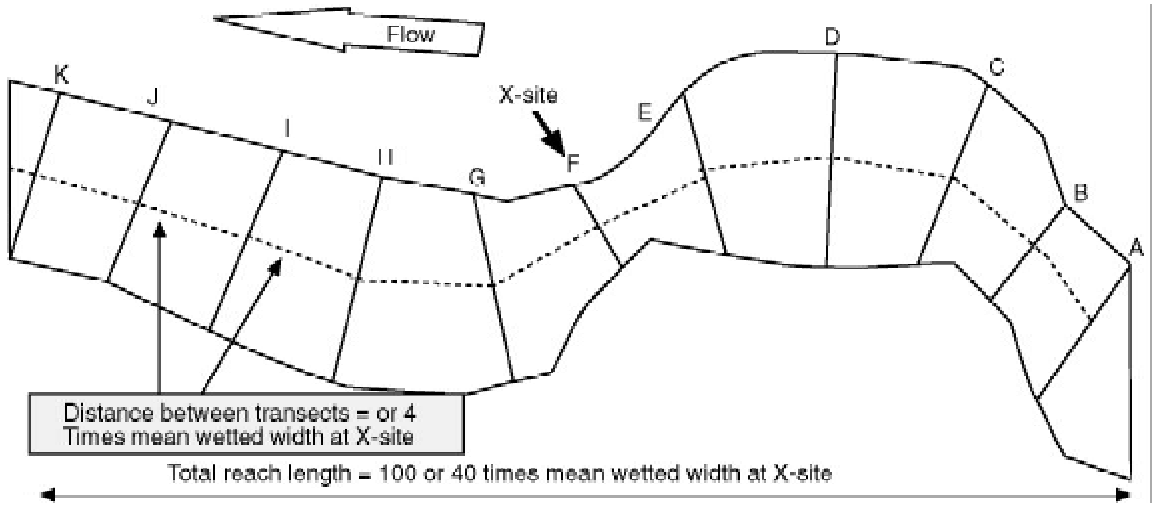

Figure 2-1. Stream reach characteristics.

used in the MAIA and Oregon Studies. Other types, numbers or sizes of boats, motors, trucks and trailers can be used as long as local boating regulations are met and the health and safety of the crews can be maintained.

2.1 Daily Operational Scenario

For western streams a 4-person field team consisted of two people in the "habitat" boat and two people in the "fish" boat. Each boat was staffed by one rower and one primary data collector. The crew in the habitat boat was primarily responsible for conducting the intensive physical habitat characterization. The crew in the fish boat is primarily responsible for collecting biological samples. Rowers assist with sampling when possible, although their primary responsibilities were rowing the rafts and river navigation. Table 2-1 presents the range of times required to conduct various field activities. Tables 2-2 a and b present the general sequence of activities conducted at each river reach for non-wadeable streams in the west and east.

In eastern non-wadeable streams a 4 or 5 person crew was used depending on whether additional research indicators were also sampled. In the eastern pilot, the electrofishing boat had 2 people while the raft crew had 2-3 people. Times presented in Table 2-1 were similar in eastern streams with the exception of additional time needed for processing additional indicator samples.

Upon arrival at a river site, the crew chief was responsible for verifying and documenting the site location, determining the length of stream reach to be sampled, and determining boat launching and retrieving locations (in the western pilot the crew chief was also establishing the required transects, Figure 2.1) (Section 4). The crew chief was also responsible for preparing samples for transport and shipment (Section 3). In addition to aquatic vertebrate sampling (Section 10), the western fish crew collected water chemistry and microbial samples (Section 5), sediment for the sediment metabolism determination (Section 8), periphyton (Section 7), macroinvertebrate samples (Sections 9),

Table 2-1. Range of Times for Field Activities.

Activity	Time Required (ranges)
Scout access locations	0 to 1.5 hours
Unload rafts and all equipment	0.5 hours
Shuttle vehicles and set up for float	0.5 to 1.5 hours
Row or float from put-in to start of reach	0 to 1 hour
Conduct field sampling activities	5 to 8 hours
Row or float from end of reach to take-out	0 to 1 hour
Load rafts and shuttle vehicles	0.5 to 1 hour
Sediment metabolism processing	0.5 hours
Sample tracking and packing	1 hour
SUMMARY	8 to 16* hours per site

*Indicates the longest total time spent on sampling activities; does not equal the sum of the greatest times spent to accomplish each task.

aquatic vertebrates (Section 10), and prepared samples for tissue contaminants (Section 11). The habitat crew conducted the intensive physical habitat characterization (Section 6), visual stream assessment (Section 12), and the habitat rower took water chemistry measurements (Section 5). The eastern three person raft crew collected all these indicators except for fish. A separate two person electroshocking crew was used for fish and also for deploying and retrieving drift nets.

2.2 Guidelines for Recording Data and Information

During the one-day visit to a river, 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.

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 river when the initial information recorded was inaccurate or illegible. Some guidelines to assist field personnel with recording information are presented in Table 2-3. Examples of completed data forms and labels are presented in the sections describing field sampling and measurement procedures for different indicators.

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

Table 2-2a. General Sequence of Activities Conducted at a Non-wadeable River Reach in the West.

NOTE: Sample odd numbered site ID's along the left shore (facing downriver); sample even numbered sites along the right shore. Large obstructions or hazards may require temporary diversion.

A. Pre-Launch

1. Pack and ship the previous day's samples (if necessary).
2. Obtain ice for the day's samples.
3. At the launch site, all crew members unload all gear and sample containers. During unloading, no time is spent loading the boats, setting up gear, etc.
4. Two crew members each shuttle a vehicle to the take-out. The two remaining crew members load each boat, label sample containers, and prepare data sheets and clipboards. The crew chief determines the transect length.
5. Set macroinvertebrate drift nets at the most ideal location (put-in or take-out ramps).
6. The shuttlers return, and all crew members complete loading preparations and launch.

B. Sampling Procedures

1. If a float is required to reach the first transect, this time should be spent completing all data form headers, labeling, situating gear on the rafts, etc. If no float, proceed to step B-2.
2. At Transect A, the habitat crew implements the habitat sampling protocol and records the GPS coordinates. The fish crew sets up the electroshocking equipment, and takes the site identification and river photos.
3. Floating between transects (e.g., Transect A downriver to Transect B, a laser rangefinder can be used to measure between transects), the habitat crew collects thalweg measurements in the deep part of the channel while the fish crew electrofishes along the designated shoreline. Both rafts cease this activity when the next transect is reached.
4. Upon reaching each transect, the habitat and fish boats pull over and "tie-off" at the designated shoreline.
   a) Habitat boat duties: The rower takes all measurements requiring instruments while the habitat lead records the measurements on field sheets. The rower then collects shore macroinvertebrate samples while the habitat lead completes the remainder of the data form and marks the transect location on the topographic map.
   b) Fish boat duties: The fish ID specialist processes the catch (identification, measurements, weights) while the rower records. Appropriate specimens are retained and stored on board. Additionally, the rower collects sediments and periphyton samples.
   c) Sample container labeling: For QA purposes, the person responsible for taking certain samples should be the one to label the sample containers for those samples.
5. Repeat steps B-3 and B-4 for each transect.
6. At the final transect (Transect K), water chemistry and microbial samples are collected.

C. Take-Out Duties

1. At the take-out location, two crew members process macroinvertebrate and periphyton composite samples, and set up for sediment metabolism processing. The two other crew members unload the rafts and load the equipment into the vehicle. All crew members assist with loading the rafts onto the trailer.
2. The crew chief reviews data forms while the other crew members tie down the rafts, clean thoroughly and stow equipment, and ready the vehicles. Data forms are reviewed for completeness, accuracy, and legibility.
3. Call in to supervisor and declare "Off River Safely."
4. Proceed to lodging facility. Take the final DO reading for sediment metabolism.

Table 2-2b. General Sequence of Activities Conducted on Eastern Non-Wadeable Streams.

NOTE: Sample odd numbered site ID's along the left shore (facing downriver); sample even numbered sites along the right shore. Large obstructions or hazards may require temporary diversion.

In the Mid-Atlantic Pilot non-wadeable sampling required two boats operating independently of each other. One boat (the fish crew) was responsible for fishing the river reach and the other boat (the bio-hab crew) collects the physical habitat, chemistry, macroinvertebrates, periphyton, and sediment (respiration and toxicity) samples, and fills in the site verification, site assessment, and TM validation forms.

A. Pre-Launch

1. Before beginning to sample, assess the river for the applicability of wadeable stream or non-wadeable stream protocols. If 50% or greater of the river reach is wadeable, than EMAP-SW Stream (Wadeable) Protocols are to be used. If not, EMAP-SW River (Non-Wadeable) Protocols will be used. For rivers non-wadeable throughout the thalweg but wadeable along the shoreline, shock thalweg and as much of the shoreline with non-wadeable protocols as possible.
2. If River protocols are used, then only one side of the river is sampled. If the site number is odd the left side (facing downstream) of the river is sample. Even numbered sites are sampled on the right side.
3. If 'X' is easily accessible, verify, using maps and GPS, its location and collect chemistry samples before traveling to the upper end of the study reach (Transect K). If 'X ' is not easily accessible, travel to the point which you believe will be the upper end of the study reach.
4. Determine average river width using one of three methods (listed in order of preference):
   a. If at 'X' measure wetted width using the rangefinder or tape;
   b. If 'X' is not accessible, estimate width based on width at nearest river crossing; or
   c. Estimate width from the 7.5 minute USGS topo map.
5. Once average width has been determined, multiply it by 40 to determine reach length to be sampled (e.g., Width=80m, reach length =3200m).
6. Once 'X' has been verified, the fish crew and one of the bio-hab crew members, travels upstream, by river or road, to the top of the reach (Transect K) and begins sampling, the bio-hab person that has gone with the fish crew drives the fish crew vehicle and trailer to the take out area and regroups with the bio-hap crew. While the fish crew and bio-hab person are launching and returning, respectively, the other two bio-hab crew members travel downstream to the bottom of the reach (Transect A) to set the drift nets.

B. Sampling Procedures

1. After setting the drift nets (drift nets should not be set by one person alone in a boat) and meeting up with the one bio-hab person that has returned the fish crew vehicle/trailer, the bio-hab crew travels to the top of the reach and begins sampling.
2. The fish crew will work downstream collecting fish through the entire reach.
3. At the end of the reach, the fish crew begins sorting fish for vouchers, tissue, and biomarkers.
4. Once the fishing crew has cleared the area, the bio-hab boat may begin their sampling of the transect. At each transect, the bio-hab boat will beach and collect physical habitat information, macroinvertebrates, periphyton, and sediment. All samples are combined with previous samples of that type (e.g., all macroinvertebrate kick samples are combined, all periphyton samples are combined and all sediment samples are combined. Sediment samples may be collected using grab samples and/ or ponar sampler.
5. Between transects the bio-hab crew will measure thalweg depths, substrate and channel form.
6. The 6th transect (Transect F) downstream should be where you verified 'X'. Chemistry samples need to be collected. TM validation form needs to be completed. Secchi depth needs to be measured and recorded in one of the comment sections on the "Field Measurements Form-Streams/Rivers".
7. Proceed downstream collecting transect, thalweg, and biology samples as in the upper half of the stream reach.

(continued)

Table 2-2b. Continued.

8. At the bottom of the reach (Transect A), the bio-hab crew sets-up metabolism, bags sediments for toxicity, collects drift nets (if deployed) and processes macroinvertebrate and periphyton samples.
9. Bio-hab crew joins fish processing or begins shuttling vehicles and carrying equipment to the vehicles.

C. TAKE-OUT DUTIES

1. At the take-out location, two crew members process macroinvertebrate and periphyton composite samples, and set up for sediment metabolism processing. The two other crew members unload the rafts and thoroughly clean and load all the equipment into the vehicle. All crew members assist with loading the rafts onto the trailer.
2. The crew chief reviews data forms while the other crew members tie down the rafts, stow equipment, and ready the vehicles. Data forms are reviewed for completeness, accuracy, and legibility.
3. Call in to supervisor and declare "Off River Safely."
4. Proceed to lodging facility. Take the final DO reading for sediment metabolism.

Table 2-3. Guidelines for Recording Field Data and Other Information.

Activity	Guidelines
	Field Measurements:
Data Recording	Record measurement values and/or observations on data forms preprinted on water-resistant paper. Record information on forms using No. 2 pencil only. Erase mistakes completely and write the correct value whenever you can. If you must line out an incorrect value, place the correct value nearby so the data entry operator can easily find it. Headers on the second pages of all forms link the data. Fill in all headers of all pages (to save time this can be filled out the night before if site is known to be accessible) or data will be lost (this is a good one to review at the end of the day). 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. Print neatly, using block capital letters in alphabetical fields. 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. Record information on each line, even if it has to be recorded repeatedly on a series of lines (e.g., fish species codes or physical habitat characteristics). Do not use "ditto marks" (") or a straight vertical line. When recording comments, print or write legibly. Make notations in comments field only. 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.
Data Qualifiers (Flags)	Use only defined flag codes and record on data form in appropriate field. K Measurement not attempted and/or not recorded.

(continued)

Table 2-3. Continued.

Activity	Guidelines
	Field Measurements:
	Q Failed quality control check; re-measurement not possible. U Suspect measurement; re-measurement not possible. Fn Miscellaneous flags (n=1, 2, etc.) assigned by a field team during a particular sampling visit (also used for qualifying samples). Explain all flags in comments section on data form.
Review of Data Forms	Field team reviews data forms for accuracy, completeness, and legibility before leaving a river. Data forms from all teams are reviewed for completeness, accuracy, and legibility before transfer to the information management staff.
	Sample Collection and Tracking
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.
Sample Collection Information	Record sample ID number from the label and associated collection information on sample collection form preprinted on water-resistant paper. Record information on field data forms using No. 2 pencil only (fine-point indelible fine-tipped markers can be used if necessary). Record collection information using correct format as provided on the collection form.
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) Fn Miscellaneous flags (n=1, 2, etc.) assigned by a field team during a particular sampling visit (also used for field measurements). Explain all flags in comments section on sample collection form.
Review of Labels and Collection Forms	The field team compares information recorded on labels and sample collection form for accuracy before leaving a stream. The field team reviews labels and collection form for accuracy, completeness, and legibility before leaving a stream. Sample collection forms are reviewed for completeness, accuracy, and legibility before transfer to the information management staff.

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 2-4. 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).

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.

Because boats are used to access sampling sites, personnel must consider and prepare for hazards associated with water conditions (e.g., obstacles, rapids), safe loading and unloading of rafts, and the operation of motor vehicles, 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). The electrofishing raft must have a fire extinguisher, and both rafts must have whistles, Personal Flotation Devices (PFD) and communication devices.

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. PFD's must always be worn while in the boat, and felted wading boots must be available for use when crew members are sampling outside of the raft.

Many hazards lie out of sight in the bottoms of rivers. 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 that is known or suspected to contain human or animal wastes or 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. [Note that nearly one-third of Oregon river reaches sampled in the summer of 1997 supported bacteria exceeding or

Table 2-4. General Health and Safety Considerations.

Training:

- First aid
- Cardiopulmonary resuscitation (CPR)
- Swiftwater rescue
- Vehicle safety (e.g., operation of 4-wheel drive vehicles)
- Boating and water safety
- 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
- Radios for boat to boat communications and cell phones for emergancies.

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

closely approaching body contact standards. Microbes can be transferred from fish or the water itself, so wash hands before eating and avoid contact between open wounds and water or fish].

Prior to a sampling trip, personnel should determine that all necessary equipment is in safe working condition. Good housekeeping practices 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. It is also recommended that at least one person on each crew should have First Aide and CPR training, especially if the crew(s) are electrofishing.

2.3.2 Safety Equipment and Facilities

Appropriate safety apparel such as PFD's, felted wading boots, lab coats, insulated gloves, safety glasses, etc. must be available and used when necessary. It is recommended that whenever two boat crews are used there are some type of communication devices on board each boat so that they can keep in contact with each other for both safety and logical reasons.

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 2-5. 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 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]).

Table 2-5. General Safety Guidelines for Field Operations

- The two rafts must be in view of each other while floating down rivers or streams.
- The river must be adequately scouted prior to sampling to avoid potential hazards (e.g., dangerous rapids, obstacles, sweepers, wood jams, portage locations).
- Exposure to river 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.

  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.

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Section 3
Base Location Activities

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Field teams conduct a number of activities at a "base" location before and after visiting each river 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 sampling 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.

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

3.1 Activities before each River Visit

Before each river visit, each field team should confirm access to the sampling 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 will be provided with dossiers containing important location and access information for each river scheduled for sampling. Before visiting a river, the crew should review the contents of the specific site dossier. The landowner(s) listed in the dossier should be contacted to confirm permission to

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[3]Dynamac International Corp., 200 SW 35th St., Covallis, OR 97333.
[4]OAO, Inc., 200 SW 35th St., Corvallis, OR 97333

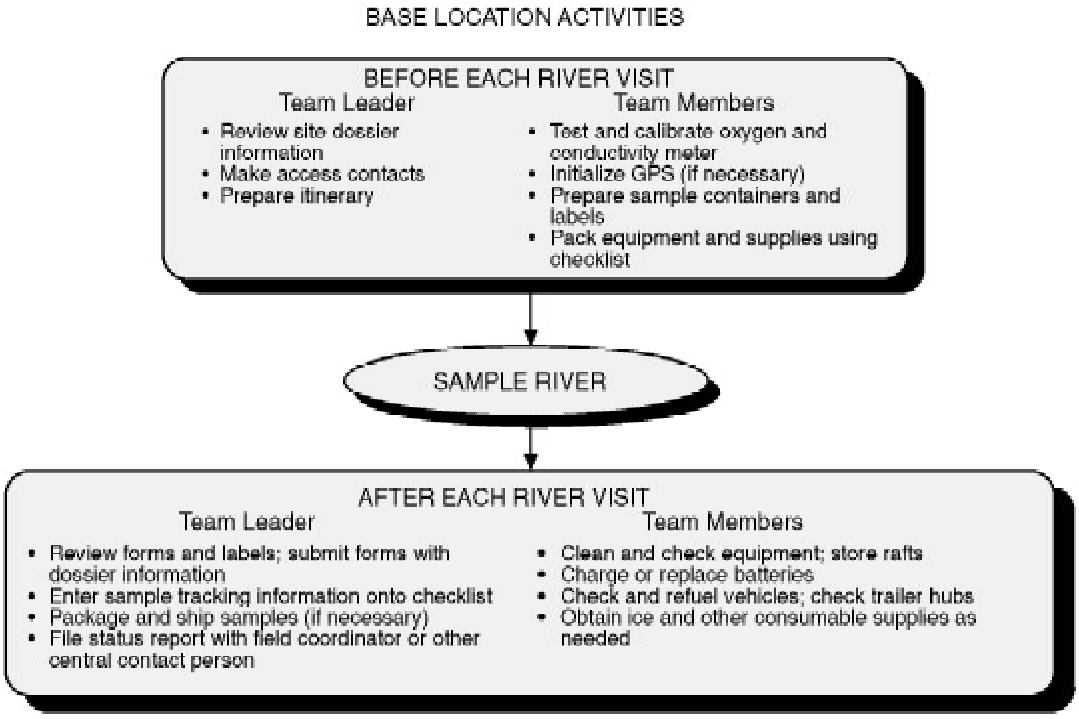

Figure 3-1. Activities conducted at base locations

sample and identify any revisions to the information contained in the dossier. Also, confirm that the proposed launch sites (e.g., public boat launches) are present and operational.

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 river dossier to ensure that it contains the appropriate maps, contact information, copies of permission letters, and access instructions. Additional activities include determining the best boat access locations, 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 river. 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 river 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 instruments prior to departure for the river site. Field instruments requiring testing and/or calibration include a global positioning system (GPS) receiver, a conductivity meter, and a dissolved oxygen meter. 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.

Before the initial use, or, in some cases, if batteries are replaced, the receiver may require inputting the coordinates of a positional reference point that is nearby (e.g., a U.S. Geological Survey benchmark identified on a topographic map). Follow the manufacturer's instructions for initializing the receiver.

3.1.3.2 Dissolved Oxygen Meter

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 at the base location before each river site visit.

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 at the terminal end of the probe. If bubbles are present, if the membrane is discolored, or if the membrane is torn, replace the screw-on membrane cap according to the manufacturer instructions.

After inspecting the meter and probe, attempt to calibrate it, 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 river site, at which time the calibration information is recorded on the field data form. If the meter cannot be successfully calibrated, replace the meter and/or probe. After the test, turn the meter off, and store the probe according to the manufacturer's instructions.

3.1.3.3 Conductivity Meters

Follow the operating manual provided with the instrument 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. The operation of the conductivity meter is checked at the base location using a standard solution of known conductivity. A daily quality control check sample (QCCS) is prepared as described in Table 3-1. The daily QCCS can be prepared as either of two dilutions of the stock standard, depending on the theoretical conductivity desired. A 1:100 dilution of the stock provides a QCCS with a conductivity of 75.3 S/cm at 25°C (Metcalf and Peck, 1993). A 1:200 dilution results in a QCCS with a con-

Table 3-1. Stock Solutions, Uses, and Instructions for Preparation.

Solution	Use	Preparation
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 pen or 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	Preservative for benthic macroinvertebrate samples.	None.

* Premade Packets can be Purchased
[a] Metcalf and Peck (1993) Premade Packets can be Purchased
[b] Peck and Metcalf (1991) Premade Packets can be Purchased
[c] Handle formalin according to 29 CFR 1910.1048.

ductivity of 37.8 S/cm at 25°C (Peck and Metcalf, 1991). A fresh lot of the daily QCCS should be prepared every two weeks from the stock standard solution. Check the performance of the conductivity meter by following the procedure presented in Table 3-2.

3.1.4 Preparation of Equipment and Supplies

To ensure that all activities at a river can be conducted completely and efficiently, field teams should check all equipment and supplies before traveling to a river site. In addition, sample containers and labels should be prepared ahead of time to the extent possible.

Check the inventory of equipment and supplies prior to departure using the river-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. Storing sensitive equipment in protective plastic cases (e.g., Pelican® cases) is recommended. Also, many smaller pieces of equipment and other supplies can be packed in labeled coolers or plastic totes. Secure the rafts to the trailer, one on top of the other with tie-down straps and stow the fishing gear in the rafts. Make sure everything stored in the rafts is secure so that nothing can blow away when traveling to and from a sample site.

If necessary, prepare stock preservative solutions as described in Table 3-1. Follow the regulations of the Occupational Safety and Health Administration (OSHA) for handling

Table 3-2. Performance Check of Conductivity Meters.

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 meter's 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 S/cm of the theoretical value, rinse the probe in deionized water. Store as described in the operating manual and package the meter for transport to the river 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.

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 and, if possible, outside the vehicle cab.

Refuel vehicles and conduct maintenance activities the night before each sampling trip, if possible. Inspect the vehicles every morning before departure. Check vehicle lights, turn signals, brake lights, and air pressure in the tires. Also inspect the flatbed trailer used to transport the rafts. Trailer hubs must be greased often, especially if submerged in water when launching the rafts.

3.2 Activities after Each River Visit

Some sample containers can be labeled before departing from the base site. Figure 3-2 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. The microbial bottle (Section 5) can also be pre-labeled as described above and stored with the cubitainer and syringes. Sample containers for biological and sediment samples should NOT be pre-labeled before reaching the river

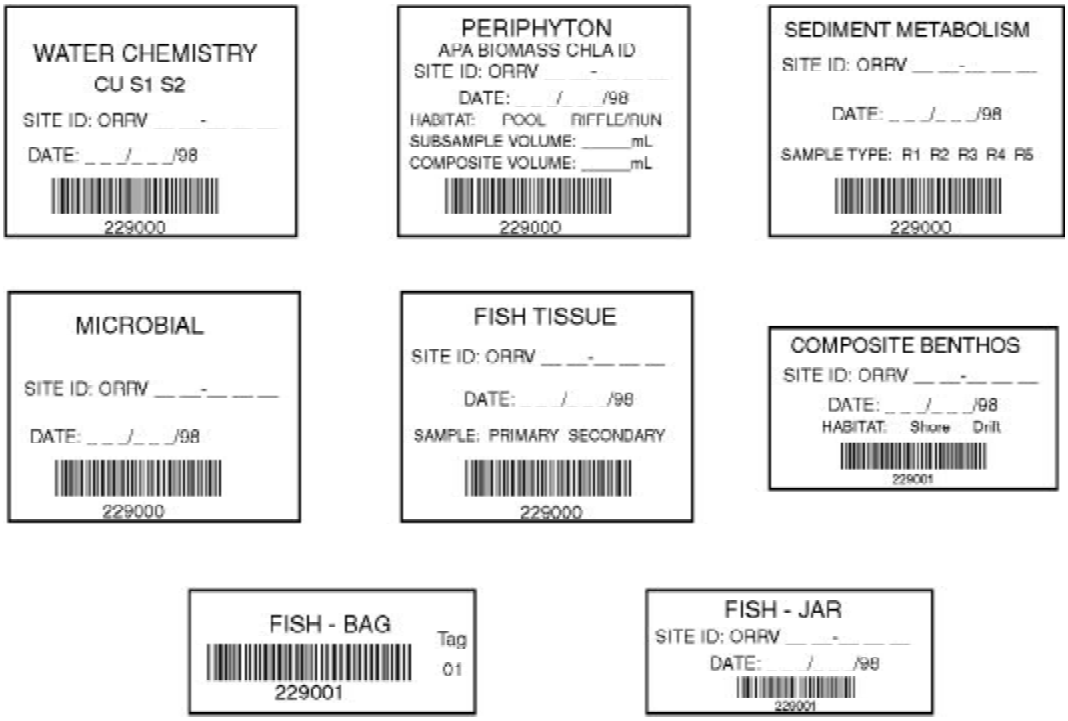

Figure 3-2. Sample container labels

site. Problems in sample tracking can result if jars are labeled and then are not used at a river.

Upon reaching lodging facilities or the base site after sampling a river, 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 must initial all data forms after review. If the team returns to the base site the day of sampling, the samples are not shipped. Under these circumstances, all sample information such as barcodes and sample condition is recorded by the team on a central log-in sheet. The team then stores all samples in their proper locations (e.g., freezer, refrigerator, chemical cabinet). The other team members should inspect and clean sampling equipment, store rafts (if at the base site), check the inventory of supplies, and prepare samples for shipment. If not already completed, the sediment metabolism samples should be processed. Other activities that must be conducted include shipping samples and communicating with the field coordinator or other central contact person.

3.2.1 Equipment Care

Equipment cleaning procedures are given in Table 3-3. Inspect all equipment, including nets, and clean off any plant or 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,

Table 3-3. Equipment Care after Each River Visit.

1. Clean for biological contaminants (e.g., plant and animal material).
   - Prior to departing each river, drain all water from the live well and buckets used to hold and process fish.
   - Inspect sampling gear for evidence of plant fragments and remove any fragments observed.
   - At the stream or base site, dry out dip nets, and kick nets, and inspect and remove any remnant vegetation or animal life. If the weather is rainy and gear cannot be dried out, then use a different (backup) set of gear, if available. If an additional set of gear is not available, disinfect gear with 10% bleach solution.
   - Clean the rafts by rinsing dirt and debris from the outside and the floors.
2. 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 site.
   - 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 river sample. Place the beakers in a 1-gallon self-sealing plastic bag with a cubitainer for use at the next river.
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.
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. Replace batteries as necessary (GPS, DO meter).
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, electrofishing generator, and spare gas container.

clean and disinfect them with a 10% chlorine bleach solution. 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.

3.2.2 Sample Tracking, Packing, and Shipment

Each field team must pack and ship samples from each sampling visit as soon as possible after collection, normally the day following the visit. Field teams will be provided with specific information for shipping destinations, contact persons, and the required shipping schedule for each type of sample. If the team returns to the base site the same day samples are collected, and the base site is the location of all sample analyses, samples are not required to be shipped.

Sample tracking information (including sample types, sample ID numbers, sample condition, number of samples, and other field-related information that is required by the laboratory to conduct analyses and associate results to a specific sample and river site) is recorded during the packing process. The field form used to record this information and accompany the sample shipment to the laboratory is illustrated in Figure 3-3. Procedures for conducting sample tracking activities should be provided to each field team by the information management staff. The sample

FIELD SAMPLE SHIPMENT TRACKING FORM

LAB NAME	CHEM ONE	AIRBILL NUMBER	1234567890
LAB CONTACT	J. DOE	IM CONTACT	S. DOE
DATE SENT	8-5-98	DATE RECEIVED	8-6-98
TEAM ID	(1) 2 3 4 5 6		
SITE ID	ORRV 98-999	VISIT	0 (1) 2 3

SAMPLE ID (BARCODE)	SAMPLE TYPE	CONDITION	COMMENTS
243223	FISH	T	PRIMARY SAMPLE (1 BAG)
243224	FISH	T	SECONDARY SAMPLE (1 BAG)
243663-667	SMET	OK	5 TUBES
243012	CHEM	OK	2 SYRINGES, 1 CUBITAINER
243113	MICR	OK	1 SAMPLE
243612	BENT	OK	DRIFT SAMPLE (1 JAR)
243611	BENT	OK	SHORE SAMPLE (1 JAR)
244013	PERI	OK	2 FILTERS, 2 TUBES
243360	VERT	OK	3 JARS

SAMPLE TYPES

BENT	=	Benthos
CHEM	=	Water Chemistry
FISH	=	Fish Tissue
MICR	=	Microbial
PERI	=	Periphyton
SMET	=	Sediment Metabolism
VERT	=	Fish Museum

CONDITION CODES

B	=	Broken Syringe Tip
C	=	Cracked Jar
F	=	Frozen
L	=	Leaking
ML	=	Missing Label
NP	=	Not Preserved
OK	=	Seems Fine
T	=	Thawed but Still Cold
W	=	Warm

Rev. 08/18/98 (strvssir.98)

FIELD SAMPLE SHIPMENTTRACKING FORM - STREAM/RIVERS - 1

Figure 3-3. Tracking form to accompany shipped samples

tracking system should identify the final destinations for each sample, and provide an informal "chain-of custody" to prevent the loss of samples and associated information.

General guidelines for packing and shipping the various types of samples described in this manual are presented in Table 3-4. Use fresh ice when shipping samples requiring 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 possible, place samples into a sealed plastic container to protect them from meltwater.

Water chemistry and microbial samples must be shipped as soon as possible after collection in order to meet holding time requirements for some laboratory analyses. To ship water chemistry and microbial samples, place a large (30-gallon) plastic bag in an insulated shipping container (e.g., a plastic cooler). The sample labels on the cubitainer, syringes, and glass microbial bottle(s) should be completely covered with clear tape to prevent damage from water or condensation during shipment. Place the syringes and microbial bottle(s) into a separate plastic container for shipment. Place the cubitainer and plastic 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 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 shipped without designating them as a hazardous material. Specific directions for packing, labeling, transporting, and shipping samples containing formalin or ethanol will be provided to each field team.

Each team leader must contact the field coordinator or other central contact person after each river visit to notify that the team is safely off the river, provide a brief update of each sampling visit, and request replenishment of supplies, if necessary. The team leader must also provide, for each shipment, the river 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 river visit and submit requests for replenishment well in advance of exhausting on-hand stocks.

3.3 Equipment and Supplies

A checklist of equipment and supplies required to conduct the activities described in Section 3 is presented in Figure 3-4. This checklist is similar to the checklist in Appen-

Table 3-4. General Guidelines for Packing and Shipping 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 (or other sample tracking report).
Microbial (200 mL glass bottle)	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 microbial bottles in a plastic container (with syringes). Place container inside of a second plastic bag (as above). Cover labels completely with clear tape. Confirm the sample ID assigned on the labels matches the ID number recorded on the field collection form (or other sample tracking report).
	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. (Portable Freezers Periphyton biomass (filter are available that can be run off a in aluminum foil cigarette lighter while in the field and electrical outlets in Motels).
Periphyton activity (50-mL centrifuge tube)	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.
Sediment metabolism (50-mL centrifuge tubes)	Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form (or other sample tracking report).
	Samples requiring freezing (-20°C) within 24 hours of collection
Fish Tissue (aluminum foil;	If samples cannot be kept frozen in the field, ship on day of collection or within two 30-gal plastic bags) 24 h by overnight courier. (Portable Freezers are available that can be run off a cigarette lighter while in the field and electrical outlets in Motels).

(continued)

Table 3-4. Continued.

Sample Type (container)	Preservative	Guidelines
		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.
	Samples requiring preservation in formalin	
Periphyton ID (50-mL centrifuge tube)	10% buffered formalin	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.
Fish Specimens (1-L , 2-L, and/or 4-L jars)	10% buffered formalin	Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form (or other sample tracking report). Special shipping containers, outside labeling, and shipping forms may be required for shipments containing formalin.
	Samples requiring preservation in ethanol	
Benthic Macroinvertebrates (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 (or other sample tracking report). Special shipping containers, outside labeling, and shipping forms may be required for shipments containing ethanol.

Base Location Activities		
Qty.	Item	
Before Departure for River		
1	Dossier of access information for scheduled river 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" Assorted extra batteries for dissolved and conductivity meters	
1 set	Completed water chemistry sample labels (3 labels with same barcode)	
1	Completed microbial sample label	
1 set	Water chemistry sample containers (one 4-L Cubitainer and two 60-mL syringes with a plastic storage container	
1	Microbial sample container (200 mL specially prepared square glass bottle)	
1 box	Clear tape strips to cover completed sample labels	
1	Checklist of all equipment and supplies required for a river visit	
Packing and Shipping Samples		
	Ice	
1 box	1-gal heavy-duty sealable plastic bags	
1-box	30-gal plastic garbage bags	
2	Insulated shipping containers (plastic coolers) for frozen samples	
2	Containers suitable to transport and/or ship samples preserved in formalin and ethanol	
	Shipping airbills and adhesive plastic sleeves	

Figure 3-4. Equipment and supply checklist for base location activities

dix A, which is used at the base location to ensure that all of the required equipment is brought to the river. Use this checklist to ensure that equipment and supplies are organized and available at the river site in order to conduct the activities efficiently.

3.4 Literature Cited

Metcalf, R. C. and D. V. Peck. 1993. A dilute standard for pH, conductivity, and acid neutralizing capacity measurement. Journal of Freshwater Ecology 8:67-72.

Peck, D. V. and R. C. Metcalf. 1991. Dilute, neutral pH standard of known conductivity and acid neutralizing capacity. Analyst 116:221-231.

Section 4
Initial Site Procedures

Alan T. Herlihy[1] and Jim Lazorchak[2]

When a field team first arrives at a non-wadeable site, they must first determine if the stream or river meets certain criteria for sampling and data collection. They must inspect the selected river reach for appropriate access, safety, and general river conditions. After locating public or private launch sites, the rafts or boats are unloaded and the vehicles are shuttled to and from launch locations. Certain conditions at the time of the visit may warrant the collection of only a subset of field measurements and samples. The crew then measures the width of the non-wadeable stream or river at several points and lays out the reach boundaries on a map, within which all subsequent sampling and measurement activities are conducted.

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4.1 Site Verification Activities

4.1.1 Locating the Index Site

Non-wadeable stream and river sampling points were chosen from the "blue line" network represented on 1:100,000- scale USGS maps, following a systematic randomized design developed for EMAP 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" or "X-site". The latitude/longitude of the X-site will be listed on an information sheet that is part of the dossier compiled for each river (see Section 3).

Complete a verification form for each non-wadeable stream or river visited (regardless of whether you decide to sampling it), following the procedures described in Table 4-1. While traveling from a base location to a site, record a detailed description of the route

Table 4-1. Site Verification Procedures.

1. Find the stream or river 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. Use a GPS receiver to record the latitude and longitude of transects "A" and "K," and if possible, confirm the X-site with 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 or river as marked on the map, including: 1:24,000 USGS map orienteering, topographic landmarks, county road maps, local contacts, etc.
4. While scouting access and shuttling vehicles, reconnoiter the river channel upstream and downstream from the X-site, and assign one of the following sampling status categories to the stream. Record the category on the Verification Form.

Target Categories

A. Regular Wadeable Stream: - The stream can be sampled with wadeable stream procedures.
B. Regular-Partial Boatable and Wadeable Combination: If over half the reach is non-wadeable, sample it with the non-wadeable protocols.
C. Regular-Boatable: A stream too deep to be safely sampled by wading following our wadeable stream protocols. The stream or river can be sampled with Non-Wadeable procedures.
D. Intermittent Stream: The flow of water is not continual, but the channel is wet. Sample using modified procedures.
E. Dry Channel: A discernible stream channel is present but there is no water at the site. Sample using modified procedures.
F. 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-target Categories

A. No Stream Channel (map error): No water body or stream channel is present at the coordinates provided for the X-site.
B. Impounded stream: The stream is submerged under a lake or pond due to man-made or natural (e.g., beaver dam) impoundments.
C. Marsh/Wetland: 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.

Inaccessible Categories

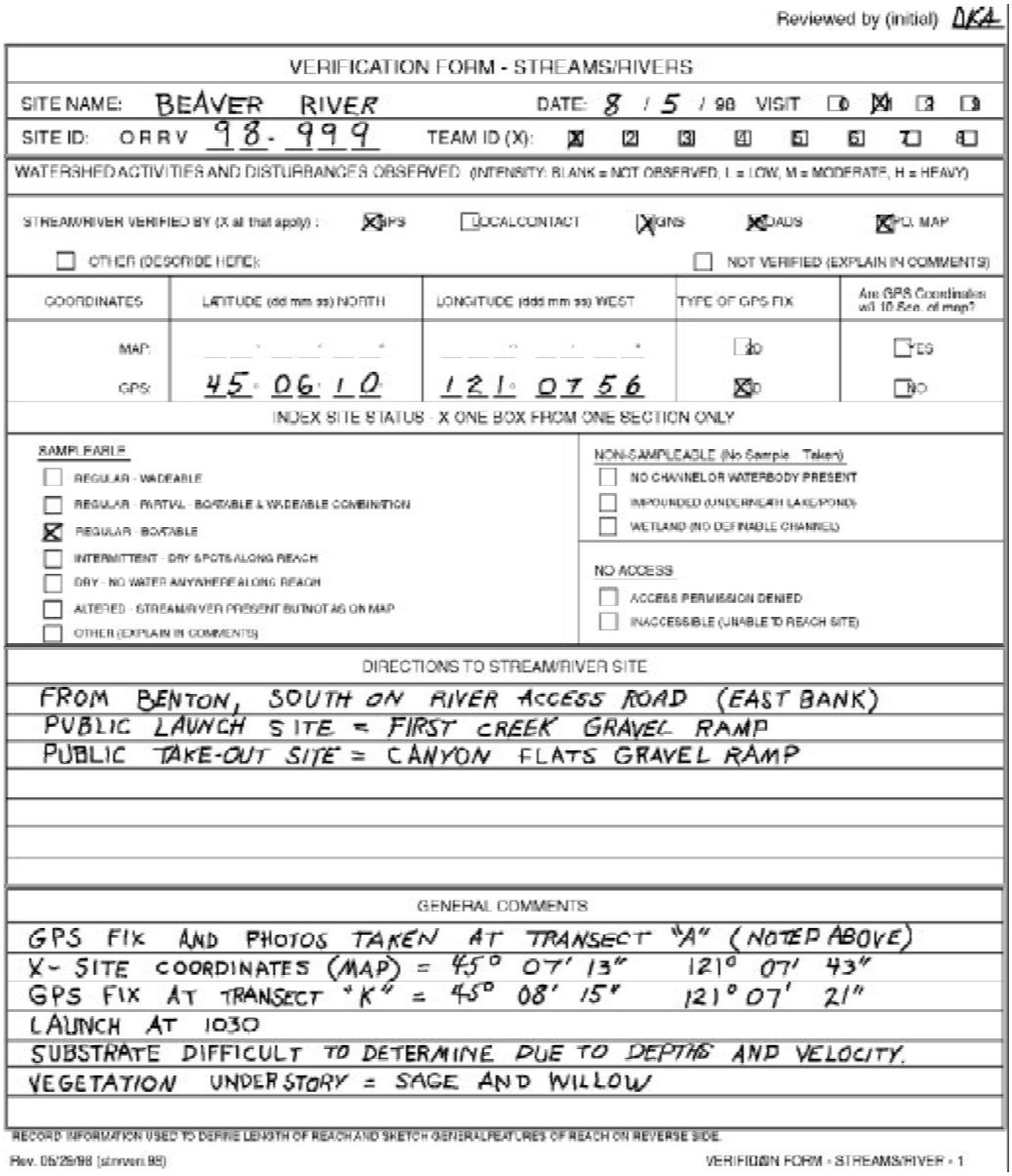
A. Physical Barriers: If you are physically unable to reach the X-site because of poor or no river access, or other obstacles that prohibit safe sampling.
B. No Permission: You are denied access to launch sites by the landowners.

5. Do not sample "Non-target" or "Inaccessible" sites. Place an "X" in the appropriate box in the "Non-Sampleable" section of the Verification Form and provide an explanation in the comments section.

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 locating the sample reach and X-site for a stream or river, confirm its location and that the team is at the correct stream or river. 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 Non-wadeable Stream or River

Not all chosen non-wadeable sites will turn out to be streams or rivers. On the basis of previous synoptic surveys, it was found



Reviewed by (initial) DKA

VERIFICATION FORM - STREAMS/RIVERS

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT ☐ ☒ ☐ ☐

SITE ID: ORRV 98-999 TEAM ID (X): ☒ ☐ ☐ ☐ ☐ ☐ ☐ ☐

WATERSHED ACTIVITIES AND DISTURBANCES OBSERVED (INTENSITY: BLANK = NOT OBSERVED, L = LOW, M = MODERATE, H = HEAVY)

STREAM/RIVER VERIFIED BY (X all that apply): ☒ GPS ☐ LOCAL CONTACT ☒ IGNS ☒ QUADS ☒ CO. MAP

☐ OTHER (DESCRIBE HERE) ☐ NOT VERIFIED (EXPLAIN IN COMMENTS)

COORDINATES	LATITUDE (dd mm ss) NORTH	LONGITUDE (ddd mm ss) WEST	TYPE OF GPS FIX	Are GPS Coordinates w/i 10 Sec. of map?
MAP:	___ ___ ___	___ ___ ___	☐ 2D	☐ YES
GPS:	45 06 10	121 07 56	☒ 3D	☐ NO

INDEX SITE STATUS - X ONE BOX FROM ONE SECTION ONLY

SAMPLEABLE
- ☐ REGULAR - WADEABLE
- ☐ REGULAR - PARTIAL - BOATABLE & WADEABLE COMBINATION
- ☒ REGULAR - BOATABLE
- ☐ INTERMITTENT - DRY SPOTS ALONG REACH
- ☐ DRY - NO WATER ANYWHERE ALONG REACH
- ☐ ALTERED - STREAM/RIVER PRESENT BUT NOT AS ON MAP
- ☐ OTHER (EXPLAIN IN COMMENTS)

NON-SAMPLEABLE (No Sample Taken)
- ☐ NO CHANNEL OR WATERBODY PRESENT
- ☐ IMPOUNDED (UNDERNEATH LAKE/POND)
- ☐ WETLAND (NO DEFINABLE CHANNEL)

NO ACCESS
- ☐ ACCESS PERMISSION DENIED
- ☐ INACCESSIBLE (UNABLE TO REACH SITE)

DIRECTIONS TO STREAM/RIVER SITE

FROM BENTON, SOUTH ON RIVER ACCESS ROAD (EAST BANK)
PUBLIC LAUNCH SITE = FIRST CREEK GRAVEL RAMP
PUBLIC TAKE-OUT SITE = CANYON FLATS GRAVEL RAMP

GENERAL COMMENTS

GPS FIX AND PHOTOS TAKEN AT TRANSECT "A" (NOTED ABOVE)
X-SITE COORDINATES (MAP) = 45° 07' 13" 121° 07' 43"
GPS FIX AT TRANSECT "K" = 45° 08' 15" 121° 07' 21"
LAUNCH AT 1030
SUBSTRATE DIFFICULT TO DETERMINE DUE TO DEPTHS AND VELOCITY.
VEGETATION UNDERSTORY = SAGE AND WILLOW

RECORD INFORMATION USED TO DEFINE LENGTH OF REACH AND SKETCH GENERAL FEATURES OF REACH ON REVERSE SIDE.

Rev. 05/29/98 (stmvsn.98) VERIFICATION FORM - STREAMS/RIVER - 1

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

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 and river length in the area. After the river and location of the X-site are confirmed, evaluate the river reach surrounding the X-site and classify the stream into one of three major sampling status categories (Table 4-1). The primary distinction between "Non-target" and "Target" streams and rivers is the flow and size characteristics of the water body and adequate access to a river site.

Record the site class and pertinent site verification information on the Verification Form (Figure 4-1). If the site is non-target 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. First, 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 4-2. 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 (digital cameras may be convenient because no film processing is required) 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 or river, start the sequence with one photograph of an 8.5 × 11 inch piece of paper with the stream ID and date printed in large letters. After the photo of the stream ID information, take one photograph at the up river transect (A) and one at the X-site. 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

Table 4-2. 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.
- 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.

habitat structure measures require sampling a certain length of a non-wadeable stream or river to get a representative picture of the ecological community. Previous EMAP pilot studies have suggested that a length of 40-100 times the channel width is necessary to collect at least 90% of the fish species occurring in the stream or river reach. In western streams a support reach that is 100 channel widths long around the X-site is required to characterize the community and habitat associated with the sampling point while 40 channel widths has been found to be adequate in eastern streams. Establish the sampling reach about the X-site using the procedures described in Table 4-3. 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 (or the midpoint of the reach) 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

Table 4-3. Laying Out The Sampling Reach.

1. Consult the 7.5 minute USGS topographic map in the site dossier. The river width is estimated and the reach boundaries are marked between the two boat launches. The total reach length equals 100 times or 40 times the mean river width.

2. Using a laser rangefinder, measure the non-wadeable stream or river width in several places, specifically the X-site and the two boat launches.

   Perform these duties while verifying the reach location and evaluating raft or boat launch and/or egress sites.

3. Confirm the river width and reach boundaries. If different than dossier information, adjust as necessary by using a map wheel and delineating new reach boundaries on the map. Multiply mean river width by 100 or 40 and delineate 50 or 20 channel widths upstream and downstream of the X-site.

   It is OK to shift the reach up or downriver to maximize effort efficiency and access. However, the X-site must be within the reach to be sampled.

4. Record the river width on page 2 of the Verification Form.

5. Extensive shallows, large log jams, absence of launch sites or vehicle access, and hazardous whitewater may preclude rafting.

6. With the map wheel, determine the distance from the raft or boat put-in location to the beginning of the reach. This is the distance to float or boat before sampling begins.

7. Using the laser rangefinder at the most upriver transect (Transect "A"), measure 10 or 4 channel widths downriver to the next transect (Transect "B"). Continue this while sampling until the final transect (Transect "K").

8. Sample odd numbered STRM_ID's along the left shore (facing downriver); sample even numbered sites along the right shore.

   Large obstructions or hazards may require temporary diversion.

Reviewed by (initial) DKA

VERIFICATION FORM - STREAMS/RIVERS

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT ☐1 ☒2 ☐3

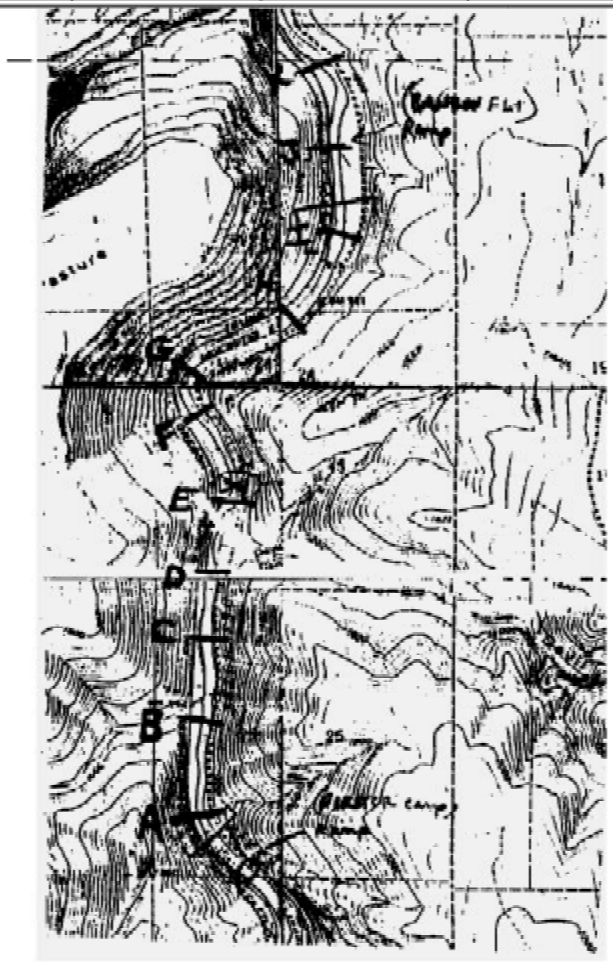
SITE ID: ORRV 98-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

STREAM/RIVER REACH DETERMINATION

CHANNEL WIDTH USED TO DEFINE REACH (M) (XX):	DISTANCE (M) FROM X-SITE		COMMENT
	UPSTREAM LENGTH	DOWNSTREAM LENGTH	
40	1900	~3000	OVERESTIMATED INTERVALS

ARROW INDICATES NORTH

N



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VERIFICATION FORM - STREAMS/RIVERS - 2

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

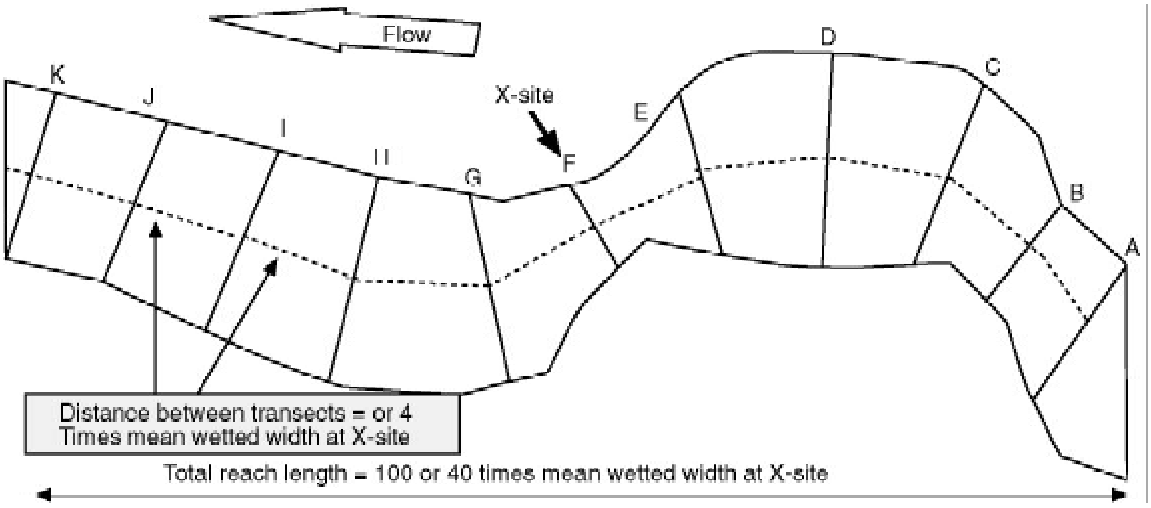

Figure 4-3. Sampling reach features.

transects used for physical habitat characterization (Section 6). Samples for periphyton (Section 7), sediment metabolism (Section 8), benthic macroinvertebrate (Section 9), aquatic vertebrates (Section 10), and tissue (Section 11) are collected only along the designated shoreline (refer to Table 4-3, step 8).

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 accommodate river access or to avoid river hazards or obstacles. If the beginning or end of the reach cannot be sampled due to obstacles or hazards, 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, access points may necessitate sliding the reach. Do not "slide" the reach so that the X-site falls outside of the reach boundaries. Sites which are 100X the average stream may be too time consuming to consider. So the field crew should avoid sliding the reach and decide whether the site really meets site selection criteria. If not, then the reach is not a target site. Also, do not "slide" a reach to avoid man-made obstacles such as bridges, rip-rap, or channelization. These represent features and effects that EMAP is attempting to study.

Before leaving the non-wadeable site, mark all transects on the supplied 7.5 minute topographic map and note that a photocopied version is attached to 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 Equipment And Supplies

A list of the equipment and supplies required to conduct the non-wadeable stream and river 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 lo-

Equipment and Supplies for Initial Site Activities		
Qty.	**Item**	
1	Dossier of site and access information	
1	Topographic map with "X-site" and proposed width and reach boundaries	
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	Laser rangefinder and clear waterproof bag	
1	Map wheel	
1	Calculator	
1	Metric ruler	
1	Waterproof camera and film	
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.

Section 5
Water Chemistry and Microbiology

Alan T. Herlihy[1] and Charles W. Hendricks[2]

There are two components to collecting water chemistry information: Collecting samples of stream or river water to ship to the analytical laboratory, and obtaining in situ measurements of specific conductance, dissolved oxygen, and temperature. At each river, teams fill one 4-L container and two 60 mL syringes with river water. 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 (Acid Neutralizing Capacity, ANC)
- 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 and dissolved inorganic carbon. Syringes are used to seal off the samples from the atmosphere because the pH and dissolved inorganic carbon (DIC) will all change if the streamwater equilibrates with atmospheric CO2. 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 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. 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

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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).

5.1 Microbial Sampling

Separate samples are collected for the culture of bacteria that are present in water. The organisms are living entities and respond to fluctuations and nutrient stress just like higher forms. They may be native to the water or introduced to the stream or river by the addition of human or animal contamination. We are currently using culturing procedures that can differentiate between the two events based on relative numbers of bacteria of cultured bacteria.

Samples of stream and river water are to be taken in laboratory prepared sterile, glass bottles. These square bottles are designed for sample collection, transit and storage, and no other container should be used for bacteriological samples. Generally one (1) sample is collected per site, but, duplicate (or triplicate) samples from the same site may be requested for statistical purposes. Occasionally, two (2) or more samples at specific sites along a stream or river reach may be requested. The number of samples to be taken will be noted on the field activity sheets.

Ideally, microbiological examination of a water sample should begin promptly after collection to avoid unpredictable changes in the numbers of organisms present in the sample. If samples cannot be processed within 1 hour after collection, an iced cooler for storage during transport (overnight express mail) to the laboratory must be used. All microbiology samples are to be iced during transport and refrigerated in the laboratory, while awaiting analysis within 24 hours of collection.

5.2 Sample Collection

Before leaving the base location, package the sample containers (one 4-L cubitainer, two 60 mL syringes, and two 200 mL sterile glass bottles) and the stream sample beaker to prevent contamination (see Section 3). Fill out a set of water chemistry and microbial sample labels as shown in Figure 5-1. Attach a completed label to the cubitainer, each syringe, and each glass bottle 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 water chemistry labels all have the same sample ID number (barcode), and that the labels are securely attached. Also, make sure the microbial 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 5-1. The sample is collected from the middle of the flowing stream whenever feasible or if the X Site is not accessible take the sample from the river channel at the last sample transect. 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 sites 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 chemi-

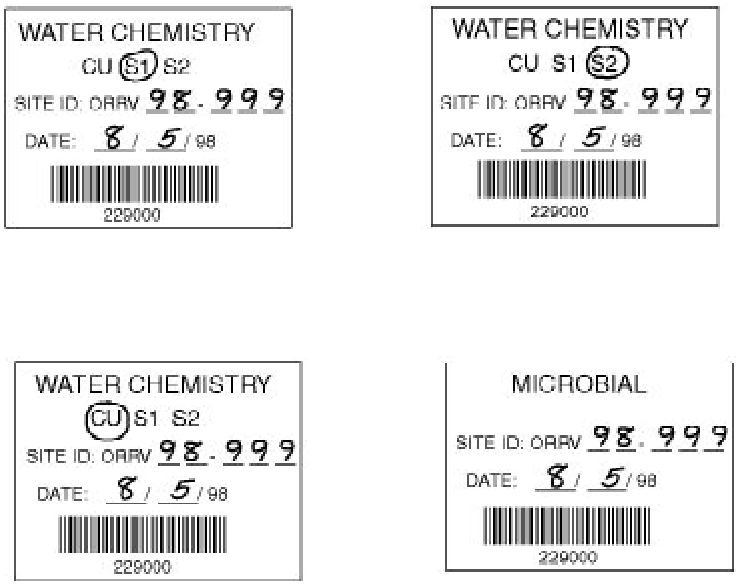

Figure 5-1. Completed sample labels for water chemistry and microbiology.

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

The procedure to collect a microbial sample is described in Table 5-2. The sample is collected from the middle of the flowing stream or river channel at the last sample transect. Collect samples that are representative of the water being tested and use aseptic techniques to avoid sample contamination. Take care to avoid contact with the bank or stream bed; otherwise, contamination of the sample may occur making it useless for analysis.

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.

5.3 Field Measurements

Table 5-3 presents the procedures for obtaining field measurement data for the water chemistry indicator. The conductivity and dissolved oxygen meters 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) is prepared according to directions presented in Section 3. The results of field checks of these meters, as well as the measured values for specific conductance, dissolved oxygen, and stream temperature, are recorded on the Field Measurement Form as shown in Figure 5-3.

5.4 Equipment and Supplies

A list of equipment and supplies required to collect samples and field data for the water chemistry and microbiology indicator is presented in Figure 5-4. This checklist is similar

Table 5-1. Sample Collection Procedures for Water Chemistry.

Collect the water samples from either the X site or the last transect in a flowing portion near the middle of the river.

1. Rinse the 500 mL sample beaker three times with river water. Discard the rinse downriver.

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 river water 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 river water 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 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 and shut the lid.

6. Submerge a 60-mL syringe halfway into the river 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 river 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 both syringes in a small plastic tote and store with the cubitainer in an ice filled cooler. Keep samples on ice until they reach the laboratory.

10. Record the site number (Sample ID) on the Sample Collection Form along with the pertinent river information (river name, ID, date, etc.). Note anything that could influence sample chemistry (heavy rain, potential contaminants) in the Comments section. 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.

Table 5-2. Sample Collection Procedures for Microbiology.

Collect the water samples from the last transect in a flowing portion near the middle of the stream. The following procedures describe sampling of Non-Wadeable streams and rivers for bacterial analysis. These techniques have been proven useful in evaluating indicators of functional diversity and sanitary significance, including the quality of all types of waters.

1. Keep the sampling bottle closed until it is to be filled.

2. Remove stopper and cap as a unit; do not contaminate inner surface of stopper or cap and neck of bottle.

3. Fill container without rinsing.

4. Water samples are taken from the upstream side of a boat by holding the bottle near its base in the hand and plunging it, neck downward, below the water's surface. Turn bottle until neck points slightly upward and mouth is directed toward the current.

5. If there is no current, as in the case of a reservoir, create a current artificially by pushing bottle forward horizontally in a direction away from the hand.

6. If it is not possible to collect samples from these situations in this way, attach a weight to base of bottle and lower it into the water.

7. When the sample is collected, leave ample air space in the bottle (at least 2.5 cm) to facilitate mixing at later stages in the examination of the water.

8. Replace stopper or cap immediately and apply electricians tape around the neck to send the bottle. If used, secure the hood around outside of the neck of the bottle and cap.

9. Place the microbial sample with the chemistry syringes in a small plastic tote and surround with ice in a cooler.

10. Repeat steps 1 through 9 if duplicate or triplicate samples are desired.

11. Record the barcode number (Sample ID) on the Sample Collection Form along with the pertinent river information (river name, ID, date, etc.). Note anything that could influence sample chemistry or microbiology (heavy rain, potential contaminants) in the Comments section. 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.

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SAMPLE COLLECTION FORM - RIVERS (continued)

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT ☐1 ☒2 ☐3 ☐

SITE ID: ORRV 98-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

CHEMISTRY AND MICROBIAL WATER SAMPLE (Chem: 4-L Cubitainer and 2 Syringes, Micro: Glass Bottle)

	SAMPLE ID (BARCODE)	TRANSECT	FLAG	COMMENTS
CHEMISTRY	243012	K		
MICROBIAL	243113	K		

SEDIMENT TOXICITY SAMPLES

SAMPLE ID (BARCODE)	FLAG	COMMENTS

FISH TISSUE SAMPLES - PRIMARY SAMPLE (min. 50g total wgt)

SAMPLE ID (BARCODE) → 243223

LINE	SPECIES CODE	COMMON NAME	NUMBER OF INDIVIDUALS	FLAG
P1	COTTZZZZ	UNKNOWN COTTIA	11	

IS COMPOSITE SAMPLE COMPOSED OF INDIVIDUALS COLLECTED FROM THROUGHOUT REACH? (X) → ☐YES ☒NO

IF NO, EXPLAIN: COLLECTED AT ONE PROFILE

FISH TISSUE SAMPLES - SECONDARY SAMPLE (where available; 5 individuals)

SAMPLE ID (BARCODE) → 243224

LINE	SPECIES CODE	COMMON NAME	TOTAL LENGTH (MM)	FLAG
S1	PTYCOREG	NORTHERN SQUAWFISH	220	
S2	PTYCOREG	NORTHERN SQUAWFISH	220	
S3	PTYCOREG	NORTHERN SQUAWFISH	265	
S4	PTYCOREG	NORTHERN SQUAWFISH	290	
S5	PTYCOREG	NORTHERN SQUAWFISH	215	

IS COMPOSITE SAMPLE COMPOSED OF INDIVIDUALS COLLECTED FROM THROUGHOUT REACH? (X) → ☒YES ☐NO

IF NO, EXPLAIN:

LINE	COMMENT OR FLAG EXPLANATION FOR FISH TISSUE

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

Rev. 05/29/98 (_rvscmb.98)

SAMPLE COLLECTION FORM - RIVERS - 2

Figure 5-2. Sample Collection Form (page 2), showing data recorded for water chemistry and microbial samples.

Table 5-3. Procedures for Streamside and In Situ Chemistry Measurements.

Specific Conductance

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

2. Insert the probe into the "RINSE" container of the quality control check sample (QCCS) and swirl for 3 to 5 seconds. Shake off the probe and transfer to the "TEST" container of QCCS; let stabilize for 20 seconds. Record the conductivity of the QCCS on the Field Measurement Form.

   If the measured conductivity is not within 10% or 10 S/cm of theoretical value, repeat the measurement process. If the value is still unacceptable, flag the conductivity data on the Field Measurement Form.

3. Submerge the probe in an 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 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. Record the results of these checks on the Field Measurement Form.

3. 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. For doing the elevation correction, the elevation of the sample site is given on the site Information sheet in the dossier for the site. Record the pertinent calibration information on the Field Measurement Form.

4. 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. 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 pertinent data flags and comments.

Reviewed by (initial) DKA

FIELD MEASUREMENT FORM - STREAMS/RIVERS

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT ☐0 ☒1 ☐2 ☐3

SITE ID: ORRV 98-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

WEATHER CONDITIONS (X)

CLOUD COVER	☒ < 5%	☐ 5-25%	☐ 25-50%	☐ 50-75%	☐ >75%
PRECIPITATION	☒ NONE	☐ LIGHT	☐ MODERATE	☐ HEAVY	
PREVIOUS PRECIPITATION (24 H)	☒ NONE	☐ LIGHT	☐ MODERATE	☐ HEAVY	
AIR TEMPERATURE XX	28 °C				

IN SITU MEASUREMENTS

STATION ID: K Assume X-site unless marked

		FLAG	COMMENTS
QCCS COND µS/CM XXXX	75		AT TRANSECT "K"
STREAM/RIVER COND µS/CM XXXX	110		AT TRANSECT "K"
STREAM/RIVER DO MG/L XX.X	8.8		AT TRANSECT "K"
STREAM/RIVER TEMP °C XX.X	17.6		AT TRANSECT "K"

FISH TISSUE SAMPLES - SECONDARY SAMPLE (where available; 5 individuals)

INITIAL O_2 (MG/L) XX.X	INITIAL INCUBATION TEMP (°C) XX.X	INCUBATION TIME (24-HR TIME) START (HH:MM)	INCUBATION TIME (24-HR TIME) FINISH (HH:MM)	DURATION OF INCUBATION (HH:MM)	FLAG	COMMENTS
9.0	18.4	18:30	22:30	4 : 00		

SAMPLE ID (BARCODE)	FINAL O_2 (MG/L) XX.X	FLAG	COMMENTS
243663	7.5		INITIAL D.O. AT 1000' ELEVATION
243664	7.2		FINAL D.O. AT 2200' - METER CALIBRATED
243665	7.8		FOR EACH ELEVATION
243666	8.1		
243667	7.0		
CONTROL	8.8		

OXYGEN METER CALIBRATION INFORMATION

MEMBRANE CHECK ☒ ELECTRONIC ZERO ☐ RED LINE ☒

CALIBRATION CHAMBER TEMPERATURE: °C

SATURATED O_2 @ @ TEMP: MG/L

STATION ELEVATION (FROM TOPO. MAP OR ALTIMETER): 1000 FT

ELEVATION CORRECTION FACTOR: x

The calibration value is obtained by multiplying the saturated DO concentration times an elevation correction factor (obtained from the tables on the back of the YSI meter). Adjust the meter reading to the calibration value.

CALIBRATION VALUE: MG/L

COMMENTS:

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.

Rev. 05/29/98 (strvfldm.98)

FIELD MEASUREMENT FORM - STREAMS/RIVERS - 1

Figure 5-3. Field Measurement Form (page 1), showing data recorded for water chemistry.

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	500-mL plastic bottle of deionized water to store conductivity probe	
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	60 mL plastic syringes (with Luer type tip) with completed sample labels attached	
1	200 mL sterile glass microbial bottles with completed sample label attached (in clean plastic bag)	
1	Plastic container with snap-on lid to hold filled syringes	
2	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 roll	Electricians tape	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for water chemistry and microbiology	

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

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 Non-Wadeable stream or river. Use this checklist to ensure that equipment and supplies are organized and available at the stream or river site in order to conduct the activities efficiently.

5.5 Literature Cited

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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.

Section 6
Physical Habitat Characterization—Non-wadeable Rivers

by
Philip R. Kaufmann

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

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

All of these attributes may be directly or indirectly altered by anthropogenic activities. Nevertheless, their expected values tend to vary systematically with river size (drainage area) and overall gradient (as measured from

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

topographic maps). The relationships of specific physical habitat measurements described in this EMAP-SW field manual 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 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.

This protocol is intended for evaluating physical habitat in non-wadeable streams and rivers. Kaufmann and Robison (1998) describe other methods for use in smaller, wadeable streams. Like the methods for wadeable streams, these methods are most efficient during low flow conditions and when leaves are on terrestrial vegetation, but may be applied during other seasons and higher flows except as limited by safety considerations. It is designed for monitoring applications where robust, quantitative descriptions of reach-scale habitat are desired, but time is limited.

Like the wadeable streams protocol (Kaufmann and Robison 1998) this habitat characterization approach employs a randomized, systematic spatial sampling design to minimize 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 crew to estimate attributes that are otherwise measurable, rather than estimating the quality or importance of the attribute to 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. In our field trials, two people typically complete the specified channel, riparian, and discharge measurements in about three hours of field time. However, the time required can vary considerably with channel characteristics, flow conditions, and the location of boat launching areas.

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

jective classification of channel units such as pools, and quantification of residual pool depth, pool volume, and total stream volume.

6.1 Components of the Field Habitat Assessment

Field data collection for the physical habitat assessment is accomplished in a single float down each river sample reach. Depending on the survey region, river sample reach lengths are defined as either 40 or 100 times the wetted width in the vicinity of the point of entry (Figure 6-1). In addition to physical habitat assessment, the 2-person habitat team of the field crew collects chemical, macroinvertebrate, and periphyton samples (if applicable). They may also recon the channel if they precede the electrofishing boat down the river. To characterize mid-channel habitat (Table 6-1), they measure a longitudinal thalweg (or mid-channel) depth profile, tally snags, classify channel habitat types, characterize mid-channel substrate, and locate the 11 systematic transect locations for littoral/riparian sampling and other habitat observations (Figures 6-1 and 6-2). At each of the 11 marked reach transect locations (A-K), they measure channel wetted width, bankfull channel dimensions, incision, channel constraint, bearing and gradient; then assess near-shore, shoreline, and riparian physical habitat characteristics by measuring or observing littoral depths, riparian canopy cover, substrate, large woody debris, fish cover, bank characteristics, riparian vegetation, and evidence of human activities (Table 6-1). They also collect benthic macroinvertebrates (Section 9), take benthic algal samples (if applicable), and measure conductivity and water temperature using procedures described in section 5.

Mid-channel habitat measurements and observations are recorded on multiple pages of the Thalweg Profile Form (Figure 6-3). Instructions for these mid-channel procedures are given in section 6.5. Measurements made while anchored or tied up to the 11 littoral/riparian plot stations ("transects") are recorded on 11 copies of the two sided Channel/Riparian Transect Form (Figures 6-4 and 6-5). Instructions for these transect or littoral/riparian assessment activities are discussed in subsection 6.6.

6.2 Habitat Sampling Locations On The Study Reach

Measurements are made at two scales of resolution along the mid-channel length of the reach; the results are later aggregated and expressed for the entire reach, a third level of resolution (Figure 6-1). We want to assess habitat and other river indices over river reach lengths that are long enough to incorporate the habitat variability due to river meandering and pool-riffle structure. To accommodate habitat variability in a way that adjusts for varying sizes of rivers, EMAP protocols specify sample reach lengths that are a multiple of their average wetted width (40 or 100 Channel-Widths). Water velocity, habitat complexity, fish abundance, and species richness may also affect capture efficiency and consequently the required sample reach length. In the Oregon river pilot, it was found that 85 channel widths is adequate for Oregon rivers (Hughes et al. In Review). In the Mid-Atlantic region, river reaches of 40 channel widths long were used in order to make this aspect of field methods consistent between wadeable and non-wadeable streams. For this field manual, we discuss the methods used to

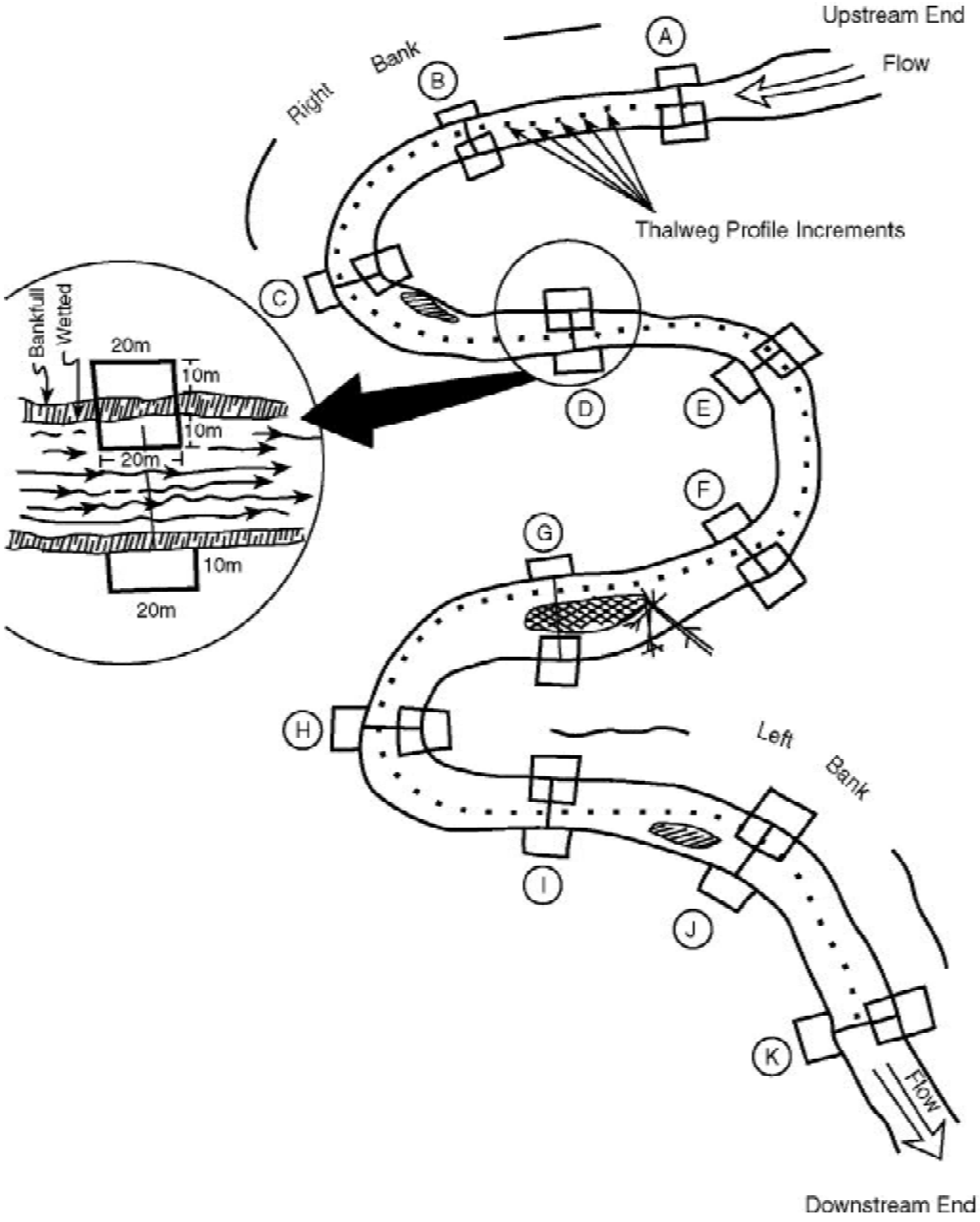

Figure 6-1. River reach sample layout.

Table 6-1. Components of River Physical Habitat Protocol.

1. Thalweg Profile:

   At 10 equally spaced intervals between each of 11 channel cross-sections (100 along entire reach):

   * Classify habitat type, record presence of backwater and off-channel habitats. (10 between cross sections, 100 total)
   * Determine dominant substrate visually or using sounding rod. (10 between cross-sections, 100 total)

   At 20 equally spaced intervals (for 100 ChW reach) or 10 equally spaced intervals (for 40 ChW reach) between each of 11 channel cross-sections:

   * Tally mid-channel snags - 10 (or 20) between cross-sections, 100(or 200) total.
   * Measure thalweg (maximum) depth using Sonar or rod - 10 (or 20) between cross-sections, 100(or 200) total.

2. Littoral/Riparian Cross-Sections: @ 11 stops ("transects") at equal intervals along reach length:

   Measure/estimate from one chosen bank on 11 channel cross-sections:

   * Gradient (clinometer or Abney level) between cross-section and next one downstream.
   * Bearing (compass) between cross-section and next one downstream.
   * Wetted width (laser range finder).
   * Mid-channel bar width (laser range finder).
   * Bankfull width and height (estimate).
   * Incision height (estimate).
   * Bank angle (estimate).
   * Riparian canopy cover (densiometer) in four directions from chosen bank.
   * Shoreline Substrate in the first 1m above waterline (est. dominant and subdominant size class).

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

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

filamentous algae	overhanging vegetation
aquatic macrophytes	undercut banks
large woody debris	boulders and rock ledges
brush and small woody debris	artificial structures

In 20m long Littoral Plot extending 10m landward starting at bankfull margin:[1]

   * Estimate areal cover class and type (e.g., woody) of riparian vegetation in Canopy, Mid-Layer, and Ground Cover
   * Observe and record human activities and disturbances and their proximity to the channel.

For largest visible Riparian Tree:

   * Estimate diameter (Dbh), height, species, and distance from river edge.

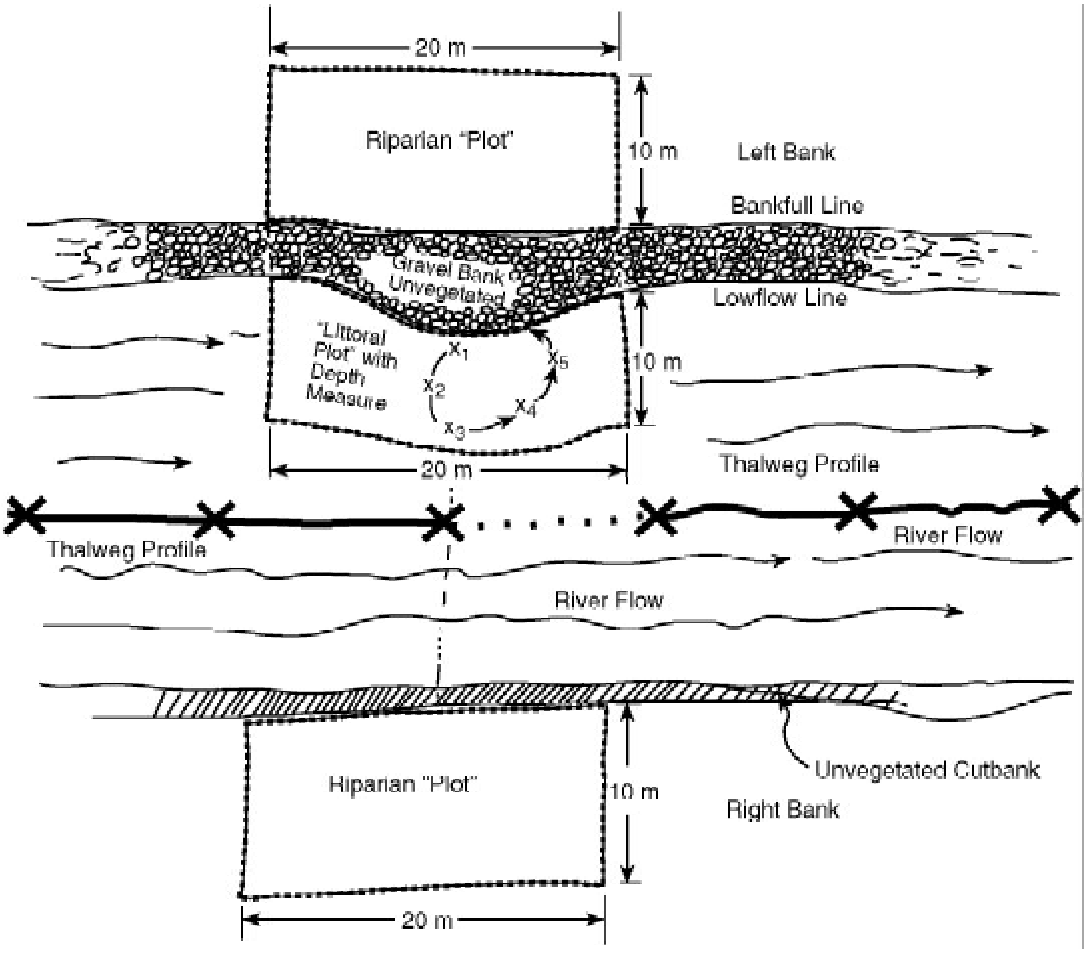

Figure 6-2. Littoral-Riparian Plots for characterizing riparian vegetation, human influences, fish cover, littoral substrate, and littoral depths.

sample reaches 40 times the mean wetted width at the vicinity of the launch point in Mid-Atlantic region streams and 100 times the mean wetted width in Oregon streams.

Section 4 describes the procedure for locating the X-site that defines the midpoint of the sample reach. This sampling location is already marked on a 1:24,000 map prior to going into the field. It has precise coordinates of latitude and longitude, and was selected by the EMAP design group using a randomized systematic sampling design. Subsections 6.3 and 6.4 describe the protocol for delineating a sample reach that is 40 or 100 times its width. Those sections also describe the protocol for measuring out (with a laser range finder) and locating the 11 littoral/riparian stations where many habitat measurements will be made. The distance between each of these stations is 1/10th the total length of the sample reach.

The thalweg profile measurements must be spaced as evenly as practicable over the entire sample reach length. 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 1/3 to 1/2 of the average channel width distance. To set the

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PHab: THALWEG PROFILE FORM - RIVERS

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT ☐1 ☒2 ☐3

SITE ID: ORRV 98-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

TRANSSECT (X) ☒A-B ☐B-C ☐C-D ☐D-E ☐E-F ☐F-G ☐G-H ☐H-I ☐I-J ☐J-K

SUBSTRATE CODES	CHANNELHABITAT CODES	OTHER
BH = Bedrock/Hardpan (smooth or rough) • (larger than a car) BL = Boulder (250 to 4000 mm) • (basketball to car) CB = Cobble (64 to 250) • (tennis ball to basketball) GR = Coarse to Fine Gravel (2 to 64) • (ladybug to tennis ball) SA = Sand (0.06 to 2 mm) • (gritty - up to ladybug size) FN = Silt/Clay/Muck • (not gritty) OT = Other (?) (Comment Please)	PO = Pool GL= Glide RI = Riffle RA= Rapid CA= Cascade FA= Falls DR = Dry Channel	Off Ch. = Off Channel or Backwater
Circle One Substrate Code for Each Station	Circle One Channel Habitat Code for Each Station	X if Yes

REMEMBER: A = Upstream End of Reach and K = Downtown End of Reach

STATION	SNAG (X)	DEPTH (ETHER) SONAR (ft) xx	DEPTH (ETHER) POLE (m) x.x	SUBSTRATE (CIRCLE ONE)	CHANNEL HABITAT (CIRCLE ONE)	OFF CH. (X)	FLAG
1		12		BH (BL) CB GR SA FN OT	PO GL (RI) RA CA FA DR		
2		10					
3		9		BH (BL) CB GR SA FN OT	PO (GL) RI RA CA FA DR		
4		9					
5		6		BH (BL) CB GR SA FN OT	PO (GL) RI RA CA FA DR		
6		5					
7		4		BH (BL) CB GR SA FN OT	PO GL (RI) RA CA FA DR		
8		6					
9	X	3		BH (BL) CB GR SA FN OT	PO GL (RI) RA CA FA DR		
10		4					
11		5		BH (BL) CB GR SA FN OT	PO GL (RI) RA CA FA DR		
12		6					
13		8		BH (BL) CB GR SA FN OT	PO GL (RI) RA CA FA DR		
14		8					
15		7		BH (BL) CB GR SA FN OT	PO GL (RI) RA CA FA DR		F1
16							
17				BH BL CB GR SA FN OT	PO GL RI RA CA FA DR		
18							
19				BH BL CB GR SA FN OT	PO GL RI RA CA FA DR		
20							

Flag Codes: K = no measurement made; U = suspect measurement; F1, F2, etc. = misc. flags assigned by each field crew . Explain all flags on Comment Form.

F1 = STATIONS 1-15 SPREAD THROUGH PROFILE

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PHab: THALWEG PROFILE FORM - RIVERS - 1

Figure 6-3. Thalweg Profile Form.

Reviewed by (initial) DKA

PHab: CHANNEL/RIPARIAN TRANSECT FORM - RIVERS

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT ☐1 ☒2 ☐3 ☐4

SITE ID: ORRV 98-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

TRANSECT (X): ☐A ☐B ☐C ☒D ☐E ☐F ☐G ☐H ☐I ☐J ☐K

"LITTORAL" Ssubstrate information

SLOPE DOM	SLOPE SEC	BOTTOM DOM	BOTTOM SEC	CLASS	BOTTOM SUBSTRATE FROM (X ONE): ☐ JUDGEMENT -OR- ☒ OBS. @ 5 LITTORAL DEPTHS
RS	RS	RS	RS		RS = BEDROCK (SMOOTH) • (LARGER THAN A CAR)
RR	RR	RR	RR		RR = BEDROCK (ROUGH) • (LARGER THAN A CAR)
DL	(DL)	DL	DL		DL = BOULDER (250 TO 4000 MM) • (BASKETBALL TO CAR)
CB	CB	(CB)	CB		CB = COBBLE (64 TO 250 MM) • (TENNIS BALL TO BASKETBALL)
(GC)	GC	GC	(GC)		GC = COARSE GRAVEL (16 TO 64 MM) • (MARBLE TO TENNIS BALL)
GF	GF	GF	GF		GF = FINE GRAVEL (2 TO 16 MM) • (LADYBUG TO MARBLE)
SA	SA	SA	SA		SA = SAND (0.06 TO 2MM) • (GRITTY - UP TO LADYBUG SIZE)
FN	FN	FN	FN		FN = SILT/CLAY/MUCK • (NOT GRITTY)
HP	HP	HP	HP		HP = HARDPAN • (FIRM, CONSOLIDATED FINE SUBSTRATE)
WD	WD	WD	WD		WD = WOOD • (ANY SIZE)
OT	OT	OT	OT		OT = OTHER (COMMENT)

SLOPE / BEARING / DISTANCE

INTENDED TRANSECT SPACING xxx (m) → 400

SLOPE xx.x %	BEARING 0 - 360°	DISTANCE xxx (m)	FLAG
0.75	183	450	F1

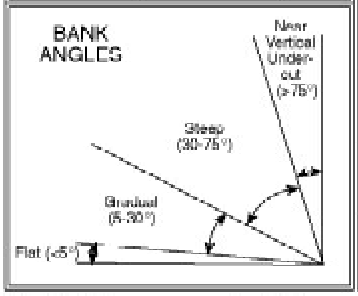
CANOPY DENSITY @ BANK
- DENSIOMETER (0 TO 17 MAX)

UP	0	DOWN	1
LEFT	8	RIGHT	0
FLAG			

BANK CHARACTERISTICS

CIRCLE ONE	ANGLE
V	Near Vertical/Undercut (>75°)
(S)	Steep (30 - 75°)
G	Gradual (5 - 30°)
F	Flat (< 5°)

	XXX (m)	FLAG
WETTED WIDTH	47	
BAR WIDTH	0	
BANKFULL WIDTH	75	
BANKFULL HT.	0.9	
INCISED HT.		K



LARGE WOODY DEBRIS in Wet Channel (10 x 20m Plot)

DIAMETER LARGE END	WOOD ALL/PART IN WETTED CHANNEL: LENGTH 5 - 15 m	15 - 30 m	> 30 m
0.3 - 0.6 m	0	0	0
0.6 - 0.8 m	0	0	0
0.8 - 1.0 m	0	0	0
> 1.0 m	0	0	0
DRY OUT ALL/PART IN BANKFULL CHANNEL			
0.3 - 0.6 m	0	0	0
0.6 - 0.8 m	0	0	0
0.8 - 1.0 m	0	0	0
> 1.0 m	0	0	0

DEPTH

SONAR (ft) xx	POLE (m) x.x
	0.4
	0.6
	0.6
	0.6
	0.6

IN SITU WATER MEASUREMENTS

WATER TEMPERATURE °C (xxxx)
15.9
CONDUCTIVITY µS/CM (xxxx)
105

FLAG	COMMENTS
	NO BIG TREES
F1 = RAPID :	
	OVERSHOT
K = NO TERRACE	

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

Rev. 05/29/98 (_rvphc_.98)

PHab: CHANNEL/RIPARIAN TRANSECT FORM - RIVERS - 1

Figure 6-4. Channel/Riparian transect form - page 1 (front side).

interval between thalweg profile measurements, measure the wetted channel width with a laser range finder at several locations near the upstream end of the reach and multiply it by 40 (100) to set the river sample reach length. Then divide that reach length by 100 (or 200) to set the thalweg increment distance. Following these guidelines, you will be making 100 or 200 evenly-spaced thalweg profile measurements, 10 or 20 between each detailed channel cross section where littoral/riparian observations are made. The number and spacing of measurements are as follows for the two different sample reach lengths:

	40 Ch-W		100 Ch-W	
	number	spacing	number	spacing
Transects and Riparian Plots	10	4 Ch-W	10	10 Ch-W
Thalweg Depth measurements	100	0.4 Ch-W	200	0.5 Ch-W
Thalweg Substrate, Habitat Class	100	0.4 Ch-W	100	1.0 Ch-W

6.3 Logistics, Work Flow, and Defining Sample Locations

The two-person habitat assessment team uses the most nimble of the selection of watercraft judged capable of navigating the river reach. In a single midstream float down the 40 or 100 Channel-width reach, the team accomplishes a reconnaissance, a sonar/pole depth profile, and a pole-drag to tally snags and characterize mid-channel substrate. The float is interrupted by stops at 11 transect locations for littoral/riparian observations. They determine (and mark -- optional) the position of each successive downstream transect using a laser range finder to measure out and mentally note each new location 4 (or 10) channel-width's distance from the preceding transect immediately upstream. The crew then floats downstream along the thalweg to the new transect location, making thalweg profile measurements and observations at 10 (or 20) evenly-spaced increments along the way. When they reach the new downstream transect location, they stop to do cross-section, littoral, and riparian measurements. Equipping the boat with a bow or stern anchor to stop at transect locations can greatly ease the shore marking operation and shoreline measurement activities. In addition, while they are stopped at a cross-section station, the crew can fill out the habitat "typing" entries retrospectively and prospectively for the portion of the stream distance that is visible up- and downstream. They can also record reconnaissance and safety notes at this time. While stopped at the transect location, the crew makes the prescribed measurements and observations, collects biological samples, backsites slope and bearing towards the previous upstream transect, and sets or mentally notes eye-level flags or reference points on shore for subsequent backsites. The habitat crew also assists the electrofishing boat crew over jams and helps to conduct shuttles (this can take considerable time where put-ins and take-outs are distant).

6.4 Reconnaissance and Reach Marking

The purpose of the reconnaissance is to locate (and optionally mark) the reach sampling location and to inform the second boat of the route, craft, and safety precautions needed during its subsequent electrofishing activities. After finding adequate put-in and take-out locations, the team may opt to mark the upstream end of the sample reach end with

colored flagging. Based on several channel width measurements using a laser range finder, they determine the sample reach length (40 x or 100 x Channel Width), the transect spacing (4 x or 10 x Channel Width) and thalweg sampling interval (0.5 x Channel Width). As the crew floats downstream, they stop (and optionally flag) 11 transect locations along the riverbank in the process of carrying out slope, bearing, and distance backsites. As the team floats downstream, they may choose and communicate to the electrofishing crew the most practical path to be used when fishing with a less maneuverable boat, taking into consideration multiple channels, blind channels, backwaters, alcoves, impassible riffles, rapids, jams, and hazards such as dams, bridges and power lines. They determine if and where tracking or portages are necessary.

6.5 Thalweg Profile

"Thalweg" refers to the flow path of the deepest water in a river channel. The thalweg profile is a longitudinal survey of maximum depth and several other selected characteristics at 100 (or 200) near-equally spaced points along the centerline of the river between the two ends of the river reach (Figure 6-1). For practical reasons, field crews will approximate a thalweg profile by sounding along the river course that they judge is deepest, but also safely navigable. Data from the thalweg profile allows calculation of indices of residual pool volume, river size, channel complexity, and the relative proportions of habitat types such as riffles and pools. The procedure for obtaining thalweg profile measurements is presented in Table 6-2. Record data on the Thalweg Profile Form as shown in Figure 6-3.

6.5.1 Thalweg Depth Profile

A thalweg depth profile of the entire 40 or 100 Channel-width reach shall be approximated by a sonar or sounding rod profile of depth while floating downstream along the deepest part of the channel (or the navigable or mid-channel path). In the absence of a recording fathometer (sonar depth sounder with strip-chart output or electronic data recorder), the crew records depths at frequent, relatively evenly-spaced downstream intervals while observing a sonar display and holding a surveyor's rod off the side of the boat (see subsection 6.5.2, below). The sonar screen is mounted so that the crew member can read depths on the sonar and the rod at the same time. The sonar sensor may need to be mounted at the opposite end of the boat to avoid mistaking the rod's echo for the bottom, though using a narrow beam (16 degree) Sonar transducer minimizes this problem. It is surprisingly easy to hold the sounding rod vertical when you are going at the same speed as the water. In our river trials, one measurement every half-channel-width (10 to 15 m) in current moving at about 0.5 m/s resulted in one measurement every 20 to 30 seconds. To facilitate accomplishing this work fast enough, the field form only requires "checks" for any observations other than depth measurements. To speed operations further, it may also be advantageous to mount a bracket on the boat to hold the clipboard.

6.5.2 Pole Drag for Snags and Substrate Characteristics

The procedure for obtaining pole drags for snags and substrate characteristics is presented in Table 6-2. While floating downstream, one crew member holds a calibrated PVC sounding tube or fiberglass surveying rod down vertically from the gunwale of the boat, dragging it lightly on the bottom to simultaneously "feel" the substrate, detect

snags, and measure depth with the aid of sonar. The number of large snags hit by this rod shall be recorded as an index of fish cover complexity (modification of Bain's "snag drag"). While dragging the sounding rod along the bottom, the crew member shall record the dominant substrate type sensed by dragging the rod along the bottom (bedrock/ hardpan, boulder, cobble, gravel, sand, silt & finer) (Figure 6-3). In shallow, "wild," fast-water situations, where pole-dragging might be hazardous, crews will estimate bottom conditions the best they can visually and by using paddles and oars. If unavoidable, suspend measurements until out of whitewater situations, but make notes and appropriately flag observations concerning your best judgements of depth and substrate.

6.5.3 Channel Habitat Classification

The crew will classify and record the channel habitat types shown in Figure 6-3 (fall, cascade, rapid, riffle, glide, pool, dry) and check presence of off-channel and backwater habitat at a spatial resolution of about 0.4 channel-widths on a 40 Channel-width reach. On a 100 Channel-width reach habitat classifications are made every 1.0 channel-widths and off-channel and backwater habitat presence is checked every 0.5 channel-width distance -- the same interval as thalweg depths. The resulting database of traditional visual habitat classifications will provide a bridge of common understanding with other studies. The procedures for classifying channel habitat are presented in Table 6-2. The designation of side channels, backwaters and other off-channel areas is independent of the main-channel habitat type. Main channel habitat units must meet a minimum size criteria in addition to the qualitative criteria listed in Table 6-3. Before being considered large enough to be identified as a channel-unit scale habitat feature, the unit should be at least as long as the channel is wide. For instance, if there is a small, deep (pool-like) area at the thalweg within a large riffle area, don't record it as a pool unless it occupies an area about as wide or long as the channel is wide.

Mid-Channel Bars, Islands, and Side Channels pose some problems for the sampler conducting a thalweg profile and necessitate some guidance. Mid-channel bars are defined here as channel features below the bankfull flow level that are dry during baseflow conditions (see Section 6.6.4 for definition of bankfull channel). Islands are channel features that are dry even when the river is at bankfull flow. If a mid-channel feature is as high as the surrounding flood plain, it is considered an island. Both mid-channel bars and islands cause the river to split into side channels. When a bar or island is encountered along the thalweg profile, choose to navigate and survey the channel that carries the most flow.

When side channels are present, the comments column of the Thalweg Profile form should reflect their presence by checking the "Off-Channel" column. These checkmarks will begin at the point of divergence from the main channel, continuing downstream to the point of where the side channel converges with the main channel. In the case of a slough or alcove, the "off-channel" checkmarks should continue from the point of divergence.

6.6 Channel Margin ("Littoral") And Riparian Measurements

Components of this section include slope and bearing, channel margin depth and sub-

Table 6-2. Thalweg Profile Procedure.

1. Determine the interval between measurement stations based on the wetted width used to determine the length of the sampling reach.
2. Complete the header information on the Thalweg Profile Form, noting the transect pair (upstream to downstream).
3. Begin at the upstream transect (station "1" of "20" or station "1" of "10").

Thalweg Depth Profile

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

b) A depth recording approximately every 0.4 (or 0.5) channel-width distance is required, yielding 10 (or 20) measurements between channel/riparian cross-section transects.

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

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

Pole Drag for Snags and Substrate Characteristics

a) From the gunwale of the boat, hold a fiberglass surveying rod or calibrated PVC sounding tube down vertically into the water.
b) Lightly drag the rod on the river bottom to "feel" the substrate and detect snags.
c) Observations are taken at half the frequency as depth measurements (i.e., at every other depth measurement point on 100 Channel-Width reaches).
d) Record the number of snags hit by the rod and the dominant substrate type sensed by dragging the rod along the bottom.
e) On the Thalweg Profile Form, circle the appropriate "SUBSTRATE" type and tally the number of "SNAGS".

Channel Habitat Classification

a) Classify and record the channel habitat type at increments of every 1.0 channel width.
b) Check for off-channel and backwater habitat at increments of every 0.4 (or 0.5) channel width.
c) If channel is split by a bar or island, navigate and survey the channel with the most discharge.
d) When a side channel is encountered, check the "OFF-CHANNEL" column beginning with the point of divergence from the main channel, continuing downriver until the side channel converges with the main channel.
e) On the Thalweg Profile Form, circle the appropriate "CHANNEL HABITAT" and check the off-channel column as described in (d) above.

4. Proceed downriver to the next station ("2"), and repeat the above procedures.
5. Repeat the above procedures until you reach the next transect. Prepare a new Thalweg Profile Form, then repeat the above procedures for each of the reach segments, until you reach the downriver end of the sampling reach (Transect "K").

Table 6-3. Channel Unit Categories.

Channel Unit Habitat Classes[a]	
Class (Code)	Description
Pools (PO):	Still water, low velocity, smooth, glassy surface, usually deep compared to other parts of the channel:
Plunge Pool	Pool at base of plunging cascade or falls.
Trench Pool	Pool-like trench in the center of the stream
Lateral Scour Pool	Pool scoured along a bank.
Backwater Pool	Pool separated from main flow off the side of the channel.
Dam Pool	Pool formed by impoundment above dam or constriction.
Glide (GL)	Water moving slowly, with a smooth, unbroken surface. Low turbulence.
Riffle (RI)	Water moving, with small ripples, waves and eddies -- waves not breaking, surface tension not broken. Sound: "babbling", "gurgling".
Rapid (RA)	Water movement rapid and turbulent, surface with intermittent whitewater and 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
Off-Channel Areas	Side-channels, sloughs, backwaters, and alcoves that are separated from the main channel.

[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.

strate, large woody debris, bank angle and channel cross-section morphology, canopy cover, riparian vegetation structure, fish cover, and human influences. All measurements are recorded on the two-sided Channel/Riparian Transect Form (Figures 6-4 and 6-5).

6.6.1 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 ma-

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PHab: CHANNEL/RIPARIAN TRANSECT FORM - RIVERS (continued)

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT ☐0 ☒1 ☐2 ☐3

SITE ID: ORRV 98-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

TRANSSECT (X): ☐A ☐B ☐C ☒D ☐E ☐F ☐G ☐H ☐I ☐J ☐K

VISUAL RIPARIAN ESTIMATES	LEFT BANK	RIGHT BANK	FLAG
RIPARIAN VEGETATION COVER (10 m x 20 m Plot)	0 = ABSENT (0%) D = DECIDUOUS 1 = SPARSE (< 10%) C = CONIFEROUS 2 = MODERATE (10 - 40%) E = BROADLEAF EVERGREEN 3 = HEAVY (40 - 75%) M = MIXED 4 = VERY HEAVY (> 75%) N = NONE		
CANOPY (> 5 m HIGH)			
VEGETATION TYPE	D C E M (N)	D C E M (N)	
BIG TREES (TRUNK > 0.3 m DBH)	(0) 1 2 3 4	(0) 1 2 3 4	
SMALL TREES (TRUNK > 0.3 m DBH)	(0) 1 2 3 4	(0) 1 2 3 4	
UNDERSTORY (0.5 to 5 m HIGH)			
VEGETATION TYPE	(D) C E M N	D C E M (N)	
WOODY SHRUBS & SAPLINGS	0 1 (2) 3 4	(0) 1 2 3 4	
NON-WOODY HERBS, GRASSES & FORBS	0 (1) 2 3 4	(0) 1 2 3 4	
GROUND COVER (< 0.5 m HIGH)			
WOODY SHRUBS & SEEDLINGS	0 (1) 2 3 4	0 (1) 2 3 4	
NON-WOODY HERBS, GRASSES & FORBS	0 1 (3) 4	0 1 2 3 (4)	
BARREN, BARE, DRY OR DUFF	0 (1) 2 3 4	(0) 1 2 3 4	
HUMAN INFLUENCE (10 m x 20 m Plot)	0 = NOT PRESENT P = > 10 m O = WITHIN 10 m, B = ON BANK		FLAG
WALL/DIKE/REVETMENT/RIPRAP/DAM	(0) P O B	(0) P O B	
BUILDINGS	(0) P C B	(0) P C B	
PAVEMENT	(0) P C B	(0) P C B	
ROAD/RAILROAD	0 (P) C B	0 (P) C B	F2
PIPES (INLET/OUTLET)	(0) P C B	(0) P C B	
LANDFILL/TRASH	(0) P C B	(0) P C B	
PARK/LAWN	(0) P C B	(0) P C B	
ROW CROPS	(0) P O B	(0) P O B	
PASTURE/RANGE/HAY FIELD	(0) P O B	(0) P O B	
LOGGING OPERATIONS	(0) P C B	(0) P C B	
MINING ACTIVITY	(0) P C B	(0) P C B	

FISH COVER/ OTHER (10 m X 20 m Plot)	COVER IN CHANNEL 0 = ABSENT (0%) 1 = SPARSE (< 10%) 2 = MODERATE (10 - 40%) 3 = HEAVY (40 - 75%) 4 = VERY HEAVY (> 75%) CIRCLE ONE	FLAG
FILAMENTOUS ALGAE	(0) 1 2 3 4	
MACROPHYTES	0 (1) 2 3 4	
WOODY DEBRIS (> 0.3 m - BIG)	(0) 1 2 3 4	
BRUSH/WOODY DEBRIS (< 0.3 m - SMALL)	(0) 1 2 3 4	
OVERHANGING VEG. (≤ 1 m OF SURFACE)	(0) 1 2 3 4	
UNDERCUT BANKS	0 (1) 2 3 4	
BOULDERS/LEDGES	0 (1) 2 3 4	
ARTIFICIAL STRUCTURES	(0) 1 2 3 4	

CHANNEL CONSTRAINT

DISTANCE FROM SHORE TO RIPARIAN VEGETATION (XX) XXX ► 0

CIRCLE ONE	
(C)	CHANNEL IS CONSTRAINED.
U	CHANNEL IS UNCONSTRAINED IN BROAD VALLEY.
B	CHANNEL IS IN BROAD VALLEY BUT CONSTRAINED BY INCISION.
N	CHANNEL IS IN NARROW VALLEY BUT NOT VERY CONSTRAINED.

CIRCLE ONE	
(YES)	I COULD READILY SEE OVER THE BANK.
NO	I COULD NOT READILY SEE OVER THE BANK.

FLAG	COMMENTS (Additional space available on Side 1)
F2	RAILROAD GRADE

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

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PHab: CHANNEL/RIPARIAN TRANSECT FORM - RIVERS - 2

Figure 6-5. Channel/Riparian transect form - page 2 (back side).

jor stream classification variables, giving an indication of potential water velocities and stream power; both of 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), Robison and Kaufmann (1994), and Kaufmann et al., (1999), 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 (Subsection 6.5). 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 "backsiting" upstream from cross-section station B to A, C to B, D to C, etc., down to the 11th cross section (Figure 6-1). To measure the slope and bearing between adjacent stations, use an Abney Level (or clinometer), and a bearing compass following the procedure presented in Table 6-4. Record data for slope and bearing in the Slope/Bearing/Distance section of the Channel/Riparian Transect Form (Figure 6-4).

It may be necessary to set up intermediate slope and bearing stations between the normal 11 stations if you do not have direct line-of-site along (and within) the channel between stations. This can happen if brush is too heavy or if there are tight meander bends or sharp slope breaks. To backsite upstream from supplemental stations, treat them just as you do a normal transect location in steps 1 to 6 of Table 6-4. Record supplemental slope, bearing, and distance backsites sequentially in the spaces provided on the field form.

6.6.2 Channel Margin Depth and Substrate

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 (as well as other benthic fishes). 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.

Channel margin depths are measured along the designated shoreline at each transect within the 10m swath of the 20m channel margin length that is centered on the transect location. Dominant and sub-dominant bottom substrates are determined and recorded at 5 systematically-spaced locations that are located by eye within the 10m x 20m plot. These methods are an adaptation of those used by the U.S.EPA for evaluating littoral substrates in lakes (Kaufmann and Whittier 1997), where the substrate size may be visually assessed or estimated by "feel" using the surveyors rod or PVC sounding tube in deep, turbid water. The procedure for obtaining channel margin depth and substrate measurements is described in more detail in Table 6-5. Record these measurements on the Channel/Riparian Transect Form as shown in Figure 6-4.

Table 6-4. Procedure for Obtaining Slope and Bearing Data.

1. Set eye-level flagging at upstream transect: Place flagging or mentally note a landmark at a standardized eye level along the shoreline at Transect A while doing shoreline measurements. To accomplish this, sit in the boat with your clinometer or Abney level held against your measuring rod at a comfortable, standardized height above the water surface (or designated place on bottom of boat). This shall be the same height you plan to use for all slope backsites from downstream. Site towards the nearby bank with the clinometer or Abney level indicating 0% slope. Note the level on the object sited and place flagging on it (optional). Accuracy of the clinometer measurements can be checked occasionally against a surveyors level.
2. Using the laser rangefinder, determine and record the intended location and distance of the next downstream Transect.
3. Float downstream (doing your thalweg profile measurements at 10 or 20 increments) to Transect B, where the next channel/riparian station is located.
4. Measure (w/ laser rangefinder) and record the distance back to the flagged upstream transect. (Note that, because of hazards and maneuvering problems, this distance may unavoidably differ from the "intended transect spacing" that is set at 4 (or 10) times the wetted width in the near vicinity of the furthest upstream transect (A).
5. Backsite the river gradient: While at the bank at Transect B, hold your Abney or clinometer at the same level on your measuring rod that you used at the previous station when you set up the eye-level flagging. Site back upstream at your flagging at Station A; read and record **percent** Slope on the field form. Be careful, the clinometer reads both percent slope and degrees of the slope angle. **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."**
6. Backsite the compass bearing: From the bank at Station B, site back with your compass to the flagging you placed at Station A and record your compass bearing ("Azimuth"). It does not matter for these measurements whether or not you adjust your compass bearings for magnetic declination, but **it is important that you are consistent in the use of magnetic (unadjusted) or true (adjusted) bearings** throughout all the measurements you make on a given reach. Write on the field form which type of bearings you take. 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."
7. Repeat step 1, setting your eye-level flagging at Transect B before floating down to a new downstream transect. Then repeat steps 2 through 7.

Again adapting methods developed for lake shorelines by Kaufmann and Whittier (1997), identify the dominant and subdominant substrate present along a shoreline swath 20 meters long and 1 meter back from the waterline. The substrate size class choices are as shown in Table 6-5.

6.6.3 Large Woody Debris

Methods for tallying large woody debris (LWD) are adapted from those described by Kaufmann and Robison (1998). This component of the EMAP Physical Habitat protocol allows estimates of the number, size, and total volume of large woody debris within the river reach. LWD is defined here as woody material with small end diameter of at least 30 cm (1ft) and length of at least 5 m (15 ft). These size criteria are larger than those used by Kaufmann and Robison (1998) in wadeable streams because of the lesser role that small wood plays in controlling velocity and morphology of larger rivers.

The procedure for tallying LWD is presented in Table 6-6. The tally includes all pieces of LWD that are at least partially in the baseflow channel (Wetted Channel). Sepa-

Table 6-5. Channel Margin Depth and Substrate Procedure.

1. If not already done, fill in the header information on page 1 of a Channel/Riparian Transect Form. Be sure to indicate the letter designating the transect location.

2. Measure depth and observe bottom substrates within a 10m swath along the 20m of the channel margin that is centered on each transect location.

3. Determine and record the depth and the dominant and subdominant substrate size class at 5 systematically-spaced locations estimated by eye within this 10m x 20m plot and 1m back from the waterline. **If the substrate particle is "artificial" (e.g. concrete or asphalt), choose the appropriate size class, flag the observation and note that it is artificial in the comment space.**

Code	Size Class	Size Range (mm)	Description
RS	Bedrock (Smooth)	>4000	Smooth surface rock bigger than a car
RR	Bedrock (Rough)	>4000	Rough surface rock bigger than a car
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)

4. On page 1 of the Channel/Riparian Transect Form, circle the appropriate shore and bottom substrate type and record the depth measurements ("SONAR" or "POLE" columns).

5. Repeat Steps 1 through 4 at each new cross section transect.

rately tally wood that is presently dry but contained within the "Bankfull" or active channel (flood channel up to bankfull stage). Include wood that spans above the active channel or spanning above the active channel with the "Dry but within Bankfull" category. For each tally (Wetted Channel and Dry but within Bankfull), the field form (Figure 6-4) 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. Woody debris is not tallied in the area between channel cross sections, but the presence of large debris dams and accumulations should be mapped and noted in the comments.

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 classes on the field form (Figure 6-4) refer to the large end diameter. The diameter classes are 0.3m to <0.6m, 0.6m to <0.8m, and 0.8m to <1.0m and >1.0m. The length classes are 5m to <15m, 15m to <30m, and >30m. Sometimes LWD is not cylindrical, so it has no clear "diameter". In these cases visually estimate what the diameter would be for a piece of wood with circular cross section that would have the same volume. When evaluating length, include only the part of the LWD piece that has a diameter greater than 0.3m (1 ft).

Table 6-6. Procedure for Tallying Large Woody Debris.

Note: Tally pieces of large woody debris (LWD) within the 11 transects of the river reach at the same time the shoreline measurements are being determined. Include all pieces whose large end is located within the transect plot in the tally.

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

Count each of the LWD pieces as one tally entry and include the whole piece when assessing dimensions, even if part of it is outside of the bankfull channel. If you encounter massive, complex debris jams, estimate their length, width, and height. Also estimate the diameter and length of large "key" pieces and judge the average diameter and length of the other pieces making up the jam. Record this information in the comments section of the form.

6.6.4 Bank Angle and Channel Cross-Section Morphology

Undercut, vertical, steep, and gradual bank angles are visually estimated as defined on the field form (Figure 6-4). Observations are made from the wetted channel margin up 5 m (a canoe's length) into the bankfull channel margin on the previously chosen side of the stream.

The channel dimensions to be measured or estimated are the wetted width, mid-channel bar width, bankfull height and width, the amount of incision, and the degree of channel constraint. These shall be assessed for the whole channel (left and right banks) at each of the 11 cross section transects. Each are recorded on the Channel/Riparian Transect Form (Figure 6-4). The procedure for obtaining bank angle and channel cross-section morphology measurements is presented in Table 6-7.

Table 6-7. Procedure for Bank Angle and Channel Cross-Section.

1. Visually estimate the bank angle (undercut, vertical, steep, gradual), as defined on the field form. Bank angle observations refer to the area from the wetted channel margin up 5 m (a canoe's length) into the bankfull channel margin on the **previously chosen side of the river**. Circle the range within which the observed band angle falls on the "Bank CHARACTERISTIC" section of the Channel/ Riparian Transect Field Form.
2. With a laser rangefinder at a cross-section transect, measure and record the **wetted width** value in the "Wetted Width" field in the bank characteristics section of the field data form. Also determine the **bankfull channel width** and the **width of exposed mid-channel bars (if present)** with the laser rangefinder and surveyor's rod. Record these values in the "Bank CHARACTERISTIC" section of the field data form.
3. To estimate bankfull height, hold the surveyor's rod vertical, with its base planted at the water's edge. Using the rod as a guide while examining both banks, estimate (by eye) 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 river sediments to terrestrial vegetation.
   - A transition from sorted river sediments to unsorted terrestrial soils.
   - Transition from bare rock to 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.
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 the 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 Characteristic section on the field data form.
5. Repeat Steps 1 through 4 at each cross-section transect. Record data for each transect on a separate field data form.

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

Bankfull height and width shall be estimated with the aid of the surveyor's rod and laser range finder. The "bankfull" or "active" channel is defined as the channel that is filled by moderate sized flood events that fill the channel to its flood banks. Measure bankfull width over and across mid-channel bars. Bankfull flows typically recur every 1 to 2 years and do not generally overtop the channel banks to inundate the valley floodplain. They are believed to be largely responsible for the observed channel dimensions in most

rivers and streams. If the channel is not greatly incised, bankfull channel height and the amount of incision will be the same. However, if the channel is incised greatly, the bankfull level will be below the level of the first terrace of the valley floodplain, making "Bankfull Height" smaller than "Incision" (Figure 6-6). You will need to look for evidence of recent flows (within about 1 year) to distinguish bankfull and incision heights, though recent flooding of extraordinary magnitude may be misleading.

Estimating 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 many types of terrestrial vegetation, the bankfull channel may be evident by a transition from exposed river sediments and water-loving plants to upland ter-

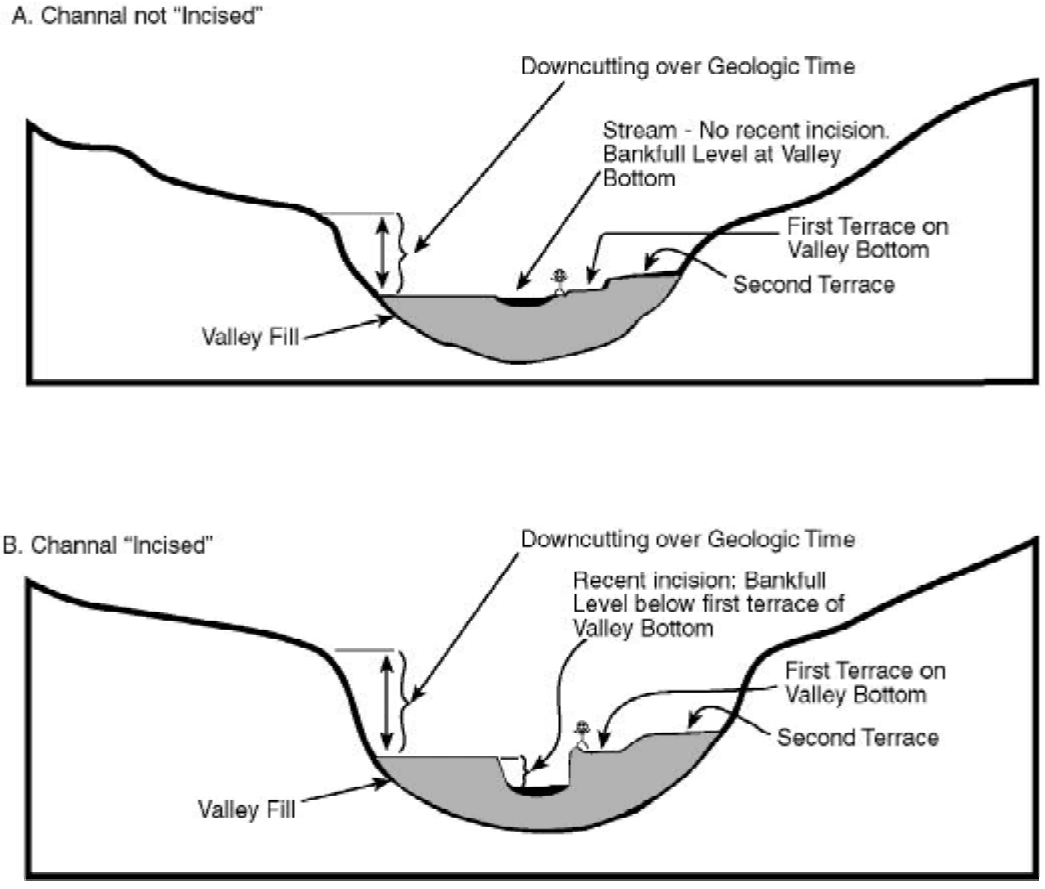

Figure 6-6. Schematic showing bankfull channel and incision for channels. (A) not recently incised, and (B) recently incised into valley bottom. Note level of bankfull stage relative to elevation of first terrace on valley bottom (Stick figure included for scale).

restrial vegetation. Similarly, it may be identified by noting where moss growth on rocks along the banks has been removed by flooding. The bankfull flow level may also be seen by the presence of drift material caught on overhanging vegetation.

As described in Table 6-7 and shown in Figure 6-6, examine both banks and estimate (by eye) the amount of channel incision from the water surface to the elevation of the first terrace of the valley floodplain. 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 make a more accurate determination of channel downcutting. Finally, assess the degree of river channel constraint by answering the four questions on the form (Figure 6-5) regarding the relationships among channel incision, valley sideslope, and width of the valley floodplain.

6.6.5 Canopy Cover (Densiometer)

Riparian canopy cover over a river is important not only for its role in moderating water temperatures through shading, but also as riparian wildlife habitat, and 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 river organisms and structure to create and maintain complex channel habitat.

Vegetative cover over the river margin shall be measured at the chosen bank at each of the 11 transect locations (A-K). This measurement employs the Convex Spherical Densiometer, model B (Lemmon, 1957). The densiometer must be taped exactly as shown in Figure 6-7 to limit the number of square grid intersections to 17. Densiometer readings can range from 0 (no canopy cover) to 17 (maximum canopy cover). Four measurements are obtained at each cross-section transect (upriver, downriver, left, and right). Concentrate on the 17 points of grid intersection on the densiometer. 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. The measure to be recorded on the form is the count (from 0 to 17) of all the intersections that have vegetation covering them. Therefore, a higher number indicates greater canopy extent and density. In making this measurement, it is important that the densiometer be leveled using the bubble level (Figure 6-7).

The procedure for obtaining canopy cover data is presented in Table 6-8. These bank densiometer readings complement your visual estimates of vegetation structure and cover within the riparian zone (Section 6.6.6). For each of the four directions, count the number of covered densiometer intersection points. Record these counts in the "Canopy Density @ Bank" section of the Channel/Riparian Transect Form as shown in Figure 6-4.

6.6.6 Riparian Vegetation Structure

The previous section (6.6.5) described methods for quantifying the cover of canopy over the river margin. The following visual estimation procedures, adapted from Kaufmann and Robison (1998), are a semi-quantitative evaluation of riparian vegetation structure, the type and amount of different types of riparian vegetation. These field characterizations shall be used to supplement interpretations of riparian vegetation from aerial photos and satellite imagery. Together, they

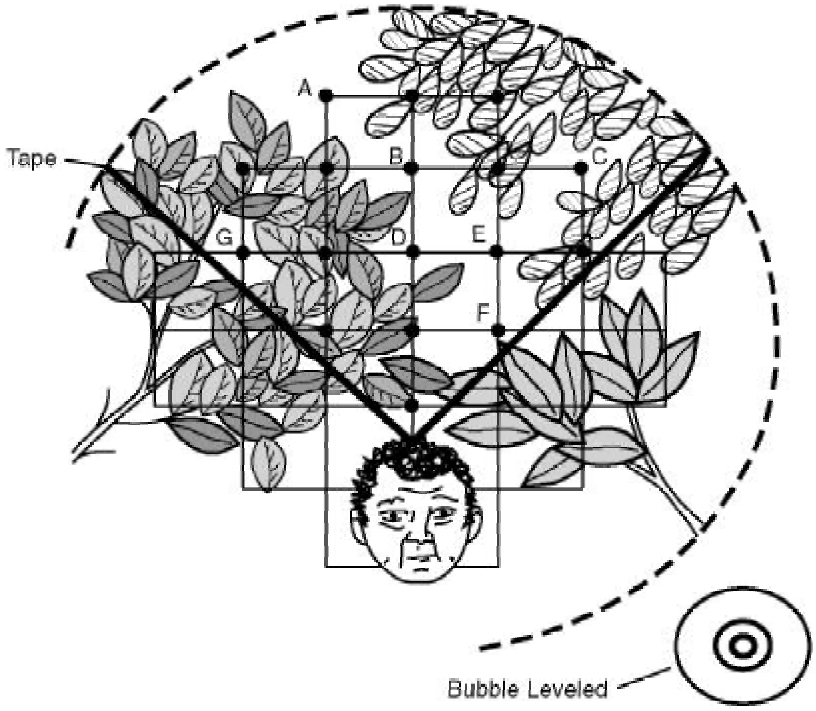

Figure 6-7. Schematic of modified convex spherical canopy densiometer (From Mulvey et al., 1992). In this example, 10 of the 17 intersections show canopy cover, giving a densiometer reading of 10. Note proper positioning with the bubble leveled and face reflected at the apex of the "V".

Table 6-8. Procedure for Canopy Cover Measurements.

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

are used to evaluate the health and level of disturbance of the river/riparian corridor. They also indicate the present and future potential for various types of organic inputs and shading. The cover and structure of riparian vegetation is estimated in three riparian layers within 10m x 20m plots along the river shoreline that are centered on the transect location with boundaries estimated by eye. As employed by Allen-Gill (unpublished manuscript), these plots shall be set back from the channel so that they describe vegetation above bankfull flow. As a result, gravel bars within the bankfull channel are not included in the vegetation plot (Figure 6-2).

Observations to assess riparian vegetation apply to the riparian area upstream 10 meters and downstream 10 meters from each of the 11 cross-section stations (Figure 6-2). They include the visible area from the river

bankfull margin back a distance of 10m (30 ft) shoreward from both the left and right banks, creating a 10m X 20m riparian plot on each side of the river (Figure 6-2). The riparian plot dimensions are estimated, not measured. On steeply sloping channel margins, the 10m X 20m 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 shall be for each side of the channel, not the bar. If an island obscures the far bank of the main channel, assess riparian vegetation on the bank of the island.

Table 6-9 presents the procedure for characterizing riparian vegetation structure and composition. Figure 6-5 illustrates how measurement data are recorded in the "Visual Riparian Estimates" section of the field form. Conceptually divide the riparian vegetation into three layers: a CANOPY LAYER (>5m high), an UNDERSTORY (0.5 to 5m high), and a GROUND COVER layer (<0.5 high). Note that several vegetation types (eg. 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 (eg. barren ground and duff, which includes fallen leaves, needles and twigs).

Before estimating the areal coverage of the vegetation layers, record the type of vegetation (**D**eciduous, **C**oniferous, Broadleaf **E**vergreen **M**ixed, or **N**one) in each of the two taller layers (Canopy and Understory). Consider the layer "Mixed" if more than 10% of the areal coverage is made up of the alternate vegetation type.

You will estimate the areal cover separately in each of the three vegetation layers. Note that the areal cover can be thought of as the amount of shadow cast by a particular layer alone when the sun is directly overhead. The maximum cover in each layer is 100%, so the sum of the areal covers for the combined three layers could add up to 300%. The four entry choices for areal cover within each of the three vegetation layers are "0" (absent: zero cover), "1" (sparse: <10%), "2" (moderate: 10-40%), "3" (heavy: 40-75%), and "4" (very heavy: >75%). These ranges of percentage areal cover corresponding to each of these codes are also shown on the Field Form. 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" with all the remaining classes either moderate ("2"), sparse ("1") or absent ("0"). Two heavy classes with 40-75% cover can both be rated "3".

As an additional assessment of the "old growth" character of riparian zones, search for the largest riparian tree visible on either side of the river from the littoral-riparian station. Identify if possible the species or the taxonomic group of this tree and estimate its height, diameter (Dbh), and distance from the wetted river margin.

6.6.7 Fish Cover, Algae, Aquatic Macrophytes

This portion of the EMAP physical habitat protocol is a visual estimation procedure modified from methods developed for lake shorelines (Kaufmann and Whittier 1997) and for wadeable streams (Kaufmann and Robison 1998). The aim is to evaluate, semi-quantitatively, the type and amount of important types of cover for fish and macroinvertebrates. Over

Table 6-9. Procedure For Characterizing Riparian Vegetation Structure.

1. Anchor or tie up at the river margin at a cross-section transect; then make the following observations to characterize riparian vegetation structure.

2. Estimate the distance from the shore to the riparian vegetation plot; record it just below the title "Channel Constraint" on the field form.

3. Facing the left bank (left as you face downstream), estimate a distance of 10 m back into the riparian vegetation, beginning at the bankfull channel margin. Estimate the cover and structure of riparian vegetation in 3 riparian layers along the river shoreline within an estimated 10m x 20m plot centered on the transect, and beginning at the bankfull river margin along the river shoreline.

    - On steeply-sloping channel margins, estimate the distance into the riparian zone as if it were projected down from an aerial view.

4. Within this 10 m × 20 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).

5. Within this 10 m × 20 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 and Thalweg Profile Form.

6. Determine separately the areal cover class of large trees (> 0.3 m [1 ft] diameter at breast height [DBH]) and small trees (< 0.3 m DBH) within the canopy layer. Estimate areal cover as the amount of shadow that would be cast by a particular layer alone if the sun were directly overhead. Record the appropriate cover class on the field data form ("0"=absent: zero cover, "1"=sparse: <10%, "2"=moderate: 10-40%, "3"=heavy: 40-75%, or "4"=very heavy: >75%).

7. Look at the UNDERSTORY layer (vegetation between 0.5 and 5 m high). Determine the dominant vegetation type for the understory layer as described in Step 5 for the canopy layer.

8. Determine the areal cover class for woody shrubs and saplings separately from non-woody vegetation within the understory, as described in Step 6 for the canopy layer.

9. Look at the GROUND COVER layer (vegetation < 0.5 m high). Determine the areal cover class for woody shrubs and seedlings, non-woody vegetation, and the amount of bare ground present as described in Step 6 for large canopy trees.

10. Repeat Steps 1 through 9 for the opposite bank.

11. Repeat Steps 1 through 10 for all cross-section transects, using a separate field data form for each transect.

a defined length and distance from shore at 11 systematically spaced plot locations, crews shall estimate by eye and by sounding the proportional cover of fish cover features and trophic level indicators including large woody debris, rootwads and snags, brush, undercut banks, overhanging vegetation, rock ledges, aquatic macrophytes, filamentous algae, and artificial structures. 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 fish cover is outlined in Table 6-10. Data are recorded in the "Fish Cover/ Other" section of the Channel/Riparian Transect Form as shown in Figure 6-5. Crews will estimate the areal cover of all of the fish cover and other listed features that are in the water and on the banks within the 10m x 20m plot (refer to Figure 6-2).

Observations to assess fish cover and several other in-channel features apply to a 10 m x 20 m inundated area adjacent to the selected bank extending 10 m out from the channel margin, and then upstream 10 m and downstream 10 m from each of the 11 transect cross-sections (Figure 6-2). These plot dimensions are estimated by eye. The ranges of percentage areal cover corresponding to each of these codes are the same as for riparian vegetation cover (Section 6.6.6) and are also shown on the Field Form.

Table 6-10. Procedure For Estimating Fish Cover.

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

Filamentous algae pertains to long streaming algae that often occur in slow moving waters. Aquatic macrophytes are water loving plants in the river, including mosses, that could provide cover for fish or macroinvertebrates. If the river channel contains live wetland grasses, include these as macrophytes. Woody debris includes the larger pieces of wood that can provide cover and influence river morphology (i.e., those pieces that would be included in the large woody debris tally [Section 6.6.3]). Brush/ woody debris pertains to the smaller wood that primarily affects cover but not morphology. The entry for trees or brush within one meter above the water surface is the amount of brush, twigs, small debris etc. that is not in the water but is close to the river and provides cover. Boulders are typically basketball to car sized particles. Many streams contain artificial structures designed for fish habitat enhancement. Streams may also have in-channel structures discarded (e.g. cars or tires) or purposefully placed for diversion, impoundment, channel stabilization, or other purposes. Record the cover of these structures on the form.

6.6.8 Human Influences

Field characterization of the presence and proximity of various important types of human activities, disturbances, and land use in the river riparian area is adapted from methods developed by Kaufmann and Robison (1998) for wadeable streams. This information shall be used in combination with riparian and watershed landuse information from aerial photos and satellite imagery to assess the potential degree of disturbance of the sample river reaches.

For the left and right banks at each of the 11 detailed Channel/Riparian Cross-Sec-

tions, evaluate the presence/absence and the proximity of 11 categories of human influences outlined in Table 6-11. Confine your observations to the river and riparian area within 10m upstream and 10m downstream from the cross-section transect (Figure 6-2). Four proximity classes are used: On the riverbank within 10m upriver or downriver of the cross-section transect, present within the 10m x 20m riparian plot, present outside of the riparian plot, and not present. Record human influences on the Channel/Riparian Transect Form (Figure 6-5).

You may mark "P" more than once for the same human influence observed outside of more than one riparian observation plot (e.g. at both Transect D and E). The rule is that you count human disturbance items as often as you see them, BUT NOT IF you have to site through a previously counted transect or its 10x20m riparian plot.

6.7 Summary of Workflow

Table 6-12 lists the activities performed at and between each transect for the physical habitat characterization. The activities are performed along the chosen river bank and mid-channel (thalweg profile).

6.8 Equipment and Supplies

Figure 6-8 lists the equipment and supplies required to conduct all the activities described for characterizing physical habitat. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the river. Use this checklist to ensure that equipment and supplies are organized and available at

Table 6-11. Procedure for Estimating Human Influence.

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

Table 6-12. Summary of Workflow - River Physical Habitat Characterization.

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

1. Move boat in a "loop" within 10 x 20 meter littoral plot, measuring five littoral depths and probing substrate.
2. Estimate dominant and subdominant littoral substrate, based on probing the five locations.
3. Estimate areal cover of fish concealment features in 10 x 20 meter littoral plot.
4. Tally LWD within or partially within the 10 x 20 meter littoral plot.
5. Measure water conductivity and temperature.
6. Do densiometer measurements at bank (facing upstream, downstream, left, right).
7. Choose bank angle class, estimate bankfull height, width and channel incision. (Note that width and incision estimates incorporate both left and right banks.).
8. Tally LWD entirely out of water but at least partially within the bankfull channel.
9. Estimate and record distance to riparian vegetation on the chosen bank.
10. Make visual riparian vegetation cover estimates for the 10 x 20 meter riparian plot on both sides of the channel. (Note that riparian plot starts at bankfull and continues back 10m away from the bankfull line).
11. Identify species, height, Dbh, and distance from riverbank of largest riparian tree within your vision.
12. Make visual human disturbance tally. It has the same plot dimensions as the riparian vegetation -- except if a disturbance item is observed in the river or within the bankfull channel, then the proximity code is "B", the closest rating. Disturbances within the plot get a rating of "C"; those visible beyond the plot are rated "P".
13. Siting clinometer level (0%) towards the near or far bank at the current transect, mark or remember an eye-level point to which you will be siting when backsiting from the next downstream transect.
14. Get out far enough from the bank so you can see downstream. Then use the laser rangefinder to site and record the distance to the intended position of the next downstream transect.

B.Thalweg Profile:

1. As soon as you get out from the bank after doing transect activities, take the first of 20 thalweg depth measurements and substrate/snag probes using sonar and pole -- also classify habitat type.
2. Estimate thalweg measurement distance increments by keeping track of boat lengths or channel-width distances traversed; each increment is 1/10th (or 1/20th) the distance between transects.
3. At the 20th thalweg measurement location, you are one increment upstream of the next transect. Backsite compass bearing mid-channel, then measure the distance and % slope back to your visual "mark" on the bank at the previous transect.

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

the river 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, fiberglass, metric scale, 7.5m extended)	
1	Clinometer (or Abney level) with percent and degree scales.	
1	Convex spherical canopy densiometer (Lemmon Model B), modified with taped "V"	
1	Bearing compass (Backpacking type)	
1 roll ea.	Colored surveyor's plastic flagging (2 colors)	
2	Covered clipboards (lightweight, with strap or lanyard to hang around neck)	
	Soft (#2) lead pencils (mechanical are acceptable)	
2 pair	Chest waders with felt-soled boots for safety and speed if waders are the neoprene "stocking" type	
1	Camera - waterproof 35mm with standard and wide angle lens	
	Film - 35mm color slide film, ASA 400 and 100	
1	Fiberglass Tape and reel (50m metric) with good hand crank and handle	
1	SONAR depth sounder - narrow beam (16 degrees)	
1	Laser rangefinder - 400 ft. distance range - and clear waterproof bag	
11 plus extras	Channel/Riparian Transect Forms	
11 plus extras	Thalweg Profile Forms	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for physical habitat characterization	

Figure 6-8. Checklist of equipment and supplies for physical habitat

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Section 7
Periphyton

Brian H. Hill[1] and Alan T. Herlihy[2]

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.

Periphyton samples are collected at the near-shore shallows when stopped at each of the cross-section transects (transects "A" through "K") established within the sampling reach (Section 4). Periphyton samples are collected at each transect at the same time as sediment samples (Section 8) and benthic macroinvertebrate samples (Section 9). One composite "index" sample of periphyton is prepared for each river site. At the completion of the day's sampling activities, but before leaving the river, four types of laboratory samples are prepared from the composite periphyton sample.

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7.1 Sample Collection

The general scheme for collecting periphyton samples from the sampling reach at each river is illustrated in Figure 7-1. At each transect, samples are collected from the shoreline assigned during the layout of the reach (Section 4). The substrate selected for sampling should be collected from a depth no deeper than can be reached by submerging your arm to mid-bicep depth. If a sample cannot be collected because the location is too deep, skip the transect. The procedure for collecting samples and preparing a composite sample is presented in Table 7-1. One sample is collected from each of the transects and composited in one bottle. The volume of the sample is recorded on the Sample Collection Form as shown in Figure 7-2.

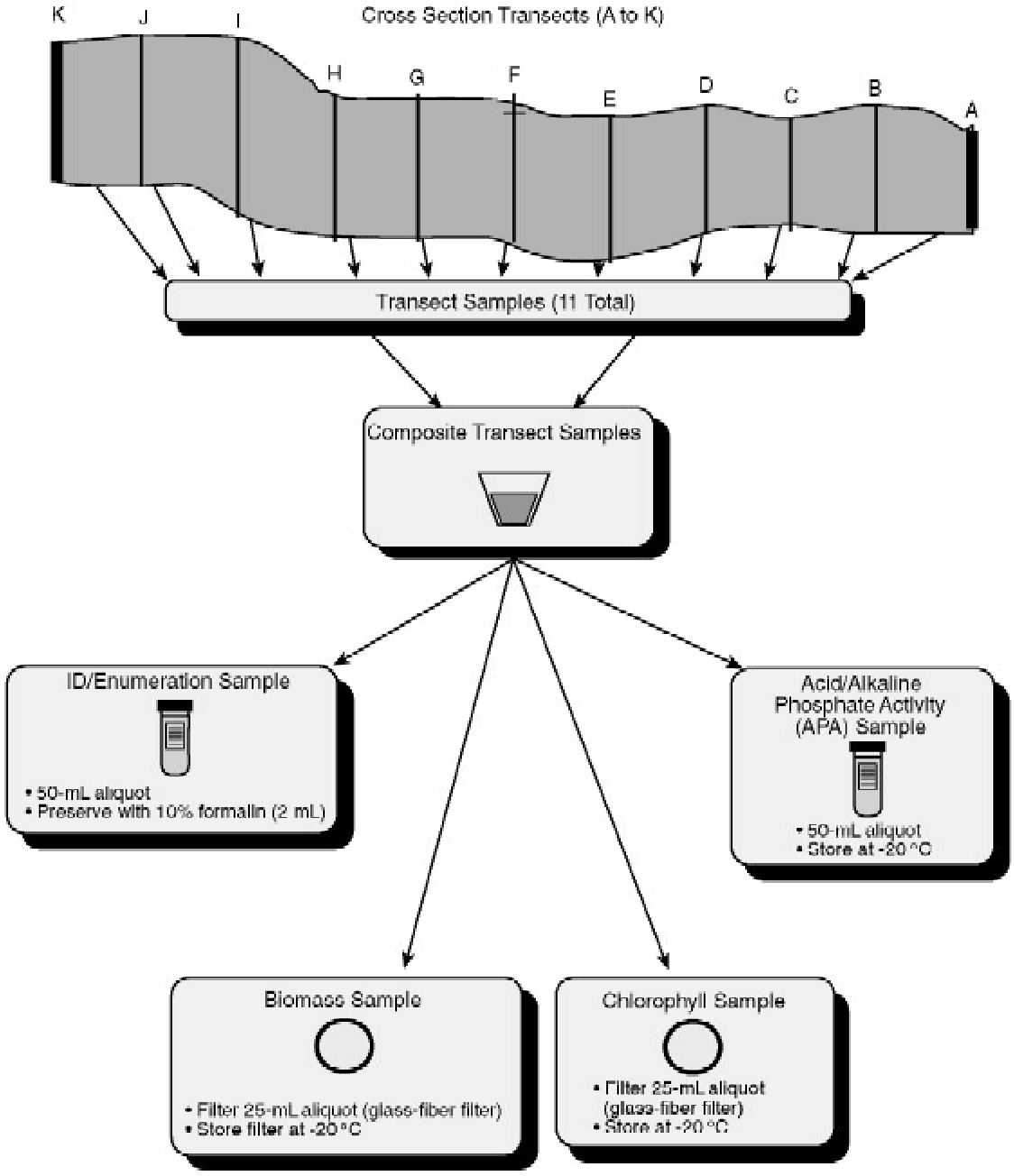

Figure 7-1. Index sampling design for periphyton.

7.2 Preparation of Laboratory Samples

Four different types of laboratory samples are prepared from the composite index samples: an ID/enumeration sample (to determine taxonomic composition and relative abundances), a chlorophyll sample, a biomass sample (for ash-free dry mass [AFDM]), and an acid/alkaline phosphatase activity

Table 7-1. Procedure for Collecting Composite Index Samples of Periphyton.

1. Starting with Transect "A", collect a single sample from the assigned shoreline using the procedure below.
   (a) Collect a sample of substrate (rock or wood) that is small enough (< 15 cm diameter) and can be easily removed from the river. Place the substrate in a plastic funnel which drains into a 500-mL plastic bottle with volume graduations marked on it.
   (b) Use the area delimiter to define a 12-cm2 area on the upper surface of the substrate. Dislodge attached periphyton from the substrate within the delimiter into the funnel by brushing with a stiff-bristled toothbrush for 30 seconds. Take care to ensure that the upper surface of the substrate is the surface that is being scrubbed, and that the entire surface within the delimiter is scrubbed.
   (c) Fill a wash bottle with river water. Using a minimal volume of water from this bottle, wash the dislodged periphyton from the funnel into the 500-mL bottle.

If no coarse sediment (cobbles or larger) are present:

   (d) Use the area delimiter to confine a 12-cm2 area of soft sediments.
   (e) Vacuum the top 1 cm of sediments from within the delimited area into a de-tipped 60-mL syringe.
   (f) Empty the syringe into the same 500-mL plastic bottle as above.
2. Repeat Step 1 for transects "B" through "K". Place the sample collected at each sampling site into the single 500-mL bottle to produce the composite index sample.
3. After samples have been collected from all 11 transects, thoroughly mix the 500-mL bottle regardless of substrate type. Record the total estimated volume of the composite sample in the periphyton section of the Sample Collection Form.

(APA) sample. All the sample containers required for an individual river 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 river shorelines.

A set of completed periphyton sample labels is shown in Figure 7-3. All labels in a set have the same sample ID number. Circle the habitat type of the composite index sample and 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, avoid covering any volume graduations and markings on the container.

7.2.1 ID/Enumeration Sample

Prepare the ID/Enumeration sample as a 50-mL aliquot from the composite index sample, following the procedure presented in Table 7-2. Preserve each sample with 2 mL of 10% formalin. Record the ID number (barcode) from the container label and the total volume of the sample in the appropriate fields on the Sample Collection Form as shown in Figure 7-2. Store the preserved samples upright in a container containing absorbent material, according to the guidelines provided for handling formalin-preserved samples.

7.2.2 Chlorophyll Sample

Prepare the chlorophyll sample by filtering a 25-mL aliquot of the composite index sample through a glass fiber filter (0.45 m nominal pore size). The procedure for preparing chlorophyll samples is presented in Table 7-3. 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 collec-

Reviewed by (initial) DKA

SAMPLE COLLECTION FORM - RIVERS

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT ☐0 ☒1 ☐2 ☐3

SITE ID: ORRV 98-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

COMPOSITE BENTHOS SAMPLES

BANK SAMPLED (LOOKING DOWNSTREAM): ☒LEFT ☐RIGHT

S = SHORE M = MID-CHANNEL D = DRIFT

SAMPLED (BARCODE)	HABITAT (CIRCLE ONE)	NO. OF JARS	FLAG	COMMENTS
243612	S M (D)	1		RIGHT BANK - SAMPLED AT LAUNCH
243611	(S) M D	1		NO SHORE KICK AT TRANSECTS
	S M D			A,G,K => TOO DEEP

STATION (CIRCLE IF DONE)	A	B	C	D	E	F	G	H	I	J
SHORE KICK	S	(S)	(S)	(S)	(S)	(S)	S	(S)	(S)	(S)
MID-CHANNEL	M	M	M	M	M	M	M	M	M	M

DRIFT NET	TRANSECT	TIME (24-HR CLOCK) START	TIME (24-HR CLOCK) END	VELOCITY (FT/SEC) XX.XX
	☒A ☐B ☐C ☐D ☐E ☐F ☐G ☐H ☐I ☐J ☐	09 : 40	19 : 45	3.0

COMPOSITE PERIPHYTON SAMPLES

SAMPLE ID (BARCODE) ➤ 244013 COMPOSITE VOLUME xxxx mL ➤ 325

ASSEMBLAGE ID (50-mL TUBE)	CHLOROPHYLL (GF/F FILTER	BIOMASS (TARED FILTER)		APA SAMPLE (50-mL TUBE)
SUB. SAMPLE VOL. xx mL	VOL. FILTERED xxxx mL	FILTER NO.	VOL. FILTERED xxxx mL	SUB. SAMPLE VOL. xxxx mL
50	25		25	50

COMMENTS:

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

Rev. 05/29/98 (_rvscmb.98)

SAMPLE COLLECTION FORM - RIVERS - 1

Figure 7-2. Sample Collection Form (page1) showing data recorded for periphyton samples.

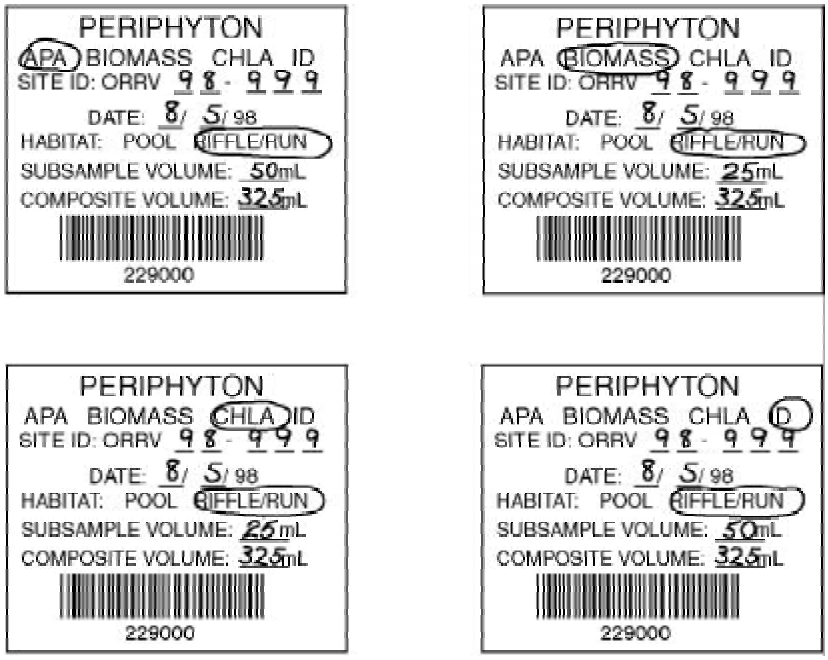

Figure 7-3. Completed set of periphyton sample labels.

Table 7-2. Preparation of ID/Enumeration Samples for Periphyton.

1. Prepare a barcoded sample label and circle the sample type ("ID") on the label. Record the volume of the subample (typically 50 mL) and the volume of the composite index sample on the label. Attach the completed label to a 50-mL centrifuge tube; avoid covering the volume graduations and markings. Cover the label completely with a clear tape strip.

2. Record the sample ID number (barcode) of the label and the total volume of the composite index sample on the form.

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

4. Thoroughly mix the bottle containing the composite sample.

5. Withdraw 50 mL of the mixed sample into the syringe. Right after mixing, place the contents of the syringe sample into the labeled 50-mL centrifuge tube.

6. Wearing gloves and safety glasses, use a syringe or bulb pipette to add 2 mL of 10% formalin solution to the tube. Cap the tube tightly and seal with plastic electrical tape. Shake gently to distribute preservative.

7. Record the volume of the sample in the centrifuge tube (excluding the volume of preservative) in the "Assemblage ID Subsample Vol." field of the Sample Collection Form.

Table 7-3. Procedure for Preparing Chlorophyll Samples for Periphyton.

1. 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.
2. Rinse the sides of the filter funnel and the filter with a small volume of deionized water.
3. Rinse a 25-mL or 50-mL graduated cylinder three times with small volumes of deionized water.
4. Mix the composite sample bottle thoroughly.
5. Measure 25 mL (±1 mL) of sample into the graduated cylinder.
   - NOTE: For a composite sample containing fine sediment, (e.g., the "DEPOSITIONAL" sample), allow grit to settle for 10 - 20 seconds before pouring the sample into the graduated cylinder.
6. Pour the 25-mL aliquot into the filter funnel, replace the cap, and pull the sample through the filter using the hand pump. NOTE: Vacuum pressure from the pump should not exceed 15 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 sample (filtrate) side folded in on itself. Wrap the folded filter in a small piece of aluminum foil.
8. Complete a periphyton sample label for chlorophyll, including the volume filtered, and attach it to the foil. Cover the label completely with a strip of clear tape. Place the foil packet into a self-sealing plastic bag.
9. Record the sample ID number (barcode) of the label and the total volume of the composite index sample on the form. Record the volume filtered in the "Chlorophyll" field on the Sample Collection Form. Double check that the volume recorded on the collection form matches the total volume recorded on the sample label.
10. Place the plastic bag containing the filter into a portable freezer or between two sealed plastic bags of ice or frozen gel packs in a cooler.
11. Rinse the filter funnel, filter holder, filter chamber, and graduated cylinder thoroughly with deionized water.

tion to minimize degradation. The filtration apparatus is illustrated in Figure 7-4. 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 15 pounds per square inch (psi) to avoid rupturing cells. If the vacuum pressure exceeds 15 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 each sample, wrap the filter in aluminum foil. Complete a sample label (Figure 7-3) and check it to ensure that all written information is complete and legible. Affix the label to the foil packet and cover it completely with a strip of clear tape. Record the barcode assigned to the sample on the Sample Collection Form (Figure 7-2). Make

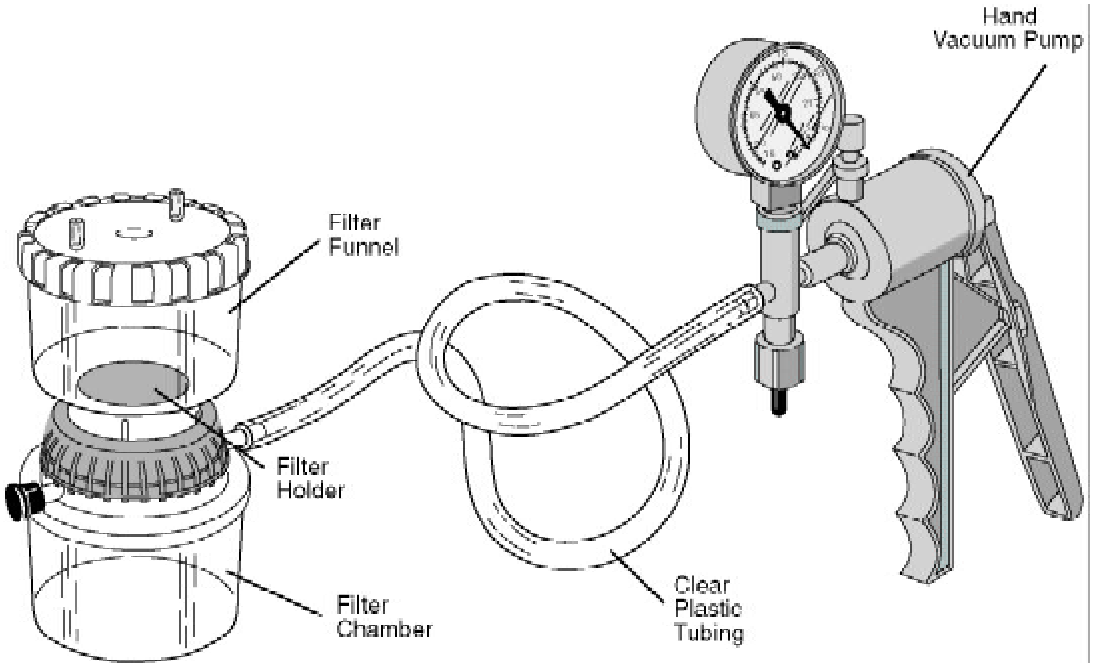

Figure 7-4. Filtration apparatus for preparing chlorophyll and biomass subsamples for periphyton. Modified from Chaloud et al. (1989).

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

7.2.3 Biomass Sample

Prepare the biomass sample from a 25-mL aliquot of the composite index sample. As with the chlorophyll sample, it is important to measure the volume to be filtered accurately (±1 mL).

After filtering each sample, complete a sample label as shown in Figure 7-3. Check each sample label to ensure that all written information is complete and legible. Affix the label to the filter container and cover it completely with clear tape. Record the bar code assigned to the sample, the container number, and the volume filtered on the Sample Collection Form as shown in Figure 7-2. Make sure the information recorded on each sample label and filters container 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 filter con-

Table 7-4. Procedure For Preparing Biomass Samples For Periphyton.

1. Using clean forceps, remove a glass-fiber filter and place it 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.
2. Rinse the filter chamber and filter with a small volume of deionized water.
3. Rinse a 25-mL or 50-mL graduated cylinder three times with small volumes of deionized water.
4. Mix the composite sample bottle thoroughly.
5. Measure 25 mL (±1 mL) of composite sample into the graduated cylinder.
   - NOTE: For a composite sample containing fine sediment, allow grit to settle for 10 - 20 seconds before pouring the sample into the graduated cylinder.
6. Pour the 25-mL aliquot into filter funnel, replace the cap, and pull the sample through the filter using the hand pump. NOTE: Filtration pressure should not exceed 15 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 filtered 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 sample portion of the filter.
8. Complete a periphyton sample label for biomass, including the volume filtered, and attach it to the foil. Cover the label completely with a strip of clear tape. Place the foil packet into a self-sealing plastic bag.
9. Record the sample ID number (barcode) of the label and the total volume of the composite sample on the form. Record the volume filtered in the "Biomass" portion on the Sample Collection Form. Double check that the volume recorded on the collection form matches the total volume recorded on the sample label.
10. Place the labeled filter container into a cooler containing two sealed plastic bags of ice.
11. Rinse the filter funnel, filter holder, filter chamber, and graduated cylinder thoroughly with deionized water.

tainer frozen until shipment to the laboratory (Section 3).

7.2.4 Acid/Alkaline Phosphatase Activity Sample

The Acid/Alkaline phosphatase activity (APA) sample is prepared from a 50-mL subsample of the composite index sample. Table 7-5 presents the procedure for preparing APA samples. No field treatment (i.e., filtration, preservation) of the APA sample is necessary. Complete a label for each sample as shown in Figure 7-3 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 7-2). 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).

7.3 Equipment and Supplies

Figure 7-5 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

Table 7-5. Procedure for Preparing Acid Alkaline Phosphatase Activity Samples for Periphyton.

1. Prepare a barcoded sample label. Circle the sample type ("APA") and the habitat type ("Riffle/ Run" or "Pool") on the label. Record the volume of the sample (typically 50 mL) and the volume of the composite index sample on the label. Attach the completed label to a 50-mL centrifuge tube; avoid covering the volume graduations and markings. Cover the label completely with a clear tape strip.
2. Rinse a 60-mL syringe with deionized water.
3. Thoroughly mix the bottle containing the composite sample.
4. Withdraw 50 mL of the mixed sample into the syringe. Place the contents of the syringe sample into the labeled 50-mL centrifuge tube. Cap the tube tightly and seal with plastic electrical tape.
5. Record the sample ID number (barcode) of the label and the total volume of the composite index sample on the form.
6. Record the volume of the sample in the centrifuge tube in the "APA Sample" field of the Sample Collection Form.

Equipment and Supplies for Periphyton		
Qty.	Item	
1	Large funnel (15-20 cm diameter)	
1	12-cm2 area delimiter (3.8 cm diameter pipe, 3 cm tall)	
1	Stiff-bristle toothbrush with handle bent at 90° angle	
1	1-L wash bottle for stream water	
1	1-L wash bottle containing deionized water	
1	500-mL plastic bottles for the composite sample.	
1	60 mL plastic syringe with 3/8" hole bored into the end	
4	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	
1	Filtration unit, including filter funnel, cap, filter holder, and receiving chamber	
1	Hand-operated vacuum pump and clear plastic tubing	
2	Aluminum foil squares (3" x 6")	
2	Self-sealing plastic bags for chlorophyll 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	
1 set	Sample labels (4 per set) with the same barcode ID number	
1	Sample Collection Form for river	
	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	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 periphyton	

Figure 7-5. Checklist of equipment and supplies for periphyton

the required equipment is brought to the river. Use this checklist to ensure that equipment and supplies are organized and available at the river site in order to conduct the activities efficiently.

7.4 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.

Section 8
Sediment Community Metabolism

Brian H. Hill and Alan T. Herlihy

This section describes procedures to collect a composite sediment sample from the sampling reach. Sediment samples are collected from each transect at the same time as periphyton samples (Section 7) and benthic macroinvertebrate samples (Section 9). At each river, a composite "index" sample of sediment is prepared and used in the determination of sediment community metabolism.

The method outlined here for determining sediment community metabolism is designed for headwater to mid-order streams, and has been adapted for larger rivers or lakes. The method measures changes in dissolved oxygen (DO) concentrations of the overlying water within microcosms containing small amounts (ca. 10 mL) of sediments as a means of assessing benthic microbial community activity. Sediments are collected from depositional habitats along the study reach. Following incubation, the DO is re-measured and the sediments are saved for ash-free dry mass (AFDM) analysis. Respiration rate, estimated as the change in DO concentration per hour within each microcosm, is adjusted for AFDM, yielding a measure of community respiration per gram of AFDM. Organic carbon turnover time can be calculated from the empirical relationship between the organic carbon content of the sediment (estimated as 0.5 × AFDM) and oxygen consumption.

8.1 Sample Collection

Table 8-1 describes the procedure for collecting the composite sediment sample. Collect sediment from depositional areas (e.g., pools, eddies, and backwaters) located at or near each of the cross-section transects within the sampling reach. If soft sediments are scarce, collect them from wherever you can within the sampling reach. At each sampling point, use a small plastic scoop to collect the top 2 cm (1 inch) of soft surface sediment. Combine sediments from different sampling points into a single jar or self-sealing plastic bag to prepare a single composite index sample

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Table 8-1. Sediment Collection Procedure.

1. At the first cross-section, locate a depositional habitat (a pool, eddy, or backwater).
   - If soft sediments are scarce, collect them wherever you can within the reach
2. Use a plastic scoop to collect a sample of surficial sediment (top 2 cm). Remove any visible organisms from the sediment. Place the sample in a plastic jar with volume graduations labeled "SEDIMENT SAMPLE".
   - Approximately 250 mL of sediment (~ 30 mL per transect) is required for sediment metabolism.
3. Repeat Steps 1 through 2 for Transects "B" through "K".

for the river reach. A composite sample volume of 250-mL is sufficient to prepare sediment metabolism samples.

8.2 Determining Sediment Respiration

The procedure to measure sediment respiration in presented in Table 8-2. A dissolved oxygen meter, equipped with a biological oxygen demand (BOD) probe and stirrer, is used for the determination of respiration rates. This may or may not be the same meter used

Table 8-2. Procedure To Measure Sediment Respiration.

1. Inspect the probe of the dissolved oxygen meter 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. Check the batteries and electronic functions of the meter as described in the meter's 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.
   - NOTE: Try to perform the calibration as close to river temperature as possible (not air temperature) by using river water to fill the calibration chamber prior to equilibration.
   - NOTE: For doing the elevation correction, the elevation of the sample site is provided on the site information sheet in the dossier for the site. Alternatively, obtain the elevation from a topographic map.
3. Prepare a set of five sediment metabolism sample labels. Note that each label will have a different sample ID number (barcode). Attach each completed label to a 50-mL screw-cap centrifuge tube.
   - NOTE: Avoid covering volume gradations on the tube with the label. Cover each label with a strip of clear tape.
4. Fill a small insulated cooler full with river water. Measure the dissolved oxygen and temperature of the water in the cooler. Record the values in the "Initial O2" and "Initial Incubation Temp." fields in the metabolism section of the Field Measurement Form.
5. Thoroughly mix the composite sediment sample. Use a small plastic spoon to transfer 10 mL of sediment from the composite sample container to each of the five labeled tubes.
6. Fill each tube to the top (no head space) with river water from the cooler and seal the tube. Fill a centrifuge tube labeled "BLANK" with river water from the cooler and seal. This tube serves as a control for changes in ambient conditions during the incubation period.
7. Place the six tubes in a 1-L plastic beaker and place the beaker inside the cooler. Record the start time in the "Incubation Time" area of the Field Measurement Form. Close the cooler and incubate the sediment samples for 2 hours.
8. After incubation, re-calibrate the oxygen probe (i.e., the meter was turned off or you have moved to a different elevation during the incubation) before the end of the incubation period.
9. At the end of the incubation period, record the end time in the "Incubation Time" area of the Field Measurement Form. Measure the DO in each tube, including the blank. Record the sample ID number of each tube and its measured DO concentration on the Field Measurement Form.
10. Decant the overlying water from each labeled tube, retaining the sediment. Tightly seal each tube and place in a cooler with bags of ice as soon as possible. Keep the samples frozen until they can be shipped. Discard the water from the "BLANK" tube.

to determine in situ dissolved oxygen concentration (Section 5). If a separate meter is used to measure sediment respiration, check the probe membrane and the meter's batteries and electronics according to the instrument's operating manual (see Sections 3 and 5, also). Calibrate the meter as directed in the instrument's operating manual.

A small cooler filled with stream water is used as an incubation chamber. The initial dissolved oxygen concentration and temperature of the water in the cooler are measured and recorded on the Field Measurement Form as shown in Figure 8-1. This concentration is assumed to be the initial concentration of all subsamples. Five sediment subsamples (10-mL ±1 mL) are prepared from the composite sediment sample. A set of completed sample labels for these subsamples is shown in Figure 8-2. A 10-mL subsample of water from the incubation cooler is used as a control for changes in ambient conditions during the incubation. The subsamples are incubated in the cooler for 2 hours. After the incubation, the final DO concentration of each tube is determined and recorded on the Field Measurement Form (Figure 8-1). The sediment in each tube is retained and stored frozen until it can be shipped to the laboratory (Section 3) to determine the AFDM.

8.3 Equipment and Supplies

Figure 8-3 is a checklist of equipment and supplies required to conduct sediment sampling and to determine sediment community respiration. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the river. Use this checklist to ensure that equipment and supplies are organized and available at the river site in order to conduct the activities efficiently.

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FIELD MEASUREMENT FORM - STREAMS/RIVERS

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT L0 [X] L2 L3

SITE ID: ORRV 98-999 TEAM ID (X): [X] 2 3 4 5 6 7 8

WEATHER CONDITIONS (X)

CLOUD COVER	[X] < 5%	[ ] 5-25%	[ ] 25-50%	[ ] 50-75%	[ ] > 75%
PRECIPITATION	[X] NONE	[ ] LIGHT	[ ] MODERATE	[ ] HEAVY	
PREVIOUS PRECIPITATION (24 H)	[X] NONE	[ ] LIGHT	[ ] MODERATE	[ ] HEAVY	
AIR TEMPERATURE XX	28 °C				

IN SITU MEASUREMENTS

STATION ID: K Assume X-site unless marked

		FLAG	COMMENTS
CCCS COND µS/CM XXXX	75		AT TRANSECT "K"
STREAM/RIVER COND µS/CM XXXX	110		AT TRANSECT "K"
STREAM/RIVER DO MG/L XX.X	8.8		AT TRANSECT "K"
STREAM/RIVER TEMP °C XX.X	17.6		AT TRANSECT "K"

FISH TISSUE SAMPLES - SECONDARY SAMPLE (where available: 5 individuals)

INITIAL O_2 (MG/L) XX.X	INITIAL INCUBATION TEMP (°C) XX.X	INCUBATION TIME (24-HR TIME) START (HH:MM)	INCUBATION TIME (24-HR TIME) FINISH (HH:MM)	DURATION OF INCUBATION (HH:MM)	FLAG	COMMENTS
9.0	18.4	18:30	22:30	4:00		

SAMPLE ID (BARCODE)	FINAL O_2 (MG/L)[2] XX.X	FLAG	COMMENTS
243663	7.5		INITIAL D.O. AT 1000' ELEVATION
243664	7.2		FINAL D.O. AT 2200' - METER CALIBRATED
243665	7.8		FOR EACH ELEVATION
243666	8.1		
243667	7.0		
CONTROL	8.8		

OXYGEN METER CALIBRATION INFORMATION

MEMBRANE CHECK [X] ELECTRONIC ZERO [ ] RED LINE [X]

CALIBRATION CHAMBER TEMPERATURE: °C

STATION ELEVATION (FROM TOPO. MAP OR ALTIMETER): 1000 FT

SATURATED O_2 @ TEMP: MG/L

ELEVATION CORRECTION FACTOR: x

The calibration value is obtained by multiplying the saturated DO concentration times an elevation correction factor (obtained from the tables on the back of the YSI meter). Adjust the meter reading to the calibration value.

CALIBRATION VALUE: MG/L

COMMENTS:

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.

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FIELD MEASUREMENT FORM - STREAMS/RIVERS - 1

Figure 8-1. Field Measurement Form (page 1), showing data for sediment metabolism samples.

Figure 8-2. Completed sample labels for sediment metabolism.

Equipment And Supplies For Sediment Metabolism		
Qty.	Item	
1	Small scoop sampler for sediments	
1	Wide-mouthed plastic jar labeled "COMPOSITE SEDIMENT SAMPLE". If sediment is only being collected for metabolism samples, a 250-mL jar is sufficient.	
1	YSI Model 95 Dissolved Oxygen meter	
1 set	Spare batteries for DO meter	
1	Small plastic spoon or spatula to transfer sediment from the composite sample container to respiration tubes	
5	50-mL, screw-top, centrifuge tubes	
1	50-mL screw-cap centrifuge tube labeled "BLANK"	
1	Small cooler used as incubation chamber	
1	1,000-mL plastic beaker to holding centrifuge tubes during incubation	
5	Sediment metabolism sample labels (each with different ID number)	
1	Field Measurement Form	
	Soft (#2) lead pencils to fill in field data forms	
	Fine tip indelible markers for preparing labels	
1 pkg	Clear tape strips for covering labels	
1	Cooler with bags of ice to store sediment metabolism samples	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for sediment community metabolism	

Figure 8-3. Checklist of equipment and supplies for sediment metabolism.

Section 9
Benthic Macroinvertebrates

Donald J. Klemm[1], James M. Lazorchak[1], and David V. Peck[2]

Benthic macroinvertebrates inhabit the sediment or live on the bottom substrates of lakes, streams, and rivers. The macroinvertebrate assemblages in rivers reflect the overall biological integrity of the benthic community such that monitoring these assemblages is useful in assessing the status of the water body and monitoring 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 environmental conditions.

EMAP scientists are currently evaluating two different approaches 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 (1994), Fore et al. (1996), Barbour et al. (1995; 1996), and Karr and Chu (1999).

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 ana-

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lyzing benthic macroinvertebrate community data.

Field procedures for collecting and processing benthic invertebrate samples from non-wadeable streams are presented in Section 9.1. These procedures are based upon draft procedures developed for the Mid-Atlantic Integrated Assessment (MAIA) study conducted in the eastern U.S. Section 9.2 contains an equipment and supply checklist for benthic invertebrate sampling.

9.1 Sampling Procedures for Non-wadeable Streams

The length of river reach established for larger non-wadeable streams and rivers is much larger than for wadeable streams, making a visual estimate of the number of riffle and pool macrohabitat units impossible. In addition, mid channel depths of larger streams and rivers will make it impractical to collect kick net samples from mid-river habitats. In non-wadeable streams and rivers, samples are collected at each of eleven transects established for physical habitat characterization. At each transect, two kick net samples are obtained from shallow area (< 1m) near the bank of the river. Kick net samples collected from each transect are composited into a single sample for the river; samples collected from different macrohabitat types are not composited separately. A kick net modified for use by one person is shown in Figure 9-1. In addition to the mesh size used in the two EMAP studies other mesh sizes such as 250 - 800 m can be used depending upon the objectives of

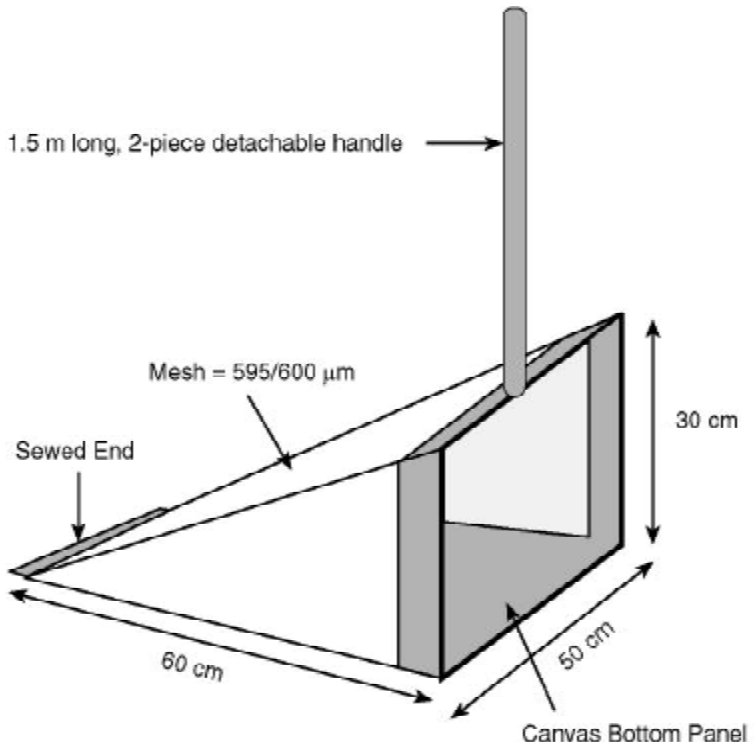

Figure 9-1. Modified kick net. (Not drawn to scale.).

the program and potential for clogging. For deep rivers that are extremely difficult or hazardous to obtain benthic samples with a kick net, a ponar or core grab sample could be used instead.

In addition, two daytime drift net samples are collected from as near to the downstream end of the defined reach as is practical. The drift net assemblies are positioned when the crew drops off a vehicle at the take-out point, and are retrieved when the crew reaches the sampling point in the boat.

9.1.1 Sample Collection Using Kick Nets

9.1.1.1 Selection of Sampling Points

Samples are collected from non-wadeable streams during a downstream traverse of the sample reach. At each transect location, locate a suitable sampling point on the same side of the river as fish sampling is conducted (Section 10). Locate the sampling point in an area away from the river margin, but at a depth less than or equal to 1 m.

9.1.1.2 Sample Collection

At each sampling point, obtain TWO kick net samples using the procedures presented in Table 9-1 (if the sampling point is located in a riffle or glide macrohabitat) or Table 9-2 (if the sampling point is located in a pool macrohabitat). If there is insufficient flow to sample a transect with the modified kick net following this protocol, spend about 60 seconds hand picking a sample from approximately 0.25 m^2 of substrate at the station and combine it with samples from other transects in the bucket. If there is too little water to collect the sample with the kick net, randomly pick up 10 rocks, and pick and wash the organisms off of them into the bucket. Keep a note of this on the field sheets and in all databases generated from sites where more than one transect has to be sampled in this manner. Results may show a bias due to the larger organisms picked in this approach.

9.1.2 Sample Processing: Kick Net Samples

After all transects have been sampled, the composite sample is processed as described in Table 9-3. Sample labels to put on and in the jar are shown in Figures 9-2 and 9.5, respectively, and the sample collection form is shown in Figure 9-4. Ensure the sample is preserved and that the jar is completely sealed. Place the sealed sample jar upright in a cooler or plastic bucket for transport. Blank labels for use inside of sample jars are presented in Figure 9-5. These can be copied onto waterproof paper.

9.1.3 Description of Drift Nets and Habitat Sampled

Drift nets are stationary nets designed to sample organisms from flowing waters such as streams and rivers. The drift net sampler is designed to obtain qualitative and quantitative samples of macroinvertebrates which either actively or passively enter the water column from all types of substrates in flowing water with a velocity of not less than 0.05 m/s. They can be used to capture organisms at and below the surface of the water. Drift nets can be used individually or in groups with nets strung out side by side or arranged vertically.

Table 9-1. Collecting Kick Net Samples From Non-wadeable Streams: Riffle/Run Macrohabitats.

1. Attach the four foot handle to the kick net. Care should be exercised to be sure the handle is on tight or the net might become twisted in strong current or while dragging it through the water causing the loss of part of the sample.
2. Position the sampler quickly and securely on the river bottom with the net opening upstream so as to eliminate gaps under the frame. Reposition the sampling point to avoid large rocks that prevent the sampler from seating properly.
3. The sampling area (or quadrate) has a width and length equal to the width of the net frame (0.5 m) or a total area = 0.25 m 2.
4. Hold the sampler in position on the substrate and check the quadrat directly in front of the net for heavy organisms, such as mussels and snails. Place these organisms into the net.
5. Continue to hold the sampler securely while vigorously kicking the substrate within the quadrat for 20 seconds (use stopwatch).
6. After 20 seconds, hold the net in place with the knees. Pick up any loose rocks in the quadrat and scrub off organisms in front of the net. Place any additional mussels and snails found in the quadrat in the net.
7. Remove the net from the water with a quick upstream motion to wash the organisms to the bottom of the net. Immerse the net several times to remove fine sediments and to concentrate organisms at the end of the net. Avoid having any water or additional material enter the mouth of the net during this operation.
8. Transfer the contents of the net into a plastic bucket half filled with water by inverting the net into the bucket.
9. Inspect the net for clinging organisms. Use forceps and remove any organisms found and place them into the bucket.
10. Carefully inspect large objects (rocks, sticks, leaves, etc.) in the bucket. Wash off any organisms, then discard the objects. Remove as much detritus, sediment, and debris as possible without losing any organisms.
11. See Table 9.3 for processing kick net samples.

The drift net consists of a bag of nylon or nylon monofilament frame. The standard drift net is approximately 1 m (39.3") long and has a closed end. The drift net open end is 30.48 cm (12") x 45.72 cm (18"). The net frame is made of stainless steel rods or PVC pipe. The frame of the drift net is anchored into the river bed by a pair of steel rods, 15.46 cm (18") long or can be attached to a "floating drift assembly" device (Figure 9-3), Wildlife Supply Co., 1999-2000. Drift net frames can also be fitted anteriorly with a mouth reducing rectangular plexiglass enclosure (Rutter and Ettinger, 1977; Wefring, 1976) to increase filtration efficiency. For EMAP, MAIA sampling in Regions 2, 3, and 4 rivers, a drift net with 600 m mesh openings has been used in conjunction with the floating drift assembly device (other mesh sizes such as 250 - 800 m can be used depending upon the objectives of the program and potential for clogging).

The drift collection usually represents a wide spectrum of the habitats found in a river. Drift nets are effective for the collection of emigrating and dislodged benthic macroin-vertebrates drifting in the water column of flowing streams and rivers. Sampling efficiency of this gear is a function of current velocity and sampling period. Data collected can be used to estimate macroinvertebrate drift densities and rates (individuals per unit volume of water per unit time passing through the net). However, this requires an estimate of the volume of water passing through the sampling nets. This is accomplished by averaging repeated measures of the water velocity at the mouth of the drift net and recording the total time the drift net is set in the

Table 9-2. Collecting Kick Net Samples From Non-wadeable Stream: Pool\Glide Macrohabitats.

1. Attach the four foot handle to the kick net. Care should be exercised to be sure the handle is on tight or the net might become twisted in strong current or while dragging it through the water causing the loss of part of the sample.
2. The sampling area (or quadrate) has a width and length equal to the width of the frame (0.5 m) or a total area = 0.25 m 2.
3. Inspect the river bottom within the quadrat for any heavy organisms, such as mussels and snails. Remove and place these organisms into the net.
4. Disturb the substrate within the quadrat by kicking vigorously with the feet. Drag the net repeatedly and continuously through the disturbed area just above the bottom whole continuing to kick for 20 seconds (use a stopwatch). Keep moving the net so that the organisms trapped in the net will not escape.
5. Remove the net from the water with a quick upstream motion to wash the organisms to the bottom of the net. Immerse the net several times to remove fine sediments and to concentrate organisms at the end of the net. Avoid having any water or additional material enter the mouth of the net during this operation.
6. Hold the net so that the mouth is out of the water and the net is partially submerged. Pick up any loose rocks in the quadrat and rub or brush any organisms found on them into the net. Also recheck the quadrat for any additional snails or clams and place them in the net.
7. Transfer the contents of the net into a bucket half filled with water by inverting the net into the bucket.
8. Inspect the net for clinging organisms. Use forceps and remove any organisms found and place them in the bucket.
9. Carefully inspect large objects (rocks, sticks, leaves, etc.) in the bucket. Wash any organisms found into the bucket, then discard the objects. Remove as much detritus, sediment, and debris as possible without losing any organisms.
10. See Table 9.3 for processing kick net samples.

water column. Repeated measures of the water velocity are most representative of the sample period if they are taken when the nets are first set and just prior to removal of the net from the system.

Limitations and hazards of daytime drift net sampling include:

- Unknown where organisms come from; terrestrial species may make up part of sample in summer and periods of wind and rain. It is not the ideal time of day for sampling drifting benthic macroinvertebrates.
- Installing and retrieving drifts from areas of swift current can be hazardous. Placing nets in highly visible areas may result in tampering or theft. Boats and skiers may damage drift net assemblies.

9.1.3.1 Drift Net Sampling Procedures

For EMAP, MAIA pilot studies of Regions 2, 3, and 4 rivers, install two drift nets at transect K (See Table 9-4), one about 25 cm from the bottom substrate and one about 10 cm below the surface in water not exceeding 3 m in depth, using cable and anchor attached to a "floating drift assembly" device. The installation procedure for drift nets is presented in Table 9-4. Install the net in an area of river that is receiving part of the main channel flow, but that can be safely accessed by wading. A location that you would consider to provide a representative water chemistry sample is probably also suitable for positioning a drift net. Do not use drift nets if the current velocity at the sampling point is less than 0.05 m/s or more than same rate.

Table 9-3. Processing Kick Net Samples: Non-wadeable Streams.

1. Fill out a sample label for the composite samples. Attach the label to a 500-mL (or 1-L) plastic jar. If the sample contains a large volume of material, complete a sample label for additional containers and attach them. Make sure the barcode numbers on each label are identical.
2. Hand-pick large organisms from the bucket containing the composite sample and place them into the appropriately labeled jar.
3. Hand-pick large rocks and sticks remaining in the bucket. Use a small brush to scrub debris from them back into the bucket. Discard the rock or stick.
4. Empty the contents of the bucket into a sieve (600 m) mesh, and then transfer into the labeled jar. NOTE: Do not fill the jar more than ½ full of material. If necessary, use a larger jar and/or distribute the sample among two or more labeled jars. Rinse residue from the bucket into the jar using a wash bottle and a small volume of water.
5. Add 95% ethanol to each labeled jar so that the final concentration of ethanol is at least 70%. If there is a small amount of water in the sample, it may not be necessary to fill the jar entirely full to reach a 70% concentration. It is very important that sufficient ethanol be used to reach a 70% concentration. Otherwise, the organisms will not be properly preserved.
6. Place the waterproof label with the following information inside each jar:
   - Stream Number
   - Type of sampler and mesh size used
   - Habitat type (riffle/run, pool/glide)
   - Name of stream
   - Date of collection
   - Collectors initials
   - Number of transects composited
7. Rinse the bucket well to eliminate any residue.
8. Complete the Sample Collection Form and on the jars (1 of 2, 2 of 2, etc). Record the barcode number of the composite sample, and the habitat type (shore). If more than one container was required for a sample, record the number of containers on the collection form. Replace the lid on the jar. Seal the container lid(s) with plastic or electrician's tape. Also note any peculiarities associated with a particular samples by using a flag code and/or a written comment on the collection form.

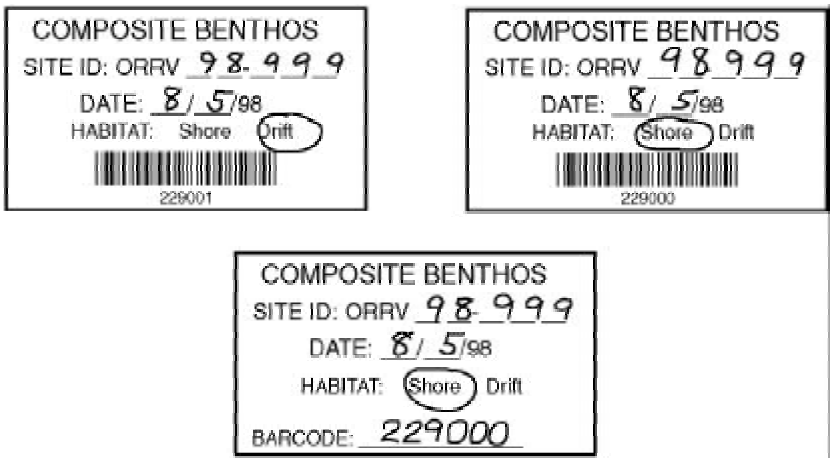

Figure 9-2. Completed labels for benthic macroinvertebrate samples. The bottom label is used if more than one jar is required for a composite sample.

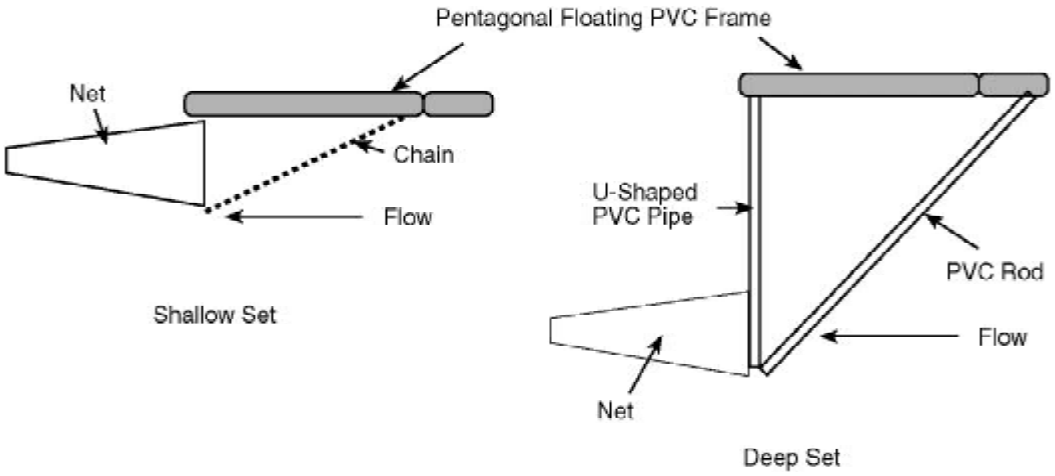

Figure 9-3. Shallow and deep set drift net assemblies. (Not drawn to scale).

Measure the current velocity at the entrance to each net at the time the net is installed and again when it is retrieved. Velocity is determined by timing a neutrally buoyant object over a known distance or using a flow meter.

9.1.3.2 Processing and Preservation of Drift Samples

After retrieving the drift nets from the stream or river, process the sample as described in Table 9-5. Sample labels are shown in Figure 9-2, and the sample collection form is shown in Figure 9-4. Note that the material from the two nets is combined to yield a single composite sample of drift for the stream or river. Blank labels for use inside of sample jars are presented in Figure 9-5. These can be copied onto water-proof paper.

9.1.3.3 Maintenance of the Drift Nets

After the drift sample has been processed and preserved, thoroughly wash the drift nets with water from the stream or river to remove all debris, etc.

9.2 Equipment Checklist

A list of all equipment and supplies required to conduct benthic invertebrate sampling is presented in Figure 9-6.

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SAMPLE COLLECTION FORM - RIVERS

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT ☐0 ☒1 ☐2 ☐3

SITE ID: ORRV 98-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

COMPOSITE BENTHOS SAMPLES

BANK SAMPLED (LOOKING DOWNSTREAM): ☒LEFT ☐RIGHT

S = SHORE M = MID-CHANNEL D = DRIFT

SAMPLED (BARCODE)	HABITAT (CIRCLE ONE)	NO. OF JARS	FLAG	COMMENTS
243612	S M (D)	1		RIGHT BANK - SAMPLED AT LAUNCH
243611	(S) M D	1		NO SHORE KICK AT TRANSECTS
	S M D			A,G,K => TOO DEEP

STATION (CIRCLE IF DONE)	A	B	C	D	E	F	G	H	I	J
SHORE KICK	S	(S)	(S)	(S)	(S)	(S)	S	(S)	(S)	(S)
MID-CHANNEL	M	M	M	M	M	M	M	M	M	M

DRIFT NET	TRANSECT	TIME (24-HR CLOCK) START	TIME (24-HR CLOCK) END	VELOCITY (FT/SEC) XX.XX
	☒A ☐B ☐C ☐D ☐E ☐F ☐G ☐H ☐I ☐J ☐	09 : 40	19 : 45	3.0

COMPOSITE PERIPHYTON SAMPLES

SAMPLE ID (BARCODE) ➤ 244013 COMPOSITE VOLUME xxxx mL ➤ 325

ASSEMBLAGE ID (50-mL TUBE)	CHLOROPHYLL (GF/F FILTER	BIOMASS (TARED FILTER)		APA SAMPLE (50-mL TUBE)
SUB. SAMPLE VOL. xx mL	VOL. FILTERED xxxx mL	FILTER NO.	VOL. FILTERED xxxx mL	SUB. SAMPLE VOL. xxxx mL
50	25		25	50

COMMENTS:

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

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SAMPLE COLLECTION FORM - RIVERS - 1

Figure 9-4. Sample Collection Form (page 1), showing information for benthic macroinvertebrate samples.

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 _____	**BENTHOS IDENTIFICATION** Site Number _____ Stream _____ Collection Date _____ Sampler _____ Habitat Type _____ Collector(s) _____ Number of Transects _____

Figure 9-5. Blank labels for benthic invertebrate samples.

Table 9-4. Collection Procedures for Drift Net Samples: Non-wadeable Streams.

NOTE: Do not use drift nets for large rivers with currents less than 0.05 m/s.

Installation and Retrieval of Drift Nets:

1. Ideally, the net should be installed at the downriver end of the designated sampling reach (transect K in non-wadeable streams). In practice, the net is installed at either the takeout point (1st choice) or put-in point, whichever is located closer to the designated sampling reach. Mark the nearest transect on the Sample Collection Form and note if the drift net location is outside of the designated sampling reach in the Comments section of the collection form.
2. Locate the drift net assembly in an area receiving the main flow of the river (i.e., avoid backwaters, river margins, eddies, etc.)
3. Anchor the net assembly using anchors and cables.
4. Record the START TIME of sampling on the Sample Collection Form.
5. Measure the current velocity at the entrance of the net, using the neutrally buoyant object technique (or a flow meter) as follows:
    A. Measure out a straight segment of the river reach just upstream of the drift net location in which an object can float relatively freely and passes through within about 10 to 30 seconds.
    B. Select an object that is neutrally buoyant, like a small rubber ball or an orange; it must float, but very low in the water. The object should be small enough that it does not "run aground" or drag bottom.
    C. Time the passage of the object through the defined river segment 3 times. Record the length of the segment and each transit time in the Comments section of the Sample Collection Form.
6. The net assembly should be left in the river for at least 3 hours or as long as possible at the site. Upon return to the net location after floating the designated sampling reach, retrieve the net assembly from the water, taking care not to disturb the bottom upstream of the net.
7. Record the END TIME on the Sample Collection Form.
8. Determine the current velocity again as described in Step 5 above. Record the three "final" velocity estimates in the Comments section of the collection form. Calculate the average velocity from the initial and final values (6 measurements). Record the average velocity in the "Velocity" field of the Sample Collection form. Exclude any gross outlier values from the computation of the average velocity.
9. Note in the comments section if the net is badly clogged, which may occur at locations with high discharge and/or where the float time of the sampling reach is long.

Table 9-5. Procedures for Processing Drift Net Samples: Non-wadeable Streams.

1. Fill out a sample label for the composite drift sample. Attach the label to a 500-mL or 1-L plastic jar. If the sample contains a large volume of material, complete a sample label for additional containers and attach it to a second jar. Make sure the barcode numbers on each label agree.
2. Concentrate the material in each net in one corner by swishing up and down in the stream or river. Wash the material into a bucket half-filled with water. Use a wash bottle and/or forceps to remove as much material as possible from the net.
3. Repeat Step 1 for the second drift net. The contents of both nets are combined into a single bucket.
4. After the two net samples are combined into a single bucket, pour the composite sample into a sieving bucket (595 micron mesh).
5. Hand-pick large organisms from the sieve bucket containing the composite sample and place them into the appropriately labeled jar.
6. Hand-pick large rocks and sticks remaining in the bucket. Use a small brush to scrub debris from them back into the bucket. Discard the rock or stick.
7. Lightly "tapping" the bottom of the sieve bucket on the surface of the stream or river helps to remove fine material. Remove as much material as possible using the sieve bucket.
8. Empty the contents of the bucket into the labeled jar. If necessary, distribute the sample among two or more labeled jars. Rinse residue from the bucket into the jar using a wash bottle and a small volume of water.
9. Add 95% ethanol to each labeled jar so that the final concentration of ethanol is at least 70%. If there is a small amount of water in the sample, it may not be necessary to fill the jar entirely full to reach a 70% concentration. It is very important that sufficient ethanol be used to reach a 70% concentration. **Otherwise, the organisms will not be properly preserved.**
10. Place a waterproof label with the following information inside each jar:
    - Stream Number
    - Type of sampler and mesh size used
    - Habitat type (drift net)
    - Name of stream
    - Date of collection
    - Collectors initials
    - Number of transects composited
11. Rinse the bucket well to eliminate any residue.
12. Complete the Sample Collection Form. Record the barcode number of the composite sample, and the habitat type (drift). If more than one container was required for a sample, record the number of containers on the collection form and on the jars. Replace the lid on the jar, and seal the container lid(s) with plastic or electrician's tape. Also note any peculiari ties associated with a particular samples by using a flag code and/or a written comment on the collection form.

Equipment and Supplies for Benthic Macroinvertebrates

Qty.	Item	
1	Modified kick net with 595 m mesh openings and closed bag (Wildco #425-J50-595)	
1	Handle for Kick Net Sampler, four foot length	
1	Floating drift net assembly (PVC frame, chains, snap-clips, and carabineers)	
2	Drift nets, 595 m mesh, closed end	
1	Sieve-bottomed bucket, 595- m mesh openings (optional)	
2 pr.	Watchmakers' forceps	
1	Wash bottle, 1-L capacity	
1	Small spatula, spoon, or scoop to transfer sample	
1	Funnel, with large bore spout	
12	Sample jars, plastic with screw caps, 500 mL and 1 L capacity, suitable for use with ethanol	
2	Buckets, plastic, eight to ten quart capacity	
1 gal	95% ethanol, in a proper container	
2 pr.	Rubber gloves, heavy rubber	
1	Cooler (with suitable absorbent material) for transporting ethanol and samples	
6	Sample labels, pre-numbered barcoded, stick-on type	
6	Sample labels, blank, stick-on type (for additional containers)	
2	Sample Collection Form for site	
1	Field check list sheet	
	Soft (#2) lead pencils	
1 pkg.	Clear waterproof tape strips	
4 rolls	Plastic electrical tape	
1	Knife, pocket, with at least two blades	
1	Stopwatch	
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 9-6. Equipment and supply checklist for benthic macroinvertebrates.

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Section 10
Aquatic Vertebrates

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

Vertebrate sampling is intended to collect all but the rarest fish and aquatic amphibian species in a reach and their abundances in the collection should be relative to their proportionate abundance in the water body. Data on species richness, species guilds, abundance, size and anomalies are used to assess ecosystem condition. In rivers, vertebrates are collected first. Boat electrofishing equipment is used as the principal sampling gear (Section 10.1.1), and only the boat personnel are involved in collecting aquatic vertebrates. In addition to gathering data on the assemblage, fish specimens are retained for analysis of tissue contaminants (Section 11).

10.1 Sample Collection

Depending on the survey region, rivers are sampled along one bank for a distance equal to either 40 or 100 times the wetted width in the vicinity of the point of entry. The mean channel width is measured with a laser range finder and estimated from maps and at the top of the reach. In the relatively fast, cold, oligotrophic, or species-depauperate rivers of some regions, lower total fish catches and efficiency of capture (compared with those in relatively slow, warm, eutrophic, or species-rich rivers) necessitate a greater sampling reach length. To capture a sufficient number of fish in rivers of some regions, sample reaches 100 Channel-Widths long may be specified for regional surveys, based largely upon fish capture requirements. River reaches 40 channel widths long are specified in the Mid-Atlantic region, for example, whereas 100 channel-width long reaches are specified in Pacific Northwest rivers.

10.1.1 Electrofishing

Because vertebrates are collected using electrofishing units, safety procedures must be followed at all times (refer to Section 2). Primary responsibility for safety while electrofishing rests with the crew leader. Electrofishing units have a high voltage output and may deliver a fatal electrical shock. While electrofishing, avoid

[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.

contact with the water unless sufficiently insulated against electrical shock and, do not touch objects outside the boat. Use watertight rubber linesman's gloves. If gloves develop a leak or become wet inside, use another pair or stop fishing until they are repaired and thoroughly dry. Avoid contact with the anode and cathode at all times due to the potential shock hazard. At no time while the electrofisher is on should a team member reach into the water for any reason. If it is necessary for a team member to reach into the water to pick up a fish or something that has been dropped, do so only after the electrical current has been turned off. Do not electrofish when navigating major rapids and wait for the second boat to clear them. Do not resume electrofishing until all nontarget individuals are clear of the electroshock hazard or obstacle. The boat units have three kill switches. Insure that these work before fishing. Do not make any modifications to the electrofishing unit that interrupt the current or that would make it impossible to turn off the electricity.

Crew members must complete CPR and first aid courses. They should be strong swimmers and, as appropriate, complete a white water rescue course. Wear a life jacket when in a boat and avoid operating electrofishing equipment within 20 feet of nontarget organisms. Discontinue activity during thunderstorms, heavy rain or if the top or inside of the boat is wet. Crew members should keep each other in constant view or communication while electrofishing. For each site, know the location of the nearest hospital with a defibrillation unit. Although the crew leader has authority, each crew member has the responsibility to question and modify an operation or decline participation if it is unsafe. Use hand signals to communicate direction and power on or off because of generator noise, and avoid colliding with obstacles overhead and in the water. Rest if the team becomes fatigued, and drink lots of water.

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. Always wear gloves and safety glasses when handling gasoline. Keep gasoline only in approved plastic containers and store in a tightly closed container.

Boat electrofishing sampling procedures are presented in Table 10-1. Record information on the Vertebrate Collection Form as shown in Figure 10-1. If the river cannot be sampled by electrofishing, complete the "NOT FISHED" field on the form. Select the initial voltage based on the measured conductivity of the river (see Section 5). Select the initial frequency based on the expected size of fish. If fishing success is poor, increase the pulse width first and then the voltage. Increase the frequency last to minimize mortality or injury to large fish.

The electrofishing boat is a 14 -16 ft. inflatable raft or john boat modified for two persons and all fishing equipment. Boat configuration consists of a frame mounted generator and electrofishing control box, port and starboard cathodes, and two anodes extending out over the bow. Alternatively, the john boat itself may be used as the cathode. The boat is maneuvered by one operator seated near the stern, and the vertebrates are collected and identified by one netter operating from the bow. Prior to fishing, determine that the netter is wearing gloves and both team members are clear of all electrodes. Wear polarized sunglasses to aid vision. Start the electrofisher, set the timer to zero, and depress the foot pedal to begin fishing. Starting at the top of the reach and along the designated

Table 10-1. Procedure to Collect Aquatic Vertebrates by Boat Electrofishing.

Onshore at launch site

a. Check generator oil and fill tank with gas (wipe up any spillage).
b. Clip cathodes to sides of frame & connect their cables to the cathode outlet (if the fishing site is distant, keep electrodes in boat).
c. Connect anode cables to outlets (if the fishing site is distant, keep anodes on poles in the boat).
d. Connect generator and pulsator.
e. Confirm that all gear for the day and a spare vehicle key are in the boat.
f. Put on a life jacket and gloves.
g. Go to step 2 & 3 below to assess electrofisher performance.

1. Complete the header information on a copy of the Vertebrate Collection Form. Indicate the transect being sampled in the "TRANSECT" field on the form.
2. Select river bank for fishing (left for odd numbered sites, right for even) unless immediate hazards or obstructions preclude this. Stay along the selected bank throughout the day's fishing to the degree it is safely navigable; do not switch back and forth between banks unless the river aspect is unchanging and the selected side is not representative (e.g., very sunny and shallow) of both. Using the rangefinder, determine a downstream point that is 10 mean channel widths distant (this is the profile length). Record this distance on the field sheet.
2. 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 all non-target organisms are clear of the water or two boat lengths away, and that boat surfaces are dry.
3. Start generator, switch to pulsed DC, a frequency of 30 pps, low range 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 overloading the generator. Switching should occur when power is off. Netter activates safety switches and insures that when either is employed current ceases. Verify that fish are rolled and relaxed but not rigid. Record settings on field sheet in comments section and start cleared clocks.
4. With system activated and safety switches on, begin fishing downstream near shore. Maneuver the boat or anode to cover a swath two-three meters wide, at an oars length from shore, near cover, and at depths less than three meters wherever possible. Do not place the boat in danger in order to fish particular habitats; cut the generator and stow the gear before negotiating hazards.
5. Place fish directly in livewell as soon as possible; 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 systems this will be impossible. Do not chase individual fish with the boat or lean far out from the boat to net them. If benthic fish are being missed, pivot the boat occasionally or hold the net behind the anode and along the bottom so some are collected.
6. Cease sampling at the end of the profile. Process the fish quickly and carefully, returning them to the water unless they are vouchered. Be sure that the data sheet is completed accurately and completely, and that voucher specimens are taken. Record the "Total Shock Time," "TOTAL FISHING TIME," and "SHOCK DISTANCE" on the Vertebrate Collection Form. If no aquatic vertebrates were collected, complete the "NONE COLLECTED" field on the Vertebrate Collection Form.
7. Complete field collections and field sheets for other indicators taken at the end of the profile. Return to step 1 for each of the subsequent 9 profiles, but begin downstream from where fish were released.

shoreline, fish in a downriver direction. Adjust voltage and output according to sampling effectiveness and incidental mortality to specimens.

The netter uses an insulated dip net to retrieve stunned individuals, which are then deposited into a livewell for later processing (Section 10.2). Change the water in the livewell

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VERTEBRATE COLLECTION FORM - STREAMS/RIVERS Page 1 of 1

SITE NAME: BEAVER RIVER DATE: 8 5 98 VISIT: 0 [X] 2 3

SITE ID: ORRV 98-999 TEAM ID (X): [X] 2 3 4 5 6 7 8

ID BY (NAME): J. DOE TRANSECT (X): [X] B C D E F G H I J A (stream)

TOTAL SHOCK (button) TIME (xxxx) 270 seconds TOTAL FISHING TIME (xxx) 6 minutes SHOCK DISTANCE (m) 400

SAMPLE ID (BARCODE) 243360 ☐ NOT FISHED ☒ NONE COLLECTED

SPECIMENS

TAG NO	SPECIES CODE	COMMON NAME	TOTAL NUMBER		VOUCHERED COUNT	LENGTH (mm)		ANOMALIES		NUMBER OF MORTALITIES	FLAG
			TALLY	COUNT		MIN.	MAX.	CODE	COUNT		
	PROS WILL	MOUNTAIN WHITEFISH	卌 11	7	1	110	390	HM	1	0	F1
	ONCOMYKI	RAINBOW TROUT	卌 1	6	0	105	390	NO	0	0	
	COTTZZZZ	UNKNOWN COTTIA	1111	4	4	30	182	NO	0	0	
	CATOMACR	LARGESCALE SUCKER	1111	4	2	103	598	LE	1	0	
	PTYCOREG	NORTHERN SQUAWFISH	1	1	1	184		NO	0	0	

FLAG	COMMENTS
	NOTE: ALL VOUCHERED SPECIMENS FOR ALL SPECIES ARE NOT TAGGED (NO TAG NUMBER)
	AND ARE LOOSE IN SAMPLE CONTAINERS. THIS APPLIES TO THIS TRANSECT, AS WELL AS
	ALL OTHERS FISHED.
F1	EIGHT ADDITIONAL WHITEFISH TAKEN OVER THE COURSE OF THE ENTIRE REACH → ANOTHER STUDY

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

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Figure 10-1. Vertebrate Collection Form.

periodically to minimize mortality prior to processing. If individuals show signs of stress (loss of righting response, gaping, gulping air, excessive mucus), stop and process them. This should only be necessary on very warm days, in long reaches, or if very large numbers of individuals are collected. Electrofishing may also need to cease at times to immediately process and release specimens (e.g., listed species or large game fish or if fish appear to be stressed due to temperature and/or low DO) as they are netted (see Section 10.2). If periodic processing is required, be sure to release individuals upriver and away from the shoreline to reduce the likelihood of collecting them again.

At the completion of electrofishing each profile, record the total operating time (shock time) and total fishing time shown on the electrofisher timer and the distance sampled by electrofishing on the Vertebrate Collection Form (Figure 10-2). If no aquatic vertebrates were collected, indicate this on the form as shown in Figure 10-2. During this project, specimens should be processed after completion of every transect when possible to provide data on catch per unit effort.

10.2 Sample Processing

Sample processing involves tallying and identifying fish, 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 (see Section 11). Process collections as quickly as possible to minimize stress to live specimens. The netter is responsible for identifying, measuring, and examining aquatic vertebrates contained in the livewell. At the end of each profile, the netter processes fish from the livewell while the operator records information on the field data forms.

10.2.1 Taxonomic Identification and Tally

Table 10-2 presents the procedure for identifying and tallying aquatic vertebrates. Record identification and tally data for each species on the Vertebrate Collection Form as shown in Figure 10-1. Also record comments and data for additional species on the Vertebrate Collection Form. The team is to be provided with a list of standardized names (required) and species codes (optional) for aquatic vertebrate species that are expected to be collected (see Appendix C for an example).

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 may be facilitated by keeping a tally count of the number of individuals of each species as it is taken from the livewell and totaling the tally once processing is complete.

To minimize handling, process threatened and endangered species first, and immediately return all individuals to the river. If conditions permit and stress to individuals will be minimal, photograph such fish for voucher purposes (Section 10.2.3). Photographs of fish, fish too large to voucher, fish anomalies, and the sites themselves are very informative to those of us who cannot be in the field. Be sure to photograph the site number so we can link photos and places. Indicate if photographed with an "F" series flag for the species on page 1 of the Ver-

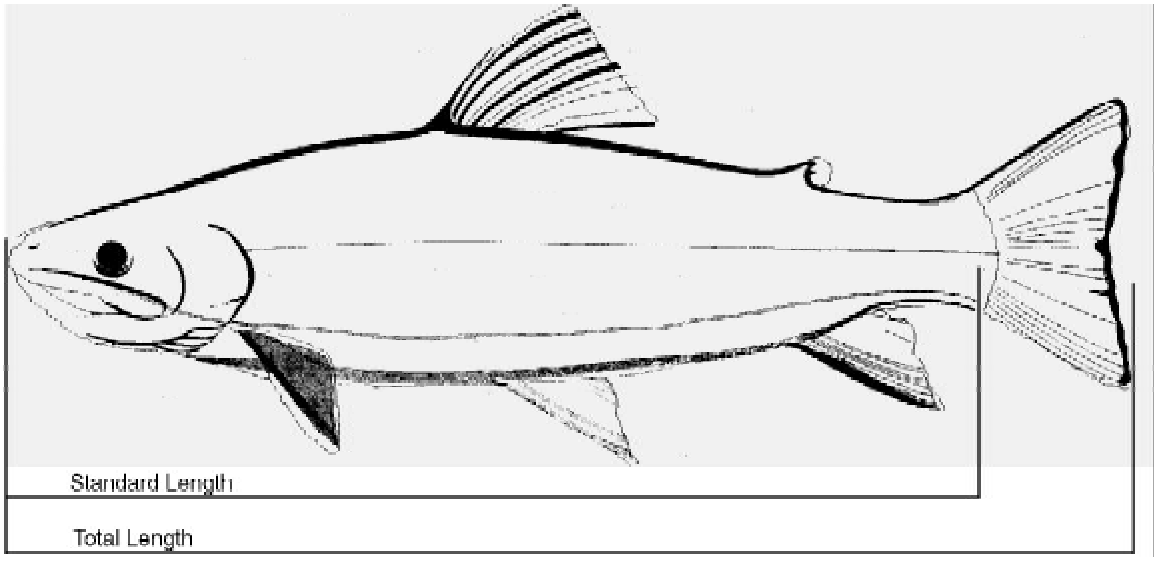

Figure 10-2. Fish length measurements (modified from Lagler, 1956).

Table 10-2. Procedure To Identify, Tally, And Examine Aquatic Vertebrates.

1. Complete the header information on the form, then record the common name (from a standardized list) and species code on the first blank line in the "SPECIMENS" section of the Vertebrate Collection Form. If a species cannot be positively identified, assign it an "unknown" species code from the list provided.
2. Examine each fish individually; small-sized fish species may be handled in small manageable groups to speed processing.
3. To minimize handling, threatened and endangered species should be identified, counted, and returned immediately to the stream. If conditions permit and stress to individuals will be minimal, photograph fish for voucher purposes. Indicate if photographed on data sheet with flags and comments. If protected fish have died, they should be vouchered in formalin. At the earliest possible time, the appropriate state officials should be notified.
4. Sport fish and very large specimens should be identified, measured for total length to the nearest mm, examined for external anomalies, and released. Record all information on the vertebrate collection form. Keep voucher specimens (up to 5) 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 should be placed on ice and then frozen.
5. Identify all other species in the livewell.
6. Tally the number of individuals collected (use the "Tally" box on the Vertebrate Collection Form if necessary) and record the total number in the "Count" field on the form.
7. Measure the total length of the largest and smallest individual to provide a size range for the species. Record these values in the "Length" area of the Vertebrate Collection Form.
8. Measure the total length of each individual (up to 30) and record the lengths in the boxes on the Vertebrate Length Recording Form (2 lines of boxes per species). For smaller species, measure and record lengths of a random set (up to 30) of the individuals collected.
9. Examine each individual for external anomalies and note the types of anomalies observed. After all of the individuals of a species have been processed, record the anomaly code and the total number of individuals affected in the "Anomalies" area of the Vertebrate Collection Form.
10. If individuals have died due to the effects of electrofishing or handling, record the total number of mortalities on the Vertebrate Collection Form.
11. Follow the appropriate procedure to prepare voucher specimens and/or to select specimens for tissue samples. Release all remaining individuals into the river.
12. Repeat Steps 1 through 11 for all other profiles.

tebrate Collection Form (Figure 10-1) and record a notation in the comments section. If protected fish have died, they should be prepared as voucher specimens and preserved in formalin. Notify the appropriate state officials as soon as possible.

If a species cannot be confidently identified in the field (e.g., small individuals or suspected hybrids), record it as an "unknown" species on the Vertebrate Collection Form, using one of the names (and code) provided for unknowns from the standardized list (see Figure 10-1 for an example). If possible, flag unknown species with an "F" series flag and provide your best guess at an identification in the comments section of the form.

10.2.2 External Examination and Length Measurements

During the tallying procedure for each species (Table 10-2), examine each individual for the presence of external anomalies. External anomalies may result from sublethal environmental or behavioral stress, diseases, and toxic chemicals. Readily identified external anomalies, include deformities, eroded fins, lesions, tumors, diseases and parasites. Codes for different types of anomalies are presented in Table 10-3. Record the types of anomalies observed and the number of individuals affected on the Vertebrate Collection Form as shown in Figure 10-1.

Blackening and exopthalmia 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 10-1). Care should be taken in the early stages of electrofishing to use the most effective combination of voltage and pulse width while minimizing injury to fish. 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 10-1).

For each species, use a measuring board or ruler to determine the total length (Figure 10-2) of the largest and smallest individuals (this is a check on your measurements of total lengths recorded on the Vertebrate Length Recording Form [Figure 10-3]). Use of "tick marks" on the length form will aid you in determining maximum and minimum lengths for a profile. Measure individuals on the right side, and slide fish to touch the "Bump Board" on the measuring board. Measure total length to the nearest millimeter (mm) and record these values on the Vertebrate Collection Form as shown in Figure 10-1. Measure the total lengths of up to 30 individuals and record these values on the Vertebrate Length Recording Form as shown in Figure 10-3.

10.2.3 Preparing Voucher Specimens

With the exception of very large individuals and protected species, collect vouchers of all species allowed by collecting permits to provide a permanent, archived, historical record of fish collections. Prepare the voucher sample for a site according to the procedure presented in Table 10-4. Retain additional specimens of the appropriate species for the fish tissue contaminants sample (Section 11). For each species, voucher specimens take priority over specimens for the tissue contaminants sample.

Voucher specimens for each species are counted and placed into individual nylon mesh bags (1 bag per species). Nylon stockings or

Table 10-3. External Anomaly Categories and Codes.

Categories	Code	Definition
Absent	AB	Absent eye, fin, tail.
Blisters	BL	In mouth, just under skin.
Blackening	BK	Tail or whole body with darkened pigmentation.
Extensive black spot disease	BS	Small black cysts (dots) all over the fins and body.
Cysts	CY	Fluid-filled swellings; may be either small or large dots.
Copepod	CO	A parasitic infection characterized by a worm-like copepod embedded in the flesh of the fish; body extends out and leaves a sore/discoloration at base, may be in mouth gills, fins, or anywhere on body.
Deformities	DE	Skeletal anomalies of the head, spine, and body shape; amphibians may have extra tails, limbs, toes.
Eroded fins	EF	Appear as reductions or substantial fraying of fin surface area.
Eroded gills	EG	Gill filaments eroded from tip.
Fungus	FU	May appear as filamentous or "fuzzy" growth on the fins, eyes, or body.
Fin anomalies	FA	Abnormal thickenings or irregularities of rays
Grubs	WG	White or yellow worms embedded in muscle or fins.
Hemorrhaging	HM	Red spots on mouth, body, fins, fin bases, eyes, and gills.
Ich	IC	White spots on the fins, skin or gills.
Lesions	LE	Open sores or exposed tissue; raised, granular, or warty outgrowths.
Lice	LI	Scale-like, mobile arthropods.
Mucus	MU	Thick and excessive on skin or gill, or as long cast from vent.
None	NO	No anomalies present.
Other	OT	Anomalies or parasites not specified (Please comment).
Scale anomalies	SA	Missing patches, abnormal thickenings, granular skin
Shortened operculum	SO	Leaves a portion of the gill chamber uncovered
Tumors	TU	Areas of irregular cell growth which are firm and cannot be easily broken open when pinched. (Masses caused by parasites can usually be opened easily.)
Leeches	WL	Annelid worms which have anterior and posterior suckers. They may attach anywhere on the body.
Exophthalmia	EX	Bulging of the eye.

panty hose may substitute for nylon bags. Each bag contains a numbered tag (Figure 10-4). Record the tag number and the number of individuals vouchered for each species on the Vertebrate Collection Form as shown in Figure 10-1. Single specimens of easily identified and distinct species (e.g., sandroller, smallmouth bass) may be placed directly in the jar.

The preceding steps are critical to enable us to link a species' field and lab identifications with the number of individuals so named. If done correctly, we can estimate the number of individuals collected from the proportions in the bag, even if a presumed single species turns out to be 2 or 3 species (this is one reason we voucher as many specimens of a species as possible). It is useful to pre-

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SITE NAME: BEAVER RIVER DATE: 8 5 98 VISIT: 1 2

SITE ID: CRRV 98-999 TEAM ID (X): 1 2 3 4 5 6 7 8

MEASURED BY (NAME): J. DOE

SPECIES CODE	COMMON NAME	LENGTH (MM) OF INDIVIDUAL FISH														
ONCOMYKI	RAINBOW TROUT	315	300	270	185	160	170	350	340	160	270	340	345	310	280	105
		180	310	160	185	260	180	390	340	315	310	110	290	360	290	180
CATOMACR	LARGESCALE SUCKER	472	426	150	103	150	120	500	485	358	486	500	560	507	598	140
		250	530	170												
PROSWILL	MOUNTAIN WHITEFISH	330	300	340	320	335	123	110	370	390	300	320	315	320	350	310
		242	340	330	325	340	305	325	335	270	320	320	110	310	320	345
PTYCOREG	NORTHERN SQUAWFISH	210	220	220	346	220	395	382	375	265	330	380	320	290	215	410
		465	330	250	240	220	430	385	325	184	285	340				
COTTZZZZ	UNKNOWN COTTIA	98	70	66	90	85	70	72	63	81	70	35	72	65	70	80
		30	30	194	182											
CATOCOLU	BRIDGEUP SUCKER	210	160	248	171	185	186	249	130	160	170	380	325	190	180	205
		230	264	230	260	175	110	100	194	260	155	260	125	180	190	220
ACROALUT	CHISELMOUTH	225	180	220	200	203										
RHINCATA	LONGNOSE DACE	150	95	70												

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VERTEBRATE LENGTH RECORDING FORM - STREAMS/RIVERS - 1

Figure 10-3. Vertebrate Length Recording Form.

Table 10-4. Guidelines and Procedures for Preparing Aquatic Vertebrate Voucher Specimens.

Determine the voucher category of a species and the number of specimens to include in the voucher sample based on the following guidelines. NOTE: Category 3 species should be processed first.

A Category 1 — Large easily identified species OR adults may be difficult to identify OR the species is uncommon in that region. Examples include:

Centrarchids Catostomids Cyprinids Ictalurids Salmonids

1. 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.

   • NOTE: Individuals with a total length > 160 mm should be slit on the lower abdomen of the RIGHT side before placing them into the container.

2. Photograph if considered too large for the jar and place in bag on ice for freezing (Do not retain large gamefish). All photographs should include (1) a card with the stream ID, date, species code, and common name, and (2) a ruler or some other object of known length to provide some indication of the size of the specimen.

3. Retain additional individuals for the tissue contaminant sample.

B. Category 2 — Small to moderate-sized fish OR difficult to identify species. Examples include:

Lampreys Minnows Sculpins Sticklebacks Sunfish

1. Preserve up to 20 adults and juveniles (ideally several per profile). If fewer than 20 individuals are collected, voucher all of them. Voucher samples take priority over tissue contaminant sample.

2. Retain additional individuals for tissue contaminants sample.

C. Category 3 — Species of "special concern." These are state or federally listed species.

1. Photograph as in Step 1.A.2 and then release immediately.

2. If specimens have died, proceed to Step 2 and include them as part of the voucher sample. Flag the species with an "F" series flag 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.

3. Place the voucher specimens in a bucket with two carbon dioxide tablets (e.g., Alka Seltzer®) and a small volume of water. When specimens are anaesthetized, transfer them to a nylon mesh bag. Record the number of individuals included in the voucher sample in the "Vouchered Count" field for the species on the Vertebrate Collection Form.

4. Select a "FISH-BAG" tag that has the same ID number (barcode) as the voucher sample jar (Step 3). 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.

(continued)

Table 10-4. Continued.

5. Immediately place the bag into a container (½ or 1 gal plastic jar) large enough to hold all voucher specimens and half-filled with 10% formalin. Use additional jars if necessary to avoid tight packing and bending of voucher specimens.

6. Repeat Steps 1 through 4 for all species collected.

7. Prepare two "FISH-JAR" labels (each having the same ID number [barcode]) 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.

8. 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 (barcode) 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.

9. 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.

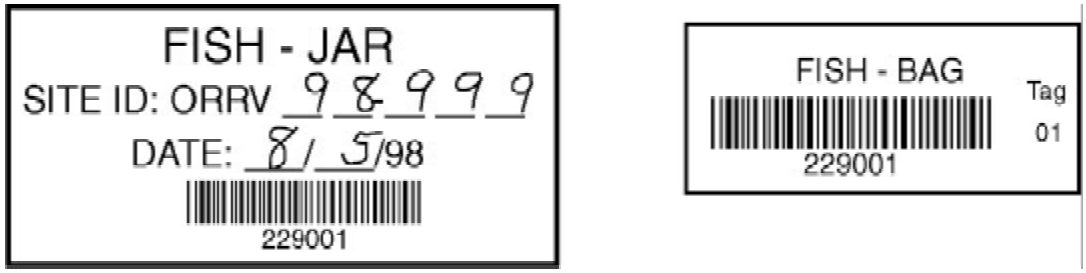

Figure 10-4. Completed voucher sample label and specimen bag tag for aquatic vertebrates.

serve vouchers of sculpins, lampreys and other difficult species from throughout the reach.

Place specimen bags together into a large plastic sample container. Preserve voucher specimens with a 10% formalin solution. See Section 3 for instructions for preparing a buffered formalin solution. Larger voucher specimens (total length > 160 mm) should be slit on the lower abdomen of the RIGHT side to allow for complete fixation of internal tissues and organs. If a fish is too large for a jar, photograph and place in bag on ice. Flag on recording sheet; freeze at lab separately from tissue. Start with a concentrated solution of formaldehyde and dilute to the final volume with water. The final volume of 10% formalin in the sample container should equal the total volume of specimens. Use

additional containers if necessary and avoid tight packing of specimen bags.

Delays in carrying out the anaesthetization and preservation procedures, overpacking a sample container, or an inadequate volume of preservative will produce unidentifiable specimens.

Formaldehyde (37%) and formalin (10% formaldehyde by volume) are extremely caustic agents and may cause severe irritation on contact of vapors or solution with skin, eyes or mucus membranes. It is a potential carcinogen. Contact with vapors or solution 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 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 10-4. Place one label inside each sample container, and attach the second label to the outside of the jar with clear tape. Record the sample ID number on the Vertebrate Collection Form as shown in Figure 10-1. Carefully complete the collection form at each transect. Tag numbers must be linked to each species, and each bag of species. Be careful with fish names and their spellings (computers see errors as different species). Some museums may also require that a separate collection card be completed and inserted into each jar of voucher specimens.

10.3 Equipment and Supplies

Figure 10-5 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 ensure that equipment and supplies are organized and available to conduct the protocols efficiently.

Equipment and Supplies for Aquatic Vertebrates		
Qty.	Item	
1	14 ft. boat with frame mounted electrofishing gear (anodes, cathodes, control box)	
3	Oars (1 as extra)	
2	Dip nets, long handled	
1	Dip net, short handled	
1	Generator and filled gas can; rag	
1	Fire extinguisher	
2	Anodes and cathodes (Spare)	
1	Livewell cooler	
1	Measuring board and ruler	
2	Pesola scales for weighing primary tissue samples	
2	Buckets (5 gallon)	
1	Seat cooler with ice	
1	Air pump, hose and fitting	
2	Personal floatation devices	
many	Boat straps and ropes	
1	Tool box (Leatherman, duct tape, spare oarlock, straps, electrical tape, fuses, zipties)	
1 roll	Aluminum foil	
2	Dry bags	
1	Boat repair kit	
1	First aid kit	
3	Taxonomic reference books and keys for fishes of the region	
1	List of vertebrate species common names (and species codes, if required)	
1	List of external anomaly codes	
15-20	Small nylon mesh bags for holding voucher specimens (bags can also be constructed from sections of nylon stockings or panty hose)	
1	Small fillet knife or scalpel for preparing larger voucher specimens for preservation	
2 ea.	1/2- or 1-gallon screw-top plastic jars for voucher sample	
2 L	10% (buffered) formalin solution	
1	Cooler to hold formalin solution and preserved voucher sample jars	
1 pr	Safety glasses	
1 pr	Chemical-resistant gloves	
1	Topographic map(s)	
1 ea	Laser rangefinder, stopwatch, camera and film, whistles	
2	Pruning saw and sheath	
	Rubber Linesman gloves, clipboards, polarized glasses, ziplock bags	
4	Carbon dioxide tablets (Alka-Seltzer® or equivalent)	
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 labels	
1 roll	Plastic electrical tape	
1 pkg.	Clear tape strips	
	Soft lead pencils for recording data and completing tags	
	Fine-tipped indelible markers for completing sample labels	
10+extras	Vertebrate Collection Form	
1 + extras	Vertebrate Length Recording Form	
1	Field operations manual	
1 set	Laminated sheets of aquatic vertebrate procedure tables and/or quick reference guides	

Figure 10-5. Equipment and Supplies for Aquatic Vertebrates.

10.4 Literature Cited

Lagler, K.R. 1956. Freshwater Fishery Biology. 2nd. Edition. William C. Brown Co., Dubuque, Iowa.

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.

Section 11
Fish Tissue Contaminants

James M. Lazorchak[1], Frank H. McCormick[1], Robert M. Hughes[2], and Spence A. Peterson[3]

In addition to gathering data on the aquatic vertebrate assemblage (Section 10), fish are retained for analysis of tissue contaminants. In general, the focus is on fish species that commonly and occur throughout the region of interest, and that are sufficiently abundant within a sampling reach. The fish tissue contaminants indicator, which measures bioaccumulation of persistent toxics, is used to estimate regional risks of consumption to fish predators, either wildlife or human. Various studies that have been done on fish tissue contaminants have focused on different parts of the fish: whole fish, fillets, livers. EMAP-SW will focus on whole fish because of its emphasis on the ecological health of the whole stream (as opposed to a focus on human health concerns). Whole fish are a good ecological indicator and 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).

For the fish contaminants indicator in EMAP-SW STREAMS, an attempt was made to collect two fish samples at as many sites as possible. One sample, of **Primary Target Species**, was stream fish whose adults are small (in Mid-Atlantic streams examples are: dace, chub, sculpins, stonerollers, shiners, and darters). The second sample, where available, of a **Secondary Target Species**, was a species whose adults are of larger size (In Mid-Atlantic streams examples are: bass, trout, sunfish, suckers, 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 ad-

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[2]Dynamac International Corp., 200 SW 35th St., Corvallis, OR 97333.
[3]U.S. EPA, National Health and Environmental Effects Laboratory, Western Ecology Division, 200 SW 35th St., Corvallis, OR 97333.

vantages over large fish. Most importantly, it may be possible to get a more representative sample of the contaminant load in that stream section (although it would be at a lower expected level of bioaccumulation) by compositing say, in the range of 20 to 200 small fish individuals than by compositing 3 to 5 large fish. Small fish may be a more appropriate indicator for assessing ecological risk, as they might be expected to be prey for a larger number of fish-eating animals (the majority of which will be piscivorous birds and small mammals). 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 having Primary and Secondary Target Species in this study.

In trying to answer these questions, the field crews' efforts to apply the protocol for sampling, handling and shipping in a **consistent manner** are **critical**. The diligence of the field crews in following the protocols is especially important in a status and trends study such as EMAP-SW where it is critical to get a **standard sample** from each site so that there is confidence that differences seen over time and between sites represents variations in the ecosystems and not differences in sampling and handling between the crews. Suggestions from field crew members on how the protocol can be improved are welcomed and will be incorporated to improve them, but protocols should be followed as written until official changes are made.

11.1 Selecting Fish Tissue Specimens

If possible, obtain one sample each, of the desired **weight** or **number** (see below) of **similarly sized*** individuals, from the **Primary** and **Secondary** target species lists (**2 composite samples** total). To judge if the proper amount of a target species is present in the fish catch, **weight** will be used for **primary** target species and **number of individuals** of sufficient size will be used for **secondary** target species.

I. Primary Target Species

Small adult fish (in priority order)	Weight
1) Blacknose Dace	50** - 400 g
2) Another Dace species	50** - 400 g
3) Creek Chub or Fallfish	50** - 400 g
4) Slimy Sculpin/Mottled Sculpin	50** - 400 g
5) Stoneroller	50** - 400 g
6) A Darter species	50** - 400 g
7) A Shiner species	50** - 400 g

A) Choose the **highest priority target species** from the above list, that has at least enough individuals to attain the minimum weight (50 g). Get as much weight of fish as possible within the desired weight range (50-400 g).

B) If **less than the desired weight of any primary target species** is collected, send individuals of a small **nontarget** species if 50 g or more are available.

* - The general rule-of-thumb for similar size is that the smallest individual in the sample should be at least 75% of the length of the largest individual. This rule applies to both primary and secondary target species. Crews just need to keep this criterion in mind while selecting the final sample. Any obviously small or large individuals should not be kept if there is a sufficient sample to return

without them. If there is a conflict between criteria, getting a sufficient sample is a higher priority than getting similar-sized individuals.

** - This weight represents the **minimum amount** of tissue needed for laboratory analysis. Crews **should not settle for the minimum amount** (weight) if more fish are present, but instead send as many fish as possible up to the 400 g weight goal.

II. Secondary Target Species

Collect and save a sample of secondary target species if such a sample of desired number of individuals of desired size is available. Collect **similar sized individuals** if enough are present.

Larger adult fish (in priority order)	Desired Size	Desired Number
1) A Bass species	120 mm	5
2) A Trout species*	120 mm	5
3) A Sunfish species	120 mm	5
4) Catfish	120 mm	5
5) White sucker	120 mm	5
6) Hogsucker	120 mm	5
7) Carp	120 mm	5

* - Collect only those trout that appear not to be recently stocked.

A) If **fewer than the desired number** of secondary target species individuals **of desired size** are collected, add smaller individuals of the same species, if available, to achieve the desired number (5).

B) If **fewer than 5 fish** of **any size** are available, you may send **as few as 3 fish** that are at least at or near the minimum desired size (120 mm).

C) If an acceptable secondary target species sample (by the above criteria) is not available send only the primary target species sample. If neither a primary or secondary species sample that meets these criteria is available, use your best judgement in sending some type of fish sample.

11.2 Preparing Composite Samples for Primary and Secondary Target Species

To determine the proper quantity for each composite sample, weight is used for the primary target species and the number of individuals of sufficient size is used for the secondary target species. Prepare each composite sample using similar sized individuals if possible, but getting a sufficient sample is a higher priority than getting similar-sized individuals.

Prepare a primary sample as described in Table 11-1. Choose a species that has at least enough individuals to attain the minimum weight (50 g). Send as many fish as possible up to the 400 g weight goal. If there is no single species with enough individuals available, prepare the sample using individuals of multiple species.

Prepare a secondary sample as described in Table 11-1. Choose a species that has 5 similar-sized individuals (minimum total length = 120 mm) available. If fewer than 5 fish of any size for any secondary species are available, prepare the composite sample using as few as 3 fish that are at least at or near the minimum desired size.

If neither a primary nor secondary sample is available, use your best judgement to obtain some type of fish tissue sample from the available species collected. Use the procedure for either primary or secondary spe-

Table 11-1. Procedure to Prepare Fish Tissue Samples.

Note: If neither a primary nor secondary species sample is available, use your best judgement in sending some type of composite fish tissue sample.

Primary Sample (P)

After all voucher specimens have been prepared, choose a primary species that has enough similarly sized individuals to weigh to 400 g (smallest to largest should not differ by more than 25% in length).

Secondary Sample (S)

After all voucher specimens have been prepared, select a large secondary species that has at least 5 individuals 120 mm. Include similar sized individuals if available (smallest to largest should not differ by more than 25% in length).

1. Place the fish into a bucket with two carbon dioxide tablets (e.g., "Alka Seltzer®") and a small volume of water. After they have been anaesthetized, use clean hands to transfer them to aluminum foil.
2. Prepare a clean work surface to prepare the primary composite sample. Keep hands, work surfaces, and wrapping materials clean and free of potential contaminants (mud, fuel, formalin, sun screen, insect repellant, etc.)

3-P. For primary samples, record the common name (from a standardized list) of the species, its species code (if required), and the number of individuals in the sample in the appropriate fields on line "P1" of the Sample Collection Form (Figure 11-1).

3-S. Measure the total length (TL) of each secondary individual. Record the common name (from a standardized list) of the secondary target species, its species code (if required), and the total length for each individual on lines S1 through S5 in the secondary sample section of the Sample Collection Form.

4. If the individuals included in composite samples were collected from throughout the sampling reach, place an "X" in the "Yes" box in the sample section of the Sample Collection Form. If the individuals were only collected from a limited segment of the sampling reach, place an "X" in the "No" box and explain in the "Explain" field on the form.

5-P. Wrap all primary fish together in a single piece of aluminum foil, making sure the **dull side of the aluminum foil is in contact with the fish**. Place the sample in a self-sealing plastic bag.

5-S. Wrap each fish of the secondary sample separately in aluminum foil, with the dull side of the foil in contact with the fish. Place all the wrapped individuals into a single self-sealing plastic bag.

6. Expel excess air and seal the bag. Wrap clear tape around the bag to seal and make a surface for each sample label..

7-P. Prepare two Fish Tissue sample labels (each having the same sample ID number [Figure 11-2]) by filling in the stream ID and the date of collection. Circle "PRIMARY" on each label. Record the sample ID number (barcode) in the primary sample section of the Sample Collection Form.

7-S. Prepare two Fish Tissue sample labels (each having the same sample ID number [Figure 11-2]) by filling in the stream ID and the date of collection. Circle "SECONDARY" on each label. Record the sample ID number (barcode) in the secondary sample section of the Sample Collection Form.

8. Attach the appropriate label to the tape surface of the bag. Cover the label with a strip of clear tape. Place the labeled bag into a second self-sealing plastic bag. Seal the bag and attach the second label to the outside of the appropriate bag. Cover the label with a strip of clear tape.
9. Place the double-bagged sample into a cooler containing bags of ice until shipment. Keep the sample **frozen** until shipment.

cies, depending upon the species used and the size range of individuals selected.

Individuals comprising the primary composite sample are wrapped together in aluminum foil and placed into a single plastic bag. Each individual comprising the secondary composite sample is wrapped separately, but all individuals are placed into a single plastic bag. Each composite sample is labeled as shown in Figure 11-1. Prepare two identical labels for each composite sample. Double-bag each sample, and place a label on each bag. Record information about each composite sample on page 2 of the Sample Collection Form (Figure 11-2). Make sure the sample ID numbers (barcodes) recorded on the collection form match those on the sample labels.

Tissue samples are stored in a cooler with several bags of ice. When using ice, double bag the ice and tape the last bag shut to prevent contamination of samples by melting ice. Store tissue samples frozen until they can be shipped (Section 3). Tissue samples can be stored and shipped with other samples requiring freezing (periphyton chlorophyll, periphyton biomass, periphyton APA, and sediment metabolism samples).

11.3 Equipment and Supplies

Figure 11-3 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 river 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.

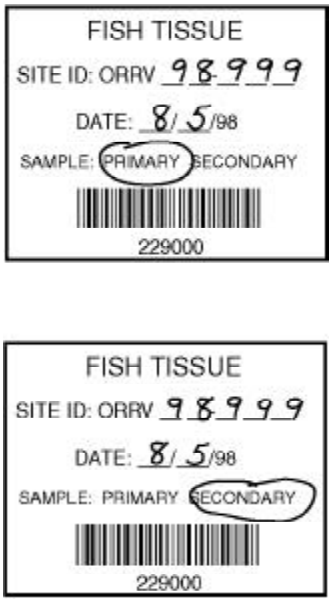

Figure 11-1. Completed sample labels for fish tissue contaminants.

Reviewed by (initial) DKA

SAMPLE COLLECTION FORM - RIVERS (continued)

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT ☐1 ☒2 ☐3

SITE ID: ORRV 98-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

CHEMISTRY AND MICROBIAL WATER SAMPLE (Chem: 4-L Cubitainer and 2 Syringes, Micro: Glass Bottle)

	SAMPLE ID (BARCODE)	TRANSECT	FLAG	COMMENTS
CHEMISTRY	243012	K		
MICROBIAL	243113	K		

SEDIMENT TOXICITY SAMPLES

SAMPLE ID (BARCODE)	FLAG	COMMENTS

FISH TISSUE SAMPLES - PRIMARY SAMPLE (min. 50g total wgt)

SAMPLE ID (BARCODE) → 243223

LINE	SPECIES CODE	COMMON NAME	NUMBER OF INDIVIDUALS	FLAG
P1	COTTZZZZ	UNKNOWN COTTIA	11	

IS COMPOSITE SAMPLE COMPOSED OF INDIVIDUALS COLLECTED FROM THROUGHOUT REACH? (X) → ☐YES ☒NO

IF NO, EXPLAIN: COLLECTED AT ONE PROFILE

FISH TISSUE SAMPLES - SECONDARY SAMPLE (where available; 5 individuals)

SAMPLE ID (BARCODE) → 243224

LINE	SPECIES CODE	COMMON NAME	TOTAL LENGTH (MM)	FLAG
S1	PTYCOREG	NORTHERN SQUAWFISH	220	
S2	PTYCOREG	NORTHERN SQUAWFISH	220	
S3	PTYCOREG	NORTHERN SQUAWFISH	265	
S4	PTYCOREG	NORTHERN SQUAWFISH	290	
S5	PTYCOREG	NORTHERN SQUAWFISH	215	

IS COMPOSITE SAMPLE COMPOSED OF INDIVIDUALS COLLECTED FROM THROUGHOUT REACH? (X) → ☒YES ☐NO

IF NO, EXPLAIN:

LINE	COMMENT OR FLAG EXPLANATION FOR FISH TISSUE

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

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SAMPLE COLLECTION FORM - RIVERS - 2

Figure 11-2. Sample Collection Form, showing information recorded for fish tissue samples.

Equipment And Supplies For Fish Tissue Contaminants		
Qty.	Item	
1	Plastic bucket for anesthetization	
4	Carbon dioxide tablets (Alka-Seltzer® or equivalent)	
1 roll	Clear tape for sealing tissue sample bags	
1	Pesola® portable scale, precision ±5g	
1 roll	Aluminum foil	
4	1-gallon self-sealing plastic bags	
1	Sample Collection Form	
2 sets	Fish tissue sample labels (each set with a different sample ID number [barcode])	
1 pkg.	Clear tape strips	
	Soft (#2) lead pencils to record data	
	Fine-point indelible markers to fill out labels	
1	Cooler with ice (double-bagged and taped)	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for fish tissue contaminants	

Figure 11-3. Equipment and supplies checklist for fish tissue contaminants.

Section 12
Visual Stream Assessments

Alan T. Herlihy[1]

After all other samples and field data have been collected, the field team makes a general visual assessment of the river, and performs a final check of the data forms and samples before leaving the river site (see Section 13). The objective of the visual river assessment is 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.

12.1 Visual Stream Assessment

The objective of the visual river assessment is to record field crew observations of catchment/river characteristics useful for future data interpretation, ecological value assessment, development of associations, and verification of stressor data. Observations and impressions of field crews are extremely valuable. Thus, it is important that these observations about river characteristics be recorded for future data interpretation and validation. 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 Comments section of the form.

Complete the assessment form after all other sampling and measurement activities have been completed. 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, reach characteristics, waterbody character, general assessment, and local anecdotal information. The procedure for conducting the visual assessment of the sampling reach is presented in Table 12-1. Record data and observations for each component of the assessment on the Assessment Form as shown in Figures 12-1 and 12-2.

[1]Dept. of Fisheries and Wildlife, Oregon State University, c/o U.S. EPA, 200 SW 35th St., Corvallis, OR 97333.

Table 12-1. Procedure for Conducting the Final Visual Assessment of a River.

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 river or driving/walking through the catchment to complete the remainder of the form.
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 river.
   - Recreational: The presence of organized public or private parks, campgrounds, beaches or other recreation areas around the river. If there are signs of informal areas of camping, swimming or boating around the river (e.g., swimming hole), record them as "primitive" parks, camping.
   - Agriculture: The presence of cropland, pasture, orchards, poultry, and/or livestock.
   - Industrial: Any industrial activity (e.g., canning, chemical, pulp), commercial activity (stores, businesses) or logging/mining activities around the river or in the catchment. Describe in more detail in the comments section.
   - Management: Any evidence of liming activity, water treatment, dredging or channelization, flow control structures, etc.

   Any oddities, or further elaboration should be recorded in the Comments section.
3. Reach Characteristics: For each type of riparian vegetation cover or land use category listed on the Assessment Form, estimate the proportion of the sampling reach immediately adjacent to the river that is affected. Place and "X" in the appropriate extent class box (Rare [< 5%], Sparse [5 to 25%], Moderate [25 to 75%], and Extensive [> 75%]) on the form.
4. Classify the overall water clarity within the sampling reach as clear, murky, or highly turbid. Place an "X" in the appropriate box on the "Water Clarity" line of the Assessment Form. If you believe that water clarity has been influenced by a recent storm event, also place an "X" in the "Storm Influenced" box.
5. 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.
5. Waterbody Character (cont.): Assign a rating to the river based on overall aesthetic quality, based on your opinion of how suitable the river 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.

   Add any comments you feel might aid data interpretation in the Comments Section.
6. General Assessment: record comments on wildlife observed, perceived diversity of terrestrial vegetation, and the estimated age class of forest (0 to 25 yr, 25 to 75 yr, or > 75 yr.) on the Assessment Form.
7. Local Anecdotal Information: Record any information regarding the past or present characteristics or condition of the river provided by local residents.

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 on a river, the rating for "Houses" would be low. If the river is in a suburban housing development, rate it as high. Simi-

Reviewed by (initial) DKA

ASSESSMENT FORUM - STREAMS/RIVERS

SITE NAME: BEAVER RIVER DATE: 8 / 5 / 98 VISIT ☐0 ☒1 ☐2 ☐3

SITE ID: ORRV 98-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

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	PARKS, CAMPGROUNDS	L M H	CROPLAND	L M H	INDUSTRIAL PLANTS	L M H	DIKING
L M H	MAINTAINED LAWNS	L (M) H	PRIMITIVE PARKS, CAMPING	L M H	PASTURE	L M H	MINES/QUARRIES	L M H	DRINKING WATER TREATMENT
L M H	CONSTRUCTION	(L) M H	TRASH/LITTER	L (M) H	LIVESTOCK USE	L M H	ORGANIC WASTES	L M (H)	ANIMAL PRESENCE
L M H	PIPES, DRAINS	L M H	OUTDOOR TOURS, GROUPS, CITIZENS	L M H	ORCHARDS	L M H	POWER PLANTS	L M H	DREDGING
L M H	DUMPING			L M H	POULTRY	L M H	LOGGING	L M H	CHANNELIZATION
(L) M H	ROADS			L M H	IRRIGATION PUMPS	L M H	EVIDENCE OF FIRE	(L) M H	WATER LEVEL FLUCTUATIONS
(L) M H	BRIDGES/CULVERTS					L M H	ODORS	L (M) H	FISH STOCKING
						L M H	COMMERCIAL	L M H	DAMS

REACH CHARACTERISTICS (percent of reach)

FOREST	☒ RARE (< 5%)	☐ SPARSE (5 TO 25%)	☐ MODERATE (25 TO 75%)	☐ EXTENSIVE (> 75%)
SHRUB	☐ RARE (< 5%)	☐ SPARSE (5 TO 25%)	☒ MODERATE (25 TO 75%)	☐ EXTENSIVE (> 75%)
GRASS	☐ RARE (< 5%)	☐ SPARSE (5 TO 25%)	☒ MODERATE (25 TO 75%)	☐ EXTENSIVE (> 75%)
WETLAND	☒ RARE (< 5%)	☐ SPARSE (5 TO 25%)	☐ MODERATE (25 TO 75%)	☐ EXTENSIVE (> 75%)
BARE GROUND	☐ RARE (< 5%)	☒ SPARSE (5 TO 25%)	☐ MODERATE (25 TO 75%)	☐ EXTENSIVE (> 75%)
MACROPHYTES	☒ RARE (< 5%)	☐ SPARSE (5 TO 25%)	☐ MODERATE (25 TO 75%)	☐ EXTENSIVE (> 75%)
AGRICULTURE - ROW CROP	☒ RARE (< 5%)	☐ SPARSE (5 TO 25%)	☐ MODERATE (25 TO 75%)	☐ EXTENSIVE (> 75%)
AGRICULTURE - GRAZING	☐ RARE (< 5%)	☐ SPARSE (5 TO 25%)	☒ MODERATE (25 TO 75%)	☐ EXTENSIVE (> 75%)
LOGGING	☒ RARE (< 5%)	☐ SPARSE (5 TO 25%)	☐ MODERATE (25 TO 75%)	☐ EXTENSIVE (> 75%)
DEVELOPMENT (RESIDENTIAL/ URBAN)	☒ RARE (< 5%)	☐ SPARSE (5 TO 25%)	☐ MODERATE (25 TO 75%)	☐ EXTENSIVE (> 75%)
WATER CLARITY	☒ CLEAR	☐ MURKY	☐ HIGHLY TURBID	☐ STORM INFLUENCED

WATERBODY CHARACTER (X ONE)

PRISTINE	☐ 6	☐ 5	☒ 4	☐ 3	☐ 2	☐ 1 — DISTURBED
APPEALING	☐ 5	☒ 4	☐ 3	☐ 2	☐ 1	UNAPPEALING

GENERAL ASSESSMENT (wildlife, vegetation diversity, forest age class (0-25 yrs, 25-75 yrs, >75)

ROCKY CANYON LANDS, SAGE/GRASS COVER — OCCASIONAL RIPARIAN TREE.
NO LWA, LIMITED FISH COVER, REGULATED FLOW / WATER LEVEL.

LOCAL ANECDOTAL INFORMATION:

Rev. 05/28/98 (strvasse.98) ASSESSMENT FORM - STREAMS/RIVER - 1

Figure 12-1. Assessment Form (page 1).

Reviewed by (initial) _____

ASSESSMENT FORUM - STREAMS/RIVERS (continued)													
SITE NAME:						DATE:	/	/ 98	VISIT	0	1	2	3
SITE ID:	O R R V	___ ___ -	___ ___ ___		TEAM ID (X):	1	2	3	4	5	6	7	8

Rev. 05/29/98 (srvasse.98)

ASSESSMENT FORM - STREAMS/RIVER - 2

Figure 12-2. Assessment Form (page 2).

larly, a small patch of clear cut logging on a hill overlooking the river would be rated as low. Logging activity right on the river shore, however, would be rated as high.

When assessing reach characteristics, make your best estimate as to the percent of the sampling reach (100 or 40 channel widths) that had each type of listed riparian zone land use immediately adjacent to the river. Also rate the water clarity, including whether you believe the clarity is influenced by recent storm events (see Section 4).

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 river a "5" rating if it is pristine, with no signs of any human development. A rating of "1" indicates a river which is totally developed (e.g., the entire river is lined with houses, or the riparian zone has been removed). For aesthetics, base your decision on any factor about the river that bothers you (e.g., trash, algal growth, weed abundance, overcrowding).

The general assessment component includes any observations that will help in data interpretation in the pertinent section. General assessment comments can include comments on wildlife observed, diversity of terrestrial vegetation, age class of forest, or any other observation. Comments from locals are often useful and should be recorded in the "Local Anecdotal Information" section. The back side of the form (Figure 12-2) is available for general comments.

12.2 Equipment and Supplies

Figure 12-3 is a checklist of the supplies required to complete the rapid habitat and visual stream assessments. 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 river 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.

Equipment and Supplies for Visual Stream Assessments

Qty.	Item	
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 12-3. Checklist of equipment and supplies required for visual assessments

Section 13
Final Site Activities

James M. Lazorchak[1] and Daniel K. Averill[2]

After the boat crews have safely reached the take-out location, team members begin final site activities. Composite samples for periphyton (Section 7) and benthos (Section 9) are processed. The incubation for sediment metabolism (Section 8) is initiated, if not already started. Equipment and supplies are unloaded from rafts, vehicles are shuttled, equipment and supplies are loaded in the vehicles, rafts are loaded onto the trailer, and data forms, labels, and samples are inspected.

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, delivering and/or shipping samples.

When reviewing field data forms, ensure that all required data forms for the river have been completed. Confirm that the site 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. A check by a team member that has not filled out the sheets for a particular section might be the best person to review the field data forms before leaving the site.

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.

Keep equipment and supplies organized so they can be inventoried using the equip-

[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.

ment and supply checklists presented in Appendix A. Clean up the take-out site and transport all waste material out of the area.

If samples are to be shipped by FEDEX, check the 1 800 number for the nearest Motel or location that samples can be left for pickup. In most eastern states, FEDEX will pick up coolers at most motels and ship the following morning. This can help reduce travel time to the next sample site. FEDEX also will ship on Saturdays for Sunday or Monday pickups as of the year 2000. Check their 1 800 number for locations that will accept Saturday drop off and Sunday delivery.

Appendix A
Equipment and Supply Checklists

Field Data Forms and Sample Labels A-2

Office Supplies and Tools A-3

Personal Equipment and Supplies A-4

Chemicals A-5

Packing and Shipping Supplies A-5

Site Verification and Sampling Reach Layout A-6

Water Chemistry and Microbiology A-6

Physical Habitat A-7

Periphyton A-7

Sediment Metabolism A-8

Benthic Macroinvertebrates A-8

Aquatic Vertebrates and Fish Tissue Contaminants A-9

Field Data Forms and Sample Labels

Number per site	Item	
1	Verification Form	
1	Sample Collection Form	
1	Field Measurement Form	
11 + extras	Channel/Riparian Transect Form	
11 + extras	Thalweg Profile Form	
1	PHAB Commentes Form	
10 + extras	Vertebrate Collection Form	
1 + extras	Vergebrate Length Recording Form	
1	Assessment Form for visual river assessment	
1	Field Sample Shipment Tracking Form	
3	Water chemistry labels (same ID number)	
1	Microbial label	
4	Periphyton labels (same ID number)	
5	Sediment metabolism labels (different ID numbers)	
2	Composite Benthic sample labels, with preprinted ID numbers (barcodes)	
4	Composite Benthic sample labels without preprinted ID numbers	
1	Sheet of preprinted aquatic vertebrate jar labels (4) and voucher bag tags (36), all with same preprinted sample ID number (barcode)	
2 sets	Fish tissue sample labels (2 labels per set; each set with a different sample ID number [barcode])	
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 and general information for scheduled river site	
1	Topographic map with "X-site," reach boundaries, and launch points marked	
1	Site information sheet with map coordinates, bank to be sampled, elevation of X-site, and other general information	
1	Portable file folder used to organize field and administrative forms	
1 ea.	Map wheel, calculator, metric ruler, shoulder bag, and field notebook	
1	Sampling itinerary notebook	
1	Safety log and/or personal safety information for each team member	
4	Covered clipboards or forms holders	
12	Soft (#2) lead pencils	
6	Fine-tip indelible markers	
1 pr	Scissors for cutting labels	
1	Pocket knife or multipurpose tool	
1	Toolbox with basic tools needed to maintain/repair sampling gear (other than electrofishing equipment)	

Personal Equipment and Supplies

Number per site	Item	
2	14 ft. inflatable rafts with custom frames	
6	Oars	
2	Extra oar locks	
2	Raft pump - AIR	
1	Raft pump - ELECTRIC	
3	Raft patch kits	
1 pr/person	Felt-soled wading boots + neoprene booties.	
1 per person	PFD's with pockets	
2 pair	Polarized sunglasses and leather gloves	
1	First aid kit, eye wash unit, sunscreen, whistle, antibacterial hand wash	
1 per person	Rain gear	
1	Water purifier	
2	Throw bags and dry bags	
2	Pruning saws	
1	5 gal. water jug	
2	Pulaski's and shovels	
2	Booster cables	
	Tie-down straps, ropes, bungee cords	

Chemicals

Number per site	Item	
1 pr	Safety glasses	
2 pr	Chemical-resistant gloves	
1	Laboratory apron, resistant to ethanol and formalin	
1	Cooler or large tote for transporting ethanol and samples	
2 gal	95% ethanol	
1	Cooler or large tote for transporting formaldehyde/formalin	
2 gal	10% (buffered) formalin solution	
1 gal	Gasoline for electrofishing generator in approved container	
2 jars	Methyl Ethyl Ketone (MEK) for patching rafts	

Packing and Shipping Supples

Number per site	Item	
2 bags	Ice	
1 box	1-gal heavy-duty self-sealingn (e.g., with a zipper-type closure) plastic bags	
1 box	30-gal plastic garbage bags	
1 roll	Clear tape for sealing tissue sample bags and shipping containers	
2 pkg	Clear tape strips for covering labels	
4 rolls	Plastic electrical tape	
3	Insulated shipping containers for samples (heavy plastic coolers)	
1	Plastic container with snap-on lide	
1	Cooler with bags of ice to store frozen samples	
2	Containers suitable to transport and/or ship samples preserved in formalin or ethanol (coolers)	
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	Laser rangefinder (400 yard range) and clear waterproof bag (dry bag)	
1	50 m fiberglass measuring tape with reel	
1	Dossier of site and access information	
1	Waterproof camera and film	
1	Topographic map and gazetteer	
1 ea	Map wheel, calculator, metric ruler	

Water Chemistry and Microbiology

Number per site	Item	
1	Dissolved oxygen/Temperature meter with probe, manual, & storage case	
1	DO repair kit containing additional membranesand probe filling solution	
1	Conductivity meter with probe, operating manual, and padded storage case	
	Extra batteries for dissolved oxygen and conductivity meters	
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	500-mL plastic bottle of deionized water to store conductivity probe	
1	Field thermometer	
1	500 mL plastic beaker with handle (in clean plastic bag)	
1	4-L cubitainer	
2	60-mL plastic syringes	
1	200 mL square glass microbial bottle	
1	Plastic container with snap-on lid to hold filled syringes	
2	Syringe valves	

Physical Habitat

Number per site	Item	
1	Surveyor's telescoping rod (round profile, metric scale, 7.5 m extended) for depth measurements and substrate estimation	
1	Clinometer (or Abney level) with percent and degree scales	
1	Convex spherical canopy densiometer, modified with taped "V"	
1	Bearing compass (Backpacking type)	
1 roll	Colored surveyor's plastic flagging	
1	Meter stick for bank angle measurements	
1	SONAR depth sounder - narrow beam (16 degrees)	

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° angle	
1	1-L wash bottle for stream water	
1	1-L wash bottle containing deionized water	
2	500-mL plastic bottles for composite samples	
1	60 mL plastic syringe with a 3/8" hole bored into the end	
4	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	
1	Filtration unit, including filter funnel, cap, filter holder, and receiving chamber	
1	Hand-operated vacuum pump with length of flexible plastic tubing	
2	Aluminum foil squares (3" x 6")	
1	Small syringe or bulb pipette for dispensing formalin	
1	Collapsible bucket	

Sediment Metabolism

Number per site	Item	
1	Small scoop sampler for sediments	
1	Wide-mouthed plastic jar labeled "COMPOSITE SEDIMENT SAMPLE". If sediment is only being collected for metabolism samples, a 250-mL jar is sufficient.	
1	YSI Model 95 Dissolved Oxygen meter	
1 set	Spare batteries for DO meter	
1	Small plastic spoon or spatula to transfer sediment from the composite sample container to respiration tubes	
5	50-mL, screw-top, centrifuge tubes	
1	50-mL screw-cap centrifuge tube labeled "BLANK"	
1	Small cooler used as incubation chamber	
1	1,000-mL plastic beaker to holding centrifuge tubes during incubation	

Benthic Macroinvertebrates

Number per site	Item	
1	Modified kick net (closed bag with 595 µm mesh) and 4-ft handle (Wildco #425-J50-595)	
	Spare net(s) for the kick net sampler or extra sampler	
2	Drift nets, 595 µm mesh, closed end	
1	Sieve bucket, 595 µm mesh openings	
2 pr	Watchmakers' forceps	
1	Wash bottle, 1-L capacity	
1	Small spatula, spoon, or scoop to transfer sample	
1	Funnel, with large bore spout	
10	Sample jars, plastic with screw caps, 500 mL and 1 L capacity, suitable for use with ethanol	
2	Buckets, plastic, eight to ten quart capacity	
1	Stopwatch	
1 pkg	Kimwipes in small self-sealing plastic bag	

Aquatic Vertebrates and Fish Tissue Contaminants

Number per site	Item	
1 set	Electrofishing equipment - cathode and anode droppers, other frame mounted electrical boxes and connections	
1	Electrofishing control box with all connectors	
2	Anodes and cathodes (SPARE)	
1	Generator and filled gas can + rag	
2	Dip nets, Long handled	
1	Dip net, Short handled	
1	Live well cooler	
1 pr	Heavy-duty rubber gloves for electrofishing	
2	Fish measuring board and rulers	
2	Portable scale, precision ±5g to weigh tissue samples	
2	Buckets (5 gallon)	
	Tools for electrofishing assembly	
1	Fire extinguisher	
1 set	Taxonomic reference books and keys for fishes and amphibians of the region	
1	List of vertebrate species codes and common names	
1	List of external anomaly codes	
15-20	Small nylon mesh bags for holding voucher specimens (bags can also be constructed from sections of nylon stockings or panty hose)	
1	Small fillet knife or scalpel for preparing larger voucher speciments for preservation	
2 ea	1, 2 or 4 L screw-top plastic jars for voucher samples	
1	Plastic bucket for anesthetization	
2 gal	10% (buffered) formalin solution	
4	carbon dioxide tablets (Alka-Seltzer® or equivalent)	
1 roll	Aluminum foil	

Appendix B
Quick Reference Guides

The following pages are tabular summaries of different field activities and procedures described in this manual. These were developed by the principal investigators for each ecological indicator to provide a field team with a quick way to access information about each procedure. They are intended to be laminated and taken to the river site after the crew has been formally trained in the detailed procedures as presented in the manual. They are arranged here in the general sequence of their use in the field.

Quick Reference Guide For Initial Site Activities B-2

Quick Reference Guide For Water Chemistry And Microbiology B-4

Quick Reference Guide For Physical Habitat Characterization B-7

Quick Reference Guide For Periphyton B-13

Quick Reference Guide For Sediment Metabolism B-15

Quick Reference Guide For Benthic Macroinvertebrates B-16

Quick Reference Guide For Aquatic Vertebrates B-20

Quick Reference Guide For Fish Tissue Contaminants B-23

Quick Reference Guide for Initial Site Activities

1. Find the river location in the field corresponding to the "X" on 7.5" topo map (X-site). Crews should use all available means to insure that they are at the correct site, as marked on the map, including: 1:24,000 USGS map orienteering, topographic landmarks, county road maps, local contacts, boat launches, and global positioning system (GPS) confirmation of site latitude and longitude.
2. Classify the site, AT THE X-SITE, as:

NON-TARGET	No Stream Channel
	Impounded River
	Marsh/Wetland
TARGET	Regular Wadeable Stream
	Regular - Partial Boatable and Wadeable Combination
	Regular Boatable
	Intermittent Stream
	Dry Channel
	Altered Channel (channel different form map representation)
INACCESSIBLE	Physical Barriers (Physically unable to reach the X-site)
	No Permission

   Record class on Site Verification form, do not sample Non-target or inaccessible sites. Take samples from Target sites as discussed in field operations and methods manual.
3. At the launch site, unload the rafts and all equipment, supplies, and sample containers. Shuttle the vehicles.
4. Using a laser rangefinder, measure the river width in several places, specifically the X-site and the two boat launches. Record the width on the site verification form. Lay out a sample reach with a length of 100 times the river width by rolling a map wheel on the topographic map and marking the reach boundaries.
5. Do a reconnaissance of the sample reach while shuttling vehicles, obtaining widths, and evaluating launch sites. Extensive shallows, large log jams, absence of launch sites or vehicle access, and hazardous whitewater may all preclude rafting.
6. Determine the float distance, if any, from the put-in to the first transect (Transect "A"), and from the last transect (Transect "K") to the take-out.
7. Using a laser rangefinder at the most upriver transect (Transect "A"), measure 10 channel widths downriver to the next transect (Transect "B"). This distance is a profile.
8. Sample odd numbered site ID's along the left shore (facing downriver); sample even numbered sites along the right shore.

NOTE: 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 accommodate river access or to avoid river hazards or obstacles. If the beginning or end of the reach cannot be sampled due to obstacles or hazards, 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, access points may necessitate sliding the reach. 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 such as bridges, rip rap, or channelization.

Quick Reference Guide for Water Chemistry and Microbiology

I. Equipment to Carry in Field for Water Chemistry and Microbiology

Rinse/Test bottles of QCCS in self-sealing plastic bag
D.O./Temperature/Conductivity Meter
Field Forms
One 500-mL plastic beaker with handle, in clean self-sealing plastic bag
One cubitainer in clean self-sealing plastic bag (barcode label attached)
Two 60-mL syringes in a plastic container (each one with a bar code label attached)
One 200-mL sterile square glass microbial bottle (barcode label attached)
Two syringe valves in the plastic container
Plastic cooler and several bags of ice
Opaque garbage bag
Electricians tape

II. Extra Equipment to Carry in Vehicle

Back-up labels, forms, cubitainers, syringes, syringe valves, and microbial bottles

III. Daily Activities after Sampling

1. Check that cubitainer lid is on tight, has a flush seal, and is taped. Also tape the microbial cap.
2. Prepare the sample for shipping (label and seal cooler, replace ice as close as possible to shipping time) OR direct delivery to the laboratory.
3. Call Overnight shipping company to arrange pick-up of cooler.
4. Rinse the sampling beaker with deionized water three times.
5. Make sure field meters are clean and are stored with moist electrodes.
6. Label the next days sample containers (cubitainer, syringes, and microbial bottle), pack cubitainer and sample beakers in clean self-sealing plastic bag, and pack two syringes, syringe valves, and a microbial bottle in a plastic container with a snap-on lid.

(continued)

Quick Reference Guide for Water Chemistry and Microbiology (continued)

Summary of Site Procedure for Water Chemistry and Microbiology

I. Collect Water Sample

A. Make sure cubitainers and syringes are labeled and have the same barcode ID.
B. Make sure the microbial bottle is labeled with a barcode ID.
C. Cubitainer, syringe, and microbial samples are taken only from the **middle of the flowing river at the last sample transect (Transect "K").**
D. Rinse the 500-mL sample beaker three times with river water from the mid-channel.
E. Rinse cubitainer three times with 25-50 mL of river water, using the sample beaker. Rinse cubitainer lid with river water.
F. Fill cubitainer with river water using the 500 mL sample beaker. Expel any trapped air and cap the cubitainer. Make sure that the lid is seated correctly and that the seal is tight. **DO NOT EXPAND CUBITAINER BY BLOWING IN IT**.
G. Rinse each of the two, 60-mL syringes three times with 10-20 mL of river water .
H. Fill each of the syringes with river water from mid-river by slowly pulling out the plunger. If any air gets into the syringe, discard the sample and draw another.
I. Invert the syringe (tip up) and cap the syringe with a syringe valve. Open the valve, tap the syringe to move any air bubbles to the tip, and expel any air and a few mL of water. Make sure there is 50-60 mL of river sample in the syringe. Close the valve and place the syringes in their transport container.
J. Keep the microbial bottle closed until filled. Do not contaminate inner surface of cap or bottle. **Fill the bottle without rinsing.**
K. Take sample from upriver side of boat by holding bottle near base and plunge neck downward below water's surface. Turn bottle until neck points slightly upward and mouth is directed toward the current.
L. After sample is collected, leave ample air in the microbial bottle (~ 2.5 cm) and tape the cap tight.
M. Place the cubitainer, syringes, and microbial bottle on ice in a cooler to keep cool (keep dark as well) until shipment.

II. In Situ Measurements

A. Conductivity
   1. Turn on and check the zero and red line (if applicable) of the conductivity meter.
   2. Measure and record the conductivity of the QCC solution. Rinse the probe in the "Rinse" bottle of QCC solution before immersing in the "Test" bottle of QCC solution.
   3. Measure and record river conductivity in mid-river **AT EACH TRANSECT**.

B. Dissolved Oxygen/Temperature
   1. Calibrate the DO meter following meter instructions.
   2. Measure the DO in mid-river at the **middle of the flowing river of the last sample transect (Transect "K")**. If water velocity is slow, jiggle the DO probe as you take the reading. Measure temperature mid-river **AT EACH TRANSECT**.

Quick Reference Guide for Physical Habitat Characterization

Field Summary: P-hab Layout And Workflow

1. Habitat Sampling Layout:

   A. Thalweg Profile: At 10 equally spaced intervals between each of 11 channel cross-sections (100 along entire reach):
      * Classify habitat type, record presence of backwater and off-channel habitats. (10 between cross-sections, 100 total)
      * Determine dominant substrate visually or using sounding rod. (10 between cross-sections, 100 total)

      At 20 equally spaced intervals between each of 11 channel cross-sections (200 along entire reach):
      * Tally mid-channel snags (20 between cross-sections, 200 total).
      * Measure thalweg (maximum) depth using Sonar or rod (20 between cross-sections, 200 total)

   B. Littoral/Riparian Cross-Sections: @ 11 stops ("transects") at equal intervals along reach length

2. Work Flow: In a single mid-river float down a 100 channel-width reach

   - At the upriver start point (Transect "A") and along the designated shoreline: Move boat in a "loop" within a 10 x 20 m littoral plot, measuring 5 littoral depths and probing substrate. Also estimate dominant and subdominant littoral substrate within the "loop." After the "loop," estimate areal fish cover within and tally LWD within or partially within the 10 x 20 m plot. Record densiometer measurements at the bank (up, down, left, right), and choose bank angle class, and estimate bankfull height, width and channel incision (for BOTH banks). Estimate and record distance to riparian vegetation on the chosen bank. Estimate visually riparian vegetation cover for the 10 x 20 m plot on BOTH sides of channel (plot starts at bankfull , continues back 10m from bankfull). For the largest riparian tree, estimate Dbh, height, species, distance from river edge. Visually tally human disturbances in the same plot as riparian vegetation. No bearing or slope at first cross section.
   - Proceed downriver between Transects "A" and "B", making 20 thalweg depth measurements and substrate snag probes; also classify habitat types. Estimate thalweg distance intervals by tracking boat lengths or channel-widths. One person measures thalweg depths and the other records those measurements. At the 20th thalweg measurement location (close to Transect "B"), backsite a compass bearing in mid-channel, then distance and % slope back to your visual "mark" on the bank at the previous transect ("A").
   - When you complete 20 thalweg intervals and reach one of 11 cross sections, stop at the chosen shore and take out a new Channel/Riparian Transect Form for Transect "B". Repeat all the Channel/Riparian measurements at this new location.
   - Repeat the cycle of thalweg and cross section measurements until you reach transect 11 ("K") at the downriver end.

(continued)

Quick Reference Guide for Physical Habitat Characterization

Field Summary: Components of P-Hab Protocol

Thalweg Depth Profile, Mid-Channel Snags, Hab. Type, Off-channel, Substrate:

- At 20 approximately equal spaced intervals between each of 11 channel cross-sections (200 along entire reach) while floating mid-channel:
  - Measure max. depth ("Thalweg") at each increment
  - Tally mid-channel snags
- At 10 approximately equal spaced intervals between each of 11 channel cross-sections (100 along entire reach) while floating mid-channel:
  - Classify habitat type and off-channel habitats
  - Determine dominant substrate

Channel and Riparian Cross-Sections:

- Measurements: Wetted width, mid-channel bar width, gradient (clinometer or Abney level), sinuosity (compass backsite), riparian canopy cover (densiometer).
- Visual Estimates: Bankfull width, bankfull height, incision height, bank angle, shoreline substrate, large woody debris, areal cover class and type of riparian vegetation in Canopy, Mid-Layer and Ground Cover; areal cover class of fish cover features, aquatic macrophytes, and filamentous algae; presence and proximity of human disturbances.

(continued)

Quick Reference Guide for Physical Habitat Characterization (continued)

Field Summary: Rip. Veg., Human Disturb., In-Channel Cover:

- Observations upriver 10 meters and downriver 10 meters from each of the 11 cross-section transects.
- For riparian vegetation and human disturbances, include the visible area from the river back a distance of 10m (30 ft) shoreward from both the left and right banks. If the wetted channel is split by a mid-channel bar, the bank and riparian measurements shall be for each side of the channel, not the bar.
- Three vegetation layers:
  CANOPY LAYER (>5 m high)
  UNDERSTORY (0.5 to 5 m high)
  GROUND COVER layer (<0.5 m high)
- Canopy and Understory Vegetation Types:
  (**D**eciduous, **C**oniferous, Broadleaf **E**vergreen, **M**ixed, or **N**one) in each of the two taller layers (Canopy and Understory). "Mixed" if more than 10% of the areal coverage made up of the alternate type.
- Areal Cover Classes for Vegetation and In-Channel Cover:
  0: (absent -- zero cover)
  1: (sparse -- cover <10%)
  2: (moderate -- cover 10-40%)
  3: (heavy -- cover 40-75%)
  4: (very heavy -- cover >75%).
- Tallying Human Disturbances:
  B: PRESENT within the defined 20 m river segment and located in the river or on the wetted or bankfull river
  C: CLOSE - Present within the 10 x 20 m riparian plot area, but above bankfull level
  P: PRESENT, but observed outside the riparian plot area
  0: NOT PRESENT within or adjacent to the 20 m river segment or riparian plot

Quick Reference Guide for Physical Habitat Characterization (continued)

Field Summaries: Substrate And Woody Debris Size Classes

Observe bottom substrates within a 10m swath along the 20m of channel margin that is centered on each transect location. Determine and record the dominant and subdominant substrate size class at 5 systematically spaced locations estimated by eye within this 10m x 20m plot and 1m back from the waterline.

Substrate Size Classes:

RS	Bedrock (Smooth)	>4000 mm	smooth surface rock or hardpan (bigger than a car)
RR	Bedrock (Rough)	>4000 mm	Rough surface rock (bigger than a car)
HP	Hardpan	>4000 mm	(consists of firm, consolidated fines)
BL	Boulders	>250 to 4000 mm	(basketball to car size)
CB	Cobbles	64 to 250 mm	(tennis ball to basketball size)
GC	Gravel(Coarse)	16 to 64 mm	(marble to tennis ball size)
GF	Gravel (Fine)	2 to 16 mm	(ladybug to marble size)
SA	Sand	0.06 to 2 mm	(smaller than ladybug size, but visible as particles - gritty between fingers).
FN	Fines	<0.06 mm	Silt-Clay-Muck (not gritty between fingers)
WD	Wood	Regardless of Size	Wood or other organic material
OT	Other	Regardless of Size	Metal, Tires, Car bodies, asphalt, concrete, etc. (Describe in comments if you enter "OT").

Large Woody Debris Size Classes:

LWD Definition: Diameter (small end) > 30 cm (>1 ft.)
Length > 5 m (> 15 ft) -- count only part with diam > 30 cm.

Two Tallys:

(1) LWD at least partially in the baseflow channel (wetted).
(2) LWD presently dry but contained within the bankfull (active) channel, and LWD spanning above the active channel.

Size Categories for Tally (12 potential combinations):

Diameter (large end):		Length:	
0.3 to <0.6 m	(1 to 2 ft.)	5 - <15 m	(16 - 49 ft)
0.6 to <0.8 m	(2 to 2.6 ft)	15 - <30 m	(49 - 98 ft)
0.8 to <1.0 m	(2.6 to 3.3ft)	>30 m	(>98 ft)
>1.0 m	(> 3.3ft)		

(continued)

Quick Reference Guide for Physical Habitat Characterization (continued)

Field Summary: Habitat Classification At Channel Unit Scale

Channel Unit Habitat Classes[a]

Class (Code)	Description
Pools (PO):	Still water, low velocity, smooth, glassy surface, usually deep compared to other parts of the channel:
Plunge Pool	Pool at base of plunging cascade or falls.
Trench Pool	Pool-like trench in the center of the stream
Lateral Scour Pool	Pool scoured along a bank.
Backwater Pool	Pool separated from main flow off the side of the channel.
Dam Pool	Pool formed by impoundment above dam or constriction.
Glide (GL)	Water moving slowly, with a smooth, unbroken surface. Low turbulence.
Riffle (RI)	Water moving, with small ripples, waves and eddies -- waves not breaking, surface tension not broken. Sound: "babbling", "gurgling".
Rapid (RA)	Water movement rapid and turbulent, surface with intermittent whitewater with breaking waves. Sound: continuous rushing, but not as loud as cascade.
Cascade (CA)	Water movement rapid and very turbulent over steep channel bottom. 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
Off-Channel Areas	Side-channels, sloughs, backwaters, and alcoves that are separated from the main channel.

[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.

Field Summary: P-hab Problem Areas

Mid-channel Bars: dry at baseflow, inundated at bankfull flow.

> Measure wetted width across and over mid-channel bars, but record bar width in the column provided on the Channel/Riparian Transect Form.

Islands: as high as the surrounding flood plain; dry even at bankfull flow.

> Measure only the width of the main channel between island and shore

Both bars and islands cause the river to split into side channels. When a bar or island is encountered along the thalweg profile, choose to navigate and survey the channel that carries the most flow.

Side channels (off-channel):

> When present, check the "Off-channel" column on the Thalweg Profile Form. Begin checking at the point of divergence continuing until convergence. In the case of a slough or alcove, "off-channel" checkmarks should continue from the point of divergence downriver to where it is no longer evident.

Dry and Intermittent rivers:

> Record zeros for depth and wetted width in places where no water is in the channel. Record habitat type as dry channel (DR).

Quick Reference Guide For Periphyton

Field Equipment

1. Large funnel (15-20 cm diameter).
2. Scrape area delimiter (3.8 cm diameter pipe, 3 cm tall).
3. Stiff-bristle toothbrush with handle bent at 90° angle.
4. Wash bottle.
5. Collection bottle to catch removed periphyton.
6. 60 mL syringes with 3/8" hole bored into the end.
7. 50 mL centrifuge tubes or similar sample vials.
8. Formalin.
9. Glass-fiber filters (0.45 m average pore size) for chlorophyll a and biomass (AFDM).
10. Forceps for filter handling.
11. Millipore®-type filtration apparatus with plastic or stainless steel filter base, and Nalgene® funnel and suction flask.
12. Nalgene® hand-operated vacuum pump (need one additional pump as a backup).
13. Aluminum foil.
14. Ice chest.

Field Protocols

1. Periphyton samples are collected from the designated shoreline at each transect location.
2. Collect a sample of substrate (rock or wood) that is small enough (< 15 cm diameter) and can be easily removed from the river. Place the substrate in a plastic funnel which drains into a 500-mL plastic bottle with volume graduations marked on it.
3. 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.
4. Fill a wash bottle with river water. Using a minimal volume of water from this bottle, wash the dislodged periphyton from the funnel into the 500-mL bottle.
5. If no coarse sediment (cobbles or larger) are present, collect soft sediments by vacuuming the upper 1 cm of sediments confined within the 12-cm^2 sampling ring into a 60-mL syringe.

6 Place the sample collected at each sampling site into the single 500-mL bottle to produce the composite index sample.

7. After samples have been collected from all 11 transects, thoroughly mix the 500-mL bottle regardless of substrate type.
8. Record total volume of composited sample before proceeding to the next step!
9. Four subsamples will be taken from each composite sample. These are:
   a. Identification/Enumeration
      1) Withdraw 50 mL of mixed sample and place in a labeled sample vial (50-mL centrifuge tubes work well). Cover label with clear tape.
      2) Preserve sample with 2 mL of 10% formalin. Gloves should be worn.
      3) Tightly cap tube and tape with electrical tape.
   b. Chlorophyll *a*
      1) Withdraw 25 mL of mixed sample and filter onto a glass-fiber filter (0.45 um pore size) using a hand-operated vacuum pump. (Note: for soft-sediment samples, allow grit to settle before withdrawing sample).
      2) Fold filter so that the sample on the filter surface is folded together, wrap in aluminum foil, and affix the tracking label to the outside, and seal with clear tape.
      3) Freeze filter as soon as possible by placing it in a freezer.
      4) Store frozen for laboratory analysis.

(continued)

Quick Reference Guide For Periphyton (continued)

c. Ash Free Dry Mass (AFDM)
   1) Withdraw 25 mL of mixed sample and filter onto a glass-fiber filter (0.45 um pore size) using a hand-operated vacuum pump. (Note: for soft-sediment samples, allow grit to settle before withdrawing sample).
   2) Fold filter so that the sample on the filter surface is folded together, wrap in aluminum foil, and affix the tracking label to the outside, and seal with clear tape.
   3) Freeze filter as soon as possible by placing it in a freezer.
   4) Store frozen for laboratory for analysis.

d. Alkaline/Acid Phosphatase
   1) Withdraw 50 mL of mixed sample and place in a labeled sample vial (50-mL centrifuge tubes work well). Cover label with clear tape.
   2) Tightly cap tube and tape with electrical tape.
   3) Freeze sample as soon as possible by placing it on dry ice.
   4) Store frozen for laboratory analysis.

Quick Reference Guide for Sediment Metabolism

Field Equipment

1. Ice chest for floating centrifuge tubes during incubation
2. 1000 mL Nalgene© beaker for holding centrifuge tubes during incubation.
3. Small scoop sampler for sediments.
4. 50-mL, screw-top, centrifuge tubes.
5. Digital dissolved oxygen meter (e.g. YSI 95).
6. Spare batteries for D.O. meter.
7. Permanent markers for labeling tubes.
8. Sample labels and field data sheets.
9. Ice chest with ice for sample freezing.

Field Protocols

Dissolved Oxygen Meter Calibration (for YSI model 95)

1. Calibrate meter using the water-saturated atmosphere chamber described in the meter's operations manual. Allow at least 15 minutes for the probe to equilibrate before attempting to calibrate.

Sediment Collection and Experimental Set-up

1. Collect and combine fine-grained, surface sediments (top 2 cm) from all depositional areas at each transect (Transects A-K) along the designated shoreline of the river reach.
2. Fill ice chest 2/3 full with river water and record temperature and dissolved oxygen (D.O.).
3. Thoroughly mix composite sediment sample.
4. Place 10 mL of sediment in each of 5 labeled, 50 mL screw-top centrifuge tubes.
5. Fill each tube to the top (no head space) with stream water from the ice chest and seal.
6. Fill one additional tube with stream water only to serve as a blank.
7. Incubate tubes in closed ice chest for 2 hours.
8. Measure D.O. in each tube, including the blank.
9. Decant overlying water and save sediment.
10. Tightly seal tubes and freeze as soon as possible.
11. Store frozen for laboratory analysis.

Quick Reference Guide For Benthic Macroinvertebrates

Table I. Base Protocols for Collecting Macroinvertebrates

1. Set drift net assembly(s) near the put-in or take-out location.
2. Shore kick net samples are collected at each of the transect locations along the designated shoreline. Drift nets collect samples during the sampling day while the crew floats the river.
3. If riffle or run, use the kick net protocol in Table II. If pool, use the kick net protocol in Table III or hand pick for 60 seconds if kick net cannot be used.
4. Go to next downriver transect and repeat. Combine all riffle and pool samples into one bucket. Check net after each sample for clinging organisms and transfer to bucket.
5. After a sample is collected from each of the transects and all kick net samples are combined into one bucket, obtain a composite sample as described in Table V.
6. Drift net(s) procedures are described in Table IV. Processing is described in Table V.
7. Preserve and label each sample as described in Table VI.

Table II. Procedures for Riffles and Glides using Kick Net Sampler

1. Attach four foot pole to the sampler.
2. Position sampler quickly and securely on river bottom with net opening upriver.
3. Hold the sampler in position on the substrate while checking for snails and clams in an area of about 0.5 m2 in front of the net; kick the substrate vigorously for about 20 seconds in front of the net.
4. Inspect and rub off with the hands any organisms clinging to the rocks, especially those covered with algae or other debris.
5. Remove the net from the water with a quick upriver motion to wash the organisms to the bottom of net.
6. Rinse net contents into a bucket containing one or two gallons of water by inverting the net in the water.
7. Inspect the net for clinging organisms. With forceps remove any organisms found and place them into the bucket.
8. Large objects (rocks, sticks, leaves, etc.) in the bucket should be carefully inspected for organisms before discarding.
9. After all transects are sampled and all samples are combined in ONE bucket (riffle/glide + pool), obtain a composite sample as described in Table V.

Table III. Procedures for Pools using the Modified Kick Net Sampler

1. Attach four-foot pole to the sampler.
2. Inspect about ½ square meter of bottom for any heavy organisms, such as mussels and snails, which have to be hand picked and placed in the net.
3. While disturbing about 0.5 m2 of substrate by kicking, collect a 20-second sample by dragging the net repeatedly through the area being disturbed. Keep moving the net all the time so that the organisms trapped in the net will not escape.
4. After 20 seconds remove the net from the water with a quick upriver motion to wash the organisms to the bottom of the net.
5. Rinse net contents into a small bucket of water (about one or two gallons) by inverting the net in the water.
6. Inspect the net for clinging organisms. With forceps remove any organisms found and place them in the bucket.
7. Large objects in the bucket should be carefully inspected for organisms which are washed into the bucket before discarding.
8. After all transects are sampled and all samples are combined in ONE bucket (pool + riffle/glide), obtain a composite sample as described in Table V.

Table IV. Collection Procedures for Drift Nets

1. Do not use drift nets for large rivers with currents less than 0.05 m/s.

(continued)

2. Install the net at the downriver end of the reach (Transect K). The take-out location is 1st choice, otherwise the put-in location - whichever is closer to the reach.
3. Set the nets in the main flow of the river (avoid backwaters, eddies, river margins) at depths of about 25 cm from the bottom substrate and 10 cm below the water's surface.
4. Anchor the net assembly using anchors and cables. Record START TIME.
5. Measure the current velocity at the entrance of the net, using a neutrally buoyant object as follows:
    a. Measure out a straight segment of the river reach just upstream of the drift net location in which an object can float relatively freely and passes through within about 10 to 30 seconds.
    b. Select an object that is neutrally buoyant, like a small rubber ball or an orange; it must float, but very low in the water. The object should be small enough that it does not "run aground" or drag bottom.
    c. Time the passage of the object through the defined river segment 3 times. Record the length of the segment and each transit time in the Comments section of the Sample Collection Form.
6. After floating the river, retrieve the net assembly from the water, taking care not to disturb the bottom upriver of the net. Record the END TIME.
7. Determine the current velocity again as described above, calculate the average from the 6 measurements, and record on the form.
8. Concentrate the material in each net in one corner by swishing up and down in the river. Wash the material into a bucket half filled with water (NOT the shore sample bucket). Remove as much as possible from the nets.
9. The contents from both nets are combined into a single bucket. After this, pour the sample over a sieving bucket (same bucket used in the kick net samples).
10. Large objects in the bucket should be carefully inspected for organisms which are washed into the bucket before discarding.
11. After both nets are combined into one bucket, obtain a composite sample as described in Table V.

Table V. Procedures for Obtaining the Composite Sample

1. Pour the contents of the composite bucket through a U.S. Standard 30 sieve. Examine the bucket while rinsing it well to be sure all organisms are washed from the bucket onto the sieve.
2. Wash contents of the sieve to one side by gently agitating in water and wash into jar using as little water from the squirt bottle as possible. Carefully examine the sieve for any remaining organisms and place them in the appropriate jar labeled as either "shore" or "drift" sample.

Table VI. Sample Preserving and Labeling

1. Fill in special pre-numbered barcoded label and place on jar. All additional jars used for a sample must be labeled with same number. Enter this number which will be used for tracking purposes in the computer.
2. Preserve samples in ethanol as follows:
    a. If jar is more than 1/4 full of water, pour off enough to bring it to less than 1/4 full using proper sieve to retain organisms.
    b. Fill jar nearly full with 95% ethanol so that the concentration of ethanol is 70%. If there is a small amount of water in the sample, it may not be necessary to fill the jar entirely full to reach a 70% concentration.
    c. Transfer any organisms on the sieve back into the jar with forceps.
3. Check to be sure that the pre-numbered stick-on barcoded label is the on jar. Cover the entire label with clear, waterproof tape.
4. Seal the caps with electrical tape.
5. Place samples in cooler or other secure container for transport.
6. Secure all equipment in the vehicle.

Quick Reference Guide for Aquatic Vertebrates

Field Protocols For Fish Collection

1. Site Selection
    a. Determine river bank to be sampled. Stay along this shore the entire day, unless river aspect is unchanging and the selected side is not representative of both.
    b. Float downriver along the designated shoreline, stopping at each transect (A to K).
    c. In case of emergency, determine location of means of easy egress from river.

2. Electrofishing
    a. Check all electrical connections and potential conductors. Place cathodes and anodes in the water. Fill livewell with river water.
    b. Start generator, switch to pulsed DC, a frequency of 30pps, low range and 40%. These are the initial settings. Set timer and depress pedal switch to begin fishing.
    c. With switch depressed and floating downriver near shore, maneuver the raft or anode to cover a swath 3-4 meters wide, at an oar's length from shore, near cover, and at depths less than 3 meters wherever possible.
    d. Deposit fish in the livewell as soon as possible; do not hold them in the electrical field.
    e. Continue fishing until the next transect.
    f. Process fish when stopped at each transect. Record total time spent collecting and shocking time on data sheets.
    g. Identify and release any threatened and endangered species.
    h. Identify and measure (TL) sport fish and very large specimens, record external anomalies, and release unharmed.
    i. Identify other specimens. Determine number of individuals in species, measure largest and smallest individuals, and voucher as described in Voucher Protocol.
    j. Large, questionable species should be placed on ice and then frozen.
    k. Retain a subsample of target species for Fish Tissue Contaminants analysis.

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Quick Reference Guide for Aquatic Vertebrates (continued)

Anomaly Categories and Codes

Categories	Code	Definition
Absent	AB	Absent eye, fin, tail.
Blisters	BL	In mouth, just under skin.
Blackening*	BK	Tail or whole body with darkened pigmentation.
Extensive Black spot disease	BS	Small black cysts (dots) all over the fins and body.
Cysts	CY	Fluid-filled swellings; maybe small dots or large.
Copepod	CO	A parasitic infection characterized by a worm like copepod embedded in the flesh of the fish; body extends out and leaves a sore/discoloration at base, may be in mouth gills, fins, or anywhere on body.
Deformities	DE	Skeletal anomalies of the head, spine, and body shape; amphibians may have extra tails, limbs, toes.
Eroded fins	EF	Appear as reductions or substantial fraying of fin surface area.
Eroded gills	EG	Gill filaments eroded from tip.
Fungus	FU	May appear as filamentous or "fuzzy" growth on the fins, eyes, or body.
Fin anomalies	FA	Abnormal thickenings or irregularities of rays
Grubs	WG	White or yellow worms embedded in muscle or fins.
Hemorrhaging	HM	Red spots on mouth, body, fins, fin bases, eyes, and gills.
Ich	IC	White spots on the fins, skin or gills.
Lesions	LE	Open sores or exposed tissue; raised, granular or warty outgrowths.
Lice	LI	Scale-like, mobile arthropod.
Mucus	MU	Thick and excessive on skin or gill, as long cast from vent.
None	NO	No anomalies present.
Other	OT	Anomalies or parasites not specified (Please comment).
Scale anomalies	SA	Missing patches, abnormal thickings, granular skin
Shortened operculum	SO	Leaves a portion of the gill chamber uncovered
Tumors	TU	Areas of irregular cell growth which are firm and cannot be easily broken open when pinched. (Masses caused by parasites can usually be opened easily.)
Leeches	WL	Annelid worms which have anterior and posterior suckers. They may attach anywhere on the body.
Exophthalmia	EX	Bulging of the eye.

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Quick Reference Guide for Aquatic Vertebrates (continued)

Guidelines and Procedures for Preparing Fish Voucher Specimens

Category 1. Large easily identified species OR adults may be difficult to identify OR the species is uncommon in that region. Preserve 1-2 small (<150 mm total length) adult individuals per site plus 2-5 juveniles. If only large adults are collected, reserve smallest individual until voucher procedure is complete and preserve ONLY if space is available. Photograph if considered too large for the jar.

Category 2. Small to moderate-sized fish OR difficult to identify species. Preserve up to 20 adults and juveniles. If less than 20 individuals are collected, voucher all of them.

Category 3. Species of "special concern." These are state or federally listed species. Photograph and release. If specimens have died, include in voucher collection, note on data sheet and notify appropriate state official as soon as possible.

a. After all individuals of a species have been processed, place the voucher sub sample in a bucket with carbon dioxide tablets and a small amount of water. Individuals > 160 mm should be slit on the lower abdomen of the RIGHT side.
b. When specimens are dead, transfer to a small nylon bag containing a waterproof label with tag #. Place in "Voucher" jar in 10% formalin. BE SURE THAT JAR IS LABELED INSIDE AND OUT WITH A VOUCHER LABEL (site ID, barcode, and date).
c. DO NOT over pack the sample jars with specimens OR use less formalin than is needed. If a fish will not fit in a jar, freeze the specimen.
d. Continue until all species are processed. Seal voucher jar with electrical or clear tape. Check that the jar is correctly labeled. Enter BARCODE ID in appropriate place on field data sheet.
e. Transport to storage depot at end of week. Store in a cool, dark, ventilated space.

Quick Reference Guide For Fish Tissue Contaminants

Selecting And Processing Fish Tissue Specimens

NOTE: If neither a primary nor secondary species sample is available, use your best judgement in sending some type of composite fish tissue sample.

Primary Sample (P)

After all voucher specimens have been prepared, choose a cottid, cyprinid, or salmonid that has enough similarly sized individuals to weigh to 400 g.

Secondary Sample (S)

After all voucher specimens have been prepared, select a large piscivore or omnivore species that has at least 5 individuals 120 mm. Include similar sized individuals if available.

1. Place the fish into a bucket with two carbon dioxide tablets (e.g., "Alka Seltzer®") and a small volume of water. After they have been anaesthetized, use clean hands to transfer them to aluminum foil.
2. Prepare a clean work surface to prepare the primary composite sample. Keep hands, work surfaces, and wrapping materials clean and free of potential contaminants (mud, fuel, formalin, sun screen, insect repellant, etc.)

3-P. For primary samples, record the common name (from a standardized list) of the species, its species code (if required), and the number of individuals in the sample in the appropriate fields on line "P1" of the Sample Collection Form (Figure 11-1).

3-S. Measure the total length (TL) of each secondary individual. Record the common name (from a standardized list) of the secondary target species, its species code (if required), and the total length for each individual on lines S1 through S5 in the secondary sample section of the Sample Collection Form.

4. If the individuals included in composite samples were collected from throughout the sampling reach, place an "X" in the "Yes" box in the sample section of the Sample Collection Form. If the individuals were only collected from a limited segment of the sampling reach, place an "X" in the "No" box and explain in the "Explain" field on the form.

5-P. Wrap all primary fish together in a single piece of aluminum foil, making sure the dull side of the aluminum foil is in contact with the fish. Place the sample in a self-sealing plastic bag.

5-S. Wrap each fish of the secondary sample separately in aluminum foil, with the dull side of the foil in contact with the fish. Place all the wrapped individuals into a single self-sealing plastic bag.

6 Expel excess air and seal the bag. Wrap clear tape around the bag to seal and make a surface for each sample label.

7-P. Prepare two Fish Tissue sample labels (each having the same sample ID number [Figure 11-2]) by filling in the stream ID and the date of collection. Circle "PRIMARY" on each label. Record the sample ID number (barcode) in the primary sample section of the Sample Collection Form.

7-S. Prepare two Fish Tissue sample labels (each having the same sample ID number [Figure 11-2]) by filling in the stream ID and the date of collection. Circle "SECONDARY" on each label. Record the sample ID number (barcode) in the secondary sample section of the Sample Collection Form.

8. Attach the appropriate label to the tape surface of the bag. Cover the label with a strip of clear tape. Place the labeled bag into a second self-sealing plastic bag. Seal the bag and attach the second label to the outside of the appropriate bag. Cover the label with a strip of clear tape.

J. Place the double-bagged sample into a cooler containing bags of ice until shipment. Keep the sample frozen until shipment.

Appendix C
Species Codes for Aquatic Vertebrates

The following table contains the unique 8-character species code, the scientific name, and the common name assigned to each aquatic vertebrate species expected to be collected by EMAP sampling protocols in the Mid-Atlantic and Western regions. Generally, the species code is composed of the first four letters of the genus plus the first four letters of the species name. Modifications to this coding scheme were made in cases where two species could be assigned the same code. Species entries are arranged first by family (alphabetically), then by the assigned species code.

Aquatic Vertebrate Species List for Mid-Atlantic Region

Common Name	Genus	Species	Family	Family Name	VERTCODE
OHIO LAMPREY	Ichthyomyzon	bdellium	Petromyzontidae	Lamprey	ICHTBDEL
CHESTNUT LAMPREY	Ichthyomyzon	castaneus	Petromyzontidae	Lamprey	ICHTCAST
NORTHERN BROOK LAMPREY	Ichthyomyzon	fossor	Petromyzontidae	Lamprey	ICHTFOSS
MOUNTAIN BROOK LAMPREY	Ichthyomyzon	greeleyi	Petromyzontidae	Lamprey	ICHTGREE
SILVER LAMPREY	Ichthyomyzon	unicuspis	Petromyzontidae	Lamprey	ICHTUNIC
LEAST BROOK LAMPREY	Lampetra	aepyptera	Petromyzontidae	Lamprey	LAMPAEPY
AMERICAN BROOK LAMPREY	Lampetra	appendix	Petromyzontidae	Lamprey	LAMPAPPE
SEA LAMPREY	Petromyzon	marinus	Petromyzontidae	Lamprey	PETRMARI
UNKNOWN LAMPREY	Lampetra		Petromyzontidae	Lamprey	LAMPZZZZ
SHORTNOSE STURGEON	Acipenser	brevirostrum	Acipenseridae	Sturgeon	ACIPBREV
LAKE STURGEON	Acipenser	fulvescens	Acipenseridae	Sturgeon	ACIPFULV
ATLANTIC STURGEON	Acipenser	oxyrhynchus	Acipenseridae	Sturgeon	ACIPOXYR
SHOVELNOSE STURGEON	Scaphirhynchus	platorynchus	Acipenseridae	Sturgeon	SCAPPLAT
PADDLEFISH	Polyodon	spathula	Polyodontidae	Paddlefish	POLYSPAT
SPOTTED GAR	Lepisosteus	oculatus	Lepisosteidae	Gar	LEPIOCUL
LONGNOSE GAR	Lepisosteus	osseus	Lepisosteidae	Gar	LEPIOSSE
SHORTNOSE GAR	Lepisosteus	platostomus	Lepisosteidae	Gar	LEPIPLAT
BOWFIN	Amia	calva	Amiidae	Bowfin	AMIACALV
AMERICAN EEL	Anguilla	rostrata	Anguillidae	Eel	ANGUROST
BLUEBACK HERRING	Alosa	aestivalis	Clupeidae	Herring	ALOSAEST

Common Name	Genus	Species	Family	Family Name	VERTCODE
SKIPJACK HERRING	Alosa	chrysochloris	Clupeidae	Herring	ALOSCHRY
HICKORY SHAD	Alosa	mediocris	Clupeidae	Herring	ALOSMEDI
ALEWIFE	Alosa	pseudoharengus	Clupeidae	Herring	ALOSPSEU
AMERICAN SHAD	Alosa	sapidissima	Clupeidae	Herring	ALOSSAPI
GIZZARD SHAD	Dorosoma	cepedianum	Clupeidae	Herring	DOROCEPE
THREADFIN SHAD	Dorosoma	petenense	Clupeidae	Herring	DOROPETE
GOLDEYE	Hiodon	alosoides	Hiodontidae	Mooneye	HIODALOS
MOONEYE	Hiodon	tergisus	Hiodontidae	Mooneye	HIODTERG
REDFIN/GRASS PICKEREL	Esox	americanus	Esocidae	Pike	ESOXAMER
NORTHERN PIKE	Esox	lucius	Esocidae	Pike	ESOXLUCI
TIGER MUSKELLUNGE	Esox	lucius x masq.	Esocidae	Pike	ESOXLUMA
MUSKELLUNGE	Esox	masquinongy	Esocidae	Pike	ESOXMASQ
CHAIN PICKEREL	Esox	niger	Esocidae	Pike	ESOXNIGE
UNKNOWN TROUT	Oncorhynchus	sp.	Salmonidae	Salmon	ONCOZZZZ
HYBRID TROUT	Oncorhynchus	sp.	Salmonidae	Salmon	ONCOHHHH
ATLANTIC SALMON	Salmo	salar	Salmonidae	Trout	SALMSALA
BROWN TROUT	Salmo	trutta	Salmonidae	Trout	SALMTRUT
BROOK TROUT	Salvelinus	fontinalis	Salmonidae	Trout	SALVFONT
LAKE TROUT	Salvelinus	namaycush	Salmonidae	Trout	SALVNAMA
YOUNG OF YEAR TROUT	SP.	SP.	Salmonidae	Salmon	SALMYYYY
UNKNOWN SALMONID			Salmonidae	Trout	SALMZZZZ

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UNKNOWN CHAR	Salvelinus	sp.	Salmonidae	Salmon	SALVZZZZ
CENTRAL MUDMINNOW	Umbra	limi	Umbridae	Mudminnow	UMBRLIMI
EASTERN MUDMINNOW	Umbra	pygmaea	Umbridae	Mudminnow	UMBRPYGM
RIVER CARPSUCKER	Carpiodes	carpio	Catostomidae	Sucker	CARPCARP
QUILLBACK	Carpiodes	cyprinus	Catostomidae	Sucker	CARPCYPR
HIGHFIN CARPSUCKER	Carpiodes	velifer	Catostomidae	Sucker	CARPVELI
LONGNOSE SUCKER	Catostomus	catostomus	Catostomidae	Sucker	CATOCATO
WHITE SUCKER	Catostomus	commersoni	Catostomidae	Sucker	CATOCOMM
BLUE SUCKER	Cycleptus	elongatus	Catostomidae	Sucker	CYCLELON
CREEK CHUBSUCKER	Erimyzon	oblongus	Catostomidae	Sucker	ERIMOBLO
LAKE CHUBSUCKER	Erimyzon	sucetta	Catostomidae	Sucker	ERIMSUCE
NORTHERN HOGSUCKER	Hypentelium	nigricans	Catostomidae	Sucker	HYPENIGR
ROANOKE HOGSUCKER	Hypentelium	roanokense	Catostomidae	Sucker	HYPEROAN
SMALLMOUTH BUFFALO	Ictiobus	bubalus	Catostomidae	Sucker	ICTIBUBA
BIGMOUTH BUFFALO	Ictiobus	cyprinellus	Catostomidae	Sucker	ICTICYPR
BLACK BUFFALO	Ictiobus	niger	Catostomidae	Sucker	ICTINIGE
SPOTTED SUCKER	Minytrema	melanops	Catostomidae	Sucker	MINYMELA
SILVER REDHORSE	Moxostoma	anisurum	Catostomidae	Sucker	MOXOANIS
BIGEYE JUMPROCK	Moxostoma	ariommum	Catostomidae	Sucker	MOXOARIO
RIVER REDHORSE	Moxostoma	carinatum	Catostomidae	Sucker	MOXOCARI
BLACK JUMPROCK	Moxostoma	cervinum	Catostomidae	Sucker	MOXOCERV

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BLACK REDHORSE	Moxostoma	duquesnei	Catostomidae	Sucker	MOXODUQU
GOLDEN REDHORSE	Moxostoma	erythrurum	Catostomidae	Sucker	MOXOERYT
RUSTYSIDE SUCKER	Moxostoma	hamiltoni	Catostomidae	Sucker	MOXOHAMI
SHORTHEAD REDHORSE	Moxostoma	macrolepidotum	Catostomidae	Sucker	MOXOMACR
V-LIP REDHORSE	Moxostoma	pappillosum	Catostomidae	Sucker	MOXOPAPP
TORRENT SUCKER	Moxostoma	rhothoecum	Catostomidae	Sucker	MOXORHOT
SMALLFIN SUCKER	Moxostoma	robustum	Catostomidae	Sucker	MOXOROBU
GREATER REDHORSE	Moxostoma	valenciennesi	Catostomidae	Sucker	MOXOVALE
HYBRID SUCKER	Catostomus	sp.	Catostomidae	Sucker	CATOHHHH
UNKNOWN CATOSTOMID			Catostomidae	Sucker	CATOZZZZ
STONEROLLER	Campostoma	anomalum	Cyprinidae	Minnow	CAMPANOM
REDSIDE DACE	Clinostomus	elongatus	Cyprinidae	Minnow	CLINELON
ROSYSIDE DACE	Clinostomus	funduloides	Cyprinidae	Minnow	CLINFUND
LAKE CHUB	Couesius	plumbeus	Cyprinidae	Minnow	COUEPLUM
SATINFIN SHINER	Cyprinella	analostana	Cyprinidae	Minnow	CYPRANAL
BLUNTFACE SHINER	Cyprinella	camura	Cyprinidae	Minnow	CYPRCAMU
WHITETAIL SHINER	Cyprinella	galactura	Cyprinidae	Minnow	CYPRGALA
RED SHINER	Cyprinella	lutrensis	Cyprinidae	Minnow	CYPRLUTR
SPOTFIN CHUB	Cyprinella	monacha	Cyprinidae	Minnow	CYPRMONA
WHITEFIN SHINER	Cyprinella	nivea	Cyprinidae	Minnow	CYPRNIVE
SPOTFIN SHINER	Cyprinella	spiloptera	Cyprinidae	Minnow	CYPRSPIL

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BLACKTAIL SHINER	Cyprinella	venustus	Cyprinidae	Minnow	CYPRVENU
STEELCOLOR SHINER	Cyprinella	whipplei	Cyprinidae	Minnow	CYPRWHIP
SLENDER CHUB	Erimystax	cahni	Cyprinidae	Minnow	ERIMCAHN
STREAMLINE CHUB	Erimystax	dissimilis	Cyprinidae	Minnow	ERIMDISS
BLOTCHED CHUB	Erimystax	insignis	Cyprinidae	Minnow	ERIMINSI
GRAVEL CHUB	Erimystax	x-punctatus	Cyprinidae	Minnow	ERIMX-PU
TONGUETIED MINNOW	Exoglossum	laurae	Cyprinidae	Minnow	EXOGLAUR
CUTLIPS MINNOW	Exoglossum	maxillingua	Cyprinidae	Minnow	EXOGMAXI
EASTERN SILVERY MINNOW	Hybognathus	regius	Cyprinidae	Minnow	HYBOREGI
WHITE SHINER	Luxilus	albeolus	Cyprinidae	Minnow	LUXIALBE
CRESCENT SHINER	Luxilus	cerasinus	Cyprinidae	Minnow	LUXICERA
STRIPED SHINER	Luxilus	chrysocephalus	Cyprinidae	Minnow	LUXICHRY
WARPAINT SHINER	Luxilus	coccogenis	Cyprinidae	Minnow	LUXICOCC
COMMON SHINER	Luxilus	cornutus	Cyprinidae	Minnow	LUXICORN
ROSEFIN SHINER	Lythrurus	ardens	Cyprinidae	Minnow	LYTHARDE
MOUNTAIN SHINER	Lythrurus	lirus	Cyprinidae	Minnow	LYTHLIRU
PINEWOODS SHINER	Lythrurus	matutinus	Cyprinidae	Minnow	LYTHMATU
REDFIN SHINER	Lythrurus	umbratilis	Cyprinidae	Minnow	LYTHUMBR
SPECKLED CHUB	Macrhybopsis	aestivalis	Cyprinidae	Minnow	MACRAEST
SILVER CHUB	Macrhybopsis	storeriana	Cyprinidae	Minnow	MACRSTOR
PEARL DACE	Margariscus	margarita	Cyprinidae	Minnow	MARGMARG

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BLUEHEAD CHUB	Nocomis	leptocephalus	Cyprinidae	Minnow	NOCOLEPT
RIVER CHUB	Nocomis	micropogon	Cyprinidae	Minnow	NOCOMICR
BIGMOUTH CHUB	Nocomis	platyrhynchus	Cyprinidae	Minnow	NOCOPLAT
BULL CHUB	Nocomis	raneyi	Cyprinidae	Minnow	NOCORANE
GOLDEN SHINER	Notemigonus	crysoleucas	Cyprinidae	Minnow	NOTECRYS
WHITEMOUTH SHINER	Notropis	alborus	Cyprinidae	Minnow	NOTRALBO
BIGEYE CHUB	Notropis	amblops	Cyprinidae	Minnow	NOTRAMBL
COMELY SHINER	Notropis	amoenis	Cyprinidae	Minnow	NOTRAMOE
PUGNOSE SHINER	Notropis	anogenus	Cyprinidae	Minnow	NOTRANOG
POPEYE SHINER	Notropis	ariommus	Cyprinidae	Minnow	NOTRARIO
EMERALD SHINER	Notropis	atherinoides	Cyprinidae	Minnow	NOTRATHE
BRIDLE SHINER	Notropis	bifrenatus	Cyprinidae	Minnow	NOTRBIFR
RIVER SHINER	Notropis	blennius	Cyprinidae	Minnow	NOTRBLEN
BIGEYE SHINER	Notropis	boops	Cyprinidae	Minnow	NOTRBOOP
SILVERJAW MINNOW	Notropis	buccatus	Cyprinidae	Minnow	NOTRBUCC
GHOST SHINER	Notropis	buchanani	Cyprinidae	Minnow	NOTRBUCH
IRONCOLOR SHINER	Notropis	chalybaeus	Cyprinidae	Minnow	NOTRCHAL
REDLIP SHINER	Notropis	chiliticus	Cyprinidae	Minnow	NOTRCHIL
DUSKY SHINER	Notropis	cummingsae	Cyprinidae	Minnow	NOTRCUMM
BIGMOUTH SHINER	Notropis	dorsalis	Cyprinidae	Minnow	NOTRDORS
WEDGESPOT SHINER	Notropis	greenei	Cyprinidae	Minnow	NOTRGREE

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BLACKCHIN SHINER	Notropis	heterodon	Cyprinidae	Minnow	NOTRHETE
BLACKNOSE SHINER	Notropis	heterolepis	Cyprinidae	Minnow	NOTRHETL
SPOTTAIL SHINER	Notropis	hudsonius	Cyprinidae	Minnow	NOTRHUDS
TENNESSEE SHINER	Notropis	leuciodus	Cyprinidae	Minnow	NOTRLEUC
SILVER SHINER	Notropis	photogenis	Cyprinidae	Minnow	NOTRPHOT
SWALLOWTAIL SHINER	Notropis	procne	Cyprinidae	Minnow	NOTRPROC
ROSYFACE SHINER	Notropis	rubellus	Cyprinidae	Minnow	NOTRRUBE
SAFFRON SHINER	Notropis	rubricroceus	Cyprinidae	Minnow	NOTRRUBR
SABINE SHINER	Notropis	sabinae	Cyprinidae	Minnow	NOTRSABI
NEW RIVER SHINER	Notropis	scabriceps	Cyprinidae	Minnow	NOTRSCAB
ROUGHHEAD SHINER	Notropis	semperasper	Cyprinidae	Minnow	NOTRSEMP
MIRROR SHINER	Notropis	spectrunculus	Cyprinidae	Minnow	NOTRSPEC
SAND SHINER	Notropis	stramineus	Cyprinidae	Minnow	NOTRSTRA
TELESCOPE SHINER	Notropis	telescopus	Cyprinidae	Minnow	NOTRTELE
MIMIC SHINER	Notropis	volucellus	Cyprinidae	Minnow	NOTRVOLU
PUGNOSE MINNOW	Opsopoeodus	emiliae	Cyprinidae	Minnow	OPSOEMIL
FATLIPS MINNOW	Phenacobius	crassilabrum	Cyprinidae	Minnow	PHENCRAS
SUCKERMOUTH MINNOW	Phenacobius	mirabilis	Cyprinidae	Minnow	PHENMIRA
KANAWHA MINNOW	Phenacobius	teretulus	Cyprinidae	Minnow	PHENTERE
STARGAZING MINNOW	Phenacobius	uranops	Cyprinidae	Minnow	PHENURAN
BLACKSIDE DACE	Phoxinus	cumberlandensis	Cyprinidae	Minnow	PHOXCUMB

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NORTHERN REDBELLY DACE	Phoxinus	eos	Cyprinidae	Minnow	PHOXEOS
SOUTHERN REDBELLY DACE	Phoxinus	erythrogaster	Cyprinidae	Minnow	PHOXERYT
MOUNTAIN REDBELLY DACE	Phoxinus	oreas	Cyprinidae	Minnow	PHOXOREA
TENNESSEE DACE	Phoxinus	tennesseensis	Cyprinidae	Minnow	PHOXTENN
BLUNTNOSE MINNOW	Pimephales	notatus	Cyprinidae	Minnow	PIMENOTA
FATHEAD MINNOW	Pimephales	promelas	Cyprinidae	Minnow	PIMEPROM
BULLHEAD MINNOW	Pimephales	vigilax	Cyprinidae	Minnow	PIMEVIGI
BLACKNOSE DACE	Rhinichthys	atratulus	Cyprinidae	Minnow	RHINATRA
CHEAT MINNOW	Rhinichthys	bowersi	Cyprinidae	Minnow	RHINBOWE
LONGNOSE DACE	Rhinichthys	cataractae	Cyprinidae	Minnow	RHINCATA
CREEK CHUB	Semotilus	atromaculatus	Cyprinidae	Minnow	SEMOATRO
FALLFISH	Semotilus	corporalis	Cyprinidae	Minnow	SEMOCORP
UNKNOWN CYPRINID			Cyprinidae	Minnow	CYPRZZZZ
SNAIL BULLHEAD	Ameiurus	brunneus	Ictaluridae	Catfish	AMEIBRUN
WHITE CATFISH	Ameiurus	catus	Ictaluridae	Catfish	AMEICATU
BLACK BULLHEAD	Ameiurus	melas	Ictaluridae	Catfish	AMEIMELA
YELLOW BULLHEAD	Ameiurus	natalis	Ictaluridae	Catfish	AMEINATA
BROWN BULLHEAD	Ameiurus	nebulosus	Ictaluridae	Catfish	AMEINEBU
FLAT BULLHEAD	Ameiurus	platycephalus	Ictaluridae	Catfish	AMEIPLAT
BLUE CATFISH	Ictalurus	furcatus	Ictaluridae	Catfish	ICTAFURC
CHANNEL CATFISH	Ictalurus	punctatus	Ictaluridae	Catfish	ICTAPUNC

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FLATHEAD CATFISH	Pylodictis	olivaris	Ictaluridae	Catfish	PYLOOLIV
MOUNTAIN MADTOM	Noturus	eleutherus	Ictaluridae	Catfish	NOTUELEU
YELLOWFIN MADTOM	Noturus	flavipinnis	Ictaluridae	Catfish	NOTUFLAV
STONECAT	Noturus	flavus	Ictaluridae	Catfish	NOTUFLAU
CAROLINA MADTOM	Noturus	furiosus	Ictaluridae	Catfish	NOTUFURI
ORANGEFIN MADTOM	Noturus	gilberti	Ictaluridae	Catfish	NOTUGILB
TADPOLE MADTOM	Noturus	gyrinus	Ictaluridae	Catfish	NOTUGYRI
MARGINED MADTOM	Noturus	insignis	Ictaluridae	Catfish	NOTUINSI
BRINDLED MADTOM	Noturus	miurus	Ictaluridae	Catfish	NOTUMIUR
NORTHERN MADTOM	Noturus	stigmosus	Ictaluridae	Catfish	NOTUSTIG
TROUT-PERCH	Percopsis	omiscomaycus	Percopsidae	Trout-perch	PERCOMIS
PIRATE PERCH	Aphredoderus	sayanus	Aphredoderidae	Pirate perch	APHRSAYA
BURBOT	Lota	lota	Gadidae	Codfish	LOTALOTA
NORTHERN STUDFISH	Fundulus	catenatus	Cyprinodontidae	Killifish	FUNDCATE
BANDED KILLIFISH	Fundulus	diaphanus	Cyprinodontidae	Killifish	FUNDDIAP
MUMMICHOG	Fundulus	heteroclitus	Cyprinodontidae	Killifish	FUNDHETE
LINED TOPMINNOW	Fundulus	lineolatus	Cyprinodontidae	Killifish	FUNDLINE
SPECKLED KILLIFISH	Fundulus	rathbuni	Cyprinodontidae	Killifish	FUNDRATH
RAINWATER KILLIFISH	Lucania	parva	Cyprinodontidae	Killifish	LUCAPARV
EASTERN MOSQUITOFISH	Gambusia	holbrooki	Poeciliidae	Livebearer	GAMBHOLB
GUPPY	Poecilia	reticulata	Poeciliidae	Livebearer	POECRETI

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BROOK SILVERSIDE	Labidesthes	sicculus	Atherinidae	Silverside	LABISICC
INLAND SILVERSIDE	Menidia	beryllina	Atherinidae	Silverside	MENIBERY
FOURSPINE STICKLEBACK	Apeltes	quadracus	Gasterosteidae	Stickleback	APELQUAD
BROOK STICKLEBACK	Culaea	inconstans	Gasterosteidae	Stickleback	CULAINCO
THREESPINE STICKLEBACK	Gasterosteus	aculeatus	Gasterosteidae	Stickleback	GASTACUL
NINESPINE STICKLEBACK	Pungitius	pungitius	Gasterosteidae	Stickleback	PUNGPUNG
BLACK SCULPIN	Cottus	baileyi	Cottidae	Sculpin	COTTBAIL
MOTTLED SCULPIN	Cottus	bairdi	Cottidae	Sculpin	COTTBAIR
BANDED SCULPIN	Cottus	carolinae	Cottidae	Sculpin	COTTCARO
SLIMY SCULPIN	Cottus	cognatus	Cottidae	Sculpin	COTTCOGN
SHORTHEAD SCULPIN	Cottus	confusus	Cottidae	Sculpin	COTTCONF
SPOONHEAD SCULPIN	Cottus	ricei	Cottidae	Sculpin	COTTRICE
POTOMAC SCULPIN	Cottus	girardi	Cottidae	Sculpin	COTTGIRA
UNKNOWN COTTID	Cottus		Cottidae	Sculpin	COTTZZZZ
WHITE PERCH	Morone	americana	Percichthyidae	Temperate Bass	MOROAMER
WHITE BASS	Morone	chrysops	Percichthyidae	Temperate Bass	MOROCHRY
STRIPED BASS	Morone	saxatilis	Percichthyidae	Temperate Bass	MOROSAXA
MUD SUNFISH	Acantharchus	pomotis	Centrarchidae	Sunfish	ACANPOMO
ROANOKE ROCKBASS	Ambloplites	cavifrons	Centrarchidae	Sunfish	AMBLCAVI
ROCK BASS	Ambloplites	rupestris	Centrarchidae	Sunfish	AMBLRUPE
FLIER	Centrarchus	macropterus	Centrarchidae	Sunfish	CENTMACR

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BANDED PYGMY SUNFISH	Elassoma	zonatum	Centrarchidae	Sunfish	ELASZONA
BLACKBANDED SUNFISH	Enneacanthus	chaetodon	Centrarchidae	Sunfish	ENNECHAE
BLUESPOTTED SUNFISH	Enneacanthus	gloriosus	Centrarchidae	Sunfish	ENNEGLOR
BANDED SUNFISH	Enneacanthus	obesus	Centrarchidae	Sunfish	ENNEOBES
REDBREAST SUNFISH	Lepomis	auritus	Centrarchidae	Sunfish	LEPOAURI
GREEN SUNFISH	Lepomis	cyanellus	Centrarchidae	Sunfish	LEPOCYAN
PUMPKINSEED	Lepomis	gibbosus	Centrarchidae	Sunfish	LEPOGIBB
WARMOUTH	Lepomis	gulosus	Centrarchidae	Sunfish	LEPOGULO
BLUEGILL	Lepomis	macrochirus	Centrarchidae	Sunfish	LEPOMACR
DOLLAR SUNFISH	Lepomis	marginatus	Centrarchidae	Sunfish	LEPOMARG
LONGEAR SUNFISH	Lepomis	megalotis	Centrarchidae	Sunfish	LEPOMEGA
REDEAR SUNFISH	Lepomis	microlophus	Centrarchidae	Sunfish	LEPOMICR
SPOTTED SUNFISH	Lepomis	punctatus	Centrarchidae	Sunfish	LEPOPUNC
SMALLMOUTH BASS	Micropterus	dolomieu	Centrarchidae	Sunfish	MICRDOLO
SPOTTED BASS	Micropterus	punctulatus	Centrarchidae	Sunfish	MICRPUNC
LARGEMOUTH BASS	Micropterus	salmoides	Centrarchidae	Sunfish	MICRSALM
WHITE CRAPPIE	Pomoxis	annularis	Centrarchidae	Sunfish	POMOANNU
BLACK CRAPPIE	Pomoxis	nigromaculatus	Centrarchidae	Sunfish	POMONIGR
UNKNOWN CENTRARCHID			Centrarchidae	Sunfish	CENTZZZZ
CRYSTAL DARTER	Ammocrypta	asprella	Percidae	Perch	AMMOASPR
EASTERN SAND DARTER	Ammocrypta	pellucida	Percidae	Perch	AMMOPELL

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SHARPHEAD DARTER	Etheostoma	acuticeps	Percidae	Perch	ETHEACUT
GREENSIDE DARTER	Etheostoma	blennioides	Percidae	Perch	ETHEBLEN
RAINBOW DARTER	Etheostoma	caeruleum	Percidae	Perch	ETHECAER
BLUEBREAST DARTER	Etheostoma	camurum	Percidae	Perch	ETHECAMU
GREENFIN DARTER	Etheostoma	chlorobranchium	Percidae	Perch	ETHECHLO
ASHY DARTER	Etheostoma	cinereum	Percidae	Perch	ETHECINE
FANTAIL DARTER	Etheostoma	flabellare	Percidae	Perch	ETHEFLAB
SWAMP DARTER	Etheostoma	fusiforme	Percidae	Perch	ETHEFUSI
BLUESIDE DARTER	Etheostoma	jessiae	Percidae	Perch	ETHEJESS
KANAWHA DARTER	Etheostoma	kanawhae	Percidae	Perch	ETHEKANA
STRIPETAIL DARTER	Etheostoma	kennecotti	Percidae	Perch	ETHEKENN
LONGFIN DARTER	Etheostoma	longimanum	Percidae	Perch	ETHELONG
SPOTTED DARTER	Etheostoma	maculatum	Percidae	Perch	ETHEMACU
JOHNNY DARTER	Etheostoma	nigrum	Percidae	Perch	ETHENIGR
TESSELATED DARTER	Etheostoma	olmstedi	Percidae	Perch	ETHEOLMS
CANDY DARTER	Etheostoma	osburni	Percidae	Perch	ETHEOSBU
DUSKYTAIL DARTER	Etheostoma	percnurum	Percidae	Perch	ETHEPERC
RIVERWEED DARTER	Etheostoma	podostemone	Percidae	Perch	ETHEPODO
REDLINE DARTER	Etheostoma	rufilineatum	Percidae	Perch	ETHERUFI
SAWCHEEK DARTER	Etheostoma	serrifer	Percidae	Perch	ETHESERR
SNUBNOSE DARTER	Etheostoma	simoterum	Percidae	Perch	ETHESIMO

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SPECKLED DARTER	Etheostoma	stigmaeum	Percidae	Perch	ETHESTIG
SWANNANOA DARTER	Etheostoma	swannanoa	Percidae	Perch	ETHESWAN
TIPPECANOE DARTER	Etheostoma	tippecanoe	Percidae	Perch	ETHETIPP
VARIEGATE DARTER	Etheostoma	variatum	Percidae	Perch	ETHEVARI
GLASSY DARTER	Etheostoma	vitreum	Percidae	Perch	ETHEVITR
WOUNDED DARTER	Etheostoma	vulneratum	Percidae	Perch	ETHEVULN
REDFIN DARTER	Etheostoma	whipplei	Percidae	Perch	ETHEWHIP
BANDED DARTER	Etheostoma	zonale	Percidae	Perch	ETHEZONA
TANGERINE DARTER	Percina	aurantiaca	Percidae	Perch	PERCAURA
BLOTCHSIDE LOGPERCH	Percina	burtoni	Percidae	Perch	PERCBURT
LOGPERCH	Percina	caprodes	Percidae	Perch	PERCCAPR
CHANNEL DARTER	Percina	copelandi	Percidae	Perch	PERCCOPE
PIEDMONT DARTER	Percina	crassa	Percidae	Perch	PERCCRAS
GILT DARTER	Percina	evides	Percidae	Perch	PERCEVID
APPALACHIA DARTER	Percina	gymnocephala	Percidae	Perch	PERCGYMN
LONGHEAD DARTER	Percina	macrocephala	Percidae	Perch	PERCMACR
BLACKSIDE DARTER	Percina	maculata	Percidae	Perch	PERCMACU
STRIPEBACK DARTER	Percina	notogramma	Percidae	Perch	PERCNOTO
SHARPNOSE DARTER	Percina	oxyrhynchus	Percidae	Perch	PERCOXYR
SHIELD DARTER	Percina	peltata	Percidae	Perch	PERCPELT
ROANOKE LOGPERCH	Percina	rex	Percidae	Perch	PERCREX

Common Name	Genus	Species	Family	Family Name	VERTCODE
ROANOKE DARTER	Percina	roanoka	Percidae	Perch	PERCROAN
DUSKY DARTER	Percina	sciera	Percidae	Perch	PERCSCIE
RIVER DARTER	Percina	shumardi	Percidae	Perch	PERCSHUM
SAUGER	Stizostedion	canadense	Percidae	Perch	STIZCANA
WALLEYE	Stizostedion	vitreum	Percidae	Perch	STIZVITR
YELLOW PERCH	Perca	flavescens	Percidae	Perch	PERCFLAV
FRESHWATER DRUM	Aplodinotus	grunniens	Sciaenidae	Drum	APLOGRUN
			Z_Hybrid		HYBR01
			Z_Unknown		UNKN09
EXOTIC/OTHER SPECIES					
TILAPIA	Tilapia	spp	Cichlidae	Cichlid	TILAZZZZ
ORIENTAL WEATHERFISH	Misgurnus	anguillicaudatus	Cobitidae	Loach	MISGANGU
GRASS CARP	Ctenopharyngodon	idella	Cyprinidae	Minnow	CTENIDEL
COMMON CARP	Cyprinus	carpio	Cyprinidae	Minnow	CYPRCARP
RUDD	Scardinius	erythrophthalmus	Cyprinidae	Minnow	SCARERYT
BITTERLING	Rhodeus	sericeus	Cyprinidae	Minnow	RHODSERI
IDE	Leuciscus	idus	Cyprinidae	Minnow	LEUCIDUS
GOLDFISH	Carassius	auratus	Cyprinidae	Minnow	CARAAURA
TENCH	Tinca	tinca	Cyprinidae	Minnow	TINCTINC
UNKNOWN AMPHIBIAN				AMPHZZZZ	
UNKNOWN FROG				Toad	FROGZZZZ

Common Name	Genus	Species	Family	Family Name	VERTCODE
UNKNOWN SALAMANDER				Salamander	SALAZZZZ
UNKNOWN TURTLE				TURTLEZZ	
UNKNOWN SNAKE					SNAKEZZZ
BEAVER	Castor	canadensis	Castoridae	Beaver	CASTCANA
MUSKRAT	Ondatra	zibethica	Cricetidae	Rodent	ONDAZIBE

Aquatic Vertebrates of the Western United States.

Code	Latin name	Common name (vouchering category)
LAMPAY	Lampetra ayresi	river lamprey 2
LAMPLE	Lampetra lethophaga	pit-klamath brook lamprey 2
LAMPRI	Lampetra richardsoni	western brook lamprey 2
LAMPTR	Lampetra tridentata	pacific lamprey 2; goose lake lamprey 3
LAMPSI	Lampetra similis	klamath river lamprey 2
LAMPZZ		unknown lamprey
COTTAL	Cottus aleuticus	coastrange sculpin 2
COTTAS	Cottus asper	prickly sculpin 2
COTTBA	Cottus bairdi	mottled sculpin 2; malheur motted sculpin 3
COTTBE	Cottus beldingi	paiute sculpin 2
COTTCF	Cottus confusus	shorthead sculpin 2
COTTGU	Cottus gulosus	riffle sculpin 2
COTTKL	Cottus klamathensis	marbled sculpin 2
COTTMA	Cottus marginatus	margined sculpin 3
COTTPE	Cottus perplexus	reticulate sculpin 2
COTTPI	Cottus pitensis	pit sculpin 3
COTTPR	Cottus princeps	klamath lake sculpin 2
COTTRH	Cottus rhotheus	torrent sculpin 2
COTTTE	Cottus tenuis	slender sculpin 3
LEPTAR	Leptocottus armatus	pacific staghorn sculpin 2
COTTZZ		unknown Cottid
ACIPME	Acipenser medirostris	green sturgeon 1
ACIPTR	Acipenser transmontanus	white sturgeon 1
ALOSSA	Alosa sapidissima	American shad 1
CLUPPA	Clupea pallasi	pacific herring 2
ONCOGO	Oncorhynchus gorbuscha	pink salmon 3
ONCOKE	Oncorhynchus keta	chum salmon 3
ONCOKI	Oncorhynchus kisutch	coho salmon 3
ONCONE	Oncorhynchus nerka	sockeye salmon 3

Code	Latin name	Common name (vouchering category)
ONCOTS	Oncorhynchus tshawytscha	chinook salmon 3
ONCOAG	Oncorhynchus aguabonita	golden trout 1
ONCOCL	Oncorhynchus clarki	cutthroat trout; Umpqua 3, Lahontan 3
ONCOMY	Oncorhynchus mykiss	rainbow trout 1
PROSWI	Prosopium williamsoni	mountain whitefish 1
SALMSA	Salmo salar	atlantic salmon 1
SALMTR	Salmo trutta	brown trout 1
SALVCO	Salvelinus confluentus	bull trout 3
SALVFO	Salvelinus fontinalis	brook trout 1
SALVNA	Salvelinus namaycush	lake trout 1
SALMZZ		unknown salmonid
HYPOPR	Hypomesus pretiosus	surf smelt 2
SPIRTH	Spirinchus thaleichthys	longfin smelt 2
THALPA	Thaleichthys pacificus	eulachon 2
ACROAL	Acrocheilus alutaceus	chiselmouth 1
CARAAU	Carassius auratus	goldfish 1
CTENID	Ctenopharyngodon idella	grass carp 1
CYPRCA	Cyprinus carpio	common carp 1
GILAAL	Gila alvordensis	alvord chub 3
GILABI	Gila bicolor	tui chub 1; Catlow, Hutton, Goose Lake, Oregon Lakes, Sheldon, Summer Basin and Warner 3
GILABO	Gila boraxobius	borax lake chub 3
GILACO	Gila coerulea	blue chub 2
LAVISY	Lavinia symmetricus	california roach 3
MYLOCA	Mylocheilus caurinus	peamouth 1
NOTECR	Notemigonus crysoleucas	golden shiner 2
PIMEPR	Pimephales promelas	fathead minnow 2
PTYCOR	Ptychocheilus oregonensis	northern squawfish 1
PTYCUM	Ptychocheilus umpquae	umpqua squawfish 1
RHINCA	Rhinichthys cataractae	longnose dace 2
RHINEV	Rhinichthys evermanni	umpqua dace 2

Code	Latin name	Common name (vouchering category)
RHINFA	Rhinichthys falcatus	leopard dace 2
RHINOS	Rhinichthys osculus	speckled dace 2; foskett speckled dace 3; millicoma dace 3
RICHBA	Richardsonius balteatus	redside shiner 2
RICHEG	Richardsonius egregius	lahontan redside 3
TINCTI	Tinca tinca	tench 1
OREGCR	Oregonichthys crameri	oregon chub 3
OREGKA	Oregonichthys kalawatseti	umpqua chub 3
CYPRZZ		unknown cyprinid
CATOCB	Catostomus columbianus	bridgelip sucker 1
CATOMA	Catostomus macrocheilus	largescale sucker 1
CATOOC	Catostomus occidentalis	sacramento sucker 1; Goose Lake sucker 3
CATOPL	catostomus platyrhynchus	mountain sucker 2
CATORI	Catostomus rimiculus	klamath smallscale sucker 1; Jenny Creek sucker 3
CATOSY	Catostomus snyderi	klamath largescale sucker 3
CATOTA	Catostomus tahoensis	tahoe sucker 3
CATOWA	Catostomus warnerensis	warner sucker 3
CHASBR	Chasmistes brevirostris	shortnose sucker 3
DELTLU	Deltistes luxatus	lost river sucker 3
CATOZZ		unknown catostomid
MISGAN	Misgurnus anguillicaudatus	oriental weatherfish 2
AMEICA	Ameiurus catus	white catfish 1
AMEIME	Ameiurus melas	black bullhead 1
AMEINA	Ameiurus natalis	yellow bullhead 1
AMEINE	Ameiurus nebulosus	brown bullhead 1
ICTAPU	Ictalurus punctatus	channel catfish 1
NOTUGY	Noturus gyrinus	tadpole madtom 2
PYLOOL	Pylodictis olivaris	flathead catfish 1
PERCTR	Percopsis transmountana	sand roller 2
LOTALO	Lota lota	burbot 1
MICRPR	Microgadus proximus	pacific tomcod 2

Code	Latin name	Common name (vouchering category)
FUNDDI	Fundulus diaphanus	banded killifish 2
LUCAPA	Lucania parva	rainwater killifish 2
GAMBAF	Gambusia affinis	western mosquitofish 2
ATHEAF	Atherinops affinis	topsmelt 2
GASTAC	Gasterosteus aculeatus	threespine stickleback 2
MOROSA	Morone saxatilis	striped bass 1
ARCHIN	Archoplites interruptus	sacramento perch
LEPOCY	Lepomis cyanellus	green sunfish 1
LEPOGI	Lepomis gibbosus	pumpkinseed 1
LEOPGU	Lepomis gulosus	warmouth 1
LEPOMA	Lepomis macrochirus	bluegill 1
LEPOMI	Lepomis microlophus	redear sunfish 1
MICRDO	Micropterus dolomieui	smallmouth bass 1
MICRSA	Micropterus salmoides	largemouth bass 1
POMOAN	Pomoxis annularis	white crappie 1
POMONI	Pomoxis nigromaculatus	black crappie 1
CENTRZZ		unknown centrarchid
PERCFL	Perca flavescens	yellow perch 1
STIZVI	Stizostedion vitreum	walleye 1
CYMAAG	Cymatogaster aggregata	shiner perch 1
PHOLOR	Pholis ornata	saddleback gunnel 2
PLATST	Platichthys stellatus	starry flounder 1
PSEURE	Pseudacris regilla	pacific tree frog
ASCATR	Ascaphus truei	tailed frog
RANAAU	Rana aurora	red-legged frog
RANABO	Rana boylii	foothill yellow-legged frog
RANACA	Rana cascadae	cascade frog
RANACT	Rana catesbiana	bullfrog
RANAPI	Rana pipiens	leopard frog
RANAPR	Rana pretiosa	spotted frog

Code	Latin name	Common name (vouchering category)
BUFOBO	Bufo boreas	western toad
BUFOWO	Bufo woodhousii	woodhouse's toad
AMBYGR	Ambystoma gracile	northwestern salamander
AMBYMA	Ambystoma macrodactylum	longtoed salamander
AMBYTI	Ambystoma tigrinum	tiger salamander
DICACO	Dicamptodon copei	cope's giant salamander
DICATE	Dicamptodon tenebrosus	pacific giant salamander
RHYACA	Rhyacotrition cascadae	cascade torrent salamander
RHYAKE	Rhyacotriton kezeri	columbia torrent salamander
RHYAVA	Rhyacotriton variegatus	southern torrent salamander
TARIGR	Taricha granulosa	rough-skinned newt
AMPHZZ	Rana sp.	unknown amphibian