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ENVIRONMENTAL MONITORING AND ASSESSMENT PROGRAM-SURFACE WATERS:

FIELD OPERATIONS AND METHODS FOR MEASURING THE ECOLOGICAL CONDITION OF WADEABLE STREAMS

Edited by

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

[1] U.S. Environmental Protection Agency
Ecosystems Research Branch
Ecological Exposure Research Division
National Exposure Research Laboratory
Cincinnati, OH 45268

[2] U.S. Environmental Protection Agency
Regional Ecology Branch
Western Ecology Division
National Health and Environmental Effects Research Laboratory
Corvallis, OR 97333

NATIONAL EXPOSURE RESEARCH LABORATORY
OFFICE OF RESEARCH AND DEVELOPMENT
U.S. ENVIRONMENTAL PROTECTION AGENCY
RESEARCH TRIANGLE PARK, NC 27711

NATIONAL HEALTH AND ENVIRONMENTAL EFFECTS RESEARCH LABORATORY
OFFICE OF RESEARCH AND DEVELOPMENT
U.S. ENVIRONMENTAL PROTECTION AGENCY
RESEARCH TRIANGLE PARK, NC 27711

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Section authors are listed on the following page. Complete addresses for authors are also provided in each section.

Section 1:	J.M. Lazorchak[1], A.T. Herlihy[2], H.R. Preston[3, 4], and D.J. Klemm[1]
Section 2:	B.H. Hill[1], F.H. McCormick[1], J.M. Lazorchak[1], D.J. Klemm[1], P.A. Lewis[1, 5], V.C. Rogers[6, 7], and M.K. McDowell[5]
Section 3:	D.J. Klemm[1], B.H. Hill[1], F.H. McCormick[1], and M.K. McDowell[5]
Section 4:	A T. Herlihy[2]
Section 5:	A T. Herlihy[2]
Section 6:	P R. Kaufmann[2]
Section 7:	P R. Kaufmann[2] and E.G. Robison[2, 8]
Section 8:	B.H. Hill[1]
Section 9:	B.H. Hill[1]
Section 10:	J.M. Lazorchak[1], and M. E. Smith[9]
Section 11:	D.J. Klemm[1], J.M. Lazorchak[1], and P.A. Lewis[1, 4]
Section 12:	F.H. McCormick[1] and R. M. Hughes[10]
Section 13:	R.B. Yeardley, Jr.[8], J.M. Lazorchak[1], and F.H. McCormick[1]
Section 14:	J.M. Lazorchak[1], A. T. Herlihy[2], and J. Green[3]
Section 15:	J.M. Lazorchak[1]

[1] U.S. EPA, National Exposure Research Laboratory, Cincinnati, OH 45628.

[2] Department of Fisheries and Wildlife, Oregon State University, Corvallis, OR 97333.

[3] U.S. EPA, Region 3, Wheeling, WV 26003).

[4] Current address: Canaan Valley Institute, Davis, WV 26260.

[5] Current Address: 1037 Wylie Road, RR #2, Seaman, OH 45679

[6] OAO Corp., Corvallis, OR 97333

[7] Current address: Linn-Benton Community College, Albany, OR.

[8] Current Address: Oregon Department of Forestry, Salem, OR 97310

[9] So Bran Environmental, Inc., Cincinnati, OH 45628.

[10] Dynamac International, Inc., Corvallis, OR 97333.

FOREWORD

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

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

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

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

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

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

1. Develop and evaluate appropriate and meaningful indicators of ecological condition and develop associated criteria to characterize condition.

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

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

PREFACE

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

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

ABSTRACT

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

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ACRONYMS, ABBREVIATIONS, AND MEASUREMENT UNITS

Acronyms and Abbreviations

AFDM	Ash-free dry mass
APA	Acid/Alkaline Phosphatase Activity
BPJ	Best Professional Judgment
BOD	Biological Oxygen Demand
CENR	(White House) Committee on the Environment and Natural Resources
CFR	Code of Federal Regulations
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
GPS	Global Positioning System
ID	identification
LWD	Large Woody Debris
MAHA	Mid-Atlantic Highlands Assessment
MAIA	Mid-Atlantic Integrated Assessment
NAWQA	National Water-Quality Assessment Program
NERL	National Exposure Research Laboratory
NHEERL	National Health and Environmental Effects Research Laboratory
ORD	Office of Research and Development
OSHA	Occupational Safety and Health Administration
P-Hab	physical habitat
PVC	polyvinyl chloride
QA	quality assurance
QC	quality control

ACRONYMS, ABBREVIATIONS, AND MEASUREMENT UNITS (CONTINUED)

Acronyms and Abbreviations (continued)

RBP	(EPA) Rapid Bioassessment Protocol
R-EMAP	Regional Environmental Monitoring and Assessment Program
SL	Standard length
SOP	Standard Operating Procedure
TIME	Temporally Integrated Monitoring of Ecosystems
TL	Total length
USGS	United States Geological Survey
WED	Western Ecology Division
YOY	young of year
YSI	Yellow Springs Instrument system

Measurement Units

amps	amperes
cm	centimeter
gal	gallon
ha	hectare
Hz	Hertz
in	inches
L	liter
m	meter
m^2	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

by
James M. Lazorchak[1], Alan T. Herlihy[2], H. Ronald Preston[3, 4] and Donald J. Klemm[1]

This manual contains procedures for collecting samples and measurement data from various biotic and abiotic components of streams. These procedures were developed and used between 1993 and 1998 in research studies of the U.S. Environmental Protection Agency's (EPA) Environmental Monitoring and Assessment Program (EMAP). 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 stream monitoring programs.

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

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

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

2 Department of Fisheries and Wildlife, Oregon State University, c/o U.S. EPA. 200 SW 35th St., Corvallis, OR 97333

3 U.S. EPA Region 3, Wheeling Office, 303 Methodist Bldg, 11th and Chaplin Streets, Wheeling, WV 26003.

4 Current address: Canaan Valley Institute, P.O. Box 673, Davis, WV 26260

1.1 OVERVIEW OF EMAP-SURFACE WATERS

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

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

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

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

The EMAP Surface Waters Resource Group (EMAP-SW) is charged with developing the appropriate tools to assess the health of lakes, streams, and wetlands in the United

States. The first phase of the program started with a study of northeastern lakes between 1991 and 1996 (Larsen and Christie, 1993; Baker et al., 1997). In 1992 and 1993, a pilot study of wetland ecosystems was conducted in the Prairie Pothole region of the northern plains region of the U.S. (Peterson et al., 1997). The specific research studies dealing with streams are described in more detail in the following section.

1.2 STREAM SAMPLING COMPONENTS OF EMAP-SURFACE WATERS

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

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

1.2.1 Mid-Atlantic Highlands Assessment Project

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

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

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

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

1.2.2 Mid-Atlantic Integrated Assessment Program

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

1.2.3 Temporal Integrated Monitoring of Ecosystems Project

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

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

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

1.2.4 Other Projects

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

1.3 SUMMARY OF ECOLOGICAL INDICATORS

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

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

1.3.1 Water Chemistry

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

1.3.2 Physical Habitat

Naturally occurring differences among surface waters in physical habitat structure and associated hydraulic characteristics contributes to much of the observed variation in species composition and abundance within a zoogeographic province. The structural complexity of aquatic habitats provides the variety of physical and chemical conditions to support diverse biotic assemblages and maintain long-term stability. Anthropogenic alterations of riparian areas and stream channels, wetland drainage, grazing and agricultural practices, and stream bank modifications such as revetments or development, generally act to reduce the complexity of aquatic habitat and result in a loss of species and ecosystem degradation.

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

1.3.3 Periphyton Assemblage

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

A hierarchical framework is being used in the development of the periphyton indices of stream condition. The framework involves the calculation of composite indices for biotic

integrity, ecological sustainability, and trophic condition. The composite indices will be calculated from measured or derived first-order and second-order indices. The first-order indices include species composition (richness, diversity), cell density, AFDM, chlorophyll, and enzyme activity (e.g., Saylor et al., 1979), which individually are indicators of ecological condition in streams. Second-order indices will be calculated from periphyton characteristics, such as the autotrophic index (Weber, 1973), community similarity compared to reference sites, and autecological indices (e.g., Lowe, 1974; Lange-Bertalot, 1979; Charles, 1985; Dixit et al, 1992).

1.3.4 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 stream provides a functional indicator of energy flow and material transformation within the ecosystem

1.3.5 Benthic Macroinvertebrate Assemblage

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

Two different approaches are currently being evaluated to developing ecological indicators based on benthic invertebrate assemblages. The first is a multimetric approach, where different structural and functional attributes of the assemblage are characterized as “metrics”. Individual metrics that respond to different types of stressors are scored against

expectations under conditions of minimal human disturbance. The individual metric scores are then summed into an overall index value that is used to judge the overall level of impairment of an individual stream reach. Examples of multimetric indices based on benthic invertebrate assemblages include Kerans and Karr (1993), Fore et al. (1996) and Barbour et al. (1995; 1996).

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

1.3.6 Aquatic Vertebrate Assemblages

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

Indicators based on vertebrate assemblages are being developed primarily using the multimetric approach described in Section 1.3.5 for benthic macroinvertebrates, and originally conceived by Karr and others (Karr et al., 1986). Simon and Lyons (1995) provide a recent review of multimetric indicators as applied to stream fish assemblages.

1.3.7 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 stream degradation, when it is shown by the condition indicators to occur, is water quality, physical habitat, or both.

The various studies that have been done on fish tissue contaminants have focused on different parts of the fish: whole fish, fillets, livers. For EMAP-SW, the focus is on whole fish because of the emphasis on the ecological health of the whole stream (as opposed to a focus on human health concerns). Whole fish are a better indicator of risk to piscivorous wildlife than fillets. It is hoped to also be able to say something about risks to human health by analyzing whole fish. Whole fish also present fewer logistical problems for field crews (no gutting required in the field) and the analytical lab (no filleting necessary).

Samples are prepared for two major categories of fish species. One sample is prepared using a species whose adults are small (e.g., small minnows, sculpins, or darters). The second sample is prepared using a species whose adults are of larger size (e.g., suckers, bass, trout, sunfish, carp). In addition to being more ubiquitous than the larger fish (and therefore more likely to be present in sufficient numbers to composite), small fish have other advantages over large fish. Most importantly, it may be possible to get a more representative sample of the contaminant load in that stream segment (although it could be at a lower level of bioaccumulation) by creating a composite sample from a larger number of small individuals than by compositing a few individuals of larger species. 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 preparing two samples in this study.

1.3.8 Sediment Toxicity

Sediment toxicity testing has been used to evaluate the contaminant levels of freshwater harbors and rivers, as well as estuaries, marine bays, and marsh lands. Most of its use in the past has been in evaluating sites that were known or suspected to be highly contaminated. EMAP-SW is the first program to use sediment toxicity on such a large scale in freshwater lakes and streams. Sediment toxicity tests, using the freshwater amphipod *Hyalella azteca*, will be used to determine the status of sediment contamination in streams. Sediment toxicity can also be used to indicate the affects of non-contaminant stressors, such as physical habitat degradation. The measurements for sediment toxicity are simple and easy to determine. The survival in each sample is determined at the end of the test and compared to survival in a test using a “reference” sediment.

1.4 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); detailed procedures will be published as a separate document.

This manual is organized to follow the sequence of field activities during the 1-day site visit. Section 2 presents a general overview of all field activities. Section 3 presents those procedures that are conducted at a “base” location before and after a stream site visit. Section 4 presents the procedures for verifying the site location and defining a reach of the stream where subsequent sampling and data collection activities are conducted. Sections 5 through 14 describes the procedures for collecting samples and field measurement data for various condition and stressor indicators. Specific procedures associated with each indicator are presented in standalone tables that can be copied, laminated, and taken into the field for quick reference. Section 15 describes the final activities that are conducted before leaving a stream site. Appendix A contains a list of all equipment and supplies required by a crew to complete all field activities at a stream. 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 provides a complete set of blank field data forms as used in 1997. Appendix D contains a list of vertebrate species names and corresponding species codes developed for use in the Mid-Atlantic region. This information documents the common and scientific names used for the various Mid-Atlantic studies, and also provides an example that can be adapted for use in other areas of the country. Appendix E presents a modified protocol for collecting benthic macroinvertebrates that has been used in EMAP studies in some parts of the U.S.

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

1.5 QUALITY ASSURANCE

Large-scale and/or long-term monitoring programs such as those envisioned for EMAP require a rigorous quality assurance (QA) program that can be implemented consistently by all participants throughout the duration of the monitoring period. Quality assurance is a required element of all EPA-sponsored studies that involve the collection of environmental data (Stanley and Verner, 1986). Field teams should be provided a copy of the QA project plan (e.g., Chaloud and Peck, 1994 for EMAP-SW activities). The QA plan contains more detailed information regarding QA/QC activities and procedures associated with general field operations, sample collection, measurement data collection for specific indicators, and data reporting activities.

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

Brian H. Hill [1], Frank H. McCormick[1], James M. Lazorchak[1], Donald J. Klemm[1], Philip A. Lewis[1, 2], Victoria C. Rogers[3, 4], and Michael K. McDowell[3]

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

2.1 DAILY OPERATIONAL SCENARIO

The field team is divided into two groups, termed the "Geomorphs" and the "Biomorphs," that reflect their initial responsibilities more than their expertise. The geomorphs are primarily responsible for conducting the intensive physical habitat characterization. The biomorphs are primarily responsible for collecting biological samples. Table 2-1 provides the estimated time required to conduct various field activities. Figure 2-1 presents the general sequence of activities conducted at each stream reach.

Upon arrival at a stream site, the geomorphs are responsible for verifying and documenting the site location, determining the length of stream reach to be sampled, and establishing the required transects (Section 4). The biomorphs collect samples and field measurements for water chemistry (Section 5) and determine stream discharge (Section 6). The biomorphs also collect sediment for the sediment metabolism determination (Section 9)

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

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

[3] OAO, Inc., 200 SW 35th St., Corvallis, OR 97333.

[4] Current address: Linn-Benton Community College,6500 SW Pacific Blvd., Albany, OR 97321.

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

Activity	Group	Est. Time Required
Site verification and establishing sampling reach and transects	Geomorphs (2 persons)	2 hours
Water chemistry sampling and stream discharge determination	Biomorphs (2 persons)	1 hour
Collecting and processing benthos, periphyton and sediment metabolism samples	Biomorphs (2 persons)	3.5 hours
Intensive physical habitat characterization	Geomorphs (2 pesons)	2 to 3 hours
Aquatic vertebrate sampling and processing	Geomorphs and Biomorphs (4 persons)	2 to 5 hours
Rapid habitat assessment Visual stream assessment	Biomorphs (2 persons)	0.5 hours
Sample tracking and packing	Geomorphs (2 persons)	1 hour
SUMMARY	28 to 32 person-hours	7 to 8 hours per team

and sediment toxicity testing (Section 10), and collect periphyton and benthos samples (Sections 8 and 11, respectively). The geomorphs conduct the intensive physical habitat characterization (Section 7). Both groups are involved with collecting aquatic vertebrates (Section 12) and preparing samples for fish tissue contaminants (Section 13). Finally, the biomorphs conduct a habitat characterization based on the Rapid Bioassessment Protocols (RBP; Plafkin et al., 1989) and a visual stream assessment (Section 14), while the geomorphs prepare samples for transport and shipment (Section 3).

2.2 GUIDELINES FOR RECORDING DATA AND INFORMATION

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

It is imperative that field and sample information be recorded accurately, consistently, and legibly. Measurement data that cannot be accurately interpreted by others

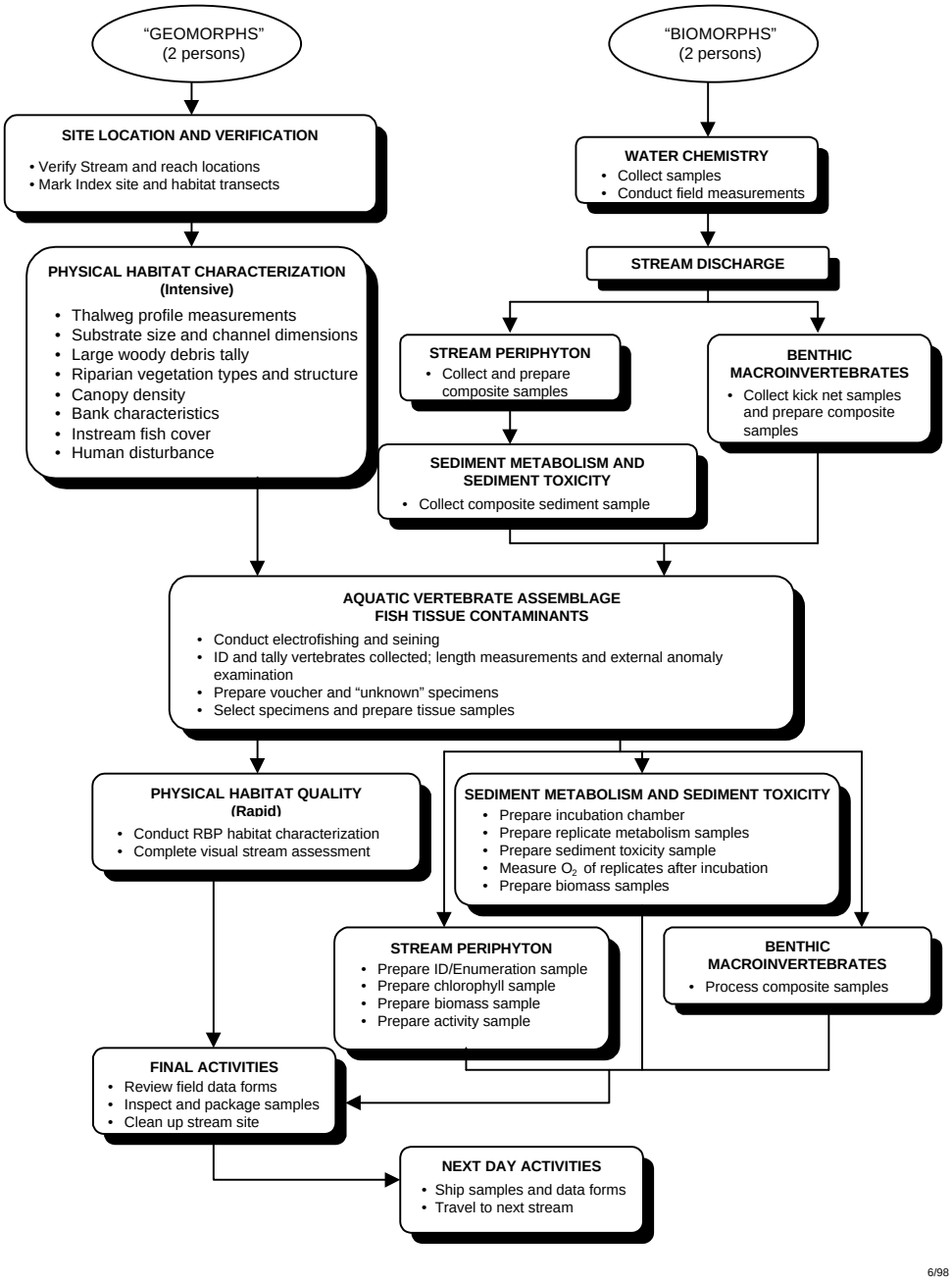

Figure 2-1. General sequence of stream sampling activities (modified from Chaloud and Peck, 1994).

besides the field teams, and/or samples with incorrect or illegible information associated with them, are lost to the program. The cost of a sampling visit coupled with the short index period severely limits the ability to re-sample a stream because the initial information recorded was inaccurate or illegible. Some guidelines to assist field personnel with recording information are presented in Table 2-2. Examples of completed data forms and labels are presented in the sections describing field sampling and measurement procedures for different indicators, and a complete set of blank field data forms are included as Appendix C.

2.3 SAFETY AND HEALTH

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

2.3.1 General Considerations

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

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

TABLE 2-2. 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 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. Q Failed quality control check; re-measurement not possible. U Suspect measurement; re-measurement not possible. F*n* Miscellaneous flags (*n*=1, 2, etc.) assigned by a field team during a particular sampling visit (also used for qualifying samples). Explain 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 stream. Data forms from all teams are reviewed for completeness, accuracy, and legibility before transfer to the information management staff.

(continued)

TABLE 2-2 (Continued)

Activity	Guidelines
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) F*n* 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.

If boats are used to access sampling sites, personnel must consider and prepare for hazards associated with the operation of motor vehicles, boats, winches, tools, and other incidental equipment. Boat operators should be familiar with U.S. Coast Guard rules and regulations for safe boating contained in a pamphlet, "Federal Requirements for Recreational Boats, " available from a local U.S. Coast Guard Director or Auxiliary or State Boating Official (U.S. Coast Guard, 1987). All boats with motors must have fire extinguishers, boat horns, life jackets or flotation cushions, and flares or communication devices.

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

TABLE 2-3. GENERAL HEALTH AND SAFETY CONSIDERATIONS

Training:
! First aid
! Cardiopulmonary resuscitation (CPR)
! Vehicle safety (e.g., operation of 4-wheel drive vehicles)
! Boating and water safety (if boats are required to access sites)
! Field safety (e.g., weather conditions, personal safety, orienteering, reconnaissance of sites prior to sampling
! Equipment design, operation, and maintenance
! Electrofishing safety
! Handling of chemicals and other hazardous materials
Communications
! Check-in schedule
! Sampling itinerary (vehicle used and its description, time of departure, travel route, estimated time of return)
! Contacts for police, ambulance, fire departments, search and rescue personnel
! Emergency services available near each sampling site and base location
Personal Safety
! Field clothing and other protective gear
! Medical and personal information (allergies, personal health conditions)
! Personal contacts (family, telephone numbers, etc.)
! Physical exams and immunizations

certified electrofishing training course. Reynolds (1983) and Ohio EPA (1990) provide additional information regarding electrofishing safety procedures and practices. 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, sun-stroke, drowning, or other dangers. Field personnel should be able to swim. Chest waders made of rubberized or neoprene material and suitable footwear must always be worn with a belt to prevent them from filling with water in case of a fall. The use of a life jacket is advisable at dangerous wading stations if one is not a strong swimmer because of the possibility of sliding into deep water.

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

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

2.3.2 Safety Equipment and Facilities

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

2.3.3 Safety Guidelines for Field Operations

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

TABLE 2-4. GENERAL SAFETY GUIDELINES FOR FIELD OPERATIONS

- **!** Two persons (three to four persons for electrofishing) must be present during all sample collection activities, and no one should be left alone while in the field.
- **!** Exposure to stream water and sediments should be minimized as much as possible. Use gloves if necessary, and clean exposed body parts as soon as possible after contact.
- **!** All electrical equipment must bear the approval seal of Underwriters Laboratories and must be properly grounded to protect against electric shock.
- **!** Use heavy gloves when hands are used to agitate the substrate during collection of benthic macroinvertebrate samples and when turning over rocks during hand picking.
- **!** Use appropriate protective equipment (e.g., gloves, safety glasses) when handling and using hazardous chemicals
- **!** Persons working in areas where poisonous snakes may be encountered must check with the local Drug and Poison Control Center for recommendations on what should be done in case of a bite from a poisonous snake.

  If local advice is not available and medical assistance is more than an hour away, carry a snake bite kit and be familiar with its use.
- **!** Any person allergic to bee stings, other insect bites, or plants must take proper precautions and have any needed medications handy.
- **!** Field personnel should also protect themselves against the bite of deer or wood ticks because of the potential risk of acquiring pathogens that cause Rocky Mountain spotted fever and Lyme disease.
- **!** All field personnel should be familiar with the symptoms of hypothermia and know what to do in case symptoms occur. Hypothermia can kill a person at temperatures much above freezing (up to 10^{o}C or 50^{o}F) if he or she is exposed to wind or becomes wet.
- **!** Handle and dispose of chemical wastes properly. Do not dispose any chemicals in the field.

health (carcinogenic) if utilized improperly. Chemical wastes can cause various hazards due to flammability, explosiveness, toxicity, causticity, or chemical reactivity. All chemical wastes must be discarded according to standardized health and hazards procedures (e.g., National Institute for Occupational Safety and Health [1981]; U.S. EPA [1986]).

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SECTION 3
BASE LOCATION ACTIVITIES

Donald J. Klemm[1], Brian H. Hill[1], Frank H. McCormick[1], and Michael K. McDowell[2]

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

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

3.1 ACTIVITIES BEFORE EACH STREAM VISIT

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

3.1.1 Confirming Site Access

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

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1 U.S. EPA, National Exposure Research Laboratory, Ecological Exposure Research Division, 26 W. Martin L. King Dr., Cincinnati, OH.

2 OAO, Inc., c/o U.S. EPA, 200 SW 35th St., Corvallis, OR 97333

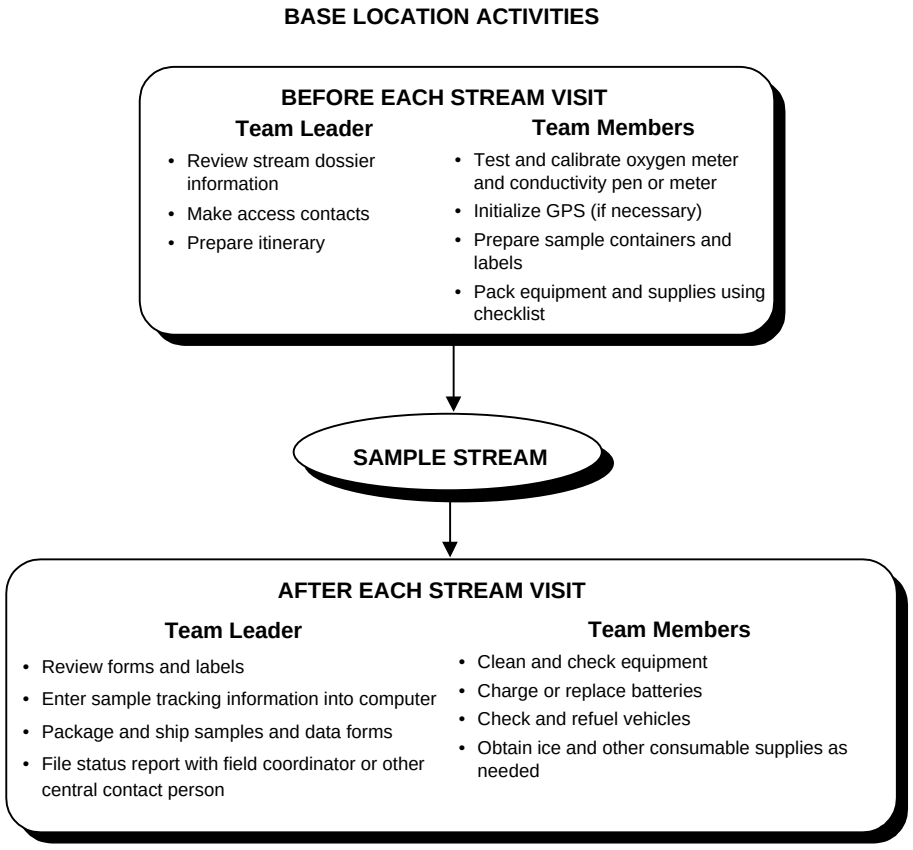

Figure 3-1. Activities conducted at base locations.

the dossier should be contacted to confirm permission to sample and identify any revisions to the information contained in the dossier.

3.1.2 Daily Sampling Itinerary

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

3.1.3 Instrument Inspections and Performance Tests

Each field team is required to test and calibrate instruments prior to departure for the stream site. Field instruments include a global positioning system (GPS) receiver, a current velocity meter, a conductivity pen (or 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 stream site visit. The inspection and testing procedure, based on the use of Yellow Springs Instruments (YSI) Model 53 oxygen meters, is summarized in Figure 3-2. Some modification to the procedure may be necessary for other models or types of dissolved oxygen meters.

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

After inspecting the meter and probe, attempt to calibrate it, following the instructions in the instrument operating manual. Do not record the calibration information obtained during the performance test. The meter is calibrated again at each stream site, 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 Pens or Conductivity Meters--

If conductivity "pens" are being used, check the pen for outward signs of fouling daily. Refer to the instrument manual for probe cleaning instructions. Do not touch the electrodes inside the probe with any object. Always keep the pen's electrode moist by keeping deionized water in the pen cap. If deionized water is not available, use streamwater or tap water rather than let the electrode dry out. Before using a pen which has been stored dry, soak the electrodes in deionized water (by filling the caps) for 24 hours. If conductivity meters are used, 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.

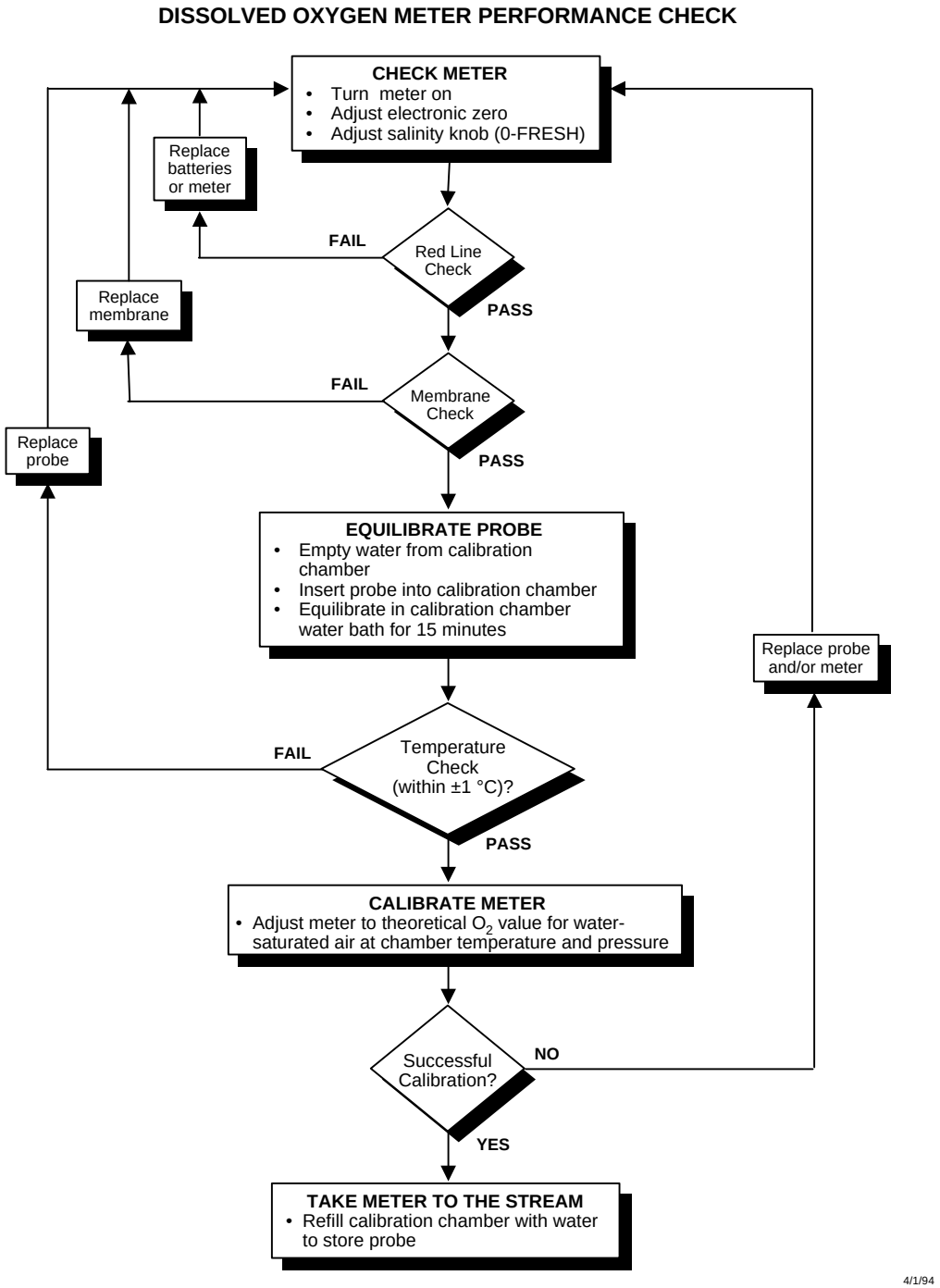

Figure 3-2. Performance test procedure for a dissolved oxygen meter.

The operation of the conductivity pen or 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 conductivity 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 pen or conductivity meter by following the procedure presented in Table 3-2.

3.1.3.4 Current Velocity Meters--

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

3.1.4 Preparation of Equipment and Supplies

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

Check the inventory of equipment and supplies prior to departure using the stream-visit checklists presented in Appendix A. Pack meters, probes, and sampling gear in such a way as to minimize physical shock and vibration during transport. If necessary, prepare stock preservative solutions as described in Table 3-1. Follow the regulations of the Occupational Safety and Health Administration (OSHA) for handling and transporting hazardous materials such as formalin and ethanol. Regulations pertaining to formalin are in the Code of Federal Regulations (CFR; specifically 29 CFR 1910.1048). These requirements should be summarized for all hazardous materials being used for the project and provided to field personnel. Transport formalin and ethanol in appropriate containers with absorbent material.

TABLE 3-1. STOCK SOLUTIONS, USES, AND INSTRUCTIONS FOR PREPARATION

SOLUTION	USE	PREPARATION
Bleach (10%)	Clean seines, dip nets, kick nets, or other equipment that is immersed in the stream	Dilute 400 mL chlorine bleach solution to 4 L with tap water.
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 :S/cm at 25 /C)[a] 1:200 dilution of standard stock solution with reagent water (theoretical conductivity = 37.6 :S/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.

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

TABLE 3-2. PERFORMANCE CHECK OF CONDUCTIVITY PENS OR CONDUCTIVITY METERS

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

2. Swirl the electrodes (pen) or probe (meter) 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 pen or probe in deionized water. Store as described in the operating manual and package the pen or meter for transport to the stream site.

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

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

   If the measured value is still not acceptable, clean the pen or 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 pen or meter.

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

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

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

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

3.2 ACTIVITIES AFTER EACH STREAM VISIT

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

3.2.1 Equipment Care

Equipment cleaning procedures are given in Table 3-4. Inspect all equipment, including nets, and clean off any plant and animal material. This effort ensures that introductions of nuisance species do not occur between streams, and prevents possible cross-contamination of samples. If nets cannot be cleaned thoroughly using water and detergent, clean and disinfect them with a 10 percent chlorine bleach solution (Table 3-1). 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.

Figure 3-3. Sample container labels.

3.2.2 Sample Tracking, Packing, and Shipment

Each field team packs and ships samples from each stream visit as soon as possible after collection, normally the day following a stream visit. Field teams must be provided with specific information for the shipping destinations, contact persons, and the required shipping schedule for each type of sample.

Sample tracking information (including sample types, sample ID numbers, and other field-related information that is required by the laboratory to conduct analyses and associate results to a specific sample and stream site) is recorded during the packing process.

TABLE 3-4. EQUIPMENT CARE AFTER EACH STREAM VISIT

1. Clean for biological contaminants (e.g., plant and animal material).

    - Prior to departing a stream, drain all water from live wells and buckets used to hold and process fish.
    - Inspect sampling gear for evidence of plant fragments and remove any fragments observed.
    - At the stream or base site, dry out seines, 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 percent bleach solution.

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 pens or conductivity meter probes are rinsed with deionized water and are stored moist.
    - Rinse all beakers used to collect water chemistry samples three times with deionized water to prevent contamination of the next stream sample. Place the beakers in a 1-gallon self-sealing plastic bag with a cubitainer for use at the next stream.

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

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

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

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

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

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

Depending upon the project, this information may be recorded manually onto paper forms, or otherwise recorded electronically into a portable computer, using such tools as barcode scanners and customized entry and reporting software. Procedures for conducting sample tracking activities should be provided to each field team by the information management staff, possibly as a separate operations manual or handbook.

The sample tracking system should also identify the intermediate and final destinations for each sample. In some cases, intermediate storage "depots" may be used to accumulate samples prior to shipment to the support laboratory. The tracking system should 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 3-5. When shipping samples using ice, use fresh ice. Use block ice when available; it should be sealed in a large plastic bags. If block ice is not available, contain the ice in several self-sealing plastic bags. Label each bag of ice as "ICE" with an indelible marker to prevent any leakage of meltwater from being misidentified by couriers as a possible hazardous material spill.

Water chemistry 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 samples, place a large (30-gallon) plastic bag in an insulated shipping container (e.g., a plastic or metal cooler). The sample labels on the cubitainer and syringes should be completely covered with clear tape to prevent damage from water or condensation during shipment. Place the four syringes into a separate plastic container for shipment. Place the cubitainer and syringe container into a second large plastic bag and close. Place the bag containing the samples inside the plastic bag lining the shipping container. Place bags of ice around the bag of samples, but inside the plastic bag lining the shipping container. Then close the outer plastic bag. Seal the cooler with clear tape. Place the required sample tracking forms in the shipping container and close it. Seal the container with shipping tape and affix any required shipping-related labels to the outside of the container. Attach an adhesive plastic sleeve to the lid of the container and insert any required shipping forms.

Sediment toxicity samples can be held for extended periods (e.g., a week), if they can be kept refrigerated in the field. Transport or ship sediment toxicity samples in a separate container from water chemistry samples if possible to avoid possible contamination of

TABLE 3-5. GENERAL GUIDELINES FOR PACKING AND SHIPPING SAMPLES

Sample Type (container)	Guidelines
Samples requiring refrigeration (4 *I*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).
Sediment Toxicity (1-gal plastic bag)	Ship on day of collection or within 24 hr by overnight courier. Keep chilled if extended storage time in the field is necessary. Use a separate shipping container from water chemistry samples. Package and ship using the same instructions as for water chemistry samples. If available, place the plastic bag containing the sample into a plastic container to protect it during transport and shipment. Cover labels completely with clear tape. 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 *I*C) within 24 hours of collection	
Periphyton chlorophyll (filter in aluminum foil)	If samples cannot be kept frozen in the field, ship on day of collection or within 24 h by overnight courier. Cover the label completely with clear tape. Protect samples from meltwater if ice is used by double bagging ice and placing samples in a plastic container. Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form (or other sample tracking report). If dry ice is used to transport or ship samples, special shipping containers, outside labeling, and shipping forms may be required.
Periphyton biomass (filter in a numbered container)	
Periphyton activity (50-mL centrifuge tube)	
Sediment metabolism (50-mL centrifuge tubes)	
Fish Tissue (aluminum foil; two 30-gal plastic bags)	If samples cannot be kept frozen in the field, ship on day of collection or within 24 h by overnight courier. Cover labels completely with clear tape. Label on each bag should have identical Sample ID number assigned. Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form (or other sample tracking report). Protect samples from meltwater if ice is used by double bagging ice. Special shipping containers, outside labeling, and shipping forms may be required for shipments containing dry ice.

(continued)

TABLE 3-5. (Continued)

Sample Type (container)	Preservative	Guidelines
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. 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.
Fish Specimens (1-L and/or 4-L jars)	10 % buffered formalin	
Samples requiring preservation in ethanol		
Benthic Macro-invertebrates (500-mL or 1-L jars)	70 % ethanol	Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form (or other sample tracking report). Special shipping containers, outside labeling, and shipping forms may be required for shipments containing ethanol.

the water samples. Pack and ship sediment toxicity samples using the same type of insulated container and plastic bag arrangement as described above for water chemistry samples. If available, sediment toxicity samples can be placed inside a plastic container (similar to the one used for syringe samples) to protect it during shipment.

Samples requiring freezing (Table 3-5) may be stored in the field in a portable freezer or on dry ice for a short period (e.g., one week). If only ice is available for field storage, the samples should be shipped to the laboratory as soon as possible after collection, using fresh ice to keep them as cold as possible. When using ice, double bag the ice and tape the last bag shut to prevent contamination of samples by melting ice. If possible, place samples into a sealed plastic container to protect them from meltwater. Dry ice may also be used for shipping. Note that dry ice is considered a hazardous material, and requires special shipping containers, shipping labels, and shipping forms for ground or air transport. If dry ice is used, the requirements and directions for packing and shipping samples ice should be provided to each field team.

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 should be provided to each field team.

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

3.3 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 Appendix

BASE LOCATION ACTIVITIES

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

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

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

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SECTION 4
INITIAL SITE PROCEDURES

by
Alan T. Herlihy[1]

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

4.1 SITE VERIFICATION ACTIVITIES

4.1.1 Locating the Index Site

Stream sampling points were chosen from the “blue line” stream network represented on 1:100,000- scale USGS maps, following a systematic randomized selection process developed for EMAP stream sampling. Sample sites were then marked with an "X" on finer-resolution 1:24,000-scale USGS maps. This spot is referred to as the “index site” or “**X-site**”. The latitude/longitude of the X-site will be listed on a stream information sheet that is part of the dossier compiled for each stream (see Section 3).

Complete a verification form for each stream visited (regardless of whether you end up sampling it), following the procedures described in Table 4-1. While traveling from a base location to a site, record a detailed description of the route taken on page 1 of the Verification Form (Figure 4-1). This information will allow others to find the site again in the future. Upon reaching the X-site for a stream, confirm its location and that the team is

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

TABLE 4-1. SITE VERIFICATION PROCEDURES

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

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

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

4. Scan the stream channel upstream and downstream **from the X-site**, 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-Not Wadeable Stream (river): The stream channel is too deep to be safely sampled by wadeable stream procedures.
   If over half of the reach is unwadeable, classify the reach as unwadeable.
   If more than half of the reach appears to be wadeable (e.g., only a couple of deep pools), classify the reach as "Regular-Wadeable" and sample those portions of the reach that can be safely sampled.

C. Intermittent Stream: The flow of water is not continual, but the channel is wet. Sample using modified procedures.

D. Dry Channel: A discernible stream channel is present but there is no water at the site. Sample using modified procedures.

E. 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.
   If the impounded stream, however, is still wadeable, record the stream as Altered (Target category E) and sample the stream.

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 heavy wetlands, steep gorge or other barrier that prohibits safe entry.

B. No Permission: You are denied access to the site 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.

Reviewed by (initial): DP

VERIFICATION FORM - STREAMS/RIVERS

SITE NAME: Mill Creek DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999___ TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

STREAM/RIVER VERIFICATION INFORMATION

STREAM/RIVER VERIFIED BY (X all that apply): ☒ GPS ☒ Local Contact ☐ Signs ☒ Roads ☒ Topo. Map

☐ Other (Describe Here): ☐ NOT VERIFIED (Explain in Comments)

COORDINATES	LATITUDE (dd mm ss) North	LONGITUDE (ddd mm ss) West	TYPE OF GPS FIX	Are GPS Coordinates w/l 10 Sec. of map?
MAP:	38° 10' 25"	77° 44' 75"	☐ 2D	☒ YES
GPS:	38° 10' 26"	77° 44' 78"	☒ 3D	☐ NO

INDEX SITE STATUS - X ONE BOX FROM ONE SECTION ONLY

SAMPLEABLE

- ☒ Regular - Wadeable
- ☐ Regular - Not Wadeable
- ☐ 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 Barnesville, go E on county road 996 to Smithtown Road (~5 mi.). Turn S onto Smithtown Rd. and go 0.6 mi to gravel road on left. Turn onto gravel road and go 0.5 mi. to farmhouse on right side of road. Owner will unlock gate to drive to stream.

GENERAL COMMENTS

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

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

at the correct stream. Use all available means to accomplish this, and record the information on page 1 of the Verification Form (Figure 4-1).

4.1.2 Determining the Sampling Status of a Stream

Not all chosen stream sites will turn out to be streams. On the basis of previous synoptic surveys, it was found that the maps are far from perfect representations of the stream network. A significant part of EMAP is the estimation of the actual extent of stream length in the area. After the stream and location of the X-site are confirmed, evaluate the stream reach surrounding the X-site and classify the stream into one of three major sampling status categories (Table 4-1). The primary distinction between "Non-target" and "Target" streams is based on the presence of a defined stream channel and its depth.

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. For one, it is often unsafe to be in the water during such times. In addition, biological and chemical conditions during episodes are often quite different from those during baseflow. On the other hand, sampling cannot be restricted to only strict baseflow conditions. It would be next to impossible to define "strict baseflow" with any certainty at an unstudied site. Such a restriction would also greatly shorten the index period when sampling activities can be conducted. Thus, some compromise is necessary regarding whether to sample a given stream because of storm events. To a great extent, this decision is based on the judgment of the field team. Some guidelines to help make this decision are presented in Table 4-2. 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 is an optional activity, but should be considered if the site has unusual natural or man-made features associated with it. If you do take any photographs at a stream, start the sequence with one photograph of an 8.5 × 11 inch piece of

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.
- **!** Do not sample if it is unsafe to wade in the majority of the stream reach.
- **!** Keep an eye on the weather reports and rainfall patterns. Do not sample a stream during periods of prolonged heavy rains.
- **!** If the stream seems to be close to normal summer flows, and does not seem to be unduly influenced by storm events, go ahead and sample it, even if it has recently rained or is raining.

paper with the stream ID and date printed in large letters. After the photo of the stream ID information, take at least two photographs at the X-site, one in the upstream direction and one downstream. Take any additional photos you find interesting after these first three pictures. For pictures of aquatic vertebrates (see Section 12) or other small objects, place the paper with the stream ID and date in each snapshot.

4.2 LAYING OUT THE SAMPLING REACH

Unlike chemistry, which can be measured at a point, most of the biological and habitat structure measures require sampling a certain length of a stream to get a representative picture of the ecological community. Previous EMAP pilot studies have suggested that a length of 40 times the channel width is necessary to collect at least 90% of the fish species occurring in the stream reach. Thus, a support reach that is 40 channel widths long around the X-site is required to characterize the community and habitat associated with the sampling point. Establish the sampling reach about the X-site using the procedures described in Table 4-3. 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 transects used for physical habitat characterization (Section 7), and specific sampling points on each cross-section transect for later collection of periphyton samples (Section 8) and benthic macroinvertebrate samples (Section 11).

TABLE 4-3. LAYING OUT THE SAMPLING REACH

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

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

2. Check the condition of the stream upstream and downstream of the X-site by having one team member go upstream and one downstream. Each person proceeds until they can see the stream to a distance of 20 times the average channel width (equal to one-half the sampling reach length) determined in Step 1 from the X-site.

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

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

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

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

4. Starting back at the X-site (or the new midpoint of the reach if it had to be adjusted as described in Step 3), measure a distance of 20 channel widths down the middle of the stream using a tape measure. Be careful not to "cut corners". Enter the channel to make measurements only when necessary to avoid disturbing the stream channel prior to sampling activities. This endpoint is the downstream end of the reach, and is flagged as transect “A”.

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

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

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

Reviewed by (initial): DF

VERIFICATION FORM - STREAMS/RIVERS (continued)

SITE NAME: Mill Creek DATE: 7 / 15 / 97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999 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	
3	75	75	

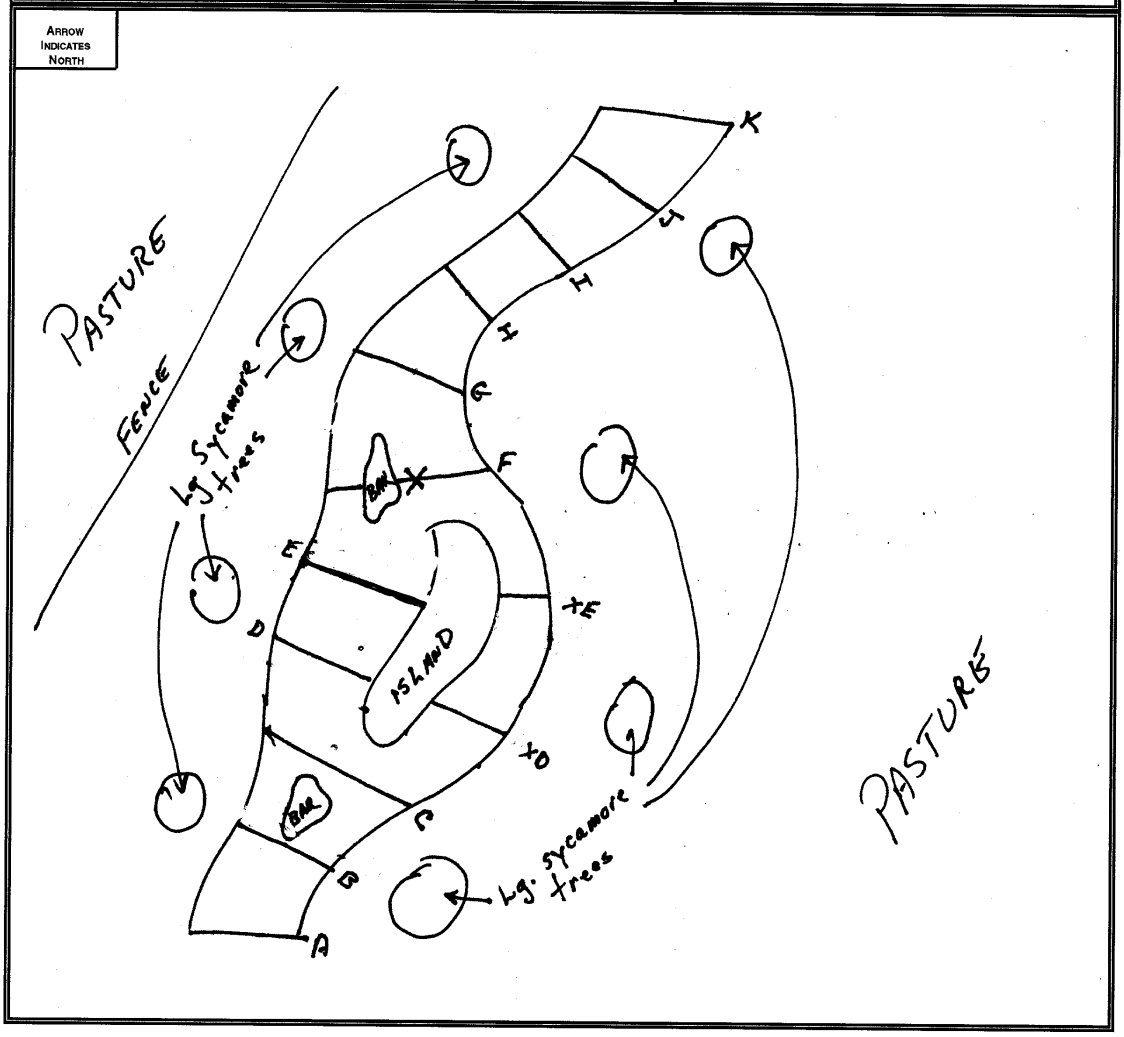

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Figure 4-2. Verification Form (page 2).

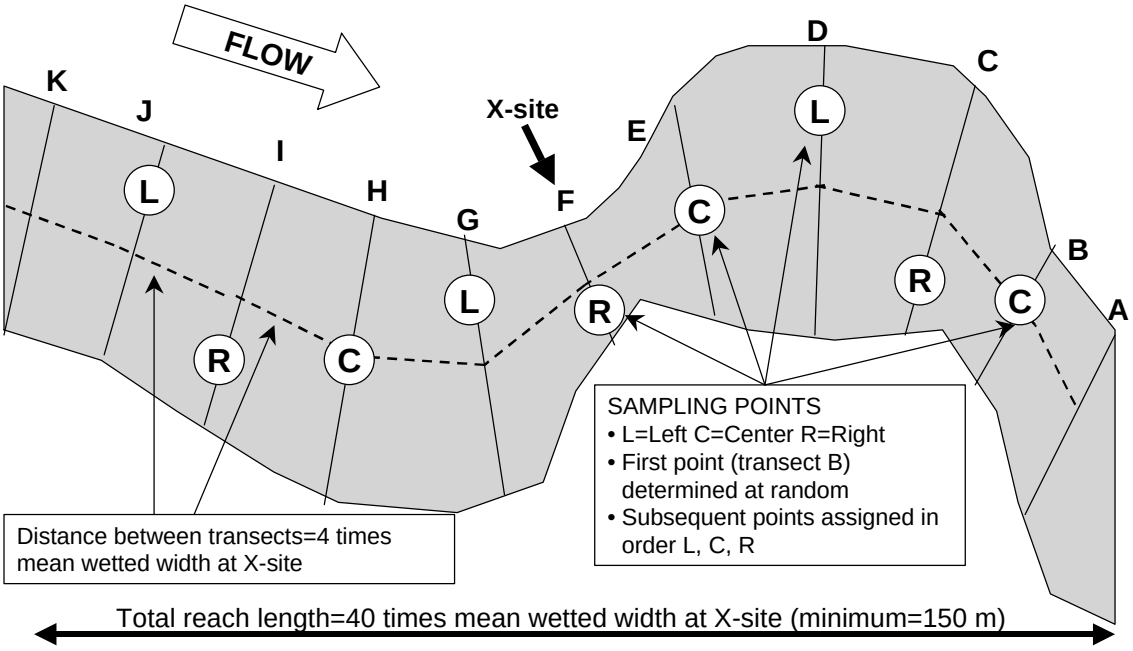

Figure 4-3. Sampling reach features.

There are some conditions that may require adjusting the reach about the X-site (i.e., the X-site no longer is located at the midpoint of the reach) to avoid features we do not wish to sample across. Do not proceed upstream into a lower order stream or downstream into a higher order stream when laying out the stream reach (order is based on 1:100,000 scale maps). If such a confluence is reached, note the distance and flag the confluence as the endpoint of the reach. Make up for the loss of reach length by moving ("sliding") the other end of the reach an equivalent distance away from the X-site. Similarly, if you run into a lake, reservoir, or pond while laying out the reach, stop, flag the lake/stream confluence as the reach end, and make up for the loss of reach length by moving the other end of the reach an equivalent distance from the X-site. Do not “slide” the reach so that the X-site falls outside of the reach boundaries. Also, do not “slide” a reach to avoid man-made obstacles

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

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

4.3 MODIFIED PROCEDURES FOR DRY AND INTERMITTENT STREAMS

The full complement of field data and samples cannot be collected from streams that are categorized as “Dry Channel” or “Intermittent” (Table 4-1). Physical habitat information (Section 7) is collected in all streams. Intermittent streams will have some cross-sections with biological measurements and some with none. Totally dry channels will have no biological sampling. Modified procedures for dry and intermittent streams are presented in Table 4-4.

Samples and measurements for water chemistry (Section 5) should be collected at the X-site (even if the reach has been adjusted by “sliding” it). If the X-site is dry, the sample and chemical measurements are taken from a location having water with a surface area greater than 1 m^2 and a depth greater than 10 cm.

All data for the physical habitat indicator (Section 7) are collected from all streams, regardless of the amount of water present in the channel or at the transects. Depth measurements along the deepest part of the channel (the “thalweg”) are obtained along the entire sampling reach for ALL target streams, whether they are dry, intermittent, or completely flowing. The thalweg profile provides a record of the "water" status of the stream for future comparisons (e.g., the percent of length with intermittent pools or no water). Other measurements associated with characterizing riparian condition, substrate type, etc. are useful to help infer conditions in the stream when water is flowing.

4.4 EQUIPMENT AND SUPPLIES

A list of the equipment and supplies required to conduct the stream verification and to lay out the sampling reach is presented in Figure 4-4. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of

TABLE 4-4. MODIFICATIONS FOR DRY CHANNELS AND INTERMITTENT STREAMS

Water Chemistry

! If the X-site is dry but there is flowing water or a pool of water having a surface area greater than 1 m^2 and a depth greater than 10 cm somewhere along the defined sampling reach, take the water sample and water chemistry measurements at the pool or flowing water location that is nearest to the X-site. Note that the sample wasn't collected at the X-site and where on the reach the sample was collected on the field data form.

! Do not collect a water sample if there is no acceptable location within the sampling reach. Record a "K" flag for the chemistry sample on the sample collection form and explain why the sample was not collected in the comments section of the form.

Physical Habitat Characterization, Periphyton, Sediment Metabolism, and Benthic Macroinvertebrates

! Obtain a complete thalweg profile for the entire reach, even if the channel is completely dry. At points where channel is dry, record depth as 0 cm and wetted width as 0 m.

! At each of the transects (cross sections), classify the stream as:

DRY CHANNEL: No surface water anywhere in cross section;
Collect all physical habitat data. Use the unvegetated area of the channel to determine the channel width and the subsequent location of substrate sampling points. Record the wetted width as 0 m. Record substrate data at the sampling points located in the unvegetated, but dry, channel.

DAMP CHANNEL: Wet spots in cross section but NO flowing water or pools > 10 cm deep;
Collect all physical habitat data.
Collect periphyton samples from the wet spots. These are great environments for algae.
Collect sediments for metabolism if there are enough fine wet sediments available.
Do not collect a benthic macroinvertebrate sample.

ENDURING POOLS: No flowing water but pools > 10 cm deep;
Collect all data and measurements for physical habitat, periphyton, sediment metabolism, and benthic macroinvertebrate indicators, using standard procedures.

FLOWING WATER: Flowing water in cross section
Collect all data and measurements for physical habitat, periphyton, sediment metabolism, and benthic macroinvertebrate indicators, using standard procedures.

Aquatic Vertebrates

! Do not sample if the entire reach is dry.

! In intermittent streams (including those having damp channels and/or enduring pools), sample any wet areas within the sampling reach that are potential habitat for aquatic vertebrates. **Do not sample downstream of Transect "A" or upstream of Transect "K", even if there appears to be good habitat present.**

EQUIPMENT AND SUPPLIES FOR INITIAL SITE ACTIVITIES

QTY.	Item	
1	Dossier of site and access information	
1	Topographic map with “X-site” marked	
1	Site information sheet with map coordinates and elevation of X-site	
1	GPS receiver and operating manual	
	Extra batteries for GPS receiver	
1	Verification Form	
	Soft lead (#2) pencils	
1	Surveyor’s telescoping leveling rod	
1	50-m fiberglass measuring tape with reel	
2 rolls	Surveyor’s flagging tape (2 colors)	
	Fine-tipped indelible markers to write on flagging	
1	Waterproof camera and film	
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.

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

SECTION 5
WATER CHEMISTRY

by
Alan T. Herlihy[1]

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

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

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

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

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

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

5.1 SAMPLE COLLECTION

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

The procedure to collect a water chemistry sample is described in Table 5-1. The sample is collected from the middle of the stream channel at the X-site, unless no water is present at that location (see Section 4). Throughout the sampling process, it is important to take precautions to avoid contaminating the sample. Rinse all sample containers three times with portions of stream water before filling them with the sample. Many of the streams have a very low ionic strength and can be contaminated quite easily by perspiration from hands, sneezing, smoking, insect repellent, or other chemicals used when collecting other types of samples. Thus, make sure that none of the water sample contacts your hands before going into the cubitainer. All of the chemical analyses conducted using the syringe samples are affected by equilibration with atmospheric carbon dioxide; thus, it is essential that no outside air contact the syringe samples during or after collection.

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

5.2 FIELD MEASUREMENTS

Table 5-2 presents the procedures for obtaining field measurement data for the water chemistry indicator. The conductivity and dissolved oxygen meters are checked in

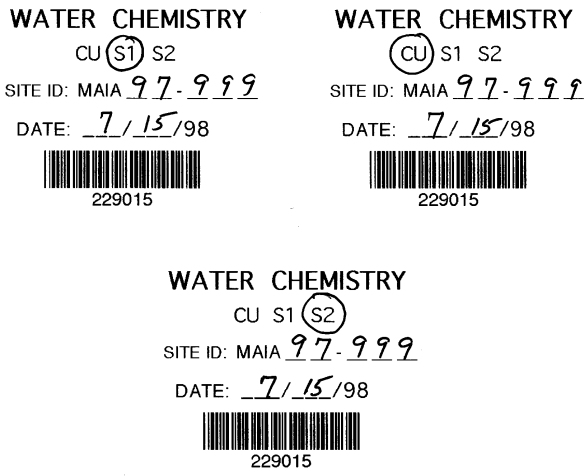

Figure 5-1. Completed sample labels for water chemistry.

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.

5.3 EQUIPMENT AND SUPPLIES

A list of equipment and supplies required to collect samples and field data for the water chemistry indicator is presented in Figure 5-4. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the stream. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

5.4 LITERATURE CITED

Kaufmann, P., A. Herlihy, J. Elwood, M. Mitch, S. Overton, M. Sale, J. Messer, K. Reckhow, K. Cougan, D. Peck, J. Coe, A. Kinney, S. Christie, D. Brown, C. Hagley, and Y. Jager. 1988. *Chemical Characteristics of Streams in the Mid-Atlantic and Southeastern*

TABLE 5-1. SAMPLE COLLECTION PROCEDURES FOR WATER CHEMISTRY

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

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

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

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

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

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

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

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

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

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

10. Record the barcode number (Sample ID) on the Sample Collection Form along with the pertinent stream information (stream name, ID, date, etc.). Note anything that could influence sample chemistry (heavy rain, potential contaminants) in the Comments section. If the sample was collected at the X-site, record an "X" in the "STATION COLLECTED" field. If you had to move to another part of the reach to collect the sample, place the letter of the nearest transect in the "STATION COLLECTED" field. Record more detailed reasons and/or information in the Comments section.

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

Reviewed by (initial): DP

SAMPLE COLLECTION FORM - STREAMS (continued)

SITE NAME: Mill Creek DATE: 7/15/97 VISIT: ☒1 ☐2 ☐__

SITE ID: MAIA97-999__ 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	229015	X		
MICROBIAL				

SEDIMENT TOXICITY SAMPLES

Sample ID (Barcode)	Flag	Comments
229011		

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

Sample ID (Barcode) → 229013

Line	Species Code	Common Name	Number of Individuals	Flag
P1	NOCOLE	Bluehead chub	16	F1

Is composite sample composed of individuals collected from throughout reach? (X) → ☒ Yes ☐ No

If No, Explain:

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

Sample ID (Barcode) → 229012

Line	Species Code	Common Name	Total Length (mm)	Flag
S1	CATOCO	White sucker	128	
S2	CATOCO	White sucker	125	
S3	CATOCO	White sucker	134	
S4	CATOCO	White sucker	128	
S5	CATOCO	White sucker	125	

Is composite sample composed of individuals collected from throughout reach? (X) → ☒ Yes ☐ No

If No, Explain:

Line	Comment or Flag Explanation for FISH TISSUE
P1	F1 = 16 individuals weighed 60 g.

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

TABLE 5-2. 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 (or a conductivity pen) 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. Transfer the probe 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 and area of flowing water near the middle of the channel at the same location where the water chemistry sample is collected. Record the measured conductivity 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.

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

3. After the calibration, submerge the probe in midstream at mid-depth at the same location where the water chemistry sample is collected. Face the membrane of the probe upstream, and allow the probe to equilibrate. Record the measured DO and stream temperature on the Field Measurement Form. 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.

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

Reviewed by (initial): DL

FIELD MEASUREMENT FORM - STREAMS/RIVERS

SITE NAME: MILL CREEK DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999 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: E — Assume X-site unless marked

		FLAG	COMMENTS
QCCS COND µS/CM	72		
STREAM/RIVER COND µS/CM	500		
STREAM/RIVER DO MG/L	4.6		
STREAM/RIVER TEMP °C	20.0		

STREAM/RIVER METABOLISM DETERMINATION

INITIAL O_2 (MG/L)	INITIAL INCUBATION TEMP. (°C)	INCUBATION TIME (24-HR TIME) START (HH:MM)	INCUBATION TIME (24-HR TIME) FINISH (HH:MM)	DURATION OF INCUBATION (HH:MM)	FLAG	COMMENTS
4.9	20.0	11:35	2:40	3:05		

SAMPLE ID (BARCODE)	FINAL O_2 (MG/L)	FLAG	COMMENTS
999991	2.0		
999992	1.9		
999993	1.8		
999994	1.9		
999995	1.1		
BLANK	4.5		

OXYGEN METER CALIBRATION INFORMATION

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

CALIBRATION CHAMBER TEMPERATURE:	27.9 °C	SATURATED O_2 @ TEMP.:	7.83 MG/L
STATION ELEVATION (FROM TOPO. MAP OR ALTIMETER):	1080 FT	ELEVATION CORRECTION FACTOR: ×	0.96
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:	7.52 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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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	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 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for water chemistry	

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

United States. Volume I: Population Descriptions and Physico-Chemical Relationships. EPA 600/3-88/021a. U.S. Environmental Protection Agency, Washington, D.C.

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

SECTION 6
STREAM DISCHARGE

by
Philip R. Kaufmann[1]

Stream discharge is equal to the product of the mean current velocity and vertical cross sectional area of flowing water. Discharge measurements are critical for assessing trends in streamwater acidity and other characteristics that are very sensitive to streamflow differences. Discharge should be measured at a suitable location within the sample reach that is as close as possible to the location where chemical samples are collected (typically the X-site; see Section 5), so that these data correspond.

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

No single method for measuring discharge is applicable to all types of stream channels. The preferred procedure for obtaining discharge data is based on "velocity-area" methods (e.g., Rantz and others, 1982; Linsley et al., 1982). For streams that are too small or too shallow to use the equipment required for the velocity-area procedure, two alternative procedures are presented. One procedure is based on timing the filling of a volume of water in a calibrated bucket. The second procedure is based on timing the movement of a neutrally buoyant object (e.g., an orange) through a measured length of the channel, after measuring one or more cross-sectional depth profiles within that length.

6.1 VELOCITY-AREA PROCEDURE

Because velocity and depth typically vary greatly across a stream, accuracy in field measurements is achieved by measuring the mean velocity and flow cross-sectional area of many increments across a channel (Figure 6-1). Each increment gives a subtotal of the

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

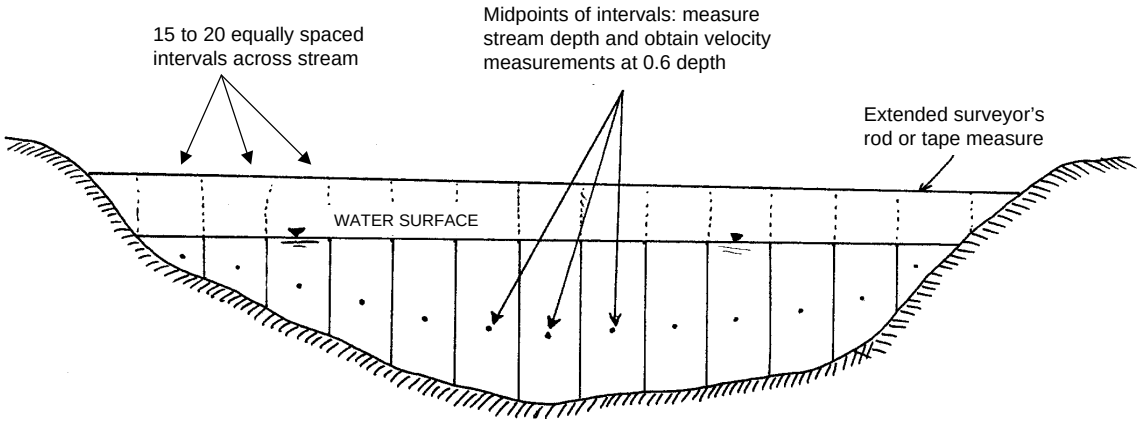

Figure 6-1. Layout of channel cross-section for obtaining discharge data by the velocity-area procedure.

stream discharge, and the whole is calculated as the sum of these parts. Discharge measurements are made **at only one carefully chosen channel cross section within the sampling reach**. It is important to choose a channel cross section that is as much like a canal as possible. A glide area with a "U" shaped channel cross section that is free of obstructions provides the best conditions for measuring discharge by the velocity-area method. You may remove rocks and other obstructions to improve the cross-section before any measurements are made. However, because removing obstacles from one part of a cross-section affects adjacent water velocities, you must not change the cross-section once you commence collecting the set of velocity and depth measurements.

The procedure for obtaining depth and velocity measurements is outlined in Table 6-1. Record the data from each measurement on page 2 of the Field Measurement Form

TABLE 6-1. VELOCITY-AREA PROCEDURE FOR DETERMINING STREAM DISCHARGE

1. Locate a cross-section of the stream channel for discharge determination that has most of the following qualities (based on Rantz and others, 1982):

   - Segment of stream above and below cross-section is straight
   - Depths mostly greater than 15 centimeters, and velocities mostly greater than 0.15 meters/second. Do not measure discharge in a pool.
   - "U" shaped, with a uniform streambed free of large boulders, woody debris or brush, and dense aquatic vegetation.
   - Flow is relatively uniform, with no eddies, backwaters, or excessive turbulence.

2. Lay the surveyor's rod (or stretch a meter tape) across the stream perpendicular to its flow, with the "zero" end of the rod or tape on the left bank, as viewed when looking downstream. Leave the tape tightly suspended across the stream, approximately one foot above water level.

3. Attach the velocity meter probe to the calibrated wading rod. Check to ensure the meter is functioning properly and the correct calibration value is displayed. Calibrate (or check the calibration) the velocity meter and probe as directed in the meter's operating manual. Place an "X" in the "VELOCITY AREA" box in the "STREAM DISCHARGE" section of the Field Measurement Form.

4. Divide the total wetted stream width into 15 to 20 equal-sized intervals. To determine interval width, divide the width by 20 and round up to a convenient number. Intervals should not be less than 10 cm wide, even if this results in less than 15 intervals.

5. Stand downstream of the rod or tape and to the side of the midpoint of the first interval (closest to the left bank if looking downstream).

6. Place the wading rod in the stream at the midpoint of the interval and adjust the probe or propeller so that it is at the water surface. Record the distance from the left bank (in centimeters) and the depth indicated on the wading rod (in centimeters) on the Field Measurement Form.

7. Stand downstream of the probe or propeller to avoid disrupting the stream flow. Adjust the position of the probe on the wading rod so it is at 0.6 of the measured depth below the surface of the water. Face the probe upstream at a right angle to the cross-section, even if local flow eddies hit at oblique angles to the cross-section.

8. Wait 20 seconds to allow the meter to equilibrate, then measure the velocity. Record the value on the Field Measurement Form.

   - For the electromagnetic current meter (e.g., Marsh-McBirney), use the lowest time constant scale setting on the meter that provides stable readings.
   - For the impeller-type meter (e.g., Swoffer 2100), set the control knob at the mid-position of "DISPLAY AVERAGING". Press "RESET" then "START" and proceed with the measurements.

9. Move to the midpoint of the next interval and repeat Steps 6 through 8. Continue until depth and velocity measurements have been recorded for all intervals.

as shown in Figure 6-2. To reduce redundancy and to conserve space, Figure 6-2 shows measurement data recorded for all three procedures. In the field, data will be recorded using only one of the three available procedures.

6.2 TIMED FILLING PROCEDURE

In channels too "small" for the velocity-area method, discharge can sometimes be determined directly by measuring the time it takes to fill a container of known volume. “Small” is defined as a channel so shallow that the current velocity probe cannot be placed in the water, or where the channel is broken up and irregular due to rocks and debris, and a suitable cross-section for using the velocity area procedure is not available. This can be an extremely precise and accurate method, but requires a natural or constructed spillway of free-falling water. If obtaining data by this procedure will result in a lot of channel disturbance or stir up a lot of sediment, wait until after all biological and chemical measurements and sampling activities have been completed.

Choose a cross-section of the stream that contains one or more natural spillways or plunges that collectively include the entire stream flow. A temporary spillway can also be constructed using a portable V-notch weir, plastic sheeting, or other materials that are available onsite. Choose a location within the sampling reach that is narrow and easy to block when using a portable weir. Position the weir in the channel so that the entire flow of the stream is completely rerouted through its notch (Figure 6-3). Impound the flow with the weir, making sure that water is not flowing beneath or around the side of the weir. Use mud or stones and plastic sheeting to get a good waterproof seal. The notch must be high enough to create a small spillway as water flows over its sharp crest.

The timed filling procedure is presented in Table 6-2. Make sure that the entire flow of the spillway is going into the bucket. Record the time it takes to fill a measured volume on the Field Measurement Form as shown in Figure 6-2. Repeat the procedure 5 times. If the cross-section contains multiple spillways, you will need to do separate determinations for each spillway. If so, clearly indicate which time and volume data replicates should be averaged together for each spillway; use additional field measurement forms if necessary.

Reviewed by (initial): DP

FIELD MEASUREMENT FORM - STREAMS (continued)

SITE NAME: MILL CREEK DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

STREAM DISCHARGE

☒ **VELOCITY AREA**

	DIST. FROM BANK (CM)	VELOCITY (M/S) XX.X	DEPTH (CM) XXX	FLAG
1	10	0.30	6	F1
2	20	0.59	6	
3	30	0.37	12	
4	40	0.34	15	
5	50	0.34	15	
6	60	0.43	24	
7	70	0.37	27	
8	80	0.43	40	
9	90	0.37	40	
10	100	0.30	46	
11	110	0.27	37	
12	120	0.27	30	
13	130	0.30	24	
14	140	0.30	18	
15	150	0.15	15	
16		.		
17		.		
18		.		
19		.		
20		.		

☒ **TIMED FILLING**

REPEAT	VOL. (L) XX.X	TIME (S)	FLAG
1	4.0	25.3	F1
2	4.0	24.0	
3	4.0	26.1	
4	4.0	24.8	
5	4.0	25.6	

☒ **NEUTRAL BUOYANT OBJECT**

MEASUREMENT	Cross Section ONE	TWO	THREE
WIDTH (m)	2.5	2.0	3.0
DEPTH 1 (cm)	4	10	5
DEPTH 2 (cm)	5	9	7
DEPTH 3 (cm)	6	9	9
DEPTH 4 (cm)	6	8	7
DEPTH 5 (cm)	2	5	5
FLOAT DISTANCE (m)	5	5	5
FLOAT TIME (s)	10.5	9.8	9.0

FLAG	COMMENTS
F1	Measurements for all three discharge procedures are shown on this form

Flag Codes: K = no measurement or observation made; U = suspect measurement or observation; Q = unacceptable QC check associated with measurement; F1, F2, etc. = miscellaneous flags assigned by each field crew. Explain all flags in comments section.

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Figure 6-2. Field Measurement Form (page 2), showing data recorded for all three discharge measurement procedures.

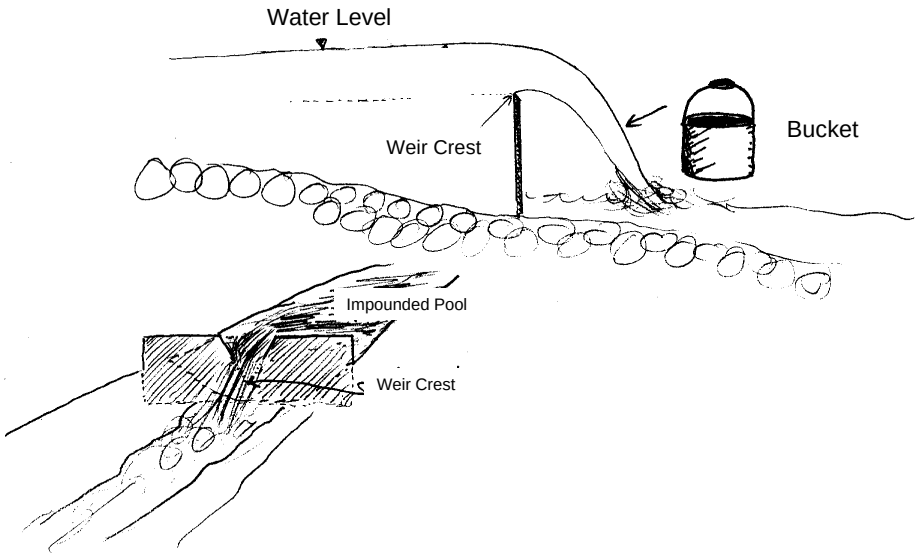


Figure 6-3. Use of a portable weir in conjunction with a calibrated bucket to obtain an estimate of stream discharge.

6.3 NEUTRALLY-BUOYANT OBJECT PROCEDURE

In very small, shallow streams with no waterfalls, where the standard velocity-area or timed-filling methods cannot be applied, the neutrally buoyant object method may be the only way to obtain an estimate of discharge. The required pieces of information are the mean flow velocity in the channel and the cross-sectional area of the flow. The mean velocity is estimated by measuring the time it takes for a neutrally buoyant object to flow through a measured length of the channel. The channel cross-sectional area is determined from a series of depth measurements along one or more channel cross-sections. Since the discharge is the product of mean velocity and channel cross-sectional area, this method is conceptually very similar to the standard velocity-area method.

TABLE 6-2. TIMED FILLING PROCEDURE FOR DETERMINING STREAM DISCHARGE

NOTE: If measuring discharge by this procedure will result in significant channel disturbance or will stir up sediment, delay determining discharge until all biological and chemical measurement and sampling activities have been completed.

1. Choose a cross-section that contains one or more natural spillways or plunges, or construct a temporary one using on-site materials, or install a portable weir using a plastic sheet and on-site materials.

2. Place an “X” in the “TIMED FILLING” box in the stream discharge section of the Field Measurement Form.

3. Position a calibrated bucket or other container beneath the spillway to capture the entire flow. Use a stopwatch to determine the time required to collect a known volume of water. Record the volume collected (in liters) and the time required (in seconds) on the Field Measurement Form.

4. Repeat Step 3 a total of 5 times for each spillway that occurs in the cross section. If there is more than one spillway in a cross-section, you must use the timed-filling approach on all of them. Additional spillways may require additional data forms

The neutrally buoyant object procedure is described in Table 6-3. Examples of suitable objects include oranges, small sponge rubber balls, or small sticks. The object must float, but very low in the water. It should also be small enough that it does not "run aground" or drag bottom. Choose a stream segment that is roughly uniform in cross-section, and that is long enough to require 10 to 30 seconds for an object to float through it. Select one to three cross-sections to represent the channel dimensions within the segment, depending on the variability of width and/or depth. Determine the stream depth at 5 equally spaced points at each cross-section. Three separate times, measure the time required for the object to pass through the segment that includes all of the selected cross-sections. Record data on the Field Measurement Form as shown in Figure 6-2.

6.4 EQUIPMENT AND SUPPLIES

Figure 6-4 shows the list of equipment and supplies necessary to measure stream discharge. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the stream. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

6.5 LITERATURE CITED

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

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

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

1. Place an "X" in the "NEUTRALLY BUOYANT OBJECT" box on the Field Measurement Form.

2. Select a segment of the sampling reach that is deep enough to float the object freely, and long enough that it will take between 10 and 30 seconds for the object to travel. Record the length of the segment in the "FLOAT DISTANCE" field of the Field Measurement Form.

3. If the channel width and/or depth change substantially within the segment, measure widths and depths at three cross-sections, one near the upstream end of the segment, a second near the middle of the segment, and a third near the downstream end of the segment.

   If there is little change in channel width and/or depth, obtain depths from a single "typical" cross-section within the segment.

4. At each cross section, measure the wetted width (m) using a surveyor's rod or tape measure, and record on the Field Measurement Form. Measure the stream depth using a wading rod or meter stick at points approximately equal to the following proportions of the total width: 0.1, 0.3, 0.5, 0.7, and 0.9. Record the depths (not the distances) in centimeters on the Field Measurement Form.

5. Repeat Step 4 for the remaining cross-sections.

6. Use a stopwatch to determine the time required for the object to travel through the segment. Record the time in the "FLOAT TIME" field of the Field Measurement Form.

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

EQUIPMENT AND SUPPLIES FOR STREAM DISCHARGE

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

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

SECTION 7
PHYSICAL HABITAT CHARACTERIZATION

by
Philip R. Kaufmann[1] and E. George Robison[2],[3]

In the broad sense, physical habitat in streams includes all those physical attributes that influence or provide sustenance to organisms within the stream. Stream physical habitat varies naturally, as do biological characteristics; thus, expectations differ even in the absence of anthropogenic disturbance. Within a given physiographic-climatic region, stream drainage area and overall stream gradient are likely to be strong natural determinants of many aspects of stream habitat, because of their influence on discharge, flood stage, and stream power (the product of discharge times gradient). Summarizing the habitat results of a workshop conducted by EMAP on stream monitoring design, Kaufmann (1993) identified seven general physical habitat attributes important in influencing stream ecology:

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

All of these attributes may be directly or indirectly altered by anthropogenic activities. Nevertheless, their expected values tend to vary systematically with stream size (drainage area) and overall gradient (as measured from topographic maps). The relationships of specific physical habitat measurements described in this section to these seven attributes

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

[2] Department of Forest Engineering, Oregon State University, Corvallis, OR 97331.

[3] Current address: Oregon Department of Forestry, 2600 State St., Salem, OR 97310

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. A comprehensive data analysis guide (Kaufmann et al., in preparation) discusses the detailed procedures used to calculate metrics related to stream reach and riparian habitat quality from filed data collected using the EMAP field protocols. This guide also discusses the precision associated with these measurements and metrics.

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

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

scale habitat types because they have been useful in past studies and enhance comparability with other work.

The time commitment to gain repeatability and precision is greater than that required for more qualitative methods. In our field trials, two people typically complete the specified channel, riparian, and discharge measurements in about three hours of field time (see Section 2, Table 2-1). However, the time required can vary considerably with channel characteristics. On streams up to about 4 meters wide with sparse woody debris, measurements can be completed in less than two hours, whereas crews may require up to five hours in large (>10 m wide), complex streams with abundant woody debris and deep water, if 100 width measurements are required. However, reducing the number of width measurements from 100 to 21 locations on sample reaches limits time to # 4 hours even on large, complex wadeable streams.

The procedures are employed on a sampling reach length 40 times its low flow wetted width, as described in Section 4. Measurement points are systematically placed to statistically represent the entire reach. Stream depth and wetted width are measured at very tightly spaced intervals, whereas channel cross-section profiles, substrate, bank characteristics and riparian vegetation structure are measured at larger spacings. Woody debris is tallied along the full length of the sampling reach, and discharge is measured at one location (see Section 6). The tightly spaced depth and width measures allow calculation of indices of channel structural complexity, objective classification of channel units such as pools, and quantification of residual pool depth, pool volume, and total stream volume.

7.1 COMPONENTS OF THE HABITAT CHARACTERIZATION

There are four different components of the EMAP physical habitat characterization (Table 7-1), including stream discharge, which is described in Section 6. Measurements for the remaining three components are recorded on 11 copies of a two-sided field form, plus an a separate form for recording slope and bearing measurements. The **thalweg profile** is a longitudinal survey of depth, habitat class, and presence of soft/small sediment at 100 equally spaced intervals (150 in streams less than 2.5 m wide) along the centerline between the two ends of the sampling reach. "Thalweg" refers to the flow path of the deepest water in a stream channel. Wetted width is measured at 21 equally spaced intervals. Data for the second component, the **woody debris tally**, are recorded for each of 10 segments of stream located between the 11 transects. The third component, the **channel and riparian characterization**, includes measures and/or visual estimates of channel dimensions,

TABLE 7-1. COMPONENTS OF PHYSICAL HABITAT CHARACTERIZATION

Component	Description
Thalweg Profile: (Section 7.4.1)	• Measure maximum depth, classify habitat and pool-forming features, and determine presence of soft sediment at 10-15 equally spaced intervals between each of 11 channel cross-section transects (100 or 150 individual measurements along entire reach). • Measure wetted width at 11 channel cross-section transects and midway between them (21 measurements).
Woody Debris Tally: (Section 7.4.2)	• Between each of the channel cross sections, tally large woody debris numbers within and above the bankfull channel according to length and diameter classes (10 separate tallies).
Channel and Riparian Characterization: (Section 7.5)	• At 11 cross-section transects placed at equal intervals along reach length: - Measure: channel cross section dimensions, bank height, bank undercut distance, bank angle, slope and compass bearing (backsite), and riparian canopy density (densiometer). - Visually Estimate[a]: substrate size class and embeddedness; areal cover class and type (e.g., woody trees) of riparian vegetation in Canopy, Mid-Layer and Ground Cover; areal cover class of fish concealment features, aquatic macrophytes and filamentous algae. - Observe & Record[a]: human disturbances and their proximity to the channel.
Discharge: (see Section 6)	• In medium and large streams (defined in Section 6) measure water depth and velocity at 0.6 depth at 15 to 20 equally spaced intervals across one carefully chosen channel cross-section. • In very small streams, measure discharge by timing the filling of a bucket or timing the passage of a neutral buoyant object through a segment whose cross-sectional area has been estimated.

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

substrate, fish cover, bank characteristics, riparian vegetation structure, and evidence of human disturbance. These data are obtained at each of the 11 equally-spaced transects established within the sampling reach. In addition, measurements of the stream slope and compass bearing between stations are obtained, providing information necessary for calculating reach gradient, residual pool volume, and channel sinuosity.

7.2 HABITAT SAMPLING LOCATIONS WITHIN THE SAMPLING REACH

Measurements are made at two scales of resolution along the length of the reach; the results are later aggregated and expressed for the entire reach, a third level of resolution. Figure 7-1 illustrates the locations within the sampling reach where data for the different components of the physical habitat characterization are obtained. We assess habitat over stream reach lengths that are approximately 40 times their average wetted width at base-flow, but not less than 150 m long. This allows us to adjust the sample reach length to accommodate varying sizes of streams (see Section 2). Many of the channel and riparian features are characterized on 11 cross-sections and pairs of riparian plots spaced at 4 channel-width intervals. The thalweg profile measurements must be spaced evenly over the entire sampling reach. In addition, they must be sufficiently close together that they do not "miss" deep areas and habitat units that are in a size range of about **a** to ½ of the average channel width. Follow these specifications for choosing the interval between thalweg profile measurements:

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

Following these guidelines, you will be making 150 evenly spaced thalweg profile measurements in the smallest category of streams, 15 between each detailed channel cross section. In all of the larger stream sizes, you will make 100 measurements, 10 between each cross section. For practical reasons, we specify width measurements only at the 11 cross-section transects and at the thalweg measurement points midway between each pair of transects (a total of 21 wetted widths). If more resolution is desired, width measurements may be made at all 100 or 150 thalweg profile locations.

7.3 LOGISTICS AND WORK FLOW

The four components (Table 7-1) of the habitat characterization are organized into three grouped activities:

1. Thalweg Profile and Large Woody Debris Tally (Section 7.4). Two people (the “geomorphs”) proceed upstream from the downstream end of the sampling reach (see Figure 7-1) making observations and measurements at the chosen

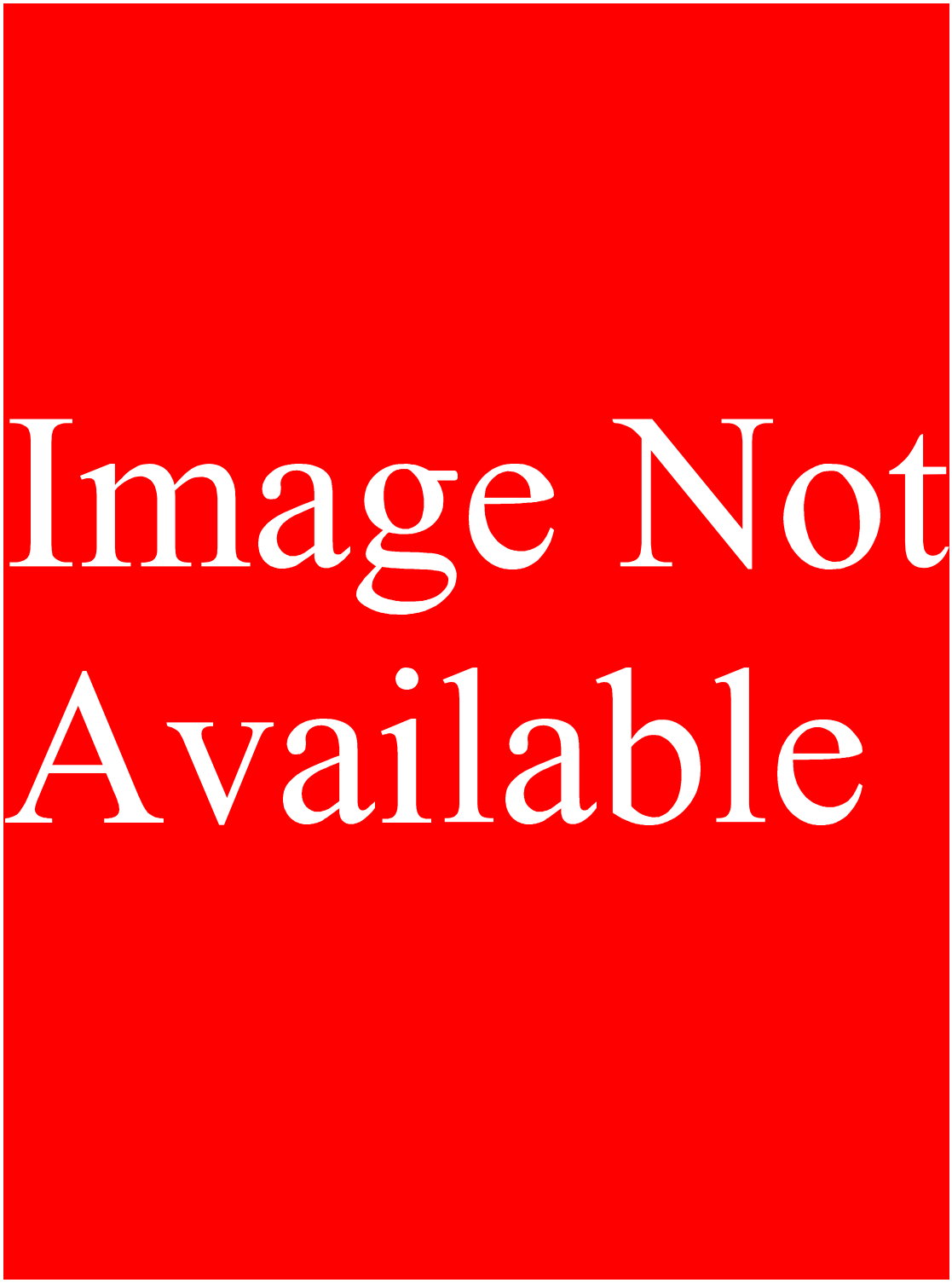


Figure 7-1. Sampling reach layout for physical habitat measurements (plan view).

increment spacing. One person is in the channel making width and depth measurements, and determining whether soft/small sediment is present under his/her staff. The other person records these measurements, classifies the channel habitat, and tallies large woody debris. Each time this team reaches a flag marking a new cross-section transect, they start filling out a new copy of the Thalweg Profile and Woody Debris Form. They interrupt the thalweg profile and woody debris tallying activities to complete data collection at each cross-section transect as it comes.

2. Channel/Riparian Cross-Sections (Section 7.5). One person proceeds with the channel cross-section dimension, substrate, bank, and canopy cover measurements. The second person records those measurements on the Channel/Riparian Cross-section and Thalweg Profile Form while making visual estimates of riparian vegetation structure, instream fish cover, and human disturbance specified on that form. Slope and bearing are determined together by backsiting to the previous transect. Intermediate flagging (of a different color) may have to be used if the stream is extremely brushy, sinuous, or steep to the point that you cannot site for slope and bearing measures between two adjacent transects. (Note that the crews could tally woody debris while doing the backsite, rather than during the thalweg profile measurements.)

3. Discharge (Section 6). Discharge measurements are made after collecting the chemistry sample. They are done at a chosen optimal cross section (but not necessarily at a transect) near the X-site. However, do not use the electromagnetic current meter close to where electrofishing is taking place. Furthermore, if a lot of channel disruption is necessary and sediment must be stirred up, wait on this activity until all chemical and biological sampling has been completed.

7.4 THALWEG PROFILE AND LARGE WOODY DEBRIS MEASUREMENTS

7.4.1 Thalweg Profile

"Thalweg" refers to the flow path of the deepest water in a stream channel. The thalweg profile is a longitudinal survey of maximum depth and several other selected characteristics at 100 or 150 equally spaced points along the centerline of the stream between the two ends of the stream reach. Data from the thalweg profile allows calculation of indices of residual pool volume, stream size, channel complexity, and the relative proportions of habitat types such as riffles and pools. The EMAP-SW habitat assessment modifies traditional methods by proceeding upstream in the middle of the channel, rather than along the thalweg itself (though each thalweg depth measurement is taken at the deepest point at each incremental position). One field person walks upstream (wearing felt-soled waders) carrying a fiberglass telescoping (1.5 to 7.5 m) surveyor's rod and a 1-m metric ruler (or a calibrated rod or pole, such as a ski pole). A second person on the bank or in the stream carries a clipboard with 11 copies of the field data form.

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

It is very important that thalweg depths are obtained from all measurement points. Missing depths at the ends of the sampling reach (e.g., due to the stream flowing into or out of a culvert or under a large pile of debris) can be tolerated, but those occurring in the middle of the sampling reach are more difficult to deal with. Flag these missing measurements using a "K" code and explain the reason for the missing measurements in the

TABLE 7-2. THALWEG PROFILE PROCEDURE

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

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

2. Complete the header information on the thalweg profile and woody debris section of a Channel/Riparian Cross-section and Thalweg Profile Form, noting the transect pair (downstream to upstream). Record the interval distance determined in Step 1 in the "INCREMENT" field on the field data form.

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

3. Begin at the downstream end (station "0") of the first transect 9Transect "A").

4. Measure the wetted width if you are at station "0", station "5" (if the stream width defining the reach length is $ 2.5 m), or station "7" (if the stream width defining the reach length is < 2.5 m). Wetted width is measured across and over mid-channel bars and boulders. Record the width on the field data form to the nearest 0.1 m for widths up to about 3 meters, and to the nearest 5% for widths > 3 m. This is 0.2 m for widths of 4 to 6 m, 0.3 m for widths of 7 to 8 m, and 0.5 m for widths of 9 or 10 m, and so on. For dry and intermittent streams, where no water is in the channel, record zeros for wetted width.

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

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

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

   - NOTE: At stations where the thalweg is too deep to measure directly, stand in shallower water and extend the surveyor's rod or calibrated rod or pole at an angle to reach the thalweg. Determine the rod angle using the external scale of the clinometer. Leave the depth reading for the station blank, and record a "U" flag. Record the water level on the rod and the rod angle in the comments section of the field data form.

(continued)

TABLE 7-2 (Continued)

6. At the point where the thalweg depth is determined, observe whether small, loose, soft sediments are present directly beneath your ruler, rod, or pole. Soft/small sediments are defined here as fine gravel, sand, silt, clay or muck readily apparent by "feeling" the bottom with the staff. Record presence or absence in the "SOFT/SMALL SEDIMENT" field on the field data form.

7. Determine the channel unit code and pool forming element codes for the station. Record these on the field data form using the standard codes provided. For dry and intermittent streams, where no water is in the channel, record habitat type as dry channel (DR).

8. If the station cross-section intersects a mid-channel bar, Indicate the presence of the bar in the "BAR WIDTH" field on the field data form.

9. Record the presence or absence of a side channel at the station's cross-section in the "SIDE CHANNEL" field on the field data form.

10. Proceed upstream to the next station, and repeat Steps 4 through 9.

11. Repeat Steps 4 through 10 until you reach the next transect. Prepare a new Channel/Riparian Cross-section and Thalweg Profile Form, then repeat Steps 2 through 10 for each of the reach segments, until you reach the upstream end of the sampling reach (Transect "K").

Reviewed by (initial): DH

PHab: THALWEG PROFILE & WOODY DEBRIS FORM - STREAMS

SITE NAME: Mill Creek DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999 ___ ___ TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

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

THALWEG PROFILE — Increment (m) → 1.5

Station	Thalweg Depth (cm) (XXX)	Wetted Width (m) (XX.X)	Bar Width [1] X	Bar Width [1] (XX.X)	Soft/Small Sediment (X for Yes)	Channel Unit Code	Pool Form Code	Side Channel (X for Yes)	Flag	Comments
0	14	3.6	X	0.8		RI	N			
1	13		X			RI	N			
2	27		X			RI	N			
3	46		X			PT	F			
4	40		X			PT	F			
5	35	4.4	X	1.0		PT	F			
6	34				X	PT	F			
7	47				X	PT	F			
8	53				X	PT	F			
9	57				X	PT	F	X		Side Channel Convergence
10										
11										
12										
13										
14										

LARGE WOODY DEBRIS (≥ 10 cm small end diameter.; ≥ 1.5 m length) - Tally Each Piece -

Diameter Large End	Pieces All/Part in Bankfull Channel: Length 1.5 - 5 m	5 - 15 m	> 15 m	Pieces Bridge Above Bankfull Channel: Length 1.5 - 5 m	5 - 15 m	> 15 m
0.1 to <0.3 m	////1 → 6	//// → 4	0	0	0	0
0.3 - 0.6 m	// → 2	0	/ → 1	0	0	0
0.6 - 0.8 m	0	0	0	/ → 1	0	0
> 0.8 m	0	0	0	0	0	0

CHANNEL UNIT CODES

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

POOL FORM CODES

N	Not a pool
W	Large Woody Debris
R	Rootwad
B	Boulder or bedrock
F	Unknown, fluvial
O	Other (note in comments)

Flag	Comments
F1	Substrate Class 'OT': Grass

Flag Codes: K = no measurement made; U = suspect measurement; F1, F2, etc. = misc. flags assigned by each field crew. Explain all flags in comments. 1 = Measure Bar Width at Station 0 and Mid-Station (5 or 7), X small column if bar present at the rest of the stations.

Rev. 06/02/97 (st_phct.97) PHab: CHANNEL/RIPARIAN CROSS-SECTION & THALWEG PROFILE FORM - STREAMS - 2

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

comments section of the field data form. At points where a direct depth measurement cannot be obtained, make your best estimate of the depth, record it on the field form, and flag the value using a "U" code (for suspect measurement), explaining that it is an estimated value in the comments section of the field data form. Where the thalweg points are too deep for wading, measure the depth by extending the surveyor's rod at an angle to reach the thalweg point. Record the water level on the rod, and the rod angle, as determined using the external scale on the clinometer (vertical = 90/).

At every thalweg measurement increment, determine by sight or feel whether soft/ small sediment is present on the channel bottom. These particles are defined as substrate equal to or smaller than fine gravel (# 16 mm diameter). These soft/small sediments are **NOT** the same as "Fines" described when determining the substrate particle sizes at the cross-section transects (Section 7.5.2). For the thalweg profile, determine if soft/small sediment deposits are readily obvious by feeling the bottom with your boot, the surveyor's rod, or the calibrated rod or pole.

Wetted width is measured at each transect (station 0), and midway between transects (station 5 for larger streams having 100 measurement points, or station 7 for smaller streams having 150 measurement points). The wetted width boundary is the point at which substrate particles are no longer surrounded by free water.

While recording the width and depth measurements and the presence of soft/small sediments, the second person chooses and records the habitat class and the pool forming element codes (Table 7-3) applicable to each of the 100 (or 150) measurement points along the length of the reach. These channel unit habitat classifications and pool-forming elements are modified from those of Bisson et al. (1982) and Frissell et al. (1986). The resulting database of traditional visual habitat classifications will provide a bridge of common understanding with other studies. With the exception of backwater pools, channel unit scale habitat classifications are to be made at the thalweg of the cross section. The habitat unit itself must meet a minimum size criteria in addition to the qualitative criteria listed in Table 7-3. 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. Bars are defined here as

TABLE 7-3. CHANNEL UNIT AND POOL FORMING ELEMENT CATEGORIES

Channel Unit Habitat Classes[a]

Class (Code)	Description
Pools:	Still water, low velocity, smooth, glassy surface, usually deep compared to other parts of the channel:
Plunge Pool (PP)	Pool at base of plunging cascade or falls.
Trench Pool (PT)	Pool-like trench in the center of the stream
Lateral Scour Pool (PL)	Pool scoured along a bank.
Backwater Pool (PB)	Pool separated from main flow off the side of the channel.
Impoundment Pool (PD)	Pool formed by impoundment above dam or constriction.
Pool (P)	Pool (unspecified type).
Glide (GL)	Water moving slowly, with a smooth, unbroken surface. Low turbulence.
Riffle (RI)	Water moving, with small ripples, waves and eddies -- waves not breaking, surface tension not broken. Sound: "babbling", "gurgling".
Rapid (RA)	Water movement rapid and turbulent, surface with intermittent 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

(continued)

[a] Note that in order for a channel habitat unit (other than a backwater pool) to be distinguished, it must be at least as wide or long as the channel is wide.

TABLE 7-3 (Continued)

Categories of Pool-forming Elements[b]

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

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

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

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

Less than 15%	Indicate the presence of a side channel on the field data form.
16 to 49%	Indicate the presence of a side channel on the field data form. Establish a secondary transect across the side channel and designate it as “X” plus the primary transect letter; e.g., XA). Complete the detailed channel and riparian cross-section measurements for the side channel, using a separate copy of the field data form.

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

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

7.4.2 Large Woody Debris Tally

Methods for large woody debris (LWD) measurement are a simplified adaptation of those described by Robison and Beschta (1990). This component of the EMAP physical habitat characterization allows quantitative estimates of the number, size, total volume and distribution of wood within the stream reach. LWD is defined here as woody material with a small end diameter of at least 10 cm (4 in.) and a length of at least 1.5 m (5 ft.).

The procedure for tallying LWD is presented in Table 7-4. The tally includes all pieces of LWD that are at least partially in the baseflow channel, the "active channel" (flood channel up to bankfull stage), or spanning above the active channel. The active (or “bank-full") channel is defined as the channel that is filled by moderate sized flood events that typically recur every one to two years. LWD in the active channel is tallied over the entire length of the reach, including the area between the channel cross-section transects. As in the thalweg profile, LWD measurements in the channel segment between each cross section transect and the next one upstream are recorded on the first 10 thalweg profile and woody debris forms (Figure 7-2). The location of the large end of each piece of LWD determines the segment to which it is assigned.

First, tally all the pieces of LWD that are at least partially in the bankfull channel (Figure 7-3, Zones 1 or 2). Then tally all the pieces of LWD that are not actually within the bankfull channel, but are at least partially spanning (bridging) the bankfull channel (Figure 7-3, Zone 3). For both the Zone 1-2 wood and the Zone 3 LWD, the field form (Figure 7-2) provides 12 entry boxes for tallying debris pieces visually estimated within three length and four diameter class combinations. Each LWD piece is tallied in only one box. Pieces of LWD that are not at least partially within Zones 1, 2, or 3 are not tallied.

For each LWD piece, first visually estimate its length and its large and small end diameters in order to place it in one of the diameter and length categories. The diameter class on the field form (Figure 7-2) refers to the large end diameter. Sometimes LWD is not cylindrical, so it has no clear "diameter". In these cases visually estimate what the diameter would be for a piece of wood with a circular cross section that would have the same volume. When evaluating length, include only the part of the LWD piece that has a diameter greater than 10 cm (4 in). Count each of the LWD pieces as one tally entry and include the whole piece when assessing dimensions, even if part of it is in Zone 4 (outside of the bankfull channel). For both the Zone 1-2 wood and the Zone 3 LWD, the field form (Figure 7-2) provides 12 entry boxes for tallying debris pieces visually estimated within three length and

TABLE 7-4. PROCEDURE FOR TALLYING LARGE WOODY DEBRIS

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

1. Scan the stream segment between the two cross-section transects where thalweg profile measurements are being made.

2. Tally all LWD pieces within the segment that are at least partially within the bankfull channel. Determine if a piece is LWD (**small end diameter $10 cm [4 in.]; length $1.5 m [5 ft.]**)

3. For each piece of LWD, determine the class **based on the diameter of the large end** (0.1 m to < 0.3 m, 0.3 m to <0.6 m, 0.6 m to <0.8 m, or >0.8 m, and the class based on the length of the piece (1.5m to <5.0m, 5m to <15m, or >15m).

    - If the piece is not cylindrical, visually estimate what the diameter would be for a piece of wood with circular cross section that would have the same volume.
    - When estimating length, include only the part of the LWD piece that has a diameter greater than 10 cm (4 in)

4. Place a tally mark in the appropriate diameter × length class tally box in the "PIECES ALL/PART IN BANKFULL CHANNEL" section of the Thalweg Profile and Woody Debris Form.

5. Tally all LWD pieces within the segment that are not actually within the bankfull channel, but are at least partially spanning (bridging) the bankfull channel. For each piece, determine the class based on the diameter of the **large end** (0.1 m to < 0.3 m, 0.3 m to <0.6 m, 0.6 m to <0.8 m, or >0.8 m), and the class based on the length of the piece (1.5 m to <5.0 m, 5 m to <15 m, or >15 m).

6. Place a tally mark for each piece in the appropriate diameter × length class tally box in the "PIECES BRIDGE ABOVE BANKFULL CHANNEL" section of the Thalweg Profile and Woody Debris Form.

7. After all pieces within the segment have been tallied, write the total number of pieces for each diameter × length class in the small box at the lower right-hand corner of each tally box.

8. Repeat Steps 1 through 7 for the next stream segment, using a new Thalweg Profile and Woody Debris Form.

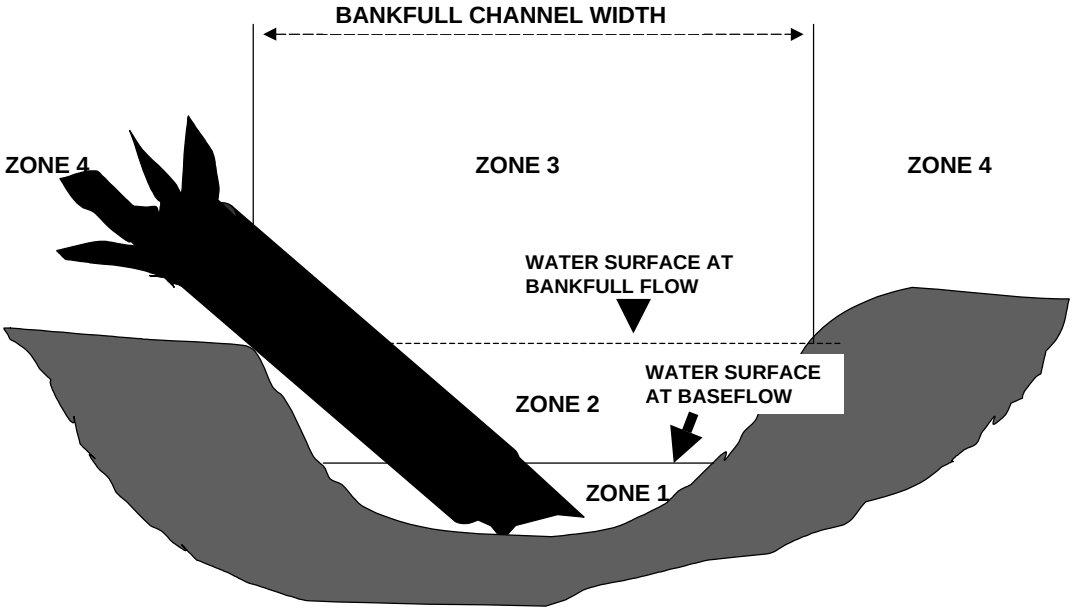


Figure 7-3. Large woody debris influence zones (modified from Robison and Beschta, 1990)

four diameter class combinations. Each LWD piece is tallied in only one box. There are 12 size classes for wood at least partially in Zones 1 and 2, and 12 for wood partially within Zone 3. Wood that is not at least partially within those zones is not tallied.

7.5 CHANNEL AND RIPARIAN MEASUREMENTS AT CROSS-SECTION TRANSECTS

7.5.1 Slope and Bearing

The slope, or gradient, of the stream reach is useful in three different ways. First, the overall stream gradient is one of the major stream classification variables, giving an indication of potential water velocities and stream power, which are in turn important con-

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

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

Slope can also be measured by two people, each having a pole that is marked at the same height. Alternatively, the second person can be “flagged” at the eye level of the person doing the backsiting. Be sure that you mark your eye level on the other person or on a separate pole beforehand while standing on level ground. Site to **your eye level** when backsiting on your co-worker. If two marked poles are used, site from the mark on one pole to the mark on the other. Also, be sure that the second person is standing (or holding the marked pole) at the water’s edge or in the same depth of water as you are. The intent is to get a measure of the **water surface slope**, which may not necessarily be the same as the bottom slope. The clinometer reads both percent slope and degrees of the slope angle; be careful to read and record percent slope. Percent slope is the scale on the right-hand side as you look through most clinometers. If using an Abney Level, insure that you are reading the scale marked “PERCENT.” With the clinometer or the Abney level, verify this by comparing the two scales. Percent slope is always a higher number than degrees of slope angle (e.g., 100% slope=45/ angle). For slopes > 2%, read the clinometer to the nearest 0.5%. For slopes < 2%, read to the nearest 0.25%. If the clinometer reading is 0%, but water is moving, record the slope as 0.1%. If the clinometer reading is 0% and water is not moving, record the slope as 0%.

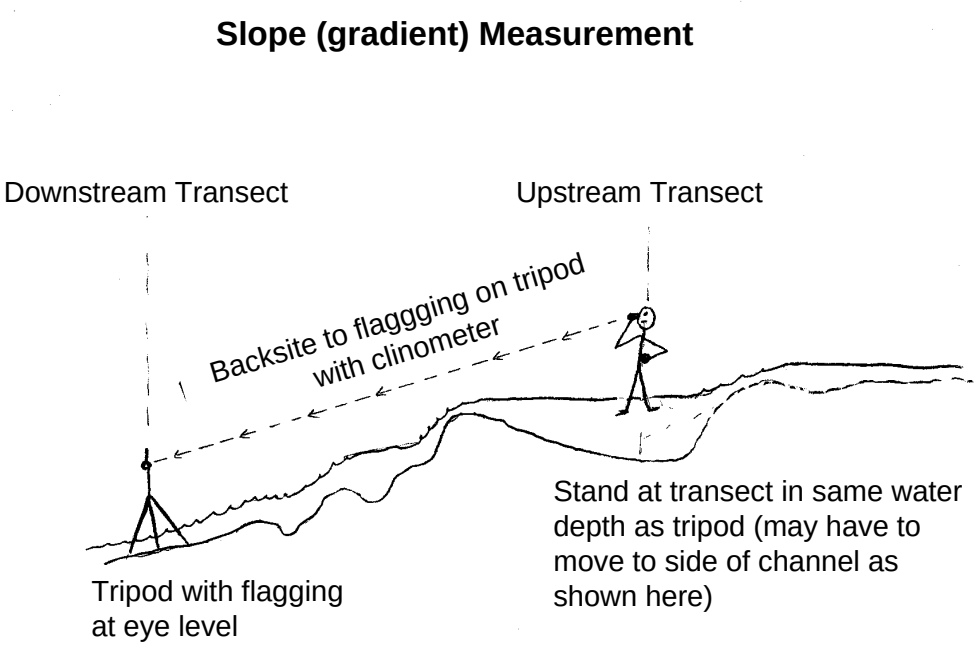


Figure 7-4. Channel slope and bearing measurements.

TABLE 7-5. PROCEDURE FOR OBTAINING SLOPE AND BEARING DATA

1. Stand in the center of the channel at the downstream cross-section transect. Determine if you can see the center of the channel at the next cross-section transect upstream. If not, you will have to take supplementary slope and bearing measurements.

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

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

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

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

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

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

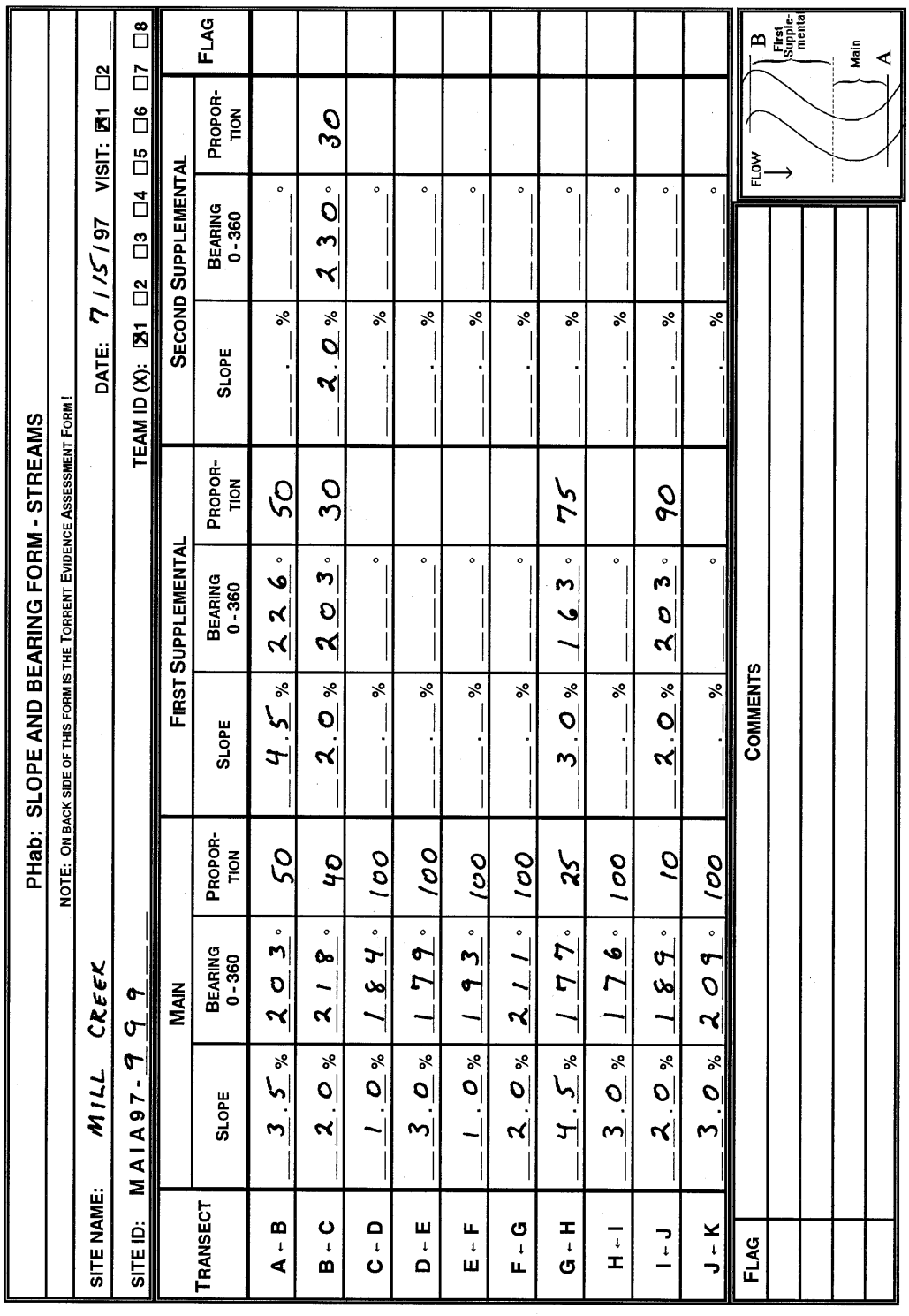
    - If you are backsiting from a supplemental point, record the slope (%) and proportion (%) of the stream segment that is included in the measurement in the appropriate "SUPPLEMENTAL" section of the Slope and Bearing Form.

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

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

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

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



PHab: SLOPE AND BEARING FORM - STREAMS

NOTE: ON BACK SIDE OF THIS FORM IS THE TORRENT EVIDENCE ASSESSMENT FORM !

SITE NAME: MILL CREEK DATE: 7/15/97 VISIT: ☒1 ☐2

SITE ID: MAIA97-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

Reviewed by (initial): JD

TRANSECT	MAIN			FIRST SUPPLEMENTAL			SECOND SUPPLEMENTAL			FLAG
	SLOPE	BEARING 0-360	PROPORTION	SLOPE	BEARING 0-360	PROPORTION	SLOPE	BEARING 0-360	PROPORTION	
A ← B	3.5 %	203°	50	4.5 %	226°	50	___.__ %	___°		
B ← C	2.0 %	218°	40	2.0 %	203°	30	2.0 %	230°	30	
C ← D	1.0 %	184°	100	___.__ %	___°		___.__ %	___°		
D ← E	3.0 %	179°	100	___.__ %	___°		___.__ %	___°		
E ← F	1.0 %	193°	100	___.__ %	___°		___.__ %	___°		
F ← G	2.0 %	211°	100	___.__ %	___°		___.__ %	___°		
G ← H	4.5 %	177°	25	3.0 %	163°	75	___.__ %	___°		
H ← I	3.0 %	176°	100	___.__ %	___°		___.__ %	___°		
I ← J	2.0 %	189°	10	2.0 %	203°	90	___.__ %	___°		
J ← K	3.0 %	209°	100	___.__ %	___°		___.__ %	___°		

FLAG	COMMENTS

Rev. 06/02/97 (st_phsl.97)

PHab: SLOPE & BEARING FORM - STREAMS - 1

Figure 7-5. Slope and Bearing Form.

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

As stated earlier, it may be necessary to set up intermediate ("supplementary") slope and bearing points between a pair of cross-section transects if you do not have direct line-of-site along (and within) the channel between stations (see Figure 7-4). This can happen if brush is too heavy, or if there are sharp slope breaks or tight meander bends. Mark these intermediate station locations with a different color of plastic flagging than used for the cross-section transects to avoid confusion. Record these supplemental slope and bearing measurements, along with the proportion of the stream segment between transects included in each supplemental measurement, in the appropriate sections of the Slope and Bearing Form (Figure 7-5). Note that the main slope and bearing observations are always downstream of supplemental observations. Similarly, first supplemental observations are always downstream of second supplemental observations.

7.5.2 Substrate Size and Channel Dimensions

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

In the EMAP protocol, substrate size and embeddedness are evaluated at each of the 11 cross-section transects (refer to Figure 7-1) using a combination of methods adapted from those described by Wolman (1954), Bain et al. (1985), Platts et al. (1983), and Plafkin et al. (1989). The basis of the protocol is a systematic selection of 5 substrate particles

from each of 11 cross-section transects (Figure 7-6). In the process of measuring substrate particle sizes at each channel cross section, you also measure the wetted width of the channel and the water depth at each substrate sample point. If the wetted channel is split by a mid-channel bar (see Section 7.4.1), the five substrate points are centered between the wetted width boundaries regardless of the mid-channel bar in between. Consequently, substrate particles selected in some cross-sections may be "high and dry". For dry channels, make cross-section measurements across the unvegetated portion of the channel.

The distance you record to the right bank is the same as the wetted channel width. (NOTE: this is the same value that is also recorded under "BANK MEASUREMENTS" on the cross-section and thalweg profile data form [Section 7.5.3]). The substrate sampling points along the cross-section are located at 0, 25, 50, 75, and 100 percent of the measured wetted width, with the first and last points located at the water's edge just within the left and right banks.

The procedure for obtaining substrate measurements is described in Table 7-6. Record these measurements on the Channel/Riparian Cross-section and Thalweg Profile Form as shown in Figure 7-7. To minimize bias in selecting a substrate particle for size classification, it is important to concentrate on correct placement of the measuring stick along the cross-section, and to select the particle right at the bottom of the stick (not, for example, a more noticeable large particle that is just to the side of the stick). Classify the particle into one of the size classes listed on the field data form (Figure 7-7) based on the middle dimension of its length, width, and depth. This “median” dimension determines the sieve size through which the particle can pass. Always distinguish “hardpan” from “fines”, coding hardpan as “HP”. Similarly, always distinguish concrete or asphalt from bedrock; denote these artificial substrates as “other” (“OT”) and describe them in the comments section of the field data form. Code and describe other artificial substrates (including metal, tires, car bodies, etc.) in the same manner. When you record the size class as “OT” (other), assign an “F"-series flag on the field data form (Figure 7-7) and describe the substrate type in the comments section of the field form, as shown in Figure 7-2.

Examine particles larger than sand for surface stains, markings, and algal coatings to estimate embeddedness of all particles in the 10 cm diameter circle around the substrate sampling point. Embeddedness is the fraction of a particle’s surface that is surrounded by (embedded in) sand or finer sediments on the stream bottom. By definition, the embeddedness of sand, silt, clay, and muck is 100 percent, and the embeddedness of hardpan and bedrock is 0 percent.

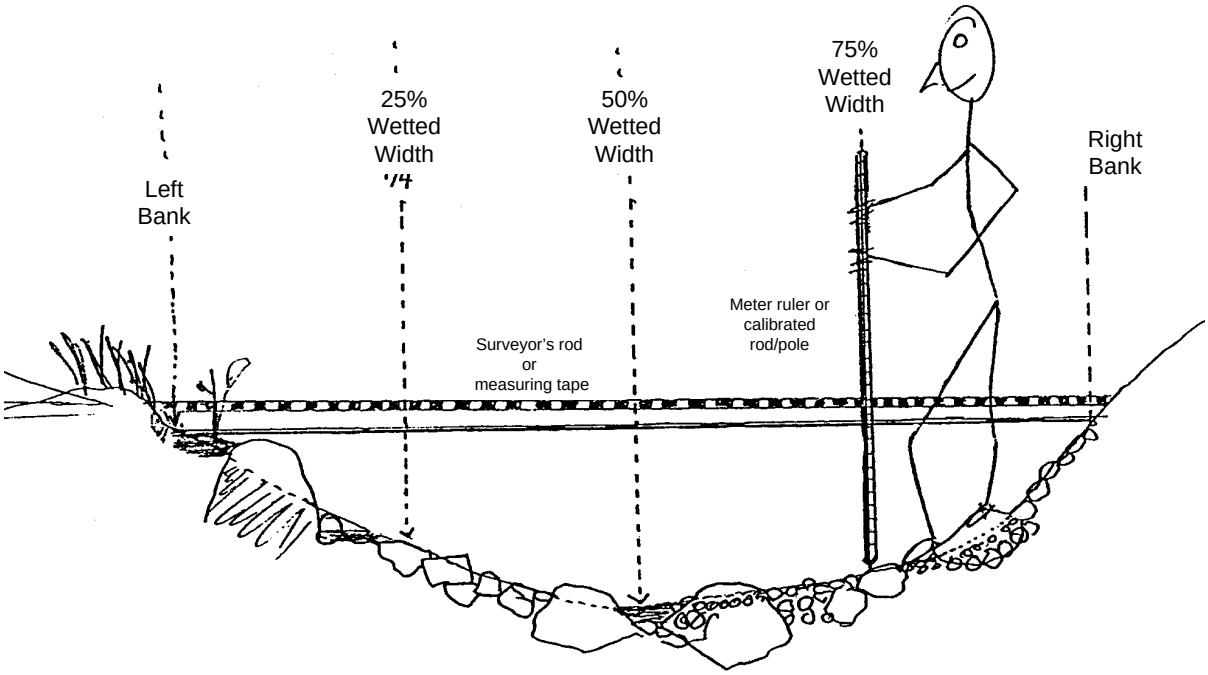


Figure 7-6. Substrate sampling cross-section.

7.5.3 Bank Characteristics

The procedure for obtaining bank and channel dimension measurements is presented in Table 7-7. Data are recorded in the "Bank Measurements" section of the Channel/Riparian Cross-section and Thalweg Profile Form as shown in Figure 7-7. Bank angle and bank undercut distance are determined on the left and right banks at each cross section transect. Other features include the wetted width of the channel (as determined in Section 7.5.2), the width of exposed mid-channel bars of gravel or sand, estimated incision height, and the estimated height and width of the channel at bankfull stage as described in Section 7.4.2 and Figure 7-3. The "bankfull" or "active" channel is defined as the channel that is filled by moderate-sized flood events that typically occur every one or two years. Such flows do not generally overtop the channel banks to inundate the valley floodplain, and are believed to control channel dimensions in most streams.

TABLE 7-6. SUBSTRATE MEASUREMENT PROCEDURE

1. Fill in the header information on page 1 of a Channel/Riparian Cross-section and Thalweg Profile Form. Indicate the cross-section transect. At the transect, extend the surveyor's rod across the channel perpendicular to the flow, with the "zero" end at the left bank (facing downstream). If the channel is too wide for the rod, stretch the metric tape in the same manner.

2. Divide the wetted width of the channel by 4 to obtain the locations of the substrate measurement points along the cross-section. In the "DISTLB" fields of the form, record the distances corresponding to 0% (LFT), 25% (LCTR), 50% (CTR), 75% (RCTR), and 100% (RGT) of the measured wetted width.

3. Place your sharp-ended meter stick or calibrated pole at the "LFT" location (0 m). Measure the depth and record it on the field data form.

   - Entries for the water's edge at the left and right banks may be 0 (zero) if the banks are gradual.
   - If the bank is nearly vertical, let the base of the measuring stick fall to the bottom, rather than holding it suspended at the water surface.

4. Pick up the substrate particle that is at the base of the meter stick (unless it is bedrock or boulder), and visually estimate its particle size, according to the following table. Classify the particle according to its "median" diameter (the middle dimension of its length, width, and depth). Record the size class code on the field data form.

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

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

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

7. Repeat Steps 1 through 6 at each new cross section transect.

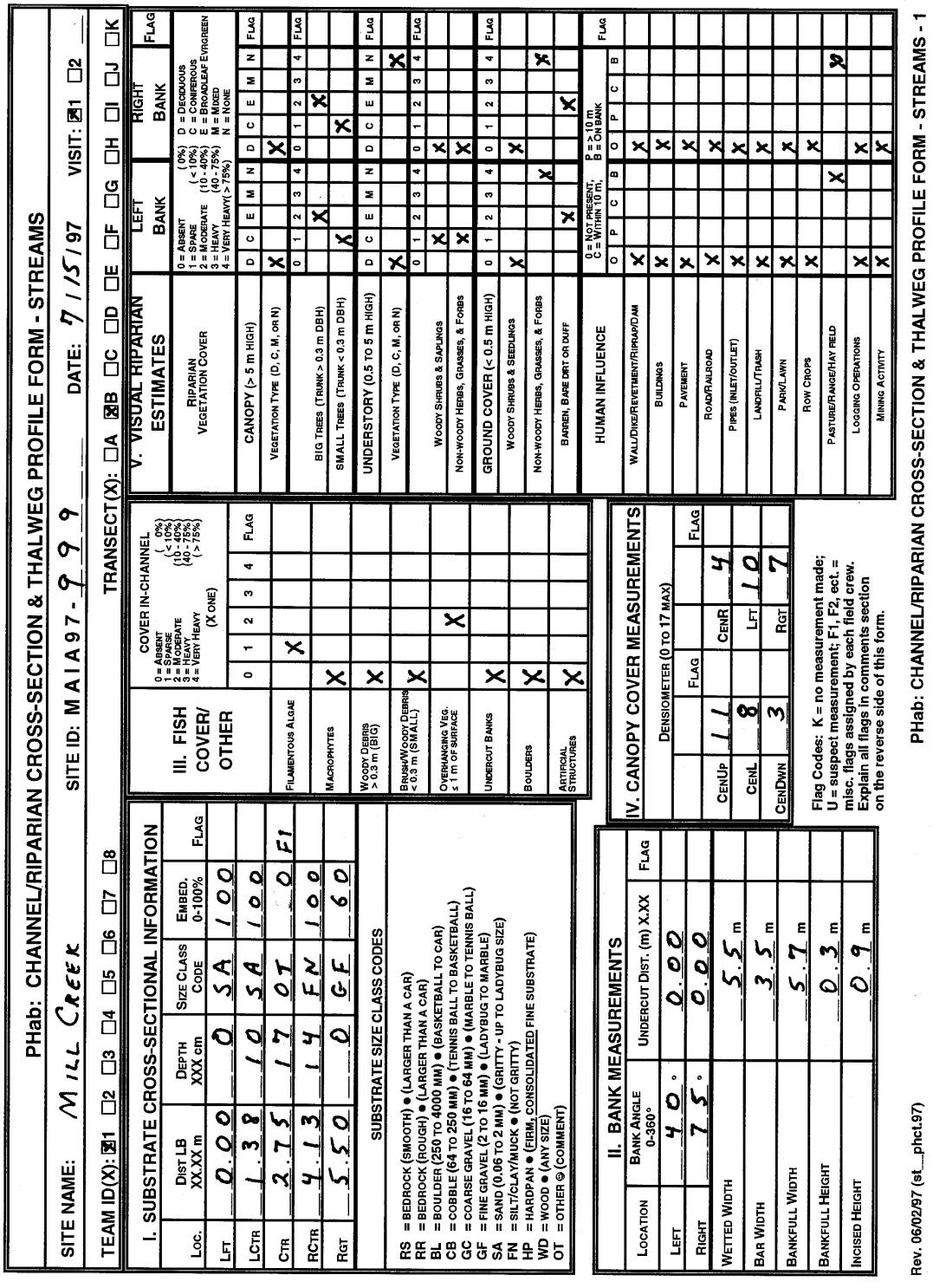


Figure 7-7. Channel/Riparian Cross-section and Thalweg Profile Form.

TABLE 7-7. PROCEDURE FOR MEASURING BANK CHARACTERISTICS

1. To measure bank angle, lay the surveyor's rod or your meter ruler down against the left bank (determined as you face downstream), with one end at the water's edge. Lay the clinometer on the rod, read the bank angle in degrees from the external scale on the clinometer. Record the angle in the field for the left bank in the "BANK MEASUREMENT" section of the Channel/Riparian Cross-section and Thalweg Profile Form.

    - A vertical bank is 90 degrees; undercut banks have angles >90 degrees approaching 180 degrees, and more gradually sloped banks have angles <90 degrees. To measure bank angles >90 degrees, turn the clinometer (which only reads 0 to 90 degrees) over and subtract the angle reading from 180 degrees.

2. If the bank is undercut, measure the horizontal distance of the undercutting to the nearest 0.01 m. Record the distance on the field data form. The undercut distance is the distance from the water's edge out to the point where a vertical plumb line from the bank would hit the water's surface.

    - Measure submerged undercuts by thrusting the rod into the undercut and reading the length of the rod that is hidden by the undercutting.

3. Repeat Steps 1 and 2 on the right bank.

4. Hold the surveyor's rod vertical, with its base planted at the water's edge. Using the surveyor's rod as a guide while examining both banks, estimate (by eye) the channel incision as the height up from the water surface to elevation of the first terrace of the valley floodplain (Note this is at or above the bankfull channel height). Record this value in the "INCISED HEIGHT" field of the bank measurement section on the field data form.

5. Still holding the surveyor's rod as a guide, examine both banks to estimate and record the height of bankfull flow above the present water level. Look for evidence on one or both banks such as:

    - An obvious slope break that differentiates the channel from a relatively flat floodplain terrace higher than the channel.
    - A transition from exposed stream sediments to terrestrial vegetation.
    - Moss growth on rocks along the banks.
    - Presence of drift material caught on overhanging vegetation.
    - transition from flood- and scour-tolerant vegetation to that which is relatively intolerant of these conditions.

6. Record the wetted width value determined when locating substrate sampling points in the "WETTED WIDTH" field in the bank measurement section of the field data form. Also determine the bankfull channel width and the width of exposed mid-channel bars (if present). Record these values in the "BANK MEASUREMENT" section of the field data form.

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

If the channel is not greatly incised, bankfull channel height and incision height will be the same. However, if the channel is incised greatly, the bankfull level will be below the level of the first terrace of the valley floodplain, making bankfull channel height smaller than incision height (Figure 7-8). You may need to look for evidence of recent flows (within about one year) to distinguish bankfull and incision heights. In cases where the channel is cutting a valley sideslope and has oversteepened and destabilized that slope, the bare "cutbank" is not necessarily an indication of recent incision. Examine both banks to more accurately determine channel downcutting.

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

7.5.4 Canopy Cover Measurements

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

Canopy cover over the stream is determined at each of the 11 cross-section transects. A Convex Spherical Densiometer (model B) is used (Lemmon, 1957). The densiometer must be taped exactly as shown in Figure 7-9 to limit the number of square grid intersections to 17. Densiometer readings can range from 0 (no canopy cover) to 17 (maximum canopy cover). Six measurements are obtained at each cross-section transect (four measurements in four directions at mid-channel and one at each bank). The mid-channel measurements are used to estimate canopy cover over the channel. The two bank measurements complement your visual estimates of vegetation structure and cover within the

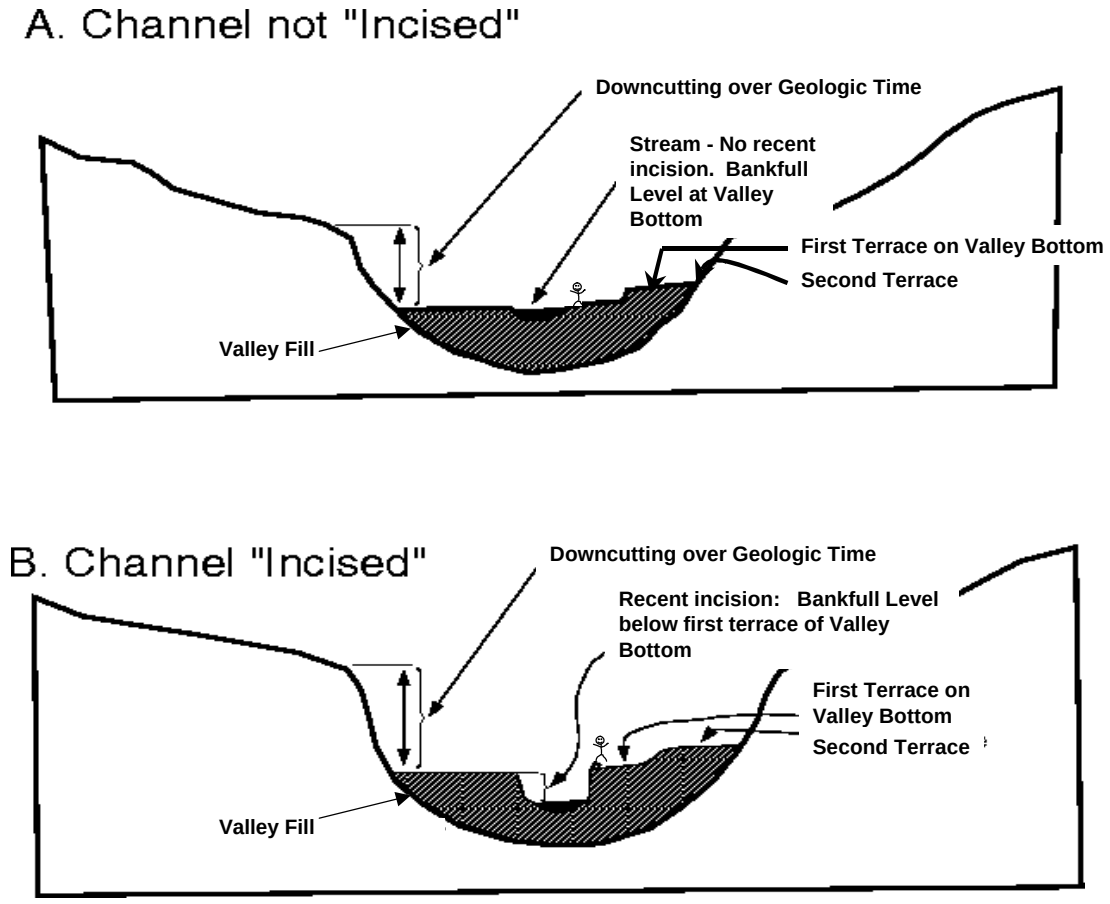


Figure 7-8. Schematic showing bankfull channel and incision for channels. (A) not recently incised, and (B) recently incised into valley bottom. Note level of bankfull stage relative to elevation of first terrace on valley bottom (Stick figure included for scale).

riparian zone itself (Section 7.5.5), and are particularly important in wide streams, where riparian canopy may not be detected by the densiometer when standing midstream.

The procedure for obtaining canopy cover data is presented in Table 7-8. Densiometer measurements are taken at 0.3 m (1 ft) above the water surface, rather than at waist level, to (1) avoid errors because people differ in height; (2) avoid errors from standing in water of varying depths; and (3) include low overhanging vegetation more consistently in the estimates of cover. Hold the densiometer level (using the bubble level) 0.3 m above the

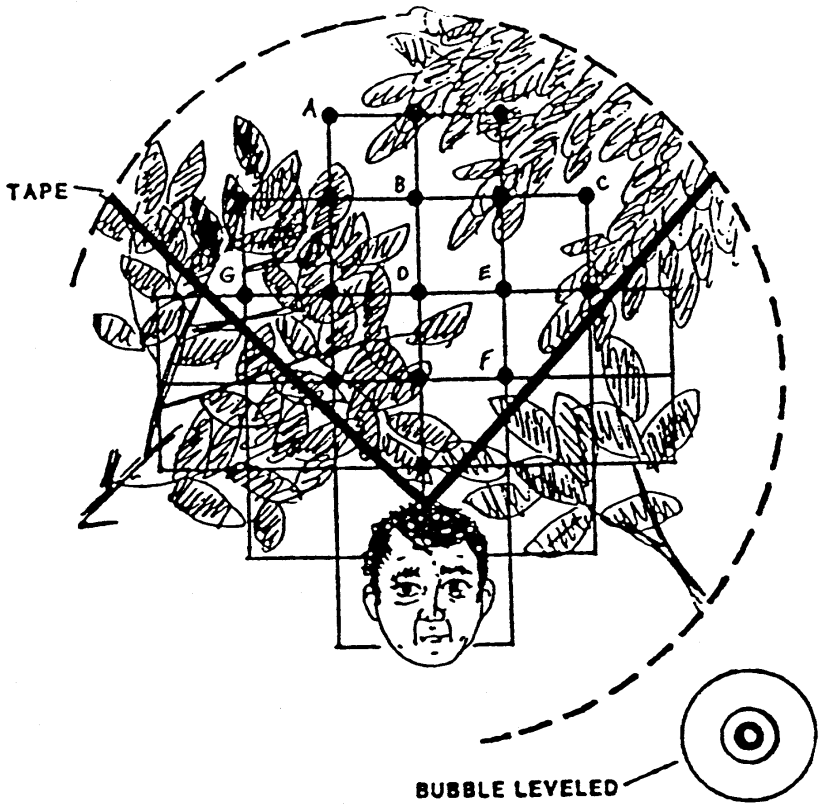


Figure 7-9. Schematic of modified convex spherical canopy densiometer (From Mulvey et al., 1992). In this example, 10 of the 17 intersections show canopy cover, giving a densiometer reading of 10. Note proper positioning with the bubble leveled and face reflected at the apex of the "V."

water surface with your face reflected just below the apex of the taped "V", as shown in Figure 7-9. Concentrate on the 17 points of grid intersection on the densiometer that lie within the taped "V". If the reflection of a tree or high branch or leaf overlies any of the intersection points, that particular intersection is counted as having cover. For each of the

TABLE 7-8. PROCEDURE FOR CANOPY COVER MEASUREMENTS

1. At each cross-section transect, stand in the stream at mid-channel and face upstream.

2. Hold the densiometer 0.3 m (1 ft) above the surface of the stream. Hold the densiometer level using the bubble level. Move the densiometer in front of you so your face is just below the apex of the taped "V".

3. Count the number of grid intersection points within the "V" that are covered by either a tree, a leaf, or a high branch. Record the value (0 to 17) in the "CENUP" field of the canopy cover measurement section of the Channel/Riparian Cross-section and Thalweg Profile Form.

4. Face toward the left bank (left as you face downstream). Repeat Steps 2 and 3, recording the value in the "CENL" field of the field data form.

5. Repeat Steps 2 and 3 facing downstream, and again while facing the right bank (right as you look downstream). Record the values in the "CENDWN" and "CENR" fields of the field data form.

6. Repeat Steps 2 and 3 again, this time facing the bank while standing first at the left bank, then the right bank. Record the values in the "LFT" and "RGT" fields of the field data form.

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

six measurement points, record the number of intersection points (maximum=17) that have vegetation covering them in the “Canopy Cover Measurement” section of the Channel/ Riparian Cross-section and Thalweg Profile Form as shown in (Figure 7-7).

7.5.5 Riparian Vegetation Structure

The previous section (7.5.4) described methods for quantifying the cover of canopy over the stream channel. The following visual estimation procedures supplement those measurements with a semi-quantitative evaluation of the type and amount of various types of riparian vegetation. These data are used to evaluate the health and level of disturbance of the stream corridor. They also provide an indication of the present and future potential for various types of organic inputs and shading.

Observations to assess riparian vegetation apply to the riparian area upstream 5 meters and downstream 5 meters from each of the 11 cross-section transects (refer to Figure 7-1). They include the visible area from the stream back a distance of 10m (~30 ft) shoreward from both the left and right banks, creating a 10 m × 10 m riparian plot on each side of the stream (Figure 7-10). The riparian plot dimensions are estimated, not measured. On steeply sloping channel margins, the 10 m × 10 m plot boundaries are defined as if they were projected down from an aerial view. If the wetted channel is split by a mid-channel bar, the bank and riparian measurements are made at each side of the channel, not the bar.

Table 7-9 presents the procedure for characterizing riparian vegetation structure and composition. Figure 7-7 illustrates how measurement data are recorded in the “VISUAL RIPARIAN ESTIMATES” section of the Channel/Riparian Cross-section and Thalweg Profile Form. Conceptually divide the riparian vegetation into three layers: a CANOPY LAYER (> 5 m high), an UNDERSTORY (0.5 to 5 m high), and a GROUND COVER layer (< 0.5 m high). Note that several vegetation types (e.g., grasses or woody shrubs) can potentially occur in more than one layer. Similarly note that some things other than vegetation are possible entries for the "Ground Cover" layer (e.g., barren ground).

Before estimating the areal coverage of the vegetation layers, record the type of vegetation (Deciduous, Coniferous, broadleaf Evergreen, Mixed, or None) in each of the two taller layers (Canopy and Understory). Consider the layer "Mixed" if more than 10% of the areal coverage is made up of the alternate vegetation type.

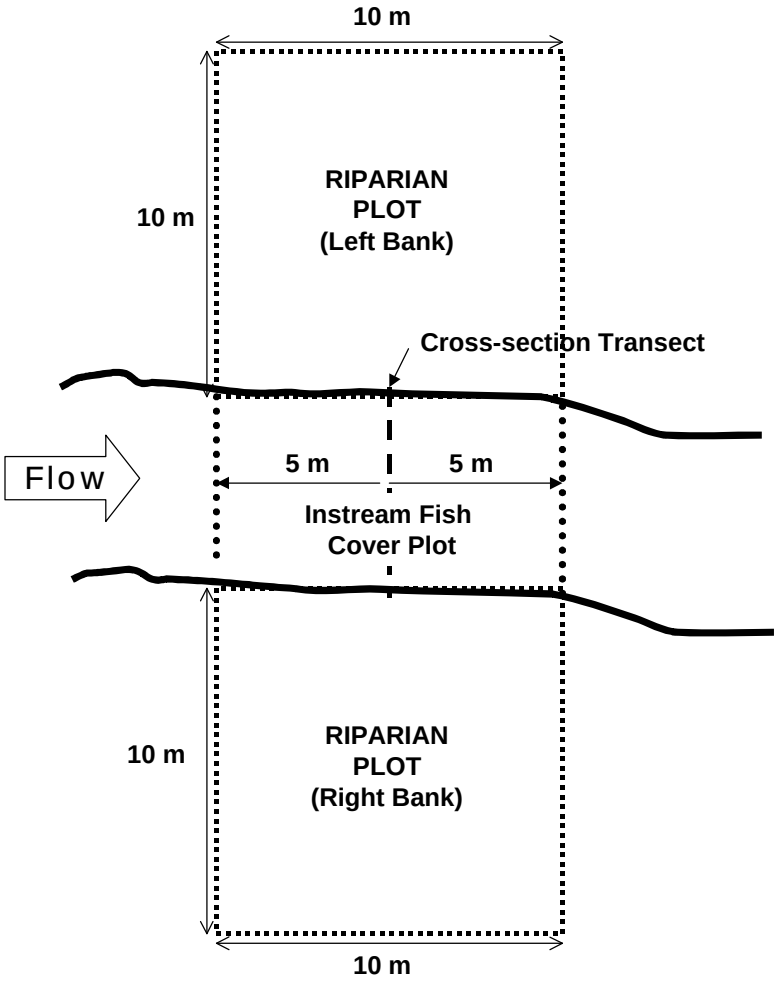


Figure 7-10. Boundaries for visual estimation of riparian vegetation, fish cover, and human influences.

Estimate the areal cover separately in each of the three vegetation layers. Note that the areal cover can be thought of as the amount of shadow cast by a particular layer alone when the sun is directly overhead. The maximum cover in each layer is 100%, so the sum of the areal covers for the combined three layers could add up to 300%. The four areal cover classes are “absent”, “sparse” (<10%), “moderate” (10 to 40%), “heavy” (40 to 75%),

TABLE 7-9. PROCEDURE FOR CHARACTERIZING RIPARIAN VEGETATION STRUCTURE

1. Standing in mid-channel at a cross-section transect, estimate a 5 m distance upstream and downstream (10 m total length).

2. Facing the left bank (left as you face downstream), estimate a distance of 10 m back into the riparian vegetation.

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

3. Within this 10 m × 10 m area, conceptually divide the riparian vegetation into three layers: a CANOPY LAYER (>5m high), an UNDERSTORY (0.5 to 5 m high), and a GROUND COVER layer (<0.5 m high).

4. Within this 10 m × 10 m area, determine the dominant vegetation type for the CANOPY LAYER (vegetation > 5 m high) as either Deciduous, Coniferous, broadleaf Evergreen, Mixed, or None. Consider the layer "Mixed" if more than 10% of the areal coverage is made up of the alternate vegetation type. Indicate the appropriate vegetation type in the "VISUAL RIPARIAN ESTIMATES" section of the Channel/Riparian Cross-section and Thalweg Profile Form.

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

6. Look at the UNDERSTORY layer (vegetation between 0.5 and 5 m high). Determine the dominant vegetation type for the understory layer as described in Step 4 for the canopy layer.

7. Determine the areal cover class for woody shrubs and saplings separately from non-woody vegetation within the understory, as described in Step 5 for the canopy layer.

8. Look at the GROUND COVER layer (vegetation < 0.5 m high). Determine the areal cover class for woody shrubs and seedlings, non-woody vegetation, and the amount of bare ground present as described in Step 5 for large canopy trees.

9. Repeat Steps 1 through 8 for the right bank.

10. Repeat Steps 1 through 9 for all cross-section transects, using a separate field data form for each transect.

and "very heavy" (>75%). These cover classes and their corresponding codes are shown on the field data form (Figure 7-6). 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 rated as either moderate ("2"), sparse ("1") or absent ("0"). Two heavy classes with 40-75% cover can both be rated "3".

7.5.6 Instream Fish Cover, Algae, and Aquatic Macrophytes

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

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

The entry “Filamentous algae” refers to long streaming algae that often occur in slow moving waters. “Aquatic macrophytes” are water-loving plants, including mosses, in the stream that could provide cover for fish or macroinvertebrates. If the stream channel contains live wetland grasses, include these as macrophytes. “Woody debris” are the larger pieces of wood that can influence cover and stream morphology (i.e., those pieces that would be included in the large woody debris tally [Section 7.4]). “Brush/woody debris” refers to smaller wood pieces that primarily affect cover but not morphology. “Overhanging vegetation” includes tree branches, brush, twigs, or other small debris that is not in the water but is close to the stream (within 1 m of the surface) and provides potential cover. “Boulders” are typically basketball- to car-sized particles. “Artificial structures” include those designed for fish habitat enhancement, as well as in-channel structures discarded (e.g., cars or tires) or purposefully placed for diversion, impoundment, channel stabilization, or other purposes.

TABLE 7-10. PROCEDURE FOR ESTIMATING INSTREAM FISH COVER

1. Standing mid-channel at a cross-section transect, estimate a 5m distance upstream and downstream (10 m total length).

2. Examine the water and the banks within the 10-m segment of stream for the following features and types of fish cover: filamentous algae, aquatic macrophytes, large woody debris, brush and small woody debris, overhanging vegetation, undercut banks, boulders, and artificial structures.

3. For each cover type, estimate the areal cover. Record the appropriate cover class in the "FISH COVER/OTHER" section of the Channel/Riparian Cross-section and Thalweg Profile Form ("0"=absent: zero cover, "1"=sparse: <10%, "2"=moderate: 10-40%, "3"=heavy: 40-75%, or "4"=very heavy: >75%).

4. Repeat Steps 1 through 3 at each cross-section transect, recording data from each transect on a separate field data form.

7.5.7 Human Influence

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

For the left and right banks at each of the 11 detailed Channel and Riparian Cross-Sections, evaluate the presence/absence and the proximity of 11 categories of human influences with the procedure outlined in Table 7-11. Relate your observations and proximity evaluations to the stream and riparian area within 5 m upstream and 5 m downstream from the station (Figure 7-10). Four proximity classes are used: In the stream or on the bank within 5 m upstream or downstream of the cross-section transect, present within the 10 m × 10 m riparian plot but not in the stream or on the bank, present outside of the riparian plot, and absent. Record data on the Channel/Riparian Cross-section and Thalweg Profile Form as shown in Figure 7-6. If a disturbance is within more than one proximity class, record the one that is closest to the stream (e.g., “C” takes precedence over “P”).

A particular influence may be observed outside of more than one riparian observation plot (e.g., at both transects “D” and “E”). Record it as present at every transect where you can see it without having to site through another transect or its 10 m × 10 m riparian plot.

7.6 EQUIPMENT AND SUPPLIES

Figure 7-11 lists the equipment and supplies required to conduct all the activities described for characterizing physical habitat. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the stream. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

TABLE 7-11. PROCEDURE FOR ESTIMATING HUMAN INFLUENCE

1. Standing mid-channel at a cross-section transect, look toward the left bank (left when facing downstream), and estimate a 5m distance upstream and downstream (10 m total length). Also, estimate a distance of 10 m back into the riparian zone to define a riparian plot area.

2. Examine the channel, bank and riparian plot area adjacent to the defined stream segment for the following human influences: (1) walls, dikes, revetments, riprap, and dams; (2) buildings; (3) pavement (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 stream and riparian plot area. Consider human disturbance items as present if you can see them from the cross-section transect. Do not include them if you have to site through another transect or its 10 m ×10 m riparian plot.

4. For each type of influence, record the appropriate proximity class in the "HUMAN INFLUENCE" part of the "VISUAL RIPARIAN ESTIMATES" section of the Channel/Riparian Cross-section and Thalweg Profile Form. Proximity classes are:

   - B ("Bank") Present within the defined 10 m stream segment and located in the stream or on the stream bank.
   - C ("Close") Present within the 10 × 10 m riparian plot area, but away from the bank.
   - P ("Present") Present, but outside the riparian plot area.
   - O ("Absent") Not present within or adjacent to the10 m stream segment or the riparian plot area at the transect

5. Repeat Steps 1 through 4 for the right bank.

6. Repeat Steps 1 through 5 for each cross-section transect, recording data for each transect on a separate field form.

EQUIPMENT AND SUPPLIES FOR PHYSICAL HABITAT

QTY.	Item	
1	Surveyor's telescoping leveling rod (round profile, metric scale, 7.5m extended)	
1	50-m fiberglass measuring tape & reel	
1	Hip chain (metric) for measuring reach lengths (Optional)	
1	Clinometer (or Abney level) with percent and degree scales.	
1	Lightweight telescoping camera tripod (necessary only if slope measurements are being determined by one person)	
2	½-inch diameter PVC pipe, 2-3 m long, each marked at the same height (for use in slope determinations involving two persons)	
1	Meter stick. Alternatively, a short (1-2 m) rod or pole (e.g., a ski pole) with cm markings for thalweg measurements, or the PVC pipe described for slope determinations can be used	
1 roll ea.	Colored surveyor's plastic flagging (2 colors)	
1	Convex spherical canopy densiometer (Lemmon Model B), modified with taped "V"	
1	Bearing compass (Backpacking type)	
1 or 2	Fisherman's vest with lots of pockets and snap fittings. Used at least by person conducting the in-channel measurements to hold the various measurement equipment (densiometer, clinometer, compass, etc.). Useful for both team members involved with physical habitat characterization.	
2 pair	Chest waders with felt-soled boots for safety and speed if waders are the neoprene "stocking" type. Hip waders can be used in shallower streams.	
	Covered clipboards (lightweight, with strap or lanyard to hang around neck)	
	Soft (#2) lead pencils (mechanical are acceptable)	
11 plus extras	Channel/Riparian Cross-section & Thalweg Profile Forms	
1 plus extras	Slope and Bearing 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 7-11. Checklist of equipment and supplies for physical habitat.

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SECTION 8
PERIPHYTON

by
Brian H. Hill[1]

Periphyton are algae, fungi, bacteria, protozoa, and associated organic matter associated with channel substrates. Periphyton are useful indicators of environmental condition because they respond rapidly and are sensitive to a number of anthropogenic disturbances, including habitat destruction, contamination by nutrients, metals, herbicides, hydrocarbons, and acidification.

The "biomorphs" (refer to Figure 2-1) collect periphyton samples after completing activities pertaining to water chemistry (Section 5) and discharge (Section 6). Periphyton samples are collected from erosional and depositional habitats located at each of the nine interior cross-section transects (transects "B" through "J") established within the sampling reach (Section 4). Periphyton samples are collected at each transect at the same time as sediment samples (Section 9) and benthic macroinvertebrate samples (Section 11). At each stream, composite "index" samples of periphyton are prepared for erosional and depositional habitats. At the completion of the day's sampling activities, but before leaving the stream, four types of laboratory samples are prepared from each composite index sample.

8.1 SAMPLE COLLECTION

The general scheme for collecting periphyton samples from the sampling reach at each stream is illustrated in Figure 8-1. The procedure for collecting periphyton samples is presented in Table 8-1. At each transect, samples are collected from an assigned sampling point (left, center, or right). Sampling points at each transect may have been assigned when the sampling reach was laid out (Figure 8-1; refer also to Section 4; Table 4-3). If not,

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

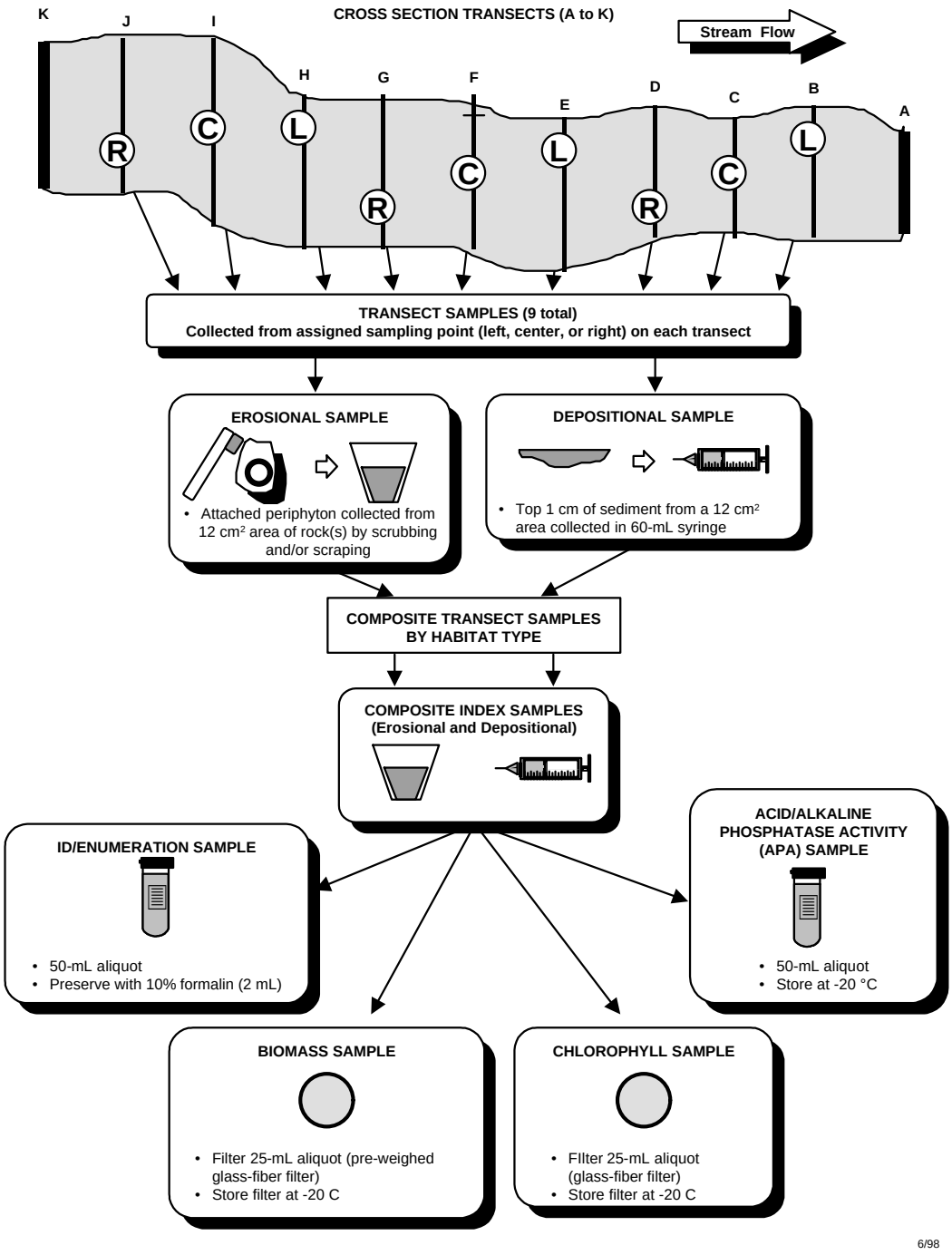


Figure 8-1. Index sampling design for periphyton.

TABLE 8-1. PROCEDURE FOR COLLECTING COMPOSITE INDEX SAMPLES OF PERIPHYTON

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

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

2A. Erosional habitats:

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

   (2) Use the area delimiter to define a 12-cm^2 area on the **upper** surface of the substrate. Dislodge attached periphyton from the substrate within the delimiter into the funnel by brushing with a stiff-bristled toothbrush for 30 seconds. **Take care to ensure that the upper surface of the substrate is the surface that is being scrubbed, and that the entire surface within the delimiter is scrubbed.**

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

2B. Depositional habitats:

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

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

   (3) Empty the syringe into a 500-mL plastic bottle with volume graduations marked on it and labeled "DEPOSITIONAL."

3. Repeat Steps 1and 2 for transects “C” through “J”. Place the sample collected at each sampling site into its appropriate 500-mL bottle ("EROSIONAL" or "DEPOSITIONAL") to produce the composite index sample for each habitat type.

4. After samples have been collected from all nine transects, mix each 500-mL bottle thoroughly. For each composite sample, place an “X” in the appropriate habitat type box (“riffle” for erosional; “pool” for depositional) and record the total estimated volume of the composite sample in the periphyton section of the Sample Collection Form.

the sampling point at Transect “B” is assigned at random using a die or other suitable means (e.g., digital watch). Once the first sampling point is determined, depositional sample, depending on whether the habitat at the site is flowing water (e.g., a riffle or run) or slack water (e.g., a pool). Composite samples for erosional and depositional habitats are prepared by combining the individual transect samples as they are collected from each habitat into separate plastic bottles. The habitat type and volume of each composite sample are recorded on the Sample Collection Form as shown in Figure 8-2.

8.2 PREPARATION OF LABORATORY SAMPLES

Four different types of laboratory samples are prepared from each of the two composite index samples: an ID/enumeration sample (to determine taxonomic composition and relative abundances), a chlorophyll sample, a biomass sample (for ash-free dry mass [AFDM]), and an acid/alkaline phosphatase activity (APA) sample. All the sample containers required for an individual stream should be sealed in plastic bags until use (see Section 3) to avoid external sources of contamination (e.g., dust, dirt, or mud) that are present at streamside.

A set of completed periphyton sample labels is shown in Figure 8-3. All labels in a set have the same sample ID number. Circle the 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.

8.2.1 ID/Enumeration Sample

Prepare the ID/Enumeration samples as 50-mL aliquots from each composite index sample, following the procedure presented in Table 8-2. Preserve each sample with 2 mL of 10% formalin. For each habitat type (riffle and pool), record the ID number (barcode) from each sample container label and the total volume of the sample in the appropriate fields on the Sample Collection Form as shown in Figure 8-2. Store the preserved samples upright in a container containing absorbent material, according to the guidelines provided for handling formalin-preserved samples.

Reviewed by (initial): _____

SAMPLE COLLECTION FORM - STREAMS

SITE NAME: MILL CREEK DATE: 7 / 15 / 97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999 ___ ___ TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

COMPOSITE BENTHOS SAMPLES

SAMPLE ID (BARCODE)	HABITAT (X ONE) R	HABITAT (X ONE) P	NO. OF JARS	FLAG	COMMENTS
229001	X		2		
229002		X	1		

STATION	A	B	C	D	E	F	G	H	I	J	K
RIFFLE OR POOL - (X ONE) →		☒R ☐P	☐R ☒P	☐R ☒P	☒R ☐P	☐R ☒P	☐R ☒P	☐R ☒P	☐R ☒P	☒R ☐P	
LEFT, CENTER, OR RIGHT - (X ONE) →		☐L ☐C ☒R	☒L ☐C ☐R	☐L ☒C ☐R	☐L ☐C ☒R	☒L ☐C ☐R	☐L ☒C ☐R	☐L ☐C ☒R	☒L ☐C ☐R	☐L ☒C ☐R	

COMPOSITE PERIPHYTON SAMPLES

HABITAT TYPE (X) → ☒ RIFFLE ☐ POOL ☐ OTHER

SAMPLE ID (BARCODE) → 229004

COMPOSITE VOLUME → 200 mL

ASSEMBLAGE ID (50-mL TUBE)	CHLOROPHYLL (GF/F FILTER)	BIOMASS (TARED FILTER)		APA SAMPLE (50-mL TUBE)
SUB. SAMPLE VOL.	VOL. FILTERED	FILTER NO.	VOL. FILTERED	SUB. SAMPLE VOL.
50 mL	25 mL	999	25 mL	50 mL

COMPOSITE PERIPHYTON SAMPLES

HABITAT TYPE (X) → ☐ RIFFLE ☒ POOL ☐ OTHER

SAMPLE ID (BARCODE) → 229003

COMPOSITE VOLUME → 300 mL

ASSEMBLAGE ID (50-mL TUBE)	CHLOROPHYLL (GF/F FILTER)	BIOMASS (TARED FILTER)		APA SAMPLE (50-mL TUBE)
SUB. SAMPLE VOL.	VOL. FILTERED	FILTER NO.	VOL. FILTERED	SUB. SAMPLE VOL.
50 mL	25 mL	1001	25 mL	50 mL

COMMENTS:

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

Rev. 06/02/97 (st_saco.97) SAMPLE COLLECTION FORM - STREAMS - 1

Figure 8-2. Sample Collection Form (page1) showing data recorded for periphyton samples.

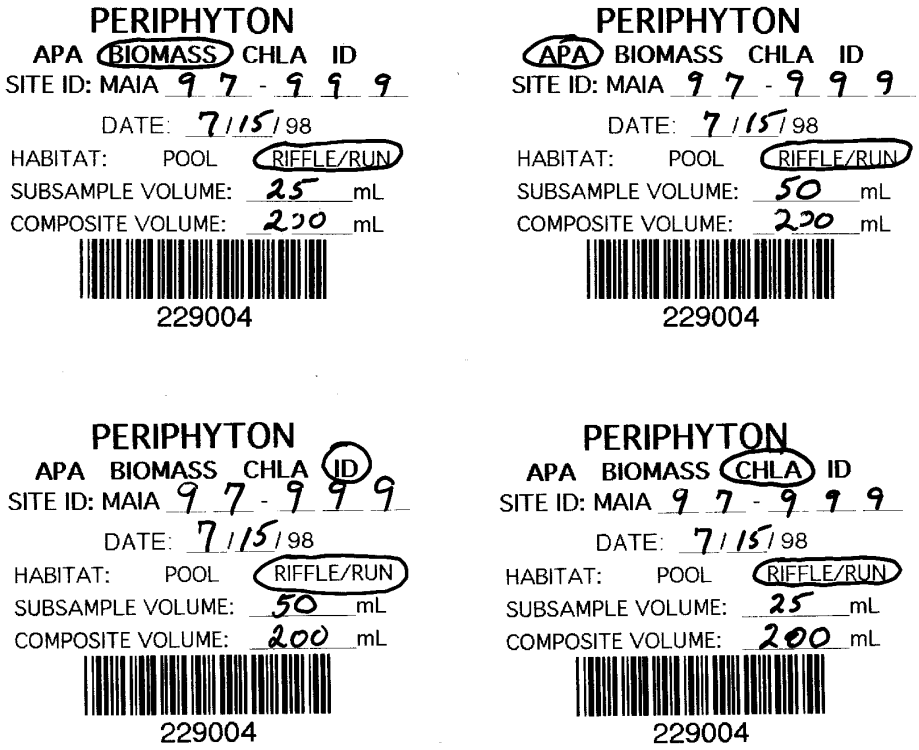


Figure 8-3. Completed set of periphyton sample labels.

8.2.2 Chlorophyll Sample

Prepare chlorophyll samples by filtering a 25-mL aliquot of each composite index sample through a glass fiber filter (0.4 to 0.6 :m nominal pore size). The procedure for preparing chlorophyll samples is presented in Table 8-3. 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 collection to minimize degradation. The filtration apparatus is illustrated in Figure 8-4. Rinse the filtration chamber with deionized water each day before use at the base site and then seal in a plastic bag until use at the stream (see Section 3). Keep the glass fiber filters in a dispenser inside a sealed plastic bag until use.

It is important to measure the volume of the sample being filtered accurately (±1 mL) with a graduated cylinder. During filtration, do no exceed 7 pounds per square inch (psi) to avoid rupturing cells. If the vacuum pressure exceeds 7 psi, prepare a new sample. If the

TABLE 8-2. PREPARATION OF ID/ENUMERATION SAMPLES FOR PERIPHYTON

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

2. Prepare a barcoded sample label. Circle the sample type ("ID") and habitat type ("RIFFLE/RUN" for EROSIONAL. "POOL" for DEPOSITIONAL) on the label. Record the volume of the sub-ample (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 mark-ings. Cover the label completely with a clear tape strip.

3. Place and "X" in the appropriate "HABITAT TYPE" box (riffle or pool) in the first "COMPOSITE PERIPHYTON SAMPLE" section of the Sample Collection Form. Record the sample ID number (barcode) of the label and the total volume of the composite index sample on the form.

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

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

6. 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.

8. Repeat Steps 1 through 7 above for the DEPOSITIONAL composite index sample. Record information in the second "COMPOSITE PERIPHYTON SAMPLE" section of the Sample Collection Form.

TABLE 8-3. PROCEDURE FOR PREPARING CHLOROPHYLL SAMPLES FOR PERIPHYTON

1. Mix the "EROSIONAL" composite index sample bottle thoroughly.

2. Using clean forceps, place a glass fiber filter on the filter holder. Use a small amount of deionized water from a wash bottle to help settle the filter properly. Attach the filter funnel to the filter holder and filter chamber, then attach the hand vacuum pump to the chamber.

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

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

   - NOTE: For composite samples containing fine sediment, (e.g., the "DEPOSITIONAL" sample), allow grit to settle before pouring the sample into the graduated cylinder.

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

   - If 25 mL of sample will not pass through the filter, discard the filter and rinse the chamber thoroughly with deionized water. Collect a new sample using a smaller volume of sample, measured to ±1 mL. Be sure to record the actual volume sampled on the sample label and the Sample Collection Form.

7. Remove both plugs from the filtration chamber and pour out the filtered water in the chamber. Remove the filter funnel from the filter holder. Remove the filter from the holder with clean forceps. Avoid touching the colored portion of the filter. Fold the filter in half, with the colored side folded in on itself. Wrap the folded filter in a small piece of aluminum foil.

9. Complete a periphyton sample label for chlorophyll, including the type of composite index sample and 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.

10. Place and "X" in the appropriate "HABITAT TYPE" box (riffle or pool) in the first "COMPOSITE PERIPHYTON SAMPLE section of the Sample Collection Form. 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.

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

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

13. Repeat Steps 1 through 12 for the "DEPOSITIONAL" composite index sample. Record information in the second "COMPOSITE PERIPHYTON SAMPLE" section on the Sample Collection Form.

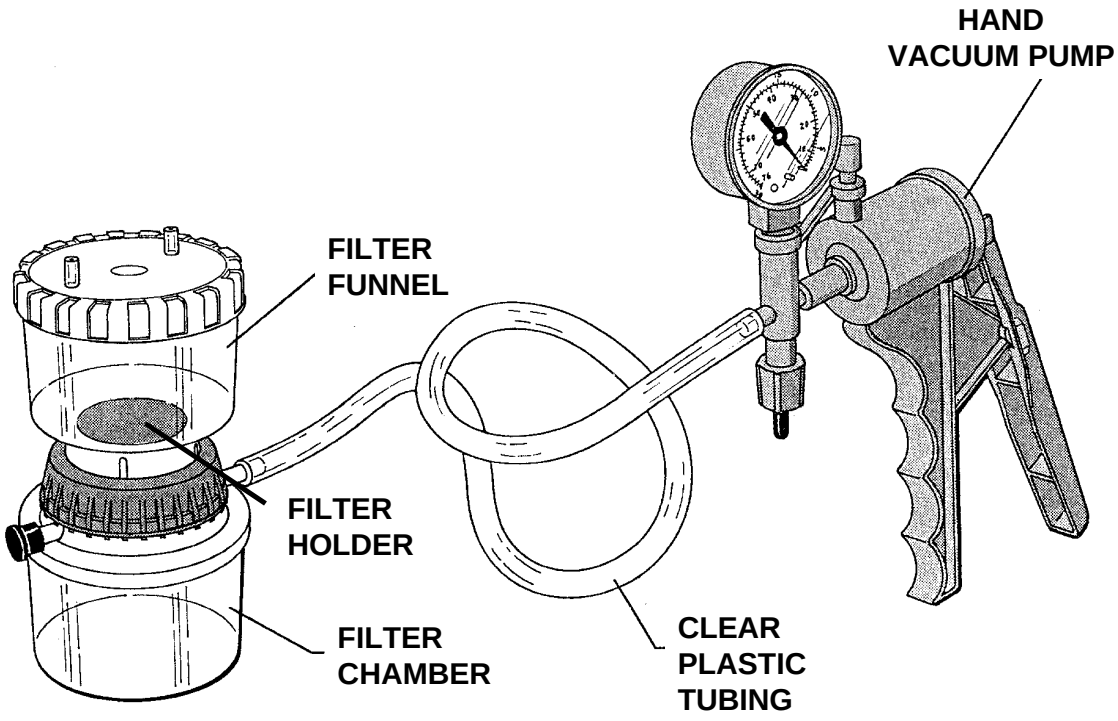


Figure 8-4. Filtration apparatus for preparing chlorophyll and biomass subsamples for periphyton. Modified from Chaloud et al. (1989).

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. Rinse the filtration unit and the graduated cylinder thoroughly with deionized water between the two composite index samples.

After filtering each sample, wrap the filter in aluminum foil. Complete a sample label (Figure 8-3) and check it to ensure that all written information is complete and legible. Affix the label to the 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 8-2). Make sure the volume recorded on each sample label matches the corresponding volume recorded on the Sample Collection Form. Record a flag and provide comments on the Sample Collec-

tion 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).

8.2.3 Biomass Sample

Prepare the biomass samples from 25-mL aliquots of each composite index sample. The filters for the biomass samples (the same type as is used for chlorophyll) may be provided in a sealed, numbered container. These filters have been prepared by combusting (30 min at 525 /C), desiccating, re-hydrating, drying (60 /C for 24 hours), then weighed to the nearest 0.01 mg. Prepare each sample according to the procedure presented in Table 8-4. Take extra care in handling the filters, as they may be very fragile as a result of their preparation. As with the chlorophyll sample, it is important to measure the volume to be filtered accurately (±1 mL). Rinse the filter chamber components (Figure 8-4) and the graduated cylinder thoroughly between the two composite index samples with deionized water.

After filtering each sample, do not fold the filter (as was done for the chlorophyll sample). Place the unfolded filter back into its numbered container. Complete a sample label as shown in Figure 8-3. 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 8-2. 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 container frozen until shipment to the laboratory (Section 3).

8.2.4 Acid/Alkaline Phosphatase Activity Sample

The Acid/Alkaline phosphatase activity (APA) samples are prepared from 50-mL subsamples of each composite index sample. Table 8-5 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 8-3 and affix it to a 50-mL centrifuge tube. Record the ID number (barcode), and the volume of the subsample on the Sample Collection Form (Figure 8-2). Check to ensure that the information recorded on the

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

1. Mix the "EROSIONAL" composite index sample bottle thoroughly.

2. Using clean forceps, remove a pre-leached, pre-weighed glass-fiber filter from its numbered container 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.

3. Rinse the filter chamber and filter with a small volume of deionized water.

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

    - NOTE: For composite samples containing fine sediment, (e.g., the "DEPOSITIONAL" sample), allow grit to settle before pouring the sample into the graduated cylinder.

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

    - If 25 mL of sample will not pass through the filter, discard the filter and rinse the chamber thoroughly with deionized water. Collect a new sample using a smaller volume of sample, measured to ±1 mL. Be sure to record the actual volume filtered on the sample label and the Sample Collection Form.

6. 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.

8. Place the **unfolded** filter back into its numbered container. Complete a periphyton sample label for biomass, including the type of index sample, the container number, and the volume filtered. Affix the label to the filter container and cover the label completely with a strip of clear tape.

9. Place and "X" in the appropriate "HABITAT TYPE" box (riffle or pool) in the first "COMPOSITE PERIPHYTON SAMPLE" section of the Sample Collection Form. Record the sample ID number (barcode) of the label and the total volume of the composite index sample on the form. Record the number from the filter container ("FILTER NO.") and 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 portable freezer, a cooler containing dry ice, or between two sealed plastic bags of ice in a cooler.

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

12. Repeat Steps 1 through 11 for the "DEPOSITIONAL" composite index sample. Record information in the second "COMPOSITE PERIPHYTON SAMPLE" section on the Sample Collection Form.

TABLE 8-5. PROCEDURE FOR PREPARING ACID/ALKALINE PHOSPHATASE ACTIVITY SAMPLES FOR PERIPHYTON

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

2. Prepare a barcoded sample label. Circle the sample type ("ID") and habitat type ("RIFFLE/RUN" for the "EROSIONAL" sample; "POOL" for the "DEPOSITIONAL" sample) 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.

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

4. Withdraw 50 mL of the composite index sample into the syringe. Place the contents of the syringe sample into the labeled 50-mL centrifuge tube. Cap the tube tightly and seal with plastic tape.

5. Place and "X" in the appropriate "HABITAT TYPE" box (riffle or pool) in the first "COMPOSITE PERIPHYTON SAMPLE" section of the Sample Collection Form. Record the sample ID number (barcode) of the label and the total volume of the composite index sample on the form.

7. Record the volume of the sample in the centrifuge tube in the "APA SAMPLE" field of the Sample Collection Form.

8. Repeat Steps 1 through 7 above for the "DEPOSITIONAL" composite index sample. Record information in the second "COMPOSITE PERIPHYTON SAMPLE" section of the Sample Collection Form.

Sample Collection Form matches the corresponding information recorded on the sample label. Store APA samples frozen until shipment to the laboratory (Section 3).

8.3 EQUIPMENT AND SUPPLIES

Figure 8-5 is a checklist of equipment and supplies required to conduct periphyton sample collection and processing activities. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the stream. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

8.4 LITERATURE CITED

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

EQUIPMENT AND SUPPLIES FOR PERIPHYTON

QTY.	Item	
1	Large funnel (15-20 cm diameter)	
1	12-cm^2 area delimiter (3.8 cm diameter pipe, 3 cm tall)	
1	Stiff-bristle toothbrush with handle bent at 90^o angle	
1	1-L wash bottle for stream water	
1	1-L wash bottle containing deionized water	
2	500-mL plastic bottles for composite index samples, labeled “EROSIONAL” and “DEPOSITIONAL”	
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 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	Pre-leached, pre-ashed, weighed glass-fiber filters in numbered containers for biomass sample	
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	
2 sets	Sample labels (4 per set) with the same barcode ID number	
1	Sample Collection Form for stream	
	Soft (#2) lead pencils for recording data on field forms	
	Fine-tipped indelible markers for filling out sample labels	
1 pkg.	Clear tape strips for covering labels	
1	Portable freezer, cooler with dry ice, or cooler with bags of ice to store frozen samples	
1 copy	Field operations and method manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for peri-phyton	

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

SECTION 9
SEDIMENT COMMUNITY METABOLISM

by
Brian H. Hill[1]

This section describes procedures to collect a composite sediment sample from the sampling reach. The "biomorphs" (refer to Figure 2-1) collect sediment samples from each transect at the same time as periphyton samples (Section 8) and benthic macroinvertebrate samples (Section 11). At each stream, a composite "index" sample of sediment is prepared. A portion of this composite sample is used in the determination of sediment community metabolism. The remaining composite sample is prepared for use in toxicity testing, if necessary (see Section 1.3.8 and Section 10).

The method outlined here for determining sediment community metabolism is designed for headwater to mid-order streams, though it may be 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 a study reach defined by 40 times the channel width. Following incubation, the DO is remeasured 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.

9.1 SAMPLE COLLECTION

Table 9-1 describes the procedure for collecting the composite sediment sample. Collect sediment from depositional areas (e.g., pools, eddies, and backwaters) located at or

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

TABLE 9-1. SEDIMENT COLLECTION PROCEDURE

1. At cross-section transect “B”, 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 3 L of sediment (- 400 mL of sediment per transect) is required for both sediment metabolism and sediment toxicity. If a sediment toxicity sample is not required, 250 mL of sediment (- 30 mL per transect) is sufficient.

3. Repeat Steps 1 through 2 for Transects “C” through “J”.

near each of the nine interior cross-section transects ("B" through "J") 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 for the stream reach. A composite sample volume of 250-mL is sufficient to prepare sediment metabolism samples. An additional 1 to 2 L of sediment is required for a sediment toxicity sample.

9.2 DETERMINING SEDIMENT RESPIRATION

The procedure to measure sediment respiration in presented in Table 9-2. 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 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 9-1. This concentration is assumed to be the initial concentration of all subsamples. Five 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 9-2. 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 9-1). The sediment in each tube is retained and stored frozen until it can be shipped to the laboratory (Section 3) to determine the AFDM.

9.3 EQUIPMENT AND SUPPLIES

Figure 9-3 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

TABLE 9-2. 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 and stirrer unit 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 stream temperature as possible (not air temperature) by using stream 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 **b** full with streamwater. Measure the dissolved oxygen and temperature of the water in the cooler. Record the values in the "INITIAL O_2" 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 th of the five labeled tubes.

6. Fill each tube to the top (no head space) with stream water from the cooler and seal the tube. Fill a centrifuge tube labeled "BLANK" with stream 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.

(continued)

TABLE 9-2 (Continued)

8. If necessary, 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 portable freezer, a container with dry ice, or 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.

11. If the remaining composite sediment sample will not be used for a sediment toxicity sample, discard the sample and rinse the composite sample container thoroughly with stream water and/or deionized water.

Reviewed by (initial): DP

FIELD MEASUREMENT FORM - STREAMS/RIVERS

SITE NAME: MILL CREEK DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999 ___ 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: E Assume X-site unless marked

		FLAG	COMMENTS
QCCS COND µS/CM	72		
STREAM/RIVER COND µS/CM	500		
STREAM/RIVER DO MG/L	4.6		
STREAM/RIVER TEMP °C	20.0		

STREAM/RIVER METABOLISM DETERMINATION

INITIAL O_2 (MG/L)	INITIAL INCUBATION TEMP. (°C)	INCUBATION TIME (24-HR TIME) START (HH:MM)	INCUBATION TIME (24-HR TIME) FINISH (HH:MM)	DURATION OF INCUBATION (HH:MM)	FLAG	COMMENTS
4.9	20.0	11:35	2:40	3:05		

SAMPLE ID (BARCODE)	FINAL O_2 (MG/L)	FLAG	COMMENTS
999991	2.0		
999992	1.9		
999993	1.8		
999994	1.9		
999995	1.1		
BLANK	4.5		

OXYGEN METER CALIBRATION INFORMATION

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

CALIBRATION CHAMBER TEMPERATURE:	27.9 °C	SATURATED O_2 @ TEMP.:	7.83 MG/L
STATION ELEVATION (FROM TOPO. MAP OR ALTIMETER):	1080 FT	ELEVATION CORRECTION FACTOR: ×	0.96
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:	7.52 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. 06/02/97 (strvfldm.97) FIELD MEASUREMENT FORM - STREAMS/RIVERS - 1

Figure 9-1. Field Measurement Form (page 1), showing data for sediment metabolism samples.

Figure 9-2. Completed sample labels for sediment metabolism.

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

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. If metabolism and toxicity samples are being prepared, use a 1-gallon jar	
1	YSI Model 58 Dissolved Oxygen meter with Model 5730 Stirring BOD probe	
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	Portable freezer, or cooler with bags of ice or dry 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 9-3. Checklist of equipment and supplies for sediment metabolism.

SECTION 10
SEDIMENT TOXICITY

by
James M. Lazorchak[1] and Mark E. Smith[2]

This section describes procedures to prepare a sediment sample for shipment to a laboratory for use in toxicity testing (see Section 1.3.8). The "biomorphs" (refer to Figure 2-1) collect sediment samples from each transect at the same time as periphyton samples (Section 8) and benthic macroinvertebrate samples (Section 11). At each stream, a composite "index" sample of sediment is prepared. A portion of this composite sample is used in the determination of sediment community metabolism (Section 9).

10.1 SAMPLE COLLECTION AND PREPARATION

The composite sediment sample remaining after the sediment respiration subsamples have been prepared is used to prepare a sediment toxicity sample. The procedure to prepare the sediment toxicity sample is presented in Table 10-1. A completed sample label for the sediment toxicity sample is shown in Figure 10-1. Record the sample ID number on the Sample Collection Form as shown in Figure 10-2. Use a heavy-duty self-sealing plastic bag as a sample container. Double-bag the sample and place it a suitably sized hard plastic container with a snap-on lid for transport and storage. Keep the sample chilled (but not frozen) until it can be shipped to the laboratory (Section 3).

10.2 EQUIPMENT AND SUPPLIES

Figure 10-3 is a checklist of equipment and supplies required to prepare the sediment toxicity sample. 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

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

2 SoBran Environmental, Inc., c/o U.S. EPA, 26 W. Martin Luther King Dr., Cincinnati, OH 45268.

TABLE 10-1. PROCEDURE FOR PREPARING SEDIMENT TOXICITY SAMPLES

1. Complete a sediment toxicity sample label with the stream ID and the date of collection. If sediment for the composite sample was collected at several cross-section transects, write "ALL" in the "STATION" field.

2. Record the sample ID number (barcode) printed on the label in the "SEDIMENT TOXICITY SAMPLES" section of the Sample Collection Form (page 2).

3. Attach the completed label to a 2-gallon polyethylene (4-mil) bag. Cover the label with a strip of clear tape.

4. Mix sediment well with a stainless steel or plastic mixing spoon, or gloved hand. Transfer at least 1 L of sediment from the composite sediment index sample container to the labeled plastic bag. Close bag, squeeze air out and tie a knot in the remaining portion of the bag to seal. Seal the bag.

5. Place the labeled bag with the sample inside a second 2-gallon polyethylene bag and tie off the top to seal. Place the sample into a hard plastic container with a snap-on lid (if available), to further protect the sample.

6. Place the sample inside a cooler containing bags of ice that is used only for sediment samples in them. Store the sediment toxicity sample chilled, but not frozen, until it can be shipped to the laboratory.

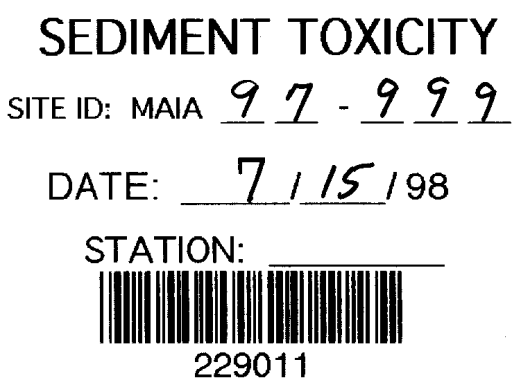


Figure 10-1. Completed sample label for sediment toxicity.

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

Reviewed by (initial): DP

SAMPLE COLLECTION FORM - STREAMS (continued)

SITE NAME: Mill Creek DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999 ___ 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	229015	X		
MICROBIAL				

SEDIMENT TOXICITY SAMPLES

Sample ID (Barcode)	Flag	Comments
229011		

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

Sample ID (Barcode) → 229013

Line	Species Code	Common Name	Number of Individuals	Flag
P1	NOCOLE	Bluehead chub	16	F1

Is composite sample composed of individuals collected from throughout reach? (X) → ☒ Yes ☐ No

If No, Explain:

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

Sample ID (Barcode) → 229012

Line	Species Code	Common Name	Total Length (mm)	Flag
S1	CATOCO	White sucker	128	
S2	CATOCO	White sucker	125	
S3	CATOCO	White sucker	134	
S4	CATOCO	White sucker	128	
S5	CATOCO	White sucker	125	

Is composite sample composed of individuals collected from throughout reach? (X) → ☒ Yes ☐ No

If No, Explain:

Line	Comment or Flag Explanation for Fish Tissue
P1	F1 = 16 individuals weighed 60 g.

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. 06/02/97 (st_saco.97) SAMPLE COLLECTION FORM - STREAMS - 2

Figure 10-2. Sample Collection Form (page 2), showing information recorded for a sediment toxicity sample.

EQUIPMENT AND SUPPLIES FOR SEDIMENT TOXICITY

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, use a 250-mL jar is sufficient. If metabolism and toxicity samples are being prepared, use a 1-gallon jar	
1	Sediment toxicity sample label	
1	Sample Collection Form	
	Soft (#2) lead pencils to fill in field data forms	
	Fine tip indelible markers for preparing labels	
2	1-gallon heavy-duty self-sealing plastic bags for the sediment toxicity sample	
1 pkg	Clear tape strips for covering labels	
1	Plastic container with snap-on lid to hold sediment toxicity sample	
1	Cooler with bags of ice to store the sediment toxicity sample	
1 copy	Field operations and methods manual	
1 set	Laminated sheets with procedure tables and/or quick reference guides for sediment toxicity	

Figure 10-3. Checklist of equipment and supplies for sediment toxicity.

SECTION 11
BENTHIC MACROINVERTEBRATES

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

Benthic invertebrates inhabit the sediment or live on the bottom substrates of streams. Benthic macroinvertebrate assemblages in streams reflect overall biological integrity of the benthic community. Monitoring these assemblages is useful in assessing the status of the water body and detecting trend in ecological condition. Benthic communities respond to a wide array of stressors in different ways so that it is often possible to determine the type of stress that has affected a macroinvertebrate community (e.g., Klemm et al., 1990). Because many macroinvertebrates have relatively long life cycles of a year or more and are relatively immobile, macroinvertebrate community structure is a function of present or past conditions.

The EMAP-SW benthic macroinvertebrate protocol is intended to evaluate the biological integrity of wadeable streams in the United States for the purpose of detecting stresses on community structure and assessing the relative severity of these stresses. It is based on the "Rapid Bioassessment Protocol III - Benthic Macroinvertebrates" published by the U.S. Environmental Protection Agency (Plafkin et al., 1989) and adopted for use by many states. The two man kick net procedure of the Rapid Bioassessment Protocol (RBP) is replaced in the EMAP-SW protocol with a kick net modified for use by one person (Figure 11-1), as is used by the U.S. Geological Survey for their National Water-Quality Assessment Program (NAWQA; Cuffney et al., 1993). This protocol requires only one person and is the preferred macroinvertebrate collecting method for streams with flowing water (a second person is often used for water safety and to keep time and record information on the field forms).

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

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

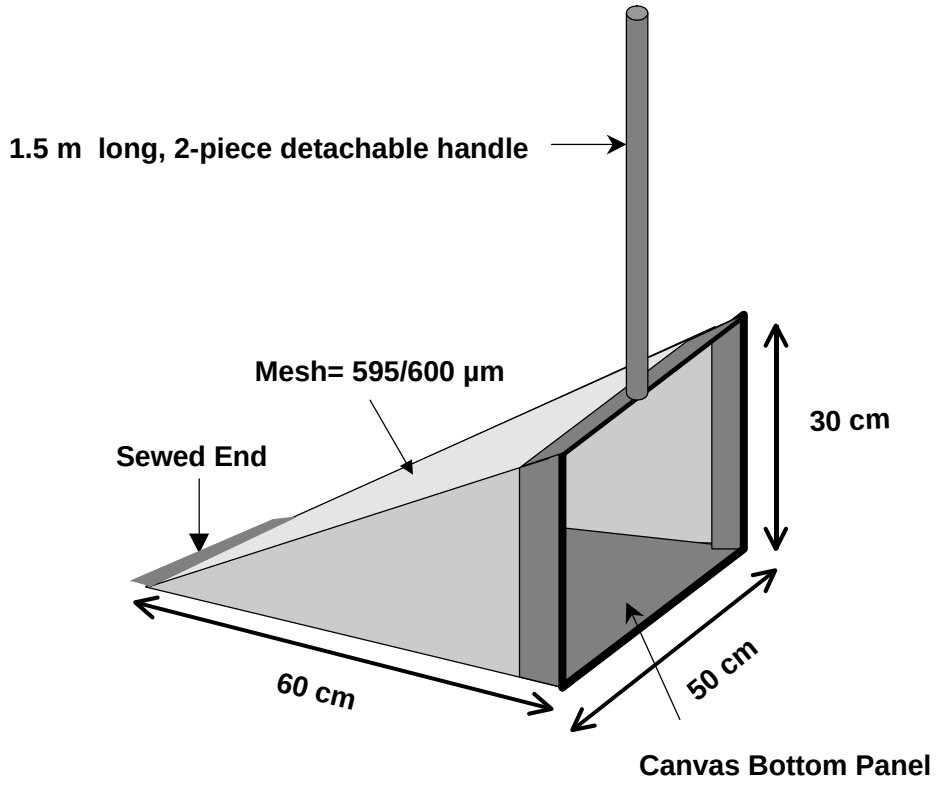


Figure 11-1. Modified kick net. (Not drawn to scale.)

The “biomorphs” (refer to Figure 2-1) collect kick net samples for benthic macro-invertebrate at sampling points located on each cross-section transect. Kick net samples are collected at the same time as periphyton samples (Section 8) and sediment samples (Section 9). Kick net samples collected from flowing water habitats (e.g., riffles, runs) are combined into a single composite sample for the stream reach. Kick net samples collected from pool habitats are combined into a separate composite sample.

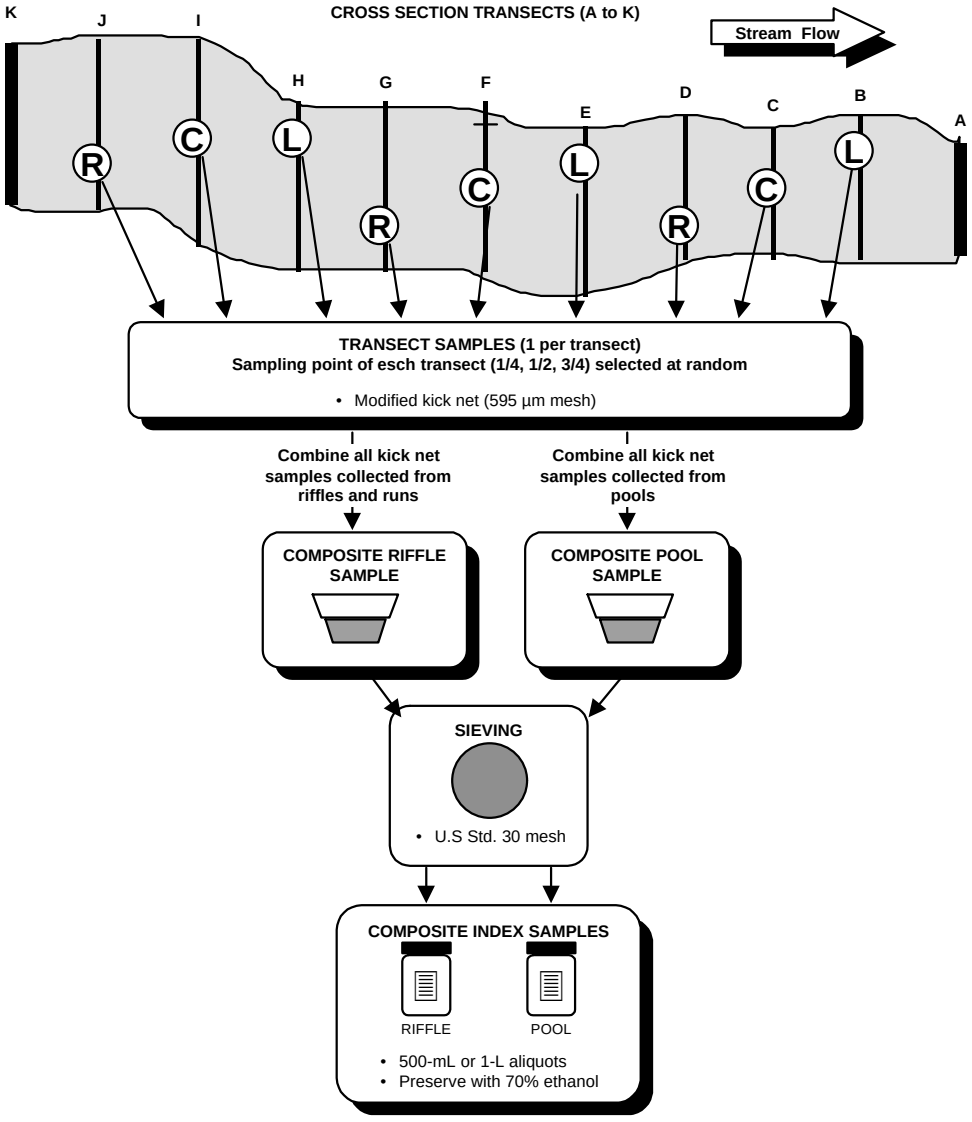
11.1 SAMPLE COLLECTION

The index sample design for benthic macroinvertebrates is shown in Figure 11-2. This design is used in the EMAP and R-EMAP stream studies in the mid-Atlantic region (refer to Section 1 for project descriptions). A modified index sample design was developed and implemented in some studies conducted in the western U.S. In the modified design, an equal number of kick net samples are collected from available riffle and pool habitats located within the sampling reach. This modified index sampling design is described in more detail in Appendix E.

A kick net sample is collected from each of the nine interior cross-section transects (Transects "B" through "J") at an assigned sampling point (Left, Center, or Right). These points may have been assigned when the sampling reach was laid out (Figure 11-2; refer also to Section 4; Table 4-3). If not, the sampling point at Transect "B" is assigned at random using a die or other suitable means (e.g., digital watch). Once the first sampling point is determined, points at successive transects are assigned in order (Left, Center, Right). These are the same sampling points as those used for periphyton samples (Section 8). At transects assigned a "Center" sampling point where the stream width is between one and two net widths wide, pick either the "Left" or "Right" sampling point instead. If the stream is only one net wide at a transect, place the net across the entire stream width and consider the sampling point to be "Center".

At each sampling point, determine if the habitat is a "riffle/run" or a "pool/glide". Any area where there is not sufficient current to extend the net is operationally defined as a pool/glide habitat. To collect a kick net sample from a sampling point classified as "riffle/run" habitat, follow the procedure presented in Table 11-1. To collect a kick net sample from a sampling point classified as a "pool/glide" habitat, follow the procedure presented in Table 11-2. Record the habitat type and sampling point for each kick net sample collected on the Sample Collection Form as shown in Figure 11-3. As you proceed upstream from transect to transect, combine all kick net samples collected from "riffle/run" habitats into a bucket or similar container labeled "RIFFLE". Combine kick net samples collected from "pool/glide" habitats into a second bucket labeled "POOL". Fill in the checklist shown in Figure 11-4 as individual activities are completed.

If it is impossible to sample at the sampling point with the modified kick net following either procedure, spend about 60 seconds hand picking a sample from about 0.25 m^2 of



9/16

Figure 11-2. Index sampling design for benthic macroinvertebrates.

TABLE 11-1. PROCEDURE TO COLLECT KICK NET SAMPLES FROM RIFFLE AND RUN HABITATS

1. At each cross-section transect, beginning with Transect "B", locate the assigned sampling point (Left, Center, or Right as you face downstream) as 25%, 50%, and 75% of the wetted width, respectively.

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

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

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

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

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

4. Holding the net in position on the substrate, visually define a rectangular quadrat that is one net width wide and two net widths long upstream of the net opening. The area within this quadrat is $\sim 0.5\ m^2$.

5. Check the quadrat for heavy organisms, such as mussels and snails. Remove these organisms from the substrate by hand and place them into the net.

6. Hold the net securely in position while kicking the substrate within the quadrat vigorously for 20 seconds (use a stopwatch).

7. After 20 seconds, hold the net in place with your knees and pick up any loose rocks within the quadrat. Use your hands to rub any clinging organisms off the rocks (especially those covered with algae or other debris) in front of the net. Also, place any additional mussels and snails found into the net. Remove the net from the water with a quick upstream motion to wash the organisms to the bottom of the net.

(continued)

TABLE 11-1. (Continued)

8. Invert the net into a bucket labeled “RIFFLE”, which is about half full of water, to rinse organisms out of the net. Inspect the net for clinging organisms. Use watchmakers’ forceps to remove any organisms from the net and place them in the bucket. Carefully inspect any large objects (such as rocks, sticks, and leaves) in the bucket and wash any organisms found off of the objects and into the bucket before discarding the object. Remove as much detritus as possible without losing any organisms.

9. Place an “X” in the appropriate habitat type and sampling point boxes for the transect on the Sample Collection Form.

10. Proceed upstream to the next transect and repeat Steps 1 through 9. Combine all kick net samples from “riffle/run” habitats into the “RIFFLE” bucket.

TABLE 11-2. PROCEDURE TO COLLECT KICK NET SAMPLES FROM POOL AND GLIDE HABITATS

1. At each cross-section transect, beginning with Transect "B", locate the assigned sampling point (Left, Center, or Right as you face downstream) as 25%, 50%, and 75% of the wetted width, respectively.

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

2. Attach the 4-ft handle to the kick net. Make sure that the handle is on tight or the net may become twisted during the collection process, causing the loss of part of the sample.

3. Determine if there is sufficient current in the area at the sampling point to fully extend the net. If so, use the sampling procedure described for "Riffle/run" habitats. If not, classify the habitat as "Pool/glide" and proceed to Step 4. NOTE: If the pool is too deep (much more than 1 m) to sample safely at the designated spot, move downstream till a safe sampling spot is found.

4. Visually define a rectangular quadrat that is one net width wide and two net widths long at the sampling point. The area within this quadrat is ~0.5 m^2.

4. Inspect the stream bottom within the quadrat for any heavy organisms, such as mussels and snails. Remove these organisms by hand and place them into the net or into a bucket labeled "POOL".

5. Vigorously kick the substrate within the quadrat with your feet while dragging the net repeatedly through the disturbed area just above the bottom. Keep moving the net all the time so that the organisms trapped in the net will not escape. Continue kicking the substrate and moving the net for 20 seconds. NOTE: If there is too little water to use the kick net, stir up the substrate with your gloved hands and use the U.S. Standard #30 sieve to collect the organisms from the water in the same way the net is used in larger pools.

6. After 20 seconds, hold the net between your legs and partially submerged. Pick up any loose rocks within the quadrat. Rub or brush any organisms found on them into the net. Also recheck the area for any additional snails or clams and place them in the net.

7. Invert the net into a bucket labeled "POOL", which is about half full of water, to rinse organisms out of the net. Inspect the net for clinging organisms. Use watchmakers' forceps to remove any organisms from the net and place them in the bucket. Carefully inspect any large objects (such as rocks, sticks, and leaves) in the bucket and wash any organisms found off of the objects and into the bucket before discarding the object. Remove as much detritus as possible without losing any organisms.

8. Place an "X" in the appropriate habitat type and sampling point boxes for the transect on the Sample Collection Form.

9. Proceed upstream to the next transect and repeat Steps 1 through 8. Combine all kick net samples from "pool/glide" habitats into the "POOL" bucket.

Reviewed by (initial): DP

SAMPLE COLLECTION FORM - STREAMS

SITE NAME: MILL CREEK DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

COMPOSITE BENTHOS SAMPLES

SAMPLE ID (BARCODE)	HABITAT (X ONE) R	HABITAT (X ONE) P	NO. OF JARS	FLAG	COMMENTS
229001	X		2		
229002		X	1		

STATION	A	B	C	D	E	F	G	H	I	J	K
RIFFLE OR POOL - (X ONE) →		☒R ☐P	☐R ☒P	☐R ☒P	☒R ☐P	☐R ☒P	☐R ☒P	☐R ☒P	☐R ☒P	☒R ☐P	
LEFT, CENTER, OR RIGHT - (X ONE) →		☐L ☐C ☒R	☒L ☐C ☐R	☐L ☒C ☐R	☐L ☐C ☒R	☒L ☐C ☐R	☐L ☒C ☐R	☐L ☐C ☒R	☒L ☐C ☐R	☐L ☒C ☐R	

COMPOSITE PERIPHYTON SAMPLES

HABITAT TYPE (X) → ☒ RIFFLE ☐ POOL ☐ OTHER

SAMPLE ID (BARCODE) → 229004 COMPOSITE VOLUME → 200 mL

ASSEMBLAGE ID (50-mL TUBE)	CHLOROPHYLL (GF/F FILTER)	BIOMASS (TARED FILTER)		APA SAMPLE (50-mL TUBE)
SUB. SAMPLE VOL.	VOL. FILTERED	FILTER NO.	VOL. FILTERED	SUB. SAMPLE VOL.
50 mL	25 mL	999	25 mL	50 mL

COMPOSITE PERIPHYTON SAMPLES

HABITAT TYPE (X) → ☐ RIFFLE ☒ POOL ☐ OTHER

SAMPLE ID (BARCODE) → 229003 COMPOSITE VOLUME → 300 mL

ASSEMBLAGE ID (50-mL TUBE)	CHLOROPHYLL (GF/F FILTER)	BIOMASS (TARED FILTER)		APA SAMPLE (50-mL TUBE)
SUB. SAMPLE VOL.	VOL. FILTERED	FILTER NO.	VOL. FILTERED	SUB. SAMPLE VOL.
50 mL	25 mL	1001	25 mL	50 mL

COMMENTS:

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

Rev. 06/02/97 (st_saco.97) SAMPLE COLLECTION FORM - STREAMS - 1

Figure 11-3. Sample Collection Form (page 1), showing information for benthic macroinvertebrate samples.

MACROINVERTEBRATE SAMPLING ACTIVITIES CHECKLIST		
Date:	**Time:**	**Site No.:**
Stream Name and Location:		
Crew ID: 1 2 3 4 5 6	**Collector:**	
1.	Initial observations, if any, on the Sample Collection Form - Streams.	
2.	Composite riffle/run sample collected with a label inside the jar.	
3.	Composite pool/glide sample collected with a label inside the jar.	
4.	Correct barcode and label on all jars and sealed with clear, waterproof tape.	
5.	All samples preserved.	
6.	With a grease pencil write site number, sample type (Riffle or Pool), and number of transects sampled for sample type on the cap. If two jars are used be sure to mark them as such.	
7.	Caps are sealed with plastic electrical tape.	
8.	Photos of the site.	
9.	Sample jars in cooler or otherwise secured.	
10.	All equipment accounted for and secured in the vehicle.	
Signature:		**Time sampling completed:**

Figure 11-4. Checklist for benthic macroinvertebrate sampling activities.

substrate at the sampling point. Place the contents of this hand-picked sample into either the "RIFFLE" or "POOL" bucket.

11.2 SAMPLE PROCESSING

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

Complete the check-off sheet (Figure 11-4). Check to be sure that the prenumbered adhesive barcoded label is on the jar and covered with clear tape, and that the waterproof label is in the jar and filled in properly. Be sure the inside label and outside label describe the same sample. Replace the cap on each jar and seal them with plastic electrical tape. Check to make sure the cap is properly marked with site number, habitat type (pool or riffle), and number of transects sampled. Record any additional pertinent information in the “Field Notes” section of the checklist. Place the samples in a cooler or other secure container for transporting and/or shipping the laboratory (see Section 3). The container and absorbent material should both be suitable for transporting ethanol. Check to see that all equipment is in the vehicle.

11.3 EQUIPMENT AND SUPPLY CHECKLIST

Figure 11-7 shows the checklist of equipment and supplies required to complete the collection of benthic macroinvertebrates from streams. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the stream. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

TABLE 11-3. PROCEDURE FOR PREPARING COMPOSITE SAMPLES FOR BENTHIC MACROINVERTEBRATES

1. Pour the entire contents of the "RIFFLE" bucket through a U.S. Standard #30 sieve (or sieve-bottomed bucket with 595 :m mesh size). Remove any large objects and wash off any clinging organisms back into the sieve before discarding.

2. Using a wash bottle filled with stream water, rinse all the organisms from the bucket into the sieve. This is the composite sample for that habitat (riffle or pool) for the site.

3. Estimate the total volume of the sample in the sieve and determine how large a jar will be needed for the sample (half gallon or gallon). Do not use more than one jar for each of the samples unless it cannot be avoided.

4. Fill in a "Composite Benthos" sample label with the stream ID and date of collection. Circle the habitat type (Riffle or Pool). Attach the completed label to the jar and cover it with a strip of clear tape.

4. Wash the contents of the sieve to one side by gently agitating the sieve in the water. Wash the sample into a jar using as little water from the wash bottle as possible. Use a large-bore funnel if necessary. If the jar is too full pour off some water through the sieve until the jar is not more than ¼ full, or use a second jar if a larger one is not available. Carefully examine the sieve for any remaining organisms and use watchmakers' forceps to place them into the sample jar.

   - If a second jar is needed, fill in a sample label that does not have a pre-printed barcode number on it. Record the barcode number from the pre-printed label prepared in Step 4 in the "BARCODE" field of the label. Attach the label to the second jar and cover it with a strip of clear tape.

5. Add 95% ethanol to each 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.

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

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

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

7. Replace the cap on each jar. Seal each jar with plastic tape. Use a grease pencil to write the site number, sample type (Riffle or Pool), and number of transects on the cap of each jar.

8. Repeat Steps 1 through 7 for the "POOL" bucket.

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

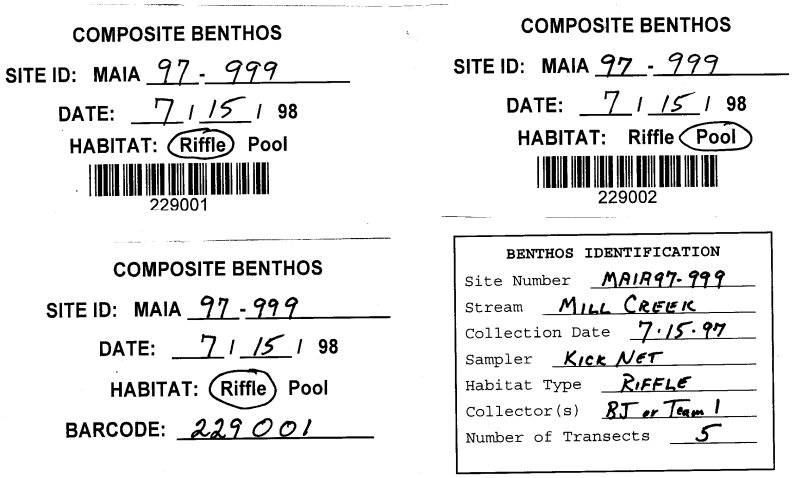


Figure 11-5. Completed labels for benthic macroinvertebrate samples. The label at lower left is used if more than one jar is required for a composite sample. The label at lower right is placed inside the sample container.

11.4 LITERATURE CITED

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

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

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

BENTHOS IDENTIFICATION

Site Number ___________________
Stream _________________________
Collection Date _______________
Sampler _______________________
Habitat Type __________________
Collector(s) __________________
Number of Transects __________

BENTHOS IDENTIFICATION

Site Number ___________________
Stream _________________________
Collection Date _______________
Sampler _______________________
Habitat Type __________________
Collector(s) __________________
Number of Transects __________

BENTHOS IDENTIFICATION

Site Number ___________________
Stream _________________________
Collection Date _______________
Sampler _______________________
Habitat Type __________________
Collector(s) __________________
Number of Transects __________

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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 11-6. Blank labels for benthic invertebrate samples.

EQUIPMENT AND SUPPLIES FOR BENTHIC MACROINVERTEBRATES

QTY.	ITEM	
1	Modified kick net (closed bag with 595/600 :m mesh) and 4-ft handle (Wildco #425-C50)	
	Spare net(s) for the kick net sampler or extra sampler	
1	Watch with timer or a stopwatch	
2	Buckets, plastic, 8- to 10-qt capacity, labeled “RIFFLE” and “POOL”	
1	Sieve, U.S. Standard 30	
1	Sieve-bottomed bucket, 595- :m mesh openings	
2 pr.	Watchmakers’ forceps	
1	Wash bottle, 1-L capacity labeled “STREAM WATER”	
1	Small spatula, spoon, or scoop to transfer sample	
1	Funnel, with large bore spout	
4 to 6 each	Sample jars, plastic with screw caps, ½ and 1 gallon capacity, suitable for use with ethanol	
2 gal	95% ethanol, in a proper container	
2 pr.	Rubber gloves, heavy rubber	
1	Cooler (with suitable absorbent material) for transporting ethanol and samples	
2	Composite Benthic sample labels, with preprinted ID numbers (barcodes)	
4	Composite Benthic sample labels without preprinted ID numbers	
6	Blank labels on waterproof paper for inside of jars	
1	Sample Collection Form for site	
1	Field check list sheet	
	Soft (#2) lead pencils	
	Fine-tip indelible markers	
	Grease pencils	
1 pkg.	Clear tape strips	
4 rolls	Plastic electrical tape	
1	Knife, pocket, with at least two blades	
1	Scissors	
1	Pocket-sized field notebook (optional)	
1 pkg.	Kim wipes in small self-sealing plastic bag	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for benthic macroinvertebrates	

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

SECTION 12
AQUATIC VERTEBRATES

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

Sampling amphibian and fish species to determine their proportionate abundances and the presence of external anomalies is conducted after all other field sampling and measurement activities are completed. The objective is to collect a representative sample of all except very rare species in the assemblage. Backpack electrofishing equipment is used as the principal sampling gear (Section 12.1.1), supplemented by block netting (when necessary) and seining (Section 12.1.2) in habitats where flow, substrate and structure affect capture of benthic species. All team personnel are involved in collecting aquatic vertebrates. In addition to gathering data on the assemblage, fish specimens are retained for analysis of tissue contaminants (Section 13).

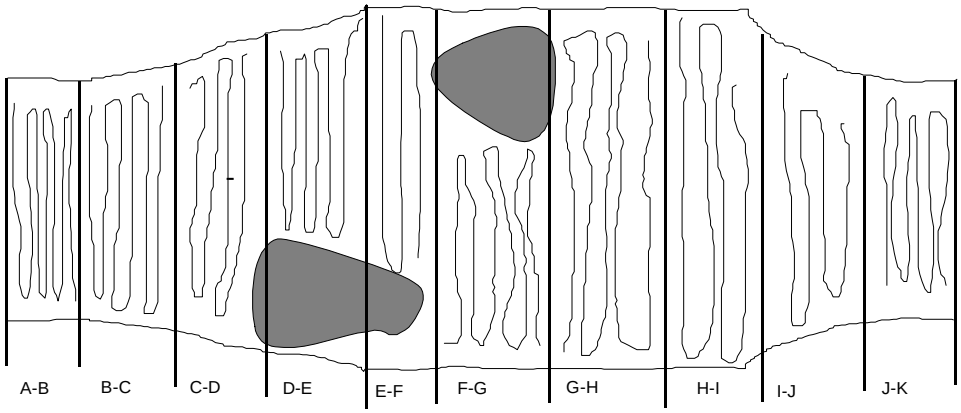
12.1 SAMPLE COLLECTION

The entire channel within the sampling reach is sampled. Complex, very large, or wide systems without clearly-defined habitat types are sampled through use of transects so that effort is distributed along the entire reach relative to the mean width of each transect, as illustrated in Figure 12-1. Fish and other aquatic vertebrates are collected according to time **and** distance criteria. Collection time should continue for not less than 45 minutes and not longer than 3 hours within the defined sampling reach (Section 4) to obtain a representative sample. Sampling information is recorded on the Vertebrate Collection Form (Figure 12-2). Record general comments (perceived fishing efficiency, missed fish, gear operation, suggestions) on the blank lines of the form.

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

Stippled areas represent habitats too hazardous to sample.



Transects correspond to cross-section transects established every 10 channel widths or 15 m

Transect	Mean Transect Width (m)	Time Allotment (sec)	Shock Time (estimated)
A-B	8m	8*84= 672	350
B-C	9m	9*84= 756	400
C-D	12m	12*84=1008	600
D-E	12m	12*84=1008	600
E-F	15m	15*84=1260	800
F-G	15m	15*84=1260	800
G-H	15m	15*84=1260	800
H-I	15m	15*84=1260	800
I-J	10m	10*84= 840	500
J-K	8m	8*84= 672	350

Example Calculation

Sum of Mean Transect Widths

8 + 9 + 12 + 12 + 15 + 15 + 15 + 15 + 10 + 8 = 128m.

3 hrs = 10800s sampling time.

10800s / 128m = 84 s/m of mean width.

Multiply mean width by # units to calculate time in seconds to be spent in each transect.

Depending on complexity of habitat, actual shock time may vary from 50% - 75% of fishing time.

Figure 12-1. Index sample design for allocating aquatic vertebrate sampling effort in very complex or very large wadeable streams. Note distribution of effort in narrow and wide sections.

12.1.1 Electrofishing

Because fishes and amphibians are collected using portable electrofishing units, safety procedures must be followed meticulously at all times (refer to Section 2). Primary responsibility for safety while electrofishing rests with the team leader. Electrofishing units have a high voltage output and may deliver a dangerous electrical shock. While electrofishing, avoid contact with the water unless sufficiently insulated against electrical shock. Use chest waders with nonslip soles and watertight rubber (or electrician's) gloves that cover to the elbows. If they become wet inside, **stop fishing until they are thoroughly dry. Avoid contact with the anode and cathode at all times due to the potential shock hazard.** If you perspire heavily, wear polypropylene or some other wicking and insulating clothing instead of cotton. While electrofishing avoid reaching into the water. 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 interrupted and the anode is removed from the water**. Do not resume electrofishing until all individuals are clear of the electroshock hazard. The electrofishing equipment is equipped with a 45° tilt switch that interrupts the current. Do not make any modifications to the electrofishing unit that would hinder turning off the electricity.

Avoid operating electrofishing equipment near unprotected people, pets, or livestock. Discontinue activity during thunderstorms or heavy rain. Team members should keep each other in constant view or communication while electrofishing. For each site, know the location of the nearest emergency care facility. Although the team leader has authority, each team member has the responsibility to question and modify an operation or decline participation if it is unsafe. Use hand signals to communicate direction and power on or off when using generators.

Gasoline is extremely volatile and flammable. Its vapors readily ignite on contact with heat, spark or flame. Never attempt to refill the generator while it is running. **Always allow the generator to cool before refilling**. Keep gasoline out of direct sunlight to reduce volatilization and vapor release. Always wear gloves and safety glasses when handling gasoline. Keep gasoline only in approved plastic containers and store in a tightly closed container in safety cabinet or cooler lined with vermiculite.

The procedure to sample with the backpack electrofisher unit is presented in Table 12-1. Record information on the Vertebrate Collection Form as shown in Figure 12-2. If the stream cannot be sampled by either electrofishing or seining, complete the "NOT FISHED" field on the form. Select the initial voltage based on the measured conductivity of the stream (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.

Determine that all team members are wearing waders and gloves and are clear of the anode. Wear polarized sunglasses to aid vision. Start the electrofisher, set the timer to zero, and depress the switch to begin fishing. Starting at the bottom of the reach, fish in an upstream direction. Adjust voltage and waveform output according to sampling effectiveness and incidental mortality to specimens. The backpack unit is equipped with an audio alarm that sounds when the output voltage exceeds 30 V. It also serves as an input current indicator for pulse cycles greater than 5Hz. It begins as a strong continuous tone and begins to beep slowly at currents of 1.25 amps. It beeps faster as input current increases. In case of an overload (in excess of 3 amps), the beep becomes very rapid and the overload indicator comes on. Release the anode switch and adjust voltage and waveform and continue fishing.

When fishing, **slowly** sweep the electrode wand from side to side in the water in riffles and pools. Sample available cut-bank and snag habitat areas as well as riffles and pools. Move the wand in and out of large snags or deep cuts or release the electrode switch, move the wand away slightly, depress the switch again and sweep the wand away from the cover to draw fish out into open. In fast, shallow water, it may be more effective to use a seine as a block net; sweep the anode and fish downstream into the net.

In extremely wide streams, it may be necessary to work from the midline of the stream channel to the banks. Be sure that deep, shallow, fast, slow, complex, and simple habitats are all sampled. In stretches with deep pools, fish the margins of the pool as much as possible, being extremely careful not to step into deep water. In larger or more complex streams, allocate the fishing time between transects based on differences in the mean wetted width of the stream (Figure 12-1).

One or two netters follow along beside or slightly behind the person operating the electrofisher (on the anode side). Each netter uses an insulated dip net to retrieve stunned individuals, which are then deposited into a bucket for later processing (Section 12.3).

TABLE 12-1. PROCEDURE TO COLLECT AQUATIC VERTEBRATES BY ELECTROFISHING

1. Survey the sampling reach and set block nets at each end (Transects "A" and "K") if necessary (e.g., the majority of the reach is one large, continuous pool). If necessary, allocate the total shocking time among transects based on mean stream widths.

2. Complete the header information on a copy of the Vertebrate Collection Form. Indicate that all transects are being sampled in the "TRANSECT" field on the form.

   - NOTE: Make an effort to search and sample for aquatic vertebrates at all streams, even if the stream is extremely small, and it appears that sampling may not collect any specimens. If no specimens are collected, complete the "NONE COLLECTED" field on the Vertebrate Collection Form. Provide an explanation in the comments section of the form.

3. If the conductivity measured during the water chemistry sampling is less than 10 uS/cm, or if the depth or velocity make electrofishing unsafe, sample by seining if possible, otherwise do not sample. If you do not sample, complete the "NOT FISHED" field on the Vertebrate Collection Form. Provide an explanation in the comments section of the form.

4. Set unit to 300 volt-amperes (VA) and pulsed DC. Select initial voltage setting (150-400 V for high conductivity [>300 :S/cm]; 500-800 V for medium conductivity [100 to 300 :S/cm]; 900-1100 V for low conductivity [<100 :S/cm] waters). In waters with strong-swimming fish (length >200 mm), use a frequency of 30 Hz with a pulse width of 2 msec. If mostly small fish are expected, use a frequency of 60-70 Hz. Start the generator, set the timer, and depress the switch to begin fishing.

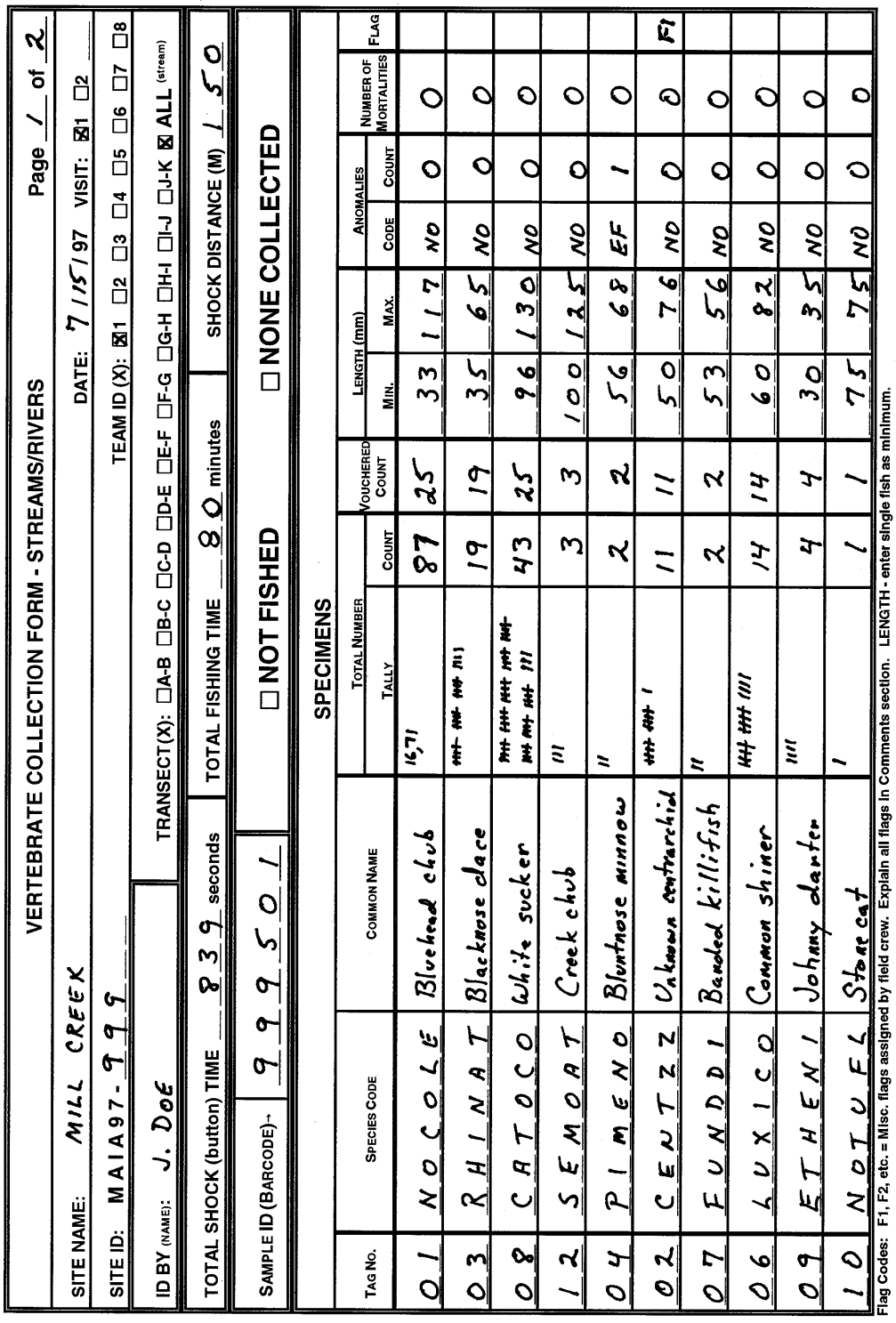
5 Beginning at the downstream end of the reach (Transect "A"), fish in an upstream direction, parallel to the current. Depress the switch and sweep the electrodes from side to side in the water. Sample available cut-bank and snag habitats as well as riffles and pools.

6. The netters follow the operator and net stunned aquatic vertebrates. Deposit individuals in buckets for processing. If necessary, use seines to block riffles, pools and snags. The operator should adjust voltage and waveform output according to sampling effectiveness and the mortality of fish specimens.

7. Continue upstream until the next transect is reached. In large or complex streams, allocate the fishing time between the two transects as calculated based on the mean transect widths (Step 1). Process fish after each transect to reduce mortality.

8. Repeat Steps 5 through 7 until Transect "K" is reached. Record the following on the Vertebrate Collection Form:
   - The reading from the electrofisher timer in the "TOTAL SHOCK TIME" field on the Vertebrate Collection Form.
   - The total distance sampled by electrofishing
   - The total fishing time, if no additional sampling is conducted (e.g., by seining) once electrofishing is completed. Total sampling time should be between 45 minutes and 3 hours.

9. If no aquatic vertebrates were collected, complete the "NONE COLLECTED" field on the Vertebrate Collection Form.



VERTEBRATE COLLECTION FORM - STREAMS/RIVERS — Page 1 of 2

SITE NAME: MILL CREEK — DATE: 7/15/97 — VISIT: ☒1 ☐2

SITE ID: MAIA97-999 — TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

ID BY (NAME): J. Doe — TRANSECT(X): ☐A-B ☐B-C ☐C-D ☐D-E ☐E-F ☐F-G ☐G-H ☐H-I ☐I-J ☐J-K ☒ ALL (stream)

TOTAL SHOCK (button) TIME 839 seconds — TOTAL FISHING TIME 80 minutes — SHOCK DISTANCE (M) 150

SAMPLE ID (BARCODE)→ 99950 1 — ☐ NOT FISHED — ☐ NONE COLLECTED

SPECIMENS

Tag No.	Species Code	Common Name	Total Number: Tally	Total Number: Count	Vouchered Count	Length (mm): Min.	Length (mm): Max.	Anomalies: Code	Anomalies: Count	Number of Mortalities	Flag
01	NOCOLE	Bluehead chub	16,71	87	25	33	117	NO	0	0	
03	RHINAT	Blacknose dace	++++ ++++ ++++ ++++ 111	19	19	35	65	NO	0	0	
08	CATOCO	White sucker	++++ ++++ ++++ ++++ ++++ ++++ ++++ 111	43	25	96	130	NO	0	0	
12	SEMOAT	Creek chub	111	3	3	100	125	NO	0	0	
04	PIMENO	Bluntnose minnow	11	2	2	56	68	EF	1	0	
02	CENTZZ	Unknown centrarchid	++++ ++++ 1	11	11	50	76	NO	0	0	F1
07	FUNDDI	Banded killifish	11	2	2	53	56	NO	0	0	
06	LUXICO	Common shiner	++++ ++++ 1111	14	14	60	82	NO	0	0	
09	ETHENI	Johnny darter	1111	4	4	30	35	NO	0	0	
10	NOTUFL	Stoat cat	1	1	1	75	75	NO	0	0	

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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VERTEBRATE COLLECTION FORM - STREAMS/RIVERS - 1

Figure 12-2. Vertebrate Collection Form (page1).

Change the water in the bucket 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 aquatic vertebrates are collected. Electrofishing may also need to cease at times to immediately process and release specimens (e.g., listed species or large game fish) as they are netted (see Section 12.2). If periodic processing is required, be sure to release individuals downstream to reduce the likelihood of collecting them again.

At the completion of electrofishing, record the total operating time (shock time) shown on the electrofisher timer and the distance sampled by electrofishing on the Vertebrate Collection Form (Figure 12-2). If sampling activities (electrofishing and seining) are completed, also record the total fishing time on the Vertebrate Collection Form. If no aquatic vertebrates were collected, indicate this on the form as shown in Figure 12-2.

12.1.2 Seining

Seining may be used in conjunction with electrofishing to ensure sampling of those species which may otherwise be underrepresented by an electrofishing survey alone (e.g., darters, sculpins, madtoms, and benthic cyprinids). Seining may also be used in sites where the stream is too deep for electrofishing to be conducted safely or in turbid, simple, soft-bottomed streams where it is more effective.

Seining procedures are presented in Table 12-2. Depending on the particular use (block netting vs. active seining) and the habitat, different sizes of seines are used. In riffle habitats, the seine is held stationary while team members disturb the substrate immediately upstream of the net. In pools, the seine is pulled back and forth across the pool, using the shore and other natural habitat breaks as barriers, or pulled rapidly downstream through the pool and then swept toward the shore. Block nets may be used in very large pools to limit escape or as seines. Large nets are typically deployed parallel to the current and swept to shore.

Proceed upstream through the reach, allocating the seining effort among habitat areas (riffles and pools) so that the entire reach is sampled within the required sampling time (45 minutes to 3 hours). Deposit aquatic vertebrates collected by seining into a bucket for later processing as described in Section 12.1. At the completion of sampling activities (electrofishing and/or seining), record the total fishing time on the Vertebrate Collection

TABLE 12-2. PROCEDURES TO COLLECT AQUATIC VERTEBRATES BY SEINING

NOTE: Seining is used in place of electrofishing where a stream is too deep for electrofishing to be conducted safely, or in turbid, simple, soft-bottomed streams where it is more effective.

1. Survey the sampling reach and set block nets at each end (Transects "A" and "K") if necessary (e.g., the majority of the reach is one large, continuous pool). Allocate the sampling effort throughout the sampling reach so that the total fishing time will be between 45 minutes (small stream) and 3 hours (large stream).

2. Complete the header information on a copy of the Vertebrate Collection Form. Indicate that all transects are being sampled in the "TRANSECT" field on the form.

   - NOTE: Make an effort to search and sample for aquatic vertebrates at all streams, even if the stream is extremely small, and it appears that sampling may not collect any specimens. If no specimens are collected, complete the "NONE COLLECTED" field on the Vertebrate Collection Form. Provide an explanation in the comments section of the form.

3. Begin at the downstream end of the sampling reach (Transect "A"). Proceed upstream, sampling available riffle and pool habitats using the appropriate method below:

   3A. Riffle habitats-- Use a small minnow seine (2 m long × 1.25 m wide; 0.6 cm mesh size).

      1. One or two persons place the seine perpendicular to the current across the downstream end of the riffle. Ensure that the lead line is on the bottom. Tilt the net slightly backward to form a pocket to trap aquatic vertebrates.
      2. Starting about 2 m upstream, the other two team members disturb the substrate in front of the net by kicking through the substrate and overturning rocks, and proceed downstream toward the nets.
      3. Raise the net and examine it carefully for aquatic vertebrates.

   3B. Pool habitats: Use a larger seine (3 m long × 2 m wide; 0.6 cm mesh size).

      1. Two people pull the seine back and forth across the pool, using the shore and other natural habitat breaks as barriers.
      2. Alternatively (in areas with some current), pull the net along in a downstream direction and then sweep toward the shore.
      3. Pull the net onto the shore and examine it carefully for aquatic vertebrates.

4. Deposit individuals in buckets for processing, and continue upstream to the next habitat area.

5. Repeat Steps 3 and 4 for successive habitat areas until Transect "K" is reached. If no aquatic vertebrates were collected by either seining or electrofishing, complete the "NONE COLLECTED" box on the Vertebrate Collection Form.

6. Record the total fishing time on the Vertebrate Collection Form. Include any time spent electrofishing in the total fishing time. Total fishing time should be between 45 minutes and 3 hours.

Form (Figure 12-2). If no aquatic vertebrates were collected, indicate this on the form as shown in Figure 12-2.

12.2 SAMPLE PROCESSING

Sample processing involves tallying and identifying fish and amphibians, examining individual specimens for external anomalies, obtaining length measurements from selected specimens, preparing voucher specimens for taxonomic confirmation and archival at a museum, and selecting specimens to prepare samples for fish tissue contaminants (see Section 13). Process collections as quickly as possible to minimize stress to live specimens. All team members can work to separate aquatic vertebrates into families or obvious "morphotypes". Alternatively, 1 or 2 persons can process fish from one bucket while the other team members continue to collect fish and deposit them into a second bucket. Once the rough sort has been completed, one person can identify, measure, and examine individuals while another person may record information on the field data forms.

12.2.1 Taxonomic Identification and Tally

Table 12-3 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 12-2. Record comments and data for additional species on page 2 of the Vertebrate Collection Form (Figure 12-3). Each team needs 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 D for an example).

Sort aquatic vertebrates by species into small buckets and containers. 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. Try to process one species completely before going on to the next. However, 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 and totaling the tally once processing is complete.

To minimize handling, process threatened and endangered species first, and immediately return all individuals to the stream. If conditions permit and stress to individuals will be minimal, photograph such fish for voucher purposes (Section 12.2.3).

TABLE 12-3. PROCEDURE TO IDENTIFY, TALLY, AND EXAMINE AQUATIC VERTEBRATES

1. Separate aquatic vertebrates retained in collecting buckets or live wells into families or obvious "morphotypes" (e.g., two dorsal fins vs. one, a sucker mouth, catfish, trout, etc.). Place each group into a separate bucket or similar container. All team members can participate in this "rough" sort. Alternatively, identify and process each individual completely, thus handling it only once.

2. Sort each group created in Step 1 by species into separate containers. This should be done only by team members who are trained ichthyologists familiar with the fish species and other aquatic vertebrate taxa of the region.

3. Select a container and record the common name (from a standardized list) and species code (if required) 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.

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

4. 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.

5. 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.

6. If the container has sport fish and other very large specimens, or if 3 or fewer species are captured at the stream, prepare a Vertebrate Length Recording Form.

    A. Complete the header information on the form, then enter the common name (from a standardized list) and the species code (if required) in the first blank line.

    B. Measure the total length of each individual (up to 30) and record the lengths in the boxes on the 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.

7. 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.

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

9. Follow the appropriate procedure to prepare voucher specimens and/or to select specimens for tissue samples. Release all remaining individuals into the stream. If there is still a portion of the sampling reach that has not been sampled, release fish downstream to avoid their recapture.

10. Repeat Steps 3 through 9 for all other species.

Reviewed by (initial): DJP

VERTEBRATE COLLECTION FORM - STREAMS/RIVERS (continued) Page 2 of 2

SITE ID: MAIA97-999 DATE: 7/15/97 VISIT: ☒1 ☐2

Sample ID (Barcode) → 999501 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

ID BY (NAME): J. Doe TRANSECT(X): ☐A-B ☐B-C ☐C-D ☐D-E ☐E-F ☐F-G ☐G-H ☐H-I ☐I-J ☐J-K ☒ALL (stream)

SPECIMENS (continued)

TAG NO.	SPECIES CODE	COMMON NAME	TOTAL NUMBER		VOUCHERED COUNT	LENGTH (mm)		ANOMALIES		NUMBER OF MORTALITIES	FLAG
			TALLY	COUNT		MIN.	MAX.	CODE	COUNT		
11	COTTZZ	Unknown cottid	III	3	3	60	80	NO	0	0	

FLAG	COMMENTS
F1	Unknown centrarchid = Lepomis sp.

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Figure 12-3. Vertebrate Collection Form (page 2).

Indicate if photographed with an “F” series flag for the species on page 1 of the Vertebrate Collection Form (Figure 12-2) and record a notation in the comments section on page 2 of the form (Figure 12-3). 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, if required) provided for unknowns from the standardized list (see Figure 12-2 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 Vertebrate Collection Form (Figure 12-3).

12.2.2 External Examination and Length Measurements

During the tallying procedure for each species (Table 12-3), 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 12-4. Record the types of anomalies observed and the number of individuals affected on the Vertebrate Collection Form as shown in Figure 12-2.

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 12-3). 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 12-2).

For each species, use a measuring board or ruler to determine the total length (Figure 12-4) of the largest and smallest individuals. Measure individuals on 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 12-2. For sport fish and other larger species, measure the total lengths of

TABLE 12-4. 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	GR	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 out-growths.
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.
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	WR	Annelid worms which have anterior and posterior suckers. They may attach anywhere on the body.
Exophthalmia	EX	Bulging of the eye.

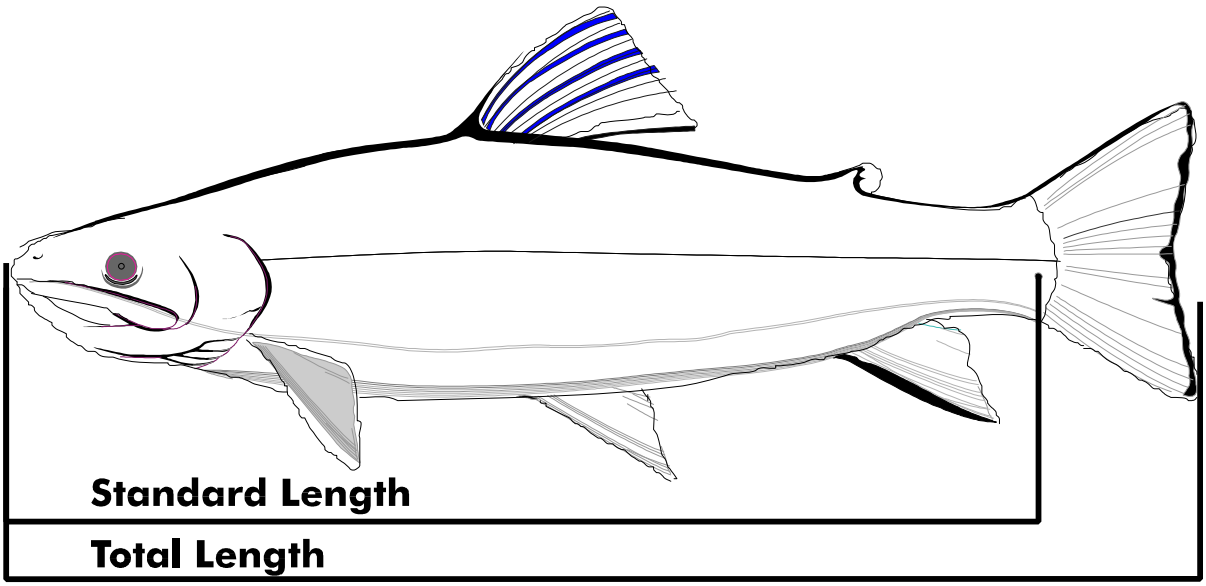


Figure 12-4. Fish length measurements.(modified from Lagler, 1956).

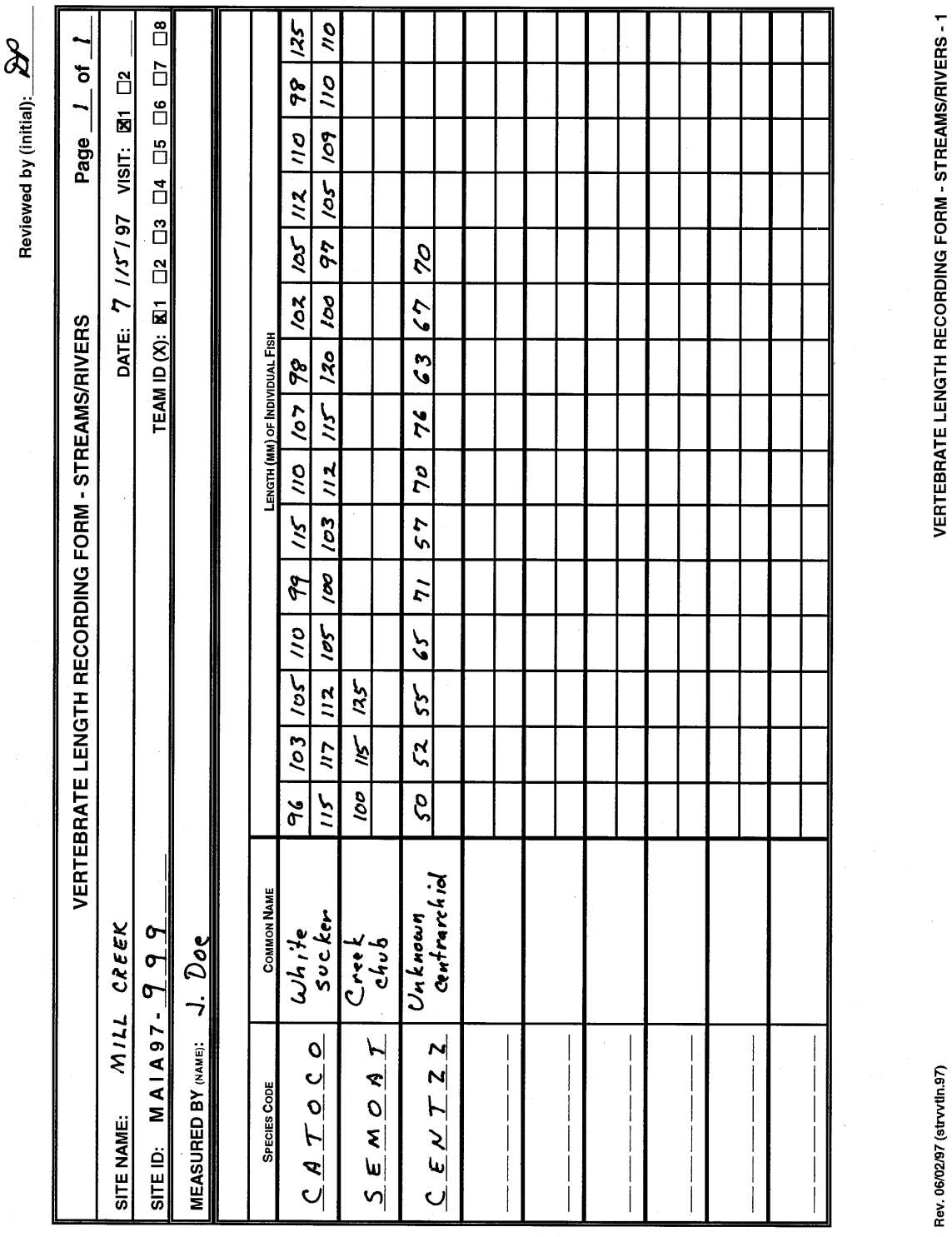
up to 30 individuals and record these values on the Vertebrate Length Recording Form as shown in Figure 12-5. If less than four species (large or small) are collected, randomly select up to 30 individuals of each species and determine the total length of each individual. Record these length measurements on the length recording form (Figure 12-5).

12.2.3 Preparing Voucher Specimens

With the exception of very large individuals of easily identified species, voucher collections of up to 25 individuals (where allowed by collecting permits) of all species are made to provide a permanent, archived, historical record of fish collections. Prepare the voucher sample for a site according to the procedure presented in Table 12-5. Retain additional specimens of the appropriate species for the fish tissue contaminants sample (Section 13). **For each species, voucher specimens take priority over specimens for the tissue contaminants sample.**

The number of voucher specimens and the method of vouchering varies with species. Large, easily identified species, larger species that are difficult to identify in the field, or species that are uncommon in the region require a few specimens of both adults and juveniles, if both were collected. Very large specimens, especially of easily identified game fish, are “vouchered” by photographing them and then releasing them alive. A larger number of voucher specimens are required for smaller species, which are typically more difficult to identify in the field. As stated previously, species of "special concern" (state and federally protected species), are processed first, vouchered by photography, and released alive. Include any individuals of protected species that die before they can be processed and released as part of the preserved voucher sample for the stream. In some cases, special restrictions may apply to protected species (e.g., sampling may have to cease upon collecting an individual). These restrictions will be stipulated on the scientific collecting permits issued by state and federal agencies.

Individuals selected as voucher specimens are first anaesthetized in a concentrated solution of carbon dioxide. Voucher specimens for each species are counted and placed into individual nylon mesh bags (1 bag per species). Nylon stockings or panty hose may be substituted in place of nylon bags. Each bag contains a numbered tag (Figure 12-6). Record the tag number and the number of individuals vouchered for each species on the Vertebrate Collection Form as shown in Figure 12-2.



VERTEBRATE LENGTH RECORDING FORM - STREAMS/RIVERS

Reviewed by (initial): DO

SITE NAME: MILL CREEK DATE: 7/15/97 VISIT: ☒1 ☐2

SITE ID: MAIA97-999 TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

MEASURED BY (NAME): J. Doe

Page 1 of 1

SPECIES CODE	COMMON NAME	LENGTH (MM) OF INDIVIDUAL FISH																	
CATOCO	White sucker	96	103	105	110	99	115	110	107	98	102	105	112	110	98	125			
		115	117	112	105	100	103	112	115	120	100	97	105	109	110	110			
SEMOAT	Creek chub	100	115	135															
CENTRZZ	Unknown centrarchid	50	52	55	65	71	57	70	76	63	67	70							

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VERTEBRATE LENGTH RECORDING FORM - STREAMS/RIVERS - 1

Figure 12-5. Vertebrate Length Recording Form (page 1).

TABLE 12-5. GUIDELINES AND PROCEDURES FOR PREPARING AQUATIC VERTEBRATE VOUCHER SPECIMENS

1. 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:

American Eel	White Sucker	Buffalo fishes	Drum
Sturgeon	Longnose Sucker	Bullhead catfish	Carp
Paddlefish	Hogsucker	Channel catfish	Salmonids
Gars	Quillback	Esocids	Crappies
Bowfin	Carpsuckers	*Morone spp*	*Micropterus spp.*
Mooneye and Goldeye	*Moxostoma spp.*	Shads	Walleye and Sauger

   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. 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 of primary and secondary target species for the tissue contaminant sample.

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

Lampreys	Troutperch	Sculpins	Madtoms
Cyprinids	Chubsucker	Sunfish	Sticklebacks
Darters	Topminnows	Silversides	Mudminnows

   1. Preserve 25 adults and juveniles. If fewer than 25 individuals are collected, voucher all of them. **Voucher samples take priority over tissue contaminant sample**.

   2. Retain additional individuals of primary and secondary target species for tissue contaminants sample.

(continued)

TABLE 12-5 (Continued)

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.

2. 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.

3. 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.

4. Immediately place the bag into a container (½ or 1 gal plastic jar) large enough to hold all voucher specimens. Add a volume of 10% formalin solution equal to the volume of fish.

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

- Add additional 10% formalin solution as bags are added so that the final volume of formalin solution is equal to the total volume of fish specimens. **Use additional jars if necessary to avoid tight packing and bending of voucher specimens**.

6. 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.

7. 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. NOTE: If more than one jar is required, use labels that have the same ID number printed on them.

8. Place the preserved sample in a suitable container with absorbent material. Store the container in a well-ventilated area during transport. Follow all rules and regulations pertaining to the transport and shipment of samples containing 10% formalin.

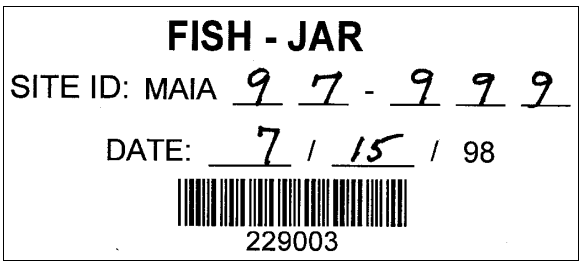

Figure 12-6. Completed voucher sample label and specimen bag tag for aquatic vertebrates.

Place specimen bags together into a large sample container. Preserve voucher specimens with a 10 percent 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. 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 result in 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 12-6. 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 12-2. Some museums may also require that a separate collection card be completed and inserted into each jar of voucher specimens.

12.3 EQUIPMENT AND SUPPLIES

Figure 12-7 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.

12.4 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.

EQUIPMENT AND SUPPLIES FOR AQUATIC VERTEBRATES

QTY.	Item	
1	Gasoline or battery-powered backpack electrofishing unit with netted anode (electrode wand)	
	Extra battery (charged) or gasoline	
3 pr	Heavy-duty rubber gloves	
3 pr	Chest waders with non-slip soles	
3 pr	Polarized sunglasses	
2	Long-handled dip nets (0.6 cm mesh) with insulated handles	
1	Watch or stopwatch to track elapsed fishing time	
	Collapsible buckets for holding and processing aquatic vertebrates	
1	Minnow seine (2m × 1.25 m, 0.6 cm mesh) with brailles	
2	Large seines (3 m × 2 m, 0.6 cm mesh) with brailles	
2	Larger sized seines for block nets (if necessary)	
1 set	Taxonomic reference books and keys for fishes and amphibians of the region	
1	Fish measuring board or ruler	
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.	½- or 1-gallon screw-top plastic jars for voucher sample	
2 gal	10% (buffered) formalin solution OR 0.2 gal buffered formaldehyde solution. Alternatively, fill each voucher sample jar one-half full of 10% formalin	
1	Container to hold formalin solution and preserved voucher sample jars	
1 pr	Safety glasses	
1 pr	Chemical-resistant gloves	

(continued)

Figure 12-7. Equipment and supplies checklist for aquatic vertebrates.

EQUIPMENT AND SUPPLIES FOR AQUATIC VERTEBRATES (Continued)

QTY.	Item	
1	Plastic bucket for anesthetization	
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	
1 + 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 12-7. (Continued).

SECTION 13
FISH TISSUE CONTAMINANTS

by
Roger B. Yeardley[1], James M. Lazorchak[2], and Frank H. McCormick [2]

In addition to gathering data on the aquatic vertebrate assemblage (Section 12), certain specimens of fish are retained for analysis of fish tissue contaminants. In general, the focus is on fish species that commonly occur throughout the region of interest, and that are sufficiently abundant within a sampling reach. Two types of composite samples of fish are prepared at each site (if possible). One composite sample is prepared using individuals of a *Primary Target Species*. Primary target species include species of fish whose adults are small (e.g., small minnows, sculpins, or darters). The second composite sample is prepared using individuals of a *Secondary Target Species*. Secondary target species include species whose adults are of larger size (e.g., suckers, bass, trout, sunfish, carp).

13.1 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. The general rule-of-thumb for "similar size" is that the smallest individual in the sample should be at least 75% of the total length of the largest individual. Keep this criterion in mind while selecting the final samples. Do not include any obviously small or large individuals if there is a sufficient sample (weight or number of individuals) without them. If there is a conflict between criteria, getting a sufficient sample is a higher priority than getting similar-sized individuals.

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1 SoBran Environmental,Inc., c/o. U.S. EPA, 26 W. Martin Luther King Dr., Cincinnati, OH 45268.

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

Prepare a composite sample of a primary target species, as described in Table 13-1. For the primary species composite sample, choose the highest priority target species 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). Do 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. If there are no primary species with enough individuals available to meet the desired weight goal, prepare the primary composite sample using individuals of a small nontarget species for which there are enough individuals available (after vouchering) to prepare a sample of at least 50 g.

Prepare a composite sample of a secondary target species as described in Table 13-2. For the secondary species composite sample, choose the highest priority target species that has the desired number (5) of similar-sized individuals (minimum total length=120 mm) available. If, for any secondary target species, you did not collect 5 individuals of the desired size, prepare the composite sample from a species having 5 individuals available (including smaller sized individuals). 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 an acceptable secondary target species sample (by the above criteria) is not available, send only the primary target species sample.

If neither a primary nor secondary species sample that meets these criteria is available, use your best judgement in preparing some type of fish tissue sample from the available species collected. Use the procedure for either primary or secondary species, 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 13-1. 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 as shown in Figure 13-2. Make sure the sample ID numbers (barcodes) recorded on the collection form match those on the sample labels.

Tissue samples are stored frozen, using either a portable freezer, a container with dry ice, or 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

TABLE 13-1. PROCEDURE TO PREPARE THE PRIMARY COMPOSITE SAMPLE FOR FISH TISSUE CONTAMINANTS

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

1. After all voucher specimens have been prepared, choose the highest priority primary target species from the list below that has at least enough individuals to attain the minimum weight (50 g). Include as many individuals as possible to attain a maximum sample weight of 400 g.

PRIMARY TARGET SPECIES (small adult fish)*

1) The most common minnow species in the region (e.g., blacknose dace)	5) Another common minnow species (e.g., stoneroller)
2) Another dace species	6) A darter species
3) Another common minnow (e.g., creek chub or fallfish)	7) A shiner species
4) The most common sculpin species in the region (e.g., Slimy sculpin or mottled sculpin)	8) If less than the desired weight of <u>any</u> primary target species is collected, send individuals of a small nontarget species if 50 g or more are available.

* The smallest individual in the sample should be at least 75% of the length of the largest individual. If there is a conflict between criteria, getting a sufficient sample is a higher priority than getting similar-sized individuals.

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. Rinse the teflon weighing beaker (to be used ONLY for weighing fish) with deionized water or stream water. Line the beaker with a sufficiently large piece of aluminum foil. Place the dull side of the foil toward the inside so it will be in contact with the fish. Place the beaker with foil on the scale and tare it.

4. If not done previously during the preparation of voucher specimens, place the individuals for the primary composite sample (Step 1) into a bucket with two carbon dioxide tablets (e.g., "Alka Seltzer®") and a small volume of water. After the individuals have been anaesthetized, use clean hands to transfer them into the beaker with foil.

5. Measure the total weight to the nearest 5g. Record the common name (from a standardized list) of the primary target species, its species code (if required), and the number of individuals in the sample in the appropriate fields on line "P1" in the primary tissue sample section of the Sample Collection Form. Enter an "F" series flag in the "Flag" field. Record the total weight of the sample in the comment/flag explanation section of the Sample Collection Form (if necessary).

(continued)

TABLE 13-1 (Continued)

6. If the individuals included in the composite sample were collected from throughout the sampling reach, place an "X" in the "YES" box in the primary 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.

7. Wrap the fish in the aluminum foil from the beaker. Make sure the dull side of the aluminum foil is in contact with the fish.

8. Place the sample in a self-sealing plastic bag. Expel excess air and seal the bag(s). Wrap clear tape around the bag to seal and make a surface for each sample label.

9. Prepare two Fish Tissue sample labels (each having the same sample ID number) 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. Attach one label to the tape surface of the bag. Cover the label with a strip of clear tape.

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

11. Place the double-bagged sample into a portable freezer, into a container with dry ice, or into a cooler containing bags of ice until shipment. Keep the sample **frozen** until shipment.

TABLE 13-2. PROCEDURE TO PREPARE THE SECONDARY COMPOSITE SAMPLE FOR FISH TISSUE CONTAMINANTS

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

1. After all voucher specimens have been prepared, select the highest priority secondary target species from the list below that has at least 5 individuals of the desired size (\$120 mm) is available. Include similar sized individuals if available.

SECONDARY TARGET SPECIES (Larger adult fish)

1) A regionally common bottom feeder (e.g., white sucker)

2) Another regionally common bottom feeder (e.g., hogsucker)

3) A regionally common piscivore (e.g., a bass species)

4) Another regionally common piscivore (e.g., a trout species)

5) Another regionally common piscivore (e.g., a sunfish species)

6) Carp

7) If fewer than 5 individuals of the desired size are collected for any target species, select a species having 5 individuals, even if some individuals are smaller than the desired size.

8) If fewer than 5 individuals of any size are available for any target species, prepare a composite sample using as few as 3 fish that are at least at or near the minimum desired size (120 mm).

9) If an acceptable secondary target species sample (by the above criteria) is not available, send only the primary target species sample.

* The smallest individual in the sample should be at least 75% of the length of the largest individual. If there is a conflict between criteria, getting a sufficient sample is a higher priority than getting similar-sized individuals.

2. Prepare a clean work surface to prepare the secondary composite sample. Keep hands, work surfaces, and wrapping materials clean and free of potential contaminants (mud, fuel, formalin, sun screen, insect repellant, etc.)

3. Measure the total length (TL) of each 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 the composite sample were collected from throughout the sampling reach, place an "X" in the "YES" box in the secondary 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. Wrap each individual 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.

(continued)

TABLE 13-2 (Continued)

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

7. Prepare two Fish Tissue sample labels (each having the same sample ID number) 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. Attach one label to the tape surface of the bag. Cover the label with a strip of clear tape.

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

9. Place the double-bagged sample into a portable freezer, a container with dry ice, or a cooler containing ice bags until shipment. Keep the sample **frozen** until shipment.

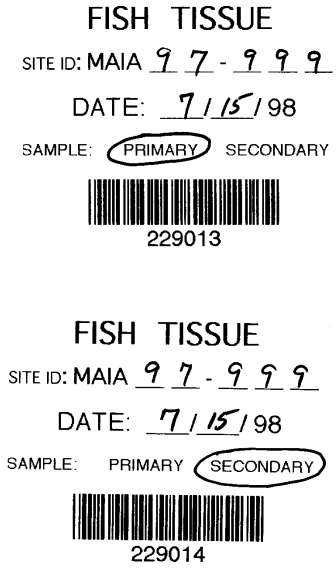

Figure 13-1. Completed sample labels for fish tissue contaminants.

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). If shipping on dry ice, special containers and shipping forms will be required.

13.2 EQUIPMENT AND SUPPLIES

Figure 13-3 is a checklist of equipment and supplies required to conduct protocols described in this section. This checklist may differ from the checklists presented in Appendix A, which are used at a base site to ensure that all equipment and supplies are brought to and are available at the stream site. Field teams are required to use the checklist presented in this section to ensure that equipment and supplies are organized and available to conduct the protocols efficiently.

Reviewed by (initial): DP

SAMPLE COLLECTION FORM - STREAMS (continued)

SITE NAME: Mill Creek DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999 ___ 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	229015	X		
MICROBIAL				

SEDIMENT TOXICITY SAMPLES

Sample ID (Barcode)	Flag	Comments
229011		

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

Sample ID (Barcode) → 229013

Line	Species Code	Common Name	Number of Individuals	Flag
P1	NOCOLE	Bluehead chub	16	F1

Is composite sample composed of individuals collected from throughout reach? (X) → ☒ Yes ☐ No

If No, Explain:

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

Sample ID (Barcode) → 229012

Line	Species Code	Common Name	Total Length (mm)	Flag
S1	CATOCO	White sucker	128	
S2	CATOCO	White sucker	125	
S3	CATOCO	White sucker	134	
S4	CATOCO	White sucker	128	
S5	CATOCO	White sucker	125	

Is composite sample composed of individuals collected from throughout reach? (X) → ☒ Yes ☐ No

If No, Explain:

Line	Comment or Flag Explanation for Fish Tissue
P1	F1 = 16 individuals weighed 60 g.

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. 06/02/97 (st_saco.97) SAMPLE COLLECTION FORM - STREAMS - 2

Figure 13-2. 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	Teflon beaker for weighing primary tissue sample	
1	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	Portable freezer, OR container with dry ice, OR 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 13-3. Equipment and supplies checklist for fish tissue contaminants.

SECTION 14
RAPID HABITAT AND VISUAL STREAM ASSESSMENTS

by
James M. Lazorchak[1], Alan T. Herlihy[2], and Jim Green[3]

After all other samples and field data have been collected, the field team conducts an overall habitat assessment of the stream, makes a general visual assessment of the stream, and performs a final check of the data forms and samples before leaving the stream site (see Section 15). The habitat assessment protocol used is adapted from EPA's "rapid" bioassessment protocols (Plafkin et al, 1989), and has been refined from various applications across the country. The approach focuses on integrating information from specific parameters on the structure of the physical habitat. The objective of the visual stream 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.

14.1 RAPID HABITAT ASSESSMENT

Based on the perception gained from collecting samples and measurements from throughout the sampling reach, classify the stream as either "Riffle/run" or "Pool/glide" prevalent based on your visual impression of the dominant habitat type. Choose the prevalent habitat type based on which habitat type occupies the majority of the length of the sampling reach. A different field data form is completed depending upon the prevalent habitat type.

For each prevalent habitat type, twelve characteristics (termed "parameters") of habitat are considered and evaluated as part of the rapid habitat assessment. These parame-

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

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

[3] U.S. EPA Region 3, Wheeling Office, 303 Methodist Bldg., 11th and Chaplin Streets, Wheeling, WV 26003.

ters are described in Table 14-1. Most of the parameters are evaluated similarly for both types of prevalent habitats. In four cases, the same parameter is evaluated differently, or a different (but ecologically equivalent) parameter is evaluated in riffle/run prevalent versus pool/glide prevalent streams. Epifaunal substrates are evaluated differently in riffle/run and pool/glide prevalent streams. Substrate embeddedness is evaluated in riffle/run prevalent streams, while pool substrate composition is evaluated in pool/glide prevalent streams. The presence of four potential types of microhabitat types based on combinations of depth and current velocity is evaluated in riffle/run prevalent streams, while the presence of four potential types of pool microhabitat based on depth and area are evaluated in pool/glide prevalent streams. The frequency of riffles is evaluated in riffle/run prevalent streams, while channel sinuosity is evaluated in pool/glide prevalent streams.

The procedure for conducting the rapid habitat assessment is presented in Table 14-2. For each of the twelve parameters, rate the overall quality of the sampling reach on a scale of 0 to 20. For riffle/run prevalent streams, record your scores for each parameter on the riffle/run version of the Rapid Habitat Assessment Form as shown in Figures 14-1 and 14-2. If the stream is classified as a pool/glide prevalent stream, record your scores for each parameter on the pool/glide version of the Rapid Habitat Assessment Form as shown in Figures 14-3 and 14-4. Transfer the scores assigned for each parameter to the box in the left-hand column of the form. Sum the scores for each parameter and record the total score in the box at the top of page 1 of the form.

14.2 VISUAL STREAM ASSESSMENT

The objective of the visual stream assessment is to record field crew observations of catchment/stream 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 stream 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

TABLE 14-1. DESCRIPTIONS OF HABITAT PARAMETERS USED IN THE RAPID ASSESSMENT OF STREAMS

Habitat Parameter	Prevalent Habitat Type R=Riffle/run P=Pool/glide	Description and Rationale
1. Instream Cover (fish)	R P	Includes the relative quantity and variety of natural structures in the stream (e.g., fallen trees, logs, and branches, large rocks, and undercut banks) that are available for refugia, feeding, or spawning. A wide variety of submerged structures in the stream provide fish with a large number of niches, thus increasing assemblage diversity.
2. Epifaunal Substrate (benthic invertebrates)	R	Essentially the amount of niche space or hard substrates (rocks, snags) available for insects and snails. Numerous types of insect larvae attach themselves to rocks, logs, branches, or other submerged substrates. As with fish, the greater the variety and number of available niches or attachments, the greater the variety of insects in the stream. Rocky-bottom areas are critical for maintaining a healthy variety of insects in most high gradient streams.
	P	The abundance, distribution, and quality of substrate and other stable colonizing surfaces (e.g., old logs, snags, aquatic vegetation) that maximize the potential for colonization.
3A. Embeddedness	R	The extent to which rocks (gravel, cobble, and boulders) are covered or sunken into the silt, sand, or mud of the stream bottom. Generally, as rocks become embedded, the surface area available to macroinvertebrates and fish for shelter, spawning, and egg incubation is decreased. To estimate the percent of embeddedness, observe the amount of silt or finer sediments overlying and surrounding the rocks. If kicking does not dislodge the rocks or cobble, they may be greatly embedded. It is useful to observe the extent of the dark area on their underside of a few rocks.
3B. Pool Substrate Characterization	P	Evaluates the type and condition of bottom substrates found in pools. Firmer sediment types (e.g., gravel, sand) and rooted aquatic plants support a wider variety of organisms than a pool substrate dominated by mud or bedrock and no plants. In addition, a stream that has a uniform substrate in its pools will support far fewer types of organisms than a stream that has a variety of substrate types.
4A. Velocity and Depth Regimes	R	There are four primary current and depth combinations: (1) slow-deep, (2) slow-shallow, (3) fast-deep, and (4) fast-shallow. The best streams in high gradient regions will have all four combinations present. The presence or availability of these four habitats relates to the ability of the stream to provide and maintain a stable aquatic environment. In general use a depth of 0.5 m to separate shallow from deep and a current velocity of 0.3 m/sec to separate fast from slow.
4B. Pool Variability	P	Rates the overall mixture of pool types found in streams, according to size and depth. The four basic types of pools are large-shallow, large-deep, small-shallow, and small-deep. A stream with many pool types will support a wide variety of aquatic species. Rivers with low sinuosity (few bends) and monotonous pool characteristics do not have sufficient quantities and types of habitat to support a diverse aquatic community. As a general guideline, consider a pool deep if it is greater than 1 m deep, and large if its length, width, or oblique dimension is greater than half the stream width.

(continued)

TABLE 14-1 (Continued)

Habitat Parameter	Prevalent Habitat Type R=Riffle/run P=Pool/glide	Description and Rationale
5. Channel Alteration	R P	Basically a measure of large-scale changes in the shape of the stream channel. Many streams in urban and agricultural areas have been straightened, deepened, or diverted into concrete channels, often for flood control purposes. Such streams have far fewer natural habitats for fish, macroinvertebrates, and plants than do naturally meandering streams. Channel alteration is present when the stream runs through a concrete channel; when artificial embankments, riprap, and other forms of artificial bank stabilization or structures are present; when the stream is very straight for significant distances; when dams and bridges are present; and when other such changes have occurred.
6. Sediment Deposition	R P	The amount of sediment that has accumulated and the changes that have occurred to the stream bottom as a result of the deposition. Deposition occurs from large-scale movement of sediment caused by watershed erosion. Sediment deposition may cause the formation of islands, point bars (areas of increased deposition usually at the beginning of meanders that increase in size as the channel is diverted toward the outer bank) or shoals or result in the filling of pools. Increased sedimentation also results in increased deposition. Usually this is evident in areas that are obstructed by natural or man-made debris and areas where the stream flow decreases, such as bends. High levels of sediment deposition create an unstable and continually changing environment that becomes unsuitable for many organisms.
7A. Frequency of Riffles	R	The sequence of riffles occurring in a stream. Riffles are a source of high-quality habitat and diverse fauna, therefore, an increased frequency of occurrence greatly enhances the diversity of the stream community. For areas where riffles are uncommon, a run/bend ratio can be used as a measure of sinuosity. A large degree of sinuosity provides for diverse habitat and fauna, and the stream is better able to handle the high energy flows that result from storms than are relatively straight streams.
7B. Channel Sinuosity	P	Evaluates the meandering or relative frequency of bends of the stream. Streams that meander provide a variety of habitats for aquatic organisms, whereas straight stream segments are characterized by monotonous habitats that are prone to flooding. A high degree of sinuosity creates a variety of pools and reduces the energy from surges when the stream flow fluctuates. The absorption of this energy by bends protects the stream from excessive erosion and flooding. In "oxbow" streams of coastal areas and deltas, meanders are highly exaggerated and transient. Natural conditions are shifting channels and bends. Alteration of these streams is usually in the form of flow regulation and diversion.
8. Channel Flow Status	R P	The degree to which the channel is filled with water. The flow status will change as the channel enlarges or as flow decreases as a result of dams and other obstructions, diversions for irrigation, or drought. When water does not cover much of the streambed, the amount of useable substrate for aquatic organisms is limited.

(continued)

TABLE 14-1 (Continued)

Habitat Parameter	Prevalent Habitat Type R=Riffle/run P=Pool/glide	Description and Rationale
9. Condition of Banks	R P	The stream banks are eroded (or have the potential for erosion). Steep banks are more likely to collapse and suffer from erosion than are gently sloping banks and are therefore considered to be unstable. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil
10. Bank Vegetative Protection	R P	The amount of the stream bank that is covered by vegetation. The root systems of plants growing on stream banks help hold soil in place, thereby reducing the amount of erosion that is likely to occur. This parameter supplies information on the ability of the bank to resist erosion, as well as some additional information on the uptake of nutrients by the plants, the control on instream scouring, and stream shading. Banks that have full, natural plant growth are better for fish and macroinvertebrates than are banks without vegetative protection or those shored up with concrete or riprap.
11. Grazing or Disruptive Pressure	R P	Disruptive changes to the riparian zone because of grazing or human interference (e.g., mowing). In areas of high grazing pressure from livestock or where residential and urban development activities disrupt the riparian zone, the growth of a natural plant community is impeded. Residential developments, urban centers, golf courses, and rangeland are the common causes of anthropogenic effects on the riparian zone.
12. Riparian Vegetated Zone Width	R P	The width of natural vegetation from the edge of the stream bank (riparian buffer zone). The riparian vegetative zone serves as a buffer zone to pollutants entering a stream from runoff, controls erosion, and provides stream habitat and nutrient input into the stream. A relatively undisturbed riparian zone reflects a healthy stream system; narrow, far less useful riparian zones occur when roads, parking lots, fields, lawns, bare soil, rocks, or buildings are near the stream bank. The presence of "old fields" (i.e., a previously developed field allowed to convert to natural conditions) will rate higher than fields in continuous or periodic use. Paths and walkways in an otherwise undisturbed riparian zone may be judged to be inconsequential to destruction of the riparian zone.

TABLE 14-2. PROCEDURE FOR CONDUCTING THE RAPID HABITAT ASSESSMENT

1. Based on observations during previous sample collection and field measurement activities, classify the sampling reach as predominantly flowing water habitat ("Riffle/run") or slow water habitat ("Pool/glide").

2. Select the appropriate version of the Rapid Habitat Assessment Form ("Riffle/Run Prevalence" or "Pool/Glide Prevalence") based on the classification in Step 1.

3. For each of the 12 habitat parameters, determine the general "quality" category ("POOR", "MARGINAL", "SUB-OPTIMAL", or "OPTIMAL") of the entire sampling reach. Assign and circle a score from the values available within each quality category. For each parameter, the sampling reach can be scored from 0 (worst) to 20 (best).

4. After the sampling reach has been scored for all parameters, transfer the score circled for each category to the corresponding "SCORE" box in the "HABITAT PARAMETER" column of the assessment form.

5. Sum the scores recorded in Step 4 over all 12 habitat parameters. Record the total score for the sampling reach in the "TOTAL SCORE" box on page 1 of the assessment form. The total score can range from 0 to 240.

Reviewed by (initial): JP

RAPID HABITAT ASSESSMENT FORM: RIFFLE/RUN PREVALENCE - STREAMS

SITE NAME: MILL CREEK DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999__ TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

HABITAT PARAMETER	CATEGORY: OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
TOTAL SCORE 162				
1. INSTREAM COVER (FISH)	Greater than 50% mix of boulder, cobble, submerged logs, undercut banks, or other stable habitat.	30 to 50% mix of boulder, cobble, or other stable habitat; adequate habitat.	10 to 30% mix of boulder, cobble, or other stable habitat; habitat availability is less than desirable.	Less than 10% of boulder, cobble, or other stable habitat; lack of habitat is obvious.
SCORE: 12	20 19 18 17 16	15 14 13 (12) 11	10 9 8 7 6	5 4 3 2 1 0
2. EPIFAUNAL SUBSTRATE	Well-developed riffle and run; riffle is as wide as stream and its length extends two times the width of stream; abundance of cobble.	Riffle is as wide as stream, but is less than two times width; abundance of cobble; boulders and gravel common.	Run area may be lacking; reduced riffle area that does not extend across entire cross section and is less than two times the width; gravel or large boulders and bedrock prevalent; cobble present.	Riffles or run virtually non-existent; gravel or large boulders and bedrock prevalent; cobble lacking.
SCORE: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
3. EMBEDDEDNESS	Gravel, cobble, and boulder particles are between 0 and 25% surrounded by fine sediment.	Gravel, cobble, and boulder particles are between 25 and 50% surrounded by fine sediment.	Gravel, cobble, and boulder particles are between 50 and 75% surrounded by fine sediment.	Gravel, cobble, and boulder particles are over 75% surrounded by fine sediment.
SCORE: 12	20 19 18 17 16	15 14 13 (12) 11	10 9 8 7 6	5 4 3 2 1 0
4. VELOCITY/DEPTH REGIMES	All four velocity regimes are present (slow-deep, slow-shallow, fast-deep, fast-shallow).	Only three of the four habitat types are present (if fast-shallow is missing, score lower than if other regimes are missing).	Only two of the four habitat types are present (if fast-shallow or slow-shallow are missing, score low).	Dominated by one velocity/depth regime (usually slow-deep).
SCORE: 15	20 19 18 17 16	(15) 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
5. CHANNEL ALTERATION	No channelization of dredging present..	Some channelization is present, usually in areas of bridge abutments; evidence of past channelization, i.e., dredging (greater than past 20 yr) may be present, but recent channelization is not present.	New embankments are present on both banks; and 40 to 80% of the stream reach is channelized and disrupted.	Banks shored with gabion or cement; over 80% of the stream reach is channelized and disrupted.
SCORE: 18	20 19 (18) 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
6. SEDIMENT DEPOSITION	Little or no enlargement of islands or point bars and less than 5% of the bottom is affected by sediment deposition.	Some new increase in bar formation, mostly from coarse gravel; 5 to 30% of the bottom is affected; slight deposition in pools.	Moderate deposition of new gravel or coarse sand on old and new bars; 30 to 50% of the bottom is affected; sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools prevalent.	Heavy deposits of fine material; increased bar development; more than 50% of the bottom is changing frequently; pools almost absent due to substantial sediment deposition.
SCORE: 14	20 19 18 17 16	15 (14) 13 12 11	10 9 8 7 6	5 4 3 2 1 0

Rev. 06/02/97 (stxxrhrr.97) RAPID HABITAT ASSESSMENT FORM: RIFFLE/RUN - STREAMS - 1

Figure 14-1. Rapid Habitat Assessment Form for riffle/run prevalent streams (page 1).

Reviewed by (initial): DP

RAPID HABITAT ASSESSMENT FORM: RIFFLE/RUN - STREAMS (continued)

SITE NAME: MILL CREEK DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999___ TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

HABITAT PARAMETER	CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
7. FREQUENCY OF RIFFLES	Occurrence of riffles is relatively frequent; the distance between riffles divided by the width of the stream equals 5 to 7; variety of habitat.	Occurrence of riffles is infrequent; distance between riffles divided by the width of the stream equals 7 to 15.	Occasional riffle or bend; bottom contours provide some habitat; distance between riffles divided by the width of the stream is between 15 to 25.	Generally all flat water or shallow riffles; poor habitat; distance between riffles divided by the width of the stream is greater than 25.
SCORE: 13	20 19 18 17 16	15 14 (13) 12 11	10 9 8 7 6	5 4 3 2 1 0
8. CHANNEL FLOW STATUS	Water reaches the base of both banks and a minimal area of channel substrate is exposed.	Water fills more than 75% of the available channel; or less than 25% of the channel substrate is exposed.	Water fill 25 to 75% of the available channel; and/or riffle substrates are mostly exposed.	Very little water in channel, and mostly present as standing pools.
SCORE: 18	20 19 (18) 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
9. CONDITION OF BANKS	Banks stable; no evidence of erosion or bank failure.	Banks moderately stable; infrequent, small areas of erosion mostly healed over.	Moderately unstable; up to 60% of banks in reach have areas of erosion.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; on side slopes, 60 to 100% of bank has erosional scars.
SCORE: 15	20 19 18 17 16	(15) 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
10. BANK VEGETATIVE PROTECTION	More than 90% of the stream bank surfaces are covered by vegetation.	70 to 90% of the stream bank surfaces are covered by vegetation.	50 to 70% of the stream bank surfaces are covered by vegetation.	Less than 50% of the stream bank surfaces are covered by vegetation.
SCORE: 16	20 19 18 17 (16)	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
11. GRAZING OR OTHER DISRUPTIVE PRESSURE	Vegetative disruption, through grazing or mowing is minimal or not evident; almost all plants are allowed to grow naturally.	Disruption is evident but is not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	Disruption is obvious; patches of bare soil or closely cropped vegetation are common; less than one-half of the potential plant stubble height remaining.	Disruption of stream bank vegetation is very high; vegetation has been removed to 2 inches or less in average stubble height.
SCORE: 11	20 19 18 17 16	15 14 13 12 (11)	10 9 8 7 6	5 4 3 2 1 0
12. RIPARIAN VEGETATION ZONE WIDTH (LEAST BUFFERED SIDE)	Width of riparian zone is greater than 18 m; human activities (i.e.; parking lots, roadbeds, clearcuts, lawns, or crops) have not impacted this zone.	Zone width is between 12 and 18 m; human activities have only minimally impacted this zone.	Zone width is between 6 and 12 m; human activities have impacted the zone a great deal.	Width of zone is less than 6 m; little or no riparian vegetation due to man-induced activities.
SCORE: 10	20 19 18 17 16	15 14 13 12 11	(10) 9 8 7 6	5 4 3 2 1 0

Figure 14-2. Rapid Habitat Assessment Form for riffle/run prevalent streams (page 2).

Reviewed by (initial): DF

RAPID HABITAT ASSESSMENT FORM: GLIDE/POOL PREVALENCE - STREAMS

SITE NAME: MILL CREEK DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999___ TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

TOTAL SCORE 118	CATEGORY			
HABITAT PARAMETER	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
1. INSTREAM COVER	Greater than 50% mix of snags, submerged logs, undercut banks, or other stable habitat; rubble or gravel may be present.	30 to 50% mix of stable habitat; adequate habitat for maintenance of populations.	10 to 30% mix of stable habitat; habitat availability is less than desirable.	Less than 10% stable habitat; lack of habitat is obvious.
SCORE: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
2. EPIFAUNAL SUBSTRATE	Preferred benthic substrate (to be sampled) is abundant throughout stream site and at a stage to allow for full colonization potential (i.e.; logs and snags that are not new fall and not transient.	Substrate is common but is not prevalent nor well-suited for full colonization potential.	Substrate frequently disturbed or removed.	Substrate is unstable or lacking.
SCORE: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
3. POOL SUBSTRATE CHARACTERIZATION	Mixture of substrate materials, with gravel and firm sand prevalent; root mats and submerged vegetation are common.	Mixture of soft sand, mud, or clay; mud may be dominant; some root mats and submerged vegetation are present	All mud or clay or sand bottom; little or no root mat; no submerged vegetation.	Hard-pan clay or bedrock; no root mat or vegetation.
SCORE: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
4. POOL VARIABILITY	Even mix of large-shallow, large-deep, small-shallow, and small-deep pools are present.	The majority of pools are large and deep; very few shallow.	Shallow pools much more prevalent than deep pools.	Majority of pools are small-shallow or pools are absent.
SCORE: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
5. CHANNEL ALTERATION	No channelization of dredging present.	Some channelization is present, usually in areas of bridge abutments; evidence of past channelization, i.e.; dredging (greater than past 20 yr) may be present, but recent channelization is not present.	New embankments are present on both banks; channelization may be extensive, usually in urban areas or drainage areas of agricultural lands; and more than 80% of the stream reach is channelized or disrupted.	Extensive channelization; banks shored with gabion or cement; heavily urbanized areas; instream habitat greatly altered or removed entirely.
SCORE: 16	20 19 18 17 (16)	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
6. SEDIMENT DEPOSITION	Less than 20% of the bottom is affected; minor accumulation of fine and coarse material at snags and submerged vegetation; little or no enlargement of islands or point bars.	20 to 50% affected; moderate accumulation; substantial sediment movement only during major storm events; some new increase in bar formation.	50 to 80% affected; major deposition; pools shallow and heavily silted; embankments may be present on both banks; frequent and substantial sediment movement during storm events.	Channelized; mud, silt, and/or sand in braided or non-braided channels; pools almost absent due to deposition.
SCORE: 7	20 19 18 17 16	15 14 13 12 11	10 9 8 (7) 6	5 4 3 2 1 0

Rev. 06/02/97 (stxxrhgp.97) RAPID HABITAT ASSESSMENT FORM: GLIDE/POOL - STREAMS - 1

Figure 14-3. Rapid Habitat Assessment Form for pool/glide prevalent streams (page 1).

Reviewed by (initial): DP

RAPID HABITAT ASSESSMENT FORM: GLIDE/POOL- STREAMS (continued)

SITE NAME: MILL CREEK DATE: 7 / 15 / 97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999 ___ TEAM ID (X): ☒1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

HABITAT PARAMETER	CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
7. CHANNEL SINUOSITY	The bends in the stream increase the stream length 3 to 4 times longer than if it was in a straight line.	The bends in the stream increase the stream length 2 to 3 times longer than if it was in a straight line.	The bends in the stream increase the stream length between 1 and 2 times longer than if it was in a straight line.	Channel is straight; waterway has been channelized for a long distance.
SCORE: 13	20 19 18 17 16	15 14 (13) 12 11	10 9 8 7 6	5 4 3 2 1 0
8. CHANNEL FLOW STATUS	Water reaches the base of both lower banks and a minimal amount of channel substrate is exposed.	Water fills more than 75% of the available channel; or less than 25% of the channel substrate is exposed.	Water fills 25 to 75% of the available channel and/or riffle substrates are mostly exposed.	Very little water in channel, and mostly present as standing pools.
SCORE: 18	20 19 (18) 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
9. CONDITION OF BANKS	Banks stable; no evidence of erosion or bank failure.	Banks moderately stable; infrequent, small areas of erosion mostly healed over.	Moderately unstable; up to 60% of banks in reach have areas of erosion.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; side slopes 60 to 100% of bank has erosional scars.
SCORE: 9	20 19 18 17 16	15 14 13 12 11	10 (9) 8 7 6	5 4 3 2 1 0
10. BANK VEGETATIVE PROTECTION	Over 90% of the stream bank surfaces is covered by vegetation.	70 to 90% of the stream bank surfaces is covered by vegetation.	50 to 70% of the stream bank surfaces is covered by vegetation.	Less than 50% of the stream bank surfaces are covered by vegetation.
SCORE: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
11. GRAZING OR OTHER DISRUPTIVE PRESSURE	Vegetative disruption minimal or not evident; almost all plants are allowed to grow naturally.	Disruption is evident but is not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	Disruption is obvious; patches of bare soil or closely cropped vegetation are common; less than one-half of the potential plant stubble height remaining.	Disruption of stream bank vegetation is very high; vegetation has been removed to 2 inches or less in average stubble height.
SCORE: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
12. RIPARIAN VEGETATIOUN ZONE WIDTH (LEAST BUFFERED SIDE)	Width of riparian zone is greater than 18 meters; human activities (i.e.; parking lots, roadbeds, clearcuts, lawns, or crops) have not impacted this zone.	Width of riparian zone is between 12 and 18 meters; human activities have only minimally impacted this zone.	Width of riparian zone is between 6 and 12 meters; human activities have impacted the zone a great deal.	Width of riparian zone is less than 6 meters; little or no riparian vegetation due to human activities.
SCORE: 7	20 19 18 17 16	15 14 13 12 11	10 9 8 (7) 6	5 4 3 2 1 0

Figure 14-4. Rapid Habitat Assessment Form for glide/pool prevalent streams (page 2).

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 14-3. Record data and observations for each component of the assessment on the Assessment Form as shown in Figures 14-5 and 14-6.

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 stream, the rating for “Houses” would be low. If the stream is in a suburban housing development, rate it as high. Similarly, a small patch of clear cut logging on a hill overlooking the stream would be rated as low. Logging activity right on the stream shore, however, would be rated as high.

When assessing reach characteristics, make your best estimate as to the percent of the sampling reach (40 channel widths) that had each type of listed riparian zone land use immediately adjacent to the stream. 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 stream a "5" rating if it is pristine, with no signs of any human development. A rating of "1" indicates a stream which is totally developed (e.g., the entire stream is lined with houses, or the riparian zone has been removed). For aesthetics, base your decision on any factor about the stream that bothers you (e.g., trash, algal growth, weed abundance, overcrowding).

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 14-6) is available for general comments.

TABLE 14-3. PROCEDURE FOR CONDUCTING THE FINAL VISUAL ASSESSMENT OF A STREAM

1. After all other sampling and measurement activities are completed, fill out the header section of an Assessment Form. Use your perceptions obtained during the course of the day, while at the stream or driving/walking through the catchment to complete the remainder of the form.

2. WATERSHED ACTIVITIES AND DISTURBANCES OBSERVED: Rate each type of activity or disturbance listed on the form as either "Not observed", "Low", "Medium", or "High", and record the rating on the Assessment Form. Keep in mind that ratings will be somewhat subjective and that an extensive effort to quantify the presence and intensity of each type of stressor is not required. General categories of activities and types of disturbance are described below:

   - Residential: The presence of any of the listed disturbances adjacent to or near the stream.
   - Recreational: The presence of organized public or private parks, campgrounds, beaches or other recreation areas around the stream. If there are signs of informal areas of camping, swimming or boating around the stream (e.g., swimming hole), record them as "primitive" parks, camping.
   - Agriculture: The presence of cropland, pasture, orchards, poultry, and/or livestock.
   - Industrial: Any industrial activity (e.g., canning, chemical, pulp), commercial activity (stores, businesses) or logging/mining activities around the stream or in the catchment. Describe in more detail in the comments section.
   - Management: Any evidence of 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 stream 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.

(continued)

TABLE 14-3 (Continued)

5. WATERBODY CHARACTER (CONT.): Assign a rating to the stream based on overall aesthetic quality, based on your opinion of how suitable the stream water is for recreation and aesthetic enjoyment today. Place and "X" in the box next to the assigned rating on the Assessment Form.

    5. Beautiful, could not be any nicer.
    4. Very minor aesthetic problems; excellent for swimming, boating, enjoyment.
    3. Enjoyment impaired.
    2. Level of enjoyment substantially reduced.
    1. Enjoyment nearly impossible.

    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 stream provided by local residents.

Reviewed by (initial): DP

ASSESSMENT FORM - STREAMS/RIVERS

SITE NAME: MILL CREEK DATE: 7/15/97 VISIT: ☒1 ☐2 ___

SITE ID: MAIA97-999 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		L	M	H		L	M	H		L	M	H		L	M	H	
X			RESIDENCES				PARKS, CAMPGROUNDS				CROPLAND				INDUSTRIAL PLANTS				LIMING
X			MAINTAINED LAWNS				PRIMITIVE PARKS, CAMPING			X	PASTURE				MINES/QUARRIES				DRINKING WATER TREATMENT
			CONSTRUCTION				TRASH/LITTER		X		LIVESTOCK USE				OIL/GAS WELLS				ANGLING PRESSURE
			PIPES, DRAINS				SURFACE FILMS, SCUMS, OR SLICKS				ORCHARDS				POWER PLANTS				DREDGING
			DUMPING								POULTRY				LOGGING				CHANNELIZATION
X			ROADS								IRRIGATION PUMPS				EVIDENCE OF FIRE				WATER LEVEL FLUCTUATIONS
			BRIDGE/CULVERTS												ODORS				FISH STOCKING
															COMMERCIAL				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	☐ 5	☐ 4	☐ 3	☒ 2	☐ 1	HIGHLY DISTURBED
APPEALING	☐ 5	☐ 4	☐ 3	☐ 2	☒ 1	UNAPPEALING

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

Low vegetation diversity. Lots of cattle. Forest age class = 25-75 yr.

LOCAL ANECDOTAL INFORMATION:

Lots of poultry farms in area, also.

Rev. 06/02/97 (strvasse.97) ASSESSMENT FORM - STREAMS/RIVERS - 1

Figure 14-5. Assessment Form (page 1).

Reviewed by (initial): _____

ASSESSMENT FORM - STREAMS/RIVERS (continued)

SITE NAME: DATE: / / 97 VISIT: ☐1 ☐2 ___

SITE ID: M A I A 9 7 - __ __ __ __ __ TEAM ID (X): ☐1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

Rev. 06/02/97 (strvasse.97) ASSESSMENT FORM - STREAMS/RIVERS - 2

Figure 14-6. Assessment Form (page 2).

14.3 EQUIPMENT AND SUPPLIES

Figure 14-7 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 stream site. Field teams are required to use the checklist presented in this section to ensure that equipment and supplies are organized and available to conduct the protocols efficiently.

14.4 LITERATURE CITED

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

EQUIPMENT AND SUPPLIES FOR RAPID HABITAT AND VISUAL STREAM ASSESSMENTS

QTY.	Item	
1	Rapid Habitat Assessment Form for Riffle/run prevalent streams	
1	Rapid Habitat Assessment Form for Pool/glide prevalent streams	
1	Assessment Form for visual stream assessment	
6	Soft (#2) lead pencils	
1	Covered clipboard or forms holder	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for rapid habitat and visual assessments	

Figure 14-7. Checklist of equipment and supplies required for rapid habitat and visual stream assessments.

SECTION 15
FINAL SITE ACTIVITIES

by
James M. Lazorchak[1]

Before leaving a stream site, the team leader reviews all of the data forms and sample labels for accuracy, completeness, and legibility. A second team member inspects all sample containers and packages them in preparation for transport, storage, or shipment. Refer to Section 3 for details on preparing and shipping samples.

When reviewing field data forms, ensure that all required data forms for the stream have been completed. Confirm that the stream identification code, the year, the visit number, and the date of the visit are correct on all forms. On each form, verify that all information has been recorded accurately, the recorded information is legible, and any flags are explained in the comments section. Ensure that written comments are legible and use no "shorthand" or abbreviations. Make sure the header information is completed on all pages of each form. After reviewing each form, initial the upper right corner of each page of the form.

When inspecting samples, ensure that each sample is labeled, all labels are completely filled in and legible, and each label is covered with clear plastic tape. Compare sample label information with the information recorded on the corresponding field data forms (e.g., the Sample Collection Form) to ensure accuracy.

The other team members should return all of the equipment and supplies to the vehicle for transport and clean up the stream site. Pack all equipment and supplies in the vehicle for transport. Keep them organized so they can be inventoried using the equipment and supply checklists presented in Appendix A. Clean up and dispose of all waste material at the stream site. Transport it out of the area if necessary.

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

APPENDIX A
EQUIPMENT AND SUPPLY CHECKLISTS

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 Cross-section and Thalweg Profile Forms	
1	Slope and Bearing Form	
1 + extras	Vertebrate Collection Form	
1 + extras	Vertebrate Length Recording Form	
1	Rapid Habitat Assessment Form for Riffle/run prevalent streams	
1	Rapid Habitat Assessment Form for Pool/glide prevalent streams	
1	Assessment Form for visual stream assessment	
3	Water chemistry labels (same ID number)	
4	Periphyton labels (same ID number)	
5	Sediment metabolism labels (different ID numbers)	
2	Sediment toxicity labels	
2	Composite Benthic sample labels, with preprinted ID numbers (barcodes)	
4	Composite Benthic sample labels without preprinted ID numbers	
6	Blank benthic sample labels on waterproof paper for inside of jars	
1	Sheet of pre-printed 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 information for scheduled stream site	
1	Topographic map with “X-site” marked	
1	Site information sheet with map coordinates and elevation of X-site	
1	Sampling itinerary form or notebook	
1	Safety log and/or personal safety information for each team member	
4	Covered clipboards or forms holders	
1	Field notebook (optional)	
12	Soft (#2) lead pencils	
6	Fine-tip indelible markers	
2	Grease pencils	
1 pr	Scissors for cutting labels	
1	Pocket knife or multipurpose tool	
1	Battery charger (if needed for electrofishing unit)	
1	Toolbox with basic tools needed to maintain/repair sampling gear	

PERSONAL EQUIPMENT AND SUPPLIES

Number per site	Item	
1 pair per person	Chest waders with felt-soled boots for safety and speed if waders are the neoprene "stocking" type. Hip waders can be used in shallower streams (except for electrofishing).	
1 per person	Life vests	
3 pair	Polarized sunglasses	
1	First aid kit	
1 per person	Rain gear	
1 or 2	Fisherman’s vest for physical habitat characterization.	

CHEMICALS

Number per site	Item	
1 pr	Safety glasses	
2 pr	Chemical-resistant gloves	
1	Laboratory apron, resistant to ethanol and formalin	
1	Cooler (with suitable absorbent material) for transporting ethanol and samples	
2 gal	95% ethanol	
1	Cooler (with suitable absorbent material) for transporting formaldehyde/formalin	
2 gal	10% (buffered) formalin solution OR 0.2 gal buffered formaldehyde solution	
	Gasoline for electrofishing unit in approved container	

PACKING AND SHIPPING SUPPLIES

Number per site	Item	
	Ice (also dry ice if it is used to ship frozen samples)	
1 box	1-gal heavy-duty self-sealing (e.g., with a zipper-type closure) plastic bags	
1-box	30-gal plastic garbage bags	
1 box	Heavy-duty plastic bags for sediment toxicity samples	
1 roll	Clear tape for sealing tissue sample bags	
2 pkg.	Clear tape strips for covering labels	
4 rolls	Plastic electrical tape	
3	Insulated shipping containers for samples	
1	Portable freezer, cooler with dry ice, or cooler with bags of ice to store frozen samples (special containers may be needed if dry ice is used)	
2	Containers and absorbent material suitable to transport and/or ship samples preserved in formalin or ethanol	
6	Shipping airbills and adhesive plastic sleeves	

SITE VERIFICATION AND SAMPLING REACH LAYOUT

Number per site	Item	
1	GPS receiver and operating manual	
	Extra batteries for GPS receiver	
1	Surveyor's telescoping leveling rod (round profile, metric scale, 7.5 m extended)	
1	50-m fiberglass measuring tape with reel	
2 rolls	Surveyor's flagging tape (2 colors)	
1	Waterproof camera and film	

WATER CHEMISTRY

Number per site	Item	
1	Dissolved oxygen/Temperature meter with probe and operating manual	
1	DO repair kit containing additional membranes and probe filling solution	
1	Conductivity meter with probe and operating manual	
	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	Plastic container with snap-on lid to hold filled syringes	
2	Syringe valves	

STREAM DISCHARGE

Number per site	Item	
1	Current velocity meter and probe, with operating manual (e.g. Marsh-McBirney Model 201, Swoffer Model 2100, or equivalent)	
1	Top-set wading rod (metric scale) for use with current velocity meter	
1	Portable Weir with 60/ “V” notch (optional)	
1	Plastic sheeting to use with weir	
1	Plastic bucket (or similar container) with volume graduations	
1	Stopwatch	
1	Neutrally buoyant object (e.g., orange, small rubber ball, stick)	

PHYSICAL HABITAT

Number per site	Item	
1	Fisherman’s vest with lots of pockets and snap fittings.	
1	Hip chain (metric) for measuring reach lengths (Optional)	
1	Clinometer (or Abney level) with percent and degree scales.	
1	Lightweight telescoping camera tripod, (necessary only if slope measurements are being determined by only one person	
2	½-inch diameter PVC pipe, 2-3 m long, each marked at the same height (for use in slope determinations involving two persons)	
1	Spherical convex canopy densiometer, modified with taped “V”	
1	Bearing compass (Backpacking type)	
1	Meter stick. Alternatively, a short (1-2 m) rod or pole (e.g., a ski pole) with cm markings for thalweg measurements	

PERIPHYTON

Number per site	Item	
1	Large funnel (15-20 cm diameter)	
1	12-cm^2 area delimiter (3.8 cm diameter pipe, 3 cm tall)	
1	Stiff-bristle Toothbrush with handle bent at 90^o angle	
1	1-L wash bottle for stream water	
1	1-L wash bottle containing deionized water	
2	500-mL plastic bottles for composite index samples, labeled "EROSIONAL" and "DEPOSITIONAL"	
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 sample	
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	Pre-leached, pre-ashed, weighed glass-fiber filters in numbered containers for biomass sample	
2	Aluminum foil squares (3" x 6")	
1	Small syringe or bulb pipette for dispensing formalin	

SEDIMENT METABOLISM AND SEDIMENT TOXICITY

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, use a 250-mL jar is sufficient. If metabolism and toxicity samples are being prepared, use a 1-gallon jar	
1	YSI Model 58 Dissolved Oxygen meter with Model 5730 Stirring BOD probe and operating manual	
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	
1	Plastic container with snap-on lid to hold the sediment toxicity sample	

BENTHIC MACROINVERTEBRATES

Number per site	Item	
1	Modified kick net (closed bag with 595/600 :m mesh) and 4-ft handle (Wildco #425-C50)	
	Spare net(s) for the kick net sampler or extra sampler	
2	Buckets, plastic, 8- to 10-qt capacity, labeled “RIFFLE” and “POOL”	
1	Sieve, U.S. Standard #30 mesh	
1	Sieve bucket, 595- :m mesh openings	
2 pr.	Watchmakers’ forceps	
1	Small spatula, spoon, or scoop to transfer sample	
1	Funnel, with large bore spout	
4 to 6 ea.	Sample jars, plastic with screw caps, ½ and 1 gallon capacity, suitable for use with ethanol	
1	Field checklist sheet	
1 pkg.	Kim wipes in small self-sealing plastic bag	

AQUATIC VERTEBRATES AND FISH TISSUE CONTAMINANTS

Number per site	Item	
1	Gasoline or battery-powered backpack electrofishing unit with electrode wand	
	Extra battery	
3 pr	heavy-duty rubber gloves for electrofishing	
2	Long-handled dip nets (0.6 cm mesh) with insulated handles	
1	Minnow seine (2m × 1.25 m, 0.6 cm mesh) with brailles	
2	Large seines (3 m × 2 m, 0.6 cm mesh) with brailles	
2	larger sized seines for block nets (if necessary)	
10	Collapsible buckets for holding and processing aquatic vertebrates	
1 set	Taxonomic reference books and keys for fishes and amphibians of the region	
1	Fish measuring board	
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 specimens for preservation	
2 ea.	½ or 1-gal screw-top plastic jars for voucher samples	
1	Plastic bucket for anesthetization	
4	carbon dioxide tablets (Alka-Seltzer® or equivalent)	
1	Teflon beaker for weighing primary tissue sample	
1	Portable scale, precision ±5g	
1 roll	Aluminum foil	
2 gal	10% (buffered) formalin solution OR 0.2 gal buffered formaldehyde solution	
1	Container to hold preserved voucher sample jars	

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 stream 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

1. Find the stream 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, 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 Stream Marsh/Wetland Unwadeable Stream (> 50% of reach is unwadeable)
TARGET	Regular Stream Intermittent Stream Dry Channel Altered Channel (stream 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. Measure the stream width at five "typical" places within 10 m of the X-site. Average and round the width to the nearest meter. Record width on the stream site verification form. Lay out a sample reach with a length of 40 times the stream width. If the stream is less than 4 m wide, use 150 m as the sample reach length.

4. Do a reconnaissance of the sample reach.

5. Proceed downstream half the required reach length; measure the distance with a tape measure down the middle of the stream. Mark it as the reach start point (Transect “A”).

6. Proceed upstream marking 10 more cross-section transects (Transects “B” through “K”) at 1/10 intervals along the calculated reach length (every 4 channel widths or 1.5 meters in small streams). At Transect “B”, assign a sampling point (Left, Center, or Right as you face downstream) for collecting periphyton and benthic macroinvertebrate samples by throwing a die. Once the initial point has been determined, assign sampling points for Transects “C” through “J” systematically using the order Left, Center, and Right.

NOTE: If there is a lake/pond or a stream order change (100,000 map-based) along the survey reach, end the sample reach at the barrier. Make up for the loss of stream length by adding length to the other end of the reach ("slide" the reach). Locations where the stream order changes will be noted on the topo maps provided to the field teams. Do not "slide" the reach to avoid bridges, riprap, small flow control structures, culverts and the like.

QUICK REFERENCE GUIDE FOR WATER CHEMISTRY

I. EQUIPMENT TO CARRY IN FIELD FOR WATER CHEMISTRY

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)

Two syringe valves in the plastic container

Opaque garbage bag

II. EXTRA EQUIPMENT TO CARRY IN VEHICLE

Cooler with 4 to 6 one-gallon self-sealing plastic bags filled with ice
Back-up labels, forms, syringes, and syringe valves

III. DAILY ACTIVITIES AFTER SAMPLING

1. Check that cubitainer lid is on tight and has a flush seal.

2. Prepare the sample for shipping (label and seal cooler, replace ice as close as possible to shipping time).

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 and syringes), pack cubitainer and sample beakers in clean self-sealing plastic bag, and pack two syringes and syringe valves in a plastic container with a snap-on lid.

(continued)

QUICK REFERENCE GUIDE FOR WATER CHEMISTRY (Continued)

.SUMMARY OF SITE PROCEDURE FOR WATER CHEMISTRY

I. COLLECT WATER SAMPLE

A. Make sure cubitainers and syringes are labeled and have the same barcode ID.

B. Rinse the 500-mL sample beaker three times with streamwater from mainstream.

C. Rinse cubitainer three times with 25-50 mL of streamwater, using the sample beaker. Rinse cubitainer lid with stream water.

D. Fill cubitainer with streamwater 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.

E. Rinse each of the two, 60-mL syringes three times with 10-20 mL of streamwater.

F. Fill each of the syringes with streamwater from mid-stream by slowly pulling out the plunger. If any air gets into the syringe, discard the sample and draw another.

G. 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 stream sample in the syringe. Close the valve and place the syringes in their transport container.

H. Place the cubitainer and syringes in cooler/stream to keep cool (keep dark as well) while the rest of the sampling is taking place. When you return to the vehicle, put the samples in the cooler and surround with 4 to 6 one-gallon self-sealing plastic bags filled with ice.

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 stream conductivity in mid-stream.

B. Dissolved Oxygen/Temperature

1. Calibrate the DO meter following meter instructions.
2. Measure the DO and temperature in mid-stream. If water velocity is slow, jiggle the DO probe as you take the reading.

QUICK REFERENCE GUIDE FOR PHYSICAL HABITAT CHARACTERIZATION

FIELD SUMMARY: P-HAB LAYOUT AND WORKFLOW

1. Habitat Sampling Layout:

Thalweg interval: 1.0 m for streams <2.5 m wide (from initial estimate).
1.5 m for streams 2.5 - 3.5 m wide
0.01 × (reach length) for streams >3.5 m wide

100 thalweg measurement intervals in each sample reach, except 150 in streams <2.5m wide

Channel/Riparian Cross Section Transect every 10th thalweg interval (every 15th for channels <2.5m wide). Eleven of them, marked "A" thru "K".

Wetted Width at every cross-section transect and halfway in between transects (total of 21 measurements).

2. Work Flow:

- At the downstream start point (Transect "A"), one person makes channel dimension, substrate, bank, and canopy densiometer measurements. The second person records those measurements while making visual estimates of riparian vegetation structure, fish cover, and human disturbance. No bearing or slope at first cross section.
- Proceed upstream between Transects “A” and “B”, making measures at each thalweg measurement station. One person in channel measures width (when required), thalweg depth, and determines presence of soft/small sediment at thalweg. The other person records those measurements, classifies channel habitat, and makes large woody debris estimates.
- When you complete 10 thalweg intervals and reach one of 11 pre-marked cross section transect flags, stop and take out a new cross-section form for Transect "B". Repeat all the Channel/Riparian measurements at this new location. In addition, do the slope & bearing backsites together. Intermediate flagging (of a different color) may have to be used if the stream is extremely brushy, sinuous, or steep to the point that you cannot site for slope and bearing measures between the 11 points. (Note that you could tally woody debris while doing the backsite, rather than during the thalweg profile measurements.)
- Repeat the cycle of thalweg and cross section measurements until you reach transect 11 ("K") at the upstream end.
- Discharge measurements made any time after choosing suitable location nearest to the "X" site. Discharge measurements are done by the Chemistry/Macroinvert pair (rather than the Habitat/Fish pair) just after chemical samples are taken.

(continued)

QUICK REFERENCE GUIDE FOR PHYSICAL HABITAT CHARACTERIZATION (Continued)

FIELD SUMMARY: COMPONENTS OF P-HAB PROTOCOL

Width, Depth Profile, Hab Classes, Woody Debris:

- At 10 (15) equally spaced intervals between each of 11 channel cross-sections (100 or 150 along entire reach):
  - Measure max. depth ("Thalweg") at each increment and wetted width at the required increments.
  - Classify habitat and pool-forming elements.
  - Determine presence of soft/small sediment at thalweg measurement points.
- Between each of the channel cross sections, tally all Large Woody debris within and above the bankfull channel according to size class. In the tally boxes provided on the form, make separate tally for LWD wholly or partially within the bankfull channel and then for LWD only bridging above the channel.

NOTE: If initial width estimate is <2.5 m, then 150 thalweg measurements are made at 1.0 m intervals over a 150 m reach. If width is 2.5 to 3.5 m, then make 100 thalweg measurements at 1.5 m intervals. In all other cases, 100 measurements are made at an interval 1/100th the length of the sample reach.

Channel and Riparian Cross-Sections:

- Measurements: Bankfull width, bankfull height, incision height, wetted width, bar width, undercut, bank angle (with rod and clinometer); gradient (clinometer), sinuosity (compass backsite), riparian canopy cover (densiometer).
- Visual Estimates: Substrate size class and embeddedness; 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

Discharge:

In medium and large streams measure water depth and velocity (at 0.6 depth from surface) at 15 to 20 equally spaced intervals across one carefully chosen channel cross-section. Let meter equilibrate to average velocity for 20 seconds. In very small streams, measure discharge by timing the passage of a neutrally-buoyant object 3 times or the filling of a bucket 5 times in succession.

(continued)

QUICK REFERENCE GUIDE FOR PHYSICAL HABITAT CHARACTERIZATION (Continued)

FIELD SUMMARY: RIP. VEG., HUMAN DISTURB., IN-CHANNEL COVER:

- Observations upstream 5 meters and downstream 5 meters from each of the 11 cross-section transects.

- For riparian vegetation and human disturbances, include the visible area from the stream 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: The human activity or structure is ON THE STREAMBANK
  C: CLOSE to the Bank (within 10m)
  P: PRESENT, but farther than 10m from the bank
  0: NOT PRESENT.

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QUICK REFERENCE GUIDE FOR PHYSICAL HABITAT CHARACTERIZATION (Continued)

FIELD SUMMARIES: SUBSTRATE AND WOODY DEBRIS SIZE CLASSES

Substrate size class and embeddedness are estimated, and depth is measured for 5 particles taken @ 5 equally-spaced points on each cross-section. The cross-section is defined by laying the surveyor's rod or tape to span the wetted channel.

SUBSTRATE SIZE CLASSES:

RS	Bedrock (Smooth)	>4000 mm	smooth surface rock or hardpan (bigger than a car)
RR	Bedrock (Rough)	>4000 mm	(bigger than a car)
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)
HP	Hardpan	>4000 mm	(consists of firm, consolidated fines)
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) ≥ 0.1 m (≥4 in)
Length ≥ 1.5 m (≥ 5 ft) -- count only part with diam ≥ 0.1m.

Two Tallys:

(1) LWD at least partially within bankfull channel.
(2) LWD not within bankfull channel, but at least partially bridging above bankfull stage (idea is that it will eventually fall into channel).

Size Categories for Tally (12 potential combinations):

Diameter (large end):		Length:	
0.1 to <0.3 m	(4 to 12 inches)	1.5 - <5 m	(5 - 16 ft)
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 m	(>49 ft)
>0.8 m	(>2.6 ft)		

(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: Still water, low velocity, smooth, glassy surface, usually deep compared to other parts of the channel:	
Plunge Pool (PP)	Pool at base of plunging cascade or falls.
Trench Pool (PT)	Pool like trench in stream center
Lateral Scour Pool (PL)	Pool scoured along one bank.
Backwater Pool (PB)	Pool separated from main flow off side of channel.
Impoundment Pool (PD)	Pool formed by impoundment above dam or constriction.
Pool (P)	Pool (unspecified type).
Glide (GL)	Water moving slowly, with 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 water surface broken in short irregular plunges, mostly whitewater. Sound: roaring.
Falls (FA)	Free falling water over vertical or near vertical drop into plunge, water turbulent and white over high falls. Sound: from splash to roar.
Dry Channel (DR)	No water in channel

Categories of Pool-forming Elements

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

(continued)

QUICK REFERENCE GUIDE FOR PHYSICAL HABITAT CHARACTERIZATION (Continued)

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 thalweg profile and cross-section 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; then if required, measure the side channel width separately (record on another form). Handle the side channels created by islands as follows:

* Visually estimate the percent of flow in the side channel.

* If <15% -- Indicate presence of side channel on field data form.

* If 16-49% -- Indicate presence of side channel, plus obtain and record detailed channel & riparian cross-section measurements on the side channel. Designate additional cross section transects as "XA", "XB", etc. corresponding to nearest main channel Transect location.

Note continuous presence of side channels on the Thalweg Profile Form until channels converge. In addition, note the points of side channel convergence and divergence in the comment section on the thalweg profile form.

Dry and Intermittent Streams, where no water is in the channel:

- Record zeros for depth and wetted width.
- Record habitat type as dry channel ("DR").
- Make all Channel Cross-section transect measures across the unvegetated portion of the channel. For substrate, DISTLB = the width of the unvegetated channel, and substrate measurements are made along that transect.

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^{o} 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.
10. Pre-leached, pre-ashed, weighed glass-fiber filters (0.45 :m average pore size) in numbered pans for ash free dry mass (AFDM).
11. Forceps for filter handling.
12. Millipore®-type filtration apparatus with plastic or stainless steel filter base, and Nalgene® funnel and suction flask.
13. Nalgene® hand-operated vacuum pump (need one additional pump as a backup).
14. Aluminum foil.
15. Ice chest.

FIELD PROTOCOLS

1. Periphyton samples will be collected using a random-systematic procedure. The location (left, middle, or right 1/3 of the channel) of the first sample (Transect B) will be chosen randomly. Subsequent samples (Transects C-J) will be collected sequentially from the left, middle, then right 1/3 of the channel, resulting in three samples from each side and middle.

2. Periphyton are collected, using the appropriate method, from flowing (riffles) and slack water (pools) habitats.

3. Rock and wood samples which are small enough (< 15 cm diameter) and can be easily removed from the stream are collected by placing the substrate in a funnel which drains into a sample bottle. A defined area of substrate surface (12 cm^2)is enclosed, and attached periphyton is dislodged with 30 seconds of brushing with a stiff-bristled toothbrush. **Care must be taken to ensure that the upper surface of the rock is the surface that is being scraped.**

4. Loosened periphyton is then washed, using stream water from a wash bottle, from the substrate into the 500-mL sample bottle.

5. Soft-sediments are collected by vacuuming the upper 1 cm of sediments confined within the 12-cm^2 sampling ring into a 60-mL syringe.

6. All samples, regardless of substrate type, are composited by habitat (riffle or pool) and mixed thoroughly.
7. **Record total volume of composited sample before proceeding to the next step!**

(continued)

QUICK REFERENCE GUIDE FOR PERIPHYTON (Continued)

8. **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.

c. Ash Free Dry Mass (AFDM)

1) Withdraw 25 mL of mixed sample and filter onto a pre-leached, pre-weighed glass-fiber filter. (Note: for soft-sediment samples, allow grit to settle before withdrawing sample).

2) **Do not fold this filter.** Return filter to it's numbered container, wrap in aluminum foil, affix tracking label to 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 58) with a stirring probe (e.g., YSI 5730).
6. Spare batteries for D.O. meter.
7. Permanent markers for labeling tubes.
8. Sample labels and field data sheets.
9. Ice chest with dry ice for sample freezing.

FIELD PROTOCOLS

Dissolved Oxygen Meter Calibration (for YSI model 58, with YSI model 5730 stirring BOD probe)

1. Zero meter according to manufacturer's directions, and
2. Calibrate meter using the water-saturated atmosphere method described in the meter's operations manual.

QUICK REFERENCE GUIDE FOR SEDIMENT METABOLISM (Continued)

Sediment Collection and Experimental Set-up

1. Collect and combine fine-grained, surface sediments (top 2 cm) from all depositional areas along the stream reach (Transects B-J) established for the Physical Habitat Characterization.

2. Fill ice chest 2/3 full with stream 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.

12. If you are collecting samples for sediment toxicity tests, save 1 to 2 L of the remaining sediment sample by placing it in a labeled plastic bag.

13. Store sediment toxicity sample chilled (but not frozen!) for laboratory analysis.

QUICK REFERENCE GUIDE FOR SEDIMENT TOXICITY

SAMPLE COLLECTION AND SHIPMENT FOR SEDIMENT TOXICITY SAMPLES

1. Use the sediment left over from the benthic (sediment) metabolism indicator in Section 9, Benthic (Sediment) Metabolism: Field Methods.

2. Mix sediment well with a stainless steel or plastic mixing spoon or gloved hand.

3. Fill a 2-gallon polyethylene (4 mil) bag with at least 1 L of sediment.

4. Close bag, squeeze the air out and tie a knot in the remaining portion of the bag to seal.

5. Fill out ID label; place label on the outside of the bag. Place this bag inside a second 2-gallon polyethylene bag and tie off the top to seal.

6. Place these bags inside a cooler with only sediment samples in them.

7. Hold sediment samples on ice (do not freeze!) for laboratory analysis.

8. Ship samples to the designated contact person or laboratory.

QUICK REFERENCE GUIDE FOR BENTHIC MACROINVERTEBRATES

TABLE I. BASE PROTOCOLS FOR COLLECTING MACROINVERTEBRATES

1. Do the water chemistry.

2. Locate first sampling station (second flag) from downstream end of the study segment and roll die to pick left (1), middle (2), or right side (3) of transect to sample. If stream is narrower than three nets, pick left or right. If wide enough for only one net, then sample entire stream width. After first transect, systematically sample remaining transects left, middle, or right so that three samples are collected on left, middle, and right at the site.

3. If riffle or run use protocol in Table II. If pool use protocol in Table III or hand pick for 60 seconds if kick net cannot be used.

4. Go to next upstream station and repeat. Combine all riffle samples in one bucket and pool samples in another. Check net after each sample for clinging organisms and transfer to bucket.

5. After a sample is collected from each of nine interior transects and all samples are combined in the proper bucket, obtain a composite sample as described in Table IV.

6. Assist with the fish collection.

7. Preserve and label each sample as described in Table V.

(continued)

QUICK REFERENCE GUIDE FOR BENTHIC MACROINVERTEBRATES (Continued)

TABLE II. PROCEDURES FOR RIFFLES AND RUNS USING KICK NET SAMPLER

1. Attach four foot pole to the sampler.

2. Position sampler quickly and securely on stream bottom with net opening upstream.

3. Hold the sampler in position on the substrate while checking for snails and clams in an area of about 0.5 m^2 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 upstream motion to wash the organisms to the bottom of net.

6. Rinse net contents into the "riffle" 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. Combine all riffle samples in the "riffle" bucket.

10. After all stations are sampled and all riffle samples combined in the "riffle" bucket, obtain a composite sample as described in Table IV.

(continued)

QUICK REFERENCE GUIDE FOR BENTHIC MACROINVERTEBRATES (Continued)

TABLE III. PROCEDURES FOR POOLS USING THE MODIFIED KICK NET SAMPLER

1. Attach four-foot pole to the sampler.

2. Inspect about 1/2 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 m^2 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 upstream 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. Combine this sample with the other pool samples in the "pool" bucket.

9. After all stations are sampled and all pool samples are combined together in the "pool" bucket, obtain a composite sample as described in Table IV.

(continued)

QUICK REFERENCE GUIDE FOR BENTHIC MACROINVERTEBRATES (Continued)

TABLE IV. PROCEDURES FOR OBTAINING THE COMPOSITE SAMPLE

1. Pour the contents of the riffle 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 jar.

3. Place properly filled out waterproof label in the jar and replace the cap.

TABLE V. 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 waterproof label is in jar with the required information on it.

4. Check to be sure that the pre-numbered stick-on barcoded label is the on jar and agrees with the inside label. Cover the entire label with clear, waterproof tape.

5. With a grease pencil write the site number, sample type (Riffle or Pool), and the number of transects sampled for either Riffle or Pool on the cap.

6. Seal the caps with electrical tape.

7. Complete the check off sheet and place samples in cooler or other secure container for transport.

8. Secure all equipment in the vehicle.

(continued)

QUICK REFERENCE GUIDE FOR BENTHIC MACROINVERTEBRATES (Continued)

TABLE VI. MACROINVERTEBRATE SAMPLING ACTIVITIES CHECKLIST			
Date:		**Time:**	**Site No.:**
Stream Name and Location:			
Crew ID: 1 2 3 4 5 6		**Collector:**	
1.	Initial observations, if any, on the Sample Collection Form - Streams.		
2.	Composite riffle/run sample collected with label inside jar.		
3.	Composite pool/glide sample collected with label inside jar.		
4.	Correct barcode and label on all jars and sealed with clear, waterproof tape.		
5.	All samples preserved.		
6.	With a grease pencil write site number, sample type (Riffle or Pool), and number of transects sampled for sample type on the cap. If two jars are used be sure to mark them as such.		
7.	Caps sealed with tape.		
8.	Photos of site.		
9	Sample jars in cooler or otherwise secured.		
10.	All equipment accounted for and secured in vehicle.		
Signature:			**Time sampling completed:**

QUICK REFERENCE GUIDE FOR AQUATIC VERTEBRATES

FIELD PROTOCOLS FOR FISH COLLECTION

1. Site Selection

   a. Determine channel width.

   b. Survey sample reach.

   c. Determine if reach requires block nets.

   d. If conductivity is below 10 :S/cm or if flow, depth or turbidity make it unsafe to electrofish, crew may elect to use seine only or not sample. THIS IS A SAFETY DECISION.

   e. **In case of emergency, determine location of means of easy egress from stream.**

2. Electrofishing

   a. Set unit to 300VA and pulsed DC. Select initial voltage setting. Start generator, set timer, and depress switch to begin fishing.

   b. Fish in an upstream direction, parallel to the current. Adjust voltage and waveform output according to sampling effectiveness and mortality fish specimens.

   c. With switch depressed, sweep electrodes from side to side in the water. Sample available cut-bank and snag habitat as well as riffles and pools.

   d. Netters follow operator and net fish. Deposit fish in buckets. Block with seines in riffles, pools and snags.

   e. Continue for 40 channel widths. Record total time spent collecting and shocking time on data sheets.

   f. Identify and release any threatened and endangered species.

   g. Identify and measure (SL, TL) sport fish and very large specimens, record external anomalies, and release unharmed.

   h. Identify other specimens. Determine number of individuals in species, measure largest and smallest individuals, and voucher as described in Voucher Protocol.

   i. Retain a subsample of target species for Fish Tissue Contaminants analysis.

3. Seining will be used in conjunction with electrofishing and in sites where stream is too deep for electrofishing to be conducted safely.

(continued)

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	GR	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.
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	WR	Annelid worms which have anterior and posterior suckers. They may attach anywhere on the body.
Exophthalmia	EX	Bulging of the eye.

(continued)

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 25 adults and juveniles. If less than 25 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 kill jar containing a strong (approximately 20%) formalin solution. 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. 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.

d. Transport to storage depot at end of week. Store in a cool, dark, ventilated space.

QUICK REFERENCE GUIDE FOR FISH TISSUE CONTAMINANTS

SELECTING FISH TISSUE SPECIMENS

If possible, obtain one sample each, containing the desired **weight** or **number** (see below) of **similarly sized individuals***, from the **primary** and **secondary** target species lists (**2 composite samples** total):

I. PRIMARY TARGET SPECIES

Small adult fish (in priority order)		**DESIRED 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). Use scale provided to determine weight. With clean hands, place the fish in fresh aluminum foil (dull side towards fish) before placing fish in weighing container.

(B) If **fewer than the desired number** of individuals of **any primary target species** are collected, send individuals of a small non-target species if 50 g or more are available.

***** - Getting a sufficient sample amount is a higher priority than getting similar-sized individuals.
****** - This weight represents the **minimum amount** needed for laboratory analysis. Crews should not settle for the minimum weight if more fish are present. They should **send as many fish as possible** up to 400 g weight goal.

QUICK REFERENCE GUIDE FOR FISH TISSUE CONTAMINANTS (Continued)

SELECTING FISH TISSUE SPECIMENS

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)	White sucker	>120 mm	5
2)	Hogsucker	>120 mm	5
3)	A Bass species	>120 mm	5
4)	A Trout species	>120 mm	5
5)	A Sunfish species	>120 mm	5
6)	Carp	>120 mm	5

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 or at least 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 nor secondary species** sample that **meets these criteria** is available, use your best judgement in sending some type of fish sample (may be mixed species).

(continued)

QUICK REFERENCE GUIDE FOR FISH TISSUE CONTAMINANTS (Continued)

PROCESSING TISSUE SPECIMENS

1. Keep hands, work surfaces, and wrapping materials clean and free of potential contaminants (mud, fuel, formalin, sun screen, insect repellant, etc.)

2. Measure **total weight** of individuals for **primary target** species and count the total number of individuals. Measure the **total length** (TL) of **each secondary target species individual**. **Record** all of this information in the fish tissue section of the Sample Collection Form.

3. Write the bar-code number(s)*** on the collection form. Make sure that the form is filled out completely.

4. Wrap fish in aluminum foil. Place the dull side of the aluminum foil in contact with the fish. The **primary target fish** sample may be wrapped as a **group**. **Secondary target fish** should be wrapped individually. Once wrapped, place each sample in a self-sealing plastic bag or a garbage bag.

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

6. Complete bar-coded fish tissue label(s). Make sure the number(s) is/are the same one(s) on the collection form. Apply it/them to the tape surface(s). Cover the label(s) with clear, waterproof tape. As labels will sometimes fall off, there should **always** be a **label on the inner bag**.

7. Place labeled bag(s) into a second plastic bag(s) and seal and label second bag(s). Repeat previous two steps.

8. Place double-bagged sample(s) in cooler with dry ice until shipment.

9. Ship weekly on dry ice by Federal Express next day service. KEEP FROZEN UNTIL SHIPMENT. If ice is used, double bag ice in self-sealing plastic bags and tape shut to avoid contamination of samples if ice should melt.

*** - If both primary and secondary target species are collected, the two samples should be wrapped and bagged separately, with separate bar codes and labels, but only one Sample Tracking Form.

APPENDIX C

FIELD DATA FORMS

Copies of field data forms are arranged according the general order of their use at each stream site:

1. Verification Form
2. Sample Collection Form
3. Field Measurement Form
4. Channel/Riparian Cross-Section & Thalweg Profile Form
5. Slope and Bearing Form
6. Vertebrate Collection Form
7. Vertebrate Length Recording Form
8. Rapid Habitat Assessment Form (Riffle/Run Prevalent)
9. Rapid Habitat Assessment Form (Pool/Glide Prevalent)
10. Assessment Form

Electronic versions of the forms may be available through the EMAP-Surface Waters Technical Director, U.S. EPA, 200 SW 35th St, Corvallis, OR 97333.

Reviewed by (initial): _____

VERIFICATION FORM - STREAMS/RIVERS

SITE NAME: **DATE:** / / **VISIT:** G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ **TEAM ID (X):** G1 G2 G3 G4 G5 G6 G7 G8

STREAM/RIVER VERIFICATION INFORMATION

STREAM/RIVER VERIFIED BY (X all that apply) : G GPS G LOCAL CONTACT G SIGNS G ROADS G TOPO. MAP

G OTHER (DESCRIBE HERE): G 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:	" ' " __ __ __ __ __ __	" ' " __ __ __ __ __ __	G 2D	G YES
GPS:	" ' " __ __ __ __ __ __	" ' " __ __ __ __ __ __	G 3D	G NO

INDEX SITE STATUS - X ONE BOX FROM ONE SECTION ONLY

SAMPLEABLE

G REGULAR - WADEABLE

G REGULAR - NOT WADEABLE

G INTERMITTENT - DRY SPOTS ALONG REACH

G DRY - NO WATER ANYWHERE ALONG REACH

G ALTERED - STREAM/RIVER PRESENT BUT NOT AS ON MAP

G OTHER (EXPLAIN IN COMMENTS)

NON-SAMPLEABLE (NO SAMPLE TAKEN)

G NO CHANNEL OR WATERBODY PRESENT

G IMPOUNDED (UNDERNEATH LAKE/POND)

G WETLAND (NO DEFINABLE CHANNEL)

NO ACCESS

G ACCESS PERMISSION DENIED

G INACCESSIBLE (UNABLE TO REACH SITE)

DIRECTIONS TO STREAM/RIVER SITE

GENERAL COMMENTS

Reviewed by (initial): _____

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

VERIFICATION FORM - STREAMS/RIVERS (continued)

SITE NAME: **DATE:** / / **VISIT:** G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ **TEAM ID (X):** G1 G2 G3 G4 G5 G6 G7 G8

STREAM/RIVER REACH DETERMINATION

CHANNEL WIDTH USED TO DEFINE REACH (M) (XX):	DISTANCE (M) FROM X-SITE		COMMENT
	UPSTREAM LENGTH	DOWNSTREAM LENGTH	

ARROW INDICATES NORTH

Reviewed by (initial): _____

SAMPLE COLLECTION FORM - STREAMS

SITE NAME: **DATE:** / / **VISIT:** G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ **TEAM ID (X):** G1 G2 G3 G4 G5 G6 G7 G8

COMPOSITE BENTHOS SAMPLES

SAMPLE ID (BARCODE)	HABITAT (X ONE)		NO. OF JARS	FLAG	COMMENTS
	R	P			
___ ___ ___ ___ ___ ___					
___ ___ ___ ___ ___ ___					

STATION	A	B	C	D	E	F	G	H	I	J	K
RIFFLE OR POOL - (X ONE) 6		GR GP	GR GP	GR GP	GR GP	GR GP	GR GP	GR GP	GR GP	GR GP	
LEFT, CENTER, OR RIGHT - (X ONE) 6		GL GC GR	GL GC GR	GL GC GR	GL GC GR	GL GC GR	GL GC GR	GL GC GR	GL GC GR	GL GC GR	

COMPOSITE PERIPHYTON SAMPLES		HABITAT TYPE (X) 6	G RIFFLE G POOL G OTHER
SAMPLE ID (BARCODE) 6	___ ___ ___ ___ ___ ___	COMPOSITE VOLUME 6	___ ___ ___ ___ mL

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

COMPOSITE PERIPHYTON SAMPLES		HABITAT TYPE (X) 6	G RIFFLE G POOL G OTHER
SAMPLE ID (BARCODE) 6	___ ___ ___ ___ ___ ___	COMPOSITE VOLUME 6	___ ___ ___ ___ mL

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

COMMENTS:

Reviewed by (initial): _____

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

Reviewed by (initial): _____

SAMPLE COLLECTION FORM - STREAMS (continued)

SITE NAME: **DATE:** / / **VISIT:** G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ **TEAM ID (X):** G1 G2 G3 G4 G5 G6 G7 G8

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

	SAMPLE ID (BARCODE)	TRANSECT	FLAG	COMMENTS
CHEMISTRY	___ ___ ___ ___ ___ ___			
	___ ___ ___ ___ ___ ___			

SEDIMENT TOXICITY SAMPLES

SAMPLE ID (BARCODE)	FLAG	COMMENTS
___ ___ ___ ___ ___ ___		

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

SAMPLE ID (BARCODE) 6 ___ ___ ___ ___ ___ ___

LINE	SPECIES CODE	COMMON NAME	NUMBER OF INDIVIDUALS	FLAG
P1	___ ___ ___ ___ ___ ___			
	___ ___ ___ ___ ___ ___			

IS COMPOSITE SAMPLE COMPOSED OF INDIVIDUALS COLLECTED FROM THROUGHOUT REACH? (X) 6 G YES G NO

IF NO, EXPLAIN:

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

SAMPLE ID (BARCODE) 6 ___ ___ ___ ___ ___ ___

LINE	SPECIES CODE	COMMON NAME	TOTAL LENGTH (MM)	FLAG
S1	___ ___ ___ ___ ___ ___			
S2	___ ___ ___ ___ ___ ___			
S3	___ ___ ___ ___ ___ ___			
S4	___ ___ ___ ___ ___ ___			
S5	___ ___ ___ ___ ___ ___			

IS COMPOSITE SAMPLE COMPOSED OF INDIVIDUALS COLLECTED FROM THROUGHOUT REACH? (X) 6 G YES G NO

IF NO, EXPLAIN:

LINE	COMMENT OR FLAG EXPLANATION FOR FISH TISSUE

Reviewed by (initial): _____

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

Reviewed by (initial): _____

FIELD MEASUREMENT FORM - STREAMS/RIVERS

SITE NAME: **DATE:** / / **VISIT:** G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ **TEAM ID (X):** G1 G2 G3 G4 G5 G6 G7 G8

WEATHER CONDITIONS (X)

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

IN SITU MEASUREMENTS

STATION ID: _____ Assume X-site unless marked

		FLAG	COMMENTS
QCCS COND µS/CM	___ ___ ___ ___		
STREAM/RIVER COND µS/CM	___ ___ ___ ___		
STREAM/RIVER DO MG/L	___ ___ . ___		
STREAM/RIVER TEMP °C	___ ___ . ___		

STREAM/RIVER METABOLISM DETERMINATION

INITIAL O_2 (MG/L)	INITIAL INCUBATION TEMP. (°C)	INCUBATION TIME (24-HR TIME) START (HH:MM)	INCUBATION TIME (24-HR TIME) FINISH (HH:MM)	DURATION OF INCUBATION (HH:MM)	FLAG	COMMENTS
___ ___ . ___	___ ___ . ___	___ ___ : ___ ___	___ ___ : ___ ___	___ ___ : ___ ___		

SAMPLE ID (BARCODE)	FINAL O_2 (MG/L)	FLAG	COMMENTS
___ ___ ___ ___ ___ ___	___ ___ . ___		
___ ___ ___ ___ ___ ___	___ ___ . ___		
___ ___ ___ ___ ___ ___	___ ___ . ___		
___ ___ ___ ___ ___ ___	___ ___ . ___		
___ ___ ___ ___ ___ ___	___ ___ . ___		
___ ___ ___ ___ ___ ___	___ ___ . ___		

OXYGEN METER CALIBRATION INFORMATION

MEMBRANE CHECK G ELECTRONIC ZERO G RED LINE: G

CALIBRATION CHAMBER TEMPERATURE: °C	SATURATED O_2 @ TEMP.: MG/L
STATION ELEVATION (FROM TOPO. MAP OR ALTIMETER): FT	ELEVATION CORRECTION FACTOR: ×
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:

Reviewed by (initial): _____

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.

Reviewed by (initial): _____

FIELD MEASUREMENT FORM - STREAMS (continued)

SITE NAME: DATE: / / VISIT: G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ TEAM ID (X): G1 G2 G3 G4 G5 G6 G7 G8

STREAM DISCHARGE

G VELOCITY AREA

	DIST. FROM BANK (CM)	VELOCITY (FT/S) XX.X	DEPTH (FEET) XX.X	FLAG
1	___ ___ ___	___ ___ . ___	___ ___ . ___	
2	___ ___ ___	___ ___ . ___	___ ___ . ___	
3	___ ___ ___	___ ___ . ___	___ ___ . ___	
4	___ ___ ___	___ ___ . ___	___ ___ . ___	
5	___ ___ ___	___ ___ . ___	___ ___ . ___	
6	___ ___ ___	___ ___ . ___	___ ___ . ___	
7	___ ___ ___	___ ___ . ___	___ ___ . ___	
8	___ ___ ___	___ ___ . ___	___ ___ . ___	
9	___ ___ ___	___ ___ . ___	___ ___ . ___	
10	___ ___ ___	___ ___ . ___	___ ___ . ___	
11	___ ___ ___	___ ___ . ___	___ ___ . ___	
12	___ ___ ___	___ ___ . ___	___ ___ . ___	
13	___ ___ ___	___ ___ . ___	___ ___ . ___	
14	___ ___ ___	___ ___ . ___	___ ___ . ___	
15	___ ___ ___	___ ___ . ___	___ ___ . ___	
16	___ ___ ___	___ ___ . ___	___ ___ . ___	
17	___ ___ ___	___ ___ . ___	___ ___ . ___	
18	___ ___ ___	___ ___ . ___	___ ___ . ___	
19	___ ___ ___	___ ___ . ___	___ ___ . ___	
20	___ ___ ___	___ ___ . ___	___ ___ . ___	

G TIMED FILLING

REPEAT	VOL. (L) XX.X	TIME (S)	FLAG
1	___ ___ . ___		
2	___ ___ . ___		
3	___ ___ . ___		
4	___ ___ . ___		
5	___ ___ . ___		

G NEUTRALLY BUOYANT OBJECT

MEASUREMENT	Cross Section		
	ONE	TWO	THREE
WIDTH (m)	___ ___ . ___	___ ___ . ___	___ ___ . ___
DEPTH 1 (cm)	___ ___ ___	___ ___ ___	___ ___ ___
DEPTH 2 (cm)	___ ___ ___	___ ___ ___	___ ___ ___
DEPTH 3 (cm)	___ ___ ___	___ ___ ___	___ ___ ___
DEPTH 4 (cm)	___ ___ ___	___ ___ ___	___ ___ ___
DEPTH 5 (cm)	___ ___ ___	___ ___ ___	___ ___ ___
FLOAT DISTANCE (m)	___ ___	___ ___	___ ___
FLOAT TIME (s)	___ ___ ___	___ ___ ___	___ ___ ___

FLAG	COMMENTS

Reviewed by (initial): _____

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.

Reviewed by (initial): _____

FIELD MEASUREMENT FORM - STREAMS (continued)

SITE NAME: DATE: / / VISIT: G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ TEAM ID (X): G1 G2 G3 G4 G5 G6 G7 G8

STREAM DISCHARGE

G VELOCITY AREA

	DIST. FROM BANK (CM)	VELOCITY (M/S) XX.X	DEPTH (CM) XX.X	FLAG
1	___ ___ ___	___ . ___ ___	___ ___ ___	
2	___ ___ ___	___ . ___ ___	___ ___ ___	
3	___ ___ ___	___ . ___ ___	___ ___ ___	
4	___ ___ ___	___ . ___ ___	___ ___ ___	
5	___ ___ ___	___ . ___ ___	___ ___ ___	
6	___ ___ ___	___ . ___ ___	___ ___ ___	
7	___ ___ ___	___ . ___ ___	___ ___ ___	
8	___ ___ ___	___ . ___ ___	___ ___ ___	
9	___ ___ ___	___ . ___ ___	___ ___ ___	
10	___ ___ ___	___ . ___ ___	___ ___ ___	
11	___ ___ ___	___ . ___ ___	___ ___ ___	
12	___ ___ ___	___ . ___ ___	___ ___ ___	
13	___ ___ ___	___ . ___ ___	___ ___ ___	
14	___ ___ ___	___ . ___ ___	___ ___ ___	
15	___ ___ ___	___ . ___ ___	___ ___ ___	
16	___ ___ ___	___ . ___ ___	___ ___ ___	
17	___ ___ ___	___ . ___ ___	___ ___ ___	
18	___ ___ ___	___ . ___ ___	___ ___ ___	
19	___ ___ ___	___ . ___ ___	___ ___ ___	
20	___ ___ ___	___ . ___ ___	___ ___ ___	

G TIMED FILLING

REPEAT	VOL. (L) XX.X	TIME (S)	FLAG
1	___ ___ . ___		
2	___ ___ . ___		
3	___ ___ . ___		
4	___ ___ . ___		
5	___ ___ . ___		

G NEUTRALLY BUOYANT OBJECT

MEASUREMENT	Cross Section		
	ONE	TWO	THREE
WIDTH (m)	___ ___ . ___	___ ___ . ___	___ ___ . ___
DEPTH 1 (cm)	___ ___ ___	___ ___ ___	___ ___ ___
DEPTH 2 (cm)	___ ___ ___	___ ___ ___	___ ___ ___
DEPTH 3 (cm)	___ ___ ___	___ ___ ___	___ ___ ___
DEPTH 4 (cm)	___ ___ ___	___ ___ ___	___ ___ ___
DEPTH 5 (cm)	___ ___ ___	___ ___ ___	___ ___ ___
FLOAT DISTANCE (m)	___ ___	___ ___	___ ___
FLOAT TIME (s)	___ ___ ___	___ ___ ___	___ ___ ___

FLAG	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.

Reviewed by (initial): ___

PHab: CHANNEL/RIPARIAN CROSS-SECTION & THALWEG PROFILE FORM - STREAMS

SITE NAME: **SITE ID:** ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ **DATE:** / / **VISIT:** G1 G2 ___

TEAM ID(X): G1 G2 G3 G4 G5 G6 G7 G8 **TRANSECT(X):** GA GB GC GD GE GF GG GH GI GJ GK

I. SUBSTRATE CROSS-SECTIONAL INFORMATION

Loc.	Dist LB XX.XX m	Depth XXX cm	Size Class Code	Embed. 0-100%	Flag
Lft	__ __ . __ __	__ __ __	__ __	__ __ __	
LCtr	__ __ . __ __	__ __ __	__ __	__ __ __	
Ctr	__ __ . __ __	__ __ __	__ __	__ __ __	
RCtr	__ __ . __ __	__ __ __	__ __	__ __ __	
Rgt	__ __ . __ __	__ __ __	__ __	__ __ __	

SUBSTRATE SIZE CLASS CODES

III. FISH COVER/ OTHER

COVER IN-CHANNEL (X ONE)

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

	0	1	2	3	4	Flag
Filamentous Algae						
Macrophytes						
Woody Debris > 0.3 m (Big)						
Brush/Woody Debris < 0.3 m (Small)						
Overhanging Veg. # 1 m of surface						
Undercut Banks						
Boulders						
Artificial Structures						

II. BANK MEASUREMENTS

Location	Bank Angle 0-360°	Undercut Dist. (m) X.XX	Flag
Left	__ __ __ °	__ . __ __	
Right	__ __ __ °	__ . __ __	
Wetted Width		__ __ . __ m	
Bar Width		__ __ . __ m	
Bankfull Width		__ __ . __ m	
Bankfull Height		__ __ . __ m	
Incised Height		__ __ . __ m	

IV. CANOPY COVER MEASUREMENTS

Densiometer (0 to 17 max)

		Flag			Flag
CenUp	__ __		CenR	__ __	
CenL	__ __		Lft	__ __	
CenDwn	__ __		Rgt	__ __	

Flag Codes: **K** = no measurement made; **U** = suspect measurement; **F1**, **F2**, ect. = misc. flags assigned by each field crew. Explain all flags in comments section

V. VISUAL RIPARIAN ESTIMATES

	Left Bank					Right Bank					Flag
Riparian Vegetation Cover	0 = Absent (0%); 1 = Spare (< 10%); 2 = Moderate (10 - 40%); 3 = Heavy (40 - 75%); 4 = Very Heavy (> 75%)					D = Deciduous; C = Coniferous; E = Broadleaf Evergreen; M = Mixed; N = None					
CANOPY (> 5 m high)	D	C	E	M	N	D	C	E	M	N	Flag
Vegetation Type (D, C, M, or N)											
	0	1	2	3	4	0	1	2	3	4	Flag
Big Trees (Trunk > 0.3 m DBH)											
Small Trees (Trunk < 0.3 m DBH)											
UNDERSTORY (0.5 to 5 m high)	D	C	E	M	N	D	C	E	M	N	Flag
Vegetation Type (D, C, M, or N)											
	0	1	2	3	4	0	1	2	3	4	Flag
Woody Shrubs & Saplings											
Non-woody Herbs, Grasses, & Forbs											
GROUND COVER (< 0.5 m high)	0	1	2	3	4	0	1	2	3	4	Flag
Woody Shrubs & Seedlings											
Non-woody Herbs, Grasses, & Forbs											
Barren, Bare dirt or duff											
HUMAN INFLUENCE	0 = Not present, C = Within 10 m, P = > 10 m, B = On bank										Flag
	O	P	C	B		O	P	C	B		
Wall/Dike/Revetment/Riprap/Dam											
Buildings											
Pavement											
Road/Railroad											
Pipes (inlet/outlet)											
Landfill/Trash											
Park/Lawn											
Row Crops											
Pasture/Range/Hay field											
Logging Operations											
Mining Activity											

Reviewed by (initial): ____

PHab: THALWEG PROFILE & WOODY DEBRIS FORM - STREAMS

SITE NAME: **DATE:** / / **VISIT:** G1 G2 ___

SITE ID: M A I A 9 7 - __ ___ _____ ___ ___ **TEAM ID (X):** G1 G2 G3 G4 G5 G6 G7 G8

TRANSECT(X): GA-B GB-C GC-D GD-E GE-F GF-G GG-H GH-I GI-J GJ-K

THALWEG PROFILE

Increment (m) 6

STATION	THALWEG DEPTH (cm) (XXX)	WETTED WIDTH (m) (XX.X)	BAR WIDTH [1]		SOFT/SMALL SEDIMENT (X FOR YES)	CHANNEL UNIT CODE	POOL FORM CODE	SIDE CHANNEL (X FOR YES)	FLAG	COMMENTS
			X	(XX.X)						
0										
1										
2										
3										
4										
5										
6										
7										
8										
9										
10										
11										
12										
13										
14										

LARGE WOODY DEBRIS ($ 10 cm SMALL END DIAMETER.; $ 1.5 m LENGTH)

- TALLY EACH PIECE -

DIAMETER LARGE END	PIECES ALL/PART IN BANKFULL CHANNEL			PIECES BRIDGE ABOVE BANKFULL CHANNEL		
	LENGTH 1.5 - 5 m	5 - 15 m	> 15 m	LENGTH 1.5 - 5 m	5 - 15 m	> 15 m
0.1 to <0.3 m						
0.3 - 0.6 m						
0.6 - 0.8 m						
> 0.8 m						

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

POOL FORM CODES	
N	Not a pool
W	Large Woody Debris
R	Rootwad
B	Boulder or bedrock
F	Unknown, fluvial
O	Other (note in comments)

FLAG	COMMENTS

Flag Codes: **K** = no measurement made; **U** = suspect measurement; **F1**, **F2**, etc. = misc. flags assigned by each field crew. Explain all flags in comments. 1 = Measure Bar Width at Station 0 and Mid-Station (5 or 7), X small column if bar present at the rest of the stations.

Reviewed by (initial): _____

PHab: SLOPE AND BEARING FORM - STREAMS

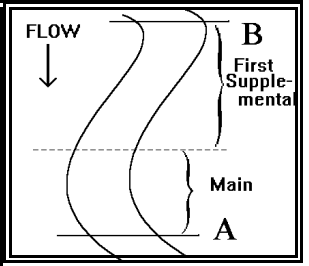
NOTE: ON BACK SIDE OF THIS FORM IS THE TORRENT EVIDENCE ASSESSMENT FORM !

SITE NAME: **DATE:** / / **VISIT:** G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ **TEAM ID (X):** G1 G2 G3 G4 G5 G6 G7 G8

TRANSECT	MAIN			FIRST SUPPLEMENTAL			SECOND SUPPLEMENTAL			FLAG
	SLOPE	BEARING 0 - 360	PROPOR-TION	SLOPE	BEARING 0 - 360	PROPOR-TION	SLOPE	BEARING 0 - 360	PROPOR-TION	
A 7 B	___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		
B 7 C	___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		
C 7 D	___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		
D 7 E	___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		
E 7 F	___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		
F 7 G	___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		
G 7 H	___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		
H 7 I	___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		
I 7 J	___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		
J 7 K	___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		___ ___ . ___ %	___ ___ ___ °		

FLAG	COMMENTS



Reviewed by (initial):__________

VERTEBRATE COLLECTION FORM - STREAMS/RIVERS

SITE NAME: DATE: / / VISIT: G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ TEAM ID (X): G1 G2 G3 G4 G5 G6 G7 G8

ID BY (NAME): TRANSECT(X): GA-B GB-C GC-D GD-E GE-F GF-G GG-H GH-I GI-J GJ-K G ALL (stream)

TOTAL SHOCK (button) TIME ___ ___ ___ ___ seconds **TOTAL FISHING TIME** ___ ___ ___ minutes **SHOCK DISTANCE (M)** ___ ___ ___

SAMPLE ID (BARCODE)6 ___ ___ ___ ___ ___ ___ G **NOT FISHED** G **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		
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				

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

Reviewed by (initial):__________

VERTEBRATE COLLECTION FORM - STREAMS/RIVERS (continued)

Page ____ of ____

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___

DATE: / /

VISIT: G1 G2 ___

Sample ID (Barcode) 6 ___ ___ ___ ___ ___ ___

TEAM ID (X): G1 G2 G3 G4 G5 G6 G7 G8

ID BY (NAME):

TRANSECT(X): GA-B GB-C GC-D GD-E GE-F GF-G GG-H GH-I GI-J GJ-K G ALL (stream)

SPECIMENS (continued)

TAG NO.	SPECIES CODE	COMMON NAME	TOTAL NUMBER		VOUCHERED COUNT	LENGTH (mm)		ANOMALIES		NUMBER OF MORTALITIES	FLAG
			TALLY	COUNT		MIN.	MAX.	CODE	COUNT		
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
___ ___	___ ___ ___ ___ ___ ___					___ ___ ___	___ ___ ___				
						___ ___ ___	___ ___ ___				

FLAG	COMMENTS

Reviewed by (initial):__________

VERTEBRATE LENGTH RECORDING FORM - STREAMS/RIVERS

SITE NAME: DATE: / / VISIT: G1 G2 ____

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ TEAM ID (X): G1 G2 G3 G4 G5 G6 G7 G8

MEASURED BY (NAME):

SPECIES CODE	COMMON NAME	LENGTH (MM) OF INDIVIDUAL FISH														
___ ___ ___ ___ ___ ___																
___ ___ ___ ___ ___ ___																
___ ___ ___ ___ ___ ___																
___ ___ ___ ___ ___ ___																
___ ___ ___ ___ ___ ___																
___ ___ ___ ___ ___ ___																
___ ___ ___ ___ ___ ___																
___ ___ ___ ___ ___ ___																
___ ___ ___ ___ ___ ___																

Reviewed by (initial): _____

RAPID HABITAT ASSESSMENT FORM: RIFFLE/RUN PREVALENCE - STREAMS

SITE NAME: DATE: / / 97 VISIT: G1 G2 ___

SITE ID: M A I A 9 7 - ___ ___ ___ ___ ___ TEAM ID (X): G1 G2 G3 G4 G5 G6 G7 G8

TOTAL SCORE		CATEGORY			
HABITAT PARAMETER		OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
1. INSTREAM COVER (FISH)		Greater than 50% mix of boulder, cobble, submerged logs, undercut banks, or other stable habitat.	30 to 50% mix of boulder, cobble, or other stable habitat; adequate habitat.	10 to 30% mix of boulder, cobble, or other stable habitat; habitat availability is less than desirable.	Less than 10% of boulder, cobble, or other stable habitat; lack of habitat is obvious.
	SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
2. EPIFAUNAL SUBSTRATE		Well-developed riffle and run; riffle is as wide as stream and its length extends two times the width of stream; abundance of cobble.	Riffle is as wide as stream, but is less than two times width; abundance of cobble; boulders and gravel common.	Run area may be lacking; reduced riffle area that does not extend across entire cross section and is less than two times the width; gravel or large boulders and bedrock prevalent; cobble present.	Riffles or run virtually non-existent; gravel or large boulders and bedrock prevalent; cobble lacking.
	SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
3. EMBEDDEDNESS		Gravel, cobble, and boulder particles are between 0 and 25% surrounded by fine sediment.	Gravel, cobble, and boulder particles are between 25 and 50% surrounded by fine sediment.	Gravel, cobble, and boulder particles are between 50 and 75% surrounded by fine sediment.	Gravel, cobble, and boulder particles are over 75% surrounded by fine sediment.
	SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
4. VELOCITY/DEPTH REGIMES		All four velocity regimes are present (slow-deep, slow-shallow, fast-deep, fast-shallow).	Only three of the four habitat types are present (if fast-shallow is missing, score lower than if other regimes are missing).	Only two of the four habitat types are present (if fast-shallow or slow-shallow are missing, score low).	Dominated by one velocity/depth regime (usually slow-deep).
	SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
5. CHANNEL ALTERATION		No channelization of dredging present..	Some channelization is present, usually in areas of bridge abutments; evidence of past channelization, i.e., dredging (greater than past 20 yr) may be present, but recent channelization is not present.	New embankments are present on both banks; and 40 to 80% of the stream reach is channelized and disrupted.	Banks shored with gabion or cement; over 80% of the stream reach is channelized and disrupted.
	SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
6. SEDIMENT DEPOSITION		Little or no enlargement of islands or point bars and less than 5% of the bottom is affected by sediment deposition.	Some new increase in bar formation, mostly from coarse gravel; 5 to 30% of the bottom is affected; slight deposition in pools.	Moderate deposition of new gravel or coarse sand on old and new bars; 30 to 50% of the bottom is affected; sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools prevalent.	Heavy deposits of fine material; increased bar development; more than 50% of the bottom is changing frequently; pools almost absent due to substantial sediment deposition.
	SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

Reviewed by (initial): _____

RAPID HABITAT ASSESSMENT FORM: RIFFLE/RUN - STREAMS (continued)

SITE NAME: DATE: / / VISIT: G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ TEAM ID (X): G1 G2 G3 G4 G5 G6 G7 G8

HABITAT PARAMETER	CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
7. FREQUENCY OF RIFFLES	Occurrence of riffles is relatively frequent; the distance between riffles divided by the width of the stream equals 5 to 7; variety of habitat.	Occurrence of riffles is infrequent; distance between riffles divided by the width of the stream equals 7 to 15.	Occasional riffle or bend; bottom contours provide some habitat; distance between riffles divided by the width of the stream is between 15 to 25.	Generally all flat water or shallow riffles; poor habitat; distance between riffles divided by the width of the stream is greater than 25.
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
8. CHANNEL FLOW STATUS	Water reaches the base of both banks and a minimal area of channel substrate is exposed.	Water fills more than 75% of the available channel; or less than 25% of the channel substrate is exposed.	Water fill 25 to 75% of the available channel; and/or riffle substrates are mostly exposed.	Very little water in channel, and mostly present as standing pools.
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
9. CONDITION OF BANKS	Banks stable; no evidence of erosion or bank failure.	Banks moderately stable; infrequent, small areas of erosion mostly healed over.	Moderately unstable; up to 60% of banks in reach have areas of erosion.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; on side slopes, 60 to 100% of bank has erosional scars.
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
10. BANK VEGETATIVE PROTECTION	More than 90% of the stream bank surfaces are covered by vegetation.	70 to 90% of the stream bank surfaces are covered by vegetation.	50 to 70% of the stream bank surfaces are covered by vegetation.	Less than 50% of the stream bank surfaces are covered by vegetation.
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
11. GRAZING OR OTHER DISRUPTIVE PRESSURE	Vegetative disruption, through grazing or mowing is minimal or not evident; almost all plants are allowed to grow naturally.	Disruption is evident but is not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	Disruption is obvious; patches of bare soil or closely cropped vegetation are common; less than one-half of the potential plant stubble height remaining.	Disruption of stream bank vegetation is very high; vegetation has been removed to 2 inches or less in average stubble height.
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
12. RIPARIAN VEGETATION ZONE WIDTH (LEAST BUFFERED SIDE)	Width of riparian zone is greater than 18 m; human activities (i.e.; parking lots, roadbeds, clearcuts, lawns, or crops) have not impacted this zone.	Zone width is between 12 and 18 m; human activities have only minimally impacted this zone.	Zone width is between 6 and 12 m; human activities have impacted the zone a great deal.	Width of zone is less than 6 m; little or no riparian vegetation due to man-induced activities.
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

Reviewed by (initial): _____

RAPID HABITAT ASSESSMENT FORM: GLIDE/POOL PREVALENCE - STREAMS

SITE NAME: DATE: / / VISIT: G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ TEAM ID (X): G1 G2 G3 G4 G5 G6 G7

TOTAL		CATEGORY			
HABITAT PARAMETER		OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
1. INSTREAM COVER		Greater than 50% mix of snags, submerged logs, undercut banks, or other stable habitat; rubble or gravel may be present.	30 to 50% mix of stable habitat; adequate habitat for maintenance of populations.	10 to 30% mix of stable habitat; habitat availability is less than desirable.	Less than 10% stable habitat; lack of habitat is obvious.
SCORE:		20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
2. EPIFAUNAL SUBSTRATE		Preferred benthic substrate (to be sampled) is abundant throughout stream site and at a stage to allow for full colonization potential (i.e.; logs and snags that are not new fall and not transient.	Substrate is common but is not prevalent nor well-suited for full colonization potential.	Substrate frequently disturbed or removed.	Substrate is unstable or lacking.
SCORE:		20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
3. POOL SUBSTRATE CHARACTERIZATION		Mixture of substrate materials, with gravel and firm sand prevalent; root mats and submerged vegetation are common.	Mixture of soft sand, mud, or clay; mud may be dominant; some root mats and submerged vegetation are present	All mud or clay or sand bottom; little or no root mat; no submerged vegetation.	Hard-pan clay or bedrock; no root mat or vegetation.
SCORE:		20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
4. POOL VARIABILITY		Even mix of large-shallow, large-deep, small-shallow, and small-deep pools are present.	The majority of pools are large and deep; very few shallow.	Shallow pools much more prevalent than deep pools.	Majority of pools are small-shallow or pools are absent.
SCORE:		20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
5. CHANNEL ALTERATION		No channelization of dredging present.	Some channelization is present, usually in areas of bridge abutments; evidence of past channelization, i.e.; dredging (greater than past	New embankments are present on both banks; channelization may be extensive, usually in urban areas or drainage areas of	Extensive channelization; banks shored with gabion or cement; heavily urbanized areas; instream habitat greatly altered or removed
SCORE:		20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
6. SEDIMENT DEPOSITION		Less than 20% of the bottom is affected; minor accumulation of fine and coarse material at snags and submerged	20 to 50% affected; moderate accumulation; substantial sediment movement only during major storm events; some new increase in bar	50 to 80% affected; major deposition; pools shallow and heavily silted; embankments may be present on both banks;	Channelized; mud, silt, and/or sand in braided or non-braided channels; pools almost absent due to deposition.
SCORE:		20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

Reviewed by (initial): _____

RAPID HABITAT ASSESSMENT FORM: GLIDE/POOL- STREAMS (continued)

SITE NAME: DATE: / / VISIT: G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ TEAM ID (X): G1 G2 G3 G4 G5 G6 G7

HABITAT PARAMETER	CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
7. CHANNEL SINUOSITY	The bends in the stream increase the stream length 3 to 4 times longer than if it was in a straight line.	The bends in the stream increase the stream length 2 to 3 times longer than if it was in a straight line.	The bends in the stream increase the stream length between 1 and 2 times longer than if it was in a straight line.	Channel is straight; waterway has been channelized for a long distance.
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
8. CHANNEL FLOW STATUS	Water reaches the base of both lower banks and a minimal amount of channel substrate is exposed.	Water fills more than 75% of the available channel; or less than 25% of the channel substrate is exposed.	Water fills 25 to 75% of the available channel and/or riffle substrates are mostly exposed.	Very little water in channel, and mostly present as standing pools.
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
9. CONDITION OF BANKS	Banks stable; no evidence of erosion or bank failure.	Banks moderately stable; infrequent, small areas of erosion mostly healed over.	Moderately unstable; up to 60% of banks in reach have areas of erosion.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; side slopes 60 to 100% of bank has erosional
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
10. BANK VEGETATIVE PROTECTION	Over 90% of the stream bank surfaces is covered by vegetation.	70 to 90% of the stream bank surfaces is covered by vegetation.	50 to 70% of the stream bank surfaces is covered by vegetation.	Less than 50% of the stream bank surfaces are covered by vegetation.
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
11. GRAZING OR OTHER DISRUPTIVE PRESSURE	Vegetative disruption minimal or not evident; almost all plants are allowed to grow naturally.	Disruption is evident but is not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	Disruption is obvious; patches of bare soil or closely cropped vegetation are common; less than one-half of the potential plant stubble height remaining.	Disruption of stream bank vegetation is very high; vegetation has been removed to 2 inches or less in average stubble height.
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
12. RIPARIAN VEGETATIOUN ZONE WIDTH (LEAST BUFFERED SIDE)	Width of riparian zone is greater than 18 meters; human activities (i.e.; parking lots, roadbeds, clearcuts, lawns, or crops) have not impacted this zone.	Width of riparian zone is between 12 and 18 meters; human activities have only minimally impacted this zone.	Width of riparian zone is between 6 and 12 meters; human activities have impacted the zone a great deal.	Width of riparian zone is less than 6 meters; little or no riparian vegetation due to human activities.
SCORE:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

Reviewed by (initial): _____

ASSESSMENT FORM - STREAMS/RIVERS

SITE NAME: **DATE: / / VISIT:** G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ **TEAM ID (X):** G1 G2 G3 G4 G5 G6 G7 G8

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

RESIDENTIAL				RECREATIONAL				AGRICULTURAL				INDUSTRIAL				STREAM MANAGEMENT			
L	M	H		L	M	H		L	M	H		L	M	H		L	M	H	
			Residences				Parks, Campgrounds				Cropland				Industrial				Liming
			Maintained Lawns				Primitive Parks, Camping				Pasture				Mines/Quarries				Drinking Water Treatment
			Construction				Trash/Litter				Livestock Use				Oil/Gas Wells				Angling Pressure
			Pipes, Drains				Surface Films, Scums, or Slicks				Orchards				Power Plants				Dredging
			Dumping								Poultry				Logging				Channelization
			Roads								Irrigation Pumps				Evidence of Fire				Water Level Fluctuations
			Bridge/Culverts												Odors				Fish Stocking
															Commercial				Dams

REACH CHARACTERISTICS (percent of reach)

Forest	G Rare (< 5%)	G Sparse (5 to 25%)	G Moderate (25 to 75%)	G Extensive (> 75%)
Shrub	G Rare (< 5%)	G Sparse (5 to 25%)	G Moderate (25 to 75%)	G Extensive (> 75%)
Grass	G Rare (< 5%)	G Sparse (5 to 25%)	G Moderate (25 to 75%)	G Extensive (> 75%)
Wetland	G Rare (< 5%)	G Sparse (5 to 25%)	G Moderate (25 to 75%)	G Extensive (> 75%)
Bare Ground	G Rare (< 5%)	G Sparse (5 to 25%)	G Moderate (25 to 75%)	G Extensive (> 75%)
Macrophytes	G Rare (< 5%)	G Sparse (5 to 25%)	G Moderate (25 to 75%)	G Extensive (> 75%)
Agriculture - Row Crop	G Rare (< 5%)	G Sparse (5 to 25%)	G Moderate (25 to 75%)	G Extensive (> 75%)
Agriculture - Grazing	G Rare (< 5%)	G Sparse (5 to 25%)	G Moderate (25 to 75%)	G Extensive (> 75%)
Logging	G Rare (< 5%)	G Sparse (5 to 25%)	G Moderate (25 to 75%)	G Extensive (> 75%)
Development (Residential & Urban)	G Rare (< 5%)	G Sparse (5 to 25%)	G Moderate (25 to 75%)	G Extensive (> 75%)
Water Clarity	G Clear	G Murky	G Highly Turbid	G Storm Influenced

WATERBODY CHARACTER (X ONE)

Pristine	G 5	G 4	G 3	G 2	G 1	Highly
Appealing	G 5	G 4	G 3	G 2	G 1	Unappealing

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

LOCAL ANECDOTAL INFORMATION:

Reviewed by (initial): _____

ASSESSMENT FORM - STREAMS/RIVERS (continued)

SITE NAME: **DATE:** / / **VISIT:** G1 G2 ___

SITE ID: ___ ___ ___ ___ ___ ___ - ___ ___ ___ ___ ___ **TEAM ID (X):** G1 G2 G3 G4 G5 G6 G7 G8

APPENDIX D

SPECIES CODES FOR AQUATIC VERTEBRATES: MID-ATLANTIC REGION

The following table contains the unique 6-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 region. Generally, the species code is composed of the first four letters of the genus plus the first two 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.

Similar lists have been compiled for EMAP-related studies occurring in regions other than the Mid-Atlantic. Information regarding the availability of species codes and associated information may be obtained through the EMAP-Surface Waters Technical Director, c/o U.S. EPA, 200 SW 35th St., Corvallis, OR 97333.

SPECIES LIST AND STATUS FOR MID-ATLANTIC REGION

CODE	Latin Name	Common Name
ICHTBD	*Ichthyomyzon bdellium*	Ohio Lamprey
ICHTFO	*Ichthyomyzon fossor*	Northern Brook Lamprey
ICHTGR	*Ichthyomyzon greeleyi*	Mountain Brook Lamprey
ICHTUN	*Ichthyomyzon unicuspis*	Silver Lamprey
LAMPAE	*Lampetra aepyptera*	Least brook lamprey
LAMPAP	*Lampetra appendix*	American brook lamprey
PETRMA	*Petromyzon marinus*	Sea lamprey
LAMPZZ		Unknown lamprey
ACIPBR	*Acipenser brevirostrum*	Atlantic sturgeon
ACIPFU	*Acipenser fulvescens*	Lake sturgeon
SCAPPL	*Scaphirhynchus platorynchus*	Shovelnose sturgeon
POLYSP	*Polyodon spatula*	Paddlefish
LEPIOC	*Lepisosteus oculatus*	Spotted gar
LEPIOS	*Lepisosteus osseus*	Longnose gar
LEPIPL	*Lepisosteus platostomus*	Shortnose gar
AMIACA	*Amia calva*	Bowfin
ANGURO	*Anguilla rostrata*	American eel
APHRSA	*Aphredoderus sayanus*	Pirate perch
LABISI	*Labidesthes sicculus*	Brook silversides
MENIBE	*Menidia beryllina*	Inland silverside
CARPCA	*Carpiodes carpio*	River carpsucker
CARPCY	*Carpiodes cyprinus*	Quillback
CARPVE	*Carpiodes velifer*	Highfin carpsucker
CATOCA	*Catostomus catostomus*	Longnose sucker
CATOCO	*Catostomus commersoni*	White sucker
CYCLEL	*Cycleptus elongatus*	Blue sucker
ERIMOB	*Erimyzon oblongus*	Creek chubsucker
ERIMSU	*Erimyzon sucetia*	Lake chubsucker
HYPENI	*Hypentelium nigricans*	Northern hogsucker
HYPERO	*Hypentelium roanokense*	Roanoke hogsucker

CODE	Latin Name	Common Name
ICTIBU	*Ictiobus bubalus*	Smallmouth buffalo
ICTICY	*Ictiobus cyprinellus*	Bigmouth buffalo
MINYME	*Minytrema melanops*	Spotted sucker
MOXOAN	*Moxostoma anisurum*	Silver redhorse
MOXOAR	*Moxostoma ariommum*	Bigeye jumprock
MOXOCA	*Moxostoma carinatum*	River redhorse
MOXOCE	*Moxostoma cervinum*	Black jumprock
MOXODU	*Moxostoma duquesnei*	Black redhorse
MOXOER	*Moxostoma erythrurum*	Golden redhorse
MOXOHA	*Moxostoma hamiltoni*	Rustyside sucker
MOXOMA	*Moxostoma macrolepidotum*	Shorthead redhorse
MOXOPA	*Moxostoma pappillosum*	V-lip redhorse
MOXORH	*Moxostoma rhothoecum*	Torrent sucker
MOXORO	*Moxostoma robustum*	Smallfin Sucker
MOXOVA	*Moxostoma valenciennesi*	Greater Redhorse
CATOZZ		Unknown catostomid
ACANPO	*Ancartharcus pomotis*	Mud sunfish
AMBLCA	*Ambloplites cavifrons*	Roanoke rockbass
AMBLRU	*Ambloplites rupestris*	Rockbass
ARCHIN	*Archoplites interruptus*	Sacramento perch
CENTMA	*Centrarchus macropterus*	Flier
ENNECH	*Enneacanthus chaetodon*	Blackbanded sunfish
ENNEGL	*Enneacanthus gloriosus*	Bluespotted sunfish
ENNEOB	*Enneacanthus obesus*	Banded sunfish
LEPOAU	*Lepomis auritus*	Redbreast sunfish
LEPOCY	*Lepomis cyanellus*	Green sunfish
LEPOGI	*Lepomis gibbosus*	Pumpkinseed
LEPOGU	*Lepomis gulosus*	Warmouth
LEPOMA	*Lepomis macrochirus*	Bluegill
LEPOME	*Lepomis megalotis*	Longear sunfish
LEPOMI	*Lepomis microlophus*	Redear sunfish

CODE	Latin Name	Common Name
MICRDO	*Micropterus dolomieu*	Smallmouth bass
MICRPU	*Micropterus punctulatus*	Spotted bass
MICRSA	*Micropterus salmoides*	Largemouth bass
POMOAN	*Pomoxis annularis*	White crappie
POMONI	*Pomoxis nigromaculatus*	Black crappie
CENTZZ		Unknown centrarcid
ALOSCH	*Alosa chrysochloris*	Skipjack herring
ALOSPS	*Alosa pseudoharengus*	Alewife (landlocked)
ALOSSA	*Alosa sapidissima*	American shad
DOROCE	*Dorosoma cepedianum*	Gizzard shad
COTTBA	*Cottus bairdi*	Mottled sculpin
COTTBL	*Cottus baileyi*	Black sculpin
COTTCA	*Cottus carolinae*	Banded sculpin
COTTCF	*Cottus confusus*	Shorthead sculpin
COTTCO	*Cottus cognatus*	Slimy sculpin
COTTGI	*Cottus girardi*	Potomac sculpin
COTTGU	*Cottus gulosus*	Riffle sculpin
COTTMA	*Cottus marginatus*	Margined sculpin
COTTRI	*Cottus ricei*	Spoonhead sculpin
COTTTE	*Cottus tenuis*	Slender Sculpin
COTTZZ		Unknown cottid
CAMPAN	*Campostoma anomalum*	Stoneroller
CLINEL	*Clinostomus elongatus*	Redside dace
CLINFU	*Clinostomus funduloides*	Rosyside dace
COUEPL	*Couesius plumbeus*	Lake chub
CYPRAN	*Cyprinella analostana*	Satinfin shiner
CYPRGA	*Cyprinella galactura*	Whitetail shiner
CYPRMO	*Cyprinella monacha*	Spotfin chub
CYPRSP	*Cyprinella spiloptera*	Spotfin shiner
CYPRWH	*Cyprinella whipplei*	Steelcolor shiner
ERIMCA	*Erimystax cahni*	Slender chub

CODE	Latin Name	Common Name
ERIMDI	*Erimystax dissimilis*	Streamline chub
ERIMIN	*Erimystax insignis*	Blotched chub
ERIMXP	*Erimystax x-punctatus*	Gravel chub
EXOGLA	*Exoglossum laurae*	Tonguetied minnow
EXOGMA	*Exoglossum maxillingua*	Cutlips minnow
HYBOHA	*Hybognathus hankinsoni*	Brassy minnow
HYBORE	*Hybognathus regius*	Eastern silvery minnow
LUXIAL	*Luxilus albeolus*	White shiner
LUXICC	*Luxilus coccogenis*	Warpaint shiner
LUXICE	*Luxilus cerasinus*	Crescent shiner
LUXICH	*Luxilus chrysocephalus*	Striped shiner
LUXICR	*Luxilus cornutus*	Common shiner
LYTHAR	*Lythurus ardens*	Rosefin shiner
LYTHLI	*Lythurus lirus*	Mountain shiner
LYTHUM	*Lythurus umbratilus*	Redfin shiner
MACRAE	*Macrhybopsis aestivalis*	Speckled chub
MACRST	*Macrhybopsis storeriana*	Silver chub
MARGMA	*Margariscus margarita*	Pearl dace
NOCOBI	*Nocomis biguttatus*	Horneyhead chub
NOCOLE	*Nocomis leptocephalus*	Bluehead chub
NOCOMI	*Nocomis micropogon*	River chub
NOCORA	*Nocomis raneyi*	Bull chub
NOTECR	*Notemigonus crysoleucas*	Golden shiner
NOTRAB	*Notropis amblops*	Bigeye chub
NOTRAL	*Notropis alborus*	Whitemouth shiner
NOTRAN	*Notropis anogenus*	Pugnose shiner
NOTRAO	*Notropis amoenis*	Comely shiner
NOTRAR	*Notropis ariommus*	Popeye shiner
NOTRAT	*Notropis atherinoides*	Emerald shiner
NOTRBC	*Notropis buccatus*	Silverjaw minnow
NOTRBH	*Notropis buchanani*	Ghost shiner

CODE	Latin Name	Common Name
NOTRBI	*Notropis bifrenatus*	Bridled shiner
NOTRBL	*Notropis blennius*	River shiner
NOTRCH	*Notropis chalybaeus*	Ironcolor shiner
NOTRDO	*Notropis dorsalis*	Bigmouth shiner
NOTRHD	*Notropis heterodon*	Blackchin shiner
NOTRHL	*Notropis heterolepis*	Blacknose shiner
NOTRHU	*Notropis hudsonius*	Spottail shiner
NOTRLE	*Notropis leuciodus*	Tennessee shiner
NOTRPH	*Notropis photogenis*	Silver shiner
NOTRPR	*Notropis procne*	Swallowtail shiner
NOTRRR	*Notropis rubricroceus*	Saffron shiner
NOTRRU	*Notropis rubellus*	Rosyface Shiner
NOTRSC	*Notropis scabriceps*	New River Shiner
NOTRSE	*Notropis semperasper*	Roughhead Shiner
NOTRSP	*Notropis spectrunculus*	Mirror Shiner
NOTRST	*Notropis stramineus*	Sand Shiner
NOTRTE	*Notropis telescopus*	Telescope Shiner
NOTRVO	*Notropis volucellus*	Mimic Shiner
OPSOEM	*Opsopoeodus emiliae*	Pugnose minnow
PHENCR	*Phenacobius crassilabrum*	Fatlips minnow
PHENMI	*Phenacobius mirabilis*	Suckermouth minnow
PHENTE	*Phenacobius teretulus*	Kanawha minnow
PHENUR	*Phenacobius uranops*	Stargazing minnow
PHOXCU	*Phoxinus cumberlandensis*	Blackside dace
PHOXEO	*Phoxinus eos*	Northern redbelly dace
PHOXER	*Phoxinus erythrogaster*	Southern redbelly dace
PHOXNE	*Phoxinus neogaeus*	Finescale dace
PHOXOR	*Phoxinus oreas*	Mountain redbelly dace
PHOXTE	*Phoxinus tennesseensis*	Tennessee dace
PIMENO	*Pimephales notatus*	Bluntnose minnow
PIMEPR	*Pimephales promelas*	Fathead minnow

CODE	Latin Name	Common Name
PIMEVI	*Pimephales vigilax*	Bullhead minnow
RHINAT	*Rhinichthys atratulus*	Blacknose dace
RHINBO	*Rhinichthys bowersi*	Cheat minnow
RHINCA	*Rhinichthys cataractae*	Longnose dace
RHINOS	*Rhinichthys osculus*	Speckled dace
SCARER	*Scardinius erythrophthal.*	Rudd
SEMOAT	*Semotilus atromaculatus*	Creek chub
SEMOCO	*Semotilus corporalis*	Fallfish
CARAAU	*Carassius auratus*	Goldfish
CYPRCA	*Cyprinus carpio*	Common carp
CTENID	*Ctenopharyngodon idella*	Grass carp
LEUCID	*Leuciscus idus*	Ide
TINCTI	*Tinca tinca*	Tench
CYPRZZ		Unknown cyprinid
FUNDCA	*Fundulus catenatus*	Northern studfish
FUNDDI	*Fundulus diaphanus*	Banded killifish
FUNDRA	*Fundulus rathbuni*	Speckled killifish
LUCAPA	*Lucania parva*	Rainwater killifish
ESOXAM	*Esox americanus*	Redfin/grass pickerel
ESOXLM	*Esox lucius x masq.*	Tiger muskellunge
ESOXLU	*Esox lucius*	Northern pike
ESOXMA	*Esox masquinongy*	Muskellunge
ESOXNI	*Esox niger*	Chain pickerel
LOTALO	*Lota lota*	Burbot
APELQU	*Apeltes quadracus*	Fourspine stickleback
CULEIN	*Culea inconstans*	Brook stickleback
GASTAC	*Gasterosteus aculeatus*	Threespine stickleback
PUNGPU	*Pungitius pungitius*	Ninespine stickleback
HIODAL	*Hiodon alosoides*	Goldeye
HIODTE	*Hiodon tergisus*	Mooneye
AMEIBR	*Ameiurus brunneus*	Snail bullhead

CODE	Latin Name	Common Name
AMEICA	*Ameiurus catus*	White catfish
AMEIME	*Ameiurus melas*	Black bullhead
AMEINA	*Ameiurus natalis*	Yellow bullhead
AMEINE	*Ameiurus nebulosus*	Brown bullhead
AMEIPL	*Ameiurus platycephalus*	Flat bullhead
ICTAPU	*Ictalurus punctatus*	Channel catfish
NOTUEL	*Noturus eleutherus*	Mountain madtom
NOTUEX	*Noturus exilis*	Slender madtom
NOTUFI	*Noturus flavipinnis*	Yellowfin madtom
NOTUFU	*Noturus flavus*	Stonecat
NOTUGI	*Noturus gilberti*	Orangefin madtom
NOTUGY	*Noturus gyrinus*	Tadpole madtom
NOTUIN	*Noturus insignis*	Margined madtom
NOTUMI	*Noturus miurus*	Brindled madtom
NOTUST	*Noturus stigmosus*	Northern madtom
PYLOOL	*Pylodictis olivaris*	Flathead catfish
MOROAM	*Morone americana*	White perch
MOROCH	*Morone chrysops*	White bass
MOROSA	*Morone saxatilis*	Striped bass
AMMOAS	*Ammocrypta asprella*	Crystal darter
AMMOPE	*Ammocrypta pellucida*	Eastern sand darter
ETHEAC	*Etheostoma acuticeps*	Sharphead darter
ETHEBL	*Etheostoma blennioides*	Greenside darter
ETHECE	*Etheostoma caeruleum*	Rainbow darter
ETHECI	*Etheostoma cinereum*	Ashy darter
ETHECM	*Etheostoma camurum*	Bluebreast darter
ETHEEX	*Etheostoma exile*	Iowa darter
ETHEFL	*Etheostoma flabellare*	Fantail darter
ETHEFU	*Etheostoma fusiforme*	Swamp darter
ETHEJE	*Etheostoma jessiae*	Blueside darter
ETHEKA	*Etheostoma kanawhae*	Kanawha darter

CODE	Latin Name	Common Name
ETHEKE	*Etheostoma kennecotti*	Stripetail darter
ETHELO	*Etheostoma longimanum*	Longfin darter
ETHEMA	*Etheostoma maculatum*	Spotted darter
ETHENI	*Etheostoma nigrum*	Johnny darter
ETHEOL	*Etheostoma olmstedi*	Tesselated darter
ETHEOS	*Etheostoma osburni*	Candy darter
ETHEPO	*Etheostoma podostemone*	Riverweed darter
ETHERU	*Etheostoma rufilineatum*	Redline darter
ETHESI	*Etheostoma simoterum*	Snubnose darter
ETHEST	*Etheostoma stigmaeum*	Speckled darter
ETHESW	*Etheostoma swannanoa*	Swannanoa darter
ETHETI	*Etheostoma tippecanoe*	Tippecanoe darter
ETHEUN	*Etheostoma unknown01*	Duskytail darter
ETHEVA	*Etheostoma variatum*	Variegate darter
ETHEVU	*Etheostoma vulneratum*	Wounded darter
ETHEZO	*Etheostoma zonale*	Banded darter
PERCAU	*Percina aurantiaca*	Tangerine darter
PERCBU	*Percina burtoni*	Blotchside logperch
PERCCA	*Percina caprodes*	Logperch
PERCCO	*Percina copelandi*	Channel darter
PERCEV	*Percina evides*	Gilt darter
PERCFL	*Perca flavescens*	Yellow perch
PERCGY	*Percina gymnocephala*	Appalachia darter
PERCMR	*Percina macrocephala*	Longhead darter
PERCMU	*Percina maculata*	Blackside darter
PERCNO	*Percina notogramma*	Stripeback darter
PERCOX	*Percina oxyrhynchus*	Sharpnose darter
PERCPE	*Percina peltata*	Shield darter
PERCRE	*Percina rex*	Roanoke logperch
PERCRO	*Percina roanoka*	Roanoke darter
PERCSC	*Percina sciera*	Dusky darter

CODE	Latin Name	Common Name
PERCSH	*Percina shumardi*	River darter
STIZCA	*Stizostedion canadense*	Sauger
STIZVI	*Stizostedion vitreum*	Walleye
PERCOM	*Percopsis omiscomaycus*	Troutperch
GAMBAF	*Gambusia affinis*	Western mosquitofish
GAMBHO	*Gambusia holbrooki*	Eastern mosquitofish
ONCOMY	*Oncorhynchus mykiss*	Rainbow trout
SALMTR	*Salmo trutta*	Brown trout
SALVFO	*Salvelinus fontinalis*	Brook trout
SALVNA	*Salvelinus namaycush*	Lake trout
SALMZZ		Unknown salmonid
APLOGR	*Aplodinotus grunniens*	Freshwater drum
UMBRLI	*Umbra limi*	Central mudminnow
UMBRPY	*Umbra pygmaea*	Eastern mudminnow

APPENDIX E
MODIFIED PROTOCOL FOR COLLECTING BENTHIC MACROINVERTEBRATES

by
Donald J. Klemm[1] and David V. Peck[2]

Field procedures described here are modified from those developed by the Oregon Department of Environmental Quality (Oregon Dept. of Environmental Quality, 1997), and the Washington Department of Ecology (Washington Dept. of Ecology, 1997). These procedures were implemented in an EMAP study of wadeable streams in Oregon in 1997, and the EPA Region 10 R-EMAP study in 1996-1997. Modifications to the basic EMAP protocol (Section 11 of the EMAP field operations manual for streams) were desired to maximize the comparability of EMAP results with both the R-EMAP project results, and with other data both State agencies routinely collect as part of their respective monitoring programs.

Within the defined sampling reach of 150 to 500 m, benthic invertebrate samples are collected from two principal macrohabitat types, erosional (operationally termed “riffle”) and depositional (operationally termed “pool”). Riffle macrohabitats include low-gradient areas that are generally more shallow than pools. Many riffles exhibit surface turbulence associated with increased velocity and shallow water depth over gravel or cobble beds. However, the riffle classification also includes shallow areas without surface turbulence such as glides. Pool macrohabitats include areas of slow, deep water with low gradient. They are typically created by scour adjacent to obstructions or impoundments of water behind channel blockages and hydraulic controls such as logjams, bedforms, or beaver dams.

Individual kick net samples are collected from up to five points within each macrohabitat type, spaced throughout the sampling reach. Individual kick net samples collected from each macrohabitat type are processed and composited into a single sample for the stream. Thus for each stream, there will be two composite samples, one for riffles and one

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

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

for pools. Each composite sample is contained in a 500-mL or 1-L plastic screw-top jar, and preserved with 95% ethanol to a final concentration of 70% ethanol.

The sampling protocols described here differ from those presented in Section 11 of the EMAP streams field operations manual in that sampling points are allocated evenly across the two major macrohabitat types, rather than by sampling at predefined points located on the transects established for physical habitat characterization. Differences from R-EMAP protocols used by Oregon DEQ (1997)and Washington Department of Ecology (1997) include not determining the depth at each sampling point and not determining the substrate particle distribution at each sampling points.

E.1 SAMPLE COLLECTION

E.1.1 Selection of Sampling Points

Table E-1 presents the procedure for selecting individual sampling points within the two major macrohabitat types (riffle and pool). Note that in some stream reaches, one macrohabitat type will predominate to the extent that the other type is not sampled. There may also be stream reaches where two kick net samples are collected from a single macrohabitat unit. It is also permissable to sample a short distance beyond the upstream end of the sample reach in order to obtain the desired number (5) of macrohabitat units of each type.

E.1.2 Collection of Kick Net Samples

The kick net is designed to obtain a qualitative and semi-quantitative sample of benthic macroinvertebrates from a variety of substrates in streams. A modified USGS kick net (Wildco # 425-J50-595) is used. This is the same net as is described in Section 10 of the EMAP field operations manual for streams for EMAP. Modifications from the standard configuration include the net mesh size (600 :m), the length of the net (61 cm or 24 in.) and the type of bag (tapering closed bag). The frame dimensions of the net are 30.48 cm (12 in.) high and 50.8 cm (20 in.) wide. The style and dimensions of this net differ from that used by Oregon DEQ and Washington Department of Ecology, who use a smaller net of a D-frame configuration. However, mesh sizes are the same.

Procedures for collecting a point sample using the kick net from riffle and pool macrohabitat units are presented in Tables E-2 and E-3, respectively. At each sampling

TABLE E-1. LOCATING SAMPLING POINTS FOR KICK NET SAMPLES: WADEABLE STREAMS

1. Before sampling, survey the stream reach to visually estimate the number of pool and riffle macrohabitat "units" contained in the defined stream reach. To be considered as a unit, the length of a stream occupied by a particular macrohabitat type unit should be at least equal to the stream's average wetted width estimate used to define the length of stream reach.

   A. Do not sample poorly represented habitats. If the reach contains < 2 macrohabitat units of a given type, then do not sample that macrohabitat type. If only one macrohabitat unit occurs in the defined reach but, more are present within 100 meters upstream, sample those as they were part of the reach.

   B. If the reach contains 3 or 4 macrohabitat units of a given type, then randomly select those macrohabitat unit(s) from which to collect a second kick net sample to bring the total number of kick net samples for the macrohabitat type to five.

   C. If the number of units is greater than five of either, skip one or more habitat units at random as you work upstream.

2. Begin sampling at the most downstream unit, and sample units as they are encountered to minimize instream disturbance. This will require separate containers for pool and riffle samples.

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

4. Visually lay out the core area of the unit sampled into 9 equal quadrats (i.e., a 3 × 3 grid). For each macrohabitat type, select a quadrat for sampling as follows:
   First unit: Lower right quadrat
   Second unit: Center quadrat
   Third unit: Upper left quadrat
   Fourth unit: Lower left quadrat
   Fifth unit: Upper right quadrat.

5. Collect the kick sample in the center of the selected quadrat, following the protocol for the type of macrohabitat unit.

6. If a second sample is required from a single macrohabitat unit, select a new quadrat.

TABLE E-2. COLLECTING A KICK NET SAMPLE FROM WADEABLE STREAMS: RIFFLE MACROHABITATS

1. Locate the sampling point within the macrohabitat unit as described in Table 1.

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

3. Hold the sampler firmly in position on the substrate. Define a quadrat immediately upstream from the mouth of the net having a width equal to the width of the net frame and a total area = 0.5 m^2. If the kick net is oriented normally, the length of the quadrat = 1 m (approx. equal to 2 times the width of the net [0.5 m]). If the net is rotated so the short side is against the substrate, the length of the quadrat = 1.67 m.

4. Lightly kick the substrate throughout the quadrat. Start at the upstream end and work toward the net.

5. Hold the net in place with the knees and pick up any loose rocks in the quadrat and rub off organisms so that they are washed into the net. With a small brush dislodge organisms from the rocks into the net. Scrub all rocks that are golf ball-sized or larger and which are over halfway into the quadrat. Large rocks that are less than halfway into the sampling area are pushed aside.

6. Keep holding the sampler securely in position and kick through the quadrat again, this time vigorously, for 20 seconds.

7. Pull the net up out of the water. Immerse the net in the stream several times to remove fine sediments and to concentrate organisms at the end of the net. Avoid having any water or material enter the mouth of the net during this operation.

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

9. Thoroughly rinse the net before proceeding to the next macrohabitat unit.

10. Repeat steps 1-9 at subsequent riffle macrohabitat units until 5 kick samples have been collected and placed into the "riffle" bucket.

TABLE E-3. COLLECTING A KICK NET SAMPLE FROM WADEABLE STREAMS: POOL MACROHABITATS

1. Locate the sampling point within the macrohabitat unit as described in table 1.

2. Define a sampling area as a quadrat having a width equal to the width of the net frame and a total area = 0.5 m^2. If the kick net is oriented normally, the length of the quadrat = 1 m (approx. equal to 2 times the width of the net [0.5 m]). If the net is rotated so the short side is against the substrate, the length of the quadrat = 1.67 m.

3. Inspect the quadrat for heavy organisms such as mussels and snails. Hand pick any of these large organisms and place them into the sieve bucket or plastic bucket marked "pool".

4. Kick vigorously with the feet within the quadrat for 10 seconds. Then drag the net repeatedly through the disturbed area just above the bottom. Keep moving the net to prevent organisms from escaping. Continue this for 1 minute.

5. Pull the net up out of the water. Immerse the net into the stream several times to remove fine sediments and to concentrate organisms at the end of the net. Avoid having any water or material enter the mouth of the net during this operation.

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

7. Thoroughly rinse the net before proceeding to the next macrohabitat unit to prevent cross-contamination of riffle and pool samples.

8. Repeat steps 1-7 at subsequent pool macrohabitat units until 5 kick samples have been collected and placed into the "pool" sieve bucket.

point, a quadrat having a total area of 0.5 m^2 is sampled. The dimensions of the quadrat will vary depending on how the kick net must be oriented against the substrate. In narrow streams, the net may have to be rotated so that the narrow side is against the stream bottom. Riffle and pool samples are kept in separate containers. Note that in pool units, the substrate is first disturbed, and the net is dragged through the disturbed area just above the substrate. Because units are sampled in the order they are encountered, it is very important to rinse the kick net thoroughly between samples to avoid carryover and possible cross-contamination of riffle and pool samples.

E.2 Sample Processing

The procedure for processing kick net samples is presented in Table E-4; the procedure is identical for riffle and pool samples. Process one sample at a time to avoid mixing riffle and pool samples in the same container. Reduce the amount of residue in each composite sample as much as possible without losing organisms. However, if there is a sizable quantity of material remaining, distribute the sample into additional containers to ensure proper preservation. A sample jar should not be more than half-full of material. Modified sample labels are shown in Figure E-1, and the modified Sample Collection Form is presented in Figure E-2.

E.3. LITERATURE CITED

Oregon Department of Environmental Quality. 1997. *Biological Assessment of Wadeable Streams of the Upper Deschutes River Basin: Quality Assurance Project Plan.*

Washington Department of Ecology. 1997. *Biological Assessment of Wadeable Streams of the Chehalis River Basin: Quality Assurance Project Plan.*

TABLE E-4. PROCESSING KICK NET SAMPLES: WADEABLE STREAMS

1. Fill out a sample label for the riffle composite samples. Attach a label to a 1-gallon plastic bag with a zipper-type closure. If the sample contains a large volume of material, complete a sample label for additional containers and attach it to a second bag. Make sure the barcode numbers on each label agree.

2. Hand pick large organisms from the bucket containing the composited riffle kick net samples and place them into the appropriately labeled plastic bag.

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 the labeled plastic bag. If necessary, distribute the sample among two or more labeled bags. Rinse residue from the bucket into the plastic bag using a wash bottle and a **small** volume of water.

5. Place each bag inside a second bag.

6. Add 95% ethanol to each labeled bag in a volume which is equal to the volume of the sample.

7. Rinse the bucket well to eliminate any residue.

8. Repeat Steps 1-7 for the pool composite sample.

9. Complete the Sample Collection Form. Record the barcode number of each composite sample (riffle and pool), and the habitat type from the sample label. If more than one container was required for a sample, record the number of containers on the collection form. Also, note any peculiarities associated with a particular sample by using a flag code and/or a written comment on the collection form.

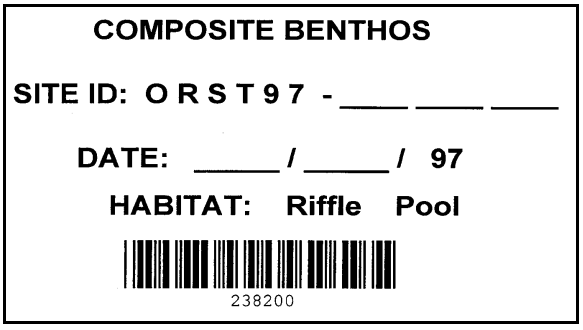


Figure E-1. Modified sample labels.

Reviewed by (initial): _____

SAMPLE COLLECTION FORM - STREAMS

SITE NAME: DATE: / / 97 VISIT: ☐1 ☐2 ___

SITE ID: O R S T 9 7 - ___ ___ ___ TEAM ID (X): ☐1 ☐2 ☐3 ☐4 ☐5 ☐6 ☐7 ☐8

COMPOSITE BENTHOS SAMPLES

SAMPLE ID (BARCODE)	HABITAT (X ONE) R	HABITAT (X ONE) P	NO. OF JARS	FLAG	COMMENTS
___ ___ ___ ___ ___ ___					
___ ___ ___ ___ ___ ___					

STATION	A	B	C	D	E	F	G	H	I	J	K
RIFFLE OR POOL - (X ONE) -		☐R ☐P	☐R ☐P	☐R ☐P	☐R ☐P	☐R ☐P	☐R ☐P	☐R ☐P	☐R ☐P	☐R ☐P	
LEFT, CENTER, OR RIGHT - (X ONE) -		☐L ☐C ☐R	☐L ☐C ☐R	☐L ☐C ☐R	☐L ☐C ☐R	☐L ☐C ☐R	☐L ☐C ☐R	☐L ☐C ☐R	☐L ☐C ☐R	☐L ☐C ☐R	

COMPOSITE PERIPHYTON SAMPLES		HABITAT TYPE (X) →	☐ RIFFLE ☐ POOL ☐ OTHER
SAMPLE ID (BARCODE) →	___ ___ ___ ___ ___ ___	COMPOSITE VOLUME →	___ ___ ___ ___ mL

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

COMPOSITE PERIPHYTON SAMPLES		HABITAT TYPE (X) →	☐ RIFFLE ☐ POOL ☐ OTHER
SAMPLE ID (BARCODE) →	___ ___ ___ ___ ___ ___	COMPOSITE VOLUME →	___ ___ ___ ___ mL

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

COMMENTS:

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

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Figure E-2. Modified Sample Collection Form.