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ENVIRONMENTAL MONITORING AND ASSESSMENT PROGRAM - SURFACE WATERS:

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

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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

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FOREWORD

Environmental measurements are required to determine the quality of ambient waters and the character of waste effluents. The Environmental Monitoring Systems Laboratory - Cincinnati (EMSL-Cincinnati) conducts research to:

- o Develop and evaluate methods to identify and measure the concentration of chemical pollutants in drinking waters, surface waters, groundwaters, wastewaters, sediments, sludges, and solid wastes.

- o Investigate and evaluate methods for the identification and measurement of viruses, bacteria and other microbiological organisms in aqueous samples and to determine the response of aquatic organisms to water quality.

- o Perform ecological assessments and measure the toxicity of pollutants to representative species of aquatic organisms and determine the effects of pollution on communities of indigenous freshwater, estuarine, and marine organisms, including the phytoplankton, zooplankton, periphyton, macrophyton, macroinvertebrates, and fish.

- o Develop and operate a quality assurance program to support the achievement of data quality objectives in measurements of pollutants in drinking water, surface water, groundwater, wastewater, sediment and solid waste.

- o Develop methods and models to detect and quantify responses in aquatic and terrestrial organisms exposed to environmental stressors and to correlate the exposure with effects on biochemical and biological indicators.

This field operations and methods 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.

Thomas A. Clark
Director
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PREFACE

The Bioassessment and Ecotoxicology Branch, Ecological Monitoring Research Division, Environmental Monitoring Systems Laboratory, U.S. Environmental Protection Agency - Cincinnati is responsible for field and laboratory methods 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 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 the 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. The Bioassessment and Ecotoxicology Branch 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-SW field operations and bioassessment methods for evaluating the health and biological integrity of wadable freshwater streams.

ABSTRACT

The Environmental Monitoring and Assessment Program (EMAP) is being implemented to provide for the assessment of status of and trends in the condition of ecological resources within the United States. In 1993 the Surface Waters Resource Group within EMAP implemented the pilot programs for streams. This document contains the updated version of the field operations and methods for biological response indicators (periphyton, benthic sediment metabolism, benthic invertebrates, fish, fish tissue collection, sediment toxicity sample collection), site protocols, water chemistry, physical assessment methods, information management, safety and health, and habitat and stream assessment forms that will be used for the U.S. Environmental Protection Agency (USEPA), Environmental Monitoring and Assessment Program (EMAP), the USEPA Regional Environmental Monitoring and Assessment Program (REMAP), and the field procedures for collection chemical indicators of acidification for the EMAP Temporally Integrated Monitoring of Ecosystems (TIME). This manual was prepared to be user friendly for field personnel that are responsible for collecting biological, chemical, and physical indicator samples.

EMAP-SURFACE WATERS-STREAMS STUDY

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SECTION 1

INTRODUCTION

1.1 This manual contains an update of the 1993 pilot field operations and procedures for biological response indicators, physical assessment methods, and research protocols that will be used in the 1994 pilot programs for the U.S. Environmental Protection Agency (USEPA), Environmental Monitoring and Assessment Program (EMAP) and the USEPA Region III Regional Environmental Monitoring and Assessment Program (REMAP). It also contains the field procedures for collection of chemical indicators of acidification for the EMAP Temporally Integrated Monitoring of Ecosystems (TIME). The purpose of the manual is to be a users manual for field teams conducting sampling and shipping of biological samples. The 1993 pilot laboratory methods have also been updated and are now contained in a separate document, "1994 Pilot Laboratory Methods Manual For Streams."

1.2 The EMAP Surface Water Program will start its second season of stream pilot activities in the mid-Appalachian region during 1994 (see Section 2, Field Operations Overview). EMAP Research will be conducted in conjunction with USEPA's TIME and Region 3 REMAP programs. Three hundred eight Appalachian Ridge and Valley, Blue Ridge, Central Appalachian Plateau, Piedmont and Atlantic Coastal Plain sites have been selected for this study. The streams will be sampled during the 10 week index period from April to July. These sites will be sampled by crews of USEPA, U.S Fish and Wildlife Service, state and contract personnel in each of the operational units of the EMAP, REMAP, and TIME projects.

1.3 The USEPA has designed the EMAP program to determine the current status, extent, changes and trends in the condition of our nation's ecological resources on regional and national scales. 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, nonpoint sources of pollution, and climate change.

1.4 The primary goal of EMAP (Whittier and Paulsen, 1992) is to provide environmental decision makers with statistically valid interpretive reports describing the health of our nation's ecosystems. 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. EMAP's ecological status and trends data will allow decision makers to assess objectively, whether or not the nation's ecological resources are responding positively, negatively, or not at all, to the regulatory programs put in place ostensibly to benefit them.

1.5 To accomplish it's goals EMAP is to document the condition of the nation's forest, wetlands, estuarine and coastal waters (USEPA, 1991a), inland surface waters, Great Lakes, agricultural lands, and arid lands in an integrated manner, on a continuing basis. Although EMAP is designed and funded by USEPA's Office of Research and Development (ORD), other offices and regions within EPA and other federal agencies have contributed to its development and will participate in the collection and use of EMAP data. When fully implemented, EMAP will form a comprehensive national monitoring network, with a large proportion of the data collection and analysis being accomplished by federal, state and local agencies.

1.6 The following three objectives guide the EMAP program:

> **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.**

1.7 The EMAP inland surface waters program (EMAP-SW) is that part of the overall program which is monitoring the health of lakes and streams in the United States. The first phase of the program started with lakes in 1991 (USEPA, 1991b, 1991c, and 1991d), continued in 1992 (USEPA, 1992a, 1992b, 1992c, and 1993), and will continue in 1993, and 1994 (USEPA, 1994). The second phase of the EMAP-SW program has concentrated on streams in 1993 and will again in 1994.

1.8 The EMAP-SW streams pilot of 1993 and 1994 is designed to focus on three classes of streams; first, second, and third order streams. The 1994 EMAP-SW streams pilot has also been integrated with two other USEPA programs, TIME and the USEPA Region III REMAP pilot.

1.9 Prior to full-scale implementation of EMAP-SW streams, a number of questions must be answered through a combination of analyses of existing data and data derived from new field activities. There are two types of field activities to be performed prior to full-scale implementation, pilot projects and demonstration projects. Pilot projects are intended to answer questions about proposed indicators (e.g. plot design, indicator sensitivity to various stressors, magnitude of variance components, alternative methods evaluations, 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 may be designed to answer many of the same questions but also have the prime objective to demonstrate the ability to estimate the condition of regional populations.

1.10 In 1993, the stream pilot initiated the USEPA's EMAP, TIME, and REMAP status and trend monitoring of the ecological condition of the stream population in USEPA Region III. In 1994 this integrated pilot will continue to estimate the condition of the entire stream population in a probability design. For the Region III pilot, we have selected stream sample sites with a randomized, systematic design using the blue lines on 1:100,000 scale USGS topographic maps as the basis for our stream population. For purposes of this pilot, we have restricted our sample population to wadeable streams (first, second, and third order on the 1:100,000 maps) in the Appalachian Mountain portion of Region III (north/west of the Blue Ridge, the Piedmont and Atlantic Coastal Plain). The Region III stream pilot consists of 308 total stream sites visited, 62 EMAP probability stream sites, 30 hand-picked reference sites, and 21 repeat visit sites. One hundred-eleven sites are associated with base EMAP sampling in the region, 195 sites are associated with the EMAP/TIME project (designed to monitor changes in surface waters associated with changes in acidic deposition), and 84 sites are associated with Region III's REMAP project.

1.11 We intend to monitor the physical, chemical, and biological components of ecological condition in single one-day visits to stream reaches. We realize that assemblages and their physical and chemical habitat change seasonally and annually at a single site. However, our data indicate that far greater

differences exist between stream sites during similar time periods. Therefore, we believe we can acquire more information and better assess this spatial variability by maximizing the number of reaches sampled and by sampling within a specific "index" period. Our chosen index period is late spring, specifically late April through July. Twenty-one of the 62 base EMAP sites will be resampled during the index period to evaluate index period variability.

1.12 A few of the questions to be answered in the 1993/1994 EMAP-SW streams project are as follows:

What is the plot design for each class of streams for each indicator?

How will each indicator be used to describe condition (good-poor) of streams and how will acceptable (good) and unacceptable (poor) be determined?

What are differences among different classes of streams?

How many discrete habitats need to be sampled in each plot?

Can teams conduct the field sampling in the time frame described (Late April through mid July)?

What is the magnitude of regional trends detectable given the indicator variability and proposed design?

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

Monitor current status and trends in chemical indicators of acidification in acid-sensitive regions of the U.S.

Relate changes in deposition to changes in surface water conditions.

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

1.14 The Region III REMAP project was designed to test the EMAP approach on a few of the most heavily impacted ecoregions of Region III, the mid-Appalachians, the Ridge and Valley, the Central Appalachians, the Piedmont and some of the coastal plain.

1.15 The Region III REMAP project is 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?

1.16 In order to meet the needs of both EMAP-SW streams and Region III REMAP several different sampling protocols have been put forward at this time. The protocols have been put forth for the 1993/1994 sample period to represent the needs of three major programs, 1994 EMAP-SW streams Pilot, the EMAP TIME project and 1994 Region III REMAP Pilot. These protocols are only final from the standpoint of this pilot activity. They represent information gained from a 2/92 Indicator Workshop of academic, State, and Federal indicator experts; discussion among the EMAP Stream Team; discussions with U.S. Geologic Service NAWQA Program leaders; U.S. Fish & Wildlife personnel; and discussions with USEPA Region III State and Regional biologists, and the 1993 pilot field activity. Many compromises had to be made to satisfy many good comments received from all these discussions. To address many of these comments the protocols described later will be tested in the 1993/1994 sampling period.

1.17 This, "Environmental Monitoring and Assessment Program Surface Waters - 1994 Pilot Operations and Methods Manual For Streams," addresses the field activities for all three sampling programs (EMAP-SW streams pilot, EMAP TIME, & Region III REMAP). Not all of the protocols will be sampled for or performed at the approximately 308 total number of sites. Listed below is a summary of which protocols will used by the sampling program:

PROTOCOLS/INDICATORS TO BE COLLECTED AT EMAP, REMAP, REMAP REFERENCE, REMAP TEST & TIME A SITES:

Fish, macroinvertebrates (> 595 microns), chemistry, and qualitative physical habitat.

PROTOCOLS/INDICATORS TO BE COLLECTED AT EMAP, REMAP REFERENCE, AND REMAP TEST SITES:

Fish, Fish Tissue, macroinvertebrates (> 595 microns), chemistry, sediment metabolism, periphyton, and quantitative & qualitative physical habitat.

1.18 The remaining sections of the EMAP-SW 1994 Pilot Field Operations and Methods Manual For Streams provide an overview of field operations, daily activities at each sample site, stream site verification, site characterization and location. The sections following these are the protocols to be conducted at each site: water chemistry, physical habitat assessment (quantitative), periphyton, benthic (sediment) metabolism, sediment toxicity sample collection, benthic invertebrates, fish, fish tissue indicator, and qualitative habitat assessment. The last two sections are on safety and health as well as sample labelling & tracking.

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SECTION 2

FIELD OPERATIONS OVERVIEW

2.1 The sampling program can be broken down into three elements:

EMAP sites -	Probability sites from base EMAP sample grid
EMAP revisits -	Repeat visits to 1994 and 1995 base EMAP sites for quantifying interannual and sampling period variability
TIME sites -	Augmented grid probability sites in acid-sensitive MAHA ecoregions.

2.1.1 A breakdown of each of these elements, listing the number of stations by state can be found in Table 2.1. The level of sampling effort varies considerably among the program elements. The full complement of protocols (Table 2.2) will be measured at EMAP and EMAP revisit sites. At TIME sites, chemistry, benthos, periphyton, and sediment metabolism will be measured.

2.2 Four sampling teams will be utilized to complete the field work. The EMAP and EMAP revisit sites will be sampled by 1 four-person teams trained in all of the protocols. The TIME sites will be sampled by 3 two-person teams. The teams will be made of personnel from DynCorp (Technology Applications Incorporated (TAI)).

TABLE 2.1

MAHA 1994 SITE DISTRIBUTION BY STATE AND ELEMENT

STATE	EMAP	EMAP Revisits	TIME	TOTAL
MD	0	0	6	6
NY	0	0	4	4
PA	7	7	83	97
VA	6	6	24	36
WV	3	3	54	60
Total	16	16	171	203

TABLE 2.2

MAHA PROTOCOLS BY SITE TYPE

Protocol	EMAP	EMAP Revisit	TIME
Chemistry	X	X	X
Fish	X	X	
Fish Tissue	X	X	
Benthos	X	X	X
Sediment Metabolism	X	X	X
Periphyton	X	X	X
Qual. (RBP) Habitat	X	X	X
Quantitative Habitat	X	X	
Sediment Toxicity	X	X	

SECTION 3

SUMMARY OF DAILY FIELD OPERATIONS FOR EMAP STREAM SAMPLING

3.1 The following schedule is a general outline of the activities that a typical 4-person crew would be involved in at an EMAP stream site. The field crew is sub-divided into two sub-crews, the Geomorphs and the Biomorphs, that reflects their initial responsibilities more than their expertise.

3.2 GEOMORPHS

1. Arrive at site and verify locations (0.5 h) **(Geomorphs)**

    a. GPS
    b. local landmarks
    c. determine average stream width for reach length determination
    d. photodocumentation

3.3 BIOMORPHS AND GEOMORPHS

1. Grab chemistry samples (0.5 h) **(Biomorphs)**

    a. cubitainer
    b. 4 syringes
    c. discharge

2. Mark lower reach boundary and lay-out 11 transects (1 h) **(Geomorphs)**

    a. 5 transects above and below "X"
    b. determine random location for biology samples
    c. collect fine sediments for benthic metabolism and seditment toxicity
    d. set-up benthic metabolism
    e. bag and label sediment toxicity sample

3. Collect bugs and periphyton at transect locations (2.0 h) **(Biomorphs)**

4. Return to lower end of stream reach and begin physical habitat analysis (3.0-3.5 h) **(Geomorphs)**

    a. channel morphology--width, depth, slope, substrate, bank slope thalweg
    b. canopy
    c. woody debris
    d. valley profile

5. Process bugs and periphyton (0.5-1.0 h) **(Biomorphs)**

6. RBP Physical Habitat Sheet, Stream Assessment Sheet (0.25 h) **(Biomorphs)**

7. Breakdown benthic metabolism study (0.25 h) **(Geomorphs)**

6. Fishing (2.0-3.0 h) **(Geomorphs and Biomorphs)**

7. Sample Tracking (0.5-1.0 h) **(Biomorphs)**

3.4 IN SUMMARY, EACH SUB-CREW IS RESPONSIBLE FOR:

GEOMORPHS	BIOMORPHS
Reach verification	Chemistry and discharge
Reach determination	Macroinvertebrate collection
Marking transects	Periphyton collection
Physical habitat assessment	RBP Physical habitat
Benthic metabolism	Sample tracking
Fishing	Fishing

SECTION 4

INITIAL SITE PROTOCOLS

4.1. INITIAL SITE SURVEY

4.1.1 Locating the Chosen Sample Site

4.1.1.1 Stream sample sites were chosen from the blue lines stream network represented on 1:100,000-scale USGS maps following a systematic randomized design 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 **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. Upon finding the X-site, fill out the stream verification form. The latitude/longitude of the site will be listed on the stream information sheet that will be available to the crews for each sample site. Record the latitude/longitude as displayed on the GPS for the X-site on the form. Also record the pertinent GPS data on strength of signal (2-D or 3-D fix) in the appropriate box on the field form. If possible, wait for the GPS to get a 3-D fix. This might require moving the GPS to a more open area near the X-site (GPS works on line of sight to orbiting satellites). If the given latitude/longitude and the GPS latitude/longitude differ by more than 10 seconds, double check that you are at the correct site. Give a detailed description of the directions to the stream site in the indicated place on the form, use additional pages, if necessary. We plan on revisiting the exact same sites in four years.

4.1.2. Determining Stream Site Status

4.1.2.1 We fully expect that not all chosen sites will turn out to be streams. On the basis of previous synoptic surveys in the area, we have 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 streams in the area. Once the crews have reached the marked sample point, evaluate the "X" site and place it into one of the following categories. The primary distinction is that the Nontarget categories have no stream channel or are too deep to wade, the Target categories have a definable stream channel.

NONTARGET

1. No Stream Channel (map error) - Examination of the x-site showed no water body or stream channel.

2. Impounded stream - stream is submerged under a lake or pond due to man-made or natural (e.g., beaver dam) impoundments. If the impounded stream, however, is still wadeable, record the stream as Altered (Target class #4) and sample the stream.

3. Marsh/Wetland - standing water 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.

4. Unwadeable Stream - A stream too deep to be safely sampled by wading following our current protocols. If over half of the reach length is not safely wadeable, classify the reach as unwadeable and do not sample it. If more than half of the survey reach is wadeable (e.g., only a couple of deep pools) classify the reach as target and sample what can be safely sampled.

NOTE: In all cases, evaluate the site at the X-site, don't "find" another spot to sample.

TARGET

1. Regular Stream - sampleable stream with our current protocols.

2. Intermittent Stream - Flow of water is not continual at the site but the channel is wet. See Subsection 4.3 for protocols for sampling intermittent streams.

3. Dry Channel - A discernible stream channel is present but there is no water at the site. See Subsection 4.3 for protocols for sampling dry streams.

4. Altered Channel - stream channel does not appear the way it is marked on the map. However, there is a stream in the field that appears to be the stream marked with the X-site on the map. An example would be a channel rerouting following a flood event that cut off a loop of stream. Position a new X-site at the same relative position in the altered channel. Make careful notes and sketches of the changes on the Site Verification form.

INACCESSIBLE

1. Physical Barriers - Crew is physically unable to reach the X-site because of heavy wetlands, steep gorge or other barrier that prohibits safe entry, explain why in the explanations field.

2. No Permission - Crew is denied access to the site by the landowners, check off "NO" on the stream sampled box on the site verification form and explain in detail in the explanations field.

4.1.2.2 Record the site class and pertinent site verification information on the site verification form. If the site is in one of the target classes, proceed with sampling. If the site is nontarget or inaccessible, the site visit is complete and sampling is finished.

4.1.3 Sampling During/After Rain Events

4.1.3.1 We wish to avoid sampling during high flow episodic 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, we can not restrict sampling to only strict baseflow conditions. It would be next too impossible to define this with any certainty at an unstudied site, and it would also greatly curtail the sampling window of the survey. Thus, we will have to make some compromise on when to sample/not sample a given system because of episodes. To a great extent, we will depend on the judgment of the field crews. If you get to a site and you think it is unduly nfluenced by episodic conditions, do not sample the site that day. Reschedule it for another visit. The following are some general guidelines:

1. The major indicator of episodic influence will be the condition of the stream itself. 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.

2. Do not sample if it is unsafe to wade in the majority of the stream reach.

3. Keep an eye on the weather reports and rainfall patterns. Do not sample a stream during periods of prolonged heavy rains.

4. If the stream seems to be close to normal spring flows, and does not seem unduly influenced by episodic events, go ahead and sample it, even if it has recently rained or is raining.

4.1.4 Taking Site Photographs

4.1.4.1 Site photographs are valuable in data analysis. If you do take any photographs at a stream, start the sequence with one photograph of an 8.5 x 11 piece of paper with the stream ID and date in big letters. After the photo of the stream ID, take 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 fish or other small objects, place a card with the stream ID and date in the snapshot.

4.2 LAYING OUT THE STREAM SAMPLE REACH

4.2.1 Unlike chemistry, which can be measured at a point, most of the biological and habitat structure measures require sampling a certain distance of stream to get an accurate picture of the ecological community. Our pilot studies have suggested that a length of 40 times the channel width is necessary to get over 90% of the species in the stream reach. Thus, we need to lay out a support reach 40 channel widths long around our X-site to characterize the community and habitat of our chosen sample point.

4.2.2 Upon reaching the site, survey the site according to the following protocol:

1. Determine channel width at the X-site by measuring the wetted width across the channel with a surveyor's rod or tape measure at three places considered to be of "typical" width within 10 meters of the X-site. Average the 3 readings together and round to the nearest 1 m. Record this width on the back side of the Stream Verification Form. For dry or intermittent channels, estimate a width based on the unvegetated width of the channel.

2. Scout out the sample reach. One crew member will go upstream and one downstream to check out the condition of the sample reach. Each person shall proceed until they can see the stream to a distance of 20 times the channel width determined in step #1 in the up/down stream direction from the X-site. The objective of the scouting is to make sure the reach is clear of obstacles that would bar a wadeable sampling approach.

NOTE: In streams less than 4 m wide, use 150 m as a minimum sample reach length. Each person would then proceed 75 m from the X-site to lay out the reach boundaries.

3. There are some conditions that may require "sliding" the stream reach around 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 based on 1:100,000 scale maps). Any such confluences near the sample site will be marked on the 1:24,000 scale maps provided to the crews. If such a confluence is reached, note the distance and flag the confluence as the reach end. 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.

NOTE: Do not slide the reach to avoid man-made obstacles such as bridges, culverts, rip-rap, or channelization. They are part of the effects that we're attempting to study.

4. Starting back at the X-site, measure a distance of 20 channel widths down the middle of the stream using a tape measure (be careful not to "cut corners"). At the 20 widths point, find the closest habitat break and flag that location as the start point (transect A). If there is no apparent habitat break for 3-4 channel widths (e.g., a long pool or glide) flag the 20 widths point as the reach end and block net it.

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 spot. 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. Proceed upstream with the tape measure and lay out the 9 additional transects at 1/10 of the reach length intervals (transects C-K). Alternate the transects in order as L,C,R after the first random selection.

6. At the last transect (K), which marks the end of the sample reach (40 channel widths), we wish to end the reach at a habitat break. Thus, mark the last transect at the habitat break closest to the 40 width endpoint. As was done for the start point, if there is no habitat break within 3-4 channel widths, mark the 40 width endpoint as the reach end. The last transect does not need an L,C or R location as biology is not collected here. The final reach/transect layout should look like the one in Figure 1 of Section 6, Physical Habitat Assessment.

4.2.2.1 Before leaving the stream, complete a rough sketch map of the stream reach you sampled on the back of the Stream Verification Form. 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 PROCEDURES FOR SAMPLING DRY AND INTERMITTENT STREAMS

4.3.1 Layout the survey stream reach as described in Subsection 4.2. For ALL target streams, whether they are dry, intermittent, or completely flowing, complete the thalweg profile (form SH-01, see Subsection 4.2) for the entire 40 channel width reach length. For dry locations, record stream width/depth as 0. The thalweg profile will give us a record of the "water" status of the stream for future comparisons (e.g., % of length with intermittent pools or no water).

4.3.1 At each of the transects (cross sections) laid out for sampling, classify the stream at that spot as:

DRY CHANNEL: No surface water anywhere in cross section;

Complete the physical habitat sampling protocols at dry cross sections, use the unvegetated area of the channel to determine channel width. This will describe riparian condition, substrate type, and diagnostic physical habitat information.

DAMP CHANNEL: Wet spots in cross section but NO flowing water or pools > 10 cm deep; Do the physical habitat series of forms and collect periphyton samples from the wet spots. These are great environments for algae; if there are enough fine wet sediments for metabolism, collect them as well.

ENDURING POOLS: No flowing water but pools > 10 cm deep;

Do complete physical habitat, algae, metabolism, and macroinvertebrate sampling. This is essentially the standard biological protocol.

FLOWING WATER: Flowing water in cross section, follow standard protocols for all indicators.

4.3.1.1 Thus, physical habitat information 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. Fish sampling and water chemistry sampling is discussed below.

4.3.2 Water chemistry sampling

4.3.2.1 The water chemistry sample needs to be collected at the X-site. If the X-site is dry but there is flowing water or a pool with greater than 1 m^2 of water surface area, more than 10 cm deep somewhere along the survey reach, take the water sample and water chemistry measurements at the pool or flowing water nearest 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 Sample Collection Form. If there is no spot on the survey reach with flowing water or a pool greater than 1 m^2 and 10 cm deep then do not collect a water sample.

4.3.3 Fish Sampling

4.3.3.1 In intermittent streams, fish sampling will take place in any wet areas along the 40 channel width reach that are potential fish habitat (electrofishing or seining). Sampling will be restricted to the reach length laid out previously. No fish sampling will be done in reaches that are entirely dry.

SECTION 5

WATER CHEMISTRY

5.1 INTRODUCTION

5.1.1 There are two components to collecting water chemistry information; collecting samples of stream water to ship to the analytical laboratory, and in situ measurements of specific conductance, dissolved oxygen, and temperature. At each stream, crews will fill one 4 L cubitainer and four 60 mL syringes with streamwater. These samples will be stored in a cooler packed with ziplock bags filled with ice and shipped to the analytical laboratory at the University of Maine within 24 hours of collection. Streamside measurements will be made using field meters and recorded on data forms. The primary function of the streamwater samples and the streamside chemistry measurements are to determine:

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

5.1.2 Streamwater from the cubitainers 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, 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 cubitainer samples stabilized (filtration and/or acidification) within a short period of time (72 hours). Most of the procedures outlined in this section are similar to the ones utilized by USEPA in streams for the National Surface Water Survey (Kaufmann et al., 1988) and have been taken from the Handbook of methods for acid deposition studies: Field operations for surface water chemistry (USEPA, 1989).

5.2 COLLECTING WATER CHEMISTRY SAMPLES

1. Before leaving the base site, package the sample containers (one 4 L cubitainer and four, 60 mL syringes) and stream sample beakers to prevent contamination; place the syringes in their plastic container and the cubitainer and beakers in a clean ziplock bag. Label the cubitainer and each of the four syringes with a barcode label.

2. In the field, make sure that the **syringes and the cubitainer have the same barcode label number.** Make sure the labels are securely attached.

3. Collect the water sample from the X-site in a flowing portion near the middle of the stream. Rinse the 500 mL sample beaker three times with streamwater, discard the rinse downstream.

4. Remove the cubitainer lid and expand the cubitainer by pulling out the sides. Fill the beaker with streamwater and slowly pour 30-50 mL into the neck of the 4-L Cubitainer. Cap and rotate it so that the water contacts all the surfaces. Discard the water. Repeat the above rinsing procedure two more times.

NOTE: DO NOT BLOW into the Cubitainers to expand them, this will cause contamination.

5. Fill the cubitainer with streamwater by pouring in 500 mL portions from the sample beaker. Let the weight of the water expand the cubitainer. Fill the cubitainer to its maximum volume. Rinse the cubitainer lid with streamwater.

6. Eliminate any air space from the Cubitainer, and cap it tightly. Make sure the cap is tightly sealed and not on at an angle. Place the cubitainer inside the 1 gallon ziplock bag it came in and seal the bag.

7. If a cooler is available, place the cubitainer in the cooler (on ice or streamwater) and shut the lid. If not, place the cubitainer in an opaque garbage bag and keep at the side of the stream in the streamwater to keep cool while the rest of the sampling is completed.

8. Collect the 60-mL syringe samples from the same place that the cubitainer sample was collected. Rinse the syringe by inserting the 60-mL syringe halfway into the stream, sucking in a 15-20 mL aliquot, pulling the plunger to its maximum extension and rotate the syringe so the water contacts all surfaces. Discard the water by depressing the syringe plunger (point downstream). Repeat the above rinsing procedure two times.

9. Insert the syringe into the stream again and slowly fill the syringe with 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.

10. Invert the syringe (tip pointing up), cap the syringe with the syringe valve, and tap the syringe lightly to detach any trapped air bubbles. Make sure the valve is open and expel the air bubbles, and a small amount of water; leave 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 9).

11. Fill all four syringes, make sure that the barcode label number on all four syringes are the same and match the barcode number on the cubitainer. Place the syringes together in their solid plastic shipping container and place in the cooler or in the streamwater with the cubitainer.

NOTE: All of the syringe measurements are affected by equilibration with atmospheric carbon dioxide, it is essential that no outside air contact the samples collected for these determinations.

12. 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 happens during the sampling process that could influence sample chemistry (heavy rain, potential contaminants). If the sample was collected at the X-site, place an X for station collected. 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 column. Record more detailed reasons and/or information on the comments line.

13. After carrying the samples out to the vehicles, place the cubitainer and syringe case with syringes in a cooler and surround with 1 gallon ziplock bags filled with ice.

14. Ship the coolers to the analytical laboratory by overnight courier at first opportunity, either that afternoon or the next day (see Section 14, Information Management, on sample shipment). Place fresh ice in the ziplocks in the cooler just before shipping. Write word "ICE" on ziplock bags with indelible marker.

15. At the base site, after sampling, rinse the sample beakers three times with deionized water to prevent contamination of the next days sample. Place the beakers in a 1-gallon ziplock bag with the next days cubitainer for transport.

CAUTION: Throughout the water chemistry sampling process it is important to take precautions to avoid contaminating the sample. Many of the streams have a very low ionic strength and can be contaminated quite easily by perspiration from your hands, sneezing, smoking, bug repellent, or other chemicals used during sample collection. Thus, make sure that none of the water sample contacts your hands before going into the cubitainer. Do not apply to oneself any bug repellant, perfume, or make up until after water sample is collected. Note any problems on field forms.

5.3 STREAM SIDE CHEMICAL MEASUREMENTS

5.3.1 Introduction

5.3.1.1 The following protocols are intended for in situ measurement of streamwater specific conductance, temperature, and dissolved oxygen. Specific conductance or conductivity is a measure of the ability of the water to pass an electrical current which is related to the ionic strength of a solution. Dissolved oxygen (DO) is a measure of the amount of oxygen dissolved in solution. In natural waters, minimal concentrations of oxygen are essential for survival of most aquatic organisms. Measures of DO and temperature are used to assess water quality and the potential for healthy aerobic organism populations.

5.3.2. Measuring Stream Specific Conductance

1. Check conductivity pen for outward signs of fouling daily. 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.

2. Perform an initial field quality control check (QCC) before the stream conductivity measurement. Remove the cap from the bottom of the conductivity pen. Swirl the pen for 3-5 seconds in the "Rinse" QCC solution (Peck and Metcalf, 1991) and let stabilize for 20 seconds. Record the QCC solution conductivity on the Field Measurement Form.

3. If the QCC solution measurements were not within 10 uS/cm of theoretical value (see bottle), repeat the measurement process. If the value is still unacceptable, flag the conductivity data on the field form. Recheck the QCC solution conductivity back at the base site with fresh bottles of QCC solution. If it is still off by more than 10 S/cm, clean the probe as described in the manual, check the batteries, soak in deionized water for 24 hours, and try again. If this doesn't work, replace the conductivity pen.

4. Rinse a 250 mL sample beaker three times with stream water and fill with 150 to 200 mL of stream water for determination of stream conductivity.

5. Swirl the conductivity pen in the beaker for 5-10 seconds. Hold unswirled, for 20 seconds in the sample to allow for the pen's temperature compensation mechanism to function. Don't let the pen touch the bottom of the beaker. Record the stream conductivity on the Field Measurement Form.

Alternatively, the pen can be placed directly in a quiescent part of the stream and conductivity read directly (needs the same equilibration time).

6. Routine Maintenance for conductivity pens

NOTE: Refer to the instrument manual for probe cleaning instructions.

a. Store the pens in deionized water by keeping deionized water in the bottom of the pen cap.

b. Before using a probe which has been stored dry, soak the electrodes in deionized water (by filling the caps) for 24 hours.

c. Always turn off the pens after use by capping them.

d. Replace the bottle of QCC solution with fresh QCC solution every 2 weeks.

5.3.3. Dissolved Oxygen/Temperature

1. Calibrate the dissolved oxygen (D.O.) meter as described in Section 8, Benthic (Sediment) Metabolism, Subsection 10, Calibration And Standardization.

2. Place the D.O. probe in mid-stream at mid-depth at the X-site. Allow the probe to equilibrate and record the D.O. and stream temperature on the Field Measurement Form. If the D.O. meter is not functioning, measure the stream temperature with the field thermometer and record the reading on the Field Measurement Form along with pertinent data flags and comments.

NOTE: Stream dissolved oxygen and temperature values may be taken at the same time that the initial dissolved oxygen/temperature values are measured for the benthic (sediment) metabolism studies instead of the same time as the cubitainer sample is collected.

5.4 REFERENCES

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

USEPA. 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, DC.

Table 5-1. CHEMICAL SAMPLING CHECK-OFF SHEETS

EQUIPMENT TO CARRY IN FIELD FOR WATER CHEMISTRY

125 mL bottle of QCC solution in ziplock bag
Conductivity Pen
Field Forms
One , 250 mL plastic beakers
One, 500 mL plastic beaker
One cubitainer in clean ziplock bag (barcode label attached)
Four, 60 mL syringes in plastic container (each one with barcode label attached)
Four syringe valves in plastic container
Opaque garbage bag

EXTRA EQUIPMENT TO CARRY IN VEHICLE

Cooler with 4-6 one gallon ziplock bags filled with ice
Back-up labels, forms, syringe valves

DAILY ACTIVITIES AFTER SAMPLING

1. Enter chemistry tracking data from today's sample (bar code ID, stream ID, date, etc.) into computer
2. Check that cubitainer lid is on tight and has a flush seal
3. Prepare sample for shipping (label and seal cooler, replace ice as close as possible to shipping time)
4. Call Overnight shipping company to arrange pick-up of cooler
5. Rinse all sample beakers with deionized water, three times
6. Make sure conductivity pens are rinsed with deionized water and are stored with moist electrodes
7. Barcode label the next days sample containers (cubitainer and syringes), pack cubitainer and sample beakers in clean ziplock bag, and pack four syringes and syringe valves in plastic container.

SECTION 6

PHYSICAL HABITAT ASSESSMENT

6.1 INTRODUCTION

6.1.1 Rationale

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

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

All of these attributes may be directly or indirectly altered by anthropogenic activities. Nevertheless, their expected values tend to vary systematically with stream size (drainage area) and overall gradient (as measured from topographic maps). The relationships of specific physical habitat measurements described in this EMAP-SW field manual to these seven attributes are discussed by Kaufmann (1993). Aquatic macrophytes, riparian vegetation, and large woody debris are included in this and other physical habitat assessments because of their role in modifying habitat structure and light inputs, even though they are actually biological measures. The field physical habitat measurements from this field habitat characterization are used in the context of water chemistry, temperature, and other data sources (e.g., remote sensing of basin land use and land cover). The combined data analyses will more comprehensively describe additional habitat attributes and larger scales of physical habitat or human disturbance than are evaluated by the field assessment alone.

6.1.1.2 This protocol is intended for evaluating physical habitat in wadeable streams. The protocol is 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. It 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 habitat assessment methods has 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.

6.1.1.3 The protocol we describe differs from other rapid habitat assessment approaches 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 crew to estimate attributes that are otherwise measurable, rather than estimating the quality or importance of the attribute to biota or its importance as an indicator of disturbance. We have included the more traditional visual classification of channel unit scale habitat types because they have been useful in past studies and enhance comparability with other work.

6.1.1.4 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 3 hours of field time. 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 as little as 2 hours, whereas crews may require up to 5 hours in large (>10m wide), complex streams with abundant woody debris and deep water.

6.1.1.5 The protocol defines the length of each sampling reach proportional to stream width and then systematically places measurements to statistically represent the entire reach. Stream thalweg 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 stream reach, and discharge is measured at one. The tightly spaced depth and width measures allow calculation of indices of channel structural complexity, objective classification of channel units such as pools, and quantification of residual pool depth, pool volume, and total stream volume.

6.1.2 Components of the Field Habitat Assessment

6.1.2.1 The Physical Habitat Protocol consists of four different components (Table 1). Measurements for the first three components are recorded on 11 copies of a two-sided field form, with a separate form for discharge measurements (both forms attached). The procedures for conducting each component are discussed in four subsections of this training manual. The first component (the THALWEG PROFILE) is a longitudinal survey of depth, width, habitat class, and presence of fine/soft sediment at 100 equally spaced points along the centerline between the two ends of the sample stream reach (150 points in some small streams – see 6.1.3.2). The second component is a continuous tally of large WOODY DEBRIS along the reach. The third component, the detailed channel and riparian CROSS-SECTIONS, includes measures and/or visual estimates of channel cross-sectional dimensions, substrate, fish cover, bank characteristics, and riparian vegetation structure at 11 equally-spaced stations along the length of the reach. This third component also includes measurements of the gradient and compass bearing between stations, providing information necessary for calculating reach gradient, residual pool volume, and channel sinuosity. The fourth and final component is a measurement of instantaneous DISCHARGE at one optimally chosen cross-section.

6.1.3 Habitat Sampling Locations on the Study Reach

6.1.3.1 Measurements are made at two scales of resolution along the mid-channel length of the reach; the results are later aggregated and expressed for the entire reach, a third level of resolution (Figure 1). We want to assess habitat and other stream indices over stream reach lengths that are approximately 40 times their average wetted width. This allows us to adjust the sample reach length to accommodate varying sizes of streams. Subsection 4.2 describes the procedures for locating the X-site that defines the

midpoint of the sample reach. This sampling location is already marked on a 1:24,000 map prior to going into field. It has precise coordinates of latitude and longitude, and was selected by the EMAP design group using a randomized systematic sampling design. Subsection 4.3 prescribes the protocol for delineating a sample reach that is 40 times its width, but no shorter than 150 meters. That section also describes the protocol for measuring out (with tape measure) and flagging the locations of the 11 channel cross sections where many habitat measurements will be made. The distance between each of these cross section is 1/10th the total length of the sample reach.

Table 1. Components of Physical Habitat Protocol

Component	Description
1. Thalweg Profile:	Measure maximum depth and wetted width, classify habitat, determine presence of fine/soft sediment at 10-15 equally spaced intervals between each of 11 channel cross-sections (100-150 along entire reach).
2. Woody Debris:	Between each of the channel cross sections, tally large woody debris numbers within the bankfull channel according to size classes.
3. Channel and Riparian Cross-Sections:	@ 11 cross-section stations placed at equal intervals along reach length: – Measure: channel cross section dimensions, bank height, undercut, angle (with rod and clinometer); gradient (clinometer), sinuosity (compass backsite), riparian canopy cover (densiometer). – Visually Estimate*: substrate size class and embeddedness; areal cover class and type (e.g., woody) of riparian vegetation in Canopy, Mid-Layer and Ground Cover; areal cover class of fish concealment features, aquatic macrophytes and filamentous algae. – Observe & Record*: human disturbances and their proximity to the channel.
6. Discharge:	In medium and large streams (defined later) measure water depth and velocity (@ 0.6 depth with electromagnetic or impeller-type flow meter) at 15 to 20 equally spaced intervals across one carefully chosen channel cross-section. In very small streams, measure discharge with a portable weir or time the filling of a bucket.

* 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. 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 station. They extend shoreward 10m from left and right banks. Fish cover types, aquatic macrophytes, and algae are observed within channel 5m upstream and 5m downstream from the cross section stations. These boundaries for visual observations are estimated by eye.

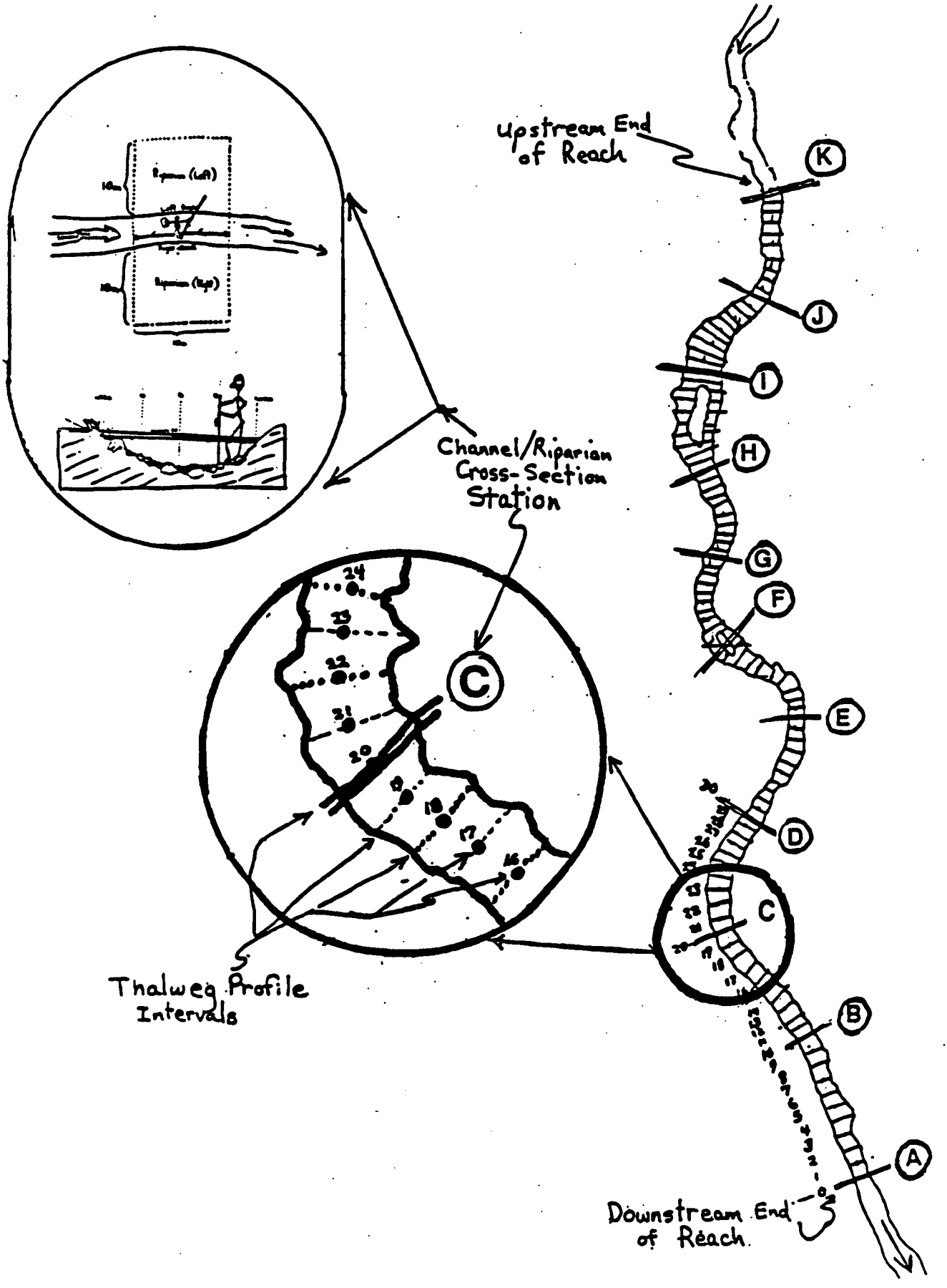


Figure 1. Sample reach layout (plan view).

6.1.3.2 The thalweg profile measurements must be spaced evenly over the entire sample reach length. In addition, they must be sufficiently close together that they do not "miss" deep areas and habitat units that are in a size range of about 1/3 to 1/2 of the average channel width. Follow these specifications for choosing the interval between thalweg profile measurements:

Channel Width < 2.5m — intervals = 1.0 m
Channel Width 2.5-3.5m — intervals = 1.5 m
Channel Width > 3.5m — intervals = 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, width measurements in stream reaches greater than 4m wide (having reach lengths greater than 150 m) shall be taken only at the 11 cross-section stations. In all the streams narrower than 4m (having reach lengths of 150 m), widths are measured at all 100 or 150 thalweg profile locations.**

6.1.4 Logistics and Work flow

6.1.4.1 The four components of the Physical Habitat data collection are organized into three grouped activities:

1. Thalweg Profile and Large Woody Debris Tally (Sections 6.2 and 6.3). Two people proceed upstream from the reach start point (see Section 4.3) making observations and measurements at the chosen increment spacing (see Subsection 6.1.3). One person is in the channel making width and depth measurements, and determining whether soft sediment is present under his/her staff. The other person records these measurements, classifies the channel habitat, and makes the large woody debris estimates. Each time this team reaches a new cross section flag, they take out a new copy of one of the 11 Channel/Riparian Cross Section forms. They interrupt the thalweg profile and woody debris assessment activity to complete each cross-section as it comes. The thalweg profile is continuous, but is measured in pieces, each recorded on the cross section form where the little segment begins.

2. Channel/Riparian Cross-Sections (Section 6.4). One person proceeds with the channel cross-section dimension, substrate, bank, and canopy cover measurements. The second person records those measurements while making visual estimates of riparian vegetation structure, fish cover, and human disturbance specified on the field form. Slope & bearing backsites are accomplished together. Intermediate flagging (of a different color) may have to be used if 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 the crews could tally woody debris while doing the backsite, rather than during the thalweg profile measurements.)

3. Discharge (Section 6.5). Discharge measurements shall be made after collecting the chemistry sample. They are done at a chosen optimal cross section 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 and sediment must be stirred up, wait on this activity until all chemical and biological sampling has been completed.

6.2 THALWEG PROFILE

6.2.1 "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 to 150 equally spaced points along the centerline of the stream between the two ends of the stream reach. The

EMAP-SW habitat assessment modifies traditional methods by proceeding upstream in the middle of the channel, rather than along the thalweg itself. One field person walks upstream (wearing felt-soled waders) carrying a 1.5 - 5 meter fiberglass telescoping surveyor's rod (metric) and a 1m metric rule (calibrated rod or pole). A second person on the bank or in the stream carries a clipboard with 11 Thalweg Profile & Woody Debris Forms. 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.

6.2.2 Use the surveyor's rod and meter stick to make the required depth and width measurements, and to measure off the distance between sampling points as you proceed upstream. Ideally, each 10th increment of thalweg measurements will bring you in line with 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. However, you may still need to make minor adjustments to align each 10th measurement with 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 each of the 11 field forms to record these measurements.

6.2.3 At each of the 100 increments of length upstream along the mid-channel line, measure and record the greatest depth (to the nearest centimeter) in the channel on either side perpendicular to the center line (Figure 1). This deepest point, the "thalweg depth", will not necessarily be along the mid-channel line. When measuring depth with the pole or surveyor's rod, read the water depth on the side of the rod, to avoid inaccuracies due to the wave formed by the rod in moving water. When placing your rod or staff on the stream bottom, record the presence or absence of fine, soft sediments beneath your staff. Fine soft sediments are defined here as fine gravel, sand, silt, clay or muck readily apparent by "feeling" the bottom with the staff.

6.2.4 At the same channel locations as for thalweg depth, measure the wetted width perpendicular to the mid-channel line . The wetted width boundary is the point at which substrate particles are no longer surrounded by free water. Width measurements are taken rapidly when they are part of the thalweg profile. Read these width measurements to the nearest 0.1 meter for widths up to about 3 meters. Thereafter, read to approximately the nearest 5%. 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.

Omit the rapid multiple width measurements when doing the thalweg profile on stream reaches greater than 4m wide (ie., those having sample reach lengths greater than 150 m). In these larger streams, widths shall be measured only at the 11 cross-section stations. In all the streams narrower than 4m (having reach lengths of 150 m), widths are measured at all 100 or 150 thalweg profile locations.

6.2.5 While recording the width and depth measurements and the presence of fine sediments, the second person chooses and records the Habitat Class and the Pool Forming Element Code applicable to each of the 100 sampling points along the length of the reach (Table 2; Figure 2). The resulting database of traditional visual habitat classifications will provide a bridge of common understanding with other studies. Channel unit scale habitat classifications are to be made at the thalweg of the cross section. The habitat unit itself must meet a minimum size criteria in addition to the qualitative criteria listed in Table 2. 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.

Table 2. Habitat Classification at Channel Unit Scale (See Figure 2). Note that in order for a channel habitat unit to be distinguished, it must be at least as wide or long as the channel is wide.

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 bank.
Backwater Pool	PB	-	Pool separated from main flow off side of channel.
Dam Pool	PD	-	Pool formed by impoundment above dam or constriction.
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, depending upon discharge.
Dry Channel	DR		No water in channel

Code	Pool-Forming Element 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
O	Other -- note in comments

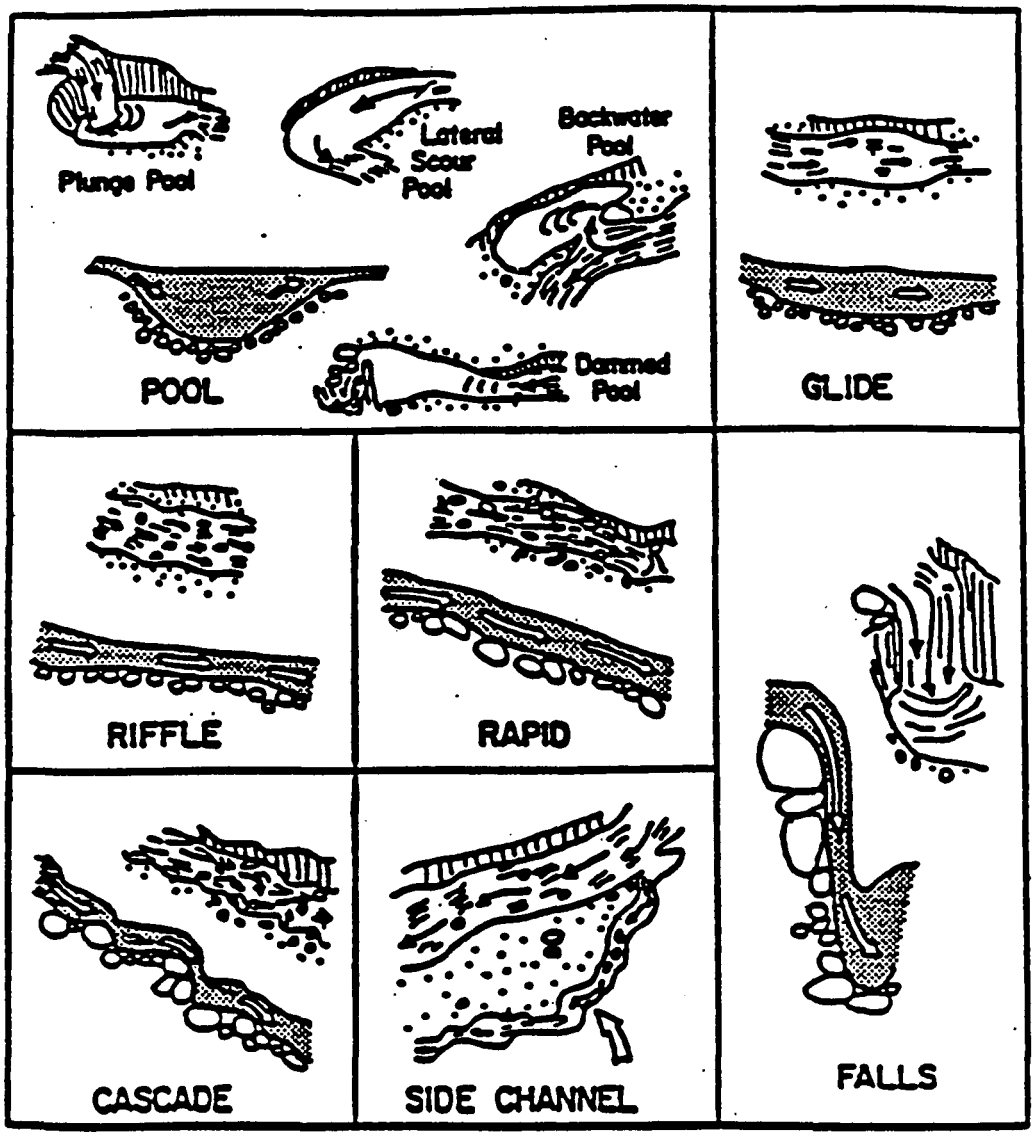


Figure 2. Sketches of channel units (From Frissell et.al.,1986)

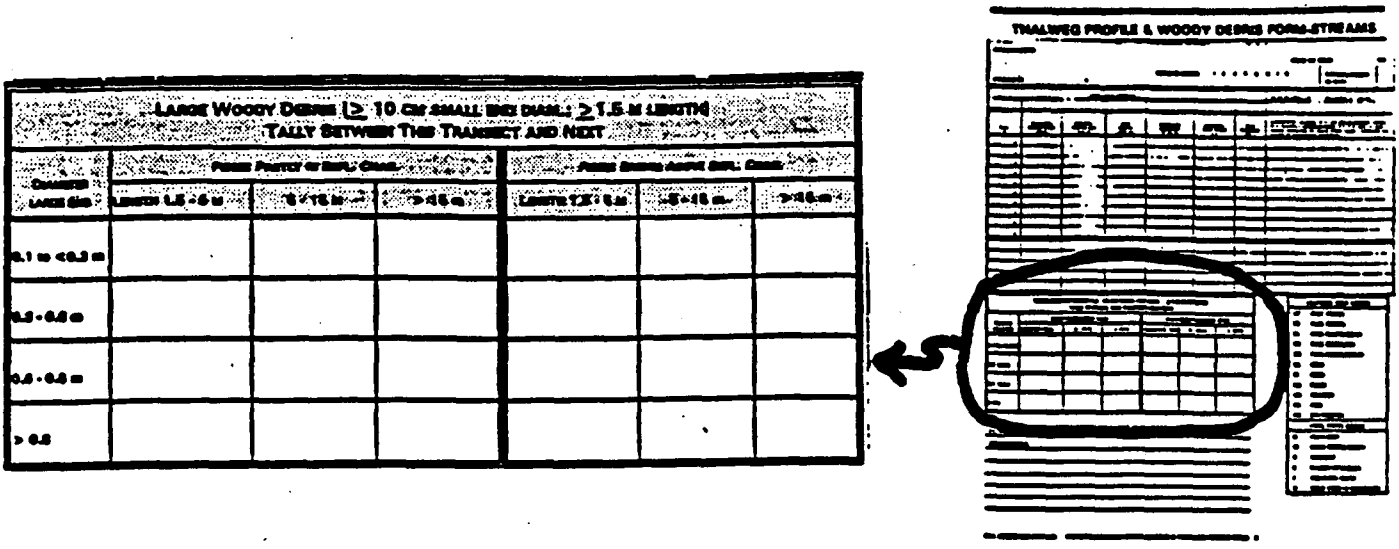
6.2.6 Mid-Channel Bars, Islands, and Side Channels pose some problems for the sampler conducting a thalweg profile. These split channel features necessitate some guidance. Bars are defined here as channel features below the bankfull flow mark that are dry during baseflow conditions (see Section 6.4.3 for definition of bankfull channel). Islands are 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 bar is encountered along the thalweg profile, it shall be noted as "BAR" in the comments column and the active channel is considered to include the bar. Therefore, the wetted width shall be measured as the distance between wetted left and right banks. It is measured across and over bars and boulders. If bars are present record bar width in the space provided.

6.2.7 If a mid-channel feature is as high as the surrounding flood plain, it is considered an island. Treat islands different than bars. The way to handle the ensuing channel shall be based visual estimates of the percent of flow of the side channel as follows:

Side/Main Channel Flow %	Action
<15%	Note side channel on form ("SC").
15-35%	Note "SC" plus detailed channel & riparian cross-section on the side channel . (Designate transect as XA, XB, etc on form)
>35%	Note "SC" plus detailed channel & riparian cross-section PLUS another thalweg profile on the side channel.

6.2.8 When side channels are present, the comments column of the Thalweg Profile form should reflect their presence with a continuous "SC" note. This comment will begin at the point of convergence with the main channel, their presence off-channel will be continuously noted, and the comments will show the point of side channel divergence. If it is a long side channel, a continuing arrow can be used to mark it in the comments column to save time. 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 as detailed above. For 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).

6.3 LARGE WOODY DEBRIS MEASUREMENTS



6.3.1 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 protocol 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 small end diameter of at least 10 cm (4 in.) and length of at least 1.5 m (5 ft.). Tally 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. First, tally together all the LWD that is at least partially in the bankfull channel Zones 1 or 2 shown in Figure 3. Then tally all the LWD that is not actually within the bankfull channel, but is at least partially spanning (bridging) the bank full channel (Zone 3 in Figure 3). For both the Zone 1-2 wood and the Zone 3 LWD, the field form provides 12 entry boxes for tallying debris pieces visually estimated within three length and four diameter class combinations. Each LWD piece is tallied in only one box. There are 12 size classes for wood at least partially in Zone 1 and 2, and 12 for wood partially within Zone 3. Wood not partially within those Zones is not tallied. LWD in the active channel is tallied over the entire length of the reach, including the area between channel cross sections. As in the thalweg profile, LWD measurements between each channel cross section and the next one upstream are recorded on the first 10 channel cross section forms. The location of the large end of the LWD determines the cross section to which it is assigned.

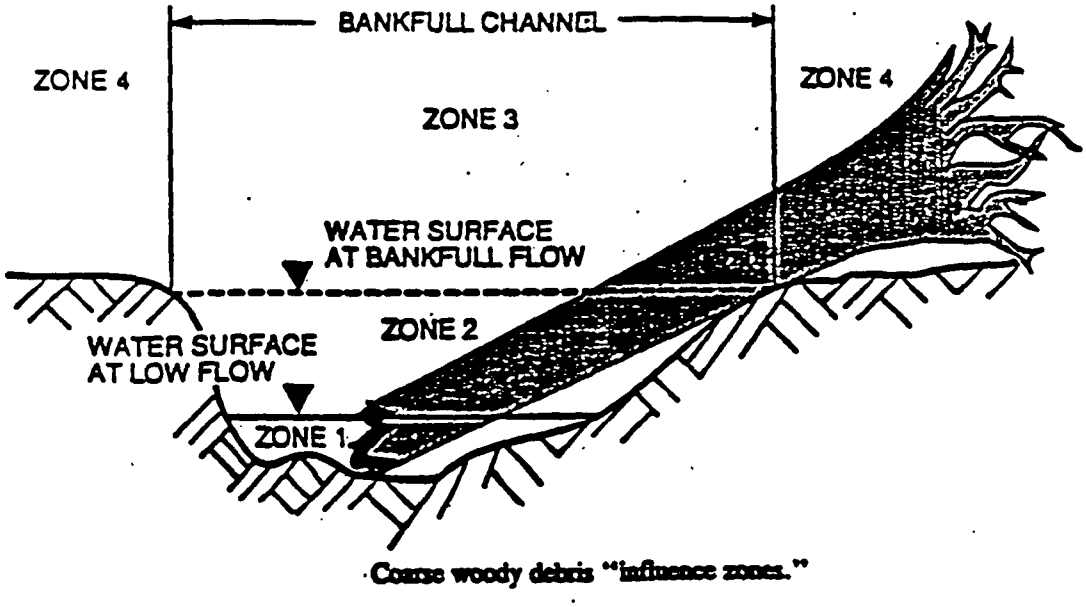
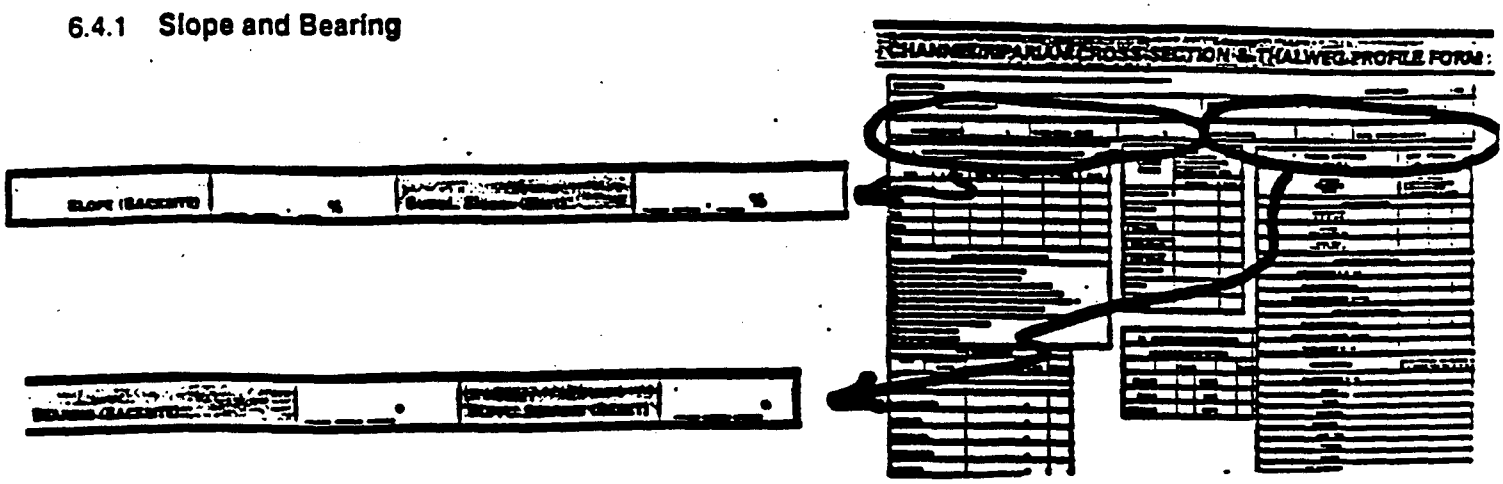


Figure 3. Large woody debris and influence zones (From Robison and Beschta, 1990).

6.3.2 For each LWD piece, first visually estimate its length and its large and small end diameters in order to place it in one of the diameter and length categories. The diameter classes are 0.1m to <0.3m, 0.3m to <0.6m, 0.6m to <0.8m, and >0.8m. The length classes are 1.5m to <5.0m, 5m to <15m, and >15m. Sometimes LWD is not cylindrical, so it has no clear "diameter". In these cases visually estimate what the diameter would be for a piece of wood with circular cross section that would have the same volume. When evaluating length, include only the part of the LWD piece that has a diameter greater than 10 cm (4 in). The diameter class on the field form (Figure 3) refers to the large end diameter. 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.

6.4 CHANNEL AND RIPARIAN CROSS-SECTIONS

6.4.1 Slope and Bearing



6.4.1.1 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; both of which are in turn important controls on aquatic habitat and sediment transport within the reach. Second, the spatial variability of stream gradient is a measure of habitat complexity, as reflected in the diversity of water velocities and sediment sizes within the stream reach. Lastly, using methods described by Stack (1989) and other methods currently being developed (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 (Subsection 6.2). 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).

6.4.1.2 You will measure slope and bearing by "backsiting" downstream between cross-section station 2 and 1, 3 and 2, 4 and 3, etc., up to the 11th cross section (Figure 4). To measure the slope and bearing between adjacent stations, use a clinometer, rangefinder compass, tripod, tripod extension, and flagging, following the following six step procedure. If you are doing these measurements with another person, replace the "tripod" with your co-worker in the following protocol, but be sure that you site to YOUR EYE LEVEL when backsiting on your co-worker. Also, be sure that he or she is standing 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.

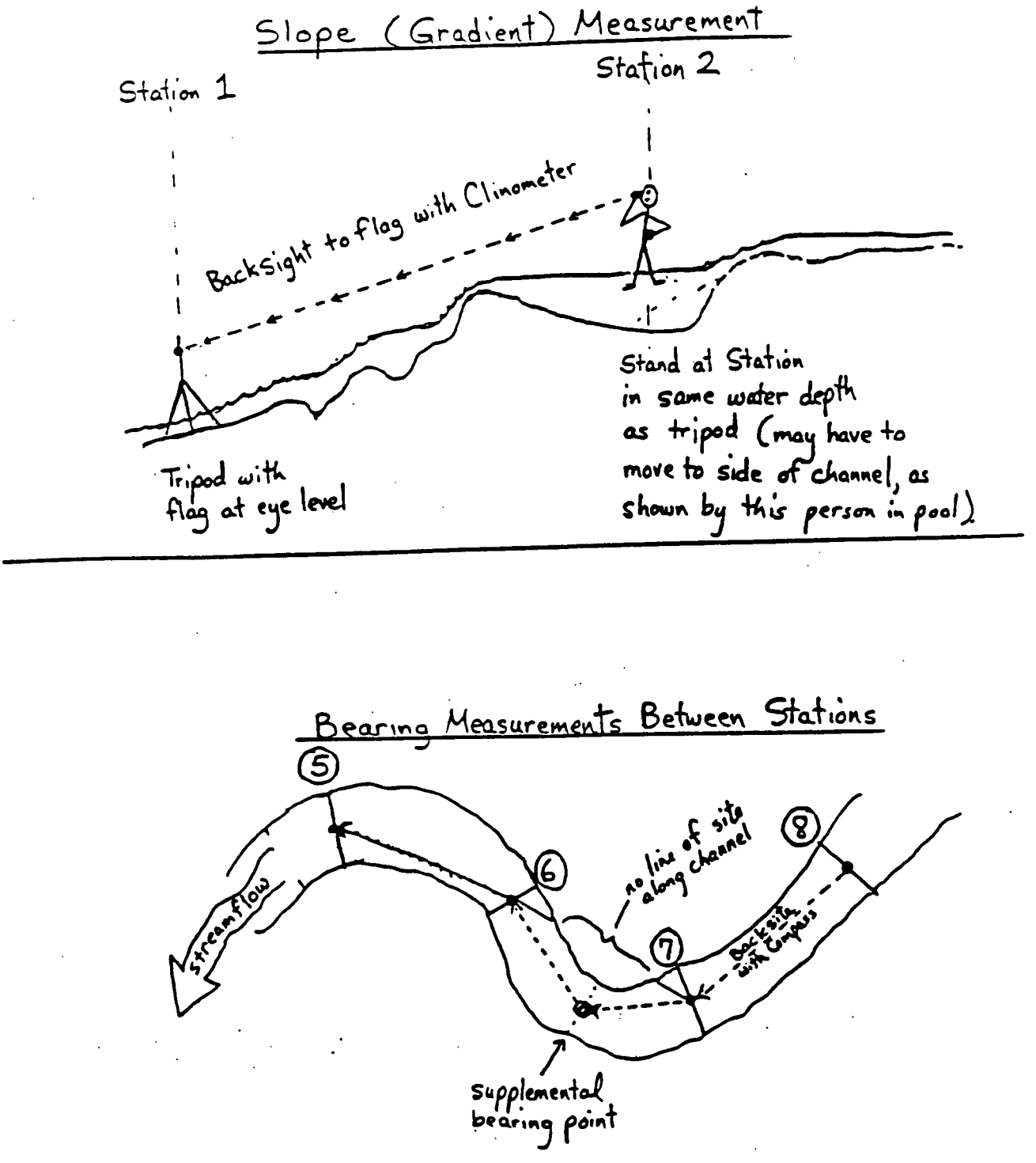


Figure 4. Channel slope and bearing measurements.

1. Set up the tripod in shallow water or at the water's edge at Station 1. Standing tall in a position with your feet as near as possible to the water surface elevation, set the tripod extension with a piece of flagging at eye level. Remember the depth of water in which you are standing when you adjust the flagging to eye level.

2. Walk upstream to Station 2 (ten "increments" upstream), where the next detailed cross section is located.

3. Find a place to stand at Station 2 that is at the same depth as where you stood at the previous station when you set up the eye-level flagging. With the clinometer, site back downstream at your flagging at Station 1; read and record PERCENT Slope on the field form. Be careful, the clinometer reads both percent slope and degrees of the slope angle. PERCENT SLOPE IS THE SCALE ON THE RIGHT HAND SIDE AS YOU LOOK THROUGH THE CLINOMETER.

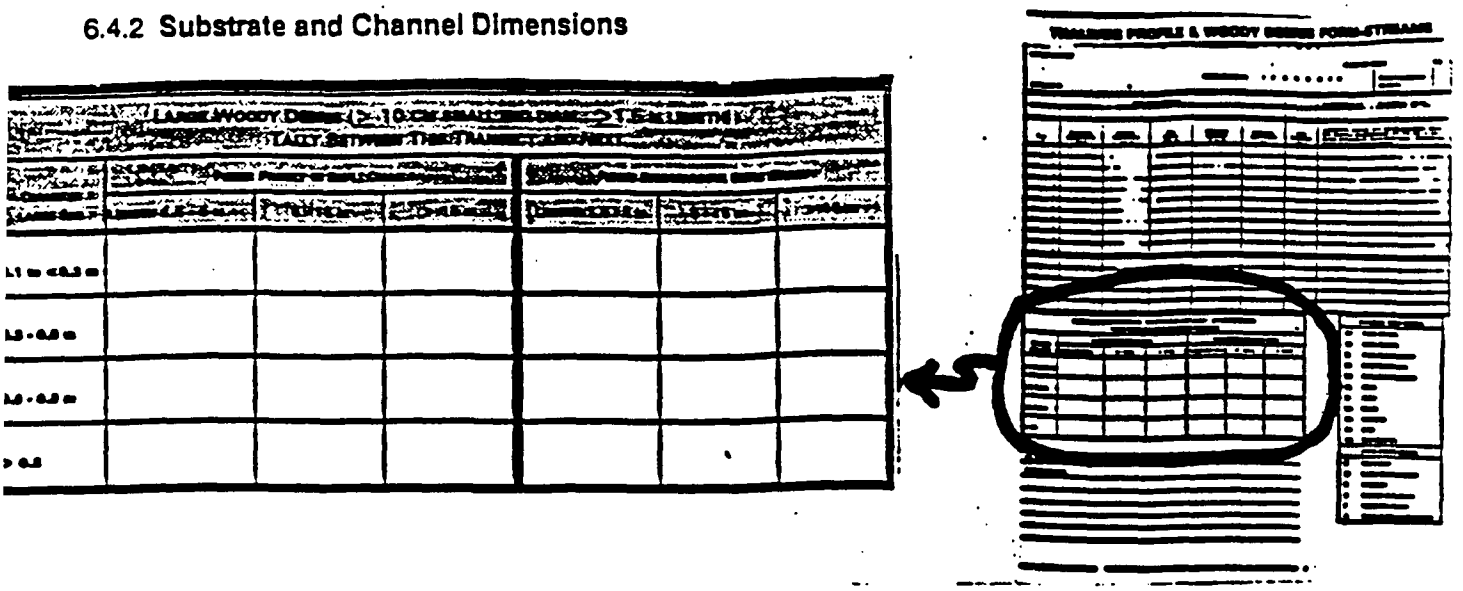
4. Stand now in the in the middle of the channel at Station 2, and site back with your compass to the middle of the channel at Station 1 and record your compass bearing ("Azimuth"). It does not matter for these measurements whether or not you adjust your compass bearings for magnetic declination, but IT IS IMPORTANT THAT YOU ARE CONSISTENT IN THE USE OF MAGNETIC OR TRUE BEARINGS throughout all the measurements you make on a given reach. 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."

5. Retrieve the tripod from the downstream cross section station (#1), (while doing a debris tally) and set it up at Station #2 (just like step 1) before proceeding to station 3 to begin a new set of cross-section measurements and backsitings.

6. When you get to each new detailed cross section station, backsite on the previous station repeating steps 2 through 5 above.

6.4.1.3 As stated earlier, it may be necessary to set up intermediate slope and bearing stations between the normal 11 stations if you do not have DIRECT LINE-OF-SITE ALONG (AND WITHIN) THE CHANNEL BETWEEN STATIONS. This can happen if bush 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 normal 11 stations, to avoid confusion. Record these supplemental slope and bearing backsites in the appropriate places on the field form. It is important also to clearly identify the channel position (increment number) of these supplemental measurements on the thalweg profile form and the cross-section & riparian form in their respective comments sections.

6.4.2 Substrate and Channel Dimensions



6.4.2.1 Substrate size is one of the most important determinants of habitat character for fish and macroinvertebrates in streams. Along with bedform (eg. 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. Sediment characteristics are often sensitive indicators of the effects of human activities on streams. Decreases in the mean sediment 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.

6.4.2.2 In this EMAP protocol, substrate size and embeddedness will be evaluated at each of the 11 detailed cross sections using a combination of procedures adapted from those described by Wolman (1954), Bain et.al. (1985), and Plafkin et.al., (1989). The basis of the procedure is a systematic selection of 5 substrate particles from each of the 11 channel cross sections (Figure 5). In the process of measuring sediment sizes at each channel cross section, you will also make measurements of the wetted width of the channel and the water depths at each sediment sample point. If the wetted channel is split by a mid-channel bar (see section 6.2), the five substrate points shall be centered between the wetted width boundaries regardless of the bar in between. Consequently, sediment 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.

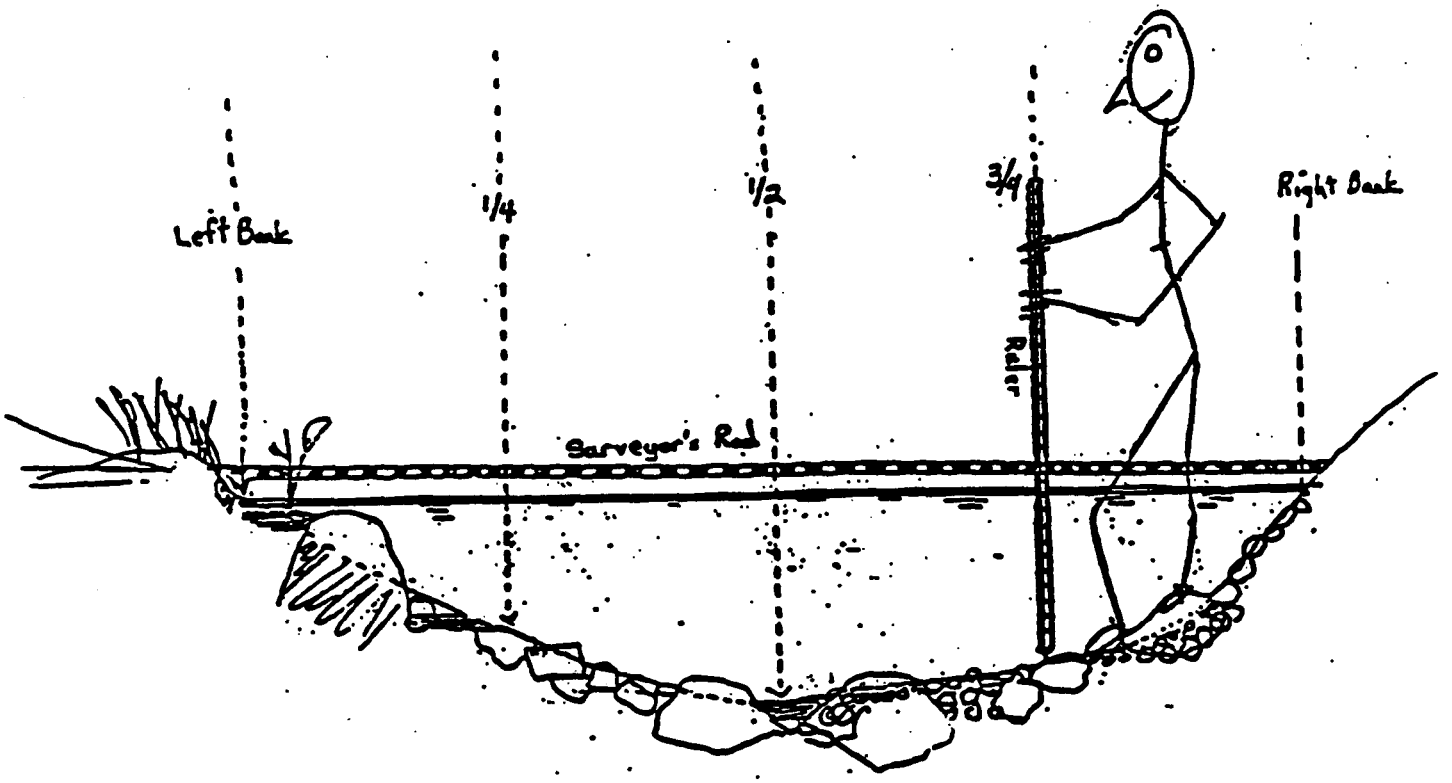


Figure 5. Substrate sampling cross-section.

1. At the cross-section station, lay the surveyors 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 width measurement of the channel by 4, and record in the left column "DistLB" of the form the distances corresponding with 1/4th, 1/2, and 3/4 the distance across the stream, starting with the left bank at zero. The distance you record to the right bank is the same as the wetted channel width. (NOTE: this is the same wetted width measurement is also recorded under "Bank Measurements" on the form (Section 6.4.3). The sediment sampling points along the rod or tape will be at the 1/4th, 1/2, and 3/4 positions, and at the water's edge just within the left and right banks.

3. Place your sharp-ended meter stick at position 1 (0m) at the end of the tape. Read and record the depth. Pick up the substrate particle (unless it is bedrock or boulder), and visually estimate its particle size, according to the following coded scale. To minimize bias in this method, it is important to concentrate on correct placement of the measuring stick along the tape 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).

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 —	(tennisball to basketball)
GC	Gravel (Coarse).....	16 to 64 mm —	(marble to tennisball)
GF	Gravel (Fine)	2 to 16 mm —	(ladybug to marble)
SA	Sand	0.06 to 2 mm —	(<ladybug size, but visible as particles - gritty between fingers).
FN	Fines	<0.06 mm —	Silt Clay Muck (not gritty between fingers)
WD	Wood	Regardless of Size	
OT	Other	Metal, Tires, Car bodies etc. regardless of size (Put in comments if use other)	

4. If the particle is larger than sand, look at the stains, markings, and algae on its surface. Estimate the percentage embeddedness (Figure 6), the fraction of the particle surface that was surrounded by (embedded in) fine sediments on the stream bottom. (For boulders not lifted out of the water, you'll have to estimate embeddedness by feel and judgment from their shape.)

5. Move successively to each of the remaining four positions along the cross section rod or tape, repeating steps 3 and 4. Repeat the entire procedure at each new cross section.

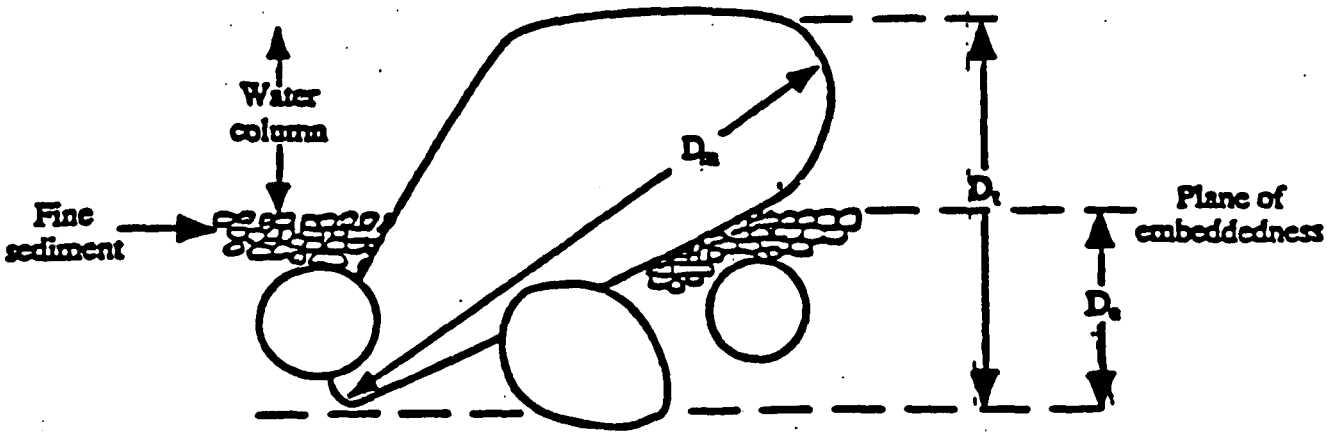
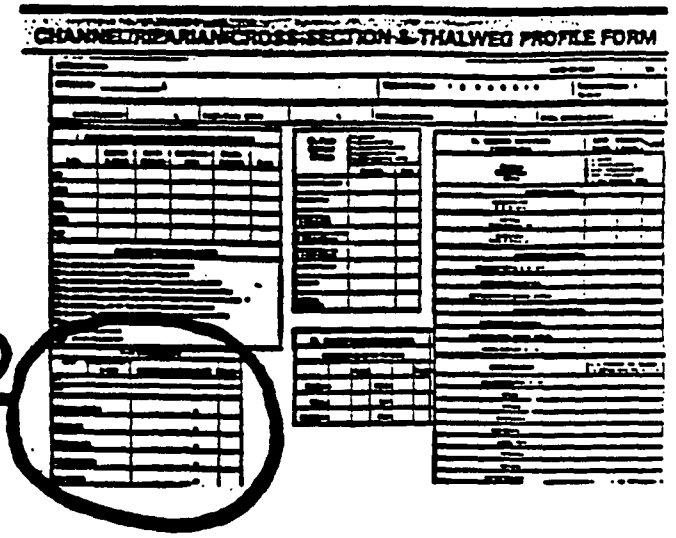


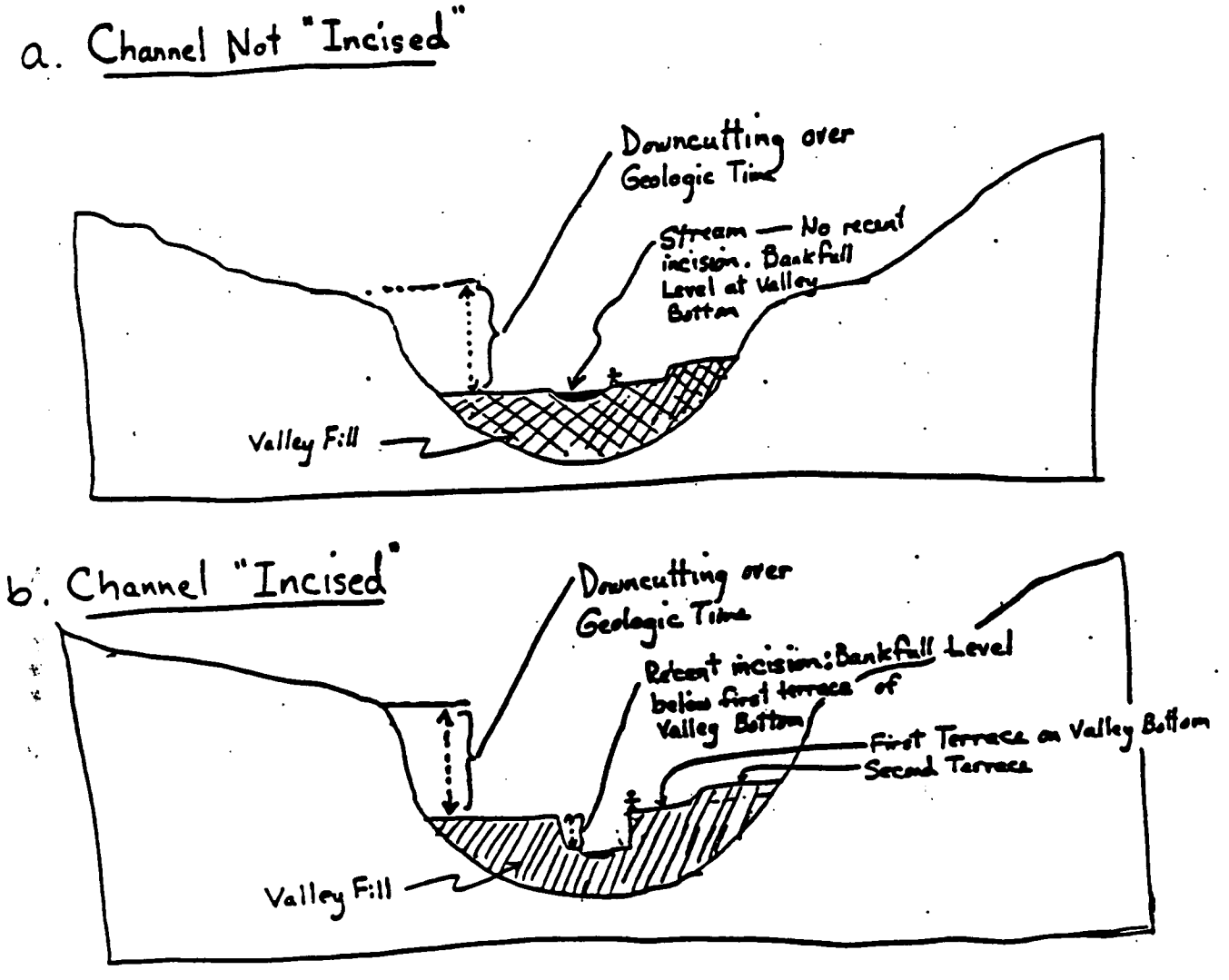
Figure 6. Schematic on determining embeddedness.

6.4.3 Bank Characteristics

II. Bank Measurements			
Loc.	Bank Angle 0-380	Undercut Dist (m) X.XX	Flag
LFT	•		
RGT	•		
Bankfull Width		__ __ . __ m	
Incised Ht.		__ __ . __ m	
Bankfull Ht.		__ __ . __ m	
Wetted Width		__ __ . __ m	
Bar Width		__ __ . __ m	



6.4.3.1 The bank characteristics to be measured are the bank angle, the amount of incision, and the amount of undercut; they shall be assessed on the left and right banks at each of the 11 cross sections. You will also measure and record the wetted width of the channel, the width of exposed gravel or sand bars in the channel, and the estimated height and width of the channel at bankfull stage. The "bankfull" or "active" channel is defined as the channel that is filled by moderate sized flood events that typically recur every 1 to 2 years. Such flows do not generally overtop the channel banks to inundate the valley floodplain, and are believed to be largely responsible for the observed channel dimensions in most streams. If the channel is not greatly incised, bankfull channel height and the amount of incision will be the same. However, if the channel is incised greatly, the bankfull level will be below the level of the first terrace of the valley floodplain, making "Bankfull Height" smaller than "Incision" (Figure 7). You will need to look for evidence of recent flows (within about 1 year) to distinguish bankfull and incision heights.



NOTE LEVEL OF BANKFULL STAGE RELATIVE TO ELEVATION OF FIRST TERRACE ON VALLEY BOTTOM (Stick figure for scale).

Figure 7. Schematic showing bankfull channel and incision for channels: (a) not recently incised, and (b) recently incised into valley bottom.

1. To measure bank angle, lay the surveyors rod or your meter stick down against the left bank, with one end at the water's edge (left bank defined when facing downstream). Lay the clinometer on the rod, read and record the bank angle in degrees, using the external scale on the clinometer. A vertical bank is 90 degrees; undercut banks have angles greater than 90 degrees approaching 180 degrees, and gradually sloped banks have angles less than 90 degrees. Procedures for measuring bank angle are shown in Figure 8, as adapted from Platts et al. (1983).

2. If the bank is undercut, measure the horizontal distance of the undercutting.

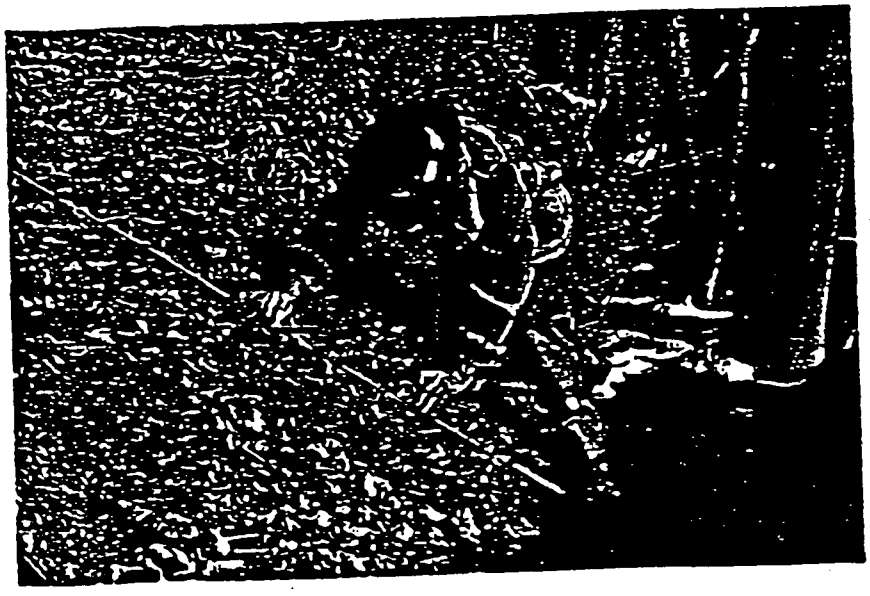
3. Repeat steps 1 and 2 on the right bank.

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 amount of 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). In cases where the channel is cutting a valley sideslope and has oversteepened and destabilized that slope, the bare "cutbank" is not necessarily an indication of recent incision. Examine both banks to make a more accurate determination of channel downcutting.

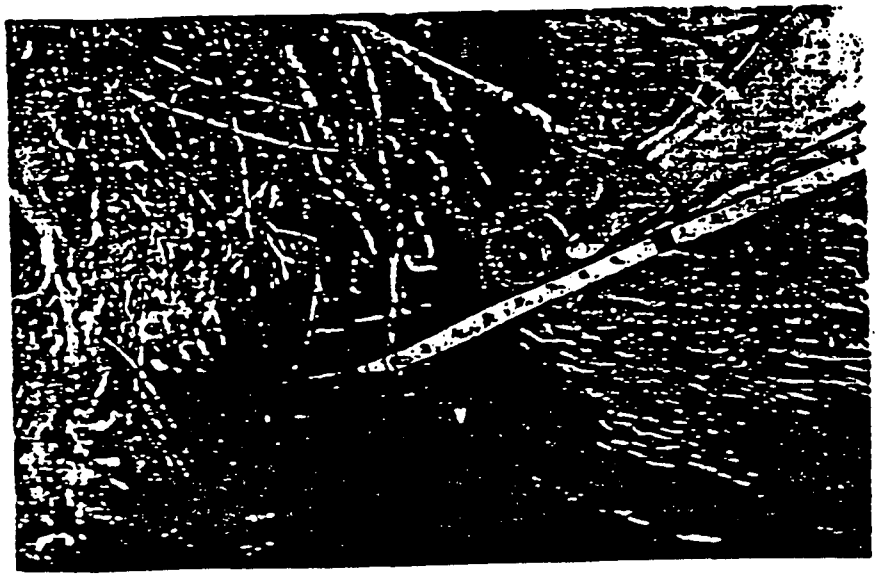
5. Still holding the surveyors rod as a guide, examine both banks to estimate and record the height of bankfull flow above the present water level. Spotting the level of bankfull flow during baseflow conditions requires judgement and practice; even then it remains somewhat subjective. In many cases there 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 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. The bankfull flow level may also be seen by the presence of drift material caught on overhanging vegetation.

6. Measure and record the bankfull channel width, the wetted width, and the width of exposed mid-channel bars (if present). The latter two measurements will probably be done while making substrate measurements across the channel.

7. Repeat steps 1 through 6 upon proceeding to each new cross section.



Using a clinometer to measure
a bank angle of 45°.

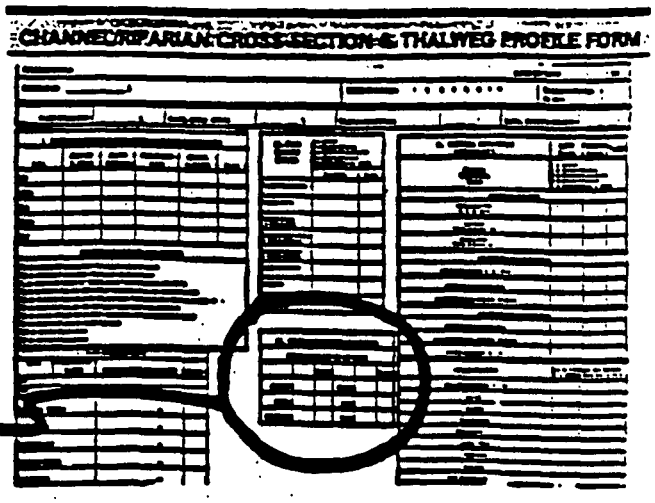


Using a clinometer to measure
a bank angle of 145°.

Figure 8. Measuring bank height and bank angle (Adapted from Platts et al., 1983).

6.4.4 Canopy Cover (Densiometer)

IV. CANOPY COVER MEASUREMENTS					
DENSIOMETER (0 TO 17 MAX)					
		FLAG			FLAG
CENUP			CENR		
CENL			LFT		
CENDWN			RGT		

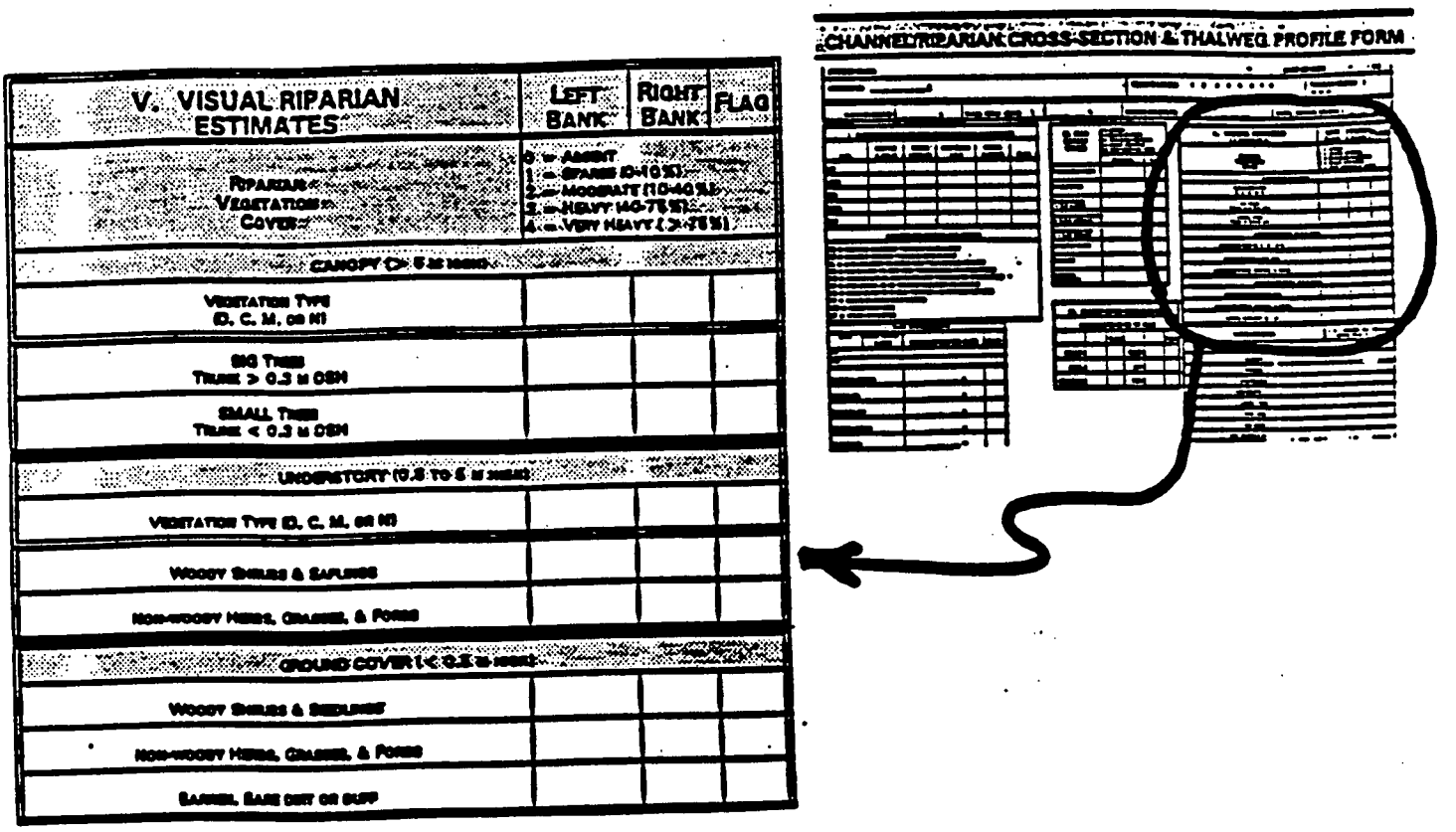


6.4.4.1 Riparian canopy cover over a stream is important not only in its role in moderating stream temperatures through shading, but also as an indicator of conditions that control bank stability and the potential for inputs of coarse and fine particulate organic material. Organic inputs from riparian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

6.4.4.2 Vegetative cover over the stream shall be measured at each of the 11 detailed cross section stations. This measurement employs the Convex Spherical Densiometer, model B (Lemmon, 1957). This instrument is available from only one vendor (see ordering information in section 6.7). The densiometer must be taped exactly as shown in Figure 8 to limit the number of square grid intersections to 17. To take a canopy cover density measurement, the observer looks down on the densiometer held just above waist level, concentrating on these 17 points of intersection. If the reflection of a tree or high branch or leaf overlies any of the intersection points, that particular intersection is counted as having cover. The measure to be recorded on the form is the count (from 0 to 17) of all the intersections that have vegetation covering them. Therefore, a greater number indicates increasing canopy extent and density. In doing the measurement, it is important that the densiometer be leveled using the bubble level (Figure 9).

6.4.4.3 For each of the 11 stations, densiometer measures are taken separately in four directions standing at the center of the stream. These measures will be used to estimate canopy cover over the channel. Then, standing at the wetted bank at the left and then the right side of the stream at each station, repeat the canopy density measurements facing the riparian vegetation (your back towards the stream). These bank densiometer readings complement your visual estimates of vegetation structure and cover within the riparian itself, and are particularly important in wide streams, where riparian canopy may not be recorded by the densiometer standing mid-stream.

6.4.5 Riparian Vegetation Structure



6.4.5.1 The previous section (6.4.4) described methods for quantifying the cover of canopy over the stream channel. The following visual estimation procedures are a semi-quantitative evaluation of the type and amount of different types of riparian vegetation. This assessment is used to evaluate the health and level of disturbance of the stream corridor. It also indicates the present and future potential for various types of organic inputs and shading.

6.4.5.2 Observations to assess riparian vegetation apply to the riparian area upstream 5 meters and downstream 5 meters from each of the 11 cross-section stations (Figure 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 10m X 10m riparian plot on each side of the stream (Figure 10). 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.

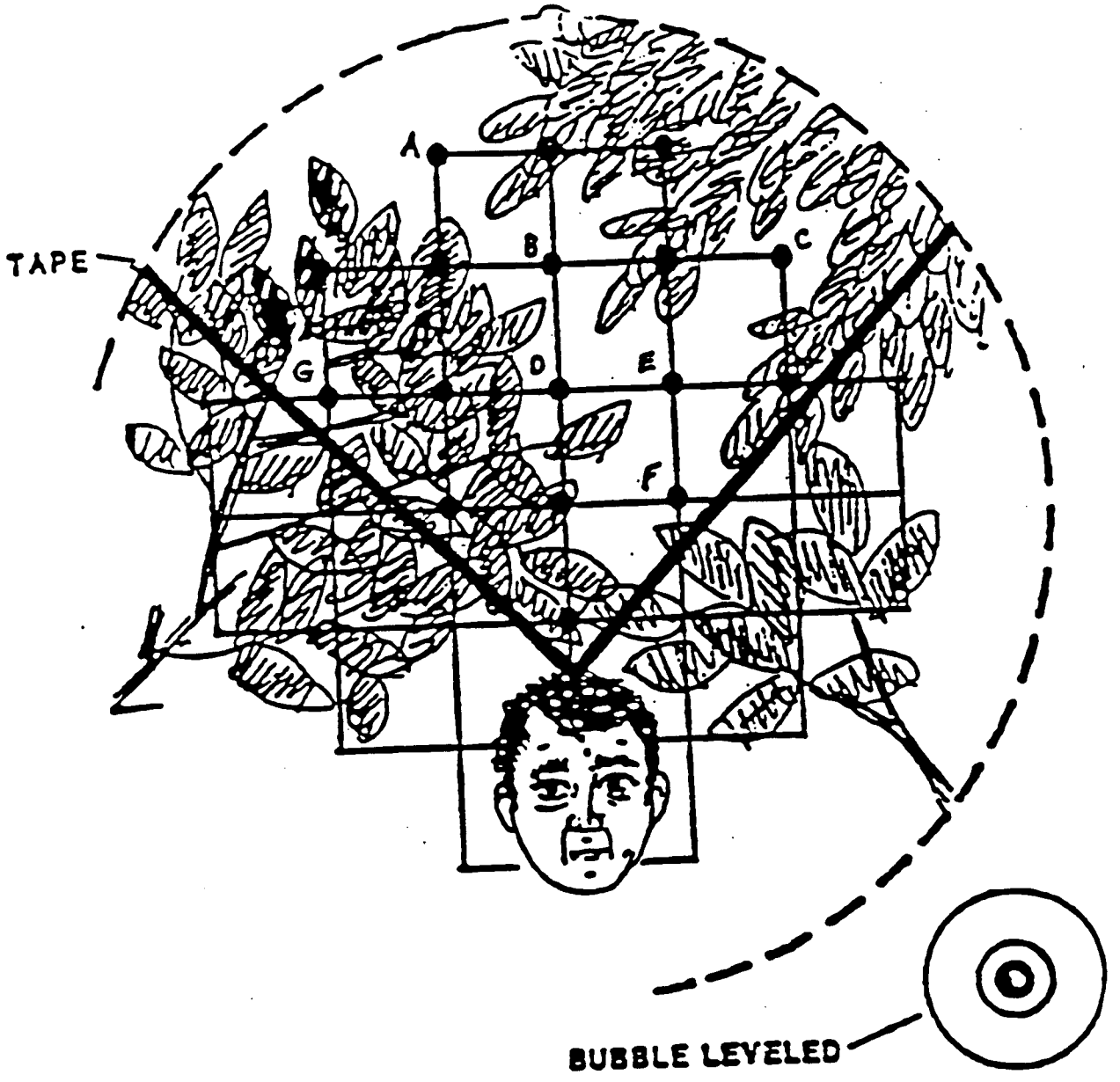
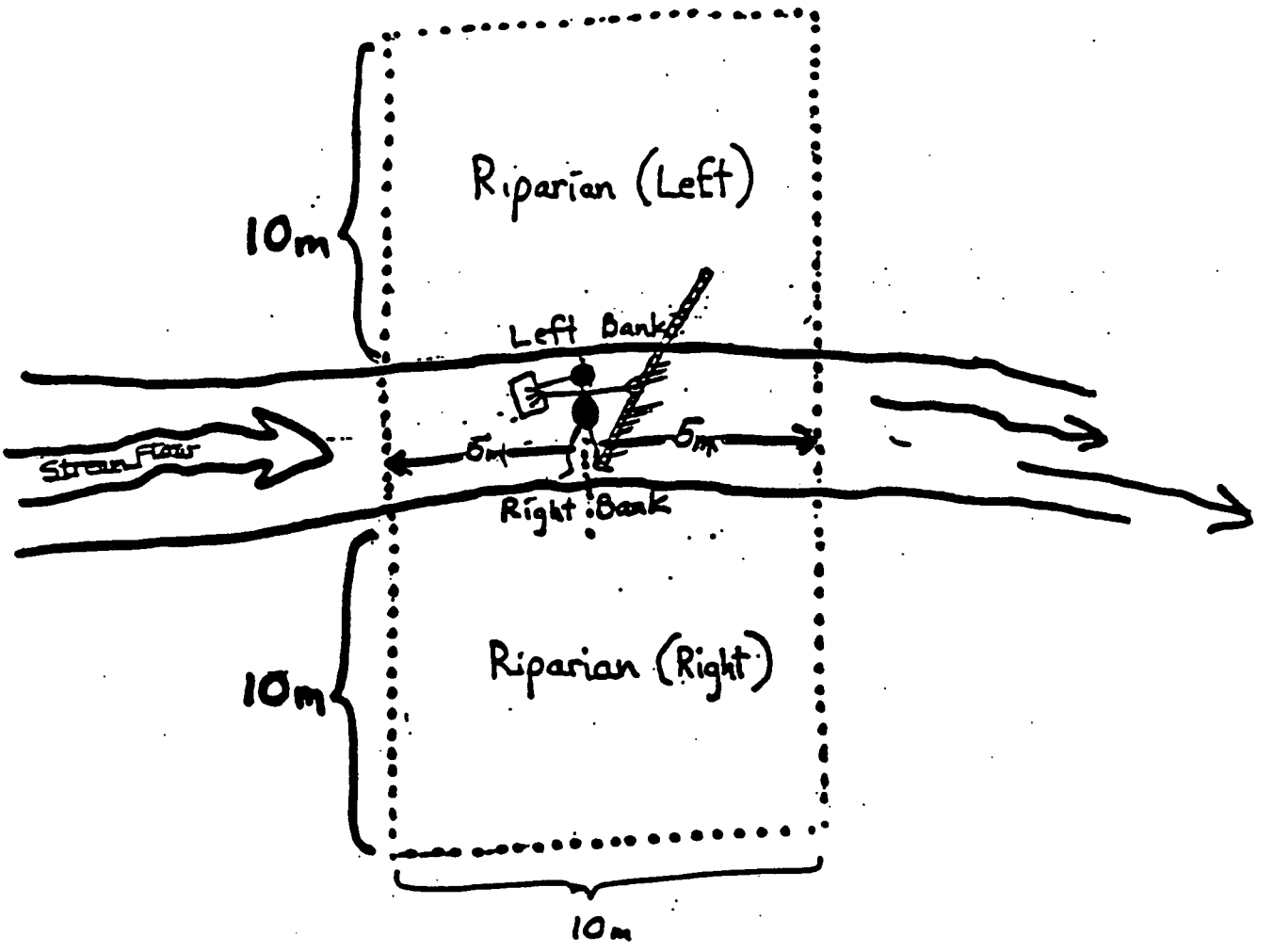


Figure 9. Schematic: Convex spherical canopy densiometer (From Mulvey et al., 1992). In this example, 10 of the 17 intersections show canopy cover, giving a densiometer reading of 10.



Riparian and Fish Cover Observation Limits Centered on Channel/Riparian Cross-Section Station

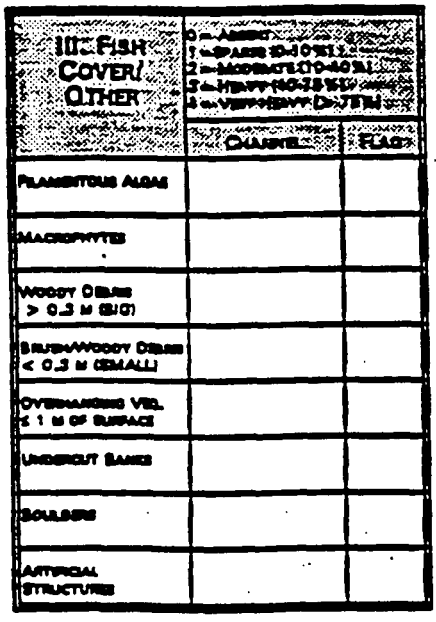
Figure 10. Boundaries for visual estimation of riparian vegetation, fish cover, and human influences.

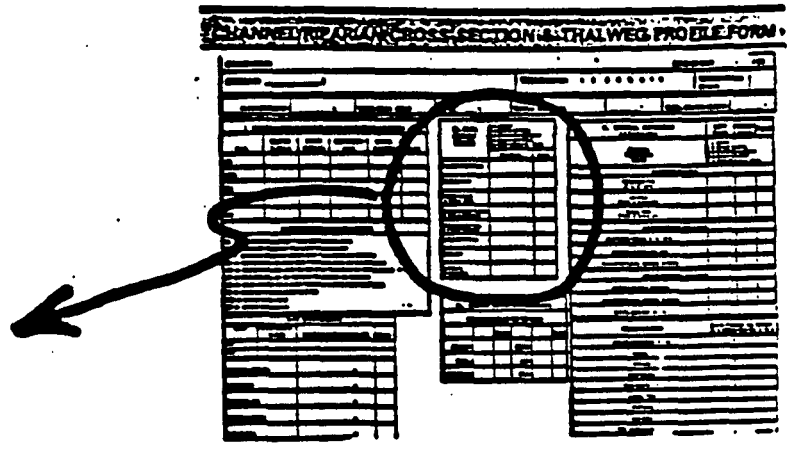
6.4.5.3 Conceptually divide the riparian vegetation into three layers: a CANOPY LAYER (>5m high), an UNDERSTORY (0.5 to 5m high), and a GROUND COVER layer (<0.5 high). Note that several vegetation types (eg. grasses or woody shrubs) can potentially occur in more than one LAYER. Similarly note that some things other than vegetation are possible entries for the "GROUND COVER" layer (eg. barren ground).

6.4.5.4 Before estimating the areal coverage of the vegetation layers, record the type of vegetation (Deciduous, Coniferous, 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.

6.4.5.5 You will estimate the areal cover separately in each of the three vegetation layers. Note that the areal cover can be thought of as the amount of shadow caste by a particular layer. The maximum cover in each layer is 100%, so the sum of the areal covers for the combined three layers could add up to 300%. The four entry choices for areal cover within each of the three vegetation layers are "0" (absent: zero cover), "1" (sparse: <10%), "2" (moderate: 10-40%), "3" (heavy: 40-75%), and "4" (very heavy: >75%). These ranges of percentage areal cover corresponding to each of these codes are also shown on the Field Form. When rating vegetation cover types, mixtures of two or more subdominant classes might all be given sparse ("1") moderate ("2") or heavy ("3") rankings. One very heavy cover class with no clear subdominant class might be ranked "4" with all the remaining classes either moderate ("2"), sparse ("1") or absent ("0"). Two heavy classes with 40-75% cover can both be ranked "3".

6.4.6 Fish Cover, Algae, Aquatic Macrophytes



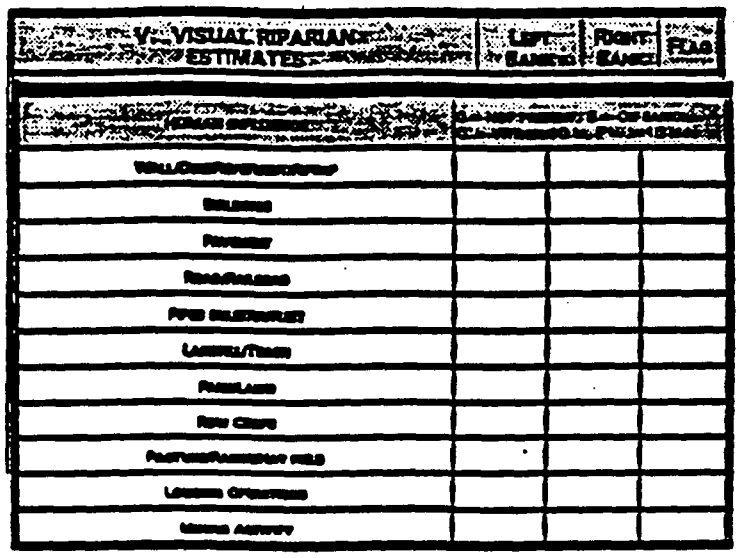


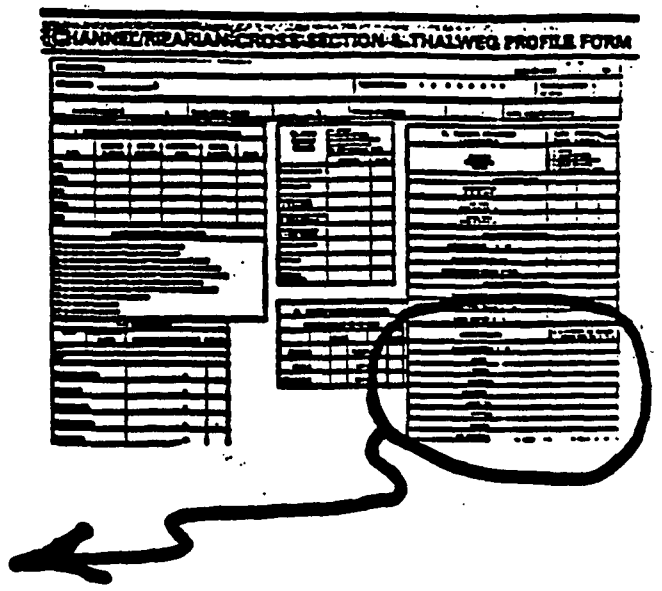
6.4.6.1 This portion of the EMAP physical habitat protocol is a visual estimation procedure that evaluates, semi-quantitatively, the type and amount of important types of cover for fish and macroinvertebrates. Alone and in combination with other metrics, this information is used to assess habitat complexity,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

6.4.6.2 Observations to assess fish cover and several other in-channel features apply to the channel area upstream 5 meters and downstream 5 meters from each of the 11 cross-section stations (Figure 10). The four entry choices for areal cover of fish concealment and other features are "0" (absent: zero cover), "1" (sparse: <10%), "2" (moderate: 10-40%), "3" (heavy: 40-75%), and "4" (very heavy: >75%). The ranges of percentage areal cover corresponding to each of these codes are the same as for riparian vegetation

6.4.6.3 Filamentous algae pertains to long streaming algae that often occurs in slow moving waters. Aquatic macrophytes are water loving plants 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. Brush/woody debris pertains to the smaller wood that primarily affects cover but not morphology. The tree or brush within one meter of the surface is the amount of brush, twigs, small debris etc. that is not in the water but is close to the stream and provides cover. Boulders are typically basketball to car sized particles. Many streams have artificial structures in them designed for fish habitat. Streams may also have in channel structures for diversions, impoundment and other purposes. Record the cover of these structures on the form.

6.4.7 Human Influences





6.4.7.1 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 landuse information to assess the potential degree of disturbance of the sample stream reaches.

6.4.7.2 At each of the 11 detailed Channel and Riparian Cross-Sections, evaluate the presence/absence and the proximity of 11 categories of human influences. Confine your observations to the stream and riparian area within 5m upstream and 5m downstream from the station (Figure 10). Entries are:

- B for any and all the items you observe ON THE STREAMBANK.

"C" for any and all the items that you observe within 10m from the streambank ("CLOSE).

"P" for those items that are PRESENT, but farther than 10m from the bank.

"0" for those NOT PRESENT.

Evaluate and record separately the left and right sides of the channel and banks. For human influence items outside of the riparian observation plots, mark "P" for each and every station where they are visible, even if you are observing the same item from station to station.

6.5 DISCHARGE MEASUREMENT

6.5.1 Background

6.5.1.1 If specified, discharge shall be measured on EMAP-SW sample reaches. Discharge measurements are critical in parts of EMAP that focus on trends in the acidity of streams, a characteristic which is very sensitive to streamflow differences. Discharge should be measured as close as possible to the EMAP "X" point where chemical samples are taken, so that the discharge and chemistry are a corresponding pair. However, different types of channel require different methods of discharge measurement, and the kinds of places within a channel where the three alternative types of discharge measurement methods can be employed differ. In streams sufficiently large to use the electromagnetic or impeller-type water velocity meter, discharge will be measured according to the standard Velocity-Area Method (Linsley et al., 1982) described in Subsection 6.6.2. In streams that are too small to employ that method, use the Portable Weir method (Subsection 6.6.3) or time the filling of a volume of water in a calibrated bucket (Subsection 6.6.4). If none of these methods can be employed, measure the cross-sectional depth profile and time the movement of a neutrally buoyant object (eg. an orange) through a measured (and recorded) length of the channel.

6.5.2 Velocity-Area Discharge Measurement

6.5.2.1 Electromagnetic Current Meter Calibration Check

NOTE: This procedure was written assuming a Marsh-McBirney Model 201D electromagnetic current meter is being used. This procedure may be used, with modification, with other meters meeting equivalent specifications.

1. Check meter calibration daily as part of morning routine, then again on site before entering the stream.

2. Calibration value should be 2.00 ± 0.05. The value obtained during morning calibration should be recorded in the comments section of the Discharge form. The values obtained onsite should also be recorded on the Discharge field form.

3. Once per week the zero value should be checked in a bucket of still water. The probe should sit for 30 minutes with no disturbance. The value obtained should be 0.0 ± 0.1. The meter zero should be adjusted if it is outside this range.

6.5.2.2 Calibration and Care of Impeller-Type Current Meter

NOTE: This procedure was written assuming a Swoffer Model 2100-14 is being used. It pertains, with modification, to other meters meeting equivalent specifications. Read the Owners' Manual and the Care and Operating instructions provided by the Manufacturer BEFORE using the instrument. As a reminder, some of the DO's and DON'TS are:

1. The propeller assembly, including the fibre-optic sensor, rotor bearing, rotor shaft and the propeller itself, is the single most important part of the instrument. Great care must be observed to insure that this precision instrument gives accurate output.

2. NEVER put the wading rod back into the carrying case, transport it, or store it with the propeller rotor assembly still attached.

3. The sensor and the rotor bearings may have to be cleaned after emersion in turbid water, or if they sink into soft sediments. Do not scratch the photo-optic sensor and do not use oil on the rotor bearing.

4. Before using, set the instrument to read in meters per second and calibrate as explained in the owners' manual.

6.5.2.3 Location for Discharge Measurements by Velocity-Area Method

6.5.2.3.1 The basis of this method is that stream discharge is equal to the product of the mean water velocity and the vertical cross sectional area flowing water. 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 11A). Each increment gives a subtotal of the stream discharge, and the whole is calculated as the sum of these parts. Discharge measurements shall be made at only one carefully chosen channel cross section in a stream reach. It is important to choose a channel cross section that is as much like a canal as possible. Pick a cross section where flow lines are directed downstream to the extent possible (low turbulence and a minimum of cross-flow). Glides with "U" shaped channel cross sections usually provide the best conditions for measuring discharge by the velocity-area method. Try to avoid cross sections with a lot of large boulders, woody debris, brush, exposed rocks, and other irregularities. Do not measure discharge in a pool.

6.5.3.3 Velocity-Area Procedures

1. Facing downstream and beginning on the left edge of the water, stretch a meter tape across the chosen stream location perpendicular to stream flow.

2. Measure and record the stream width. Leave the tape tightly suspended across the stream, approximately one foot above water level.

3. Divide the total stream width into 15 to 20 equal-sized intervals. To determine interval width, divide the total stream wetted width by 15 and round down to a convenient number. This should yield a sufficient number of intervals.

4. If using the electromagnetic current meter, attach its probe to the wading rod and check the internal electronics by turning the switch to "CAL." If the meter calibrates to 2.00 ± 0.05 in air, record this value in the comments section and proceed with the measurements. If using the impeller-type current meter, install its propeller on the sensor boom of the wading rod. Prior to making measurements, the meter shall have been set and calibrated to read in meters per second.

5. Move to the center of the interval number 1, closest to the left bank (looking downstream).

6. Read and record the stream depth in centimeters at the center of the interval.

7. Place the current meter probe at 0.6 of the total depth (measured from the surface). Orient the probe or propeller to face upstream, and make sure that your feet or legs are not deflecting or impounding flow that passes over the probe or propeller.

8. For the electromagnetic current meter (eg. Marsh-McBirney), use the lowest time constant scale setting on the meter that provides stable readings. Wait 20 seconds to allow the meter to

equilibrate, then measure and record the current velocity at the center of the interval. For the impeller-type meter (eg Swoffer 2100), set the control nob at the mid-position of "DISPLAY AVERAGING". Press "RESET" then "START" and proceed with the measurements. Wait 20 seconds for the meter reading to stabilize and read current velocity in meters per second.

9. Repeat steps (6)(7)(8) for all intervals across the stream.

6.5.4 Portable Weir Discharge Measurement

6.5.4.1 Discharge in channels too "small" for the velocity-area method can be measured with a portable, sharp-crested, 60 degree V-notch weir. Small can be defined as a channel so shallow that the probe can hardly be placed in the water or the stream is so broken up and irregular due to rocks and debris that a velocity area method measurement is not possible. The weir must completely block the flow and reroute it through its notch. It must be high enough to create a small spillway as water flows over its sharp crest. In other words, don't let the water level downstream interfere with the smooth plunge of the water flowing over the weir. In addition, the weir must create a relatively flat surface of impounded water upstream for a distance at least equal to the width of the stream.

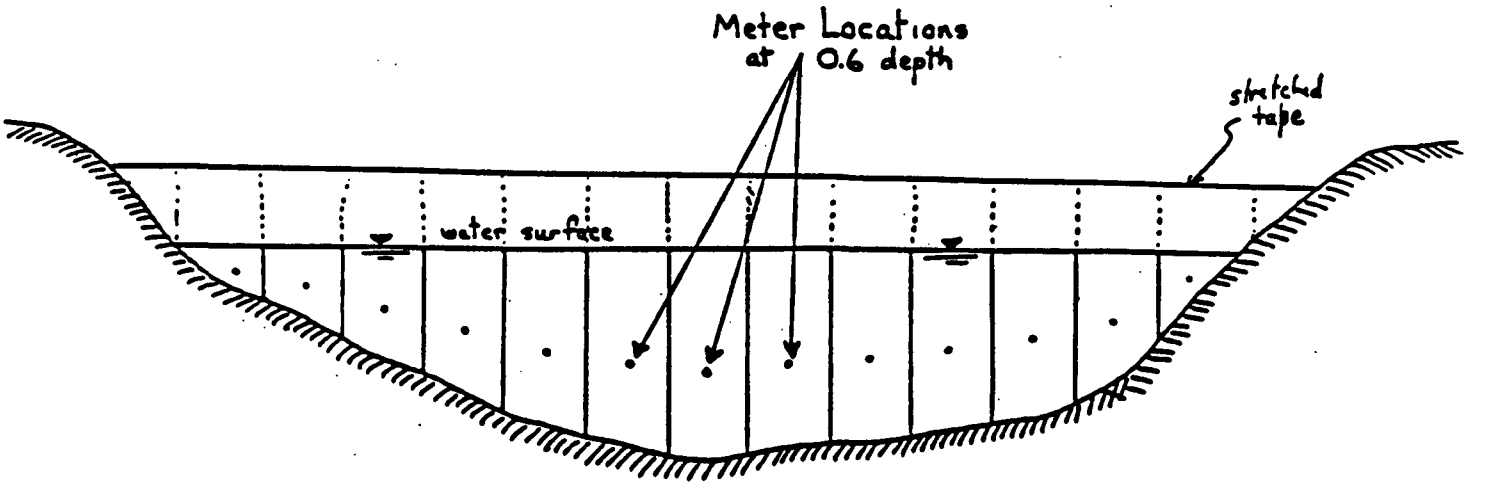
6.5.4.2 Choose a stream cross section that is narrow and easy to block with the portable weir. Ideal sections might be the "lip" or "tail out" below a pool. A bad choice would be right in the middle of a cascade. Impound the flow with the weir, making sure that water is not flowing beneath or around the side of the weir. Use mud or stones and plastic sheeting to get a good waterproof seal. Insure that the weir is level from side to side, and that it is perpendicular to the streamflow. When the depth of flow stabilizes, read and record the height of impounded water above the V-notch (Figure 11B). It may take some time for the flow to equilibrate (eg., 15 min), as the impounded pond builds upstream from the weir. For small discharges, water velocities through the weir will be slow, allowing this height to be measured very close to the weir. Larger discharges result in more rapid velocities, producing a pronounced downward curvature of the water as it approaches the crest of the weir. The height measurement in these cases must be well back upstream (say 0.5 m) from the weir, and may necessitate using the ruler and clinometer as a level to match the pool height against the weir.

IF INSTALLING THE TEMPORARY WEIR REQUIRES A LOT OF CHANNEL DISTURBANCE OR STIRS UP A LOT OF MUD, WAIT TO DO THIS UNTIL AFTER ALL BIOLOGICAL AND CHEMICAL MEASUREMENTS AND SAMPLING HAVE BEEN COMPLETED.

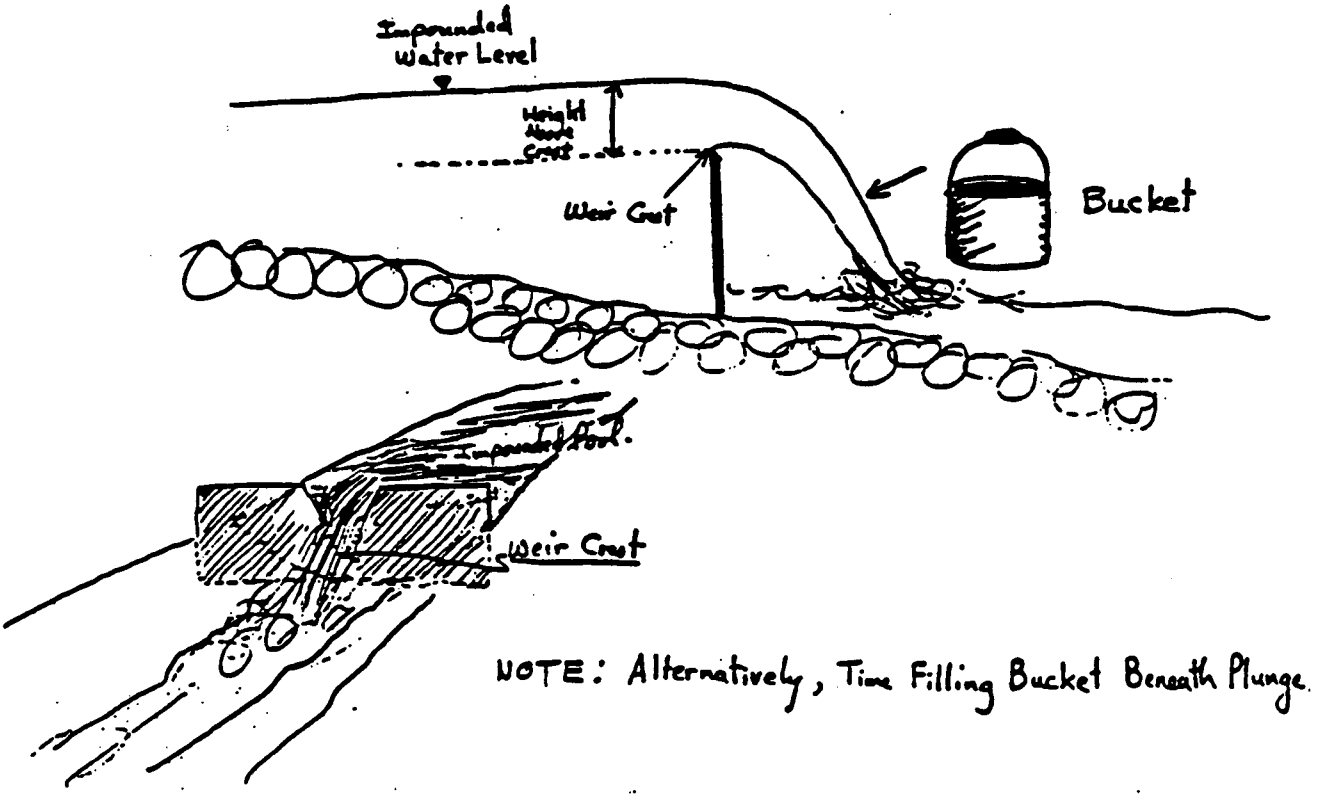
6.5.5 Timed Bucket Discharge Measurement

6.5.5.1 Where feasible in very small streams, discharge can be measured directly, by recording the time it takes to fill a known volume. This can be an extremely precise and accurate method. Choose a natural spillway or plunge, or construct a temporary one using a weir, using plastic sheet or on-site materials. Using a stopwatch and a calibrated bucket held beneath the spillway, record the filling time and the volume of water collected in the bucket (see Note in Figure 11B). Make sure that the entire flow of the stream is going into the bucket. Repeat the procedure 10 separate times. You may need to time the volumes obtained from more than one different spillway in the same channel cross section. If so, indicate clearly the time and volume data replicates that should be averaged together for each spillway.

IF THE TIMED BUCKET DISCHARGE MEASUREMENTS INVOLVE A LOT OF CHANNEL DISTURBANCE OR STIR UP A LOT OF MUD, WAIT TO DO THIS UNTIL AFTER ALL BIOLOGICAL AND CHEMICAL MEASUREMENTS AND SAMPLING HAVE BEEN COMPLETED.



A. Velocity-Area Discharge Measurement



B. Portable Wier/Bucket Discharge Measureme

Figure 11. Discharge measurements

6.6 EQUIPMENT LIST

For Physical Habitat Profile Measurements:

1. Surveyors Rod (Metric, Fiberglass, 3-part Telescoping, approx 5m extended about 1.5 m collapsed) – for width and depth meas's. (about $80)

2. Clinometer (SUUNTO Model PM5/360PC or equivalent) must read both in percent and degrees slope — for gradient meas's (about $105)

3. Spherical (Convex) Canopy Densiometer. (Lemmon, Model B —only one manufacturer: Robert Lemmon, Forest Densiometers 5733 S.E. Cornell Dr., Bartlesville, Oklahoma 74006, phone 918-333-2830) (about $80).

4. Tripod (modest price camera tripod, light, telescoping) for backsite on previous cross-section to obtain gradient (tripod necessary only if Physical habitat work is done by one person alone). (about $10-$30)

5. Bearing Compass (Backpacking type, e.g., Silva Ranger, as used in NSS) (About $40)

6. Colored plastic Flagging (2 colors)

7. "Rebar" to monument site, sheet aluminum label and aluminum nails for to nail into tree.

8. 2 Covered Clipboards (lightweight, with strap or lanyard to hank around neck – Camera straps work great for straps.)

9. Pencils, soft lead (good "fat lead" mechanical, disposable or whatever works best – need lots of extras)

10. Chest Waders (1 each person) – with felt-soled boots for safety and speed if waders are the neoprene "stocking" type.

11. Hip Waders (1 each person) – with felt soles for safety and speed on slippery rocks.

12. Camera, waterproof 35mm with standard and wide angle lense (small point and shoot would probably be better as well as cheaper). (About $100-$250)

13. Film (35mm, color slide film, ASA 400 and 100) Will use 10 to 20 frames per stream reach.

14. Fiberglass Tape & reel (50 m metric), with good hand crank and handle for measuring cross-sections. (About $30-$45)

15. Meter stick (cheap wooden ruler with metric calibrations) for bank angle meas's.

16. Hip Chain for measuring of reach and valley transects. (Optional)

Note: The surveyors rod, clinometer, bearing handheld compass, fiberglass tape, and hip chain can be bought from either Forestry Suppliers or Ben Meadows Company. All other items can be bought from department or sporting goods stores.

For Discharge Measurement :

1. Electromagnetic Water Velocity Meter (Mgf: Marsh-McBirney Mod 201D or similar) or Impeller-Type Water Velocity Meter (Mgf: Swoffer Model 2100-14), with adjustable wading rod (metric calibrations).

2. Portable Weir -- 60 degree V-notch (this is a plastic plate to impound and measure flow in streams too small for the Velocity-Area Method).

3. Bucket, Plastic with volume graduations -- for timed bucket discharge measurements.

4. Stopwatch -- I prefer mechanical over electronic --mechanical ones more reliable in wet situations. (Used for timed volume discharge measurements.)

Note: The Weirs and Water Velocity meters can be bought from suppliers like Ben Meadows or Forestry Suppliers. The Meters can also be bought directly from manufacturer.

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6.8 APPENDIX: FIELD FORMS FOR PHYSICAL HABITAT PROTOCOL

CHANNEL/RIPARIAN CROSS-SECTION & THALWEG PROFILE FORM

STREAM NAME: DATE OF VISIT: / / 94

STREAM ID: _ _ _ _ _ _ S | TEAM ID (circle): 1 2 3 4 5 6 7 8 | Transect Station (A to K):

Slope (Backsite)	_ _ . _ %	Suppl. Slope (Bkst)	_ _ . _ %	Bearing (Backsite):	_ _ _ °	Suppl. Bearing (Bksst)	_ _ _ °

I. Substrate Measurements Cross-Sectional Information

Loc.	Dist LB X.XX m	Depth XXX cm	Size Class Code	Embed. 0-100%	Flag
LFT					
LCTR					
CTR					
RCTR					
RGT					

Substrate Size Class Codes

RS = Bedrock (smooth) *(larger than a car)
RR = Bedrock (rough) *(larger than a car)
BL = Boulder (250 to 4000 mm) *(basketball to car)
CB = Cobble (64 to 250 mm) *(tennis ball to basketball)
GC = Coarse gravel (16 to 64 mm) *(marble to tennis ball)
GF = Fine gravel (2 to 16 mm) *(ladybug to marble)
SA = Sand (0.06 to 2 mm) *(gritty - up to ladybug size)
FN = Silt/clay.muck (not gritty)
WD = Wood (any size)
OT = Other (comment)

II. Bank Measurements

Loc.	Bank Angle 0-360	Undercut Dist (m) X.XX	Flag
LFT	°		
RGT	°		

Bankfull Width	_ _ . _ m	
Incised Ht.	_ _ . _ m	
Bankfull Ht.	_ _ . _ m	
Wetted Width	_ _ . _ m	
Bar Width	_ _ . _ m	

III. Fish Cover/ Other

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

	Channel	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		

IV. Canopy Cover Measurements

Densiometer (0 to 17 max)

		Flag			Flag
CenUp			CenR		
CenL			Lft		
CenDwn			Rgt		

V. Visual Riparian Estimates

Riparian Vegetation Cover	Left Bank	Right Bank	Flag
0 = Absent 1 = Sparse (0-10%) 2 = Moderate (10-40%) 3 = Heavy (40-75%) 4 = Very Heavy (> 75%)			
Canopy (> 5 m high)			
Vegetation Type (D, C, M, or N)			
Big Trees Trunk > 0.3 m DBH			
Small Trees Trunk < 0.3 m DBH			
Understory (0.5 to 5 m high)			
Vegetation Type (D, C, M, or N)			
Woody Shrubs & Saplings			
Non-woody Herbs, Grasses, & Forbs			
Ground Cover (< 0.5 m high)			
Woody Shrubs & Seedlings			
Non-woody Herbs, Grasses, & Forbs			
Barren, Bare dirt or duff			

Human Influence	0 = Not present, B = On bank C = Within 10 m, P = > 10 m		
Wall/Dike/Revetment/Riprap			
Buildings			
Pavement			
Road/Railroad			
Pipes (Inlet/Outlet)			
Landfill/Trash			
Park/Lawn			
Row Crops			
Pasture/Range/Hay field			
Logging Operations			

Reviewed by (initial)_

THALWEG PROFILE & WOODY DEBRIS FORM-STREAMS

STREAM NAME:

DATE OF VISIT: / /

STREAM ID: _ _ _ _ _ _ S

TEAM ID (circle): 1 2 3 4 5 6 7 8

Transect Station (A to K):

THALWEG PROFILE | REACH LENGTH m | INCREMENT m

STA. #	THALWEG DEPTH (CM) XXX	WETTED WIDTH (M) XX.X	BAR WIDTH (XX.X)	SOFT/FINE SEDIMENT Y OR N	CHANNEL UNIT CODE XX	POOL FORM CODE	SIDE CHANNEL (SC)?	COMMENTS
0								
1								
2								
3								
4								
5								
6								
7								
8								
9								

LARGE WOODY DEBRIS (≥ 10 CM SMALL END DIAM.: ≥1.5 M LENGTH)
TALLY BETWEEN THIS TRANSECT AND NEXT

DIAMETER LARGE END	PIECES PARTLY IN BKFL. CHAN.			PIECES BRIDGE ABOVE BKFL. CHAN.		
	LENGTH 1.5 - 5 M	5 - 15 M	> 15 m	LENGTH 1.5 - 5 M	5 - 15 m	> 15 m
.1 to <0.3 m						
.3 - 0.5 m						
.5 - 0.8 m						
> 0.8						

FLAG CODES: K = NO MEASUREMENT MADE; U = SUSPECT MEASUREMENT: F1, F2, ETC. = MISC. FLAGS ASSIGNED BY EACH FIELD CREW. EXPLAIN ALL FLAGS IN COMMENTS SECTION

COMMENTS:

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 comment

SECTION 7

STREAM PERIPHYTON: FIELD PROCEDURES

7.1 SAMPLING RATIONALE

7.1.1 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 acids.

7.2 FIELD EQUIPMENT

1. Large funnel (15-20 cm diameter).
2. Scrape area delimiter (3.8 cm diameter pipe, 3 cm tall).
3. Stiff-bristle toothbrush with handle bent at 90° angle.
4. Wash bottle.
5. Collection bottle to catch removed periphyton.
6. 60 mL syringes with 3/8" hole bored into the end.
7. 50 mL centrifuge tubes or similar sample vials.
8. Formalin.
9. Glass-fiber filters (0.45 um average pore size) for chlorophyll a.
10. Pre-leached, pre-ashed, weighed glass-fiber filters (0.45 um average pore size) in numbered pans for ash free dry mass (AFDM).
11. Forceps for filter handling.
12. Millipore-type filtration apparatus with 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.
16. Dry ice or other means of freezing sample.

7.3 FIELD PROTOCOLS

7.3.1 Collection

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!**

8. Four subsamples will be taken from each composite sample. These are:

a. <u>Identification/Enumeration</u>

1) Withdraw 50 mL of mixed sample and place in a labelled sample vial (50 mL centrifuge tubes work well). Cover label with clear tape.

2) Preserved sample with 2 mL of 10% formalin. Gloves should be worn.

3) Tightly cap tube and tape with electrical tape.

b. <u>Chlorophyll a</u>

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, and 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 labelled 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.

NOTE: ALL SAMPLES MUST BE CAREFULLY LABELLED WITH THE APPROPRIATE BAR-CODE AND ADHESIVE TAGS!

7.4 SHIP SAMPLES TO:

Dr. James M. Lazorchak
U.S. EPA
3411 Church Street
Cincinnati, OH 45244
Phone: (513) 569-7076
Fax: (513) 569-7078

SECTION 8

BENTHIC (SEDIMENT) METABOLISM: FIELD METHODS

1. SCOPE AND APPLICATION

1.1 Ecosystems are complex, self-regulating, functional units defined by rates and processes, such as energy flow or material cycling. Functional indicators are those metrics that measure energy flow and material transformation within the ecosystem.

1.2 The method outlined here is designed for headwater to mid-order streams, though it may be adapted for larger rivers or lakes.

2. SUMMARY OF METHOD

2.1 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 40X channel width. Following incubation, DO is remeasured and the sediments saved for ash free dry mass (AFDM) analysis. Respiration, the decline in DO within each microcosm, is adjusted for AFDM, yielding a measure of community respiration /g AFDM. Organic carbon turnover time can be calculated from the empirical relationship between organic carbon (estimated as AFDM X 0.5) concentration of the sediment and oxygen consumption.

3. DEFINITIONS

3.1 AFDM - ash free dry mass

3.2 DO - dissolved oxygen

4. INTERFERENCES

4.1 Conditions which may interfere with this method are unknown. The method has been performed successfully in waters containing high metal concentrations, high conductivity, and moderate salinity.

5. SAFETY

5.1 Gloves should be worn when collecting, compositing, mixing, and subsampling sediments.

6. EQUIPMENT AND SUPPLIES

6.1 Ice chest for incubating centrifuge tubes

6.2 1 L Nalgene beaker to support centrifuge tubes during incubation

6.3 Scoop sampler for sediments.

6.4 50 mL, screw-top, centrifuge tubes.

6.5 YSI Model 58 Dissolved Oxygen meter with Model 5905 Stirring BOD probe.

6.6 Spare batteries for D.O. meter.

6.7 Permanent markers for labelling tubes.

6.8 Sample labels and field data sheets.

6.9 Zip-lock sandwich bags to contain replicates from each treatment and site.

6.10 Ice chest with dry ice for sample freezing (or other means of freezing samples).

7. REAGENTS AND STANDARDS

None.

8. SAMPLE COLLECTION, PRESERVATION, AND STORAGE

8.1 Collect sediment samples by gently scooping the top 4 cm of soft, surface sediments from depositional habitats (pools, eddys, backwaters) within the study reach. Samples are collected near Transects B-J, as defined by the Physical Habitat reach delineation. If soft sediments are abundant, collect samples at or near each transect. If soft sediments are scarce, collect them wherever you can within the reach. A total of 3 L of sediments are needed for metabolism and sediment toxicity analyses.

8.2 Fill ice chest 2/3 full with stream water and record temperature and dissolved oxygen (D.O.).

8.3 Thoroughly mix composited sediments.

9. QUALITY CONTROL

9.1 All field work conducted during this study will be done by personnel having prior experience with the designated methods, or under the direct supervision of experienced personnel.

9.2 Sediment metabolism will be sampled at each site on all site visits.

9.3 Quality Assurance Objectives:

9.3.1 Precision--Precision of measurements is assured by carefully following the method protocols, including calibration of DO meter.

9.3.2 Completeness--Valid data are required from 80% of the sites visited.

9.3.3 Representativeness--Sediment metabolism is being determined for depositional areas of streams. Replicate samples from these areas in a stream segment (defined by fish survey) are composited and mixed to provide a representative site sample.

9.4 Documentation and Review: Sediment metabolism field data are recorded and checked for completeness and accuracy before leaving the site. All data sheets are inspected for completeness,

accuracy, and legibility before proceeding to the next sample. Raw data sheets are retained in a file, and the data is entered into an ASCII file.

10. CALIBRATION AND STANDARDIZATION

10.1 Before the field season, meters will be calibrated against the Winkler titration method of DO determination.

10.2 Dissolved Oxygen Meter Calibration (for YSI model 58, with YSI model 5905 stirring BOD probe)

10.3 Each day, before leaving lab, motel, or base station:

10.3.1 Check meters batteries to ensure that meter and stirring probe are operational.

10.3.2 Check probe membrane to ensure that it is not frayed or torn and that there are no bubbles under the membrane. If membrane is not intact or has entrapped bubbles, it should be replaced according to the manufacturer's directions.

10.4 Upon arriving at each site:

10.4.1 Zero meter according to manufacturer's directions.

10.4.2 Calibrate meter using the water-saturated atmosphere method described in the meter's operations manual.

11. PROCEDURES

11.1 Remove any visible organisms from the sediment before placing approximately 10 mL of sediment in each of 5 labelled, 50 mL screw-top centrifuge tubes. Labels are covered with clear tape.

11.2 Fill each tube to the top (no head space) with stream water from the ice chest and seal.

11.3 Fill 1 additional tube with stream water only and seal.

11.4 Incubate in closed ice chest for 2 hours.

11.5 Measure D.O. in each tube.

11.6 Respiration is the difference between initial and final D.O.

11.7 Decant overlying water and save sediment.

11.8 Seal tubes and place on dry ice as soon as possible.

11.9 Store frozen for laboratory analysis.

11.10 If you are collecting samples for sediment toxicity tests, save remaining composite sediment sample by double-bagging in labelled, zip-lock freezer storage bags. Label is covered with clear plastic tape.

11.11 Store sediment toxicity sample chilled (but not frozen!) for laboratory analysis.

11.12 NOTE: All samples must be carefully labelled with the appropriate bar-code and adhesive tags. Tags need to be covered with clear tape.

11.13 Ship samples to:

Dr. James M. Lazorchak
US EPA-EMSL
3411 Church Street
Cincinnati, OH 45244
Phone 513/569-7076
Fax 513/569-7078

12. DATA ANALYSIS AND CALIBRATION

N/A

13. METHOD PERFORMANCE

N/A

14. POLLUTION PREVENTION

N/A

15. WASTE MANAGEMENT

1.5.1 For normal respiration measurements, no waste is produced. Water may simply be decanted into the stream from which it came.

16. REFERENCES

N/A

17. TABLES, DIAGRAMS, FLOWCHARTS, AND VALIDATION DATA

None.

SECTION 9

BENTHIC INVERTEBRATE FIELD METHODS

9.1 INTRODUCTION

9.1.1 Rationale

9.1.1.1 This protocol is intended for evaluating the biological integrity of wadeable streams of EPA Region III of 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 (RBP) 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 RBP is replaced in this protocol with a kick net modified for use by one person.

9.1.2 Importance of Monitoring Benthic Assemblages

9.1.2.1 Benthic invertebrates inhabit the sediment or live on the bottom substrates of streams. The benthic macroinvertebrate assemblages in streams reflect overall biological integrity of the benthic community such that monitoring these assemblages are useful in assessing the status of the water body and monitoring trends. 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 (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.

9.1.3 Sampling Design

9.1.3.1 The protocols described here employ a randomized, systematic spatial sampling design that minimizes bias in positioning the sampling stations. Sampling stations are randomly located along the transects established according to the Physical Habitat Assessment Protocol. Eleven transects will be systematically marked off equal distance apart in proportion to stream size. Macroinvertebrate assemblages will be collected at the nine interior transects at either the 1/4th (left), 1/2 (middle), or 3/4 (right) points on each transect as determined by the throw of a die. If the die comes up a one or two the sample will be collected at the 1/4 point, a three or four would indicate the 1/2 point, and a five or six would indicate the 3/4 point. Left and right are determined when facing downstream.

9.1.4 Components of the Macroinvertebrate Indicator Protocols

9.1.4.1 There are two Macroinvertebrate Indicator Protocols. The BASE Protocol will be utilized at all EMAP-SW and REMAP Sites. It consists of the collection of nine samples with the 595 µm modified kick net and combining the riffle samples in one bucket and the pool samples in another bucket. The combined riffle samples become the composite riffle sample and the combined pool samples become the composite pool sample. The SORTING-IDENTIFICATION Protocol describes the procedure that must be followed during the sorting and identification of the samples (see the Laboratory Manual).

9.1.5 Objectives of the Study

9.1.5.1 The primary objective of the 1994 EMAP-SW pilot study is to determine if nine modified kick net samples separated into a composited riffle sample and a composited pool sample will adequately characterize a stream segment in 1st and 2nd order streams throughout EPA Region III. A second objective is to determine the relative importance of the riffle and the pool sample in assessing stream condition.

9.2 PROCEDURES FOR FIELD CREWS

9.2.1 Preparing for the Sampling Day

9.2.1.1 Before leaving the Base Site for the day's sampling site(s). Check the conductivity meter and DO meter for proper operation and be sure the batteries are well charged. Go over the equipment checklist (Table 1) to be sure all equipment is loaded in the vehicle and in good working condition. Place one gallon of ethanol per site in a cooler which has vermiculite on the bottom. Check maps for all sampling sites to be visited. DO NOT place barcodes on the jars before going into the field because doing so causes tracking problems if any labeled jars are not used at the site they were prepared for.

9.2.2 Upon Arrival at the X-site.

9.2.2.1 Record the time, weather conditions, and initial observations in the field note book and fill in the information requested at the top of the Checklist (Table 2). While the habitat assessment team marks off the 11 transects, the benthos crew will do the water chemistry following the Protocol for Water Chemistry. By the time the water chemistry measurements are completed and the water chemistry samples are collected, the habitat assessment crew should have the downstream transects marked off so that the benthos crew can begin to collect the benthic invertebrate samples.

9.2.3 BASE Protocol for Collection of the Macroinvertebrate Composite Samples

9.2.3.1 Carry benthos sampling gear to the first sampling station. The first sampling station is transect B (the first transect upstream from the downstream end of the study segment and will be marked by flag bearing the letter B). If the transects have not been established by the physical habitat crew, the benthos crew will need to set up the transects following the Physical Habitat Protocols.

9.2.3.2 From a random numbers table or roll of a die pick a number (if not already done by the Physical Habitat Crew) between one and three to determine if the sample will be taken at the 1/4 (left side), 1/2 (middle), or 3/4 (right side) point of the transect (1 or 2 = 1/4, 3 or 4 = 1/2, 5 or 6 = 3/4). After randomly picking sampling point at the first transect, systematically sample from left, middle, right for the remaining eight transects. In streams only two nets wide pick left or right, or if the stream is only one net wide collect entire stream width. Be sure to circle R, C, or L on field forms for each transect sampled.

9.2.3.3 Determine if the stream at this station consists of a riffle, run, or pool and collect the sample following the protocol for that habitat (See Subsection 9.2.3.4 - Sampling Protocol for Riffles and Runs or Subsection 9.2.3.5 - Sampling Protocol for Pools). For the purpose of this section any area where there is not sufficient current to collect a sample by the riffle protocol, would be defined as a glide-pool and

sampled according to the protocol for pools. If it is impossible to sample the station with the modified kick net following either of these protocols, spend about 60 seconds hand picking a sample from about 0.5 m^2 of substrate at the station and combine it with the other samples in either the "riffle" or "pool" bucket. Repeat the procedure to collect a sample from each of the nine established interior transects (transects B-J). All of the riffle samples will be combined in one bucket and the pool samples will be combined in another bucket.

9.2.3.4 Procedures for Collecting Macroinvertebrates from Riffles and Runs with the Modified Kick Net Sampler

9.2.3.4.1 A modified Kick Net with 595 µm mesh openings and 0.5 m nylon bag (Wildco # 425-C50) will be used to collect macroinvertebrates in riffles and runs. Begin with the most downstream station so as not to disturb the organisms upstream of the sampling location. This procedure requires only one person and is the preferred macroinvertebrate collecting method for streams with flowing water (a second person is often used to keep time and record information on the field forms).

9.2.3.4.2 The following steps are used to collect the sample from riffles and runs:

1. Attach the four foot pole to the sampler. Care should be exercised to be sure the handle is on tight or the net might become twisted in strong current causing the loss of part of the sample.

2. Position the sampler quickly and securely on the stream bottom so as to eliminate gaps under the frame with the net opening upstream. Large rocks that prevent the sampler from seating properly should be avoided.

3. Hold the sampler in position on the substrate while checking for heavy organisms, such as mussels and snails in an area of about 0.5 m^2 in front of the net. These organisms will have to be hand picked and placed in the net.

4. Hold the sampler securely while kicking the substrate vigorously for 20 seconds (Use stopwatch) in an area of about 0.5 m^2 in front of the net.

5. After 20 seconds sampling at the station (transect), hold the net in place with the knees and pick up any loose rocks in the sampled area and rub off organisms in front of the net. Also any additional mussels and snails found should be placed in the net. Remove the net from the water with a quick upstream motion to wash the organisms to the bottom of the net.

6. Rinse net contents into the "riffle" bucket about half full of water (about one or two gallons) by inverting the net in the water.

7. Inspect the net for clinging organisms. With forceps remove any organisms found and place them in the bucket.

8. Large objects (rocks, sticks, leaves, etc.) in the bucket should be carefully inspected, and any organisms found should be washed into the bucket before discarding. Remove as much detritus as possible without losing any organisms.

9. 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 the riffle bucket.

10. Combine all the riffle samples in the same "riffle" bucket.

11. After all nine stations are sampled and the riffle samples combined in the "riffle" bucket, obtain a composite riffle sample as described in Subsection 9.2.3.6.

9.2.3.5 Procedures for Collecting Macroinvertebrates from Pools with the Modified Kick Net Sampler

9.2.3.5.1 A modified Kick Net Sampler with 595 µm mesh openings and a 0.5 m nylon bag attached to a four foot pole is used for collecting macroinvertebrates from pools. This sampler, described as a modified kick net, is available from Wildco (Catalog number 425-C50).

9.2.3.5.2 The following steps are used to collect the sample from pools and slow water areas:

1. Attach the four-foot pole to the Kick Net sampler. Care should be exercised to be sure the handle is on tight or the net might become twisted while dragging it through the water causing the loss of part of the sample.

2. If the pool is too deep (much over one meter) to safely sample at the designated spot, move downstream till a safe sampling spot is found.

3. Inspect the stream bottom for any heavy organisms, such as mussels and snails, in the 0.5 m^2 area to be sampled. These organisms will have to be hand picked and placed into the net or bucket.

4. While disturbing about 0.5 m^2 of the substrate by kicking vigorously with the feet, collect a 20 second sample by dragging the net repeatedly through the disturbed area just above the bottom while continuing to kick. Keep moving the net all the time so that the organisms trapped in the net will not escape.

5. After 20 seconds, hold the sampler between the legs with the net partially submerged and pick up any loose rocks in the sampled area and 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.

6. Rinse net contents into the "pool" bucket about half filled with water (about one or two gallons) by inverting the net in the water.

7. Inspect the net for clinging organisms. With forceps remove any organisms found and place them in the bucket.

8. Large objects in the bucket should be carefully inspected and any organisms found must be washed into the bucket before discarding the objects. Remove as much detritus as possible without losing any organisms.

9. If there is too little water to use the kick net, stir up the substrate with the gloved hands and use the U.S. 30 sieve to collect the organisms from the water in the same way the net is used in larger pools.

10. Combine all the pool samples in the same "pool" bucket.

11. After all nine stations are sampled and the pool samples are combined in the "pool" bucket, obtain a composite pool sample as described in Subsection 9.2.3.6.

9.2.3.6 Procedures for Obtaining a Composite Sample

9.2.3.6.1 Nine transects will be collected from all the sites. All the riffle samples from each site will be combined and all the pool samples will be combined in the field and a composite sample of each obtained as follows:

1. Save the entire combined sample as the composite sample for that habitat (riffle or pool) at that site and record on the sample collection form the number of samples that make up the composite sample.

2. Pour the entire contents of the bucket through a U.S. Standard 30 sieve. Remove any large objects and wash off any clinging organisms onto the sieve before discarding.

3. Using a squirt bottle, rinse all the organisms from the bucket onto the sieve. This is the composite sample for that habitat (riffle or pool) for the site.

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

5. Wash the contents of the sieve to one side by gently agitating the sieve in the water and wash the sample into a jar using as little water from the squirt bottle as possible (jar should not be over 1/4 full of water). If the jar is too full pour off some water through the sieve until the jar is not more than 1/4 full or use a second jar if a larger one is not available. Carefully examine the sieve for any remaining organisms and with forceps place them in the sample jar.

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

1. Stream Number
2. Type of sampler and mesh size used
3. Habitat type (riffle or pool)
4. Name of stream
5. Date of collection
6. Collectors initials
7. Number of transect samples composited

7. Replace the cap and with grease pencil put site number and sample type (Riffle or Pool) on the cap. If two jars are used be sure to mark them as such.

9.2.4 After Collection of benthic Invertebrate Sample(s)

9.2.4.1 Return samples and the sampling gear to the vehicle.

9.2.4.2 Assist the rest of the field crew with the fish collection.

9.2.5 Upon Returning to Vehicle

9.2.5.1 Each sample is assigned a unique prenumbered barcoded label to be used for tracking purposes. This unique barcode number must be placed on the jar and the label containing the barcode number must be filled in with a waterproof pen or pencil. This unique number must appear on all data sheets and must not be removed from the jar except as described in the Sample Sorting and Identification Protocol (See Benthic Invertebrate Laboratory Manual). If more than one jar is used for the sample this number must appear on each jar (use the extra labels and write the number on the label). DO NOT use two different Barcode Numbers on two jars containing one single sample. Either at the vehicle or at the motel, enter the barcoded numbers into the computer.

9.2.5.2 All samples are preserved 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 the U.S. Standard No. 30 sieve so as not to lose any 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. It is very important that sufficient ethanol be used to reach 70% concentration.

c. Transfer any organisms on the sieve back into the jar with forceps.

9.2.5.3 Check to be sure that the prenumbered stick-on barcoded label is on the jar and cover the entire label with clear, waterproof tape. Check to be sure the waterproof label is in the jar and properly filled in as described in the Protocol for Obtaining the Composite Sample (Subsection 9.2.3.7). Be sure the inside label and outside label describe the same sample. Replace the cap and seal it with electrical tape. Check to make sure the cap is properly marked with site number and habitat type (pool or riffle). Copy Barcoded Sample ID Number from the sample jar onto the Sample Collection Form-Streams and indicate which is the pool sample and which is the riffle sample.

9.2.5.4 Complete the check off sheet (Table 2) and enter any additional pertinent information in the field note book. Place the samples in a cooler or other secure container for transporting. Check to see that all equipment is in the vehicle.

9.2.6 After Each Week of Sampling

9.2.6.1 All samples must be transported or shipped to the designated depot site at the end of each week of sampling or at other agreed upon intervals. Tracking numbers of all samples transferred to the depot must be entered into the computer. A printout of the sample tracking numbers (barcodes) of the samples shipped must be included with the shipment and a copy will be mailed to the Indicator Lead along with the computer diskette each week.

9.2.6.2 When the shipment arrives at the depot the tracking numbers on the jars will be checked with the numbers on the printout. Any discrepancies will be cleared up by contacting the field crew responsible for the shipment as soon as possible. The samples will be picked up by Region III biologists and transferred to EMSL - Cincinnati at the end of the sampling period. All samples will be checked when they arrive at Cincinnati to be sure all samples listed on the shipping label are in the shipment. The samples will be shipped to the designated taxonomist(s) as soon as possible after they arrive at Cincinnati so that the analyses can be completed and the data analyzed within the required time frame. A copy of the computer printout listing all samples shipped will be included with the shipment and a copy will be sent to the Indicator Lead at EMSL-Cincinnati.

9.3 REFERENCES

Klemm, D.J., P.A. Lewis, F. Fulk, 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, OH 45268.

Plafkin, J.L., M.T. Barbour, K.D. Porter, S.K. Gross, 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, DC. 20460.

TABLE 1. EQUIPMENT LIST FOR MACROINVERTEBRATE SAMPLING CREWS

1. Modified Kick Net with 595 µm mesh openings and closed bag (Wildco # 425-C50)
2. Spare nets for the Kick Net Sampler or extra sampler
3. Sample jars, one quart, one per site, US Plastic # 66165
4. Sample jars, half gallon, one per site
5. Sample jars, one gallon, one per site
6. Maps of sampling sites
7. Labels, waterproof, 6 per site
8. First aid kit
9. Rubber gloves, heavy rubber, two pair
10. Rain gear for each person
11. Pencils, No. 2 soft lead, a dozen
12. Grease pencils, two
13. Knife, pocket, with at least two blades
14. Scissors, one pair
15. Forceps, two pair
16. Clear waterproof Tape
17. Electrical tape, four rolls
18. Labels, prenumbered barcoded, stick-on type, 6 per site
19. Compass
20. Wash bottle, one liter capacity
21. Sieve, U.S. Standard 30
22. Two Buckets, plastic, eight to ten quart capacity
23. Sieve bucket, 595 µm mesh openings (optional)
24. Kim wipes in small ziplock bag
25. Field check list sheets
26. Ethanol, one gallon per site, in proper container
27. Funnel, with large spout
28. Waders, hip length, pair for each person
29. Computer for sample tracking
30. Cooler for transporting ethanol and samples
31. Watch with timer or a stop watch
32. Pole attachment for Kick Net Sampler, four foot length
33. Camera and film
34. Small spatula
35. Field note book - pocket size
36. Dice for random numbers determination

TABLE 2. MACROINVERTEBRATE SAMPLING PROTOCOL CHECKLIST FOR BASE PROTOCOL

Date: _____ Time: _____ Site Number: _____

Name of Stream: _____ Location: _____

Crew ID (Circle): A B C D E F G; Collector: _____

___ 1. Weather and time recorded on Habitat Assessment Data Sheet.

___ 2. Initial observations in field notebook.

___ 3. Composite riffle sample collected with label inside.

___ 4. Composite pool sample collected with label inside.

___ 5. Correct barcode and label on all jars and sealed with clear, waterproof tape.

___ 6. All samples preserved.

___ 7. Caps sealed with tape.

___ 8. Samples logged in computer.

___ 9. Sample jars in cooler or otherwise secured.

___ 10. All equipment accounted for and secured in vehicle.

Signature: _____ Time: _____

APPENDIX

I. SAMPLE COLLECTION AND SHIPMENT FOR SEDIMENT TOXICITY SAMPLES

SAMPLE COLLECTION AND SHIPMENT FOR SEDIMENT TOXICITY SAMPLES

SEDIMENT COLLECTION AND SHIPMENT

1. Use the sediment left over from the benthic (sediment) metabolism indicator in Section 8, Benthic (Sediment) Metabolism: Field Methods.

2. Mix sediment well with a stainless steel or plastic mixing spoon.

3. Fill a 1 gallon ziplock bag with at least 1 L of sediment.

4. Close bag, making sure seal is secure.

5. Fill out ID label; place label and sediment inside another ziplock bag. Seal.

6. Place these bags inside a half gallon wide-mouth, screw top plastic jar. Place a label with stream ID on outside of the jar. Make sure cap seals tight.

7. Hold sediments samples on ice (do not freeze!) for laboratory analysis.

8. Ship samples to:

DynCorp, C/O U.S. EPA
3411 Church St.
Cincinnati, OH 45244
Phone (513) 569-7095
FAX (513) 569-7081

SELECTING FISH TISSUE SPECIMENS - 1994 EMAP-SW STREAMS PILOT

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 or secondary species sample that meets these criteria is available, use your best judgement in sending some type of fish sample (may be mixed species).

PROCESSING TISSUE SPECIMENS

- Keep hands, work surfaces, and wrapping materials clean and free of potential contaminants (mud, fuel, formalin, sun screen, insect repellant, etc.)
- 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 on the Fish Tissue Sample Tracking Form.
- Write the bar-code number(s)*** on the Fish Tissue Sample Tracking Form. Make sure that the form is filled out completely.
- 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 ziplock or garbage bag.
- 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.
- Complete bar-coded fish tissue label(s). Make sure the number(s) is/are the same one(s) on the Tracking 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.
- Place labeled ziplock or garbage bag(s) into second plastic bag(s) and seal and label second bag(s) (repeat previous two steps).
- Place double-bagged sample(s) in cooler with dry ice until shipment.
- Ship weekly on dry ice by Federal Express next day service. KEEP FROZEN UNTIL SHIPMENT. If ice is used, double bag ice in ziplock 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.

PROBLEMS?, QUESTIONS? - CALL: Roger Yeardley, (513) 569-7093 (# has voicemail also)

APPENDIX

H. SELECTING FISH TISSUE SPECIMENS

SELECTING FISH TISSUE SPECIMENS - 1994 EMAP-SW STREAMS PILOT

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 nontarget 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.

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).

FIELD PROTOCOLS FOR FISH COLLECTION

EXTERNAL ANOMALIES

Code	Categories	
N	NONE	No anomalies present.
D	Deformities	Skeletal anomalies of the head, spine, and body shape.
E	Eroded fins	Reductions of fin surface area or hemorrhage of fin rays.
L	Lesions	Open sores or exposed tissue.
T	Tumors	Irregular cell growth which are firm and cannot be easily broken.
F	Fungus	Filamentous or "fuzzy" growth on the fins or body.
B	Black spot disease	Parasite that appears as small black cysts on the fins and body.
I	"Ich"	Appears as white spots on the fins and body.
A	Anchor worm	Parasitic infection characterized by a worm embedded in the flesh of the fish.
W	Leeches	Worms which have anterior and posterior suckers. They may attach anywhere on the body.
X	Exopthalmia"	Popeye disease" is an anomaly seen as the bulging of the eye.
M	Missing	Eyes missing, blindness or deterioration of the lens should also be noted.
O	Other	Anomalies or parasites not specified.

FIELD PROTOCOLS FOR FISH COLLECTION

VOUCHERING FISH SPECIES AND UNIDENTIFIED SPECIMENS

Category 1. Large species, difficult to identify, OR uncommon in region. Preserve 1-2 adults plus 2-5 YOY and juveniles. Document with photograph.

American Eel	White Sucker	Shads
Sturgeon	Longnose Sucker	Bullhead catfish
Paddlefish	Hogsucker	Channel catfish
Gars	Quillback	Esocids
Bowfin	Carpsuckers	*Morone* spp.
Mooneye and Goldeye	*Moxostoma* spp.	Drum
Carp	Buffalo fishes	Salmonids
Walleye and Sauger	*Micropterus* spp.	Crappies

Category 2. Small fish OR difficult to identify species. Preserve up to 25 adults, juveniles and YOY per site.

Lampreys	Troutperch	Sculpins	Madtoms
Cyprinids	Chubsucker	Sunfish	Sticklebacks
Darters	Topminnows	Silversides	

a. Place subsample of 25 individuals of total catch into a kill jar containing 10% buffered formalin solution.

b. When specimens are dead, transfer to a nylon bag containing a waterproof label with site ID, barcode, date, crew ID and species name.

c. Repeat process for all species identified.

d. If fewer than 25 individuals are collected, preserve entire sample.

e. Place all ziplock bags in labelled voucher jar.

Category 3. State and federal species listed as protected. **Photograph and release.** Voucher if specimens have died.

NEW YORK
Mooneye
Round whitefish
Silver chub
Black redhorse (SC)
Longear sunfish
Eastern sand darter
Bluebreast darter
Longhead darter
Gilt darter

PENNSYLVANIA
Eastern sand darter

VIRGINIA (All SC)
Spotfin chub
Yellowfin madtom
Orangefin madtom
Roanoke logperch

WEST VIRGINIA (ALL SC)
Mountain brook lamprey
Shovelnose sturgeon
Paddlefish
Mooneye
Potomac sculpin
Slimy sculpin
Shield darter
Crystal darter
Grass pickerel
Redside dace
Satinfin shiner
Popeye shiner
New River shiner
Kanawha minnow
Cheat minnow
Longnose sucker
Mountain madtom

MARYLAND

Striped Bass
Maryland darter

APPENDIX

G. FIELD PROTOCOLS FOR FISH COLLECTION

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 uS 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, BD) 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.

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 1/2 square meter 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.

TABLE IV. PROCEDURE 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 prenumbered bar coded 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 prenumbered stick-on bar coded label is the on jar and agrees with the inside label. Cover the entire label with clear, waterproof tape.

5. Seal the caps with electrical tape.

6. Complete the check off sheet and place samples in cooler or other secure container for transport.

7. Secure all equipment in the vehicle.

TABLE VI. MACROINVERTEBRATE SAMPLING PROTOCOL CHECKLIST

Date: _____ Time: _____ Site No.: _____
Stream Name & Location: _____
Crew ID: 1 2 3 4 5 6 7 8; Collector: _____

__ 1. Habitat Assessment Sheet Completed with weather conditions described.

__ 2. Initial observations in field notebook.

__ 3. Composite riffle sample collected with waterproof label inside.

__ 4. Composite pool sample collected with waterproof label inside.

__ 5. Bar code and label on all jars covered with clear waterproof tape.

__ 6. All samples preserved.

__ 7. Caps sealed with tape.

__ 8. Photos of site.

__ 9. Samples logged in computer.

__ 10. Sample jars stored for travel.

__ 11. All equipment accounted for and secured in vehicle.

Time sampling completed _____

APPENDIX

F. BENTHIC INVERTEBRATES

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 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.

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 square meter 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.

SUMMARY FIELD PROTOCOLS FOR BENTHIC (SEDIMENT) METABOLISM

1.3 Ship samples to: Dr. James M. Lazorchak
US EPA-EMSL
3411 Church Street
Cincinnati, OH 45244
Phone 513/569-7076
Fax 513/569-7078

APPENDIX

E. SUMMARY FIELD PROTOCOLS FOR BENTHIC (SEDIMENT) METABOLISM

SUMMARY FIELD PROTOCOLS FOR BENTHIC (SEDIMENT) METABOLISM

1.1 Dissolved Oxygen Meter Calibration (for YSI model 58, with YSI model 5905 stirring BOD probe)

1. Each day, before leaving lab, motel, or base station: a) check meter's batteries to ensure that meter and stirring probe are operational and b) check probe membrane to ensure that it is not frayed or torn and that there are no bubbles under the membrane. If membrane is not intact or has entrapped bubbles, it should be replaced according to the manufacturer's directions.

2. Upon arriving at each site: a) zero meter according to manufacturer's directions and b) calibrate meter using the water-saturated atmosphere method described in the meter's operations manual.

1.2 Sediment Collection and Experimental Set-up

1. Collect sediment samples by gently scooping the top 4 cm of soft, surface sediments from depositional habitats (pools, eddys, backwaters) within the study reach. Samples are collected near Transects B-J, as defined by the Physical Habitat reach delineation. If soft sediments are abundant, collect samples at or near each transect. If soft sediments are scarce, collect them wherever you can within the reach. A total of 3 L of sediments are needed for metabolism and sediment toxicity analyses.

2. Fill ice chest 2/3 full with stream water and record temperature and dissolved oxygen (D.O.).

3. Thoroughly mix composited sediments.

4. Remove any visible organisms from the sediment before placing approximately 10 mL of sediment in each of 5 labelled, 50 mL screw-top centrifuge tubes. Labels are covered with clear tape.

5. Fill each tube to the top (no head space) with stream water from the ice chest and seal.

6. Fill 1 additional tube with stream water only and seal.

7. Incubate in closed ice chest for 2 hours.

8. Measure D.O. in each tube.

9. Decant overlying water and save sediment.

10. 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 remaining composite sediment sample by double-bagging in labelled, zip-lock freezer storage bags. Label is covered with clear plastic tape.

13. Store sediment toxicity sample chilled (but not frozen!) for laboratory analysis.

NOTE: All samples must be carefully labelled with the appropriate bar-code and adhesive tags. Tags need to be covered with clear tape.

SUMMARY FIELD PROTOCOLS FOR PERIPHYTON COLLECTION

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, and 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 labelled 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.

NOTE: ALL SAMPLES MUST BE CAREFULLY LABELLED WITH THE APPROPRIATE BAR-CODE AND ADHESIVE TAGS!

1.2 Ship Samples To:

Dr. James M. Lazorchak
U.S. EPA
3411 Church Street
Cincinnati, OH 45244
Phone: (513) 569-7076
Fax: (513) 569-7078

APPENDIX

D. SUMMARY FIELD PROTOCOLS FOR PERIPHYTON

SUMMARY FIELD PROTOCOLS FOR PERIPHYTON COLLECTION

1.1 Collection

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!**

8. Four subsamples will be taken from each composite sample. These are:

   a. <u>Identification/Enumeration</u>

      1) Withdraw 50 mL of mixed sample and place in a labelled sample vial (50 mL centrifuge tubes work well). Cover label with clear tape.

      2) Preserved sample with 2 mL of 10% formalin. Gloves should be worn.

      3) Tightly cap tube and tape with electrical tape.

   b. <u>Chlorophyll a</u>

      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).

FIELD SUMMARY: HABITAT CLASSIFICATION PICTURES

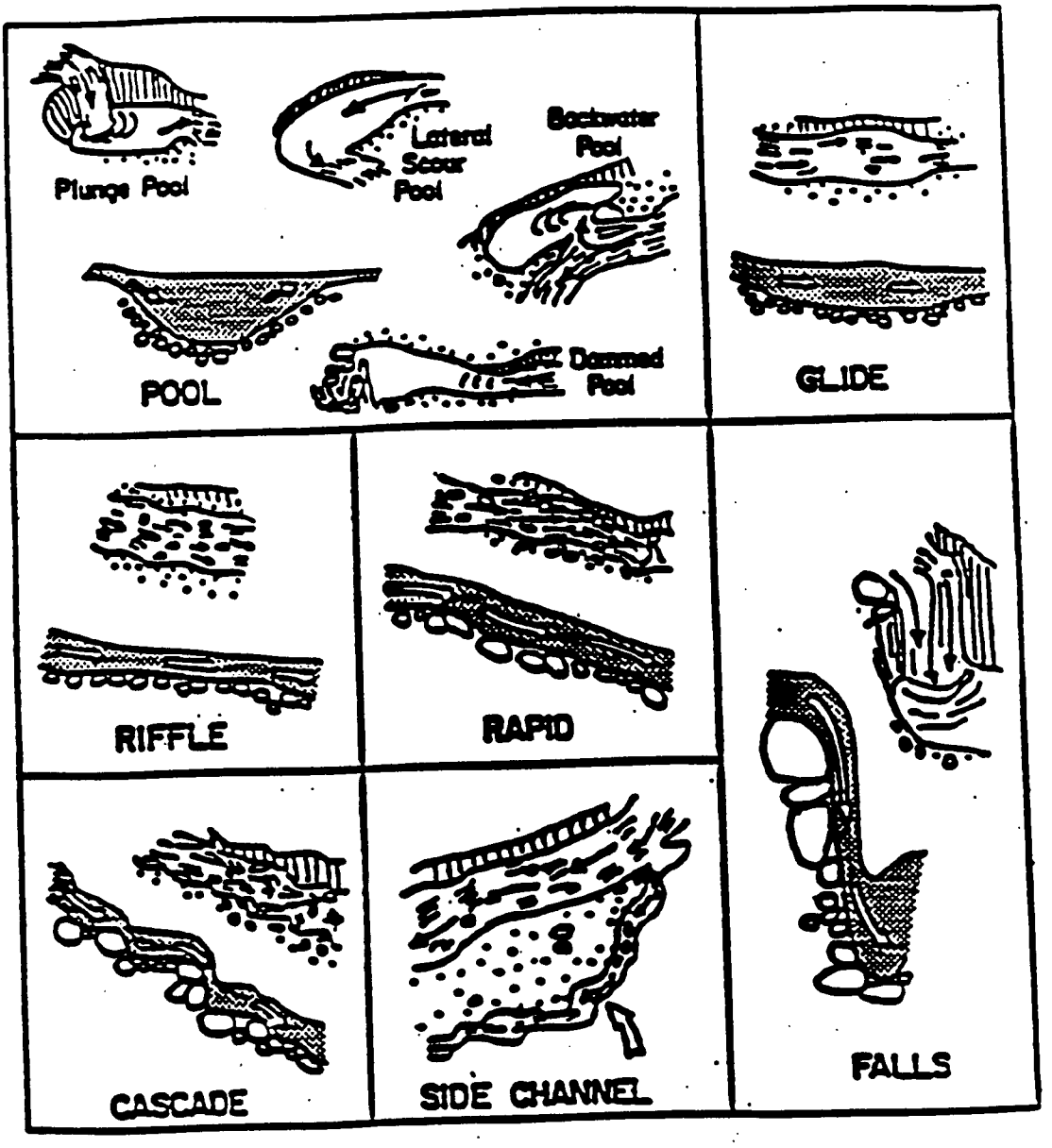

Figure 2. Sketches of channel units (From Frissell et.al.,1986)

FIELD SUMMARY: P-HAB PROBLEM AREAS

Bars: dry at baseflow, inundated at bankfull flow.

Measure wetted width across and over bars, but record bar width in the column provided on thalweg profile or 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 in the measures; 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% – Note side channel with "SC" in side channel column.

* If 15-35% – Note "SC" plus detailed channel & riparian Cross Sections on the side channel. Label additional new Cross Section Transect Forms as "XA", "XB", etc. corresponding to nearest main channel Transect location.

* If >35% – Note "SC" plus detailed channel & riparian profiles PLUS another thalweg profile on the side channel. As above, label additional new Cross Section Transect and Thalweg Profile Forms as "XA", "XB", etc. corresponding to nearest main channel Transect location.

When side channels are present, write "SC" in the space provided on the Thalweg Profile form. The thalweg form should reflect their presence with a continuous "SC" note until channels converge. If it is a long side channel, a continuing arrow can be used to mark it in the comments column to save time.

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 measures across the unvegetated portion of the channel.

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 — (tennisball to basketball)
GC	Gravel(Coarse)	16 to 64 mm — (marble to tennisball)
GF	Gravel (Fine)	2 to 16 mm — (ladybug to marble)
SA	Sand	.06 to 2 mm — (<ladybug size, but visible as particles - gritty between fingers).
FN	Fines	<.06 mm —Silt Clay Muck (not gritty between fingers)
WD	Wood	Regardless of Size
OT	Other	Metal, Tires, Car bodies etc. regardless of size (Put in comments if use other)

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):	0.1 to <0.3 m	(4 to 12 inches)
	0.3 to <0.6 m	(1 to 2 ft)
	0.6 to <0.8 m	(2 to 2.6 ft)
	>0.8 m	(>2.6 ft)
Length:	1.5 - <5 m	(5 - 16 ft)
	5 - 15 m	(16 - 49 ft)
	>15 m	(>49 ft)

FIELD SUMMARY: HABITAT CLASSIFICATION AT CHANNEL UNIT SCALE

Table 2. Habitat Classification at Channel Unit Scale (See Figure 2). Note that in order for a channel habitat unit to be distinguished, it must be at least as wide or long as the channel is wide.

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 bank.
Backwater Pool	PB	-	Pool separated from main flow off side of channel.
Dam Pool	PD	-	Pool formed by impoundment above dam or constriction.
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, depending upon discharge.
Dry Channel	DR		No water in channel

Code	Pool-Forming Element 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
O	Other – note in comments

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") and wetted width.
  - Classify habitat and pool-forming elements.
  - Determine presence of soft/fine 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. If the channel width is initially estimated greater than 4.0 m (and the reach length therefore exceeds 150 m), then omit the multiple width measures at the 100 thalweg intervals.

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 (densitometer).

* 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 human disturbances

Discharge:

In medium and large streams measure water depth and velocity (@ 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 filling of a bucket 10 times in succession.

FIELD SUMMARY: RIP. VEG., HUMAN DISTURB., IN-CHANNEL COVER:

* Observations upstream 5 meters and downstream 5 meters from each of the 11 cross-section stations.

* For Rip. Veg. and Human Disturbances, include the visible area from the stream back a distance of 10 m (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 (>5m high)
UNDERSTORY (0.5 to 5m high)
GROUND COVER layer (<0.5 high).

* Canopy and Understory Vegetation Types:

(Deciduous, Coniferous, Mixed, or None) 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 Scores 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%).

* Scoring 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.

APPENDIX

C. PHYSICAL HABITAT

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 meas' increments 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".

2. Work Flow:

* At downstream start point (Cross-Section "A"), one person makes channel dimension, substrate, bank, and canopy densitometer measurements. 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 Cross Sections A and B, making measures at each Thalweg distance interval. One person in channel measures width (when required), thalweg depth, and determines presence of fine/soft 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 flags, stop and take out a new cross-section form for Cross Section "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 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 station 11 ("K") at the upstream end.

* Discharge measurements made any time after choosing suitable location nearest to the EMAP "X" location. Discharge meas's are done by the Chemistry/Macroinvertebrate pair (rather than the Habitat/Fish pair) just after chemical samples are taken.

SUMMARY OF SITE PROCEDURE FOR WATER CHEMISTRY

I. COLLECT WATER SAMPLE

A. Make sure cubitainers and syringes are labelled and have the same barcode ID.

B. Rinse the 500 mL sample beaker three times with streamwater from mid-stream.

C. Rinse cubitainer three times with 25-50 mL of streamwater using sample beaker.

D. Fill cubitainer with streamwater using the 500 mL sample beaker. Cap the cubitainer and make sure that the seal is tight.

DO NOT EXPAND CUBITAINER BY BLOWING IN IT.

E. Rinse each of the four, 60 mL syringes three times with 10-20 mL of streamwater.

F. Fill each of the four 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 - 5 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 4-6 one gallon ziplock bags filled with ice.

II. IN SITU MEASUREMENTS

A. Conductivity

1. Measure and record the conductivity of the QCC solution.
2. Measure stream conductivity in sample beaker or in quiescent part of stream.

B. Dissolved Oxygen/Temperature

1. Calibrate DO meter following meter instructions.
2. Measure DO/Temperature in mid-stream.

APPENDIX

B. WATER CHEMISTRY

WATER CHEMISTRY SAMPLING CHECKLIST

I. EQUIPMENT TO CARRY IN FIELD FOR WATER CHEMISTRY

Rinse/Test 125 mL bottles of QCC solution in ziplock bag
Conductivity Pen
Field Forms
One, 250 mL plastic beakers
One, 500 mL plastic beaker in clean ziplock bag
One cubitainer in clean ziplock bag (barcode label attached)
Four, 60 mL syringes in plastic container (each one with bar code label attached)
Four syringe valves in plastic container
Opaque garbage bag

II. EXTRA EQUIPMENT TO CARRY IN VEHICLE

Cooler with 4-6 one gallon ziplock bags filled with ice
Back-up labels, forms, syringe valves

III. DAILY ACTIVITIES AFTER SAMPLING

1. Enter chemistry tracking data from today's sample (bar code ID, stream ID, date, etc.) into computer.

2. Check that cubitainer lid is on tight and has a flush seal.

3. Prepare sample for shipping (label and seal cooler, replace ice as close as possible to shipping time).

4. Call Overnight shipping company to arrange pick-up of cooler.

5. Rinse all sample beakers with deionized water, three times.

6. Make sure conductivity pens are rinsed with deionized water and are stored with moist electrodes.

7. Barcode label the next days sample containers (cubitainer and syringes), pack cubitainer and sample beakers in clean ziplock bag, and pack four syringes and syringe valves in plastic container.

APPENDIX

A. SUMMARY OF INITIAL SITE PROTOCOLS

SUMMARY OF INITIAL SITE PROTOCOLS

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:

NONTARGET	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 from map representation)
INACCESSIBLE	Physical Barriers (physically unable to reach the X-site) No Permission

Record class on Site Verification form, do not sample Nontarget or Inaccessible sites. Take samples from Target sites as discussed in field operations manual.

3. Measure the stream width at three "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. Layout 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.

6. Find the closest habitat break to the downstream reach endpoint, mark it as the reach start point (transect A). If no habitat break is present within 3-4 channel widths of the endpoint, mark the endpoint as Transect A and block net it.

7. Proceed upstream marking 10 more sample transects (sites B-K) at 1/10 intervals along the calculated reach length (every 4 channel widths or 1.5 meters in small streams). Assign each transect a Left, Center or Right sample site by throwing a die.

8. For the last transect (K), end the sample reach at the habitat break nearest the measured 40 channel width endpoint. If no habitat break is present for 3-4 channel widths, end the reach at the endpoint.

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 crews. Do not "slide" the reach to avoid bridges, stream side flow control structures, culverts and the like.

APPENDICES

SUMMARY OF FIELD PROTOCOLS FOR EMAP INDICATORS

A. SUMMARY OF INITIAL SITE PROTOCOLS

B. WATER CHEMISTRY

C. PHYSICAL HABITAT

D. SUMMARY FIELD PROTOCOLS FOR PERIPHYTON

E. SUMMARY FIELD PROTOCOLS FOR BENTHIC (SEDIMENT) METABOLISM

F. BENTHIC INVERTEBRATES

G. FIELD PROTOCOLS FOR FISH COLLECTION

H. SELECTING FISH TISSUE SPECIMENS

I. SAMPLE COLLECTION AND SHIPMENT FOR SEDIMENT TOXICITY SAMPLES

SECTION 10

FISH INDICATOR

10.1 EXPERIMENTAL DESIGN

10.1.1 The EMAP Surface Water Program will continue its stream pilot activities in the mid-Atlantic region during FY 94. Research will be conducted in conjunction with USEPA's TIME and Region 3 REMAP programs. One hundred ninety-nine Appalachian Ridge and Valley, Blue Ridge, Central Appalachian Plateau, Piedmont and Atlantic Coastal Plain sites will be selected for study. These sites will be sampled by teams of USEPA, USFWS, state and contract personnel in each of the operational units of the EMAP, REMAP, and TIME projects. The streams will be sampled during the 12 week index period. The objective is to collect a representative sample of the fish assemblage by methods designed to 1) collect all except very rare species in the assemblage and 2) provide a measure of the relative abundance of species in the assemblage.

10.1.2 Data will be summarized (means, standard deviations, etc.), analyzed for correlations among biotic and physico-chemical data (both parametric and distribution-free), and tested for site differences using analysis of variance. In addition, multivariate analyses of the data will be compared to metrics (e.g., diversity, evenness, relative abundance, IBI) to determine which approach yields the most information about the status of stream ecosystems. Cumulative distribution frequencies of community metrics will be evaluated to determine success of sampling method in obtaining a representative qualitative sample of the fish assemblage. Reference sites and historical databases will be used as a basis for this evaluation.

10.2 METHODS

10.2.1 Protocols for Fish Sampling

10.2.1.1 Site Selection. Sampling sites have been selected according to the base EMAP grid design. The candidate reference streams were identified by Region 3 and state biologists. Geographic Positioning System technology and USGS 7.5 minute base maps will be used to locate the latitude and longitude of each EMAP sampling reach (x-site). The x-site will be used as the center of the sampling reach.

10.2.1.2 Fish will be collected according to time and distance criteria. Collection time should continue for no less than 45 minutes and no greater than 3 hours for a distance of between 150 - 500 meters in order to obtain a representative sample over a distance equivalent to 40 channel widths. Homogeneous (or large systems) without clearly-defined habitat types should be sampled wherever best fish habitat is found. Sampling gear will consist of backpack electrofishing equipment supplemented by block netting and seining in habitats where flow, substrate and structure affect capture of benthic species. These protocols are designed for use in wadeable streams.

10.2.2 At the sampling site

a. Once x-site has been located, determine fish sampling reach as a function of mean channel width taken at the x-site (40 channel widths). The x-site will serve as the midpoint of the sample reach (except in cases indicated in Section 6, Physical Habitat Assessment).

b. Walk the length of the sample reach to determine pool depths, habitat composition, barriers and obstructions which may impede or aide in fish capture. Determine if reach requires block nets be placed at upstream and downstream ends of stream. (This will usually be required only in those places where sample reach is a large continuous pool.)

c. If the downstream or upstream limits of the sampling reach (pre-determined linear distance from x-site) fall in the middle of a habitat unit, extend the planned fishing reach to the bottom of the downstream habitat or the top of the upstream habitat. If no habitat break is evident within 20 m of either endpoint (e.g., a long pool or glide), block net at the endpoint. Record total length of stream reach sampled on data sheets, noting such exceptions to the protocol on the site description and fish field data forms.

d. Decision to use electrofishing equipment will depend on size of site, flow, conductivity and turbidity. If conductivity is below 10 µS or if flow is too high, site too deep or water is too turbid to assure safe footing or locate stunned fish, crew may consider use of seine only or determine that site is "Unsampleable". THIS IS A SAFETY DECISION.

e. In case of emergency, determine location of means of easy egress from stream.

10.2.3 Electrofishing

10.2.3.1 A generator-powered backpack electrofishing unit with a 45° tilt cutout switch will be used. **Always wear waders and heavy-duty insulated rubber gloves when working with electricity in water.** Wear polarized sunglasses to aid vision. All four crew members will participate in electrofishing.

a. Set unit to 300VA and pulsed DC. Based on stream conductivity, select initial voltage setting (150-400V for high conductivity >300µS; 500-800V for medium conductivity 100uS; 900-1100V for low conductivity <100µuS waters). Determine that all crewmembers are wearing waders and gloves and are clear of the anode. Start generator, set timer, and depress switch to begin fishing. Starting at the bottom of the most downstream riffle, pool, or other habitat type from the x-site and fish in an upstream direction, parallel to the current.

Adjust voltage and waveform output according to sampling effectiveness and incidental mortality to specimens. Backpack unit is equipped with an audio alarm when output voltage greater than 30V is present. 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 overload (in excess of 3 amps), the beep becomes very rapid and the LED overload indicator comes on. Release the anode switch and adjust voltage and waveform and continue fishing.

b. With switch depressed, slowly sweep electrodes from side to side in the water in riffles and pools. Sample available cut-bank and snag habitat as well as riffles and pools. Move 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 streams wider than can be spanned by sweeping the electrode from side to side, it may be necessary to work from the midline of the stream reach to the banks. At the bottom of the

reach, start by samplintg from the midline to the LEFT BANK (FACING DOWNSTREAM). At each subsequent transect, switch to the opposite side from the one which has just been fished. In stretches with deep pools, fish the margins of the pool as much as possible, being extremely careful not to step into deep water.

c. Netters follow along beside or slightly behind person operating shocker on the anode side and net stunned fish which are then deposited in separate buckets or holding tanks. (Water should be changed periodically.) Minnow seines (4m x 2m x 0.5cm) and kick nets (2m x 2m x 0.5cm) may be used to block in riffles, pools and snags.

d. Continue until 40 channel widths or a segment of at least 150 m has been sampled. Sampling should continue for at least 45 min. Distance sampled should not exceed 500 m; sampling time should not exceed 3 hours. **Record total time spent collecting and shocking time on data sheets.**

e. If fish show signs of stress (loss of righting response, gaping, gulping air, excessive mucus), stop at intervals and work up catch. Be sure to release fish downstream to reduce likelihood of resampling. This should only be necessary on very warm days, in long reaches or if very large numbers of fish are collected.

10.2.4. Seining

10.2.4.1. Seining will be used in conjunction with electrofishing to ensure sampling of those species which may otherwise be underrepresented by an electrofishing survey alone (eg. darters, sculpins). Seines will be used as block or kick nets to selectively isolate sections of the stream being electrofished (eg. snags, riffles, cutbanks). This method has proven effective in collecting sculpins, darters, dace and other benthic cyprinids. Seining may also be used in sites where stream is too deep for electrofishing to be conducted safely. In those sites, seining in riffles and pool margins will provide some indication of the assemblage of common species.

a. Riffle kicknet sampling consists of placing the net perpendicular to the current with the leadline on the bottom. The net (2m L x 1.25m W x .6cm mesh) is tilted slightly to form a pocket for trapping fish. Starting about 2m upstream, crewmembers disturb the substrate by shuffling their feet and overturning rocks. Seiners raise the net and it is carefully examined for fish.

b. Pool seining consists of pulling the net (3m x 2m x .6cm mesh) back and forth across the pool, using the shore and other natural habitat breaks as barriers. Another technique is to pull the net along in a downstream direction and sweeping toward the shore. Net is examined as above.

c. Block nets may be used in very large pools to limit fish escape or as seines. Large nets are typically deployed parallel to the current and swept to shore.

10.2.5. Sample Processing

10.2.5.1 Work as quickly as possible to minimize stress to live fish. All members of crew could work to separate fish into families or obvious "morphotypes" (two dorsal fins vs. one, sucker mouth, catfish,

trout). Once the rough sort has been completed, one person should identify, measure and examine the fish while another writes.

a. Sort fish by species into small buckets and containers. Taxonomic identification should be performed only by trained ichthyologists familiar with the fish species of the region.

b. To minimize handling, threatened and endangered species should be identified, counted and returned immediately to the stream. Consult the AFS Publication "Protected Fishes of the United States and Canada", J.E. Johnson, 1978. If conditions permit and stress to individuals will be minimal, photograph fish for voucher purposes. Indicate if photographed on data sheet. If protected fish have died, they should be vouchered in formalin. At the earliest possible time, the appropriate state officials should be notified.

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 on the second line of the identification column for each species and totalling the tally once processing is complete. Each fish must be examined individually but small species may be handled in small manageable groups to speed processing.

c. Sport fish and very large specimens (suckers, etc.) should be identified, measured to the nearest mm (standard length, total length, body depth, Fig. 10.1), examined for external anomalies (Table 10.3.), and released. Record all information on field data sheets. Separate a subsample of the target species for the fish tissue contaminants analysis. Voucher specimens (up to 25) of smaller individuals of each species should be kept (see Section 10.2.7 for voucher protocols). If only large individuals are collected, photograph each species and indicate if photographed on data sheet.

d. Identify other species. Measure the largest and smallest individuals (standard length and total length) to provide a size range for the species. Use the space beneath line for the common name on the data sheet to tally the number of individuals. Examine individuals in the sample for external anomalies and note proportion and type of anomalies observed keeping a running tally count in the column for comments and anomalies. Once all individuals have been processed, record total number under count and the species code beneath the common name. Separate a subsample of the target species for the fish tissue contaminants analysis. Voucher a subsample of 25 individuals from each species identified (see Section 10.2.7 for voucher protocols). If fewer than 25 individuals are collected of a species, keep all of the specimens. Count the number of unidentifiable specimens (if a large number, estimate number of individuals and note as comment) and preserve them in the jar labeled UNKNOWN. Record all information on field data sheets.

e. Preserve voucher and unidentifiable ("unknown") specimens in 10% formalin solution (37% formaldehyde:water). **Start with a concentrated solution of formaldehyde and dilute to final volume.**

f. Affix labels containing stream ID number, date, team ID and sample type to appropriate jars from that site. **Enter barcode ID in the space provided on the field data sheet.**

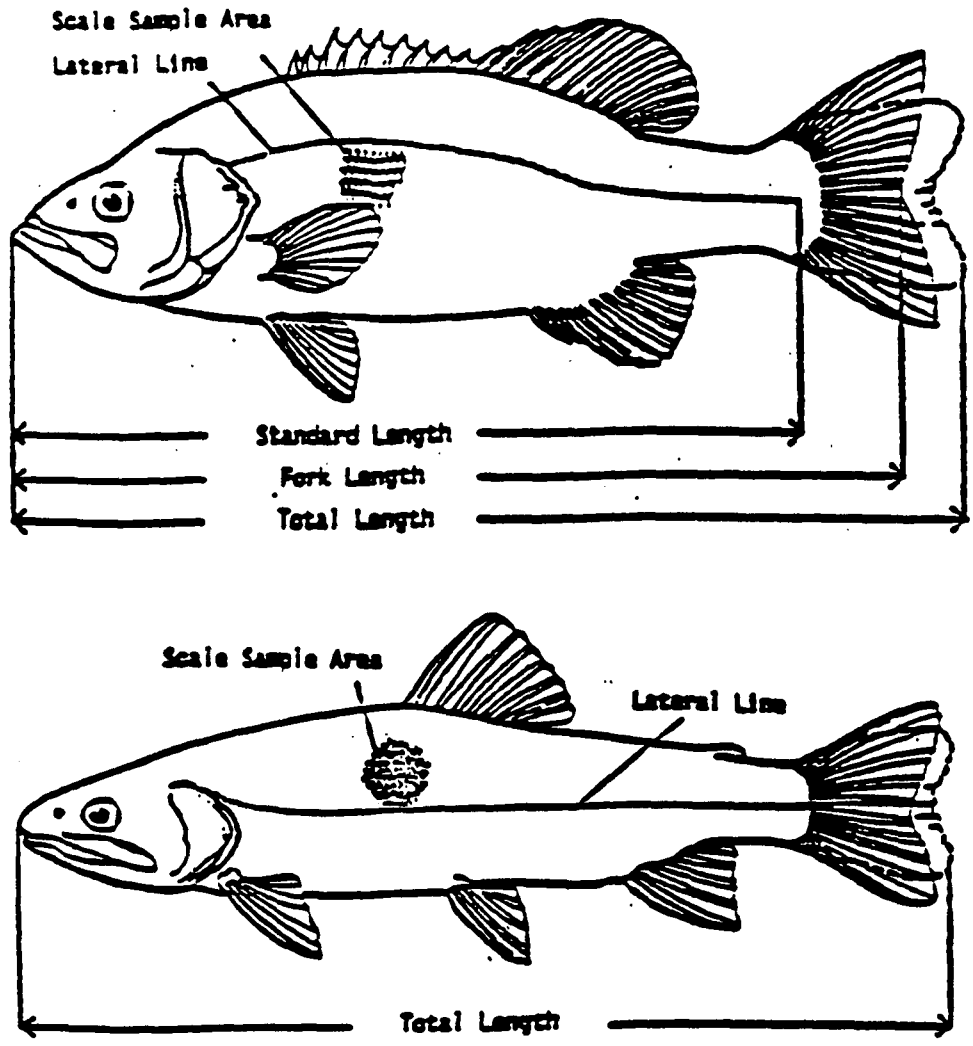

Figure 10.1. Fish measurement. With mouth closed and fish on right side, slide fish to touch "bump board". Read scale to nearest mm. (modified from Lagler, 1956).

10.2.6 External Anomalies

10.2.6.1 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.

Categories and Codes:

NONE (Code = N). No anomalies present.

Deformities (D) are skeletal anomalies of the head, spine, and body shape.

Eroded fins (E) appear as reductions of fin surface area or hemorrhage of fin rays.

Lesions (L) are open sores or exposed tissue.

Tumors (T) are areas of irregular cell growth which are firm and cannot be easily broken open when pinched. (Masses caused by parasites usually can.)

Fungus (F) may appear as filamentous or "fuzzy" growth on the fins or body.

Black spot disease (B) is caused by a parasite and appears as small black cysts on the fins and body.

"Ich" (I) is caused by a protozoan infestation and appears as white spots on the fins and body.

Anchor worm (A) is a parasitic infection characterized by a worm embedded in the flesh of the fish.

Leeches (W) are annelid worms which have anterior and posterior suckers. They may attach anywhere on the body.

Exopthalmia (X) or "popeye disease" is an anomaly seen as the bulging of the eye.

Missing (M) eyes, blindness or deterioration of the lens should also be noted.

Other (O) anomalies or parasites not specified.

Caution: Spinal cord injury, lesions, and exopthalmia or missing eyes may occasionally result from the sampling process, especially when electro-fishing. 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.

10.2.7 Voucher Protocol

10.2.7.1 With the exception of very large individuals of easily identified species, voucher collections of all species will be made to provide a permanent, archived, historical record of fish collections. Voucher material falls into three categories.

Category 1. Large easily identified species OR adults may be difficult to identify OR the species is uncommon in region. Preserve 1-2 small (<150 mm TL) adult individuals per site plus 2-5 YOY and juveniles. If only large adults are collected, reserve smallest individual until voucher procedure is complete and preserve ONLY if space is available. In specimens >100mm make a small slit on the lower abdomen on the RIGHT side. Document with photograph.

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

Category 2. Small to moderate-sized fish OR difficult to identify species. Preserve up to 25 adults, juveniles and YOY per site.

Lampreys	Troutperch	Sculpins	Madtoms
Cyprinids	Chubsucker	Sunfish	Sticklebacks
Darters	Topminnows	Silversides	Mudminnows

Category 3. Species of "special concern". These are state and federal species listed as protected. **Photograph and release.** If specimens have died, voucher them, note on datasheet and notify appropriate state official as soon as possible. Listed species are protected unless otherwise noted. SC = special concern. It is unlikely crews will encounter protected species in Maryland.

NEW YORK
Mooneye
Round whitefish
Silver chub
Black redhorse (SC)
Longear sunfish
Eastern sand darter
Bluebreast darter
Gilt darter
Longhead darter

PENNSYLVANIA
Eastern sand darter

VIRGINIA (All SC)
Spotfin chub
Yellowfin madtom
Orangefin madtom
Roanoke logperch

WEST VIRGINIA (All SC)
Mountain brook lamprey
Shovelnose sturgeon
Paddlefish
Mooneye
Grass pickerel
Redside dace
Popeye shiner
Satinfin shiner
New River shiner
Kanawha minnow
Cheat minnow
Longnose sucker
Mountain madtom
Crystal darter
Shield darter
Slimy sculpin
Potomac sculpin

MARYLAND
Maryland darter
Striped bass

a. Once all individuals of a species have been processed, place voucher subsample in kill jar containing a strong (approx. 20%) formalin solution. (Use the "Unidentified" jar for this.) Individuals >150mm SL should be slit on the lower abdomen of the RIGHT side.

b. When specimens are dead, transfer to a perforated ziplock bag containing waterproof label with site ID, barcode, species name and code. Place in "Voucher" jar in 10% formalin. BE SURE THAT JAR IS LABELED WITH VOUCHER LABEL with site ID, barcode and date.

c. Continue until all species are processed. Seal voucher jar with electricians' or clear tape. Check that jar is correctly labeled. Enter BARCODE ID in appropriate place on field data sheet.

d. Place all individuals of unidentified fish into UNKNOWN jar with label with site ID, barcode and date. Seal jar with tape. Enter BARCODE ID in appropriate place on field data sheet.

e. Transport to storage depot at end of week. Store in a cool, ventilated space.

10.3 SAFETY

10.3.1 Primary responsibility for safety while electrofishing rests with the fish team leader. All crew members should receive training in First Aid and CPR. Electro-fishing units have a high voltage output and may deliver dangerous electrical shock. While electrofishing, avoid contact with water unless sufficiently insulated against electrical shock. Use chest waders with non-slip soles and water-tight rubber (or electrician's) gloves that cover to the elbows. If they become wet inside, **stop fishing until thoroughly dry. Avoid contact with anode at all times. Cathode is also a potential shock hazard. At no time while electrofishing should a crewmember reach into the water for any reason.** If it is necessary for a crewmember to reach into the water to pick up a fish or something which has been dropped, **DO SO ONLY WHEN THE ELECTRICAL CURRENT HAS BEEN INTERRUPTED AND THE ANODE REMOVED FROM THE WATER. Do not resume electrofishing until crew is sure that all is clear.** The electrofishing equipment provided is equipped with a 45° tilt switch which interrupts the current. Do not make any modifications to the electrofishing unit which would make it impossible to turn off the electricity.

10.3.2 General safety guidelines should be observed. If waders or gloves develop leaks, leave the water immediately. Avoid operating electrofishing equipment near people, pets or livestock. Discontinue any activity in streams during thunderstorms or heavy rain. Rest if crew becomes fatigued.

10.3.3 Formaldehyde is an extremely caustic agent 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.

10.3.4 Ethanol has been shown to have deleterious effects on the central nervous system and may cause skin irritation. Care should be taken to avoid contact with vapors. Absolute ethanol is flammable and should be kept away from open flame or sparks. Store in sealed containers in safety cabinet or cooler lined with vermiculite.

10.3.5 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 containers and store in tightly closed container in safety cabinet or cooler lined with vermiculite.

10.4 QUALITY ASSURANCE/QUALITY CONTROL

10.4.1 The U.S. Environmental Protection Agency requires that all monitoring and measurement projects have written and approved quality assurance project plans. This QA/QC plan is presented in the

Environmental Monitoring and Assessment Program Integrated Quality Assurance Project Plan for the Surface Waters Resource Group (EPA600/X-91/080, Rev. 2.00).

10.4.1.1. Fish Sampling Quality Assurance Plan.

1. Sampling Design. Fish will be collected according to time and distance criteria. Results from the 1993 MAHA pilot project indicate a significant relationship between time sampled and distance sampled and number of individuals collected. A significant relationship also exists between time and distance sampled and number of species collected (this is subject to validation by museum). The length will be determined by a multiple of 40 channel widths indexed at the x-site. Stream segment length should be at least 150 m, but not more than 500 m. Collection time should be at least 45 min and not more than 3 hr per site. The length of channel surveyed will be used to define the stream segment for other sampling activities. The EMAP research indicator team will continue its evaluation of reach length criteria for larger wadeable streams.

2. Sampling and Analytical Methodologies. Fish collection protocols for electrofishing and seining are given in Subsection 1.2.3 - 1.2.4.

3. Quality Assurance Objectives

Precision--Precision of fish samples will be accomplished by careful application of methods and thorough sampling of all available fish habitats.

Taxonomic Accuracy--Taxonomic accuracy will be achieved by careful adherence to protocols, thorough sample preparation and analysis, and re-analysis of a subset of samples at a later date. Organisms will be identified to the lowest possible taxonomic level, using standard references and keys, and enumerated. Taxonomic identification will be performed only by trained ichthyologists familiar with the fish species of the region. Verification of taxonomic identification will be performed on voucher material by professional ichthyologists at the National Museum of Natural History, Smithsonian Institution, Washington, D.C. or other regional museums and academic institutions. Taxonomic nomenclature will follow Common and Scientific Names of Fishes from the United States and Canada, American Fisheries Society Special Publication 20.

Completeness--Valid fish data are required from 90% of the sites visited from which fish were collected.

Representativeness--Fish samples will be collected from all available habitats along the entire length of stream segment.

4. Quality Control Procedures

Field Operations--All personnel involved in fish sampling are trained in the selection of sample locations and in the use and maintenance of both seines and electrofishing gear. The minimum qualifications for fish team leader are a BS in fisheries with 3-5 yr experience in fishes in region. Preferred qualifications are MS in fisheries or zoology with specialization in fishes of region.

Laboratory Operations--Quality control of laboratory processing of fish samples will be achieved by proper selection of taxonomic techniques and references, and preparation, documentation, and archiving of reference specimens.

Information Management--Fish samples returned to the laboratory are logged in and their processing tracked until the samples are completed. All data sheets are inspected for completeness, accuracy, and legibility before proceeding to the next sample. Raw data sheets are retained in a file, and the data is entered into an ASCII file.

10.5 REFERENCES

Lagler, K.R. 1956. Freshwater fishery biology. 2nd. Edition. William C. Brown Co., Dubuque, IA.

TABLE 10.1. 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 uS 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, BD) 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.

TABLE 10.2. PROCEDURE FOR VOUCHERING FISH SPECIES AND UNIDENTIFIED SPECIMENS.

Category 1. Large species, difficult to identify, OR uncommon in region. Preserve 1-2 adults plus 2-5 YOY and juveniles. Document with photograph.

American Eel	White Sucker	Shads
Sturgeon	Longnose Sucker	Bullhead catfish
Paddlefish	Hogsucker	Channel catfish
Gars	Quillback	Esocids
Bowfin	Carpsuckers	*Morone* spp.
Mooneye and Goldeye	*Moxostoma* spp.	Drum
Carp	Buffalo fishes	Salmonids
Walleye and Sauger	*Micropterus* spp.	Crappies

Category 2. Small fish OR difficult to identify species. Preserve up to 25 adults, juveniles and YOY per site.

Lampreys	Troutperch	Sculpins	Madtoms
Cyprinids	Chubsucker	Sunfish	Sticklebacks
Darters	Topminnows	Silversides	Mudminnows

a. Place subsample of 25 individuals of total catch into a kill jar containing 10% buffered formalin solution.

b. When specimens are dead, transfer to perforated ziplock bag containing a waterproof label with site ID, barcode, date, crew ID and species name.

c. Repeat process for all species identified.

d. If fewer than 25 individuals are collected, preserve entire sample.

e. Place all ziplock bags in labelled voucher jar.

Category 3. State and federal species listed as protected. **Photograph and release.** Voucher if specimens have died.

NEW YORK	VIRGINIA (All SC)	Paddlefish
		Mooneye
Mooneye	Spotfin chub	Grass pickerel
Round whitefish	Yellowfin madtom	Redside dace
Silver chub	Orangefin madtom	Satinfin shiner
Black redhorse (SC)	Roanoke logperch	Popeye shiner
Longear sunfish	Mountain madtom	New River shiner
Eastern sand darter		Kanawha minnow
Bluebreast darter	WEST VIRGINIA (All SC)	Cheat minnow
Longhead darter		Longnose sucker
Gilt darter	Mountain brook lamprey	Crystal darter
Gilt darter	Shovelnose sturgeon	Shield darter

TABLE 10.2. PROCEDURE FOR VOUCHERING FISH SPECIES AND UNIDENTIFIED SPECIMENS (CONTINUED)

Category 3. State and federal species listed as protected. **Photograph and release.** Voucher if specimens have died. (Continued)

WEST VIRGINIA (ALL SC) (CONTINUED)	PENNSYLVANIA	MARYLAND
Slimy sculpin	Eastern sand darter	**Maryland darter**
Potomac sculpin		**Striped bass**

TABLE 10.3. EXTERNAL ANOMALIES

Categories and Codes:

NONE (Code = N). No anomalies present.

Deformities (D) are skeletal anomalies of the head, spine, and body shape.

Eroded fins (E) appear as reductions of fin surface area or hemorrhage of fin rays.

Lesions (L) are open sores or exposed tissue.

Tumors (T) are areas of irregular cell growth which are firm and cannot be easily broken open when pinched. (Masses caused by parasites usually can.)

Fungus (F) may appear as filamentous or "fuzzy" growth on the fins or body.

Black spot disease (B) is caused by a parasite and appears as small black cysts on the fins and body.

"Ich" (I) is caused by a protozoan infestation and appears as white spots on the fins and body.

Anchor worm (A) is a parasitic infection characterized by a worm embedded in the flesh of the fish.

Leeches (W) are annelid worms which have anterior and posterior suckers. They may attach anywhere on the body.

Exopthalmia (X) or "popeye disease" is an anomaly seen as the bulging of the eye.

Missing (M) eyes, blindness or deterioration of the lens should also be noted.

Other (O) anomalies or parasites not specified.

SECTION 11

FISH TISSUE CONTAMINANTS INDICATOR

11.1 INTRODUCTION

11.1.1 When coupled with a study design such as EMAP, the fish tissue contaminants indicator which measures bioaccumulation of toxic chemicals, 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 diagnostic indicators (physical habitat, water chemistry, land use, population density, other records of relevant anthropogenic stresses) and response indicators (fish, macroinvertebrates, periphyton) to diagnose whether the probable cause of stream degradation, when it is shown by the response indicators to occur, is water quality or physical habitat.

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

11.1.3 For the fish contaminants indicator in the 1994 pilot of EMAP-SW STREAMS, an attempt will be made to collect two fish samples at as many sites as possible. One sample, of **Primary Target Species**, will be of stream fish whose adults are small (dace, chub, sculpins, stonerollers, shiners, and darters). The second sample, where available, of a **Secondary Target Species**, will be of a species whose adults are of larger size (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 section (although it would be at a lower expected level of bioaccumulation) by compositing say, in the range of 20 to 200 small fish individuals than by compositing 3 to 5 large fish. Small fish may be a more appropriate indicator for assessing ecological risk, as they might be expected to be prey for a larger number of fish-eating animals (the majority of which will be piscivorous birds and small mammals). Small fish should afford savings in time and money, being easier to composite in the analytical lab, and offering potential savings in field processing. The major advantage that larger fish could potentially offer, whether predators (piscivores) or bottom feeders, is a higher level of bioaccumulation and thus greater sensitivity to detect contaminants. The relative bioaccumulation of contaminants by large and small stream fish is not known, thus the reason for having Primary and Secondary Target Species in this study.

11.1.4 Therefore one of the goals of the pilot studies, is to examine if small fish in the same stream section are bioaccumulating significantly relative to the larger fish. If they are, their other advantages would warrant targeting them for collection in further studies. Some group of these species with small adult individuals, perhaps representing a functional feeding group would be logical candidates for target species in a national streams survey. It is also possible to get relative bioaccumulation data from this kind of study, which may be of use in extrapolating from levels found in small fish to what might be found in a species with large adults (which are more appropriate for human risk assessment) if relationships between some prevalent species could be established.

11.1.5 In trying to answer these questions, the field crews' efforts to apply the protocol for sampling, handling and shipping in a **consistent manner are critical**. The diligence of the field crews in following the protocols is especially important in a status and trends study such as EMAP-SW where it is critical to get a **standard sample** from each site so that there is confidence that differences seen over time and between sites represents variations in the ecosystems and not differences in sampling and handling between the crews. Suggestions from field crew members on how the protocol can be improved are welcomed and will be incorporated to improve them, but protocols should be followed as written until official changes are made. If any questions arise for which the answers are not contained in the field guide, crews are encouraged to call the indicator lead, Roger Yeardley, at (513) 569-7093. If it is necessary to leave a message on the voicemail at this number, be sure and leave a phone number or fax number through which a reply can be made. If necessary, a reply to a crew's question can be incorporated into the voicemail greeting and the crew can call back to the above number that night or the next day and get the answer. If message is received and satisfactory, leave a second message acknowledging this so that the voicemail greeting can be changed.

11.2 SELECTING FISH TISSUE SPECIMENS

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

I. PRIMARY TARGET SPECIES

Small adult fish
(in priority order)

	WEIGHT
1) Blacknose Dace	50** - 400 g
2) Another Dace species	50** - 400 g
3) Creek Chub or Fallfish	50** - 400 g
4) Slimy Sculpin/Mottled Sculpin	50** - 400 g
5) Stoneroller	50** - 400 g
6) A Darter species	50** - 400 g
7) A Shiner species	50** - 400 g

A) Choose the **highest priority target species** from the above list, that has at least enough individuals to attain the minimum weight (50 g). Get as much weight of fish as possible within the desired weight range (50-400 g). Use scale provided to determine weight. With clean hands, place the fish on a fresh piece of aluminum foil (dull side towards fish) before placing in weighing container.

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

there is a conflict between criteria, getting a sufficient sample is a higher priority than getting similar-sized individuals

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

II. SECONDARY TARGET SPECIES

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

Larger adult fish (in priority order)	DESIRED SIZE	DESIRED NUMBER
1) 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 least at or near the minimum desired size (120 mm).

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

11.3 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 (to the nearest 5g) and count the total number of individuals. Make sure that the scale is tared and the teflon weighing beaker (to be used ONLY for weighing fish) has been rinsed with deionized or stream water. Measure the **total length** (TL) of <u>each</u> **secondary target species individual**. **Record** all of this information on the Fish Tissue Sample Tracking Form.

3. Write the **bar code number(s)*** ** on the Fish Tissue Sample Tracking Form. Record if the target species chosen was found throughout the measured EMAP stream reach (check-yes or no), with an explanation or some further details if the answer is no. Make sure that the form is filled out completely.

4. Wrap fish in aluminum foil. The primary target fish sample may be wrapped as a group. Secondary target fish should be wrapped individually. The dull side of the aluminum foil should be in contact with the fish. Once wrapped, place each sample in a ziplock or 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 as the one(s) on the Tracking Form. Apply it/them to the tape surface(s). Cover the label(s) with clear, waterproof tape. As labels may sometimes fall off, there should always be a label on the inner bag.

7. Place labeled ziplock or garbage bag(s) into second plastic bag(s) and seal and label second bag(s) (i.e., repeat steps 5 & 6).

8. Place double-bagged sample(s) in portable freezer until shipment. Keep samples frozen until shipment.

*** - 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.

11.4 SHIPPING TISSUE SPECIMENS

11.4.1 Crews collecting fish tissue samples will have a portable freezer for freezing and storage of samples until shipment. Samples should be shipped weekley. Crews may want to have them picked up at the motel in the morning, or ship in the evening and include that day's samples. Samples held by crews over the weekend must be kept frozen.

11.4.2 Ship samples on ice or dry ice. When using ice, double bag the ice and tape the last bag shut to prevent contamination of samples by melting ice. Place ice or dry ice in a cooler with a week's worth of samples (fish tissue, chlorophyll,periphyton,sediment). Place the week's field forms as well as the computer-generated shipping forms for these indicators in a ziplock bag and include with the shipment (tape to the underside of cooler lid). Tape coolers shut. Ship by Federal Express next day service. If shipping on dry ice, attach a filled-out DRY ICE warning label to the shipping container and enter the necessary information pertaining to dry ice in sections 3 & 4 on the FedEx Airbill, when filling out the FedEx Airbill.

Address shipment to:

U.S. EPA
Attn: EMAP
3411 Church St.
Cincinnati, OH 45244
(513) 569-7095

11.5 TARGET ANALYTES

11.5.1 For details of how the fish are processed by the analytical laboratory to determine tissue concentrations of EMAP-SW's target analytes, refer to the "1994 Pilot Stream Laboratory Methods Manual For Streams."

ANALYTE (CAS NUMBER)
Aldrin (309-00-2)
Aluminum (7429-90-5)
Arsenic (7440-38-2)
Cadmium (7440-43-9)
Chlordane-cis (5103-71-9)
Chlordane-trans (5103-74-2)
Chromium (7440-47-3)
Copper (7440-50-8)
2,4'-DDD (53-19-0)
4,4'-DDD (72-54-8)
2,4'-DDE (3424-82-6)
4,4'-DDE (72-55-9)
2,4'-DDT (789-02-6)
4,4'-DDT (50-29-3)
Dieldrin (60-57-1)
Endosulfan-I (959-98-8)
Endosulfan-II (33213-65-9)
Endrin (72-20-8)
Heptachlor (76-44-8)
Heptachlor Epoxide (1024-57-3)
Hexachlorobenzene (118-74-1)
Hexachlorocyclohexane [Gamma-BHC/Lindane] (58-89-9)
Iron (7439-89-6)
Lead (7439-92-1)
Mercury (7439-97-6)
Mirex (2385-85-5)
Nickel (7440-02-0)
trans-Nonachlor (3765-80-5)
cis-Nonachlor (5103-73-1)
Oxychlordane (27304-13-8)

PCB Congeners
2,4-Dichlorobiphenyl, #8 (34883-43-7)
2,2',5-Trichlorobiphenyl, #18 (37680-65-2)
2,4,4'-Trichlorobiphenyl, #28 (7012-37-5)
2,2',5,5'-Tetrachlorobiphenyl, #52 (35693-99-3)
2,2',3,5'-Tetrachlorobiphenyl, #44 (41464-39-5)
2,3',4,4'-Tetrachlorobiphenyl, #66 (32598-10-0)
2,2',4,5,5'-Pentachlorobiphenyl, #101 (37680-73-2)
2,3',4,4',5-Pentachlorobiphenyl, #118 (31508-00-6)
2,2',4,4',5,5'-Hexachlorobiphenyl, #153 (35065-27-1)
2,3,3',4,4'-Pentachlorobiphenyl, #105 (32598-14-4)

PCB Congeners (Continued)

2,2',3,4,4',5-Hexachlorobiphenyl, #138 (35065-28-2)
2,2',3,4',5,5',6-Heptachlorobiphenyl, #187 (52663-68-0)
2,2',3,3',4,4'-Hexachlorobiphenyl, #128 (38380-07-3)
2,2',3,4,4',5,5'-Heptachlorobiphenyl, #180 (35065-29-3)
2,2',3,3',4,4',5-Heptachlorobiphenyl, #170 (35065-30-6)
2,2',3,3',4,4',5,6-Octachlorobiphenyl, #195 (52663-78-2)
2,2',3,3',4,4',5,5',6-Nonachlorobiphenyl, #206 (40186-72-9)
Decachlorobiphenyl, #209 (2051-24-3)
3,3',4,4' Tetrachlorobiphenyl, #77 (32598-13-3)
3,3',4,4',5 Pentachlorobiphenyl, #126 (?)
3,3',4,4',5,5' Hexachlorobiphenyl, #169 (32774-16-6)

Selenium (7782-49-2)
Silver (7440-22-4)
Tin (7440-31-5)
Zinc (7440-66-6)

SECTION 12

SAMPLE COLLECTION AND SHIPMENT FOR SEDIMENT TOXICITY SAMPLES

12.1 SEDIMENT COLLECTION AND SHIPMENT

12.1.1 Use the sediment left over from the Sediment Metabolism Indicator, Section 8. Benthic (Sediment) Metabolism: Field Methods.

12.1.2 Mix sediment well with a stainless steel or plastic mixing spoon.

12.1.3 Fill a 1 gallon ziplock bag with at least 3 L of sediment.

12.1.4 Close bag, making sure seal is secure.

12.1.5 Fill out I.D. label; place label and sediment inside another ziplock bag. Seal.

12.1.6 Place these bags inside the third and final ziplock bag and seal.

12.1.7 Hold sediments samples on ice (do not freeze!) for laboratory analysis.

12.1.8 Ship samples to: Mark Smith/Lori Herrin/Ann Kneipp
TAI, C/O U.S. EPA
3411 Church St.
Cincinnati, OH 45244
Phone (513) 569-7091
FAX (513) 569-7081

12.2 SEDIMENT TESTING METHODS

12.2.1 For further information on the testing of these sediments, see the "1994 EMAP-SW Pilot Laboratory Methods Manual For Streams," Section 8, Sediment Toxicity Test Methods.

SECTION 13

SAFETY AND HEALTH

13.1 INTRODUCTION

13.1.1 Individuals working in the field and/or in the laboratory must become thoroughly familiar with all EMAP safety and health requirements and procedures. This Section is found in both the EMAP 1994, Pilot Field Operations Manual and the Laboratory Methods Manual. Specific safety and health requirements may be found in the various indicator sections of the above mentioned documents.

13.1.2 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. The Safety and Health Section must be available to all personnel and should be included in the field training.

13.1.3 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). Management should assign health and safety responsibilities and establish a program for training in safety, accident reporting, and medical and first aid treatment. The laboratory safety document and standard operating procedures (SOPs) containing necessary and specific safety precautions should be available to all persons involved in fish sample collecting and processing. Field and laboratory safety requirements for biomonitoring laboratories are found also in USEPA (1986) and Ohio EPA (1990).

13.1.4 A planned line of communication for safety and emergency situations is essential. Field personnel should have a daily check-in procedure for safety. An emergency communications plan should include contacts for police, ambulance, fire departments, search and rescue personnel. All field personnel need to be fully aware of all lines of communication, and the communication strategy should be included in all indicator sections of the field training and operations manual.

13.2 GENERAL PRECAUTIONS

13.2.1 Good housekeeping practice should be followed both in the field and in the laboratory. These practices should be aimed at protecting the staff from physical injury, preventing or reducing exposure to hazardous or toxic substances, avoiding interferences with laboratory operations, and producing valid data.

13.2.2 Field personnel and sampling crews must have mandatory training in Red Cross first aid, cardiopulmonary resuscitation (CPR), boating and water safety, field survey safety (weather conditions, personal safety, and vehicle safety), presurvey safety requirements (equipment design, equipment maintenance, reconnaissance of survey area), and electrofishing safety (American Red Cross, 1979; National Institute For Occupational Safety and Health, 1981; U.S. Coast Guard, 1987; Ohio EPA, 1990). It is the responsibility of the group safety officer or field sampling leader to ensure that the necessary safety courses are taken by all field personnel and that all safety policies and procedures are followed. Persons using sampling devices should become familiar with the hazards involved and establish appropriate safety practices prior to using them.

13.2.3 Operation of fish sampling devices involves potential hazards that must be addressed by the individuals using the equipment. Electrofishing equipment should be operated carefully, and all safety procedures must be followed. Electrofishing should always be done with at least three individuals. Persons using electrofishers should become familiar with the hazards involved and establish appropriate safety practices prior to using them (Reynolds, 1983; Ohio EPA, 1990). **Note:** Individuals involved in electrofishing must be trained by a person experienced in this method or by attending a certified electrofishing training course.

13.2.4 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.

13.2.5 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.

13.2.6 Personnel must consider and parepare 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 pamplet, "Federal Requirements for Redcreational Boats, " available from your local U.S. Coast Guard Director or Auxiliary or State Boating AOfficial (U.S. Coast Guard, 1987).

13.2.6 Prior to a sampling trip, personnel should determine that all necessary equipment is in safe working condition and that the operators are properly trained to use the equipment.

13.2.7 Safety equipment and first aid supplies must be available in the laboratory and in the field at all times. All boats with motors must have fire extinguishers, boat horns, cushions, and flares or communication devices.

13.2.8 Proper field clothing should be worn to prevent hypothermia, heat exhaustion, sunstroke, drowning, or other dangers.

13.2.9 The following safety elements should be included in the field training of field personnel:

1. Clothing and safety gear

2. Safety log for field personnel

    a. itinerary (vehicle used and its description, time of departure, travel route, estimated time of return)

    b. medical and personal information (allergies, personal health conditions)

    c. personal contacts (family, telephone numbers, etc.)

3. Emergency action
    a. first aid/CPR
    b. communication
    c. search and rescue
4. Emergency services required near base sites
5. Safety plan schedules

13.3 SAFETY EQUIPMENT AND FACILITIES

13.3.1 Necessary and appropriate safety apparel such as waders, lab coats, gloves, safety glasses, etc. must be available and used in accordance with the EMPA-SW for streams field operations and methods indicators and in the processing of all laboratory samples.

13.3.2 Bright colored caps (e.g., orange) must be available and worn in all field operations and sample collecting activities.

13.3.3 First aid kits, fire extinguishers, and blankets must be readily available in the field, and first aid kits, fire estinguishers, safety showers, and emergency spill kits must be available in the laboratory at all times.

13.3.4 A properly installed and operating hood must be provided in the laboratory for use when working with carcinogenic chemicals (e.g., formaldehyde, formalin) that may produce dangerous fumes (see Subsection 13.4.6).

13.3.5 Communication equipment and posted emergency numbers must be available to field personnel and those working in mobile labs in remote areas for use in case of an emergency.

13.3.6 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.

13.4 FIELD AND LABORATORY OPERATIONS

13.4.1 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.

13.4.2 Staff training in basic first aid and cardio-pulmonary resuscitation is strongly recommended.

13.4.3 All surface waters should be considered potential health hazards due to toxic substances or pathogens and exposure to them should be minimized as much as possible. Exposed body parts should be cleaned immediately after contact with these waters.

13.4.4 All electrical equipment must bear the approval of Underwriters Laboratories and must be properly grounded to protect against electric shock.

13.4.5 Heavy gloves should be used when hands are used to agitate the substrate during collection of kick net or square-foot type samples and when turning over rocks during hand picking.

13.4.6 During sample or specimen processing, persons must become familiar with the health hazards when using fixing and/or preserving chemical agents. Note: when using formaldehyde or formalin in the field, care must be taken because it is highly allergenic, toxic, and dangerous to human health (carcinogenic) if utilized improperly (see Subsection 13.3.3).

13.4.7 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 or other insect bites must take proper precautions and have any needed medications handy.

13.4.8 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 or State Safety requirements.

13.4.9 All field personnel should be familiar with the symptoms of hypothermia and know what to do in case symptoms occur. Hypothermia can kill a person at temperatures much above freezing (up to 10°C or 50°F) if he or she is exposed to wind or becomes wet.

13.5 DISEASE PREVENTION

13.5.1 Unknown pollutants and pathogens in surface waters and sediments should be considered potential health hazards and exposure to them kept to a minimum.

13.5.2 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. 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.

13.6 CHEMICAL WASTES

13.6.1 Chemical wastes can cause various hazards due to flammability, explosibility, toxicity, causticity, or chemical reactivity. All chemical wastes must be discarded according to standardized health and hazards procedures (National Institute for Occupational safety and Health, 1981; USEPA, 1986).

13.7 REFERENCES

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SECTION 14

INFORMATION MANAGEMENT PROCEDURES

14.1 INTRODUCTION

14.1.1 Data are collected from each site using field data recorders, sample collection and tracking, and receipt of analytical data from laboratories. The first two methods are used at the site and are essential to ensure that laboratory results will be available and accurately linked to each site. Without proper data and information collection methods, data can be lost and may result in revisiting the site.

Therefore, you are a major link in making sure the Mid-Appalachian Stream Pilot is complete. The Information Management (IM) personnel have made every effort to make all aspects of the sample shipment and tracking system easy and as painless as possible.

14.2 Field Data Entry Methods.

The primary method of data collection will be using Portable Data Recorders (PDR). This is a small hand held computer that has a full QWERTY keyboard, is air and water tight, will withstand being dropped into the water and has a viable battery life to be used for field data entry. The secondary method is paper backup (see figures 14-5 thru 14-15) for the PDR.

In both methods, each field has a representative significant figure shown and in most cases with have the proper unit of measure. If the measuring device used is in a different unit of measure and this is not stated somewhere, the results of the data may be unpredictable. These units and significant figures are important to follow. Just one digit longer or a different unit of measure may result in a value being represented incorrectly and changing the total analysis of the sampling. In all cases, please be concise in any entries and comment on anything notable in the comment sections provided.

14.2.1 PDR data entry

The field data recorders are a simple computing device. Each of the prior years field data entry forms have been converted from paper to electronic format and are available for field use.
The programs are organized in three different segments, general field data collection, fish collection and physical habitat. In all cases, the general field data collection program must be entered first to establish the sampling site. Even if the site was not sampled nor visited for any reason, enter the information on the PDR. Once this has been done, the rest of the programs will contain the information. Most of the fields can be changed except certain ones to protect the integrity of the connection within the data (like Stream ID, Date of collection, Visit Number).

The programs were written to be patterned off of the 1994 Streams Pilot field forms. Each section within the program(s) is as close as possible to the field data sheet that it represents. The same data is being collected and will be organized in the same manner as the prior year's data. A users guide is available that has examples of each screen and a full explanation as to the entry (see Information Management Handbook, PDR user instructions).

This type of data entry does not allow you full view of the data as the forms did. You need to look at each screen and make sure that you have at least entered that section and then check it again for completeness. You can change the data as many times as needed until the data is transferred from the PDR to the laptop. And even then, you do have one last opportunity to modify the data on the laptop. After the data leaves the laptop, you no longer can change it.

The data is transferred from the PDR to the laptop nightly. It is essential to the sample tracking (STARS) system. Table 14.2 has a brief description on the process. refer to the Information Management

Handbook for full instructions. After you have completed the sample tracking information, the data will need to be transferred from the laptop to the ERL-Corvallis VAX computer. This process is fully automated and can run on it's own. You first need to make sure that the connection is made from your computer to ERL-Corvallis. After that, the two (2) computers will take it from there.

If the data fails to transfer to ERL-Corvallis or you have no way to connect your computer to a phone, then please be sure to do a backup from the Main Menu on the laptop. This will save the data on something else other than the computer. Then, mail the diskette at the of the week as described in table 14.2

Recharge the PDR, the PDR batter and the laptop computer each night. This is essential to ensure that there will be enough power for the equipment to be used the next day. Please refer to the Information Management Handbook for full instruction. If at any time the PDR is not available, be it failure, power failure, or not available, use the field data sheets that are provided (see the next section, 14.2.2).

14.2.2 Paper data entry

The forms are printed on specially treated paper, Rite in the Rain, and are designed for the type of work that you will be doing. They have actually been dropped in the water and have retained all entries. A pen has been selected to ensure that the entries will not smear and/or rub off this specially treated paper. This pen (SANFORD Sharpie) will be issued and used by all team members. Also, a soft lead pencil (NO. 2 or less) will be provided. If the form gets wet, only the pencil will write on the form. **DO NOT USE ANY OTHER TYPE PEN OR PENCIL.**

Depending on the type of site (EMAP or TIME), the number and type of forms needed will vary. Section 14.5, table 14.1A-B illustrates the type and number of forms needed for the different sites. Check this table to see what set of forms you will need to have for your team and get a weeks supply plus a few days each week. The forms will be supplied at the beginning of field season. If more forms of any type are needed during the season, contact your field coordinator or IM and some will be mailed to you.

Now, on to the importance of the forms. Each form is essential to the project. They provide information on each indicator with either field measurements and/or sample collection and tracking. The entries must be legible and within the guidelines provided on each form. Illegible information is equivalent to NO information. If any additional information is deemed necessary for the indicator, provide this in the comment section on the specific form of indicator concentration. Review the forms at the end of the day at the site. This will allow you time to complete something you may have forgotten. The lead on each team will do a final check of the forms and enter their initials at the top of each page to indicate a check was made.

At the beginning of each week, prepare a FED-EX envelope to put each days set of forms in. Keep the envelope in your vehicles. Keep collecting the forms for each site until the end of the week. When you get ready to send the weeks samples that go to Newtown, again check the forms for completeness and place the envelope on the top of all the samples so that the ice does not saturate it. See section 14.3.3 on sample shipping for any further details.

Each Indicator section in the 1995 Pilot Operations and Methods Manual has information regarding completing each field data form. The specific entries and values for the forms will be covered in field training. Any other questions can be directed to IM and/or the indicator lead for clarification.

14.3 Field Sample Processing

The collection of samples is just as important and in some cases the most important part of the particular indicator the sample is associated to. Proper handling, preparation and documentation of any sample is integral to a successful stream visit. Taking time to properly fill out the labels, making sure that

the samples are collected as instructed by the protocols, and processing the samples for shipment by tracking them will ensure safe arrival to the laboratories for analysis. This section will assist you on the process to accomplish this.

14.3.1 Sample Labels and Preparation.

Each sample collected will have a label to identify it (see figure 14-16). Fill in any known information (e.g., stream name, stream id, date, etc.) on each label before departing from the vehicle so long as the stream has been determined samples will be collected. Pre-label and prepackage Cubitainers, syringes, and sample containers into sample kits prior to departure. Store an extra supply kit of sampling supplies (field data forms, sample labels, sample containers, etc.) in the vehicles. Inventory these extra supplies daily. Sample checklists are provided in tables 14-1A-1B

14.3.2 Sample Tracking and Reporting.

The sample tracking, shipping and reporting process will be completed back at the base site (hotel/motel/etc.). Follow the instructions for hotel/motel connection and data downloading (see the Information Management Book for further instructions).

Sample tracking is initiated at this time by using the notebook computer, bar code reader and the Information Management Handbook that will be provided to each crew. Table 14-2 is a brief version of this process. Any further instructions will be provided in the Information Management Handbook.

14.3.3 Sample Shipment

First and foremost, follow the indicator protocols on proper sample collection and storage. Then make sure all labels are completely filled out, legible and taped with clear plastic tape to prevent the label from smearing and/or getting wet. Make sure all the samples and labels are properly prepared. Just like the field data forms, labels are an important part of this project. If the samples are labelled incorrectly or illegibly, the site may have to be re-sampled and/or the data thrown out!

There are three (3) different sample shipping/destination locations that will be used. Table 14-3 lists the destinations for each sample type, the shipping times, and the preservatives required. The water chemistry is the only one that will be shipped on the day of collection. The others are either shipped or transported once a week. In the case of samples going to the supply depots, a designated crew member will be transporting the sample(s) there (see table 14-4 for depot locations).

14.3.3.1 Newtown shipments

Samples of fish tissue, periphyton chlorophyll, biomass, APA, ID, sediment tubes, and sediment toxicity bags remain with the crews until you return to EPA-Newtown. Keep the samples that need to be kept frozen (see table 14-3) in the provided freezer. If more samples are collected than the freezer can hold, ship samples in a cooler by FEDEX. Follow the cooler preparation as stated below, process the samples using STARS, complete the pre-printed FEDEX airbill and call FED-EX with the location of the cooler for pick up. Be sure to include the weeks worth of field data sheets.

Line each shipping cooler with some kind of paper (use the provided absorbent paper or paper towels, brown paper shopping bags or even pizza boxes work real well!). Place a garbage bag in each cooler. Prepare several bags of ice by writing on the bags the word "ICE" in large print. Then fill each bag with fresh ice and seal the bags. Place at least one or two bag(s) on the bottom of the cooler inside the garbage bag. Take the samples out of the freezer, Check all sample labels and make sure the information is correct, complete and legible. Place them on top of the ice, and then place more bagged ice on top. If the samples and the ice do not fit, get another cooler and prepare it the same way the first one was. Seal the garbage bag with either tieing it in a knot or using a twist tie (if available).

If there are field data sheets and you are making a mid-week FEDEX shipment, prepare the weeks worth of field data sheets by placing them in the FEDEX envelope and sealing it. Then place the envelope on the top of the sealed garbage bag. For all shipments, place the packing slip in a clear plastic bag and enclose it in the cooler before sealing. Seal the cooler with clear tape and place the completed FED-EX airbill in the attached plastic sleeve. Call FEDEX with the drop off location. Call Newtown (see table 14-3) to expect a Saturday shipment (for Friday shipments only).

14.3.3.2 University of Maine shipments

Water samples (cubi's/syringes) are shipped to University of Maine the day after collection. Each cooler will have a bag of sealed ice, the samples, packing slip and a label tapped to the inside of the cooler top with one of the re-supply depots' address for cooler return.

Line the cooler with a garbage bag. Prepare several bags of ice by writing on the bags the word "ICE" in large print. Place the sealed bags of ice in the garbage bag. Check all sample labels and make sure the information is correct, complete and legible. Place the water sample(s) on the ice, add more bags of ice on top of the samples, seal the garbage bag by knot, twist tie or tape, place the packing slip in a plastic bag on the garbage, and then attach the supply depot return card to the top of the cooler. Seal the cooler with clear tape and place the completed FEDEX airbill in the attached plastic sleeve. Call FEDEX with the drop off location. If the shipment date is on a Friday, call the contact person (see table 14-3) or leave a message that a Saturday delivery is coming.

14.3.3.3 Depot shipments

Biological specimens (fish and macroinvertebrae) will be taken to a depot (see table 14-4 for locations) by a designated crew member. Due to the length of time the samples are being stored, take a few extra moments to be sure the sample is properly prepared for storage. Reverify the fish voucher is preserved correctly, each perforated Ziploc bag contains a water proof label and the jar has the jar label taped on it. The macroinvertebrae should have the waterproof label in the jar and the bar coded label outside of the jar and taped. Be sure the packing slip of all samples taken to the specific depot. This packing slip is a tracking mechanism that will follow the sample(s) at all times.

14.4 Supplies

Depending on your team, resupplies can be acquired at your designated supply depot (see table 14-4). Coolers that were sent to the University of Maine will be sent back to one of the resupply depots. Each team will be supplied with address cards and will be instructed at field training as to how to prepare the coolers for return. This way you can get more coolers and other supplies at each location. Again be sure to collect one weeks supply plus some extras of each item to carry you over until the next week.

14.5 Forms, Labels and Checklists.

Figures 14.1 - 14.15 are the field data forms that will be used throughout the field season. Figure 14.16 illustrates the sample labels for sample collection. Table 14-1A-B are checklists for Information Management and stream visit needs for each site. The table itself declares which type of site it is to be used for. Table 14-2 is a brief description on how to run the STARS sample tracking and reporting program. Table 14-3 provides the shipping addresses and a brief synopsis of sample shipping. Table 14-4 is a list of each states' supply depot, contact and address.

TABLE 14-1A STREAM VISIT CHECKLIST - EMAP, REFERENCE AND REVISIT SITES

Stream Verification Form	1
Stream Assessment Form	1
Rapid Habitat Assessment Form: Glide/Pool	1
Rapid Habitat Assessment Form: Riffle/Run	1
Channel/Riparian Cross-Section Form	11
Fish Collection and Sample Tracking Form	1
Field Measurement Form	1
Sample Collection Form	1
Information Management Book	1
Stream Information Packet	1
Sampling Permit	1
EMAP Pamphlet	10
Field Operations and Method Manual	1
Composite Benthos Labels - Bar Coded	2
Benthos Identification Label Sheet - Non Bar Coded	1
Fish Voucher Label Sheet - Bar Coded	2
Fish Voucher Label Sheet - Non Bar Coded	1
Fish Tissue Labels - Bar Coded	2
Periphyton Labels (APA, BIOMASS, CHLA, ID) - Bar Coded	8
Sediment Metabolism Labels - Bar Coded	5
Sediment Toxicity Label - Bar Coded	1
Water Chemistry Labels - Bar Coded	5
Clear Tape Gun - Loaded	1
Clear Tape Pad	2
Cheat Sheets	1 set
Computer Equipment (see Information Management Book)	1 set
Clipboard/Sharpie/pencil	1 each
Shipping Airbills	2

TABLE 14-1B STREAM VISIT CHECKLIST - TIME SITES

Stream Verification Form	1
Stream Assessment Form	1
Rapid Habitat Assessment Form: Glide/Pool	1
Rapid Habitat Assessment Form: Riffle/Run	1
Fish Collection and Sample Tracking Form	1
Field Measurement Form	1
Sample Collection Form	1
Information Management Book	1
Stream Information Packet	1
Sampling Permit	1
EMAP Pamphlet	10
Field Operations and Methods Manual	1
Composite Benthos Labels - Bar Coded	2
Benthos Identification Label Sheet - Non Bar Coded	1
Periphyton Labels (APA, BIOMASS, CHLA, ID) - Bar Coded	2
Water Chemistry Labels - Bar Coded	5
Clear Tape Gun - Loaded	1
Clear Tape Pad	2
Cheat Sheets	1 set
Computer Equipment (see Information Management Book)	1 set
Clipboard/Sharpie/pencil	1 each
Shipping Airbill	1

TABLE 14-2 SAMPLE AND DATA TRACKING PROCEDURE

1. Upon return from the sampling site, connect the PDR and computer to the provided power strip, plug the power strip into a wall outlet and turn all equipment on. Connect the PDR to the computer. Select the PDR downloading process from the menus on the PDR and the computer (See the Information Handbook for detail instructions).

2. To begin entering sample tracking data, select the STARS menu option from the laptop menu system. The sample tracking screen will be displayed.

2. Using the bar code scanner, scan the bar code of the corresponding sample.

3a. (PDRs used) verify the information is correct and press F4 to save the record. Repeat the process for all samples.

3b. (No PDRs used) enter the STREAM ID, DATE, CREW ID, SAMPLE CLASS and location sample collected. Verify the information is correct and press F4 to save the record. Repeat the process for all samples.

4. After all data have been entered for the day, press ESC to return to STARS main menu. Select option 2 to generate shipping forms. A menu will appear. Select to either produce a new shipping form or reprint a previous. A list of the sample tracking numbers available for shipping will appear. Highlight the sample(s) to be shipped in a single cooler and press SPACEBAR to select it.

5. After all samples have been selected to be printed, press ESC to start printing. Scan in the FEDEX airbill number. Check printer to ensure that shipping and sample tracking form(s) are being printed. A shipping form should be printed for each cooler shipped. The program will ask if the printing was successful. If not, select reprint. The program will then ask, if there are more coolers to be shipped. Answer Y(es) to continue and follow steps 4-5 or enter N(o) to return to STARS main menu and exit the program.

6. Samples are ready to be shipped to the respective laboratories/locations. The shipping form(s) should be placed in a Ziploc bag(s) and taped to the inside of the cooler top. Follow the cooler sample packing process as described in section 14 of the fields method manual.

7. If this is one of the days to prepare a backup disk (Tuesday or Friday), place a diskette in drive A of the computer and select the Backup selection on the Main Menu of the laptop. Drop the sealed diskette packet into any US mail outlet on Friday of each week.

9. Last process is to select the option on the Main Menu to transfer data from the laptop to the Corvallis vax system. Follow the instructions provided in the Information Management Handbook. This is automatic once connection has been made.

10. Recharge all equipment by first turning the computer and PDR off. Remove the battery from the PDR, connect the PDR and the extra battery into the power strip using the provided AC adapters. Leave the computer plugged in along with the PDR and let them charge over night.

TABLE 14-3 SHIPPING DESTINATIONS, TIMES AND PRESERVATIVES

Media	Preservative	Shipping Times*	Destination
Composite Benthos (jar)	Ethanol Solution	Upon arrival to the depot center.	Depot supply center.[1]
Fish Vouchers (jar)	Formalin	Upon arrival to the depot center.	Depot supply center.[1]
Fish Tissue	Freezer/ Ice	Weekly to Newtown**	US EPA - Newtown[2]
Periphyton - APA	Freezer/ Ice	Weekly to Newtown**	US EPA - Newtown[2]
Periphyton - BIOMASS	Freezer/ Ice	Weekly to Newtown**	US EPA - Newtown[2]
Periphyton - CHLA	Freezer/ Ice	Weekly to Newtown**	US EPA - Newtown[2]
Periphyton - ID	Freezer/ Ice	Weekly to Newtown**	US EPA - Newtown[2]
Sediment Metabolism	Freezer/ Ice	Weekly to Newtown**	US EPA - Newtown[2]
Sediment Toxicity	Freezer/ Ice	Weekly to Newtown**	US EPA - Newtown[2]
Water Chemistry (Cubitainer, syringes)	Ice in Ziplocs	Day of collection	University of Maine[3]

* It may be necessary to ship items the following day if sampling activities pass the time for courier pickup of dropoff. Make sure samples requiring ice remain frozen and in darkness until shipment.
** there may be a mid-week shipment if the freezers become full. Otherwise, transport the samples to Newtown.

1. Depot supply center.

2. US EPA - Newtown
   3411 Church Street
   Cincinnati, OH 45244
   (513) 569-7075
   ATTN: EMAP - Jim Lazorchak

3. Sawyer Environmental Research Center
   2nd Floor
   University of Maine
   Orono, ME 04469
   (207) 581-3287
   ATTN: Therese Anderson

TABLE 14-4 SUPPLY DEPOTS

DEPOT	CONTACT	ACCESS	PHONE #	ADDRESS
CLEARFIELD	Wayne Wynick	M-F 8AM-5PM	814-765-3741	PA DER Bureau of Forestry Moshannon State Forest, RD1 Penfield, PA 15849
ELKINS	Bruce Evans	M-F 7am-5pm	304-637-0245	WV DEP Ward Road Elkins, WV 25241
St. ALBINS	Janice Smithson	Call Ahead	304-759-0701	WV Dep 694 Winfield Rd. St. Albans, WV 25177
WHEELING	Jim Green	Call Ahead	304-234-0243	US EPA - Wheeling Office 303 Methodist Bldg. Wheeling, WV 26003
WILKES-BARRE	Ed Kupsky	M-F 7am-5pm	717-826-2553	PA DER Water Management Program 2 Public Square Wilkes-Barre, PA 18711

Reviewed by (initial): _____

STREAM ASSESSMENT FORM

STREAM NAME: DATE OF VISIT: / / 94

STREAM ID: __ __ __ __ __ S TEAM ID (circle): 1 2 3 4 5 6 7 8

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

RESIDENTIAL	RECREATIONAL	AGRICULTURAL	INDUSTRIAL	STREAM MANAGEMENT
__ Residence	__ Public Campgrounds	__ Cropland	__ Industrial Plants	__ Logging
__ Maintained Lawns	__ Primitive Public Campsites	__ Pasture	__ Mines/Quarries	__ Surface Water Withdr.
__ Construction	__ Trash/Litter	__ Livestock	__ Oil/Gas Wells	__ Animal Passage
__ Public Docks	__ Surface Public Boaters, or Skiers	__ Orchards	__ Power Plants	__ Dredging
__ Dumping		__ Poultry	__ Logging	__ Channelization
__ Roads			__ Sewage or Pipe	__ Water Level Fluctuations
__ Bridge/Culverts			__ Docks	__ Fish Stocking
			__ Commercial	

QUALITATIVE ASSESSMENT OF ENVIRONMENTAL ATTRIBUTES

TROPHIC STATE: ☐ Highly Productive ☐ Moderately Productive ☐ Low Production

WATER CLARITY: ☐ CLEAR ☐ MURKY ☐ BOTTOM USUALLY NOT VISIBLE ☐ VISIBILITY < 10 CM

PERIPHYTON/MACROPHYTES:

VISUAL ASSESSMENT:

FISHABILITY: ☐ Excellent ☐ Good ☐ Fair ☐ Poor

CONDITIONS:

COMMENTS FROM LOCALS:

OBSERVATIONS:

OVERALL BIOTIC INTEGRITY ☐ Excellent ☐ Impacted ☐ Severely Impacted

GENERAL ASSESSMENT:

OBSERVATIONS:

WATERBODY CHARACTER (CIRCLE ONE)

OVERALL	5	4	3	2	1	MORE DEVELOPED
APPEARANCE	5	4	3	2	1	UNAPPEALING

COMMENTS:

Rev. 3/16/94 (stmass2.94)

STREAM ASSESSMENT FORM - 1

Figure 14.1. Stream Assessment Form - 1.

Reviewed by (initial): _____

STREAM VERIFICATION FORM

STREAM NAME: DATE OF VISIT: / / 94 VISIT #: 1 2 3

STREAM ID: _ _ _ _ _ _ S TEAM ID (circle): 1 2 3 4 5 6 7 8

STREAM VERIFICATION INFORMATION

PHOTO ROLL #: _____ FRAME #'S: _____ TO _____

STREAM VERIFIED BY (✓ all that apply): ☐ GPS ☐ LOCAL CONTACT ☐ SIGNS ☐ ROADS ☐ TOPO. MAP

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

COORDINATES	LATITUDE (dd mm.mm North)	LONGITUDE (ddd mm.mm) West	DOP FROM GPS		Are GPS Coordinates within 30 Sec. of map?
Map:	_ _ ° _ _ ' _ _ °	_ _ _ ° _ _ ' _ _ °	☐ 2D	DQ _____	☐ YES
Index Site:	_ _ ° _ _ ' _ _ °	_ _ _ ° _ _ ' _ _ °	☐ 3D	DQ _____	☐ NO

X-SITE STATUS - CHECK ONE BOX FROM ONE SECTION ONLY

SAMPLEABLE

- ☐ Regular - Flowing Water
- ☐ Intermittent - Dry Spots Along Reach
- ☐ Dry - No Water Anywhere along Reach
- ☐ Altered - Stream 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)
- ☐ NOT WADEABLE - > 1 M DEEP OVER HALF OF REACH

NO ACCESS

- ☐ Access Permission Denied
- ☐ Inaccessible (unable to reach site)

DIRECTIONS TO STREAM SITE

GENERAL COMMENTS

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

Rev. 3/16/94 (stmver1.94) Stream Verification Form- 1

Figure 14.2. Stream Verification Form - 1.

Reviewed by (initial): _____

STREAM VERIFICATION FORM (continued)

STREAM REACH DETERMINATION

CHANNEL WIDTH USED TO DEFINE REACH (M)	DISTANCE (M) FROM X SITE		COMMENT
	UPSTREAM LENGTH	DOWNSTREAM LENGTH	

Rev. 3/16/94 (sswver1.94)

Stream Verification Form- 2

Figure 14.2. Stream Verification Form - 2 (Continued).

Reviewed by (Initial): _____

FIELD MEASUREMENT FORM: STREAMS

STREAM NAME: DATE OF VISIT: / / 94

STREAM ID: _ _ _ _ _ _ S TEAM ID (circle): 1 2 3 4 5 6 7 8

WEATHER CONDITIONS (Circle)

CLOUD COVER	< 5%	5-25%	25-50%	50-75%	>75%
PRECIPITATION	NONE	LIGHT	MODERATE	HEAVY	
PREVIOUS PRECIPITATION (24 hr)	NONE	LIGHT	MODERATE	HEAVY	
AIR TEMPERATURE	_____ (°C)				

IN SITU MEASUREMENTS

STATION ID: ____

GPS Coordinates (XXXXX)			
Stream Conductance (XXXX)			
Stream DO mg/L (XX.X)			
Stream pH (XX.X)			

STREAM METABOLISM DETERMINATION

INITIAL DO (mg/L) (XX.X)	INITIAL INCUBATION TEMP (°C) (XX.X)	INCUBATION TIME (START TIME)	DURATION OF INCUBATION	FLAG	COMMENTS

TUBE	FINAL DO (mg/L) (XX.X)	FLAG	COMMENTS
R1			
R2			
R3			
R4			
R5			
Blank			

OXYGEN METER CALIBRATION INFORMATION

MEMBRANE CHECK ☐ INITIAL ELECTRODE ZERO ☐ RED LINE ☐

CALIBRATION CHAMBER TEMPERATURE (XX.X °C) SATURATED O2 @ AMBIENT (XX.X) mg/L

STATION ELEVATION (FROM TOPO MAP OR ALTIMETER) (XXXX) FT ELEVATION CORRECTION FACTOR (X.XXX)

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 (XX.X) mg/L

COMMENTS:

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

Rev. 3/16/94 (swfld1.94) Field Measurement Form- Streams - 1

Figure 14.3. Field Measurement Form-Streams - 1.

15.2.3.1 The goal of EMAP-SW is to assess three major ecological values with respect to streams: Trophic State, Fishability, and Biotic Integrity. To this end, any comments field crews can make in assessing these values, based on their field experience, would be most valuable. Comments on these values should be written in this section. The keywords on the left side of this section are there to stimulate thought and are not comprehensive, nor do they all have to be addressed. In this section, we are looking for an assessment of these values based on the field visit.

Trophic State - is defined as "rate or amount of algae & macrophytes produced or present in a stream". Listing any observed potential nutrient sources to the stream (e.g. septic tanks, agricultural runoff). Give your visual impression of trophic status as either low productivity (little/no biomass in streamwater), moderate productivity (intermediate amounts of biomass in streamwater), or high productivity (large amounts of biomass in streamwater).

Fishability - is defined as "a fish assemblage containing fish that are catchable, desirable, and safe to consume by wildlife and humans". Write down any observations about fishability; impressions of fish habitat, conversations with locals, presence of fish, fishermen.

Biotic Integrity - is defined as "the ability to support and maintain a balanced, integrated, adaptive community with a biological diversity, composition, and functional organization comparable to natural streams of the region". Record your overall impression of the "health" of the biota in the stream. The presence of higher order consumers (fish eating birds, mammals) is an indication of a healthy food web and could be noted here. Similarly, the lack of an organism that you expected to see would be an important observation. Also, any input on possible causes of impairment would be appropriate.

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". We're attempting (pilot) to define water body character through two attributes, degree of human development, and aesthetics. We'd like you to rate each of these attributes on a scale of 1 to 5. For development, give the stream a "5" if it is pristine, with no signs of any human development. A "1" would indicate a stream is totally developed, for example 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 (trash, algal growth, weed abundance, overcrowding). Circle the number that best describes your opinion on how suitable the stream water is for recreation and aesthetic enjoyment today:

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.

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.

15.1.4 Pool/Glide Prevalence Form

15.1.4.1 *Instream Cover for Fish* (see Subsection 15.1.3.1)

15.1.4.2 *Epifaunal Substrate for Macroinvertebrates*, in muddy bottom streams are mostly on submerged logs or snags, and aquatic vegetation (also see Subsection 15.1.3.2)

15.1.4.3 *Pool Substrate Characterization* 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.

15.1.4.4 *Pool Variability* 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 is it is greater than 1 m deep, and large if it length, width, or oblique dimension is greater than half the stream width.

15.1.4.5 *Channel Alteration* (see Subsection 15.1.3.5)

15.1.4.6 *Sediment Deposition* (see Subsection 15.1.3.6)

15.1.4.7 *Channel Sinuosity* 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.

15.1.4.8 *Channel Flow Status* determines the percent of the channel that 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. The water will not cover as much of the streambed, thus decreasing the amount of living areas for aquatic organisms. In muddy bottom streams, the decrease in water level will expose logs and snags, thus reducing the areas with good habitat.

15.1.4.9 *Condition of Banks* (see Subsection 15.1.3.9).

15.1.4.10 *Bank Vegetative Protection* (see Subsection 15.1.3.10).

15.1.4.11 *Grazing/Disruptive Pressure* (see Subsection 15.1.3.11).

15.1.4.12 *Riparian Zone Vegetative Width* (see Subsection 15.1.3.12).

15.2 STREAM ASSESSMENT FORM

15.2.1 The objective of the stream assessment form 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. This 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. It is meant to be completed at the end of the sampling, taking into account all observations the sampling crew has made while on site. The form consists of two major sections: Site Activities/Disturbances, and Environmental Value Assessment (see Section 14, Information Management for forms).

15.2.2 Site Activities/Disturbances

15.2.2.1 Record any of the following stressors that were observed either while on the stream or driving/walking through the stream catchment. For activities/stressors that you observe, rate them as to abundance or influence as Low, Medium, or High by putting an L, M, or H in the line next to the listed stressor. Leave the line blank for any stressor not observed. The distinction between low, medium, and high will be subjective. For example, if there are 2-3 houses on a stream, I would mark the Houses line with a "L" for low. If the stream is in a suburban housing development, I'd rate it as high (H). Similarly, a small patch of clear cut logging on a hill overlooking the stream, would rate a low ranking. Logging activity right on the stream shore, however, would get a high disturbance ranking. The form is broken down into the following Major Categories:

Residential:	Note the presence of any of the listed disturbances adjacent to or near the stream.
Recreational:	Record in this section 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:	Note the presence of cropland, pasture, orchards, poultry, and/or livestock.
Industrial:	Note 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:	Note 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.

15.2.3 Environmental Value Assessment

SECTION 15

RAPID HABITAT AND STREAM ASSESSMENT FORMS

15.1 RAPID PHYSICAL HABITAT ASSESSMENT FORM

15.1.1 The habitat assessment approach used in this protocol is adapted from EPA's Rapid B[illegible] assessment approach and refined from various applications across the country. The approach focuses on integrating information from specific parameters on the structure of the physical habitat. The field data sheets that summarize the major attributes of each parameter are shown in Section 14, Information Management. Specific instruction and training are necessary for an adequate assessment of habitat quality. For each of the parameters listed on the form, circle the point score that best describes the condition of the survey reach based on the described categories.

15.1.2 Different assessment forms are used for streams that are riffle/run prevalent versus those that are pool/glide prevalent. After making the initial survey of the 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 occupies the majority of the reach length. Then fill out the habitat assessment form corresponding to the chosen habitat type.

15.1.3. Riffle/Run Prevalence Form

15.1.3.1 *Instream Cover for Fish* includes the relative quantity and variety of natural structures in the stream, such as fallen trees, logs, and branches, large rocks, and undercut banks, that are available for refugia, feeding, or laying eggs. A wide variety of submerged structures in the stream provides the fish with a large number of niches, thus increasing the diversity.

15.1.3.2 *Epifaunal Substrate for Macroinvertebrates* are 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.

15.1.3.3 *Embeddedness* refers to the extent to which rocks (gravel, cobble, and boulders) are covered or sunken in to the silt, sand, or mud of the stream bottom. Generally, as rocks become embedded, the surface area available to macroinvertebrates and fish (shelter, spawning, and egg incubation) is decreased. 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.

15.1.3.4 *Varying Velocity and Water Depth* examine the availability of each of the four primary current/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 habitat types present. The presence or availability of these four habitats relates to the stream's ability to provide and maintain a stable aquatic environment. The general guidelines are 0.5 m depth to separate shallow from deep, and 0.3 m/sec to separate fast from slow.

15.1.3.5 *Channel Alteration* is 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.

15.1.3.6 The *Sediment Deposition* parameter measures 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 increase 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 an continually changing environment that becomes unsuitable for many organisms.

15.1.3.7 *Frequency of Riffles* is a way to measure 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.

15.1.3.8 *Channel Flow Status* is 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.

15.1.3.9 *Condition of Banks* measures whether the stream banks are eroded (or have the potential for erosion). Steep banks are more likely to collapse and suffer form 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.

15.1.3.10 *Bank Vegetative Protection* measures 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.

15.1.3.11 *Grazing/Disruptive Pressure* is a measure of 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.

15.1.3.12 The *Riparian Vegetated Zone Width* measures 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

Reviewed by (initial): ____

RAPID HABITAT ASSESSMENT FORM: RIFFLE/RUN-STREAMS (continued)

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 streambank surfaces are covered by vegetation.	70 to 90% of the streambank surfaces are covered by vegetation.	50 to 70% of the streambank surfaces are covered by vegetation.	Less than 50% of the streambank 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 streambank 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

Figure 14.9. Rapid Habitat Assessment Form: Riffle/Run-Streams - 2 (Continued).

FISH - JAR

STREAM ID: _ _ _ _ _ S

DATE: ____ / ____ / 93

SAMPLE TYPE: UNKNOWN VOUCHER

283098

FISH VOUCHER - BAG

STREAM ID: _ _ _ _ _ S

DATE: ____ / ____ / 93

FISH CODE: _____

TOTAL #: _____

283098

PERIPHYTON - APA

STREAM ID: _ _ _ _ _ S

DATE: __/__/94

HABITAT: POOL RIFFLE/RUN

SUBSAMPLE VOLUME: _____ mL

COMPOSITE VOLUME: _____ mL

100201

PERIPHYTON - BIOMASS

STREAM ID: _ _ _ _ _ S

DATE: __/__/94

HABITAT: POOL RIFFLE/RUN

SUBSAMPLE VOLUME: _____ mL

COMPOSITE VOLUME: _____ mL

100200

FISH TISSUE

STREAM ID: _ _ _ _ _ S

DATE: __/__/94

100200

COMPOSITE BENTHOS

STREAM ID: _ _ _ _ _ S

DATE: __/__/94

HABITAT: RIFFLE POOL

100200

PERIPHYTON - CHLA

STREAM ID: _ _ _ _ _ S

DATE: __/__/94

HABITAT: POOL RIFFLE/RUN

SUBSAMPLE VOLUME: _____ mL

COMPOSITE VOLUME: _____ mL

100201

WATER CHEMISTRY

STREAM ID: _ _ _ _ _ S

DATE: __/__/94

100200

COMPOSITE BENTHOS

STREAM ID: _ _ _ _ _ S

DATE: __/__/94

HABITAT: RIFFLE POOL

BARCODE No.: _____

PERIPHYTON - ID

STREAM ID: _ _ _ _ _ S

DATE: __/__/94

HABITAT: POOL RIFFLE/RUN

SUBSAMPLE VOLUME: _____ mL

COMPOSITE VOLUME: _____ mL

100201

SEDIMENT TOXICITY

STREAM ID: _ _ _ _ _ S

DATE: __/__/94

STATION: _____

100201

SEDIMENT METABOLISM

STREAM ID: _ _ _ _ _ S

DATE: __/__/94

SAMPLE TYPE: R1 R2 R3 R4 R5

100201

PERIPHYTON

BIOMASS / ID / CHLA / APA

STREAM ID: _ _ _ _ _ S

DATE: __/__/94

HABITAT: POOL RIFFLE/RUN

FILTER #: _____

ACTUAL VOLUME: _____ mL

TOTAL VOLUME: _____ mL

BARCODE No.: _____

Figure 14.10. Examples of Indicator Sample Labels.

Reviewed by (initial): _____

RAPID HABITAT ASSESSMENT FORM: GLIDE/POOL-STREAMS (continued)

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 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	Over 90% of the streambank surfaces is covered by vegetation.	70 to 89% of the streambank surfaces is covered by vegetation.	50 to 79% of the streambank surfaces is covered by vegetation.	Less than 50% of the streambank 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 streambank 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 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

Figure 14.8. Rapid Habitat Assessment Form: Glide/Pool-Streams - 2 (Continued).

Reviewed by (initial): ____

RAPID HABITAT ASSESSMENT FORM: RIFFLE/RUN PREVALENCE

STREAM NAME: DATE OF VISIT: / / 94

STREAM ID: _ _ _ _ _ _ S TEAM ID (circle): 1 2 3 4 5 6 7 8

TOTAL SCORE				
HABITAT PARAMETER	**CATEGORY**			
	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 boulders 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

Rev. 3/16/94 (version 1.94) RAPID HABITAT ASSESSMENT FORM: RIFFLE/RUN- STREAMS - 1

Figure 14.9. Rapid Habitat Assessment Form: Riffle/Run-Streams - 1.

Reviewed by (initial): _____

THALWEG PROFILE & WOODY DEBRIS FORM-STREAMS

STREAM NAME: DATE OF VISIT: / / 94

STREAM ID: _ _ _ _ _ _ S TEAM ID (circle): 1 2 3 4 5 6 7 8 Transect Station (A to K)

THALWEG PROFILE			Reach Length (m)			Increment (m)		
STA. #	THALWEG DEPTH (CM) (XXX)	WETTED WIDTH (M) (XX.X)	BAR WIDTH (XX.X)	SOFT/FINE SEDIMENT (Y or N)	CHANNEL UNIT CODE	POOL FORM CODE	SIDE CHANNEL (SC)	COMMENTS
0								
1								
2								
3								
4								
5								
6								
7								
8								
9								

LARGE WOODY DEBRIS (≥ 10 CM SMALL END DIAM. ≥ 1.5 M LENGTH) TALLY BETWEEN THIS TRANSECT AND NEXT

DIAMETER LARGE END	[illegible]			[illegible]		
	[illegible]	[illegible]	[illegible]	[illegible]	[illegible]	[illegible]
0.1 to <0.3 m						
0.3 - 0.6 m						
0.6 - 0.8 m						
> 0.8 m						

FLAG CODE: K = NO MEASUREMENT MADE; [illegible] = SUSPECT MEASUREMENT; F1, F2, ETC. = MISC. FLAGS ASSIGNED BY EACH FIELD CREW. EXPLAIN ALL FLAGS IN COMMENTS SECTION

CHANNEL UNIT CODES

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

POOL FORM CODES

S	Scour pool
W	Large Woody Debris
R	Backwater
B	Boulder or bedrock
P	Debrisform dam
O	Other (note in comments)

COMMENTS:

Rev. 3/15/94 (chnrip1.94) CHANNEL/RIPARIAN CROSS-SECTION & THALWEG PROFILE FORM - 2

Figure 14.7. Thalweg Profile and Woody Debris Form-Streams. Channel/Riparian Cross-Section and Thalweg Profile Form - 2.

Reviewed by (initial): _____

RAPID HABITAT ASSESSMENT FORM: GLIDE/POOL PREVELANCE

STREAM NAME: DATE OF VISIT: / / 94

STREAM ID: _ _ _ _ _ _ S TEAM ID (circle): 1 2 3 4 5 6 7 8

TOTAL SCORE

HABITAT PARAMETER	CATEGORY			
	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 or 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 concrete; heavily urbanized areas; instream habitat greatly altered or removed entirely.
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 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:	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

Rev. 3/16/94 (mpsdsp2.94)

RAPID HABITAT ASSESSMENT FORM: GLIDE/POOL- STREAMS - 1

Figure 14.8. Rapid Habitat Assessment Form: Glide/Pool-Streams - 1.

Reviewed by (initial):_____

VOUCHER SPECIMENS (continued)								
	SPECIES*	COUNT	TL* (mm)	SL* (mm)	BD* (mm)	Weight* (g)	FLAG	ANOMALIES/COMMENTS
13								
14								
15								
16								
17								
18								
19								
20								
21								
22								
23								
24								
25								
26								
27								
28								
29								

* Record species code on first line, then print common name on second line of each entry.
* Record minimum value on first line, and maximum value on second line of each entry.
Flag Codes: K= Sample not collected; U= Suspect sample. F1, F2, etc. = Misc. flags assigned by field crew. Explain all flags in Comments section.

Rev. 3/16/94 (stmfsh1.94)

Fish Collection & Sample Tracking Form - Streams - 1.

Figure 14.5. Fish Collection and Sample Tracking Form-Streams - 1 (Continued).

Reviewed by (initial): _____

CHANNEL/RIPARIAN CROSS-SECTION & THALWEG PROFILE FORM

STREAM NAME:

DATE OF VISIT: / / 94

STREAM ID: _ _ _ _ _ _ S

TEAM ID (circle): 1 2 3 4 5 6 7 8

Transect Station (A to K)

Slope (Backsite)	. %	Bkgd. Slope (Backsite)	_ _ . _ %	Bearing (Backsite)	_ _ _ °	Suppl. Bearing (Backsite)	_ _ _ °

I. Substrate Measurements Cross-Sectional Information

Loc.	Dist LB X.XX m	Depth XXX cm	Size Class Code	Embed. 0-100%	Flag
Lft					
LCtr					
Ctr					
RCtr					
Rgt					

Substrate Size Class Codes

RS = Bedrock (Smooth) *(Larger than a car)
RR = Bedrock (Rough) *(Larger than a car)
BL = Boulder (250 to 4000 mm) *(Basketball to car)
CB = Cobble (64 to 250 mm) *(Tennis ball to basketball)
GC = Coarse gravel (16 to 64 mm) *(Marble to tennis ball)
GF = Fine gravel (2 to 16 mm) *(Ladybug to marble)
SA = Sand (0.06 to 2 mm) *(Gritty - up to ladybug size)
FN = Silt/clay/muck (not gritty)
WD = Wood (any size)
OT = Other (comment)

II. Bank Measurements

Loc.	Bank Angle 0-360	Undercut Dist (m) X.XX	Flag
Lft	°		
Rgt	°		

Bankfull Width	_ _ _ . _ m	
Incised Ht.	_ _ _ . _ m	
Bankfull Ht.	_ _ _ . _ m	
Wetted Width	_ _ _ . _ m	
Bar Width	_ _ _ . _ m	

III. Fish Cover/Other

0 = Absent
1 = Sparse (0-10%)
2 = Moderate (10-40%)
3 = Heavy (40-70%)
4 = Very Heavy (> 70%)

	Channel	Flag
Floodplain Plant		
Macrophyte		
Woody Debris > 0.3 m (DBH)		
Small/Woody Debris < 0.3 m (SMALL)		
Overhanging Veg. < 1 m of Surface		
Undercut Banks		
Boulders		
Artificial (Structures)		

IV. Canopy Cover Measurements

Densiometer (0 to 17 max)

		Flag			Flag
Cen/Up			Cen/		
Cen/L			Lft		
Cen/Dwn			Rgt		

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

V. Visual Riparian Estimates

	Left Bank	Right Bank	Flag
Riparian Vegetation Cover (0 = Absent; 1 = Sparse (0-10%); 2 = Moderate (10-40%); 3 = Heavy (40-70%); 4 = Very Heavy (> 70%))			
Canopy (> 5 m high)			
Vegetation Type (D, C, M, or N)			
Big Trees Trees > 0.3 m DBH			
Small Trees Trees < 0.3 m DBH			
Understory (0.5 to 5 m high)			
Vegetation Type (D, C, M, or N)			
Woody Shrubs & Saplings			
Non-woody Herbs, Grasses, & Ferns			
Ground Cover (< 0.5 m high)			
Woody Shrubs & Saplings			
Non-woody Herbs, Grasses, & Ferns			
Barren, Bare soil or duff			
Human Influence (0 = Not present, B = On bank, C = Within 10 m, P = > 10 m)			
Walls/Dikes/Revetments/Riprap			
Buildings			
Pavement			
Roads/Railroad			
Pipes (Sewer/Outlets)			
Landfill/Trash			
Park/Lawn			
Row Crops			
Pasture/Range/Hay field			
Logging Operations			
Mining Activity			

Rev. 3/15/94 (chnlrip1.94) CHANNEL/RIPARIAN CROSS-SECTION & THALWEG PROFILE FORM - 1

Figure 14.6. Channel/Riparian Cross-Section and Thalweg Profile Form - 1.

Reviewed by (initial): _____

SAMPLE COLLECTION FORM- STREAMS (continued)

SEDIMENT METABOLISM SAMPLES (50 mL Tubes)

SAMPLE ID # (Barcode)	SAMPLE TYPE	FLAG	COMMENTS
	R1		
	R2		
	R3		
	R4		
	R5		
	R_		

SEDIMENT TOXICITY SAMPLES

SAMPLE ID # (BARCODE)	FLAG	COMMENTS

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

Sample ID (Barcode)

LINE #	SPECIES CODE	COMMON NAME	TOTAL WEIGHT (g)	NUMBER OF INDIVIDUALS	FLAG
L1					
L2					

Is composite sample composed of individuals collected from throughout reach? Yes No

If No, Explain: _____

FISH TISSUE SAMPLES - SECONDARY SAMPLE (where available)

Sample ID (Barcode)

LINE #	SPECIES CODE	COMMON NAME	TOTAL LENGTH (mm)	FLAG
L1				
L2				
L3				
L4				
L5				

Is composite sample composed of individuals collected from throughout reach? Yes No

If No, Explain: _____

LINE #	COMMENT OR FLAG EXPLANATION FOR FISH TISSUE

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

Figure 14.4. Sample Collection Form-Streams - 2 (Continued).

Reviewed by (initial): _____

FISH COLLECTION AND SAMPLE TRACKING FORM- STREAMS

STREAM NAME: DATE COLLECTED: / / 94

STREAM ID: __ __ __ __ __ S TEAM ID (circle): 1 2 3 4 5 6 7 8

SHOCK TIME (0000) _____ SECONDS* FISHING TIMEDUP (0000) _____ MINUTES!* ID BY (last name):

SAMPLE ID # (BAR CODE)	No. OF SPECIMENS	SAMPLE TYPE (L or V)	FLAG	COMMENTS

VOUCHER SPECIMENS

	SPECIES	COUNT	TL* (mm) 00.00000	SL1 (mm) 00.00000	SL2 (mm) 00.00000	Weight (g) (max 4)	FLAG	ANOMALIES/COMMENTS

* Record species code on first line, then print common name on second line of each entry.
* Record minimum value on first line, and maximum value on second line of each entry.

☐ NOT FISHED ☐ NONE COLLECTED

Flag Codes: K = Sample not collected: U = Suspect sample. F1, F2, etc. = Misc. flags assigned by field crew. Explain all flags in Comments section.

Rev. 3/16/94 (strwfsh1.94) Fish Collection & Sample Tracking Form - Streams - 1.

Figure 14.5. Fish Collection and Sample Tracking Form-Streams - 1.

Reviewed by (initial): _____

FIELD MEASUREMENT FORM STREAMS (continued)

STREAM DISCHARGE

☐ Velocity Area	Machine ID #				☐ Timed Filling				
	DIST FROM BANK (cm) XXX	VELOCITY (ft/s) XX.X	DEPTH (FEET) XX.X	FLAG	PATHE	TREP	VOLUME	TIME (s)	FLAG
1									
2									
3									
4									
5									
6									
7									
8									
9									
10									
11									
12									
13									
14									
15									
16									
17									
18									
19									
20									

☐ Portable Weir	☐ Neutral Buoyant Object	
	Float Distance (m) XXX	Total Time (sec) XXX
V. Notch (60 deg)	1. _____	1. _____
Water Ht. (_____ (cm) XXX)	2. _____	2. _____
	3. _____	3. _____

Flag	Comments

Flag Codes: E = 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.

Field Measurement Form- Streams - 2

Rev. 3/16/94 (strwfld1.94)

Figure 14.3. Field Measurement Form-Streams - 2 (Continued).

Reviewed by (initial): _____

SAMPLE COLLECTION FORM-STREAMS

STREAM NAME: DATE OF VISIT: / / 94

STREAM ID: _ _ _ _ _ _ S TEAM ID (circle): 1 2 3 4 5 6 7 8

WATER CHEMISTRY (4-L Cubitainers and 4 syringes)

SAMPLE ID # (Barcode)	SAMPLE TYPE	STATION COLLECTED	FLAG	COMMENTS
	R1			

COMPOSITE BENTHOS SAMPLES

SAMPLE ID #	HABITAT (R = RIFFLE, P = POOL)	NUMBER OF JARS	FLAG	COMMENTS

STATION												TOTAL
RIFFLE												
POOL												
CIRCLE ONE		LCR	LCR	LCR	LCR	LCR	LCR	LCR	LCR	LCR		

COMPOSITE PERIPHYTON SAMPLES		HABITAT TYPE (CIRCLE)	Riffle	Pool	Other
SAMPLE ID # (Barcode):		COMPOSITE VOLUME			mL

ASSEMBLAGE ID (150-mL)	CHLOROPHYLL (10-mL)	ASH FREE DRY MASS (1/2 filter)	APA SAMPLE (50-mL)
SUB-SAMPLE VOL	VOL FILTERED	FILTER	VOL FILTERED	SUB SAMPLE VOL
mL	mL		mL	

COMPOSITE PERIPHYTON SAMPLES		HABITAT TYPE (CIRCLE)	Riffle	Pool	Other
SAMPLE ID # (Barcode):		COMPOSITE VOLUME			mL

ASSEMBLAGE ID (150-mL)	CHLOROPHYLL (10-mL)	ASH FREE DRY MASS (1/2 filter)	APA SAMPLE (50-mL)
SUB-SAMPLE VOL	VOL FILTERED	FILTER	VOL FILTERED	SUB SAMPLE VOL
mL	mL		mL	

COMMENTS:

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

Rev. 3/16/94 (streams1.94) Sample Collection Form- Streams - 1

Figure 14.4. Sample Collection Form-Streams - 1.