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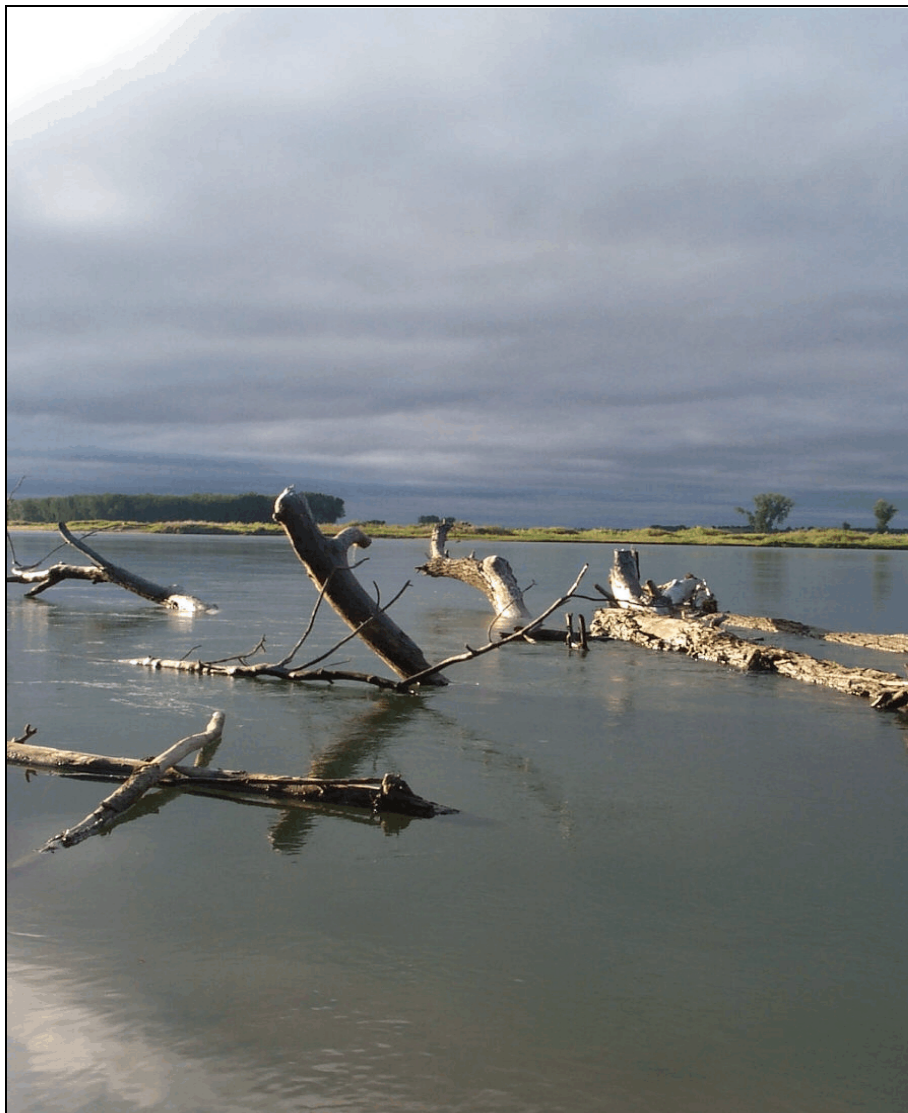
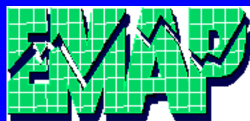
Great River Ecosystems Field Operations Manual





Environmental Monitoring and
Assessment program

EPA/620/R-06/002
February 2006

ENVIRONMENTAL MONITORING AND ASSESSMENT PROGRAM GREAT RIVER ECOSYSTEMS (EMAP-GRE)

FIELD OPERATIONS MANUAL

Ted R. Angradi[1] (editor), E. William Schweiger[5], Brian H. Hill[1], David W. Bolgrien[1], James M. Lazorchak[2], Erich B. Emery[3], Terri M. Jicha[1], Jeff A. Thomas[3], Donald J. Klemm[2], Spence A. Peterson[4], David M. Walters[2], Brent R. Johnson[2], and Mark Bagley[2]

[1]U.S. Environmental Protection Agency
Office of Research and Development
National Health and Environmental Effects Research Laboratory
Mid-Continent Ecology Division
Duluth, MN 55804

[2]U.S. Environmental Protection Agency
Office of Research and Development
National Exposure Research Laboratory
Ecological Research Division
Cincinnati, OH 45268

[3]Ohio River Valley Water Sanitation Commission
Cincinnati, OH 45228

[4]U.S. Environmental Protection Agency
Office of Research and Development
National Health and Environmental Effects Research Laboratory
Western Ecology Division
Corvallis, OR 97333

[5]National Park Service
1201 Oakridge Drive
Fort Collins, CO 80525

NOTICE

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SECTION AUTHORS

Addresses for authors are provided in each section.

Section 1: Ted R. Angradi, David W. Bolgrien, Terri Jicha, E. William Schweiger, and Brian H. Hill

Section 2: Ted R. Angradi

Section 3: Ted R. Angradi and Terri M. Jicha

Section 4: E. William Schweiger, Ted R. Angradi, and David W. Bolgrien

Section 5: Terri M. Jicha, Ted R. Angradi, and Brian H. Hill

Section 6: Ted R. Angradi and E. William Schweiger

Section 7: E. William Schweiger and Ted R. Angradi

Section 8: Erich B. Emery, Jeff A. Thomas, Mark Bagley, and Ted R. Angradi

Section 9: James M. Lazorchak, Erich B. Emery, David M. Walters, and Spence A. Peterson

Section 10: Ted R. Angradi, Donald J. Klemm, Jim M. Lazorchak, and Brent R. Johnson

Section 11: Brian H. Hill and James M. Lazorchak

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ACRONYMS, ABBREVIATIONS AND MEASUREMENTS UNITS USED IN THIS DOCUMENT

Acronyms and abbreviations

AFDM	ash-free dry mass
AFS	American Fisheries Society
ALK	alkalinity
AP	aquatic plant
BPJ	best professional judgment
CENR	(White House) Committee on the Environment and Natural Resources
CHL	chlorophyll
CPR	cardio-pulmonary resuscitation
CFR	Code of Federal Regulations
DBH	diameter at breast height
DC	direct current
DELT	deformities, erosions, lesions, and tumors
DFS	distance from shore
DI	de-ionized
DLG	digital line graph
DNR	Department of Natural Resources
DO	dissolved oxygen
EERD	Ecological Exposure Research Division
EMAP	Environmental Monitoring and Assessment Program
EMAP-SW	EMAP-Surface Waters Resource Group
EMAP-WP	EMAP Western Pilot Study
EPA	U.S. Environmental Protection Agency
GCM	geochemical markers
GPS	Global Positioning System
GIS	Geographic Information System
GRE	Great River Ecosystems
HDPE	high density polyethylene
ID	identification
IM	information management
INHS	Illinois Natural History Survey
IP	invasive plant

LTRMP	Long Term Resource Monitoring Program
LWD	large woody debris
MCS	main channel shoreline
MDC	Missouri Department of Conservation
MED	Mid-Continent Ecology Division
MSDS	Materials Safety Data Sheet
NAWQA	National Water-Quality Assessment Program
NHD	National Hydrography Database
NERL	National Exposure Research Laboratory
NHEERL	National Health and Environmental Effects Research Laboratory
NIOSH	National Institute for Occupational Safety and Health
NTU	nephelometric turbidity units
ORD	Office of Research and Development
OSHA	Occupational Safety and Health Administration
PDF	portable document format
PE	polyethylene
PFD	personal flotation device
QA	quality assurance
QCCS	quality control check sample
REMAP	Regional Environmental Monitoring and Assessment Program
SAV	submersed aquatic vegetation
SL	standard length
SMSU	Southwest Missouri State University
SOP	standard operating procedure
TBD	to be determined
TOC	total organic carbon
TSS	total suspended solids
TL	total length
UMR	Upper Missouri River
UN	United Nations
UMESC	Upper Midwest Environmental Sciences Center
USCG	U.S. Coast Guard
USEPA	U.S. Environmental Protection Agency
USDA	U. S. Department of Agriculture
USGS	U. S. Geological Survey
VHF	very high frequency

Acronyms and abbreviations, continued

VSS	volatile suspended solids
WAAS	wide-area augmentation system
WCC	water chemistry composite
WED	Western Ecology Division
YOY	young of the year
YSI Inc	Yellow Springs Instruments, Incorporated

Measurement units

amps	amperes
C	degrees Celsius
cm	centimeter
g	gram
gal	gallon
ha	hectare
Hz	Hertz
km	kilometer
L	liter
m	meter
m^2	square meters
mg/L	milligram per liter
mL	milliliter
mm	millimeter
µm	micrometer
µS/cm	microsiemens per centimeter
msec	millisecond
ppm	parts per million
psi	pounds per square inch
V	volts
VA	volt-ampere

Section 1

Introduction

Ted R. Angradi[1], David W. Bolgrien[1], Terri Jicha[1], E. William Schweiger[2], and Brian H. Hill[1]

This manual describes procedures for collecting samples and field measurements for biotic assemblages and abiotic characteristics of the Great Rivers of the Central Basin of the United States: the Missouri, Upper Mississippi, and Ohio Rivers. The purpose of this manual is to document the field procedures to be used in the Environmental Monitoring and Assessment Program for Great River Ecosystems (EMAP-GRE). In addition to the technical and logistic aspects of field operations, this manual emphasizes health and safety considerations, data quality assurance (QA), and information management (IM). Not included in this manual are laboratory protocols, sample-design details, or protocols for data analysis or interpretation.

The procedures in this manual are based on a variety of sources, including previously published EMAP manuals (e.g., Baker et al. 1997, Strobel and Heitmuller 2001, Peck et al., unpublished drafts), other EPA documents (Klemm et al. 1990, Barbour et al. 1999, Kaufmann et al. 1999, Flotermersch et al. 2000), an unpublished Upper Missouri River EMAP Field Operations Manual (Angradi et al. 2002), the unpublished findings of an EPA-sponsored workshop on Great River indicators (Angradi and Hill 2003), USGS NAWQA (Moulton et al. 2002) and USGS LTRMP protocols (e.g., Yin et al. 2000), the scientific literature, and the experience and professional judgement of section contributors.

This manual is designed to be a comprehensive set of required procedures and checklists for EMAP-GRE field sampling. This manual is also an informal safety and QA guidance document for field crews; it is a reference document for planning EMAP-GRE field operations, including office-based reconnaissance activities; it is a training tool for EMAP-GRE crews; and it serves as documentation of methods in support of EMAP-GRE publications and products.

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1 U.S. Environmental Protection Agency, Office of Research and Development, National Health and Environmental Effects Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804

2 National Park Service, 1201 Oakridge Drive, Fort Collins, CO 80525

Earlier versions of this manual were used in the 2004 and 2005 field seasons. The current version of the manual supports ongoing EMAP sampling for EMAP-GRE and other large river sampling programs.

1.1 Overview of EMAP

EMAP is a long-term research program focused on developing indicators and unbiased statistical designs for assessing the condition of aquatic ecosystems at a variety of spatial scales (USEPA 2002). Ecosystems included in past EMAP efforts have included lakes, estuaries, wetlands, and surface waters (wadeable and non-wadeable streams). The two main goals of EMAP (USEPA 2002) are relevant to the selection of indicators and approaches for assessment and to the field operations that generate the data upon which the assessments are based:

> **Develop the science needed for a state-based statistical monitoring framework to detect trends in condition of the Nation's aquatic ecosystems**. EMAP indicators are chosen for their usefulness in revealing the condition of aquatic ecosystems. Field operations must be designed so that the samples and measurements are collected in a manner that maximizes the reliability and sensitivity of the indicators. This includes the equipment, procedures, and personnel used; the spatial arrangement of samples; the timing of data collection and the habitats in which data are collected.
>
> **Transfer EMAP science and technology to the states, tribes, and EPA regions.** Indicators and methods developed for use by states and tribes must have attributes that favor their adoption. These attributes include cost-efficiency, public acceptance, and relevance to management. High quality assurance (QA) during field operations is also central to this EMAP goal since only data sets with known performance characteristics can be efficiently combined and shared (Barbour et al. 1999).

1.2 Overview of EMAP-GRE

1.2.1 Objectives and scope

The objectives of EMAP-GRE are to 1) develop monitoring and assessment tools for the Great Rivers of the Central Basin: the Upper Mississippi, Missouri, and Ohio Rivers; and 2) to demonstrate those tools in a regional assessment of Great River ecosystem condition. Because of the size and complexity of these river ecosystems, they represent a major assessment challenge for states and tribes. In general, robust GRE monitoring and assessment tools proven to be useful across the region are not yet available.

EMAP-GRE will address 3 general questions:

1. What proportion of the GREs of the Central Basin, expressed in river miles, are in good, fair, and poor condition?

2. What is the extent of aquatic, floodplain, and riparian habitat in the GREs of the Central Basin?

3. What is the relative importance of stressors (e.g., bank stabilization, excess nutrients, metals, invasive species, riparian disturbance) in the GREs of the Central Basin?

The resource population of interest for the EMAP-GRE assessment is selected habitats (described below) of the Missouri River from Fort Peck Dam in Montana to the confluence with the Mississippi River, the Mississippi River from Lower St. Anthony Falls in Minneapolis to the confluence with the Ohio River, and the Ohio River from the confluence of the Monongahela and Allegheny Rivers to the confluence with the Mississippi River (Figure 1-1). Fifteen states are included within the scope of EMAP-GRE.

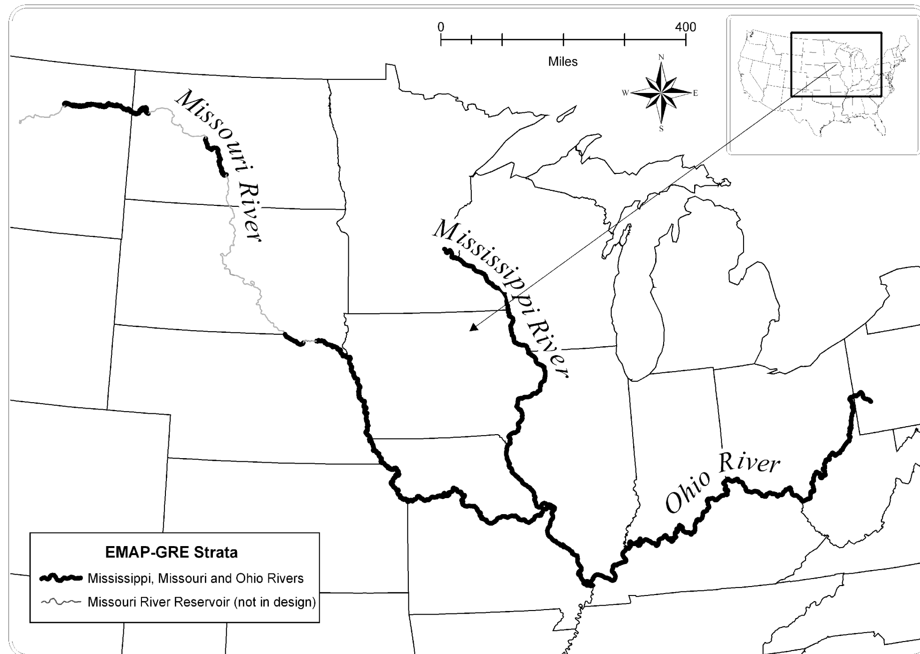

Figure 1-1. Geographic scope of EMAP-GRE. Mainstem Missouri River reservoirs are excluded from EMAP-GRE.

1.2.2 The EMAP-GRE sample design

The details of the EMAP-GRE sample design are beyond the scope of this manual. This manual describes activities that occur after field personnel have the “design file” in hand. The design file is a spreadsheet containing the locations and support information of all candidate sample sites for each river. The design file is discussed in Section 4 of this manual.

1.2.3 The index period

The index period is the period of the year in which sampling may occur. The index period for EMAP-GRE is July 1 – September 30. This period corresponds to a season of high biological activity, clement weather, and relatively stable flows.

1.2.4 EMAP-GRE habitats

Habitats included in EMAP-GRE are relevant to the goals of EMAP-GRE either because they 1) are specified in the Clean Water Act (i.e., water quality in the river channel), 2) are well documented to be essential for a reliable assessment of river ecosystem condition (e.g., vegetation in the riparian zones), or 3) reveal aspects of ecological condition not captured by sampling other aquatic habitats (e.g., littoral benthos). Off-channel habitat types, including backwaters, tributary mouths, and floodplain lakes are not sampled in EMAP-GRE.

Three basic habitats are included in EMAP-GRE:

1. Main channel. This habitat includes the fluvial channel containing the most discharge. Navigation pools on the Upper Mississippi River an Ohio Rivers are included. Lake Pepin on the Upper Mississippi River is included. Mainstem reservoirs on the Upper Missouri River are excluded.

2. Main-channel littoral zone. This habitat includes the main-channel margin to a maximum depth of 6 m or a maximum distance of 30 m from the wetted perimeter (whichever is closer to the shoreline). The depth criteria is defined by the effective sampling range of the electrofishing gear.

4. Main-channel riparian zone. This habitat includes terrestrial habitat in a 30-m wide zone adjacent to the main channel.

1.3 EMAP-GRE indicators

Indicators are measurements that characterize an ecosystem or one of its critical physical, biological, or chemical components. Indicators vary in their cost, variability, sensitivity to different stressors, and societal value (Jackson et al. 2000). Presumptive performance relative to these criteria provides the rationale for including each indicator in the EMAP-GRE assessment.

In April, 2003, an EMAP-GRE Indicator Workshop was held in Minneapolis, MN (Angradi and Hill 2003) to identify elements of GREs for which indicators should be developed and to identify appropriate measurements and methods for each indicator. Experts on indicators, the ecology of large rivers, and the EMAP approach participated in the workshop. The advantages and shortcomings of a large number of potential Great River indicators were considered. The indicators and methods included in this manual reflect those and subsequent discussions. Due to financial and other constraints, not all of the indicators identified in the workshop as potentially useful for assessing GREs are currently included in EMAP-GRE.

EMAP-GRE includes four types of indicators: condition indicators, stressor indicators, exposure indicators, and function indicators. *Condition indicators* are biotic or abiotic characteristics of an ecosystem that reveal the condition of an ecosystem or habitat relative to an environmental value or reference condition. Fish assemblage structure is an example of a biotic condition indicator. *Stressor indicators* characterize or quantify anthropogenic effects on the condition of ecosystems. River nutrient concentration is an example of a stressor indicator. *Exposure indicators* quantify exposure of the biota at one or more trophic levels to toxic contaminants. Fish tissue contamination is an example of an exposure indicator. *Function indicators* measure the magnitude or rate of an ecosystem process relevant to ecosystem condition. Sediment enzyme activity is an example of a function indicator.

Not all indicators will be sampled or measured in all habitats. In some cases, the indicator is only relevant to or present in a single habitat (e.g., riparian vegetation structure). In other cases, logistic considerations render collection of an indicator more feasible in some habitats than in others (e.g., benthic macroinvertebrates in littoral areas versus the main channel). Some indicators are presumed to reliably characterize GRE condition when measured in a particular habitat and are not measured elsewhere to reduce the overall effort required. For example, fish are sampled in the main-channel littoral zone of the main channel rather than in the thalweg or in backwaters.

1.3.1 Water chemistry and plankton (Section 5)

In EMAP-GRE, water chemistry data are primarily used to define reference conditions

and to identify stressor gradients. Great River stressors associated with water chemistry may include nutrient enrichment, inorganic contamination, anoxia, temperature stress, turbidity, and suspended sediment. Water chemistry sampling includes a grab sample of water for laboratory analysis and in-situ measurements, including dissolved oxygen, conductivity, pH, turbidity, total suspended solids (TSS), and seston geochemistry.

Plankton include algae (phytoplankton) and microinvertebrates (zooplankton) suspended in the water column. Plankton assemblages are potentially useful indicators of environmental condition because they are important to the trophic structure of larger rivers, and they are likely sensitive to a number of anthropogenic disturbances, including flow regulation, habitat alteration, invasive species, and contamination by nutrients, metals, and herbicides.

1.3.2 Aquatic vegetation (Section 6)

Aquatic vegetation has multiple ecological functions in Great River ecosystems. Aquatic plant communities generate dissolved oxygen, stabilize bed sediments, filter suspended sediment, and immobilize nutrients and toxic substances. Plant parts are an important food source for waterfowl and other wildlife. Submerged, floating, and emergent aquatic plants provide substrate for invertebrates and habitat for fish. Submerged aquatic vegetation (SAV) is sensitive to anthropogenic stressors, including excessive turbidity, sedimentation, flow modification, and exotic herbivores. Relating SAV community structure, abundance, and distribution to stressors can provide a biological basis for water quality criteria. For example, understanding the influence of turbidity on SAV beds could lead to development of light-related water quality criteria for Great Rivers (UMRCC 2003).

1.3.3 Riparian habitat (Section 7)

Interactions among aquatic and riparian ecosystem components are important in Great River ecosystem functioning and condition. Riparian ecosystems contribute to and moderate the flux of materials and energy between terrestrial and aquatic habitats within GREs. Riparian and shoreline habitat characteristics (vegetation, shoreline stability, human disturbance)

influence affect channel form, water velocity, substrate and other physical habitat attributes at multiple spatial scales. Riparian measurements are also important in EMAP-GRE because they provide robust abiotic indicators of human disturbance at the site scale that are useful for identifying reference sites.

1.3.4 Fish (Section 8)

EMAP-GRE fish sampling methods are designed to collect all but the rarest fish in near-shore littoral habitats at each site. The sample collected is assumed to accurately represent the proportional abundance of the targeted assemblage at the site. Benthic species that inhabit the thalweg or other deep main channel habitats may not be adequately sampled by electrofishing. Fish sample data include species composition, size distribution, and the occurrence of anomalies on individual fish. Other measures of assemblage structure and function can be calculated from the data and combined into indices of condition useful for assessing the condition of Great Rivers (Emery et al. 2003, Simon and Emery 1995).

1.3.5 Fish tissue (Section 9)

Fish tissue contaminants are an indicator of bioaccumulation of persistent toxic substances in the environment, and can be used to estimate exposure to contaminants associated with fish consumption for higher trophic levels. EMAP-GRE will focus on whole fish rather than on fillets because of its emphasis on the health of the ecosystem. Although whole-fish contamination is primarily an indicator of contaminant exposure to piscivorous wildlife, whole fish data are still relevant for estimating human exposure to contaminants through fish consumption.

1.3.6 Benthic macroinvertebrates (Section 10)

Benthic macroinvertebrates inhabit river bed sediments or adhere to hard substrates. Macroinvertebrates have several advantages as condition indicators (Barbour et al. 1999, Klemm et al. 1990). Macroinvertebrates are ubiquitous throughout all GRE aquatic habitats and

are relatively easy to collect in large numbers in most habitats. In some situations of multiple stressors, macroinvertebrate assemblages have diagnostic power to identify stressors. Benthic macroinvertebrates generally live one or two years, and they usually recolonize substrates relatively rapidly after disturbance. Therefore, they are likely most useful for detecting stressors at temporal scales of several months up a year or two. Mobility partly determines the spatial scale at which organisms respond to stress. Macroinvertebrates are relatively sessile and substrate specific, and are very sensitive to sedimentation, sediment contamination, and habitat alterations (e.g., rip rap). EMAP-GRE includes two types of macroinvertebrate sampling: shoreline (littoral) kick sampling, and snag surface sampling from a boat.

Two types of condition indicators can be developed based on macroinvertebrate assemblages: multimetric additive indices of condition, and multivariate approaches which use predictive modeling to assess condition. Multimetric indices combine various ecological attributes of the benthic assemblage into an index that reflects the condition of the assemblage and is compared to a reference value for assessment. Examples of the multimetric approach include Kerans and Karr (1994), Barbour et al. (1996), and Klemm et al. (2003). In the multivariate approach, abiotic data associated with each sample are used to predict which organisms should be in the sample based on a model developed from reference sites. Examples of this approach include Wright (1995) and Reynoldson et al. (1995).

1.3.7 Periphyton and sediment (Section 11)

Periphyton include algae, fungi, bacteria, protozoa, and associated organic matter on the surface of aquatic substrata. Periphyton is a useful indicator of environmental condition because periphyton is easy to collect and it responds rapidly to a number of anthropogenic disturbances, including habitat alteration and contamination by nutrients, metals, herbicides, hydrocarbons, and acids (Pan et al. 1996, Hill et al. 2003). Indicators based on periphyton may be based on assemblage species composition, cell density, ash-free dry mass, chlorophyll concentration, and enzyme activity. As for macroinvertebrates, a index can be developed which combines multiple ecological characteristics of periphyton into an additive index useful for bioassessment (Hill et al. 2003).

Benthic organisms are in intimate contact with river sediments and are influenced by the

physical and chemical properties of sediment. Sediment characteristics serve as exposure indicators for benthos, fish, and other wildlife (e.g., sediment toxicity) and as functional indicators of key ecosystem processes (e.g., nutrient dynamics as revealed by sediment enzyme activity) (Sinsabaugh and Foreman 2001, Hill et al. 2002).

1.4 Quality assurance/quality control

EMAP-GRE has a rigorous QA/QC program that includes all aspects of the project, including field operations, lab analysis, and information management. Generic field-operations-related QA/QC considerations are outlined in Table 1-1. More detailed QA/QC considerations are included in each section of this manual.

Table 1-1. Generic QA activities for EMAP-GRE field operations.

Category	Considerations
Training	All crews will be thoroughly and consistently trained for assigned field tasks, safety, and project QA procedures. Initial training is supplemented by annual "booster" training.
Standardization	Crews will receive standardized training based on this manual. Standardized field forms and labels will be used by all crews. Field instruments, sampling equipment, and supplies will be specified or supplied.
Calibration	Calibration of field instruments will be integrated with field operations.
Objectivity	Field operations are designed to minimize unnecessary subjectivity in measurements. To the extent possible, rules will be provided for site verification and other field decisions.
Communication	Regular communication between field crews and coordinating EPA personnel forestalls problems, misinterpretations, and supply shortages and promotes inter-river and among-crew uniformity. Field-season QA audits and post-field-season debriefings improve QA.
Documentation	Non-standard or unusual situations or conditions are documented with data quality flags and notes on the field forms and by communication with EPA scientists and IM personnel.
Information management	Web-based sample tracking and 100% data proofing are fully integrated into the program

1.5 Safety

Safety is paramount. Operating on large rivers is inherently hazardous and involves significant risks to crew health and safety. Obtaining data is always less important than maintaining the safety of the crew. Specific safety practices and precautions related to field procedures and logistics are integrated into this manual. These practices do not supercede or replace the safety practices or guidelines of other agencies whose personnel are conducting EMAP field work. Refer to Flotermersch et al. (2001) for additional guidance on operational safety when working on rivers.

1.6 Training

Training sessions were conducted prior to the first EMAP-GRE field season. The training included a full day of classroom instruction and a day of on-the-river demonstration of field operations. At a minimum, all crew leaders receive the complete training. Crew members

not present at the EPA training were trained in the field. Prior to subsequent field seasons, a "booster" training is held which reinforces the initial training, trains new crews leaders, and emphasizes any changes in procedures that may have occurred.

1.7 EPA support of field operations

EMAP-GRE field sampling will typically not be conducted by EPA personnel, but the EPA principal investigators will be available during the field season for consultation with field crews on sample design, logistics, methods, and other issues that arise during the field work. Table 1-2 includes contact information for project principal investigators and the EMAP data manager.

Table 1-2. Contact information for EPA-GRE principal investigators.

David Bolgrien (218) 529-5216 fax: (218) 529-5003 bolgrien.dave@epa.gov Lead role: Project administration, project public relations, site verification	Ted Angradi (218) 529-5243 (office) (720) 480-7321 (mobile) fax: (218) 529-5003 angradi.theodore@epa.gov Lead role: Field Operations Manual, logistics
Marlys Cappaert (541) 754-4467 fax: (541) 754-4338 cappaert.marlys@epa.gov Lead role: EMAP data manager, sample tracking	Terri Jicha (218) 529-5153 fax: (218) 529-5003 jicha.terri@epa.gov Lead role: Information management, water sampling, sample tracking

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Section 2
Overview of Field Operations

Ted Angradi[1]

This section describes the daily operational scenario for EMAP-GRE field activities. Included are field-crew configuration and responsibilities, a discussion of boat operations, a flow chart of daily operations, guidelines for recording data, and general safety considerations.

2.1 Crew configuration and responsibilities

EMAP-GRE field operations require at least two crews: a three-person fish-sampling crew and a three-or four-person river-sampling crew. In some cases the same crew may perform both functions. These crew sizes are only recommendations; alternative crew configurations are acceptable. Crew responsibilities are outlined in Table 2-1. Many logistical aspects of field operations such as site verification, lodging, transportation, and sample tracking and shipping may overlap between crews. In the field, each crew is supervised by a crew leader, who is responsible for daily operational planning, data quality, and safety.

Whenever possible, the crews should coordinate their activities. By visiting the same site on the same day, crews can share equipment, coordinate laying out the reach, and provide mutual support in case of a breakdown or safety emergency. River and fish-sampling at a site should be completed within five days of each other whenever possible.

2.2 Boat operations

2.2.1 Operational logistics

Each crew requires a boat for sampling. Care must be taken to maintain the boats in good order. Consult Flotermersch et al. (2001) for guidance on the logistics of boat operations for river sampling.

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1 U.S. Environmental Protection Agency, Office of Research and Development, National Health and Environmental Effects Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804

2.2.2 Navigation

The boat trip from the ramp to the sample site may be many miles, and may involve potential hazards. All boats should be equipped with a high-quality dash-mounted GPS/sonar unit with preloaded basemaps. Site location (latitude, longitude) data from the design file should also be loaded into the GPS units as waypoints. As part of pre-visit activities (described in more detail in Sections 3 and 4), crews should plan their route to make sure they use the closest suitable ramp, and that they are aware of any hazards, including locks, rapids, and shoals.

Table 2-1. Outline of the responsibilities of each field crew.

Crew	Habitats	Sampling responsibilities	Section of manual
Fish sampling	Near-shore littoral	Fish assemblages	8
		Substratum	8
		Fish cover	8
		Fish tissue for contaminants	9
		Fish tissue for DNA	8
River sampling	Main channel	Water chemistry	5
		Phytoplankton	5
		Macrozooplankton	5
		Microzooplankton	5
		LWD	10
	Near-shore littoral	LWD	10
		Benthic macroinvertebrates	10
		Snag macroinvertebrates	10
		Sediment	11
		Periphyton	11
		Aquatic vegetation	6
	Riparian zone	Bank characteristics	7
		Riparian vegetation structure	7
		Human disturbance	7
		Invasive plants	7

2.2.3 Boating safety

Boating on large rivers presents multiple safety hazards. The river must always be treated with respect to avoid situations that threaten the health and safety of crews. Table 2-2

lists safety recommendations related to general boat operations. Crews should also receive safe-boating training. Other safety consideration related to sampling are described in subsequent chapters.

2.3 Flow of daily operations

This section outlines a proposed general flow of daily operations for the fish and river sampling crews (Figure 2-1). The two different crews may operate independently or together depending on the schedule of operations. The details of each activity are provided in subsequent chapters. At any particular site, circumstances may require that the order of the activities be altered.

2.3.1 River-sampling crew

After completing base location activities (Section 3) and navigating to the sample site, the crew leader evaluates whether the site is safe to sample under the existing conditions (sampleability may be apparent at the boat ramp). See Section 4.6 for more guidance on sampleability. If the site is safely sampleable, the river-sampling crew collects depth- and width-integrated water and plankton samples and makes water quality measurements (Section 5). The crew then locates and flags two 500-m shoreline transects (if they have not already been flagged by the fish-sampling crew; site layout is described in Section 4). Two crew members collect a composite littoral macroinvertebrate sample (Section 10), a composite sediment sample (Section 11), and a composite periphyton sample (Section 11) at each of 11 stations along the primary 500-m shoreline transect. While this is occurring, the other crew member collects bank and riparian habitat data (Section 7) and aquatic vegetation data (Section 6), Following this, the crew returns to the boat, collects a macroinvertebrate sample from the surface of a snag in the channel, and collects LWD abundance data (Section 10). Before departing the site, the river-sampling crew performs a subjective overall site assessment (Section 7).

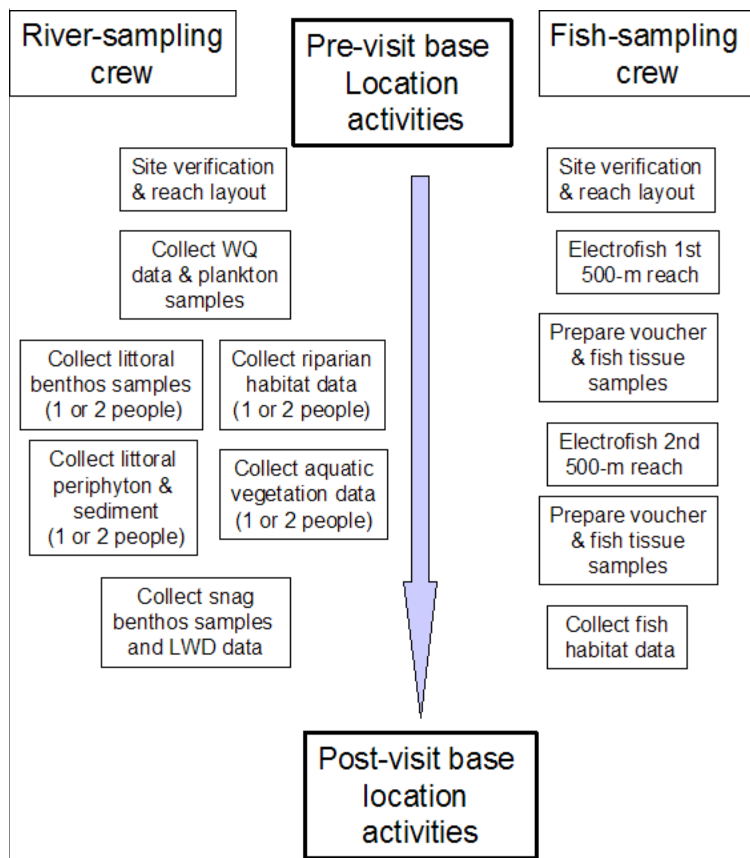

Figure 2-1. Flow chart of field activities for EMAP-GRE river- and fish-sampling crews (proposed). Each crew consists of at least three people. Site verification and the location and flagging of the MCS transects is done by the first crew to arrive at the site. The river-sampling crew may split up for the littoral sampling. One or two people can collect benthos, periphyton, and sediment samples, while the other crew member(s) collects riparian habitat and aquatic vegetation data. Pre- and post-visit activities are described in Section 3; site verification and reach layout is described in Section 4; sampling is described in Sections 5-11.

2.3.2 Fish-sampling crew

After completing base location activities (Section 3) and navigating to the sample site, the crew leader evaluates whether the site is sampleable under the existing conditions (sampleability may be apparent at the boat ramp). The fish-sampling crew locates and flags two 500-m near-shore transects to be electrofished (if they have not already been flagged). The

crew electrofishes each 500-m transect (Section 8). Captured fish are identified, measured, weighed, and released. A subsample of captured fish are retained as taxonomic vouchers and for fish tissue analysis (Section 9). The crew then collects substrate-size and fish cover data along each transect (Section 8).

2.3.2 River teams

The fish-sampling and river-sampling crew for each section of river together comprise a river team. Each team is assigned a three letter code that is used in certain field forms to aid with information management and sample tracking:

Team code	River	Name (headquarters location)
LCM	Mississippi	Lake City (Lake City, MN)
BVM	Mississippi	Bellvue (Bellvue, IA)
OLM	Mississippi	Onalaska (La Crosse, WI)
HSM	Mississippi	Havana (Havana, IL)
GRM	Mississippi	Great Rivers (Brighton, IL)
ORM	Mississippi	Open River (Jackson, MO)
MRU	Missouri	Upper Missouri River (Bismarck, ND)
MRM	Missouri	Middle Missouri River (Lincoln, NE)
MRL	Missouri	Lower Missouri River in Missouri and Kansas (Rolla, MO)
OHR	Ohio River	Ohio River (Cincinnati, OH)

Table 2-2. Safety considerations related to general boat operations.

- All field personnel should receive U.S. Coast Guard-approved safe boating and water safety training.

- Be conservative navigating the river. Grounding on sandbars, snags, or wing dams is extremely dangerous and damaging. Be aware that the safest and deepest approach to sites in secondary channels, backwaters, or among sandbars is usually from downriver.

- Silver carp (*Hypophthalmichthys molitrix*) >10 kg can leap >2 m out of the water. Several people have been seriously injured in carp collisions. Silver carp are present in the lower reaches of all three rivers. Be alert for leaping fish while running the river and during electrofishing.

- Trust the GPS unit for back-navigating shoals and at night. All crew members should know how to use the “man-overboard” feature on the GPS.

- Secure and stow dangerous objects forward when running. They become unguided missiles when the boat stops suddenly.

- Always double check the anchor set when landing on shore. Be aware that river stage may change dramatically in a short time and plan accordingly. **Always** anchor from the bow eye when in any current.

- Make sure the boat is equipped with enough suitable PFDs for everyone aboard, a throwable device, a fire extinguisher, and other required safety items.

- A large fluke anchor with a section of chain and a line buoy is recommend for anchoring in current in sand-bottom reaches. At least a 7:1 ratio of anchor line length:depth is recommended for anchoring in current.

- Carry a cell phone, tools, first aid supplies, engine oil, sun block, and insect repellent on board. Carry sufficient line to tow another boat back to the ramp.

- Check the VHF weather channel frequently for storm alerts. Thunder or lightning means **get off the river.**

- Perform a regular inspection of boat tie-downs, trailer connections, winch parts and cable, tires, bearings, and lights.

- When it is safe to do so, render immediate assistance to other boats or boaters in distress. Insure that a safe situation exists before returning to field work.

- In clear water, polarized sunglasses greatly improve the ability of the boat driver to see underwater hazards.

2.4 Guidelines for recording data and information

Following the guidelines for filling out field forms and sample labels (Table 2-3) is essential for quality assurance. Errors or sloppiness in recording data can result in data being lost from the program. Forms are designed to be optically scanned into electronic files. Photocopying alters the dimensions of forms so that they are not readable using the scanning system. Therefore, **only original forms and labels provided by EPA can be used.** Originals should be photocopied as soon as possible after field work.

Table 2-3. Guidelines for recording field data and other information (adapted from Peck et al., unpublished draft)

Recording data on field forms

- Use forms preprinted on water-resistant paper supplied by EPA.
- Header information on each field form links the data. Make sure headers are filled in completely on both sides of each form.
- Never mark on or around the corner blocks or ID box on field forms. These preprinted markings are necessary for the optical scanning software; obscuring them will affect performance.
- Write legibly using a soft pencil or pen so information can be read by the optical scanner. Erase mistakes completely and write in the correct value whenever possible.
- If a value must be lined out, write the correct value adjacent to the line-out so the data entry operator can find it.
- USE ALL CAPITALS WHEN ENTERING TEXT ON THE FIELD FORMS. Clearly distinguish letters from number (e.g., 0 vs. O, 2 vs Z, 7 vs T). Do not put lines through 7's, 0's, or Z's. Do not use slashes.
- WRITE IN ALL CAPITALS IN THE COMMENTS SECTION. Be concise but avoid abbreviations or shorthand notation. Attach additional sheets if necessary.
- Always record the full AFS common name for fish.
- Record data and information so that all the entries are obvious. Enter data completely in every field that is used. Follow the "comb" guidelines – print each number or letter in the individual space provided. Keep letters and numerals from overlapping.
- When values need to be circled, use these proportions: x
- Record data to the number of decimal places provided on the forms.
- If the measurement is zero, enter a zero. Blanks will be interpreted as missing data (which should be flagged).
- If the field calls for meters, enter the value in meters. Do not use different units and a notation to that effect. If the number is negative, enter the value and flag it as a negative value in the comments.
- Do not enter longitudes as negative values (as in the dossier). Use 2 spaces left of the decimal place for longitudes <100 degrees west; use 3 spaces left of the decimal place for longitudes ≥100 degrees west.
- Record information on each line even if it has to be recorded repeatedly (e.g., fish species names). Do not use a vertical line to indicate repeated entries.
- Make a copy of completed forms and return the original field forms to the data manager (Corvallis). Retain the copy in a master file for the site. Keep forms in order (they are numbered) and do not staple them together. Three-ring binders are a good option for the form copies.

Continued

Table 2-3. Guidelines for recording field data and other information, continued.

Data Flags

- Use only defined flag codes from the list below and record them on the data form in the appropriate field. If data are collected for which there is no space on the form, choose a flag and record the data in the comment section.

Flag	Comment
F1, F2, Fn	Miscellaneous comments assigned by field crew; use only once per form
K	Sample not collected or lost; no measurement made
U	Suspect sample, measurement, or observation; sample collected using a nonstandard procedure
Q	Unacceptable QC check associated with measurement

Sample Labels and Tracking

- Use adhesive labels with preprinted sample ID numbers for each type of sample.
- Record information on labels using a fine-point permanent marker (e.g., Sharpie). Cover labels with clear tape.
- Record sample ID numbers from the label on field forms and on sample tracking forms.
- Reconcile sample ID numbers on samples and tracking forms before shipping samples.
- Include a copy of the sample tracking form with each sample shipment (the original is part of data form packet for the site that is mailed to the information manager).

2.5 Collecting permits

All states require collecting permits for fish sampling. Federal permits may also be required. Some states require permits for collecting plants or macroinvertebrates. **Obtaining collecting permits and filing collecting reports is the responsibility of the field crews.** Copies of the permits should be carried on boats when sampling. Crews should closely follow the specifications of the permit(s). These specifications may include destruction of certain species if captured (e.g., Asian carp), notification of the permitting agency prior to field sampling, and submission of an annual report listing the fish collected and their disposition. Consult Walsh and Meador (1998) for a summary of state permitting agencies and their reporting requirements.

2.6 General safety considerations for field operations

Field work on Great Rivers is inherently hazardous and involves significant risks to crew safety and health. Safety considerations for boat operations are given in Table 2-2. Additional general safety considerations are presented in Table 2-4. Safety considerations associated with specific sampling activities and gear are presented in subsequent chapters. Additional resources include the American Red Cross and Handal (1992), Ohio EPA (1990), USCG (1987), and USEPA (1986). Web sites with useful safety information include www.cdc.gov/niosh (occupational safety), www.nws.noaa.gov/safety (weather safety), www.uscgboating.org (boating safety), and www.firstaidguide.net (includes insect bite information).

Personnel on EMAP-GRE field crews should be in sound physical condition, be able to swim, and have a physical exam annually or in accordance with their agency policy. **Crew members with "MedicAlert" health conditions (e.g., severe allergies, diabetes, susceptibility to seizures) should make crew leaders and other crew members aware of their condition, the symptoms, and the actions required in a health emergency.**

Water and sediment samples handled in the field should be considered potential health hazards due to toxic substances or pathogens. Personnel must be familiar with health hazards associated with using chemical fixing or preserving agents. Material Safety Data Sheets for all chemicals used in field operations must be available to personnel. Chemical wastes can cause

various hazards due to flammability, explosiveness, toxicity, causticity, or chemical reactivity. All chemical wastes must be discarded according to standardized health and hazards procedures (NIOSH 1981, USEPA 1986).

During the course of field activities, crews may observe apparent violations of environmental regulations, may discover improperly disposed hazardous materials, or may observe or cause an accidental spill or release of hazardous materials. In such cases, it is important that the proper actions be taken and that field personnel do not become exposed to harmful substances. The following guidelines apply (Peck et al., unpublished drafts):

- First and foremost during any environmental incident, it is extremely important to protect the health and safety of all personnel. Take any necessary steps to avoid injury or exposure to hazardous materials. If you have been trained to take action such as cleaning up a minor field spill during fueling of a boat, do so. However, always err on the side of personal safety.

- Field personnel should never disturb or retrieve improperly disposed hazardous materials from the field and bring them back to the facility for disposal. This action may worsen the impact to the area of the incident, incur liability, cause personal injury, and waste time and money. However, field personnel should not ignore environmental incidents. There is a requirement to notify the proper authorities of any incident of this type.

- For most environmental incidents, the following emergency telephone numbers should be provided to field crews as part of a Health and Safety Plan: state and tribal Departments of Environmental Quality or Protection, United States Coast Guard and the United States Environmental Protection Agency Regional Office. **In the event of a major environmental incident, the National Response Center and Terrorist Hotline should be contacted at 1-800-424-8802.**

2.7 Equipment and Supplies for EMAP-GRE sampling

Table 2-5 is a checklist of the generic equipment and supplies needed for all for EMAP-GRE field operations. Each section of the manual also has a checklist of specific equipment and supplies needed for sampling.

Table 2-4. General safety guidelines for field operations.

- Crews should receive adequate training including first aid, CPR, vehicle safety, boating and water safety, electrofishing safety, and laboratory safety.
- Crews should carry cell phones to maintain reliable communications, and should carry contact information for local police, ambulance, fire and rescue departments, and should program important numbers into the cell phones.
- All crew members should know the location of the truck keys.
- Serious health problems may be associated with working in polluted waters. Exposure to river water and sediments should be minimized, especially near effluent discharge points. Use gloves if necessary.
- All electrical equipment must bear the approval seal of Underwriters Laboratories and must be properly grounded.
- Use appropriate protective equipment (e.g., gloves, safety eyewear) when handling and using hazardous chemicals.
- Be aware of risks posed by and first aid for poisonous snake bites, bee stings, ticks, and poisonous plants. Plan for potential allergic reactions.
- Be aware of hypothermia and heat exhaustion risks, symptoms, and first aid.
- Use extreme care walking on ramps and shorelines, especially on riprap.
- All crew members should be aware of any "MedicAlert" conditions among crew members.

Table 2-5. Generic equipment and supply checklist for all EMAP-GRE field operations. Basic boat safety equipment not listed. Quantities are per boat.

Qty	Item	
1	River crew. Boat with a winch and GPS/sonar unit loaded with appropriate base maps	
1	Fish crew. Boat wired for electrofishing with GPS/sonar unit loaded with appropriate base maps	
1	Hand-held WAAS-enabled GPS unit with extra batteries	
6	Soft pencils for filling in field forms	
6	Fine- and medium-point permanent markers for labeling	
2	Form-holder clip-boards	
1 box	Clear tape strips for covering labels	
1	Multi-tool	
1	Scissors for cutting labels	
1	First-aid kit	
1 copy	EMAP-GRE Field Operations Manual	
1 set	State and federal collecting permits	
1	Health and Safety Plan with emergency contact information	
1	Cell phone	
1 or 2	Coolers with ice in sealed bags	
1	Portable freezer (optional)	

2.8 Literature cited

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Section 3
Base-Location Activities

Ted R. Angradi[1] and Terri M. Jicha[1]

Field crews conduct a number of activities at "base locations" before and after visiting each river site. Base locations will usually be either the crews' temporary lodging facility or a state or federal facility. Base-location activities are usually conducted on the same day as the sampling visit.

3.1 Pre-visit base-location activities

Pre-visit activities include confirming suitability and location of launching facilities, inspecting equipment, calibrating instruments, and assembling supplies and sample containers. Procedures and guidelines for these activities are described in this section.

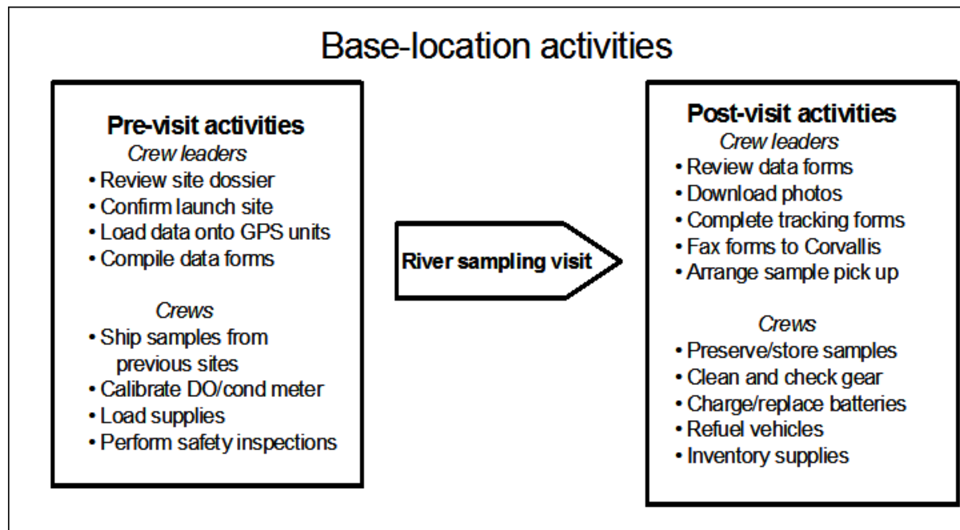

Figure 3-1. EMAP-GRE base-location activities. Fish- and river-sampling crew activities combined.

1 U.S. States Environmental Protection Agency, Office of Research and Development, National Health and Environmental Effects Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804

3.1.1 Confirming site location and status

The crew leaders will be provided with a “site dossier” containing location and access information for each site. The crew leaders should confirm that the nearest launch facilities are adequate and determine if there are any hazards or other special circumstances for the site. Additional reconnaissance of the site may be necessary. The site dossier and site verification are described in detail in Section 4.

3.1.2 Water quality meters

Five water quality parameters must be measured as part of EMAP-GRE field sampling: temperature, dissolved oxygen (DO), conductivity, pH, and turbidity (turbidity may be measured at the base location). No preferred manufacturer or model of meter is specified. Any reliable meter that can be properly calibrated and adapted to the depth-integrated sampling protocol (Section 5) is acceptable. A combination of meters (e.g., a DO/conductivity/temperature meter and a pH meter) or a single multi-parameter sensor may be used. Specific calibration and maintenance procedures and instructions for preparing calibration standards are not included in this manual and are the responsibility of the field crew.

3.1.3 Meter calibration

At the beginning of the sample season, the accuracy of the DO meter should be tested using a modified Winkler titration kit. Before each calibration attempt, inspect the membranes. If bubbles are present, or if the membrane is discolored or torn, replace the membrane according to manufacturer instructions. The DO meter should be calibrated daily either at the base location or at the sample site. Record all calibration details in a calibration log book including (as applicable) meter model and serial number, date, calibration standards used, elevation, and meter maintenance performed. Each entry in the log should be signed. The calibration log will be inspected during field QA audits and will become part of the EMAP-GRE data record.

Conductivity meters should be re-calibrated at least weekly. Follow instructions in the instrument’s operations manual and use a conductivity standard that is appropriate for the expected range. Calibration with a 1000 µS/cm standard is usually satisfactory. Record

calibration results in the calibration log book.

pH meters should be calibrated at least weekly at the base location using two standards, pH 7 and pH 10. Record calibration results in the calibration log book.

Turbidity meters are usually factory calibrated for a wide range of turbidity values, but should be calibrated at the beginning of the field season and about every two or three weeks thereafter using a standard in the range of expected values. Record calibration results in the log book.

Calibration of global positioning system (GPS) receivers should be done according to the manufacturers specifications for initialization, after replacing batteries, or if a new reference point is needed. Whenever possible, older units should be upgraded to modern units with built in or up-loadable base maps. Crews that use multiple GPS receivers should check them against each other and replace units that produce outlier coordinates.

3.1.4 Preparation of equipment and supplies

To ensure that all activities at a site can be conducted efficiently, field crews should check all equipment and supplies before leaving the base location. Sample containers and labels should be prepared ahead of time to the extent possible. Crews should inventory equipment and supplies prior to departure using the checklists appropriate for their responsibilities. Meters, probes, cameras, rangefinders, and other sensitive gear should be packed to avoid shock, exposure, and other damage.

Prepare stock preservative solutions as described in Table 3.1. Regulations pertaining to formalin and ethanol are in the Code of Federal Regulations. These requirements should be summarized for all hazardous material being used for the project and an MSDS file should be available to field personnel. Transport and store formalin and ethanol solutions in clearly-labeled, non-breakable containers within secondary containment and outside vehicle cabs or other unventilated spaces.

Refuel vehicles and conduct maintenance and repairs the night before sampling, if possible. Inspect vehicles every morning before departure. Check lights, boat tie downs, trailer connections, and tire pressure. Make sure the spare tire for the trailer is in good condition. Grease trailer hubs and jet drives (if applicable) frequently.

Table 3-1. Stock preservative solutions and instructions for their preparation. All stock solutions should be stored in clearly-labeled non-breakable containers. Labels should include container contents, date of preparation, and the initials of the preparer.

Solution	Use	Recipe
100% borax-buffered formalin[a] (pH 7-8)	Preservative for phytoplankton and periphyton; stock solution for fish	Add 20 g borax (hydrated sodium borate: $Na_2B_4O_7$-$10H_2O$) detergent (20 Mule Team®) per L 100% formalin (37% formaldehyde). Test pH with paper.
100% carbonate-buffered formalin (pH 10)[b]	Stock solution for macroinvertebrate preservative	Add 35 g Na_2CO_3 (also called “washing soda”) per L 100% formalin (37% formaldehyde). Test pH with paper.
12% buffered formalin-sugar solution[c] (pH 7-8)	Preservative for zooplankton	Add 600 mL 100% formalin, 5 tablespoons borax and 400 g table sugar (sucrose) to 4.4 L tap water (makes 5 L). Test pH with paper.
10% borax-buffered formalin	Preservative for fish[d]	Add 1 part 100% borax-buffered formalin to 9 parts tap water.
95% benzene-free ethanol	Stock solution for fish preservation (2005 DNA sites)[e]	Full strength agriculture-derived.
85% benzene-free ethanol	Field preservation of fish (2005 DNA sites)	Add 9 parts 95% ethanol to 1 part tap water.
75% benzene-free ethanol	Lab preservation of fish (2005 DNA sites)	Add 8 parts 95% ethanol to 2 parts tap water.
10% carbonate-buffered formalin (pH10)	Preservative for macroinvertebrates	Add 1 part 100% carbonate buffered formalin to 9 parts tap water. Test pH with paper.
Concentrated rose bengal solution	Stain added to macroinvertebrate samples	Add 1 teaspoon rose bengal powder to 1 L of 10% carbonate-buffered formalin stock solution.
1% bleach solution[f] (optional)	Used to decontaminate field gear	Add 1 part bleach to 99 parts water in a plastic spray bottle.
Clorox Formula 409® degreaser[g] (optional)	Used to decontaminate field gear	Full strength or 50% dilution.

a Formalin is a potential human carcinogen. Formalin should be handled only in well-ventilated areas while wearing chemical-resistant gloves and approved eye protection.

b High pH solution required to preserve mollusk shells (Merritt et al. 1997).

c Modified from Haney and Hall (1975); this solution should be kept on ice in the boat.

d Unbuffered 10% formalin may be used to preserve fish.

e Ethanol allows preserved fish to be sampled for DNA analysis.

f From Moulton et al. (2002); solution used to reduce risk of translocation of living organisms.

g Kills operculate snails (e.g., New Zealand mud snails).

3.2 Post-visit base-location activities

Upon reaching the base location or mobile laboratory after sampling, subsamples of river water are filtered for chlorophyll *a*, total suspended solids (TSS), and geochemical markers, and turbidity is determined (described in Section 5). The crew leader reviews data forms and sample labels for accuracy, completeness, and legibility; attempts to fill in (and flag – see Section 2.4) missing information as accurately as possible; and initials the data forms. Data files from digital cameras should be recorded, downloaded, and backed up as soon as possible. The other crew members should inspect and clean sampling equipment and boats as needed, check the inventory of supplies, preserve and store samples, and prepare unpreserved samples for shipment the next morning. Equipment maintenance tasks are listed in Table 3-2.

Invasive plants, fish, and invertebrates are potentially present at every EMAP-GRE sample site. Drain bilges and live wells on site and inspect all sampling equipment, including nets and boats, and remove any plant or animal material to prevent transporting nuisance species between sites. Decontaminate gear (Table 3-2) when there is a risk of organism translocation.

Crews should use discretion and common courtesy when using public facilities like river access parking areas for sample processing. Do not litter or dispose of sediment, fish, or preservative inappropriately. The curious public should be treated with respect and patience.

3.2.1 Sample packing, shipping, and tracking

An important aspect of program QA is sample handling between the field and the lab. Inconsistent practices compromise QA and complicate information management. Crews should follow the packing and shipping guidelines as closely as possible (Table 3-3 to 3-5) and should ship fresh, unpreserved samples (water, sediment, aquatic plant voucher specimens) as soon as possible after collection. Sample types, sample codes, shipping destinations, and which tracking form to use for each sample type are summarized in Table 3-6.

Samples that must be frozen (chlorophyll filters and fish tissue) may be shipped in

weekly batches but should be shipped as soon as possible if a freezer is not available. Samples preserved by drying (TSS filters, geochemical markers) can be shipped as a batch at the end of the field season. Formalin- or ethanol-preserved samples can be shipped in batches at the end of the season (4% formalin) or retained for later transport to the laboratory (10% formalin, 75% ethanol).

All samples, whether they are to be shipped immediately or retained and shipped later are recorded on laboratory-specific sample tracking forms (Figure 3-2 to 3-9) which are copied and included with each shipment. These forms also serve as a chain-of-custody between field crews and the laboratory contact (the sample tracking responsibilities of the laboratory contact are described in Section 3.3). Original sample tracking forms are faxed to the EMAP data manager in Corvallis (541-754-4637) and then are added to the form package for the site which will eventually be shipped to the data manager. Customer copies of courier airbills should be saved in case of loss by the courier. Sample tracking is described in more detail in Table 3-7.

3.2.2 Samples not preserved with formalin

Samples not preserved with formalin include water, chlorophyll, TSS and geochemistry filters, sediment, fish tissue samples for DNA analysis, and aquatic plant voucher specimens. Each sample should be entered as a separate line on the appropriate sample tracking form.

Guidelines for packing and shipping these samples are given in Table 3-3. Use ice substitute packs whenever possible to avoid leakage due to melting ice. When shipping with ice, use block ice when available. Ice and ice substitute packs should be sealed in plastic bags labeled "ice." Seal cooler lids with tape to prevent leakage in transit. A completed return airbill (including billing account information) should be enclosed in each cooler so coolers do not accumulate at laboratories.

Ship water chemistry and sediment samples **as soon as possible** after collection to meet holding time requirements. Water and sediment samples that cannot be shipped immediately should be refrigerated at 4°C. Samples collected on Friday and held over the weekend should be shipped "same-day delivery" on Monday. Follow cooler packing instructions

in Table 3-3. Be sure that at least half the final weight of the packed cooler is ice. Frozen samples (fish tissue, chlorophyll filters) can be shipped in small batches comprising no more than a week's worth of samples. However, if a freezer is not available, ship the chlorophyll and fish tissue samples as soon as possible. Be sure labels are protected and the samples are well sealed. Double-bag ice so that meltwater does not contaminate samples.

Table 3-2. Equipment care after each site visit.

- Prior to departing the river, drain all water from bilges and live wells.
- At the ramp or at the base location, inspect boats, trailers, and other gear for plant fragments or animal remains and remove them. If appropriate, decontaminate gear with a 1% bleach solution to reduce risk of organism translocation.
- Rinse water quality apparatus several times with distilled water after use.
- Rinse the periphyton sampling equipment with tap water after use.
- Rinse conductivity probes with deionized water and store moist.
- Drain water sampling hoses.
- Check fish dip nets, sieves, plankton, and benthos nets for holes and repair or replace.
- Store nets dry to prevent mildew.
- Inventory equipment and supplies and relay needs to field coordinator.
- Examine DO meter membrane for cracks, wrinkles, or bubbles; replace if necessary.
- Charge or replace all batteries as needed.
- Refuel boats, trucks, and generators.

3.2.3 Samples preserved with formalin or ethanol

Formalin or ethanol-preserved samples include macroinvertebrates and fish specimens preserved in 10% borax- or carbonate-buffered formalin, periphyton and plankton samples preserved in 4% borax-buffered formalin, and 2005 fish specimens for DNA analysis preserved in 85% or 75% ethanol. Samples preserved in ethanol or 10% formalin must be stored and transported in secondary containers (tubs or coolers) and stabilized to prevent spillage.

For each sample, enter the sample ID from the label, the sample type, number of containers, and any comments on the appropriate sample tracking form. For forms that include only retained samples, leave the date sent and airbill number fields blank and note in the comments which samples are being retained. When these samples are to be shipped or transported to a lab, fill in the missing information and re-fax the completed forms to Corvallis. Samples preserved in ethanol or 10% formalin should be retained by the crews until they can be conveniently transported to the appropriate laboratory. Leave copies of the completed sample tracking forms with the samples where they are being stored prior to delivery to the lab. Tables 3-4 and 3-5 provide guidance for formalin- and ethanol-preserved samples that need to be shipped by commercial carrier.

3.2.4 Inter-laboratory sample shipments

Some sample types are created in the laboratory by subsampling the original sample (Table 3-6). There are three types of inter-laboratory samples: unpreserved sediment samples shipped from NERL to MED, and preserved water samples for TOC and dissolved metals shipped from UMESC to MED. All the sample packing, shipping, and tracking guidelines for samples shipped from the field apply to inter-lab shipments. The sample tracking form and label for inter-laboratory shipments is shown in Figures 3-2 and 3-3.

3.3 Sample tracking responsibilities of laboratory contacts

Sample tracking forms faxed from the field to the EMAP data manager will be forwarded via email to the appropriate laboratory contact in the form of *.tiff image files. This email should

arrive in advance of the shipped samples and serves to notify the laboratory contact that the samples are in route **or** are being retained by the crew for later shipment or hand delivery. When the samples arrive at the laboratory, the laboratory contact should check the contents of the shipment against the tracking forms included with the shipment and forwarded by the data manager, and assign condition codes to each sample. If there are problems with the shipment (e.g., the sample is warm or mislabeled), the laboratory contact should communicate with the field crew to prevent future problems.

As soon as possible after the samples are checked in, the laboratory contact must enter the sample information into the EMAP Surface Water Information Management (SWIM) sample tracking web page at https://emapsw.cor.epa.gov/tracking/labsamples.php3. When sample information is entered in SWIM, the EMAP data manager is informed that samples have been received at the laboratory. Inter-laboratory samples are treated the same as samples shipped from the field. To access the SWIM sample tracking web page, the IP address from the lab's computer(s) must be supplied to the EMAP data manager (Section 3.4). A username and password will be provided to each laboratory contact.

3.4 Information management

A copy should be made of each set of completed and reviewed field forms. The originals are arranged in numeric order (do not staple forms together) and mailed or shipped to the EMAP data manager in Corvallis approximately monthly. The copies should be retained by the field crew. Crews should maintain a master file for each site including the site dossier (described in Section 4), copies of all field forms, sample tracking forms, air bills, data disks, and any other documents that pertain to the site. Mail forms to:

Marlys Cappaert
EMAP Data Manager
Computer Sciences Corporation, c/o U.S. EPA, NHEERL/WED
200 S.W. 35th Street
Corvallis, OR 97333
(541) 754 - 4467

3.5 Equipment and supplies

A checklist of equipment and supplies required for base-location activities is presented in 3-8. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

Table 3-3. General guidelines for packing and shipping unpreserved samples (adapted from Peck et al., unpublished drafts).

Sample type (container)	Guidelines
	Samples requiring refrigeration (4° C)
Water chemistry (4-L cubitainer and 500-mL bottle) Sediment (PE bag) Plant voucher specimens (plastic bag)	• Ship on day of collection or within 24 h by overnight courier. • Use fresh block ice in labeled ("ice") plastic bags. • Final weight of cooler should be at least half ice. • Line cooler with a plastic bag. • Cover sample labels with clear tape to prevent label loss. • Confirm that sample ID data on the sample tracking form and field forms agree. • Enclose a copy of the completed sample tracking forms and a pre-paid return airbill in each cooler in a self-sealing plastic bag. • Minimize the volume of air space in the packed cooler.
	Samples requiring freezing (-20° C) within 24 h of collection
Fish tissue (plastic bags) Chlorophyll (filter holders)	• If samples cannot be kept frozen in field, ship on day of collection or within 24 h by overnight courier. • Frozen fish tissue samples should be shipped as soon as they are frozen solid. • Frozen chlorophyll samples should be shipped in weekly batches. • Use fresh block ice in labeled ("ice") plastic bags. • Final weight of cooler should be at least half ice. • Cover sample labels with clear tape to prevent label loss. • Package samples so that meltwater does not contaminate them. • Confirm that sample ID data on the sample tracking form and field forms agree. • Enclose a copy of the sample tracking form(s) in a self-sealing plastic bag. • Minimize the volume of air space in the packed cooler.
	Samples requiring drying (30 - 50° C) for 24 h
Geochemical markers (filter holder) TSS (filter holder)	• If no oven is available, place in an air-conditioned room for 24-48 h with lids ajar; cover with a sheet of foil to exclude dust. • Cover sample labels with clear tape to prevent label loss. • Confirm that sample ID data on the sample tracking form and field form agree. • Enclose a copy of the sample tracking form in a self-sealing plastic bag. • Dried filters should be shipped as a batch at the end of the season in the tray boxes they came in (if available).

Table 3-4. General guidelines for storing and shipping formalin-preserved samples (adapted from Peck et al., unpublished drafts). In most cases, formalin-preserved samples will not be shipped, but will be carried by truck to the laboratory.

Sample type (container)	Final preservative strength of sample	Guidelines
Periphyton (500 mL bottle)[a] Macrozooplankton (250 mL bottle)[a] Microzooplankton (250 mL bottle)[a] Phytoplankton (2 L bottle)[a]	4% borax-buffered formalin	• Labels or tags placed inside sample jars must be of water-resistant paper. • The adhesive label on the outside of the container should be completely covered with clear tape. • Confirm that sample ID data on the sample tracking forms and field forms agree. • Store samples in secondary containers at base location. • Enclose copies of the sample tracking forms in a self-sealing plastic bag with stored samples.
Fish specimens (jars of various sizes)	10% borax-buffered formalin	
Macroinvertebrate samples (250-mL and 500-mL jars)	10% carbonate-buffered formalin	

Packaging and Shipping Guidance for samples preserved in formalin (IATA instructions 914, no limit)	
Inside packaging	HDPE bottles with leakproof screw-top cap that meets UN spec IP2. Seal cap with a strip of plastic tape. Fill jar to shoulder to provide headspace.
Outside packaging	Screw top plastic bucket (20 L) with ratcheted lid is recommended (UN spec 1H2). Line container with plastic bag meeting IP5 specifications.
Absorbent material	Not required. Stabilize contents with packing peanuts.
Labeling	Outside package marked with UN shipping name and ID no.: “Environmentally hazardous substance, liquid, n.o.s. (Formalin < 5%), UN3082", and a “Class 9 Miscellaneous” label, and at least two package orientation labels should be affixed to the container.
Shipping forms	Include packing list with each container. Note total quantity of formalin in liters and the gross container weight in pounds. Prepare Shipper Manifest prior to shipment.

[a] Preserved periphyton and plankton samples may be shipped as unpreserved samples because formalin concentration is low (<5%).

Table 3-5. General guidelines for storing and shipping ethanol-preserved samples (adapted from Peck et al., unpublished drafts). In most cases, ethanol-preserved samples will not be shipped, but will be carried by truck to the laboratory.

Sample type (container)	Final preservative strength of sample	Guidelines
2005 fish tissue DNA specimens (jars of various sizes)	75-85% ethanol	• Labels or tags placed inside sample jars must be of water-resistant paper. • The adhesive label on the outside of the container should be completely covered with clear tape. • Confirm that sample ID data on the sample tracking forms and field forms agree. • Store samples in secondary containers at base location. • Enclose copies of the sample tracking forms in a self-sealing plastic bag with stored samples.
Packaging and Shipping Guidance for samples preserved in formalin (IATA instructions 307, 60-L limit)		
Inside packaging	HDPE bottles with leakproof screw-top cap that meets UN spec IP2. Seal cap with a strip of plastic tape. Fill jar to shoulder to provide head space.	
Outside packaging	Screw top plastic bucket (20 L) with ratcheted lid is recommended (UN spec 1H2). Line container with plastic bag meeting IP5 specifications. Each pail can hold no more than 5.0 L total liquid.	
Absorbent material	Sufficient volume of absorbent material (vermiculite, UN A100) Absorbent sheets or equivalent) to absorb contents of all inner packaging. Stabilize contents with packing peanuts.	
Labeling	Outside package marked with UN shipping name and ID no.: “Alcohol, flammable, toxic, n.o.s. (Denatured alcohol), UN1986, and a “Class 3 flammable” label, and a “Class 6 Toxic” label, and at least two package orientation labels should be affixed to the container.	
Shipping forms	Include packing list with each container. Note total quantity of formalin in liters and the gross container weight in pounds. Prepare Shipper Manifest prior to shipment. There is a 60-L limit for a single shipment.	

Table 3-6. Summary of sample type, sample codes amd shipping destination. Each sample destination (lab) has a unique sample tracking form (Figures 3-2 to 3-9). Preserved samples are usually retained and shipped in batches or transported to the lab at the end of the season. Shipped samples are not preserved and are shipped as soon as possible after collection. Fish voucher specimens are retained by the crew for later laboratory identification.

Sample type	Type code	Shipped immediately or preserved	Sample destination	Comments
Phytoplankton composite	PP	Retained	MED	Carried to lab or shipped at end of season.
Benthos kick sample composite	BK	Retained	MED	Carried to lab.
Benthos snag sample	BS	Retained	MED	Carried to lab.
Periphyton composite	PA	Retained	MED	Carried to lab or shipped at end of season.
Sediment chemistry subsample	SC	Shipped	MED	Inter-lab sample. Shipped fresh from UMESC to MED.
Total organic carbon	TC	Shipped	MED	Inter-lab sample. Shipped in weekly batches from UMESC to MED.
Dissolved metals	DM	Shipped	MED	Inter-lab sample. Shipped in weekly batches from NERL to MED.
Small fish tissue	ST	Shipped	NERL	Shipped frozen. May be shipped in weekly batches.
Large fish tissue	LT	Shipped	NERL	Shipped frozen. May be shipped in weekly batches.
Sediment grab sample	SG	Shipped	NERL	Shipped fresh.
Fin tissue DNA	DS	Shipped	NERL	Shipped frozen. Shipped weekly with ST and LT samples
Water chemistry composite	WC	Shipped	UMESC	Shipped fresh.
Alkalinity grab	AL	Shipped	UMESC	Shipped fresh.
Aquatic plant vouchers	AP	Shipped	UMESC	Shipped fresh.
Geochemical markers filter	GF	Retained	Stroud	Shipped dried. Ship at end of season.
TSS filter pair 1	SS1	Retained	Stroud	Shipped dried. Ship at end of season.
TSS filter pair 2	SS2	Retained	Stroud	Shipped dried. Ship at end of season.
Chlorophyll filter	CF	Retained	Univ Louisville	Shipped frozen weekly.
Macrozooplankton (63-µm mesh)	BZ	Retained	Univ Louisville	Samples from Ohio River only. Ship at end of season.
Microzooplankton (20-µm mesh)	LZ	Retained	Univ Louisville	Samples from Ohio River only. Ship at end of season.
Macrozooplankton (63-µm mesh)	BZ	Retained	INHS	Samples from Mississippi River only. Ship at end of season.
Microzooplankton (20-µm mesh)	LZ	Retained	INHS	Samples from Mississippi River only. Ship at end of season.
Macrozooplankton (63-µm mesh)	BZ	Retained	SMSU	Samples from Missouri River only. Ship at end of season.
Microzooplankton (20-µm mesh)	LZ	Retained	SMSU	Samples from Missouri River only. Ship at end of season.
Composite fish vouchers (field)	CV	Retained	MED	Processed in lab of fish-sampling crew; tracked on MED form.
Fish species vouchers	SV	Retained	MED	Carried to MED from crews' labs; tracked on MED form.

Table 3-7. Sample tracking procedures.

1.	Complete the laboratory-specific sample tracking forms for the site (if it is sampled) including all samples collected regardless of whether they are to be immediately shipped to a lab or are to be preserved and retained. Fill in the site ID, sample date, visit number, crew leader, and team (see Section 2.3.2)
2.	Fill in the sample ID for each sample and circle the sample type that applies. Enter the number of containers holding the sample and (if applicable) whether the samples are to be shipped immediately or retained by the crew and shipped as a batch or retained until the end of the season. The condition code is filled in at the lab on receipt of the shipment.
3.	Fill in the date that samples are to be shipped and the airbill number. If the form only includes samples that are to be retained and not shipped immediately, do not fill in the date sent or the airbill Number. Include a copy of the sample tracking form(s) in each container shipped to each lab. Place forms in a self sealing plastic bag in the container. If the samples are delivered to a FEDEX location directly after sampling before the form can be copied), a hand made copy of the sample tracking form should be made and faxed in as soon as possible
4.	Fax all the completed sample tracking forms (including inter-lab transfers and retained samples) and the field verification form to the EMAP data manager (contact information in Table 1-2). The field verification form must be faxed in after every site visit, even if the site is not sampled for some reason. The faxed sample tracking forms will be forward via email as *.tiff files to the appropriate lab contacts so they will have a prior notification of sample shipments.
5.	Make copies of the sample tracking forms so a copy can accompany each shipment to the lab and can be associated with every preserved and retained sample.
6.	Ship the samples via overnight carrier. Friday samples should be shipped Monday morning for same-day delivery. Retain the customer copies of the airbills in the master file for the site. The laboratory will contact the field crews if the shipment does not arrive as expected, or if the shipment does not match the sample tracking form.
7.	When the retained samples are to be shipped or carried to the lab, fill in the Date Sent and Airbill Number (if the information is missing) on the original sample tracking form, and re-fax the form to Corvallis.
8.	Return the original sample tracking form to the form packet for the site.

Table 3-8. **Checklist of supplies for base-location activities.**

Qty	Item	
Before departure to site		
1	Site dossier	
1	Instrument calibration supplies	
Packing and shipping supplies (per site)		
2	1-gallon self-sealing plastic bags for tracking forms	
1-2	30-gallon plastic garbage bags to line coolers for shipping	
1-2	Coolers for shipping samples	
1	Containers suitable to transport and/or ship preserved samples	
1-2	Shipping air bills and sleeves	
variable	Block ice or ice substitute	
1 roll	Clear tape to seal coolers	

3.6 Literature cited

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Rivers and Streams, U.S. Environmental Protection Agency, Washington, DC.

Peck, D. V., Herlihy, A. T., Hill, B. H., Hughes, R. M., Kaufmann, P. R., Klemm, D. J., Lazorchak, J. M., McCormick, F. H., Peterson, S. A., Ringold, P. L., Magee, T. and Cappaert, M. R. Unpublished draft. Environmental Monitoring and Assessment Program - Surface Waters Western Pilot Study: Field Operations Manual for Wadeable Streams, U.S. Environmental Protection Agency, Office of Research and Development, Washington, DC.

Draft

EMAP-GRE TRACKING - MED

SITE ID: GRW04449-

DATE COLLECTED: ___ / ___ / 2 0 0 __

TEAM: ____

ANNUAL VISIT NUMBER: ☐ 1 ☐ 2

CREW LEADER: ________

AIRBILL NUMBER: ________

DATE SHIPPED: ___ / ___ / 2 0 0 __

Sample ID	Sample Type	# of Containers	Cond. Code	Shipped/ Retained	Comments
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	
	PP BK BS PA CV SV			S R	

Sample Types	Condition Codes	Chain of Custody	Contact Information
PP = Phytoplankton BK = Benthos kick BS = Benthos snag PA = Periphyton CV = Composite fish voucher SV = Fish species voucher	C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK =Sample OK **Filled in at lab**	Date Received: ____ / ____ / ______ Received by: __________ **Filled in at lab**	**Tracking** Marlys Cappaert p) 541-754-4467 **Lab** Terri M Jicha US Environmental Protection Agency- MED 6201 Congdon Blvd Duluth, MN 55804 p) 218-529-5153 f) 218-529-5003 jicha.terri@epa.gov

FAX TO
Tracking: 541-754-4637

1.

Figure 3-2. Sample tracking form for samples shipped or carried to the Mid-Continent Ecology Division (MED) Lab.

Draft

EMAP-GRE TRACKING - NERL

PAGE: ____ OF ____

SITE ID: GRW04449-

DATE COLLETED: ___ / ___ / 2 0 0 _

TEAM: ____ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 CREW LEADER: ____

AIRBILL NUMBER: ____ DATE SHIPPED: ___ / ___ / 2 0 0 _

Sample ID	Sample Type	# of Containers	Cond. Code	Shipped/ Retained	Common name, photo file, or comments
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	
	DS ST LT SG			S R	

Sample Types	Condition Codes	Chain of Custody	Contact Information
DS = DNA ST = Small fish tissue LT = Large fish tissue SG = Sediment grab	C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK =Sample OK **Filled in at lab**	Date Received: ___ / ___ / ___ Received by: ______ **Filled in at lab**	**Tracking** Marlys Cappaert p) 541-754-4467 **Lab** Mark Smith Sobran Environmental National Exposure Research Laboratory 26 W. Martin Luther King Dr Cincinnati, OH 45268 p) 513-569-7091 f) 513-569-7554 smith.marke@epa.gov

FAX TO Tracking: 541-754-4637

3.

Figure 3-3. Sample tracking form for fish tissue and sediment samples shipped to the EPA National Exposure Research Laboratory (NERL). Write the fish species collected under "comments."

Draft

EMAP-GRE TRACKING - UMESC

SITE ID: GRW04449-______ DATE COLLECTED: __/__/2 0 0_

TEAM: ____ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 CREW LEADER: ____________

AIRBILL NUMBER: ____________ DATE SHIPPED: __/__/2 0 0_

Sample ID	Sample Type	# of containers	Cond. Code	Comments
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			
	WC AL AP			

*All samples to be shipped ASAP

Sample Types	Condition Codes	Chain of Custody	Contact Information
WC = Water chemistry AL = Alkalinity AP = Aquatic plants	C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK =Sample OK **Filled in at lab**	Date Received: ____/ ____/ ______ Received by: ____________ **Filled in at lab**	**Tracking** Marlys Cappaert p) 541-754-4467 **Lab** Shirley Yuan, Chemist U.S.G.S., Upper Midwest Environmental Sciences Center 2630 Fanta Reed Road La Crosse, WI 54603 p) 608-781-6302 f) 608-783-6066 Xiaoli_Yuan@usgs.gov

FAX TO
Tracking: 541-754-4637

4.

Figure 3-4. Sample tracking form for water samples and aquatic plant specimens shipped or carried to the Upper Midwest Environmental Sciences Center (UMESC).

Draft

EMAP-GRE TRACKING - STROUD

SITE ID: GRW04449-

DATE COLLECTED: ___ / ___ / 2 0 0 _

TEAM: ______

ANNUAL VISIT NUMBER: ☐ 1 ☐ 2

CREW LEADER: ______

AIRBILL NUMBER: ______

DATE SHIPPED: ___ / ___ / 2 0 0 _

Sample ID	Sample Type	# of containers	Cond. Code	Comments
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			
	GF SS1 SS2			

* Retain until end of field season. Be sure sample is dry.

Sample Types	Condition Codes	Chain of Custody	Contact Information
GF = Geochemical markers filter SS1 = Total suspended solids filter pair 1 SS2 = Total suspended solids filter pair 2	C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved (wet) W = Warm OK =Sample OK	Date Received: ____/ ____/ ______ Received by: ______ **Filled in at lab**	**Tracking** Marlys Cappaert p) 541-754-4467 **Lab** Mark Monk Geochemistry Department Stroud Water Research Center 970 Spencer Road Avondale, PA 19311 p) 610-568-2153 x 273 f) 610-268-0490 mmonk@stroudcenter.org

FAX TO
Tracking: 541-754-4637

5.

Figure 3-5. Sample tracking form for filters shipped to Stroud Water Research Center.

Draft

EMAP-GRE TRACKING - University of Louisville

SITE ID: GRW04449- DATE COLLECTED: ___/___/ 2 0 0 _

TEAM: ____ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 CREW LEADER: ____________

AIRBILL NUMBER: ____________ DATE SHIPPED: ___/___/ 2 0 0 _

Sample ID	Sample Type	# of containers	Cond. Code	Shipped/ Retained	Comments
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	
	CF BZ LZ			S R	

* Ship weekly

Sample Types	Condition Codes	Chain of Custody	Contact Information
CF = Chlorophyll filter BZ = Macrozooplankton (63 um) LZ = Microzooplankton (20 um)	C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK =Sample OK Filled in at lab	Date Received: ___/___/____ Received by: ________ Filled in at lab	Tracking Marlys Cappaert p) 541-754-4467 Lab Richard Schultz Environmental Analysis Laboratory Department of Biology 139 Life Sciences University of Louisville Louisville KY 40292 EXCEPT Fed EX uses 40208 p) 502-852-2543 f) 502-852-0725 reschu01@gwise.louisville.edu

FAX TO
Tracking: 541-754-4637

6.

Figure 3-6. Sample tracking form for all chlorophyll filters and Ohio River zooplankton samples shipped or carried to the University of Louisville.

Draft

EMAP-GRE TRACKING - **Illinois Nat. Hist. Surv.**

SITE ID: GRW04449-

DATE COLLECTED: ___ / ___ / 2 0 0 _

TEAM: ______ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 CREW LEADER: ______

AIRBILL NUMBER: ______ DATE SHIPPED: ___ / ___ / 2 0 0 _

Sample ID	Sample Type	# of containers	Cond. Code	Comments	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	
	BZ LZ			S R	

* Shipped in batches

Sample Types	Condition Codes	Chain of Custody	Contact Information
BZ = Macrozooplankton (63 um) LZ = Microzooplankton (20 um)	C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK =Sample OK **Filled in at lab**	Date Received: ___ / ___ / ___ Received by: ______ **Filled in at lab**	**Tracking** Marlys Cappaert p) 541-754-4467 **Lab** Eric Ratcliff Illinois Natural History Survey Great Rivers Field Station 8450 Montclair Ave. Brighton, IL 62012 p) 618-466-9690 f) 618-466-9688 ratcliff@inhs.uiuc.edu

FAX TO
Tracking: 541-754-4637

7.

Figure 3-7. Sample tracking form for Mississippi River zooplankton samples shipped or carried to the Illinois Natural History Survey, Great Rivers Field Station.

56259

EMAP-GRE TRACKING - SMSU

SITE ID: GRW04449-

DATE: ___ / ___ / 2 0 0 _

TEAM: ______

VISIT NUMBER: ☐ 1 ☐ 2 ☐ 3 ☐ 4

CREW LEADER: ______________________

AIRBILL NUMBER: ______________________

DATE SENT: ___ / ___ / 2 0 0 _

Sample ID	Sample Type	# of containers	Cond. Code	Comments
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			
	BZ LZ			

Sample Types	Condition Codes	Chain of Custody	Contact Information
BZ = Macrozooplankton (63 um) LZ = Microzooplankton (20 um)	C = Cracked jar F = Frozen L = Leaking ML = Missing label NP = Not preserved W = Warm OK =Sample OK **Filled in at lab**	Date Received: ____ / ____ / ______ Received by: ________________ **Filled in at lab**	**Tracking** Marlys Cappaert p) 541-754-4467 **Lab** Kim Medley or John Havel Department of Biology Southwest Missouri State University 901 S. National Av. Springfield, Missouri 65804 USA p) 417-836-5308 f) 417-836-8886 kam797s@smsu.edu (Medley) johnhavel@smsu.edu (Havel)

FAX TO
Tracking: 541-754-4637

8.

Figure 3-8. Sample tracking form for Missouri River zooplankton samples shipped or carried to Southwest Missouri State University.

EMAP-GRE TRACKING - INTERLAB TRANSFERS

AIRBILL NUMBER: ____________________ DATE SENT: ___/___/ 2 0 0 __ TEAM: ________

Draft

Site ID	Date Sample Collected MM/DD/YYYY	Visit	Sample ID	Sample Type	# of Containers	Cond. Code	Comments
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			
				TC SC DM			

FAX TO
Tracking: 541-754-4637

Figure 3-9. Sample tracking form for inter-lab sample transfers. This form is used when shipping samples between laboratories. SC = sediment chemistry subsample shipped from NERL to MED, TC = preserved total organic carbon water sample shipped from UMESC to MED, DM = preserved dissolved metals water sample shipped from UMESC to MED.

INTER-LAB SAMPLE TRANSFER

SC TC DM

GRW04449- ___ ___ ___

________ / ________ / 200__

Sample ID ____________________

Figure 3-10. Label for inter-lab sample transfers. This label is only used when shipping samples between laboratories. SC = sediment chemistry subsample shipped from NERL to MED, TC = preserved total organic carbon water sample shipped from UMESC to MED, DM = preserved dissolved metals water sample shipped from UMESC to MED. Labels for shipping samples from the field are depicted in subsequent sections. Not actual size.

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Section 4
Site Verification

E. William Schweiger[1], Ted R. Angradi[2], and David W. Bolgrien[2]

Site verification is the process by which 1) available background information for potential EMAP-GRE sample sites is evaluated in the office, and 2) sites are visited to determine if a site can and should be sampled. It includes procedures for adjusting sampling locations at each site to avoid safety hazards and to correct for errors in sample location from the design. Parts of this section are adapted from Herlihy (2003).

4.1 The EMAP sample design

EMAP uses an unequal selection probability randomized design to select sample sites (for details, see http://www.epa.gov/nheerl/arm/). EMAP-GRE sample locations were selected from a river-centerline GIS data layer (or "sample-frame") developed from the National Hydrography Database (NHD). Sites were stratified by river, creating three explicit and independent sample designs for the Missouri, upper Mississippi, and Ohio Rivers. Subsets of sites within each strata were partitioned into sections defined by state boundaries (Missouri and upper Mississippi Rivers) or multi-state river reaches (Ohio River) (see Figure 1-1). Strata and sections are important in structuring how sites are replaced (see Section 4.1.1) and during data analysis.

The list of sites generated through the design process is stored in a database called the design file. The design file includes geographic (e.g., state, river section, river bank to sample) and other coding fields identifying the characteristics of each sample location. All sites are referenced with a Site ID number. Each crew will receive a copy of the design file. Additional data fields may be added to a crew's copy of the design file to assist in field operations; however, the structural relationships of the cells in a design file should never be altered (to prevent a Site ID becoming associated with incorrect attribute fields).

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1 National Park Service, 1201 Oakridge Drive, Fort Collins, CO 80525

2 U.S. Environmental Protection Agency, Office of Research and Development, National Health and Environmental Effects Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804

Table 4-1. EMAP-GRE sample sites and site visits by river and section for 2004 and 2005 sample seasons. The total number of site visits is a sum of all initial site visits and revisits within each section. To calculate the number of sites (or site visits) in a *specific* state, sum all values in rows that include the state. For example, Nebraska has sites shared with Missouri (5), South Dakota (10), and Iowa (31), resulting in a total of 46 Nebraska sites (or 56 site visits).

Section	Strata (river)	Section name (reach or river left/river right state)	Primary site N	Total number of site visits	Total number of site visits
1	Ohio	Lower	36	49	114
2		Middle	27	33	
3		Upper	27	32	
4	Upper Mississippi	Illinois/Missouri	30	40	118
5		Illinois/Iowa	23	28	
6		Wisconsin/Iowa	12	17	
7		Wisconsin/Minnesota	22	24	
8		Minnesota/Minnesota	7	9	
9	Missouri	Missouri/Missouri	19	23	160
10		Missouri/Nebraska	5	5	
11		South Dakota/Nebraska	10	15	
12		Montana/Montana	19	22	
13		North Dakota/North Dakota	22	22	
14		Iowa/Nebraska	31	36	
15		Missouri/Kansas	30	37	

4.1.1 Site types and site replacement

Each section has a unique set of primary and oversample sites. All primary sites in a section will eventually be sampled if they are not classified as non-target or unsampleable. If a primary site is classified as non-target or unsampleable during the site verification process it may not be sampled later, even if it becomes sampleable. Oversample sites are used to replace non-target or unsampleable primary sites. Oversample sites are used for site replacement in the order in which they appear in the oversample list within a section in most cases. However, because some sections have a small expected primary sample size, it is possible that oversample replacement sites in these sections may be from another section.

As site verification forms are returned to the data information manager, the list of sites to sample within a section will be updated and returned to each crew.

4.1.2 Panels

Primary sites within each section are divided by year into subsets called panels. Primary sites should be sampled within their designated panel. Oversample sites acquire the panel designation of the primary site they replace.

4.1.3 Site revisits

A subset of sites will be revisited two or three times. Data from these revisits are used to refine the population estimates generated from EMAP sampling (for more details go to http://www.epa.gov/nheerl/arm/). The design file designates which sites are revisited within each strata. Allocation of revisits is across each strata (rivers). Therefore, individual sections within a strata are not guaranteed any revisit sites (e.g., the Missouri/Nebraska section of the Missouri River, Table 4-1). The revisit schedule is a combination of a “4-visit” and “3-visit” approach (Table 4-2) which optimizes the allocation of effort between site-specific estimates and the spatial distribution of revisits. Within each strata, the first four sites in panel 1 are revisited three times (for a total of four site visits). The next six panel 1 sites are revisited two times (for a total of three site visits). The first three of these six sites receive their intra-annual visit in 2004 and their inter-annual visit in 2005. The second three sites are revisited twice in 2005. The first visit to revisit sites should be early in the sampling season (ideally they should be among the first sites visited). Intra-annual revisits should be among the last sites sampled, maximizing the time interval between visits to a site. If an oversample site is used to replace a primary site designated a revisit site, the oversample site acquires the revisit schedule of the original primary site.

Table 4-2. Revisit schedule for each river. "x" denotes a site visit.

Panel 1 primary site order	2004		2005	
	Original visit *(early in index period)*	Intra-annual 1 *(late in index period)*	Inter-annual 1 *(early in index period)*	Inter/Intra-annual 2 *(late in index period)*
1	x	x	x	x
2	x	x	x	x
3	x	x	x	x
4	x	x	x	x
5	x	x	x	not revisited in 2005
6	x	x	x	
7	x	x	x	
8	x	not revisited in 2004	x	x
9	x		x	x
10	x		x	x

4.2 The EMAP-GRE sample site

Sampling at EMAP-GRE sample sites is done at point locations, within plots, or along transects associated with an "X-site" location (latitude and longitude) from the design file (Figures 4-1 and 4-2). Sample stations and their default (pre-adjustment) locations are as follows:

1. **Three main-channel sample locations**. Water samples and plankton are collected in the thalweg and at point locations half the distance from the thalweg to each shoreline along a cross-channel transect oriented perpendicular to the river centerline. The thalweg is defined as the line connecting the deepest points in the main channel.

2. **Two 500 m main-channel shoreline (MCS) transects.** Each MCS transect begins at the intersection of the cross-channel transect and the river right or river left (facing downriver) MCS as designated in the design file. The primary MCS transect extends 500 m upriver from this point. A secondary 500 m MCS transect extends down river from the starting point of the primary transect (Figure 4-1). Fish assemblages and fish habitat are sampled in the near-shore littoral zone along both MCS transects. In addition, large woody

debris, bank, and channel morphology measurements are taken along the primary MCS transect.

3. **Sample stations and plots along the primary MCS transect**. Eleven locations, spaced 50 m apart along the primary MCS transect (Figure 4-2), define sample locations for riparian measurements, macroinvertebrates, sediment and periphyton, bank morphology, and aquatic vegetation. Not all indicators are sampled at all stations.

4. **Other sample locations**. A single macroinvertebrate sample is collected from a snag (large woody debris in the channel) near the X-site. A subjective assessment is made of the reach adjacent to the X-site.

4.3 The site dossier

Site dossiers contain aerial imagery and other site attribute information used in site verification and sampling activities. Dossiers will be provided to crews by EMAP Duluth staff. An example is given in Figure 4-3. Dossiers include approximate (GIS-derived) locations for the X-site, cross-channel transect, and main-channel sample locations, and show the approximate linear extent of the primary and secondary 500-m MCS transects. These data are displayed on aerial imagery and in tables. Point coordinates are given in decimal degrees to facilitate importation into GPS units. Crews may use a different local coordinate projection for navigation, but all data must be reported as decimal degrees in NAD83 datum.

Dossiers contain a typographic error: main-channel locations for water and plankton sampling locations identified as point ID numbers 2 and 3 in the data table in each dossier are described (“detail”) as “1/3" and “2/3" points. These points are actually half the distance between the thalweg and the main-channel shoreline, and their locations are not accurately characterized by a fraction of channel width. Point identified in the dossier as “250 m” and “500 m” points (ID numbers 7 and 8) can be ignored. These locations refer to sample locations relevant for bathymetry procedures that have been dropped from EMAP-GRE.

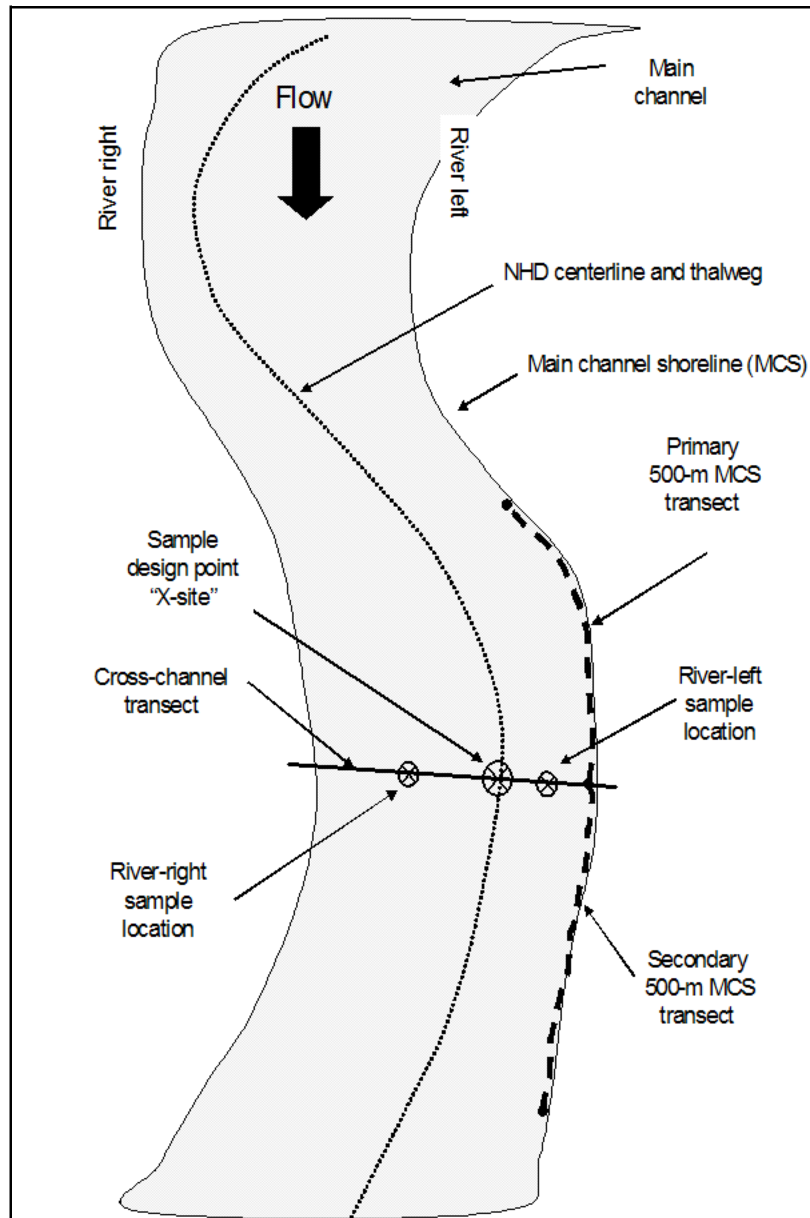


Figure 4-1. Idealized sampling site. In this view, the target shoreline is at river left and the NHD centerline and thalweg are in the same location.

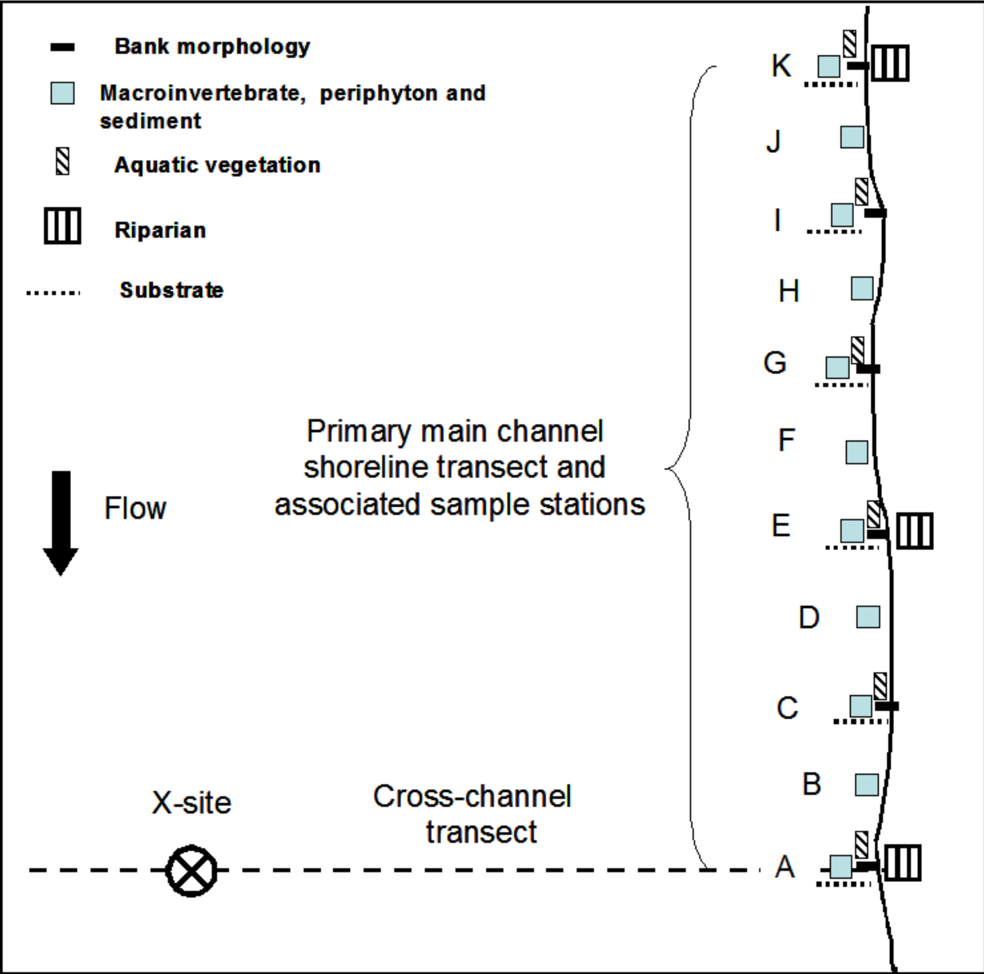


Figure 4-2. Detail of the 500-m long primary main channel shoreline (MCS) transect showing a subset of samples/measurements collected. Shoreline stations are spaced 50 m apart along the river margin.

EMAP-GRE Site Dossier

GRW04449-300

River Thalweg Shoreline Transect
Points Lines: Attribute Information

NAME	NED_ELEV	SECTION	SAMPLE BANK	CLOSEST RIVER MILE	# PLANNED VISITS	POOL/REACH
Mississippi River	165	Illinois Iowa	Left	477	1	Pool 16

DETAIL	TRAN	ID	DESIGN_NO	LON_DD	LAT_DD	OR_AZ	SB_DIST	NSB_DIST	CH_WIDTH
X-Site	X	#01	300	-90.6535	41.4619	--	--	--	--
1/3 Point	X	#02	300	-90.6524	41.4609	--	--	--	--
2/3 Point	X	#03	300	-90.6545	41.4627	--	--	--	--
Cross Channel Transect/MCS Int.	X	#04	300	-90.6512	41.4599	--	--	--	--
Cross Channel Transect/MCS Int.	X	#05	300	-90.6555	41.4636	--	--	--	--
Transect X	X	#06	300	--	--	315.94	295.52	248.98	544.50
250m Site	Y	#07	300	-90.6513	41.4633	--	--	--	--
500m Site	Z	#08	300	-90.6492	41.4650	--	--	--	--
Primary 500m Upstream MCS Transect	X	#09	300	-90.6473	41.4631	--	--	--	--
Secondary 500m Downstream MCS Transect	X	#10	300	-90.6562	41.4575	--	--	--	--

DATA DICTIONARY

DETAIL = Description
TRAN = Transect
ID = ID number from map page 2
DESIGN_NO = Sample number
LON_DD = Longitude in decimal degrees
LAT_DD = Latitude in decimal degrees
OR_AZ = Orthogonal azimuth (degrees clockwise from north)
SB_DIST = Distance to sample bank in meters
NSB_DIST = Distance to non-sample bank in meters
CH_WIDTH = Total width of channel in meters

**All distance values are in meters.
**All azimuth values are in degrees.
**All coordinate values were derived using:
Projection: Geographic
Datum: NAD83
Spheroid: GRS1980
Units: Degrees

Semimajor Axis: 6378137.000000000000000000
Semiminor Axis: 6356752.314140356100000000
Inverse Flattening: 298.257222101000020000

US EPA, Mid-Continent Ecology Division
6201 Congdon Blvd
Duluth, MN 55804

Map layout produced under the FAIR II Contract
68-W-02-032 Task Order 2024

EMAP-GRE Site Dossier

GRW04449-300

Page 1 of 2

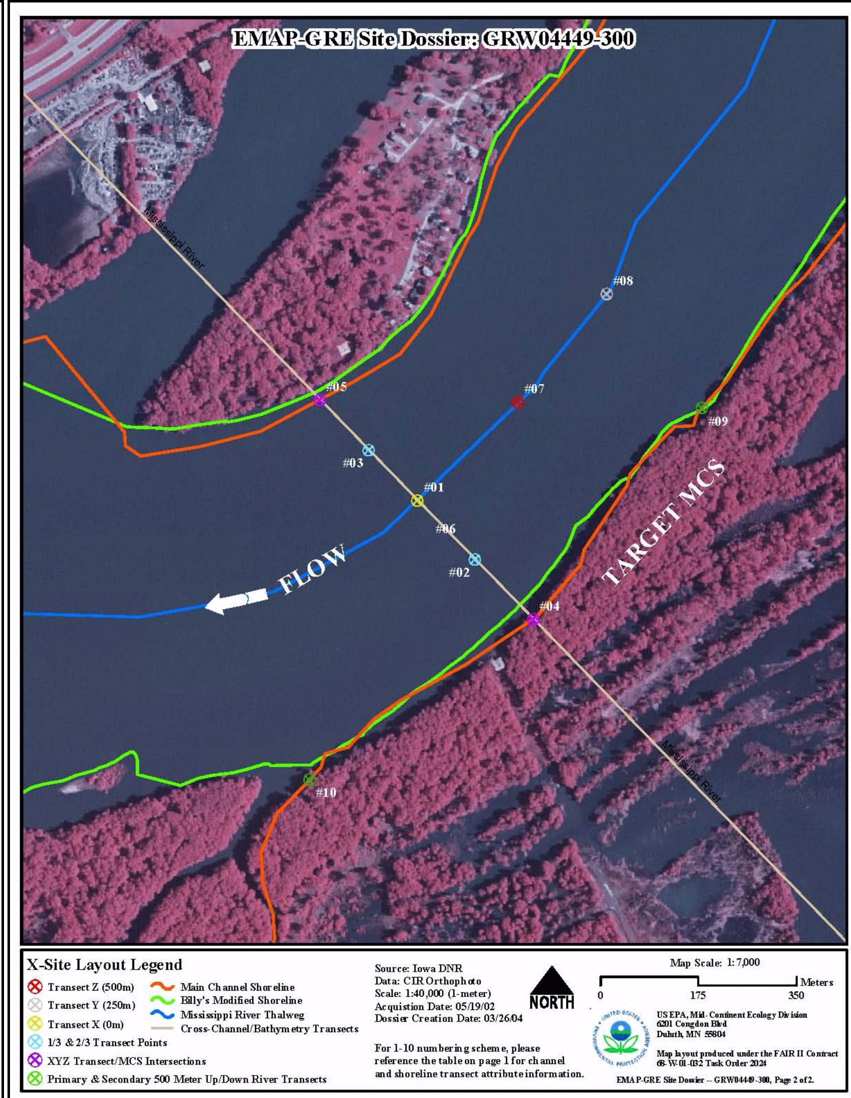


Figure 4-3. Example site dossier (size and resolution are reduced here; each dossier panel is a full 8.5 x 11" sheet and is in high resolution color.

4.4 Site verification

Site verification includes three phases for determining whether a site can and should be sampled (its “sampling status”): 1) office-based verification, 2) field reconnaissance, and 3) field-based verification. Each verification phase has a unique (but similar) data form. The sampling status of a site is determined primarily by two factors: 1) whether or not the X-site falls within one of the Great Rivers of interest, and 2) whether the site can be safely sampled. Site verification results in the assignment of a status code to each site (Table 4-3)

Office-based verification and field reconnaissance occur before the sample season. The completed verification forms must be returned to the EMAP data manager (Table 1-2) at least two weeks prior to the first day of sampling. If a site is determined to be target, but is not sampleable when the crew arrives for sampling, the EMAP-GRE design contact (Table 1-2) should be consulted for guidance.

Site verification (both office and field phases) includes an evaluation of the main-channel sample locations (see Section 4.2) and the MCS transects for position errors and any possible safety hazards. Both office and field verification procedures include rules for adjusting sampling locations when errors in their position or a safety hazard exists. These procedures should be followed as closely as possible to maintain the validity of the EMAP sample design.

Non-target sites (NTS) are sites that fall in an Upper Missouri River reservoir (navigation pools are considered river for EMAP-GRE). These sites will not be sampled. This status may be apparent from office-based review, but given fluctuations in reservoir elevations, field verification for sites in reservoir delta areas may be necessary. Local definitions and professional judgement should be used to define the boundary between the river and the reservoir into which it flows.

Target, not sampleable sites (TNS) are target (non-reservoir) but are unsafe to sample even after all allowable adjustments (described in Section 4.4.4) to sample locations are attempted. Classifying a site as TNS requires the following conditions: 1) If a site cannot be safely electrofished, it is assumed that littoral and riparian sampling is also unsafe and the site is classified as TNS; or 2) If it is unsafe to operate on the cross-channel transect because it is

too close to a safety hazard (e.g., falls, dam, lock) then the site is TNS. If only a portion of the cross-channel transect is unsafe, the sampleability is up to the discretion of the crew leader(s). If electrofishing can be safely conducted, but some littoral or riparian stations appear unsafe, the site should not be classified as TNS and all data that can be safely collected should be collected.

Target, wrong strata (TWS) sites are sites near confluence areas that fall in the wrong river. Target, wrong section (TWE) sites are sites that are in the wrong river section (state or reach; Table 4-1). These sites may be sampled if safe. However, in the analysis phase, data generated from the site will be grouped with their correct strata or section. An oversample site that is in the correct strata or section must also be sampled to meet the primary N requirements for the original strata/section. Crew leaders must determine if they want to sample a TWS or TWE site and a correct strata or section oversample replacement site. TWS and TWE sites should be identifiable through office-based review.

Sites that are not classified as NTS, TNS, TWS, or TWE during office verification are classified as target, office-only sites (TOO) and do not require a separate reconnaissance visit and field reconnaissance form. The final status of TOO sites is determined by field-based verification at the time of sampling. Sites that are not classifiable with office-based verification alone and that appear to be at risk of being NTS or TNS should be classified as unknown (UNK) by office verification. These sites will require a separate field reconnaissance.

Table 4-3. Site status codes.

Code	Meaning	Description	Applicable phase
NTS	Non-target	Non-target because the site falls in a Missouri River mainstem reservoir.	Office-based Field reconnaissance Field-based
TNS	Target, not sampleable	Target but electrofishing and/or water quality sampling not possible due to safety hazards. See Section 4.4.	Office-based Field reconnaissance Field-based
TWS	Target, wrong strata	Target but belongs to another strata. See Section 4.4.1.	Office-based Field reconnaissance Field-based
TWE	Target, wrong section	Target but belongs to another state or reach section. See Section 4.4.1.	Office-based Field reconnaissance Field-based
TOO	Target, office only	Probably target. High degree of certainty allows omission of field reconnaissance.	Office-based
UNK	Unknown	Unknown from office verification alone, requires field reconnaissance.	Office-based
TSA	Target, sampleable	Verified with a field reconnaissance visit.	Field reconnaissance
TSS	Target, successfully sampled	Target and successfully sampled.	Field sampling

4.4.1 Office-based site verification

Office-based site verification relies on information in the site dossier, the crews' experience, and local knowledge. Crews will evaluate and return an office-based site verification form (Figures 4-4 and 4-5) for all primary sites in each section and for a subset of oversample sites (approximately 10% of primary site N within each river strata). Office-based site verification may be omitted for revisits. Procedures are described in detail in Table 4-4.

Although the sample design process will often place the X-site on or near the thalweg (the line connecting points of maximum discharge in the main channel), errors may occur in its position such as when the X-site falls away from the thalweg, outside of the main channel, or on an island or bar. Furthermore, the NHD centerline (used to locate the X-site) may not necessarily be parallel to the azimuth of either the thalweg or the MCS, causing the azimuths used to create the cross-channel transect to be incorrect.

The shorelines used to locate the MCS in the dossiers may also be incorrect due to the resolution of the source data. These situations can lead to incorrect placement in all or some of the sample locations in the dossier. Generally, office-based adjustment of incorrect sample locations should be done in cases when an error in X-site position, cross-channel transect azimuth, or MCS delineation is obvious and the possible solution is apparent in the dossier image. In these cases, sample station locations may be adjusted by hand (e.g., by drawing on a copy of the dossier) or in a GIS (data layers will be provided to crews by EMAP Duluth staff upon request). If adjusted by hand on the dossier, a flag and explanation of these changes must be provided on the office-based site verification form and the copy of the corrected dossier returned with the office-based site verification form. If new values are calculated by a crew in a GIS, these should be reported on the office-based site verification form. Office-based site verification forms do not need to be faxed to Corvallis. They can be mailed or FedExed in with the other field forms.

Table 4-4. Office-based site verification procedures.

1. Fill out the header information on the office-based site verification form (Figure 4-5) including the site ID, the date the form was filled out, the team (see Section 2.3.2), the verifier, and the closest river mile (from the dossier).

2. Transfer the coordinates for the X-site, river left/right sample locations, MCS intersections, and cross-channel transect azimuth from the dossier. Do not record negative numbers for longitudes. Round dossier azimuths to the nearest degree.

3. Using all available data, answer the questions about apparent frame errors or safety hazards. Answers to these questions assist with remaining steps in office-based site verification.

4. Assign a site status code to the site from among the options in Table 4-3. Sites classified as UNK during office-based verification require field reconnaissance prior to sampling. Use flag/comments to explain status assignments if necessary. Hazards include (but are not limited to) falls, rapids, high and low dams, locks, and barge fleeting areas. A site is considered target, not sampleable (TNS) if it appears that electrofishing cannot be safely conducted or if the main-channel sample locations on the cross-channel transect cannot be safely sampled after all allowable adjustments have been made (see steps 6-11). If only a subset of littoral or riparian sampling is prevented by unsafe conditions, the site is still considered sampleable.

5. If the site is classified as NTS or TNS, explain the reasons in the comments. In this case, site verification is complete; otherwise go to step 6.

6. Following the rules in Section 4.4.4, determine if a lateral shift or longitudinal slide is required for either the primary and/or secondary MCS transect to avoid a safety hazard. During office-based review, this should be done only as the quality of the dossier data allows. Shifts or slides may only be apparent after going through steps 8 - 12.

7. Longitudinal slides only apply to MCS transects. The X-site and cross-channel transect may not be slid up- or downriver.

8. On the provided imagery, examine the X-site, the river right/left points, the cross-channel transect, and the shoreline intersection points. River right and river left are defined looking downriver. If the location of the NHD centerline at the X-site, and the GIS-derived azimuth of the cross-section transect and the left and right banks of the MCS appear valid, the intersection of the channel cross-sections with the left and right bank should be approximately at the correct position. A "valid" cross-channel transect azimuth will appear to be approximately perpendicular to the general orientation of the thalweg (deepest point in the main channel) and/or the MCS. A "valid" MCS will be reasonably close in position and orientation to the main-channel shoreline on the imagery. See Section 4.4.4 for further details.

Continued

Table 4-4. Office-based site verification procedures, continued.

9. If any of the locations from step 8 are suspect, a new cross-channel transect azimuth and associated sample station locations should be drawn in by hand on the dossier (and/or in a GIS). Suspect situations include an X-site that falls on an in-channel terrestrial feature or outside of the bankfull channel, or the X-site on the NHD centerline appears to not fall in the thalweg. In all of these cases, mark on the dossier image an adjusted position of the X-site – the "thalweg sample location" in the thalweg closest to the original X-site along the cross-channel transect. An approximate thalweg location can often be detected in imagery provided in the dossier (e.g., the thalweg is generally in the navigation channel which may be apparent and is usually close to outside bend shorelines). Estimate by eye the azimuth of the thalweg or MCS. Draw by hand (and/or in a GIS) a new cross-channel transect line perpendicular to the thalweg or MCS and through the X-site (or the substitute X-site). Estimate (or calculate in a GIS) its new azimuth and record on the form.

10. The MCS transects should be evaluated and adjusted if necessary. The MCS on the imagery can often be determined by examining the lateral positions of the most obvious or prominent bank line. Draw a new MCS on the dossier maps (and/or in a GIS). With the thalweg location as a reference, extend, if necessary, the cross-channel transect laterally to the new MCS. Place a point where the cross-channel transect intersects the MCS by hand and/or in a GIS.

11. Examine the position of the river-right and river-left sampling locations on the cross-channel transect (Figure 4-1). If they appear to be in the wrong location or unsampleable (e.g., on land) they should be adjusted by moving them to the closest suitable position along the cross-channel transect. The reasons for any adjustments should be recorded in the comment or explanation sections of the office-based site verification form.

12. If any of the above adjustments are made in a GIS, record the new sample site coordinates on the office-based site verification form. If adjustments were made by hand, note this on the form and include the marked copy of the dossier in the mailing to the EMAP data manager.

13. Record the name and location information for the boat ramp nearest the X-site.

14. (Optional) Repeat step 11 for a secondary ramp in case the primary ramp is unavailable or unsuitable.

15. Use all available resources (e.g., consulting local resource contacts, maps, aerial photos) to identify and describe any safety hazards associated with accessing the site.

16. Record contact information for any sources used to complete the office-based site verification form.

4.4.2 Field reconnaissance

Sites classified as unknown (UNK) during office-based verification require a field reconnaissance visit prior to sampling. For sites that receive a reconnaissance visit, a completed field reconnaissance form (Figure 4-6) must be returned to the data manager. Table 4-5 presents field reconnaissance procedures in detail. The primary purpose of field reconnaissance is to establish the status of the site. However, to do this effectively, sample locations may need to be visited, and the crew may elect to locate and flag the MCS transects during the field reconnaissance visit.

4.4.3 Field-based site verification

Field-based site verification activities are largely analogous to office-based verification but are conducted at the site. Table 4-5 presents field-based site verification procedures in detail. Field-based site verification forms (Figures 4-7 and 4-8) are filled out for all site visits (including revisits), regardless of whether the site is sampled or not. These forms are returned with all other data forms from a sampling event.

4.4.4 Identifying the MCS and making adjustments in sample locations in the field

Identifying the MCS is a necessary step in establishing the set of sample locations at each EMAP site. Once the MCS is identified, the sampleability (based on safety) of the 500-m primary and secondary main-channel shoreline transects must be evaluated. To reduce bias in sample location, all adjustments in the MCS transect, either laterally (shift) or longitudinally (slide), must adhere to the rules below. Figure 4-4 presents a flow chart for identifying the MCS, associated sample locations, and any adjustments that may be required.

The MCS is defined as the interface between the main channel (the channel with the most discharge) and landward terrestrial habitat. It is usually the most obvious and closest shoreline that borders the main channel. By definition, it is a terrestrial shoreline that an electrofishing boat can safely navigate. In relatively straight, un-braided, single-channel reaches, the MCS will likely follow a high bank that bounds bankfull channel flow. This situation is also likely in reaches that are extensively modified (e.g., riprapped) and along outside bends where the main channel is in direct contact with a high bank that separates floodplain terraces from the active

channel. In complex reaches (deltaic, braided, or with at least two significant channels), or where a new floodplain has formed below an abandoned pre-regulation floodplain (as has occurred along the upper Missouri River), the MCS will not necessarily follow the high bank. In these cases, the MCS is the interface between the main channel and terrestrial habitat that is navigable for electrofishing and is closest to the position of the X-site moving laterally along the cross-channel transect. The following rules must be followed when the MCS is shifted laterally:

1. If the target shoreline habitat adjacent to the X-site is an apparent island that is obviously less than 2 km in length (estimated in the field or from the dossier), the shoreline of the bar or island is probably not the MCS. The MCS will instead likely fall on the next navigable shoreline landward along the line of the cross-channel transect.

2. If there is no navigable channel (> 5 m wide; > 2 m maximum depth) along the MCS landward of the apparent island, the MCS is located on the island regardless of its size.

3. If the location of the thalweg is unclear (e.g., discharge in two channels appears equal), and there are no other clues to the location of the thalweg (e.g., location of the navigation channel) then the MCS transect is established on the shoreline nearest to the original X-site location if the above rules are not violated. If the nearest shoreline is not navigable, the MCS transect is established on the next closest navigable shoreline.

4. Primary and secondary MCS transects are shifted together. For example, if the primary MCS transect must shift landward to get off an island < 2 km in length, the secondary MCS would also shift laterally to the same shoreline.

5. All 500 m of the transect is shifted together so that part of the 500 m transect is not on one shoreline and the rest on another shoreline.

6. The opposite shoreline may not be used to replace the target MCS specified in the dossier.

Once the MCS is identified, the sampleability of the primary MCS transect (the 500 m upriver from the intersection of the cross-channel transect and the MCS; see Figure 4-1) must be evaluated. If a tributary or unconnected secondary channel (backwater or slack water) greater than 5 m wide merges with the main channel or if there are less than 500 m of safely sampleable MCS habitat along this reach, the entire set of MCS sampling stations should be "slid" up- or downriver along the MCS a distance that places a sufficient buffer between sampling stations and the safety hazard or tributary/secondary channel. For example, if a navigation lock is 200 m upriver from the X-site or cross-channel transect, the 500-m primary MCS transect can be slid approximately 400 m downriver from the X-site or cross channel transect so that a 100-m buffer exists between the end of the transect and the lock. The degree of hazard presented and the length of the buffer needed is up to the crew leaders (explain decisions in comments on the field-based verification form). If necessary, all 500 m of the primary and secondary MCS transects may be slid. If a connected secondary channel (running water but not a tributary) greater than 5 m wide at the mouth and with a maximum depth ≥ 2 m is encountered along the MCS and it is safely navigable, the MCS transect will follow this shoreline into the secondary channel. Tributaries and secondary channels less than 5 m wide at their mouth are ignored when locating the MCS transect. Several rules apply when sliding the MCS transect:

1. The MCS transects must not be slid more than 500 m up- or downriver so that the both the up and downriver end of the primary MCS transect are > 500 m from intersection of the cross-channel transect and the shoreline.

2. The opposite shoreline may not be used to replace the target MCS specified in the dossier.

3. Safety or access issues that only apply to riparian plots are not grounds for sliding the MCS transect. Unsafe or inaccessible riparian plots are not sampled (see Section 7).

4. If an up- or down-river slide in one of the transects will cause the primary and secondary transect to overlap, the non-slid transect must be slid in conjunction with the adjusted MCS transect. For example, if the downriver end of the primary MCS transect is slid 100 m downriver, the upriver end of the secondary MCS transect is also slid downriver 100 m so that they will not overlap.

5. If a safety hazard or tributary or unconnected secondary channel > 5 m wide at the mouth is encountered along the secondary MCS transect, the reach may be slid up- or down-river to avoid the tributary or channel (following all the rules above). The upriver end of the secondary transect must be < 500 m from the downriver end of the primary 500 m MCS transect after sliding.

6. If the primary or secondary transect must be slid up- or down-river but the other transect cannot be slid an equal distance because of a safety hazard, then the transects can be slid in opposite directions (following all the rules above).

7. If the transects cannot be slid in opposite directions to avoid a hazard or tributary, the secondary transect should be truncated in favor of the primary MCS transect (but is still sampled to the degree possible). Flag any data not collected as missing and explain the reasons.

There are many possible combinations of safety hazards and adjustments that may be encountered in the field, not all of which are treated here. The operational goal of crew leaders should be to conduct as much of the complete sampling suite as is possible without subjecting the crew to unacceptable risk. Flag and explain any non-standard methods used or sampling decisions made in the field. Do the best you can and consult the authors of this section for additional guidance in special cases that do not seem to fit the circumstances described herein.

Table 4-5. Field reconnaissance and field-based site verification procedures.

1.	Fill out the header information on the appropriate form (field reconnaissance form or field-based verification form) including site ID, the date the form was filled out, the team (see Section 2.3.2), the verifier, and the closest river mile.
2.	Transfer the original (dossier-based) or adjusted (from office verification) X-site coordinates, cross-channel transect azimuth, and MCS intersections from the office-based site verification form or field reconnaissance form. Do not record negative numbers for longitudes.
3.	Using all available data, answer the questions about apparent frame errors or safety hazards (or confirm the answers from the office-based site verification form/field reconnaissance form).
4.	Assign a site status code to the site from among the options in Table 4-3. Sites classified as UNK during office-based verification require field reconnaissance prior to sampling. Use flag/comments to explain status assignments if necessary. Hazards include (but are not limited to) falls, rapids, high and low dams, locks, and barge fleeting areas. A site is considered target, not sampleable (TNS) if it appears that electrofishing cannot be safely conducted <u>or</u> if the main channel sample locations on the cross-channel transect cannot be safely sampled after all allowable adjustments are made (steps 5-12 below). If only a subset of littoral or riparian sampling is prevented by unsafe conditions, the site is still considered sampleable. If the site is classified as NTS or TNS, explain the reasons in the comments and site verification is complete; otherwise move on to step 5.
5.	Following the rules in Section 4.4.4 determine if a lateral shift or longitudinal slide is required for either the primary or secondary MCS. The need for shifts or slides may only be apparent after going through steps 8-12.
6.	Longitudinal slides only apply to MCS transects. The X-site and cross-channel transect may not be slid up- or down-river.
7.	Navigate to the X-site using coordinates from the dossier, office-based site verification or field reconnaissance forms. It is strongly recommended that the relevant site coordinates (i.e., points 1-6, 9 and 10 from the dossier) be pre-loaded into the boat's GPS unit.
8.	If the location of the X-site is incorrect (e.g., not in the thalweg, on an island or the floodplain), a new location should be established in the field. Locate the position of the estimated thalweg closest to the X-site along the cross channel transect. Using the boat's GPS, acquire the coordinates of the actual thalweg sample location and record them on the field reconnaissance or field-based site verification form.

Continued

Table 4-5. Field reconnaissance or field-based site verification procedures, continued.

9.	Verify the azimuth of the thalweg and/or the general direction of the MCS while stationed on the original X-site (if not moved in step 6) or generate a new one if the thalweg sample location is not the same as the X-site. Maintain the boat position as close as possible to the X-site or its substitute and back-sight along the direction of thalweg flow with a bearing compass. If the safest or most convenient location is on the MCS, back-sight along the shoreline upriver. Generate the approximate azimuth of the new cross-channel transect by calculating an azimuth perpendicular (offset 90 degrees) to the thalweg/MCS orientation. Record this azimuth on the field reconnaissance or verification form. If the original X-site is actually in the thalweg and the field-calculated cross channel transect azimuth is within 5 degrees of the original cross-channel transect azimuth, the original dossier azimuth value should be used.
10.	While stationed on X-site or at the thalweg sample location (if they are not the same point), visually extend the cross-channel transect laterally to the suspected MCS by sighting along the bearing compass using the azimuth from the dossier/office-based site verification form or the new value calculated in step 9. The intersection of the cross-channel transect and the MCS should be identified using a fixed feature on the shoreline that is closest to the intersection.
11.	Navigate to the river-right and river-left sample locations on the cross-channel transect. If they appear to be in the wrong location (not half the distance from the shore to the thalweg) or are unsampleable, they should be adjusted by moving them to the closest sampleable location along the cross-channel transect. Record the final GPS location for each site on the form. Be sure to note with a flag any adjustment made in the sample locations.
12.	Navigate towards the MCS using the original or adjusted azimuth (from step 9). Determine if the MCS as identified in the dossier or through office-based site verification is correct. See Section 4.4.4 for a description of the MCS. If the MCS needs to be adjusted, locate the correct MCS along the original or corrected azimuth. Verify or acquire new coordinates for the intersection of the cross-channel transect and the MCS. Record these on the form.
13.	Conditions apparently preventing complete and safe sampling of a subset of indicators do not cause a site to be classified as unsampleable. All data that can be safely collected should be collected from a given site.
14.	All adjustments in sampling stations should be explained in the comment or explanation sections of the site verification form.
15.	Locate and flag the primary and secondary MCS transects (Section 4.5) and acquire the GPS coordinates for the up- and down-river end of each MCS transect.
16.	Make a sketch of the site (Figure 4-10), including any hazards, or frame error corrections. Take digital photographs of the site (additional photos should be taken opportunistically during sampling).
17.	Record crew personnel.

4.5 Locating and flagging the 500 m MCS transects

Because the position of one MCS can effect the other MCS, the two sampling crews should communicate to make sure all adjustments are mutually understood. The first sampling crew (fish or river) at the site locates and flags the primary and secondary 500-m MCS transects after the intersection between the cross-channel transect and the MCS has been verified and flagged using all the rules detailed above. If both crews arrive at the site at the same time, crew leaders should decide which crew is responsible for flagging the stations. The simplest method is to use the trip odometer on a GPS unit to determine distance along the shoreline from the boat. A heavy steel washer with a flag streamer can be thrown from the boat onto the bank at the station location to avoid landing the boat. Other methods may be used and should be available as a backup if GPS reception is poor. For the purposes of EMAP-GRE, dikes, wing dams, and other man-made structures that project into the channel are treated as fish habitat at the site but are not treated as extensions of the river's shoreline. The contour of the MCS cuts across the base of these structures. During site layout, stations on the primary and secondary transect are flagged at 100-m intervals to 500 m. The intermediate littoral stations on the primary MCS transect are spaced 50 m apart and are established during littoral sampling.

4.6 Determining sampleability during the sampling visit

Even after site verification has indicated that a site is target and sampleable, conditions encountered at the time of sampling may make sampling the site unsafe or impossible. The most likely situations include high flows, high turbidity, barge activity, or gear failure. The decision whether to sample on a given day is up to the crew leader(s). If the decision is made not to sample, the crew should schedule another visit for the site and perhaps attempt to sample an alternate target site. All reasons for aborting the site sampling attempt should be noted on the Field-based site verification form Any data forms that are completed should be returned (with appropriate flags) to the data manager. The field-based site verification form is also filled out during site revisits.

4.7 Site photographs

Site photographs should be taken at all sites using a digital camera (Table 4-6). In addition to the recommended scenes, photos may also be taken of any stressors or other unusual features at or around the site. Image file names (or the series) should be recorded on the field-based verification form (Figure 4-10). All photos should be downloaded into a folder named with the Site ID and backed up. The image files should be delivered to the EMAP data center on a disk after the sample season.

Table 4-6. Recommended site photographs.

Image scene	Comments
X-site and MCS	If possible, include a placard with site name and sampling date.
At cross-channel transect - MCS intersection: along MCS looking upriver	Taken at the shoreline
At cross-channel transect - MCS intersection: along MCS looking downriver	Taken at the shoreline
At cross-channel transect - MCS intersection: riverward	Taken at the shoreline
On riparian bank at bank station A looking landward	Taken on the riparian bank
On riparian bank at bank station E looking landward	Taken on the riparian bank
On riparian bank at bank station K looking landward	On riparian bank
Other 1 - 4	Stressors, notable features

4.8 Equipment and supplies

A list of the equipment and supplies required to conduct site verification and to lay out the sampling reach is presented in Table 4-7. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

Table 4-7. Equipment and supplies for site verification.

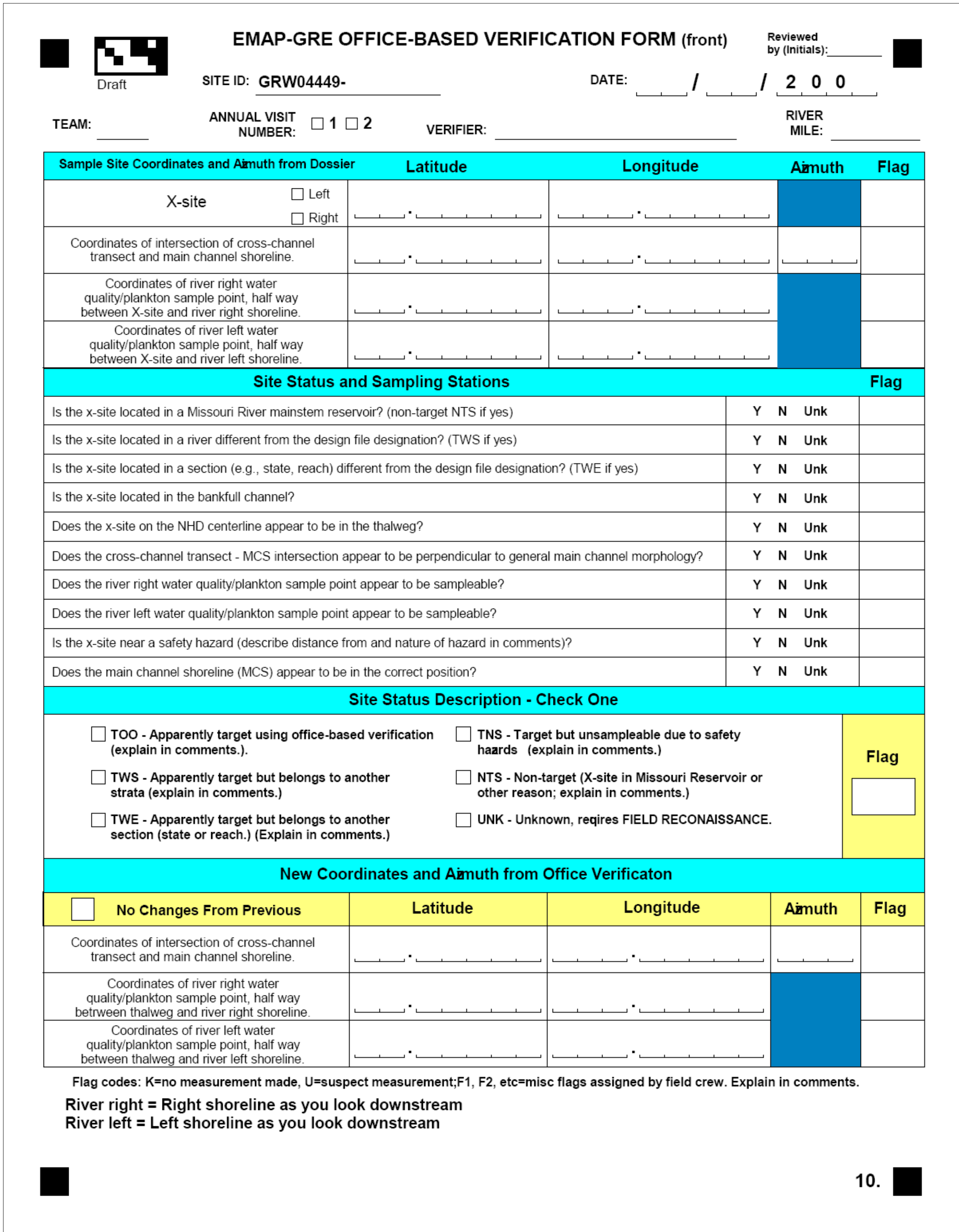
Qty	Item	
1	Dossier of site and safety/access information	
1	Gazetteers, topographic maps, and/or other navigation aids	
1 set	Office-based, field reconnaissance, and field-based site verification forms	
2 rolls	Biodegradable flagging, 2 colors (one for 100-m MCS intervals; one for 50-m intervals)	
1	Laser range finder ($\geq$1000 m range) (optional)	
1	Digital camera and extra memory cards	

4.9 Literature cited

EPA EMAP Design Web Site. http://www.epa.gov/nheerl/arm.

Peck, D. V., Averill, D. K., Herlihy, A. T., Hughes, R. M., Kaufmann, P. R., Klemm, D. J., Lazorchak, J. M., McCormick, F. H., Peterson, S. A., Cappaert, M. R., Magee, T. and Monaco, P. A. Unpublished draft. Environmental Monitoring and Assessment Program - Surface Waters Western Pilot Study: Field Operations Manual for Non-Wadeable Rivers and Streams, U.S. Environmental Protection Agency, Washington, DC.

Peck, D. V., Herlihy, A. T., Hill, B. H., Hughes, R. M., Kaufmann, P. R., Klemm, D. J., Lazorchak, J. M., McCormick, F. H., Peterson, S. A., Ringold, P. L., Magee, T. and Cappaert, M. R. Unpublished draft. Environmental Monitoring and Assessment Program - Surface Waters Western Pilot Study: Field Operations Manual for Wadeable Streams, U.S. Environmental Protection Agency, Office of Research and Development, Washington, DC.



Draft

EMAP-GRE OFFICE-BASED VERIFICATION FORM (front)

Reviewed by (Initials): ________

SITE ID: GRW04449-

DATE: ___ / ___ / 2 0 0 _

TEAM: ______ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 VERIFIER: ____________ RIVER MILE: ______

Sample Site Coordinates and Azimuth from Dossier	Latitude	Longitude	Azimuth	Flag
X-site ☐ Left ☐ Right	___ . ______	___ . ______		
Coordinates of intersection of cross-channel transect and main channel shoreline.	___ . ______	___ . ______	___	
Coordinates of river right water quality/plankton sample point, half way between X-site and river right shoreline.	___ . ______	___ . ______		
Coordinates of river left water quality/plankton sample point, half way between X-site and river left shoreline.	___ . ______	___ . ______		

Site Status and Sampling Stations		Flag
Is the x-site located in a Missouri River mainstem reservoir? (non-target NTS if yes)	Y N Unk	
Is the x-site located in a river different from the design file designation? (TWS if yes)	Y N Unk	
Is the x-site located in a section (e.g., state, reach) different from the design file designation? (TWE if yes)	Y N Unk	
Is the x-site located in the bankfull channel?	Y N Unk	
Does the x-site on the NHD centerline appear to be in the thalweg?	Y N Unk	
Does the cross-channel transect - MCS intersection appear to be perpendicular to general main channel morphology?	Y N Unk	
Does the river right water quality/plankton sample point appear to be sampleable?	Y N Unk	
Does the river left water quality/plankton sample point appear to be sampleable?	Y N Unk	
Is the x-site near a safety hazard (describe distance from and nature of hazard in comments)?	Y N Unk	
Does the main channel shoreline (MCS) appear to be in the correct position?	Y N Unk	

Site Status Description - Check One

- ☐ TOO - Apparently target using office-based verification (explain in comments.).
- ☐ TWS - Apparently target but belongs to another strata (explain in comments.)
- ☐ TWE - Apparently target but belongs to another section (state or reach.) (Explain in comments.)
- ☐ TNS - Target but unsampleable due to safety haards (explain in comments.)
- ☐ NTS - Non-target (X-site in Missouri Reservoir or other reason; explain in comments.)
- ☐ UNK - Unknown, reqires FIELD RECONAISSANCE.

Flag: ____

New Coordinates and Azimuth from Office Verificaton

☐ No Changes From Previous	Latitude	Longitude	Azimuth	Flag
Coordinates of intersection of cross-channel transect and main channel shoreline.	___ . ______	___ . ______	___	
Coordinates of river right water quality/plankton sample point, half way betrween thalweg and river right shoreline.	___ . ______	___ . ______		
Coordinates of river left water quality/plankton sample point, half way between thalweg and river left shoreline.	___ . ______	___ . ______		

Flag codes: K=no measurement made, U=suspect measurement;F1, F2, etc=misc flags assigned by field crew. Explain in comments.

River right = Right shoreline as you look downstream
River left = Left shoreline as you look downstream

10.

Figure 4-4. Office-based site verification form (front).

Draft

EMAP-GRE OFFICE-BASED VERIFICATION FORM (back)

Reviewed by (Initials):________

SITE ID: GRW04449-________ DATE: ___ / ___ / 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2

Location Information	Primary Boat Ramp	Secondary Boat Ramp
Ramp Name		
Ramp Shoreline	☐ River Right ☐ River Left	☐ River Right ☐ River Left
Location from x-site	☐ Upriver ☐ Downriver	☐ Upriver ☐ Downriver
Distance from x-site (miles)		
Nearest town		

Driving directions to primary boat ramp

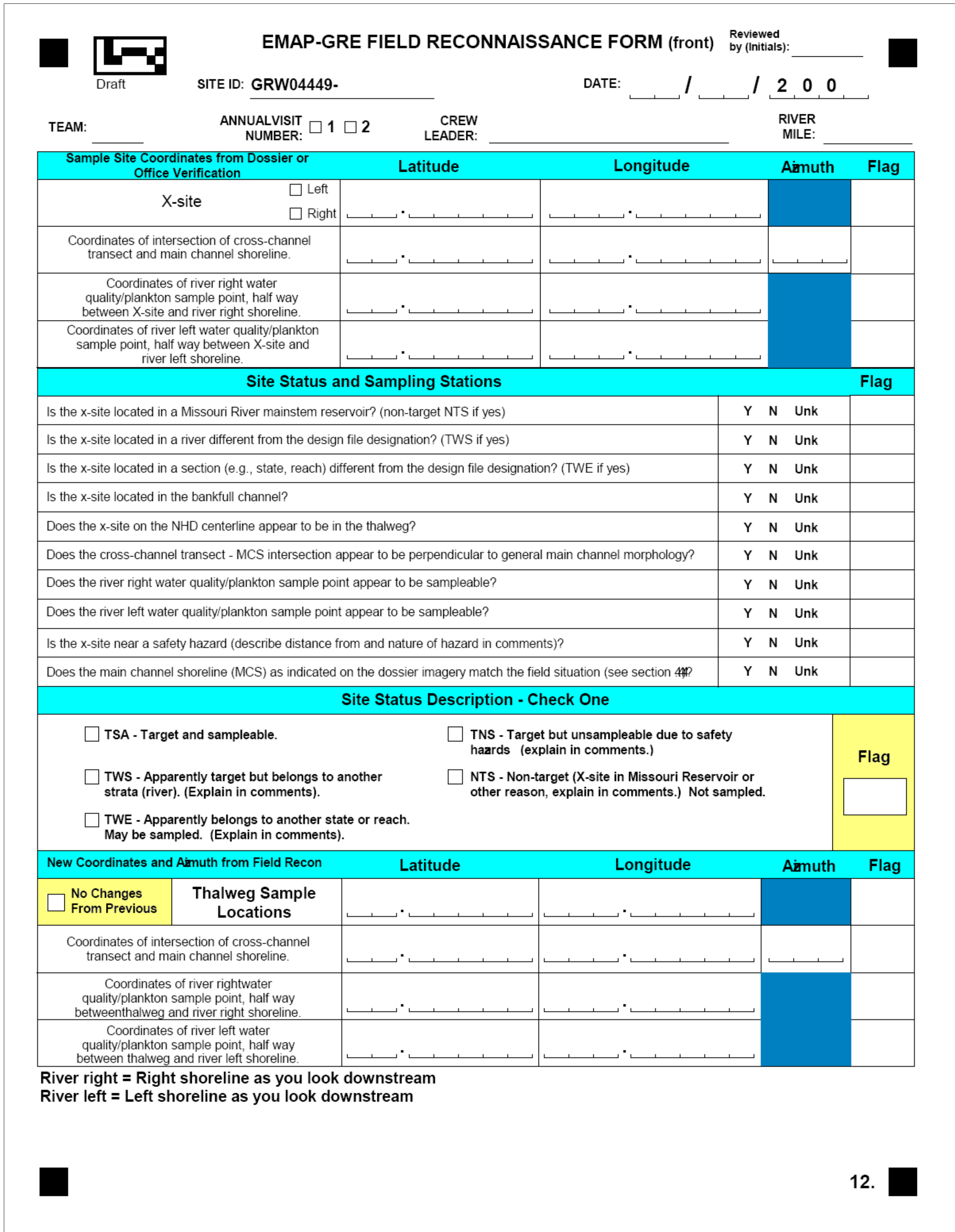
Information Sources - Names, Affiliations, Phone Numbers

Flag	Comments (Description of MCS position, safety hazards, etc.)

Flag codes: K=no measurement made, U=suspect measurement;F1, F2, etc=misc flags assigned by field crew. Explain in comments.

11.

Figure 4-5. Office-based site verification form (back).



Draft

EMAP-GRE FIELD RECONNAISSANCE FORM (front) Reviewed by (Initials): ________

SITE ID: GRW04449- DATE: ___ / ___ / 2 0 0

TEAM: ______ ANNUALVISIT NUMBER: ☐ 1 ☐ 2 CREW LEADER: ________ RIVER MILE: ______

Sample Site Coordinates from Dossier or Office Verification	Latitude	Longitude	Azimuth	Flag
X-site ☐ Left ☐ Right	.	.		
Coordinates of intersection of cross-channel transect and main channel shoreline.	.	.		
Coordinates of river right water quality/plankton sample point, half way between X-site and river right shoreline.	.	.		
Coordinates of river left water quality/plankton sample point, half way between X-site and river left shoreline.	.	.		

Site Status and Sampling Stations		Flag
Is the x-site located in a Missouri River mainstem reservoir? (non-target NTS if yes)	Y N Unk	
Is the x-site located in a river different from the design file designation? (TWS if yes)	Y N Unk	
Is the x-site located in a section (e.g., state, reach) different from the design file designation? (TWE if yes)	Y N Unk	
Is the x-site located in the bankfull channel?	Y N Unk	
Does the x-site on the NHD centerline appear to be in the thalweg?	Y N Unk	
Does the cross-channel transect - MCS intersection appear to be perpendicular to general main channel morphology?	Y N Unk	
Does the river right water quality/plankton sample point appear to be sampleable?	Y N Unk	
Does the river left water quality/plankton sample point appear to be sampleable?	Y N Unk	
Is the x-site near a safety hazard (describe distance from and nature of hazard in comments)?	Y N Unk	
Does the main channel shoreline (MCS) as indicated on the dossier imagery match the field situation (see section 4.4)?	Y N Unk	

Site Status Description - Check One

☐ **TSA - Target and sampleable.**

☐ **TWS - Apparently target but belongs to another strata (river). (Explain in comments).**

☐ **TWE - Apparently belongs to another state or reach. May be sampled. (Explain in comments).**

☐ **TNS - Target but unsampleable due to safety hazards (explain in comments.)**

☐ **NTS - Non-target (X-site in Missouri Reservoir or other reason, explain in comments.) Not sampled.**

Flag

New Coordinates and Azimuth from Field Recon		Latitude	Longitude	Azimuth	Flag
☐ No Changes From Previous	Thalweg Sample Locations	.	.		
Coordinates of intersection of cross-channel transect and main channel shoreline.		.	.		
Coordinates of river rightwater quality/plankton sample point, half way betweenthalweg and river right shoreline.		.	.		
Coordinates of river left water quality/plankton sample point, half way between thalweg and river left shoreline.		.	.		

River right = Right shoreline as you look downstream
River left = Left shoreline as you look downstream

12.

Figure 4-6. Field-based reconnaissance form (front).

Draft

EMAP-GRE FIELD RECONNAISSANCE FORM (back)

Reviewed by (Initials):________

SITE ID: GRW04449- ________ DATE: __ / __ / 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2

Sketch of X-site (indicate direction of flow)

Flag	Comments

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

13.

Figure 4-7. Field-based reconnaissance form (back).

Draft

EMAP-GRE FIELD-BASED VERIFICATION FORM (front)

Reviewed by (Initials): ______

SITE ID: GRW04449-______ DATE: ___ / ___ / 2 0 0 _

TEAM: ______ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 CREW LEADER: ______ RIVER MILE: ______

Sample Site Coordinates from Dossier/Office Verification/Field Recon	Latitude	Longitude	Aimuth	Flag
X-site ☐ Left ☐ Right	___ . _______	___ . _______		
Coordinates of intersection of cross-channel transect and main channel shoreline.	___ . _______	___ . _______	____	
Coordinates of river right water quality/plankton sample point, half way between X-site and river right shoreline.	___ . _______	___ . _______		
Coordinates of river left water quality/plankton sample point, half way between X-site and river left shoreline.	___ . _______	___ . _______		

Site Status and Sampling Stations		Flag
Is the x-site located in a Missouri River mainstem reservoir? (non-target NTS if yes)	Y N Unk	
Is the x-site located in a river different from the design file designation? (TWS if yes)	Y N Unk	
Is the x-site located in a section (e.g., state, reach) different from the design file designation? (TWE if yes)	Y N Unk	
Is the x-site located in the bankfull channel?	Y N Unk	
Does the x-site on the NHD centerline appear to be in the thalweg?	Y N Unk	
Does the cross-channel transect X appear to be perpendicular to general main channel morphology?	Y N Unk	
Does the river right water quality/plankton sample point appear to be sampleable?	Y N Unk	
Does the river left water quality/plankton sample point appear to be sampleable?	Y N Unk	
Is the x-site near a safety hazard (describe distance from and nature of hazard in comments)?	Y N Unk	
Does the main channel shoreline (MCS) as indicated on the dossier imagery match the field situation (see section 4.4.4)?	Y N Unk	

Site Status Description - Check One

☐ **TSS - Target and sampled.**

☐ **TWS - Target but belongs to another strata. May be sampled (explain in comments.)**

☐ **TWE - Target but belongs to another state or reach. May be sampled (explain in comments.)**

☐ **TNS - Target but unsampleable due to safety haards (explain in comments.)**

☐ **NTS - Non-target (X-site in Missouri Reservoir or other reason, explain in comments.) Not sampled.**

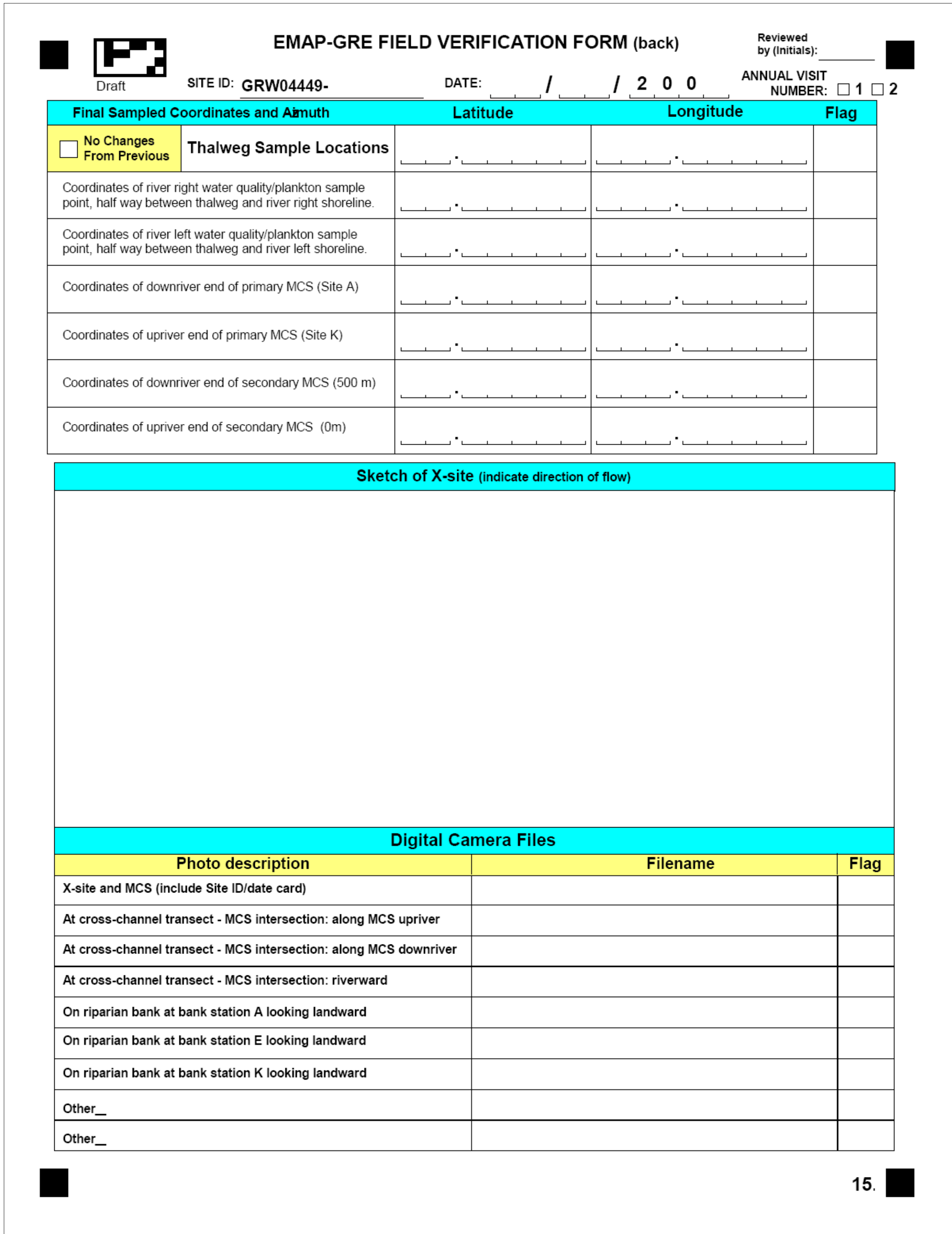
Flag ______

Flag	Comments (Description of MCS position, safety haards, etc.)

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

14.

Figure 4-8. Field-based site verification form (front).



Draft

EMAP-GRE FIELD VERIFICATION FORM (back)

Reviewed by (Initials): ______

SITE ID: GRW04449- DATE: ___ / ___ / 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2

Final Sampled Coordinates and Aimuth	Latitude	Longitude	Flag
☐ No Changes From Previous — Thalweg Sample Locations	___ . _____	___ . _____	
Coordinates of river right water quality/plankton sample point, half way between thalweg and river right shoreline.	___ . _____	___ . _____	
Coordinates of river left water quality/plankton sample point, half way between thalweg and river left shoreline.	___ . _____	___ . _____	
Coordinates of downriver end of primary MCS (Site A)	___ . _____	___ . _____	
Coordinates of upriver end of primary MCS (Site K)	___ . _____	___ . _____	
Coordinates of downriver end of secondary MCS (500 m)	___ . _____	___ . _____	
Coordinates of upriver end of secondary MCS (0m)	___ . _____	___ . _____	

Sketch of X-site (indicate direction of flow)

Digital Camera Files

Photo description	Filename	Flag
X-site and MCS (include Site ID/date card)		
At cross-channel transect - MCS intersection: along MCS upriver		
At cross-channel transect - MCS intersection: along MCS downriver		
At cross-channel transect - MCS intersection: riverward		
On riparian bank at bank station A looking landward		
On riparian bank at bank station E looking landward		
On riparian bank at bank station K looking landward		
Other__		
Other__		

15.

Figure 4-9. Field-based site verification form (back).

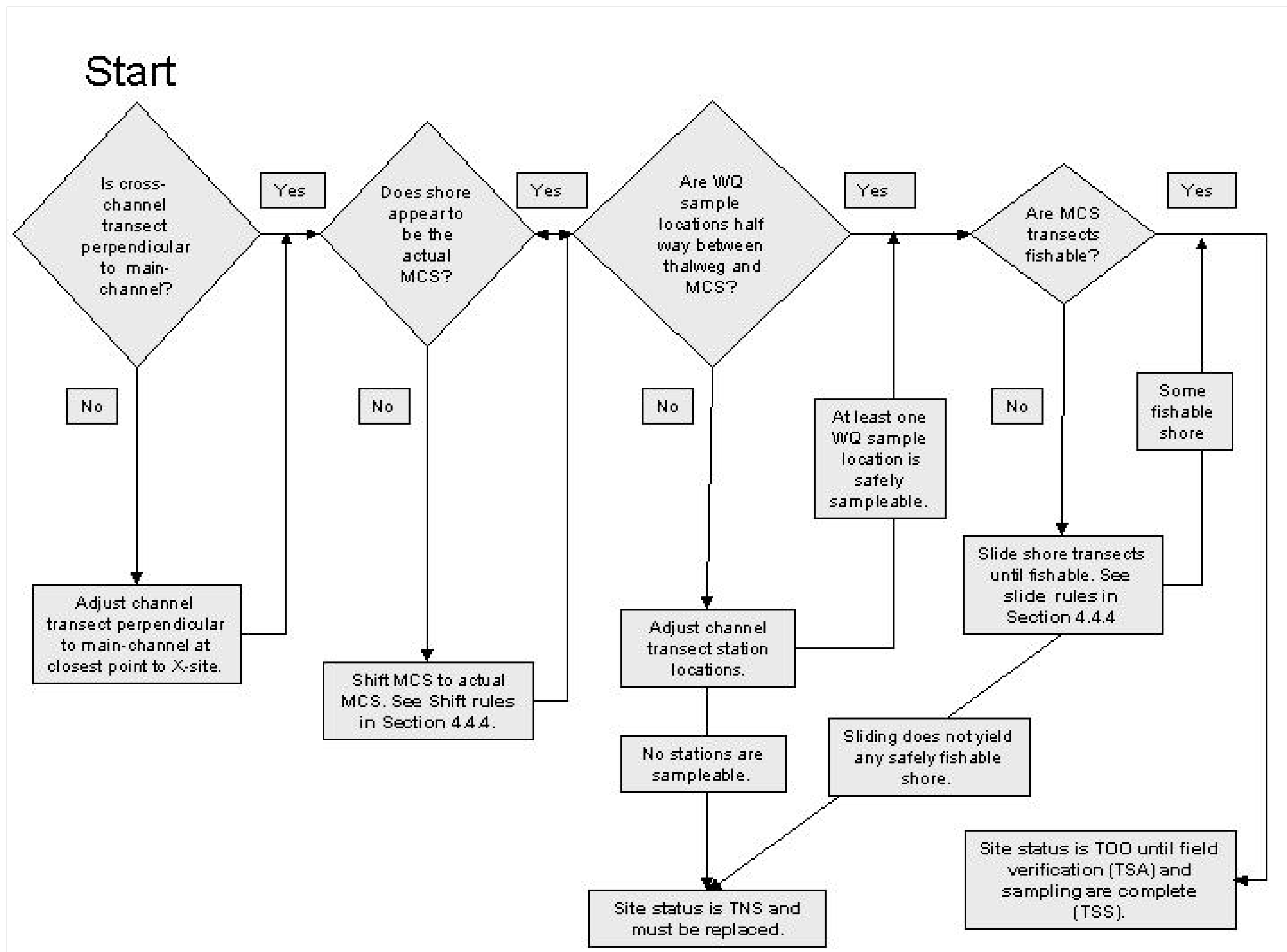


Figure 4-10. Flow chart for adjusting layout of sites known to be target. MCS = main channel shoreline. Other acronyms defined in Table 4.3.

Section 5
Water Chemistry and Plankton

Terri M. Jicha[1], Ted R. Angradi[1], and Brian H. Hill[1]

In EMAP-GRE, water chemistry data will be used to define reference conditions and to identify stressor gradients. Great River stressors associated with water chemistry may include nutrient enrichment, inorganic contamination, hypoxia, temperature stress, turbidity, and suspended sediment. Water chemistry sampling includes depth-integrated water samples for laboratory analysis and depth-integrated in-situ measurements, including dissolved oxygen, conductivity, and pH. At the base location, turbidity is measured, and chlorophyll, total suspended solids (TSS), and geochemical marker samples are collected from subsamples of a composite water sample.

Plankton includes algae (phytoplankton) and microinvertebrates (zooplankton) suspended in the water column. Zooplankton are potentially useful indicators of environmental condition. They are important to the food web of large rivers because they link primary producers (algae) to larger invertebrates, and to fish and other vertebrates (Baker et al. 1997). Plankton assemblage structure, body size distribution, and trophic structure are likely sensitive to a number of anthropogenic disturbances, including flow regulation, habitat alteration (including floodplain disconnection), invasive species, and contamination by nutrients, metals, and herbicides.

Water chemistry and plankton sampling are combined in this section of the manual because the activities are conducted together at the same sampling locations in the channel and the data are recorded on the same forms. Water samples are collected from a transect across the main channel rather than from littoral areas, because the emphasis is on an unbiased and representative composite sample from the site rather than on relating water chemistry to the biota at a site. Aspects of this section are adapted from Peck et al. (Unpublished drafts).

5.1 Water samples

Water sampling is conducted by the river-sampling crew. The coordinates of the sample

1 U.S. Environmental Protection Agency, Office of Research and Development, National Health and Environmental Effects Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804

locations are specified in the site dossier (Section 4). The three sample locations are in the thalweg, and half the distance from the thalweg sampling location to each shoreline along the cross-channel transect (Figure 4-1). The water sample consists of two depth-integrated 4-L cubitainer samples (composite water samples 1 and 2) and one 500-mL grab sample. A 2-L subsample of composite water sample 1 and 2, combined will be shipped to the laboratory for determination of major cations and anions, nutrients, organic carbon, and dissolved metals. The 500-mL sample is collected as a single grab at the thalweg sample location and is used to determine total alkalinity. Subsamples from composite water sample 1 and 2, combined (the portion not shipped) will be used to measure chlorophyll *a*, geochemical markers, TSS, and turbidity.

The 2-L composite water sample and the 500-mL grab sample are packed on ice in coolers and shipped as soon as possible (Section 3.2.2) to the laboratory for analysis. The rest of the composite water sample is processed at the base location. Procedures for collecting water samples are described in Table 5-1. Subsample volumes for depth integrated composite samples are given in Table 5-2.

Whenever possible, schedule site visits to occur in a downriver to upriver direction to avoid re-sampling the same parcel of water. This is especially important when sampling a group of adjacent sites over a short period of time.

5.2 Water-quality measurements

5.2.1 Dissolved oxygen, conductivity, pH, and temperature

In-situ measurements for DO, conductivity, pH, and temperature are made at each subsample depth at each of the three sample locations at the site. The DO and pH meters are calibrated daily at the base location or the sample site. Calibration requirements are described in Section 3.

5.2.2 Water clarity and turbidity

Water transparency will be estimated using a Secchi disk at each sample location (Table 5-3). Turbidity will be measured using a turbidimeter at the base location or mobile lab. Turbidity readings in NTUs are made for a subsample from composite water sample 1 and 2, combined (Table 5-4).

5.2.3 Chlorophyll *a*, TSS, and geochemical markers

Chlorophyll *a*, TSS, and geochemical marker samples for laboratory analysis will be collected by filtering a subsample from composite water sample 1 and 2 (combined) at the base location or mobile lab. Geochemical markers include percent organic matter, percent nitrogen, and stable isotopes ($\delta^{13}C$, $\delta^{15}N$). The chlorophyll *a* filters must be kept refrigerated and dark until frozen. Geochemical marker and TSS filters are dried before shipment. Procedures for filtering water are described in Table 5-4.

5.3 Collection of plankton samples

Depth-integrated composite phytoplankton and zooplankton samples are collected in the main channel at the water sampling locations. Phytoplankton samples are collected as a ≈2-L composite of pumped river water. Macrozooplankton samples are collected by pouring about 180 L of pumped river water through a plankton net with 63-µm mesh. Microzooplankton are collected by filtering 18 L of pumped river water through 20-µm mesh. Microzooplankton samples are not prefiltered through the macrozooplankton net. Table 5-5 describes plankton sampling procedures in detail. Composite plankton samples are preserved with formalin (phytoplankton) or a formalin-sugar solution (zooplankton) and stored until they can be transported or shipped to the laboratory.

Table 5-1. Procedure for collecting water chemistry samples.

1. Fill in the site ID and date on the front and back of the water chemistry and plankton form (Figure 5-1 and 5-2).

2. Calibrate the DO meter (if the meter has not already been calibrated at the base location), record calibration details in the log book. Indicate on the water chemistry and plankton form (Figure 5-1) whether or not the DO and pH meter have been calibrated on the day of sampling.

3. Using a fine-point waterproof marker (e.g., Sharpie), fill out a pre-printed sample label (Figure 5-3) for one 4-L cubitainer and one 500-mL bottle (these containers will be shipped to the lab). On each label, circle the sample type (WCC = water chemistry composite in cubitainer; ALK = 500-mL bottle), affix the labels to the cubitainer and bottle, and cover them with clear tape. Record the sample ID numbers (from the labels) on the forms (water sample ID for the 4-L cubitainer; alkalinity sample ID for the 500-mL grab). On a second 4-L cubitainer, write the site ID and date directly on the cubitainer with a waterproof marker (this container is not shipped). Containers may also be labeled at the base location prior to departing for the sample site. If the site has been identified as a QA/QC (Section 5.4) site for water chemistry, additional containers will be needed for field blanks and duplicate samples.

4. Attach the sounding weight to the winch cable. Attach the end of the hose from the peristaltic pump (Tygon size 24) and the Guzzler pump (garden hose with check valve on the end) and the DO/conductivity and pH probe(s) (or datasonde) to the cable above the sounding weight (Figure 5-4).

5. Using the coordinates provided in the site dossier, navigate to the first sampling location located halfway between the thalweg sample location and one of the shorelines (river-right or river-left).

6. At the sample location, anchor if possible. Otherwise, the driver should hold the boat on the sample location facing upriver. Remove the lids from both of the 4-L amber cubitainers and pull them open. One cubitainer (label covered with tape) will hold composite water sample 1; the other cubitainer (site ID written on side) will hold composite water sample 2. Do not blow into the cubitainers to inflate them; this will contaminate the samples. Sun block, insect repellent, etc., will contaminate the sample so avoid touching the inside of the cubitainer or cap.

7. From the sonar unit, determine and record the depth under the boat on the form. If the depth is > 2 m, a subsample will be collected at 0.5 m off the bottom, at mid-depth and at 0.5 m from the surface (each subsample is about 445 mL (Table 5-2): 4L composite volume/three sample sites/three subsample depths). If the depth is < 2 m and > 1 m, subsamples will be collected only at 0.5 m off the bottom and 0.5 m below the surface (each subsample is about 665 mL). If the depth is < 1 m, the entire 1300-mL subsample for the location is collected at mid-depth. The pumps may lack sufficient lifting power beyond about 6 m. If a sample location on the cross-channel transect cannot be sampled, the subsample volume for each remaining location should be adjusted so that the total sample volume for the site is maintained.

Continued

Table 5-1. Procedure for collecting water chemistry samples, continued.

8.	Lower the hose/sensor cluster to the deepest subsample depth using the depth dial on the winch, by counting marks on the cable, or by allowing the sounding weight to contact the bottom and then raising it to the proper depth. Adjust the depth to account for the distance between the bottom of the weight and the sensors. Turn on the peristaltic pump and the DO/conductivity/pH meter(s). Make sure the sounding weight is not bouncing along the bottom during sampling. Use plastic cable ties to secure hoses and the sensor cable(s) to the winch line as the weight is lowered. Pump overboard more water than the entire length of the peristaltic pump hose can hold (determined beforehand). Once the hose is refreshed, pump about 100 mL into each cubitainer to rinse it out. Repeat this two more times making sure rinse water comes in contact with all interior surfaces.
9.	Pump the first subsample (of 445, 665, or 1300 mL depending on the number of subsample depths) into cubitainer 1 and cap it. Use graduations on the cubitainer to estimate when the subsample volume is attained. Pump a second subsample of equal volume into cubitainer 2 and cap it (total of 888 mL per depth at each sample location). Record sample depth, DO (mg/L) and conductivity (µS/cm), temperature and pH following procedures in the instruments operating manuals. Record the actual sample depth (from the surface). Flag depths (K flag) for which no instrument readings are made.
10.	Collect phytoplankton and zooplankton subsamples using the procedures described in Table 5-5.
11.	Repeat steps 6-9, as appropriate, for the other subsample depths at the sample location. Record the total depth or sample depth each sample location on the form. Estimate Secchi depth (Table 5-3).
12.	Repeat steps 6-10 for the two other water quality sample locations at the site. By sliding the cable ties down the winch line as the line is winched up, it should be possible to move between stations without having to disassemble the sampling apparatus. At the thalweg sample location only, collect a 500-mL grab sample for alkalinity. Rinse the bottle and cap three times with river water (discard downriver). Take the sample by filling and capping the bottle under water at arms-length; cap the bottle underwater with no head space. Alternatively, fill a clean bucket with river water from the surface at the thalweg location and take the sample by filling and capping the bottle under water.
13.	After all the water and plankton has been collected, the boat can be beached at the first shoreline sample station for sample processing. Place the water samples in a cooler with ice.

5.4 QA considerations for water chemistry and plankton sampling

Instrument calibration and maintenance, avoiding contamination, and proper container labeling and tracking are all essential for maintaining high QA standards for water chemistry sampling. Table 5.6 describes some specific QA considerations for water chemistry sampling.

At the second site sampled and at the 11th and 21st site sampled by a crew in a season, field duplicates and field DI blanks are required. Field duplicates and blanks are processed and tracked using the same procedures as for regular water samples.

5.5 Equipment and supplies

A list of the equipment and supplies required for collecting water and plankton is presented in Table 5-7. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

Table 5-2. Subsample volumes for depth-integrated composite water samples. Volumes are totals for each depth for each of three sample locations on the cross-channel transect. If a sample location on the cross-channel transect cannot be sampled, the subsample volume for each remaining location should be adjusted so that the total sample volume for the site is maintained. This table should be reproduced and affixed to the top of the peristaltic pump or other convenient location for reference in the field.

Sample type (Total volume per site)	Total depth at sample location		
	> 2 m (3 depths)[1]	< 2 m and > 1 m (2 depths)[2]	< 1 m (1 depth)[3]
Water chemistry (8L)	888 mL	1333 mL	2667 mL
Phytoplankton (1935 mL)	215 mL	322 mL	645 mL
Macrozooplankton (63-µm mesh) (180 L)	20 L	30 L	60 L
Microzooplankton (20-µm mesh) (18 L)	2 L	3 L	6 L

[1] Collect a subsample 0.5 m above the bottom, at mid-depth and 0.5 m below the surface.
[2] Collect a subsample 0.5 m above the bottom and 0.5 m below the surface.
[3] Collect a subsample at mid-depth.

Table 5-3. Procedure for determining Secchi depth (after Strobel and Heitmuller 2001).

1. Remove sunglasses. If the water is relatively clear, lower the Secchi disk over the shaded side of the boat until it disappears. Slowly raise the disk until it becomes visible and record the depth indicated on the line. (interpolate to the nearest 10 cm). If the disappearance depth is < 0.5 m, retrieve the disk and go to step 2.

2. Use the “Secchi on a stick” and read the disappearance depth from the scale on the stick to the nearest cm. In current, it may work best to estimate Secchi depth while drifting.

3. If the disk is visible resting on the river bottom, mark the appropriate box on the form.

Table 5-4. Base-location procedures for turbidity measurements and for subsampling and filtering a water sample for chlorophyll *a*, TSS, and geochemical markers (based, in part, on personal communication with Anthony Aufdencampe, Stroud Water Research Center, Avondale, PA).

1. Shake composite water samples 1 and 2 vigorously and completely pour both composites into an 8-L plastic churn-splitter. Use a subsample dispensed from the churn to rinse the cubitainers into the churn. Churn for five strokes with the dasher touching the bottom of the churn on each stroke. Dispense subsamples while continuing to churn.

2. Dispense 2000 mL from the churn back into the labeled 4-L cubitainer that originally held composite water sample 1. Cap the cubitainer and return it to the cooler with ice. This is the sample that will be shipped to the lab for analysis.

3. **Turbidity**. Dispense about 75 mL of composite from the churn into a beaker for turbidity analysis. Follow the operating instructions for the turbidimeter and make three replicate readings (three subsamples from the 75 mL of composite). Record the values in NTUs on the water chemistry and plankton form. Record the temperature of the sample. Clean sample tubes as required in the turbidimeter operating instructions.

4. Set up the filtering apparatus by connecting a vacuum pump to the filter reservoir.

5. **Chlorophyll *a*.** Filter chlorophyll in shade or subdued light. Place the filter holder on the reservoir and position a 47-mm Whatman GF/F glass-fiber filter on the manifold (not pre-ashed or pre-weighed). Handle filters with forceps. The top of the filter is the side opposite the "checked" or "gridded" side (i.e., filters are placed "grid to grid" on the manifold). Wetting the manifold screen with DI water will allow the filter to adhere better. Secure the top of the apparatus.

6. Dispense (while churning) between 50 to 250 mL at a time using a graduated cylinder. If the water appears clear, dispense 250 mL at a time; if the water appears turbid or green, dispense 50 to 100 mL at a time. Turn on the vacuum pump (or start pumping with a hand pump). Try not to exceed a vacuum of 7 psi (15 inches of Hg) during filtration. Pour the entire contents of the graduated cylinder into the filter funnel each time. Continue dispensing until the filters begins to clog and filtration slows. Keep track of the volume dispensed (to the nearest mL). It is important to filter as much water as possible, in order to collect enough sample for analysis. The total volume filtered can range from 50 mL to 1500 mL, depending on turbidity. If the filters clog completely before all the sample in the filter funnel has been filtered, discard the sample and filter and prepare a new sample with a smaller volume of water.

7. Record the final volume filtered to the nearest mL on the form and sample label (there is no filter ID for chlorophyll filters). Remove the filter with forceps and place it in a foil-wrapped scintillation vial. The filter may be folded in half.

8. **Geochemical markers**. Place the filter holder on the reservoir and position a pre-weighed and pre-combusted (450° C, 4-6 h) 47-mm Whatman GF/F glass-fiber filter on the manifold. Handle filters with forceps The top of the filter is the side opposite the "checked" or "gridded" side (i.e., filters are placed "grid to grid" on the manifold). Record the filter ID number from the filter container on the water chemistry and plankton form. Wetting the manifold screen with DI water will allow the filter to adhere better. Secure the top of the apparatus.

Continued

Table 5-4. Procedure for filtering a water sample for chlorophyll *a*, TSS, and geochemical markers, continued.

9.	Dispense (while churning) between 50 to 250 mL at a time using a 100 or 250 mL graduated cylinder. If the water appears clear, dispense 250 mL at a time; if the water appears turbid or green, dispense 50 to 100 mL at a time. Turn on the vacuum pump (or start pumping with a hand pump). Try not to exceed a vacuum of 7 psi (15 inches of Hg) during filtration. Pour the entire contents of the graduated cylinder into the filter funnel each time. Continue dispensing until the filters begins to clog and filtration slows. Keep track of the volume dispensed (to the nearest mL). It is important to filter as much water as possible, in order to collect enough sample for analysis. The total volume filtered can range from 50 mL to 1500 mL, depending on turbidity. If the filters clog completely before all the sample in the filter funnel has been filtered, discard the sample and filters and prepare a new sample with a smaller volume of water.
10.	Record the filter ID and final volume filtered to the nearest mL on the form and the EPA sample label (transfer the filter ID from the small label on top). Remove the filter funnel with the vacuum still on. Turn the vacuum off and carefully peel the filter off the manifold screen and return the filter to its container (dirty side up) using blunt forceps. If the filter tears, start over.
11.	**Total suspended solids (TSS)**. Place the filter holder on the reservoir and position a pair of pre-weighed 47-mm membrane filters on the manifold (they will be labeled and prepackaged as a pair). Handle filters with forceps and make sure the top filter from the filter holder is on top in the manifold. Wetting the manifold screen with DI water will allow the filters to adhere better.
12.	Record the filter pair ID number on the water chemistry and plankton form. Secure the top of the apparatus.
13.	Dispense (while churning) between 50 to 250 mL at a time using a 100 or 250 mL graduated cylinder. If the water appears clear, dispense 250 mL at a time; if the water appears turbid or green, dispense 50 to 100 mL at a time. Turn on the vacuum pump (or start pumping with a hand pump). Try not to exceed a vacuum of 7 psi (15 inches of Hg) during filtration. Pour the entire contents of the graduated cylinder into the filter funnel each time. Continue dispensing until the filters begins to clog and filtration slows. Keep track of the volume dispensed (to the nearest mL). It is important to filter as much water as possible, in order to collect enough sample for analysis. The total volume filtered can range from 50 mL to 1500 mL, depending on turbidity (membrane filters have a 10-20% lower capacity than GFF filters). If the filters clog completely before all the sample in the filter funnel has been filtered, discard the sample and filters and prepare a new sample with a smaller volume of water.
14.	Record the filter ID and the final volume filtered to the nearest mL on the form and on the EPA sample label (transfer the filter ID from the small label on top). Remove the filter funnel with the vacuum still on. Turn the vacuum off and carefully peel the filter off the manifold screen and return the filter to its container (dirty side up) using blunt forceps. If the filter tears, start over.
15.	Repeat step 11-14 for a second TSS filter pair. The total volume filtered should be about the same as for the first TSS sample.
16.	Rinse the filter apparatus, churn, and graduated cylinder with DI water.

Table 5-4. Procedure for filtering a water sample for chlorophyll *a*, TSS, and geochemical markers, continued.

17.	Complete filling out sample labels (Figure 5-3) for each filter or filter pair (TSS). Place a label on the chlorophyll vial and cover with tape. Place the labels on the bottom of the“Petrislide” containers (TSS and geochemical marker filters). Be sure they do not cover the lid or obscure other information. Keep chlorophyll filters cold (near 4° C) until they can be frozen.
18.	Chlorophyll filters are preserved by freezing. Geochemistry and TSS filters are preserved by drying in an oven. Dry filters overnight in a drying oven at 30 - 50° C with the Petrislide container upright and the lids ajar (the Petrislide containers will melt at >70° C). Drape a sheet of aluminum foil over the filters to keep out dust. If a drying oven is not available, filters will usually dry in 24-48 h in an air-conditioned room by setting the filters out on a table with lids ajar and a sheet of aluminum foil draped on top to keep out dust. Store dried filters in a dry location prior to shipment. Do not ship filters that have not been dried.

Table 5-5. Procedures for collecting plankton samples. Plankton sample collection is integrated with water sample collection (Table 5-1).

1. Navigate to the first sampling location half the distance between the thalweg and one of the shorelines.

2. **Phytoplankton.** Follow steps 1-8 in Table 5-1 (Procedure for collecting water chemistry samples). After the water subsample has been collected, continue pumping water at that depth for a phytoplankton sample. Pump a subsample into a graduated cylinder. Total sample volume at each location will be 650 mL (1,950 L total sample volume/three sample locations – total volume is less than 2 L to allow room for preservative). Subsample volume at each depth will be ≈215, 322, or 645 mL (Table 5-2), depending on the number of depths at the sample location (e.g., 322 mL ≈ 1,935 mL total composite/three sample locations/two depths). Pour the subsample into a 2-L bottle and cap the bottle (this same bottle will hold the entire composite phytoplankton sample for all depths and all three sample locations).

3. **Microzooplankton**. After the phytoplankton sample has been collected, continue pumping at that depth into the graduated cylinder for a microzooplankton sample. Total microzooplankton sample volume for each location is 6 L, so if there are three depths to be sampled, the subsample volume will be 2 L; for two depths the subsample volume will be 3 L; and for 1 depth the subsample volume will be 6 L. Pour the subsample from the graduated cylinder into a PVC microzooplankton filter with 20-µm mesh. Turn off the peristaltic pump. Alternatively, the Guzzler pump (see step 4) can be used to collect the microzooplankton composite.

4. **Macrozooplankton**. Pump the Guzzler pump to prime it. Pump over-board for about 15 seconds to refresh the hose contents. Total sample volume for each sample location is 60 L, so if there are three depths, the subsample volume will be 20 L for a macrozooplankton sample; for two depths the subsample volume will be 30 L; and for 1 depth the subsample volume will be 60 L Table 5-2. Pump into a graduated plastic bucket and pour the bucket contents through the plankton net (63-µm mesh) 10 L at a time.

5. Repeat steps 2-4 for each depth at the sample location. Be sure to pump water overboard before collecting plankton samples to refresh the hose contents at each new depth. Keep track of the volume of water pumped for zooplankton.

6. Using a minimum amount of filtered (20-µm mesh) river water from a wash bottle, rinse the contents of the microzooplankton filter into a 100-mL bottle (use a funnel) and cap the bottle (this same bottle will hold the entire composite microzooplankton sample).

7. Wash the (macro)plankton net contents down to the cod end using the on-board washdown pump. Release the pinchcock and discharge the contents of the cod end into a 250-mL sample bottle. Using a minimum amount of filtered river water from a wash bottle, rinse the cod end into the sample bottle and cap the bottle (this same bottle will hold the entire composite macrozooplankton sample).

8. Repeat steps 2-7 at each sample location. By sliding the cable ties down the winch line as the line is winched up, it should be possible to move between stations without having to disassemble the sampling apparatus. Be sure to pump water overboard before collecting the sample to refresh the hose contents at the new location/depth.

Continued

Table 5-5. Procedures for collecting plankton samples, continued.

9.	Using a small beaker, add 80 mL of borax-buffered 100% formalin to the 1,950 mL phytoplankton composite (4% formalin final concentration). Use gloves and safety glasses when handling formalin.
10.	Using filtered (20-µm mesh) river water, raise the level in the macrozooplankton (250 mL) and microzooplankton (100 mL) sample bottles to 2/3 full. Put half of an Alka-Seltzer® tablet in each bottle and let it dissolve. Top off each bottle with the chilled 12% borax-buffered formalin-sugar solution to achieve a final preservative strength of 4% formalin. Record the total volume filtered for macrozooplankton and for microzooplankton on the water chemistry and plankton form.
11.	Prepare labels (Figure 5-3) for outside the jars (if not pre-labeled). Fill in the site number, enter the sample date, and composite sample volume (phytoplankton) or the volume filtered (zooplankton) and site visit number. Place the labels on the jars and cover them with clear tape. Record the sample ID and other data on the water chemistry and plankton form.
12.	Seal each capped jar with plastic electrician's tape. Store the preserved samples upright in a container to await transport or shipment to the laboratory (see Table 3-7).

Table 5-6. QA considerations for water chemistry and plankton sampling.

- The laboratory water quality analyses are very sensitive, and attention to detail is needed to avoid sample contamination. Possible sources of contamination include substances on sampler's hands, boat exhaust, and sediment from the river bed.

- Be vigilant about refreshing the hose contents at each new depth or sample location.

- Cap the composite water samples (4-L cubitainers) between subsamples to avoid atmospheric contamination.

- Keep the 12% formalin-sugar solution (for preserving zooplankton) on ice in the boat.

- Whenever possible, schedule site visits to occur in a downriver to upriver direction to avoid re-sampling the same parcel of water. This is especially important when sampling a group of adjacent sites over a short period of time.

- Calibrate the dissolved oxygen meter before or during every site visit.

- Avoid contacting the river bed with the sounding weight while collecting water and plankton.

- Avoid sending the sounding weight down through or adjacent to snags. If snags are observed or suspected under the boat, move several meters away to sample and flag the samples.

- If possible, the same crew member should estimate Secchi depth at each sample location and site.

- Using a survey pole or other gage, occasionally confirm the accuracy of the boat's sonar depth readings.

- Always pour the entire 4-L grab samples (water chemistry composites 1 and 2) into the churn for subsampling. Rinse the cubitainers into the churn with an aliquot from the churn.

- Make certain that the TSS and geochemical marker filters are thoroughly dried before shipping them to the lab.

Table 5-7. Equipment and supplies for water quality and plankton sampling. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

Qty	Item	
1	Winch with calibrated cable or depth dial and 30, 50, or 100 lb sounding weight depending on water velocity	
1	Masterflex 115V peristaltic pump or equivalent with 10 m of size 24 Tygon hose (Fisher Scientific 13-310-490 or equivalent)	
1	Guzzler hand pump (Model 400h with aluminum epoxy coated clamp ring, 3/4" mail garden hose fittings and 74MGH check valve on submerged hose end) www.thebosworthco.com. Mount the pump on a board large enough to step on with 2 feet.	
1	3/4" flexible garden hoses (10 m section with female fittings both ends, 2 m section with 1 female fitting)	
1	Modified sounding weight hanger or other apparatus for attaching sensors and hoses to winch line (See Figure 5-4)	
25	Plastic cable-ties long enough to secure hoses and sensor cables to the winch line or cable	
1 pr	Side cutters for removing cable ties	
1	Nephelometric turbidimeter (used at the base location)	
1 set	Turbidity standards (e.g., 1, 10, 100, 250 NTU)	
4	4-L amber pre-DI-washed cubitainer for water samples. Only two are needed per site; extras should be carried in case of contamination (Fisher Scientific 11-375-115B or equivalent).	
1	500-mL Nalgene bottle for pH and alkalinity subsample	
1	DO/Conductivity meter with 7.5-m cable and manufacturers manuals	
1	pH meter with 7.5-m cable and manuals	
1	200-mm Secchi disk with calibrated chain or line (0.5-m increments)	
1	200-mm Secchi disk affixed to a calibrated stick (0.01-m increments)	
1	Plankton tow net with 63-µm mesh net, drain hose and pinchcock (Wildco 426-A28 or equivalent)	
1	250-mL plastic graduated cylinder for filtering samples	
1	100-mL plastic graduated cylinder for filtering samples	
1	Graduated plastic bucket	
1	20-µm mesh plankton filter (sieve or home made from PVC pipe, coupler and Nitex). Put a bead of silicone caulk around inside edge of pipe where it meets the Nitex.	
1	Square HDPE sample jars, 250-mL capacity (for macrozooplankton samples) (Fisher Scientific 03-311-3D or equivalent)	
1	Square HDPE sample jars, 100-mL capacity (for microzooplankton samples) (Fisher Scientific 03-311-3C or equivalent)	
1	Square HDPE bottle, 2-L capacity (for phytoplankton samples) (Fisher Scientific 03-311-3G or equivalent)	
1	Vacuum pump	
1	8-L churn sample splitter (Wildco 1831-C80 or equivalent)	

Continued

Table 5-7. Equipment and supplies for water quality and plankton sampling, continued.

Qty	Item	
1	Lab thermometer for turbidity samples	
1	Chlorophyll filter apparatus (including tubing) (Fisher Scientific 09-740-23E or equivalent)	
1	Whatman GF/F filters (47 mm) for chlorophyll analysis (Fisher Scientific 09-874-71 or equivalent)	
1	Pre-weighed and pre-combusted (450° C, 4-6 h) Whatman GF/F filter (47 mm) for geochemical markers in a Millipore Petrislide container **(Provided by lab)**.	
1	Pre-weighed membrane filter pair (47 mm, 0.45 µm) for suspended sediment in a Millipore petrislide container **(Provided by lab)**.	
1	Scintillation vials for chlorophyll filters (Fisher Scientific 03-337-14 or equivalent)	
1 box	Aluminum foil to wrap chlorophyll vials	
1	Small funnel for transferring plankton samples from nets into jars	
1	Wash bottle	
2 pr	Powder free lab gloves	
1 pr	Filter forceps	
2	100-mL plastic beakers for turbidity procedures and for adding formalin to phytoplankton samples (Fisher Scientific 02-591-27 or equivalent)	
1	Water chemistry and plankton form	
1 set	Sample labels	
1 L	Borax-buffered formalin (100%) for preserving phytoplankton	
1	Gloves, safety glasses and apron for handling formalin	
1 L	Buffered formalin-sugar solution (12%) for preserving zooplankton	
2	Alka-Seltzer® tablets to anaesthetize zooplankton prior to preservation	
1 roll	Plastic electrician's tape for sealing plankton sample jars	

5.6 Literature cited

Baker, J.R., D.V. Peck, and D.W. Sutton (editors). 1997. Environmental Monitoring and Assessment Program Surface Waters Field Operations Manual for Lakes. EPA/620/R-97/001. U.S. Environmental Protection Agency, Washington, DC.

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Draft

EMAP-GRE WATER CHEMISTRY AND PLANKTON FORM (front)

SITE ID: GRW04449- DATE: ___ / ___ / 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 Reviewed by (Initials): ______

Water Chemistry

Water Sample			FLAG
Sample ID 2 L composite			
Sample	Collected?	Composite of 3 Stations?	FLAG
2 L composite 1	☐ Yes	☐ Yes ☐ No	
500 mL grab	☐ Yes	☐ Yes ☐ No	
2 L Composite Field Duplicate Sample ID			
DI Blank Sample ID			

DO/PH Calibration		FLAG
Altitude at Calibration (m)		
Was DO meter calibrated on day of sampling? ☐ Yes ☐ No		
Was pH meter calibrated on day of sampling? ☐ Yes ☐ No		
Alkalinity	Sample ID	
Alkalinity Duplicate	Sample ID	

Water Quality Measurements

* See below for rules on which depths to take readings;flag depths not used.	River Left			Thalweg			River Right		
	0.5 m from surface	Mid depth	0.5 m from bottom	0.5 m from surface	Mid depth	0.5 m from bottom	0.5 m from surface	Mid depth	0.5 m from bottom
	Total depth	*Sample depth*	*Sample depth*	*Total depth*	*Sample depth*	*Sample depth*	*Total depth*	*Sample depth*	*Sample depth*
Depth xx.x m									
DO (mg/L)									
Conductivity (uS/cm)									
Temperature (C)									
pH									
Flag									

Phytoplankton Composite Desired Sample (1935-ml composite excluding preservative)

Sample ID	Composite vol. (mL)	Number of Locations Sampled (0-3):	FLAG

63-um Macrozooplankton Composite Sample (180-L composite filtration desired)

Sample ID	Volume filtered (L)	Number of Locations Sampled (0-3):	FLAG

20-um Microzooplankton Composite Sample (18-L composite filtration desired)

Sample ID	Volume filtered (L)	Number of Locations Sampled (0-3):	FLAG

* If depth at the station >2m, collect meter readings and a subsample 0.5 m above the bottom, mid-depth, and 0.5m from the surface; subsample volumes: 445mLfor water; 215mL for phytoplankton, 20L for macrozooplankton, 2L for microzooplankton.

* If depth at the station $\leq$2m and $\geq$1m, collect meter readings and a subsample 0.5 m above the bottom, mid-depth, and 0.5m from the surface; subsample volumes: 665mLfor water; 325mL for phytoplankton, 30L for macrozooplankton, 3L for microzooplankton.

* If depth at the station $\leq$1m, collect meter readings and a sample at mid-depth 1.3L for water; subsample volumes: 650mL for phytoplankton, 60L for macrozooplankton, 6L for microzooplankton.

Flag codes: K=no measurement made, U=suspect measurement;F1, F2, etc=misc flags assigned by field crew. Explain in comments.

16.

Figure 5-1. Water chemistry and plankton form (front). This version of the form has a typographic error in the information at the bottom of the form: no sample is collected at mid-depth if depth at the station is <2 m and >1 m.

EMAP-GRE WATER CHEMISTRY AND PLANKTON FORM (back)

SITE ID: GRW04449- DATE: ___ / ___ / 2 0 0 __ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 Reviewed by (Initials): ______

Water Chemistry (cont.)			
Secchi Depth (cm)	River Left	Thalweg	River Right
Is disk visible resting on bottom?	Y N	Y N	Y N
FLAG			

Turbidity						
Measurement	Replicate 1	Replicate 2	Replicate 3	Duplicate 1	Duplicate 2	Duplicate 3
Turbidity (NTU's)						
Sample Temperatures (C)						
FLAG						

Chlorophyll *a* Filtration (GFF filter)					
Sample ID	Volume filtered (mL)	FLAG	Duplicate Sample ID	Duplicate Volume filtered (mL)	FLAG

Geochemical Markers (GFF filter)			
Sample ID	Volume filtered (mL)	Filter ID	FLAG
		G	
Duplicate Sample ID	Duplicate Vol. filtered (mL)	Duplicate Filter ID	FLAG
		G	

Total Suspended Solids Membrane Filter Pair 1			
Sample ID	Volume filtered (mL)	Filter Pair ID	FLAG
		M	

Total Suspended Solids Membrane Filter Pair 2			
Sample ID	Volume filtered (mL)	Filter Pair ID	FLAG
		M	
Duplicate Sample ID	Duplicate Vol. filtered (mL)	Duplicate Filter Pair ID	FLAG
		M	

FLAG	COMMENTS

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

17.

Figure 5-2. Water chemistry and plankton form (back).

WATER CHEMISTRY

WC AL

GRW04449- ___ ___ ___

___/___/ 200__

Site visit number 1 2

300213

PHYTOPLANKTON (PP)

(4% formalin)

GRW04449-___ ___ ___

___/___/ 200__

Composite volume _____ L

Site visit number 1 2

300214

ZOOPLANKTON (4% formalin)

BZ (63µm) LZ (20µm)

GRW044449-___ ___ ___

___/___/ 200__

Volume filtered _____ L

Site visit number 1 2

300215

FILTERS

CF GF SS1 SS2

GRW04449-___ ___ ___

___/___/ 200__

Volume filtered _____ mL

Filter ID ___________

Site visit number 1 2

300216

____

Sample type

GRW04449- ___ ___ ___

___/___/ 200__

Comp/filtered vol.________

Site visit number 1 2

Sample ID______________

Figure 5-3. Labels for water quality and plankton samples. WC = water chemistry composite, BZ = macrozooplankton, LZ = microzooplankton, AL = alkalinity, CF = chlorophyll filter, GF = geochemical markers filter, SS1 = total suspended solids filter pair 1. The bottom number on the labels is a unique sample ID. The bottom continuation label is used if additional containers are needed for a sample. Not actual size.

Figure 5-4. Apparatus for attaching instrument sensors and sample-collecting hoses to the sounding weight (30 lbs in this picture). A sounding-weight hanger-bar has been fabricated to accommodate a Hydrolab DataSonde. The DataSonde is attached to the hanger bar, facing forward, with stainless hose clamps (covered by tape in this picture). The hose to the peristaltic pump (small diameter) is taped to the DataSonde. The hose to the high volume Guzzler pump (large diameter) is attached to the hanger bar with plastic cable ties. A one-way valve is attached to the end of the Guzzler hose. When the apparatus is lowered, the instrument cables and hoses are attached to the winch line with cable ties. Other configurations are acceptable.

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Section 6
Aquatic Vegetation

Ted R. Angradi[2], and E. William Schweiger[1]

Aquatic vegetation has multiple ecological functions in Great River Ecosystems. Aquatic plant beds generate dissolved oxygen, stabilize bed sediments, filter suspended sediment, and immobilize nutrients and toxic substances (Rogers and Theiling 1999). Aquatic plants are an important food source for waterfowl and other wildlife, and they provide substrate for invertebrates and habitat for fish (Rogers and Theiling 1999). Submerged aquatic vegetation (SAV) is sensitive to anthropogenic stressors, including excessive turbidity, sedimentation, flow modification, and exotic herbivores. Relating SAV community structure, abundance, and distribution to stressors can provide a biological basis for water quality criteria. For example, understanding the influence of turbidity on SAV beds could lead to development of light-related water-quality criteria for Great Rivers (UMRCC 2003). The EMAP-GRE method is adapted from Yin et al. (2000).

6.1 Aquatic vegetation sampling

Littoral aquatic vegetation is sampled from the main-channel shoreline (MCS) by the river crew. Plant cover and species occurrence is noted visually, and total and species-specific density is estimated from samples collected with a vegetation rake. Samples are processed in the field, although voucher specimens should be retained for identification when necessary.

6.1.1 Sample locations

At each site, two 500-m main channel shoreline (MCS) transects starting at the intersection of the cross-channel transect and the MCS (Figure 4-1), are located and flagged out by either the fish- or river-sampling crew, depending on which crew arrives at the site first. The primary transect is initially flagged at 100-m intervals; intermediate littoral stations at 50-m

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1 National Park Service, 1201 Oakridge Drive, Fort Collins, CO 80525

2 U.S. Environmental Protection Agency, Office of Research and Development, National Health and Environmental Effects Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804

intervals are located and flagged using a handheld GPS or by visual estimation during littoral sampling (use a different flag color than at the 100 m stations). At every other shoreline station (A, C, E, G, I, K; Figure 4-2) visual and quantitative methods are used to quantify aquatic vegetation in a 2 x 5 m littoral plot (Figure 6-1). Plots are slightly offset from the station location to avoid the effects of disturbance from other littoral sampling (macroinvertebrates, sediment, and periphyton) and are slightly offset from the wetted margin to avoid the effects of wave action. For guidance on where to collect aquatic vegetation samples in a dike field, see Figure 10-1. In swift, turbid reaches such as the lower Missouri River, aquatic plant beds will rarely be encountered in the main channel.

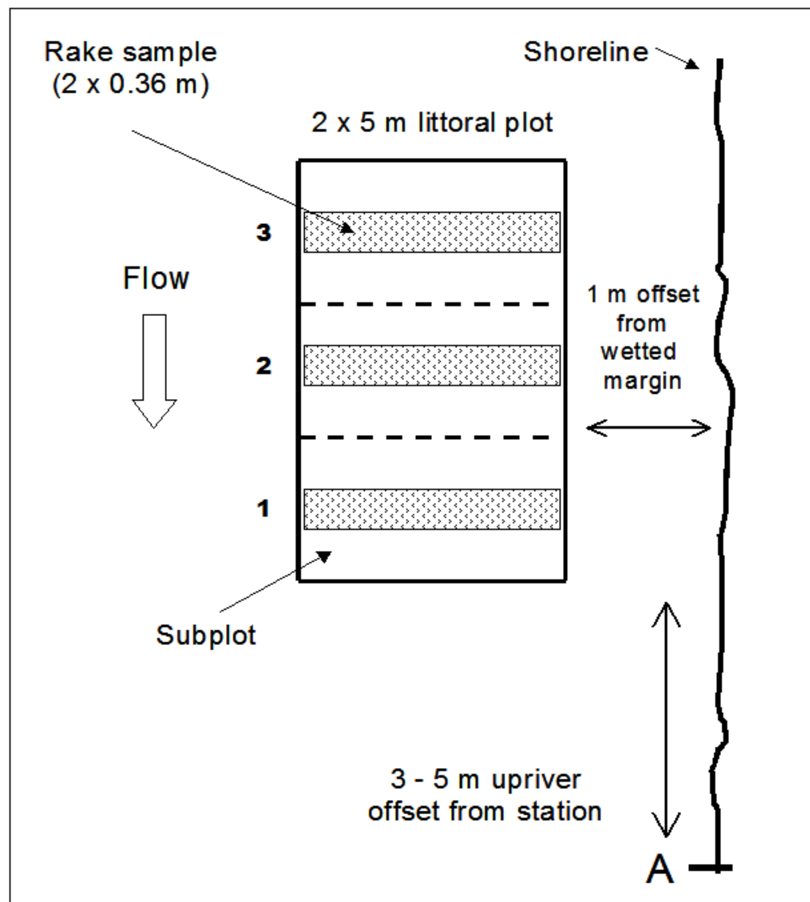

Figure 6-1. Littoral plot for aquatic vegetation sampling. Not to scale.

6.1.2 Sampling procedures

In each littoral plot, percent cover by life form and taxa richness are estimated visually. Habitat characteristics of each plot, including depth, velocity, and substrate are also quantified. The relative abundance of each submersed taxa is estimated from a 0.72-m^2 rake sample (Figure 6-2) in each of three subplots. The density of vegetation is estimated from the amount of vegetation caught on the rake. Procedures for aquatic vegetation sampling are described in Table 6-1.

Figure 6-2. Double-headed sampling rake. Each tine is marked into 5 sections of equal length. In this sample, total density would be scored as "2" based on rake fullness.

6.1.3 Taxa codes and voucher specimens

Nomenclature and synonymy follow protocols and references established in Yin et al. (2000). Table 6-2 lists expected species and their codes. Codes are derived from the USDA Plants database (http://plants.usda.gov/) and Yin et al. (2000). Codes and nomenclature for species not in Table 6-2 (identified in the field or for voucher specimens in the lab) should be derived from the Plants database. If genus is known but not species, use the first four letters of the genus with "?" inserted between the second and third letters or use the genera level USDA code.

Table 6-1. Aquatic vegetation sampling procedures.

1.	Fill in the site ID and date on the aquatic vegetation form (Figure 6-3).
2.	Go to littoral sample station A (aquatic vegetation is sampled at stations A, C, E, G, I, and K). Visually establish a 2 m wide x 5 m long littoral plot starting 3 - 5 m upriver from the flagged station and offset 1 m from the wetted margin (Figure 6-1). If there is no aquatic vegetation in the plot, go to step 12.
3.	If vegetation is present, visually estimate the percent cover, by life-form categories, of vegetation within the **entire 2 x 5 meter plot**. Life-form categories include non-rooted floating-leaved, rooting floating-leaved, and emergent. Use the following cover codes: Cover (%) — Cover code 81-100 — 5 61- 80 — 4 41- 60 — 3 21- 40 — 2 1- 20 — 1 None — 0
4.	Estimate the non-submersed taxa richness at or above the water surface in the 2 x 5 meter plot. If taxa are not readily distinguishable, use distinct morphological variants as proxies for taxa.
5.	Prior to rake sampling, evaluate the sampleability of the 2 x 5 meter plot at the station. If the plot is not sampleable due to safety concerns or excess depth, place a flag in the “Plot characteristics” section. Explain the flag in comments.
6.	If rake sampling is possible, visually divide the plot into three evenly-spaced subplots (numbered from down river: 1, 2, 3; Figure 6-1) and collect a rake sample by extending the rake 2 meters outward from the shoreline at the midpoint of each subplot and lowering the head to the bottom. Drag the rake along the bottom back to the shoreline for 2 m. Twist the rake 180 degrees as it is raised out of the water.
7.	The density of vegetation is estimated from the amount of vegetation caught on the upper set of tines on the rake (after it has been twisted 180 degrees) or the “rake fullness”. Each tine should be marked into 5 even vertical segments (requiring 4 horizontal lines on each tine, Figure 6-2). Vegetation density ranges from 0 to 5. If there is no vegetation on the rake, the density is 0; if there is vegetation on the rake but the amount does not reach the first horizontal mark, the density is scored as “1”; if the vegetation on the rake reaches between the first and second horizontal mark (from the bottom) the vegetation density is scored as “2”, etc. If vegetation is not evenly distributed on the rake, visually distribute the catch across the tines before assigning a density score.
8.	Estimate the total submerged vegetation density of each rake sample (Figure 6-3). Next, record the taxa code (Table 6-2) and the density code (0 - 5) for each taxa collected on the rake on the back of the form (Figure 6-4). Record “UNKN” for unknowns (QE codes 3 and 4, see below). Each taxa may have a density up to 5. In contrast with the surface richness estimate, submersed taxa are also sampled. Use a continuation form(s) if necessary (Figure 6-5).

Continued

Table 6-1. Aquatic vegetation sampling procedures, continued.

9. For all taxa code designations used, characterize the quality of the taxonomic evaluation in the "QE" field using the following scale:

Taxonomic evaluation	QE code
Species code matches definition in Table 6-3	1
Genus certain	2
Genus and species suspected	3
Unknown	4

10. Collect specimens for taxa with a QE code other than 1. Place each specimen in a plastic bag with an Aquatic Plant Specimen Tag (one taxa per bag; Figure 6-6). Record the tag number on the form. Attempt to collect at least two specimens with roots and flowers (if possible). Duplicate specimens may be placed in the same bag and have the same specimen tag number. Place bagged specimens in a larger bag. Place an Aquatic Plant Specimen Label (Figure 6-6) on the outer bag and cover with clear tape. Place the specimens in a cooler on ice.

11. Based on the sediment brought up with the rake samples or visually from the shoreline, classify the dominant substrate in the 2 x 5 m plot and note the presence/absence of detritus (Figure 6-3).

12. Assign a depth category to the 2 x 5 m plot based on the midpoint of the rake samples. Estimate water velocity in the plot using the qualitative scale on the form (Figure 6-3).

13. Search for aquatic plants along the shoreline between littoral plots. If present, check the appropriate box (Figure 6-3) and list taxa codes in comments.

14. Repeat steps 2 -13 at the remaining five shoreline stations.

15. Place each bagged plant specimen in a larger bag. Place an Aquatic Plant Specimen Label (Figure 6-6) on the outer bag and cover with clear tape. Record the sample ID from the label on the form (Figure 6-4). Place the specimens in a cooler on ice.

Table 6-2. Taxa codes for aquatic plants. Life form codes: A = filamentous algae; N = non-rooted floating-leaved; E = emergent; F=Floating; S = submersed; U = not applicable.

Code	Life form	*Scientific name*	Common name	Code	Life form	*Scientific name*	Common name
ACSA2	E	*Acer saccharinum*	silver maple	NELU	F	*Nelumbo lutea*	American lotus
ALPL	E	*Altissima plantago-aquatica*	American waterplantain	NULU	F	*Nuphar variegata*	yellow pond lily
AM?AR	E	*Amaranthus spp.*	pigweed	NYTU	F	*Nymphaea odorata tuberosa*	white waterlily
AMTU	E	*Amaranthus tuberculatus*	roughfruit amaranth	PAFL5	E	*Paspalum fluitans*	horsetail paspalum
AMTR	E	*Ambrosia trifida*	great ragweed	PESE6	E	*Penthorum sedoides*	ditch stonecrop
AMCO	E	*Ammannia coccinea*	valley redstem	PHAR3	E	*Phalaris aruninaceae*	reed canary grass
AMCOP	E	*Ammannia coccinea purpurea*	valley redstem	PHAU7	E	*Phragmites australis*	common reed
AP?OC	E	*Apocynum spp.*	dogbane	PH?RA	E	*Phragmites spp.*	reed
ASIN	E	*Asclepias incarnata*	swamp milkweed	PHLA3	E	*Phyla lanceolata*	lanceleaf fogfruit
AZCA	N	*Azolla caroliniana*	Carolina mosquitofern	PH?YS	E	*Physostegia spp.*	dragonhead
AZME	N	*Azolla mexicana*	Mexican mosquitofern	POPR	E	*Poa pratensis*	Kentucky bluegrass
AZ?OL	N	*Azolla spp.*	mosquitofern	PO?A	E	*Poa spp.*	bluegrass
BARO	E	*Bacopa rotundifolia*	disk waterhyssop	POACEA	E	*Poaceae*	grass family
BIAR	E	*Bidens aristosa*	bearded beggarticks	POAM8	E	*Polygonum amphibium*	water knotweed
BOCY	E	*Boehmeria cylindrica*	false nettle	POAME	E	*Polygonum amphibium v. emersum*	longroot smartweed
CASES	E	*Calystegia sepium sepium*	false hedge bindweed	POHY2	E	*Polygonum hydropeperiodes*	swamp smartweed
CA?RE	E	*Carex spp.*	sedge	POLO13	E	*Polygonum pensylvanicum*	Pennsylvania smartweed
CEOC2	E	*Cephalanthus occidentalis*	common buttonbush	POPE2	E	*Polygonum pensylvanicum*	Pennsylvania smartweed
CEDE4	S	*Ceratophyllum demersum*	coon's tail	POPU5	E	*Polygonum punctatum*	dotted smartweed
CHSE4	E	*Chamaesyce serpens*	matted sandmat	PO?LY	E	*Polygonum spp.*	smartweed
CH?AR	S	*Chara spp.*	chara	POCO14	E	*Pontederia cordata*	pickerelweed
CODI5	E	*Commelina diffusa*	climbing dayflower	PODE3	E	*Populus deltoides*	eastern cottonwood
CYER2	E	*Cyperus erythrorhizos*	redroot flatsedge	POAL8	S	*Potamogeton alpinus*	alpine pondweed
CY?PE	E	*Cyperus spp.*	flatsedge	POCR3	S	*Potamogeton crispus*	curly pondweed
CYST	E	*Cyperus strigosus*	straw-colored flatsedge	POEP2	S	*Potamogeton epihydrus*	ribbonleaf pondweed
DEVE	E	*Decodon verticillatus*	swamp loosestrife	POFO3	S	*Potamogeton foliosus*	leafy pondweed
DEIL	E	*Desmanthus illinoiensis*	prairie bundleflower	NLPW	S	*Potamogeton foliosus/pusillus*	narrow-leaved pondweeds
DI?GI	E	*Digitaria spp.*	crabgrass	PONO2	S	*Potamogeton nodosus*	longleaf pondweed
DIVI5	E	*Diospyrus virginiana*	common persimmon	POPU7	S	*Potamogeton pusillus*	small pondweed
DUAR3	E	*Dulichium arundinaceum*	threeway sedge	PORI2	S	*Potamogeton richardsonii*	Richardson's pondweed
ECCR	E	*Echinochloa crus-galli*	barnyardgrass	POZO	S	*Potamogeton zosteriformis*	flatstem pondweed
ECES	E	*Echinochloa esculenta*	Japanese millet	RALO2	S	*Ranunculus longirostris*	longbeak buttercup
ECMU2	E	*Echinochloa muricata*	rough barnyardgrass	RA?NU	S	*Ranunculus spp.*	buttercup
ECWA	E	*Echinochloa walteri*	coast cockspur grass	RATR	S	*Ranunculus trichophyllus*	thread leaf crowfoot
ECPR	E	*Eclipta prostrata*	false daisy	RIFL4	N	*Riccia fluitans*	slender liverwort
EL?EO	E	*Eleocharis spp.*	spikerush	RINA2	N	*Ricciocarpos natans*	liverwort
ELCA7	S	*Elodea canadensis*	Canadian waterweed	RONA2	E	*Rorippa nasturtium-aquaticum*	watercress
ELVI3	E	*Elymus virginicus*	Virginia wildrye	RUCR	E	*Rumex crispus*	curly dock
EQ?UI	E	*Equisetum spp.*	horsetail	RU?ME	E	*Rumex spp.*	dock
ERFR	E	*Eragrostis frankii*	sandbar lovegrass	SACU	E	*Sagittaria cuneata*	arumleaf arrowhead
ERPE	E	*Eragrostis pectinacea*	tufted lovegrass	SALA2	E	*Sagittaria latifolia*	broadleaf arrowhead
ER?IG	E	*Erigeron spp.*	fleabane	SARI	E	*Sagittaria rigida*	stiff arrowhead
ALGA	A	*filamentous algae*	filamentous algae	SA?GI	E	*Sagittaria spp.*	arrowhead

Continued

Table 6-2. Taxa codes for aquatic plants, continued.

Code	Life form	Scientific Name	Common Name	Code	Life form	Scientific Name	Common Name
FOAC	E	*Forestiera acuminata*	eastern swampprivet	SAEX	E	*Salix exigua*	sandbar willow
FRPE	E	*Fraxinus pennsylvanica*	green ash	SANI	E	*Salix nigra*	black willow
GAOBO	E	*Galium obtusum obtusum*	bluntleaf bedstraw	SA?LI	E	*Salix spp.*	willow
GA?LI	E	*Galium spp.*	bedstraw	SCFL	E	*Schoenoplectus fluviatilis*	river bulrush
ZODU	S	*Heteranthera dubia*	water stargrass	SCVA	E	*Schoenoplectus tabernaemontani*	softstem bulrush
HILA2	E	*Hibiscus laevis*	halberdleaf rosemallow	SC?IR	E	*Scirpus spp.*	bulrush
IPLA	E	*Ipomoea lacunosa*	whitestar	SE?NE	E	*Senecio spp.*	ragwort
JUEF	E	*Juncus effusus*	common rush	SEVI4	E	*Setaria virdis*	green bristlegrass
LEOR	E	*Leersia oryzoides*	rice cutgrass	SIAN	E	*Sicyos angulatus*	oneseed burrcucumber
LE?ER	E	*Leersia spp.*	cutgrass	SM?IL	E	*Smilax spp.*	greenbrier
LEMI3	N	*Lemna minor*	small duckweed	SPEU	E	*Sparganium eurycarpum*	giant burreed
LETR	N	*Lemna trisulca*	star duckweed	SPPO	N	*Spirodela polyrrhiza*	big duckweed
LEMNA	N	*Lemnaceae*	duckweed family	POPE6	S	*Stuckenia pectinatus*	sago pondweed
LEFI	E	*Leptochloa filiformis*	muronate sprangletop	SYLAL7	E	*Symphyotrichum lateriflorum v. alateriflorum*	calico aster
LEFA	E	*Leptochloa fusca fascicularis*	bearded sprangletop	TYAN	E	*Typha angustifolia*	narrowleaf cattail
LEPA3	E	*Leptochloa panicoides*	Amazon sprangletop	TYLA	E	*Typha latifolia*	common cattail
LUDE4	F	*Ludwigia decurrens*	wingleaf primrosewillow	TY?PH	E	*Typha spp.*	cattail
LUPE5	E	*Ludwigia peploides*	floating primrosewillow	ULAM	E	*Ulmus americana*	American elm
LU?DW	F	*Ludwigia spp.*	primrosewillow	URDI	E	*Urtica dioica*	stinging nettle
LYSA2	E	*Lythrum salicaria*	purple loosestrife	UR?TI	E	*Urtica spp.*	nettle
MYSI	S	*Myriophyllum sibiricum*	northern watermilfoil	UTMA	S	*Utricularia macrorhiza*	common bladderwort
MYSP2	S	*Myriophyllum spicatum*	Eurasian watermilfoil	VAAM3	S	*Vallisneria americana*	wild celery
MY?RI	S	*Myriophyllum spp.*	watermilfoil	VEHA2	E	*Verbena hastata*	swamp verbena
NAFL	S	*Najas flexilis*	nodding waternymph	VICI2	E	*Vitis cinerea*	graybark grape
NAGR	S	*Najas gracillima*	slender waternymph	VI?TI	E	*Vitis spp.*	grapevine
NAGU	S	*Najas guadalupensis*	southern waternymph	WOCO	N	*Wolffia columbiana*	Columbian watermeal
NAMI	S	*Najas minor*	brittle waternymph	WO?LF	E	*Wolffia spp.*	watermeal
				XAST	E	*Xanthium strumarium*	rough cocklebur
NOSMPL	U	*no sample*		ZAPA	S	*Zannichellia palustris*	horned pondweed
UNKN	U	*Unknown*		ZIAQ	E	*Zizania aquatica*	wild rice

6.2 QA considerations for aquatic vegetation sampling

Table 6-3. QA considerations for aquatic vegetation sampling.

- Attempt to standardize the rake sampling among all substrate and vegetation conditions encountered among sites.
- Do not attempt to sample where conditions prevent a good rake sample (e.g., depth >2 m or in areas of fast current).
- Be sure to fill out the header information on the continuation forms properly so they can be associated with the main forms

6.3 Equipment and supplies for sampling aquatic vegetation

Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

Table 6-4. Checklist of equipment and supplies for aquatic vegetation sampling.

Qty	Item	
2	Double headed vegetation sampling rake with 3-m long handle. Buy 2 steel garden rakes, cut the head off one and weld it to the head of the other at the same angle from the handle. The rake head should be 36 cm wide and have about 14 5-cm-long tines on each side. Add a rope extension to the handle and mark the handle in 10-cm increments. Mark the tines into 5 equal vertical increments by scoring the tines or with paint. See Figure 6-2 .	
1	Aquatic vegetation species list (Table 6-3)	
1 set	Aquatic plant identification guides (not provided by EPA)	
20	1-gallon self-sealing plastic bags	
20	2-gallon self-sealing plastic bags	
1 set	Aquatic vegetation forms	
1 set	Labels (voucher tags, labels)	

6.4 Literature cited

Rogers, S. and C.Theiling 1999. Submersed aquatic vegetation. *In* Ecological status and trends of the Upper Mississippi River System 1998: A Report of the Long Term Resource Monitoring Program. U.S. Geological Survey, Upper Midwest Environmental Sciences Center, La Crosse, WI. April 1999. LTRMP 99-T001.

Upper Mississippi River Conservation Committee, Water Quality Technical Section (UMRCC). 2003. Proposed water quality criteria necessary to sustain submersed aquatic vegetation in the Upper Mississippi River. Rock Island, IL. 6p .

Yin, Y., J.S. Winkelman, and H.A. Langrehr. 2000. Long term resource monitoring program procedures: aquatic vegetation monitoring. U.S. Geological Survey, Upper Midwest Environmental Sciences Center, 2630 Fanta Reed Road, Lacrosse, WI 54603 http://www.umesc.usgs.gov/data_library/vegetation/vegetation_page.html ftp://ftp.umesc.er.usgs.gov/pub/media_archives/documents/reports/1995/95p00207.pdf

Draft

EMAP-GRE AQUATIC VEGETATION FORM (front)

Reviewed by (Initials): ______

SITE ID: GRW04449-______ DATE: ___/___/ 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2

Cover and Taxa Richness in entire 2 x 5m littoral plot						
Station	Non-rooted floating	Rooted floating leaved	Emergent	Richness	Flag	
A	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0			Cover Codes: 5 = 81 - 100% 4 = 61 - 80% 3 = 41 - 60% 2 = 21 - 40% 1 = 1 - 20% 0 = 0%
C	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0			
E	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0			
G	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0			
I	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0			
K	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0			

Plot Characteristics in entire 2 x 5m littoral plot						
Station	Velocity Code	Depth Code	Dominant Substrate Code	Detritus presence/ absence	Flag	Code Keys and Descriptions
A	2 1 0	2 1 0		Y N		XB = BOULDER (1000 TO 4000 mm) METERSTICK TO CAR SB = SM.BOULDER (250 TO 1000mm) BASKETBALL TO METERSTICK CB = COBBLE (64 TO 250 mm) TENNIS BALL TO BASKETBALL GC = COARSE GRAVEL (16 TO 64 mm) MARBLE TO TENNISBALL GF = GRAVEL (2 TO 64 mm) LADYBUG TO MARBLE SA = SAND (0.06 TO 2 mm) GRITTY - UP TO LADYBUG SIZE FN = SILT/ CLAY / MUCK NOT GRITTY HP = HARDPAN FIRM, CONSOLIDATED FINE SUBSTRATE WD = WOOD ANY SIZE WOOD OT =OTHER FLAG AND COMMENT VELOCITY: 2 = > 1m/s, 1 = < 1m/s and >0, 0 = 0m/s. DEPTH: 2 = > 1m, 1 = > 0.5m and < 1m, 0 = < 0.5m.
C	2 1 0	2 1 0		Y N		
E	2 1 0	2 1 0		Y N		
G	2 1 0	2 1 0		Y N		
I	2 1 0	2 1 0		Y N		
K	2 1 0	2 1 0		Y N		

Total Submersed Vegetation Density (rake fullness) in Subplots						
Subplot	Station					
	A	C	E	G	I	K
1	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0
2	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0
3	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0
* Outside of plots	☐	☐	☐	☐	☐	☐
Flag						

* Presence/absence of vegetation outside of plot. If present, check box and list taxa codes in comments.

Flag	COMMENTS

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

18.

Figure 6-3. Aquatic vegetation form (front).

Draft

EMAP-GRE AQUATIC VEGETATION FORM (back)

Reviewed by (Initials): ______

SITE ID: GRW04449- ______ DATE: ___ / ___ / 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 PAGE: 1 OF ___

Rake Sample of Submersed Taxa

Sample ID ______

Density of individual submersed taxa from rake samples (rake fullness)

Sta.	Taxa Code	Rake Sample 1	Rake Sample 2	Rake Sample 3	QE Code	Specimen Tag #	Flag
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		

QE Codes: 4 = Unknown, 3 = Genus and species suspected, 2 = Genus certain, 1 = Code match. Collect one specimen if QE Code not = 1.

COMMENTS

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

19.

Figure 6-4. Aquatic vegetation form (back).

Draft

EMAP-GRE AQUATIC VEGETATION FORM (cont.)

Reviewed by (Initials): ______

SITE ID: GRW04449- ______ DATE: ___ / ___ / 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 PAGE: ____ OF ____

Rake Sample of Submersed Taxa

Sample ID ______

Density of individual submersed taxa from rake samples (rake fullness)

Sta.	Taxa Code	Rake Sample 1	Rake Sample 2	Rake Sample 3	QE Code	Specimen Tag #	Flag
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		
		5 4 3 2 1 0	5 4 3 2 1 0	5 4 3 2 1 0	4 3 2 1		

QE Codes: 4 = Unknown, 3 = Genus and species suspected, 2 = Genus certain, 1 = Code match. Collect one specimen if QE Code not = 1.

COMMENTS

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

20.

Figure 6-5. Aquatic vegetation continuation form (one sided form).

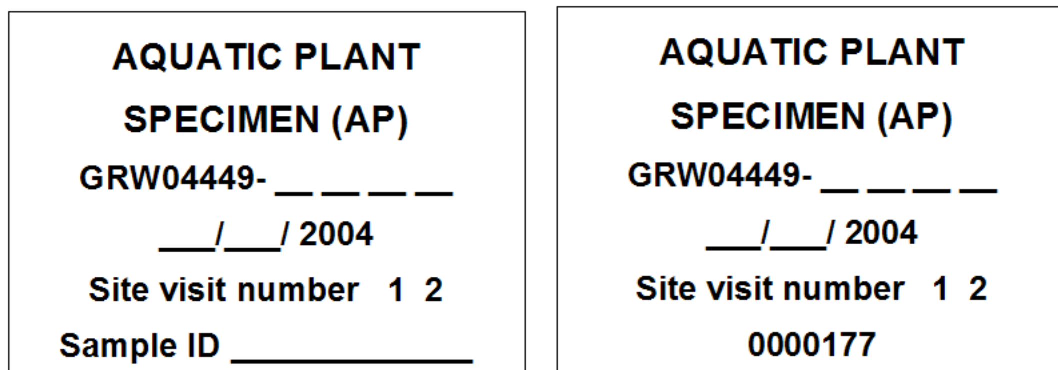

Figure 6-6. Labels and tags for aquatic plant specimens. Plant specimen tags are placed in the bags containing individual specimens. Specimen labels are placed on the larger outer bag holding the smaller bags. Use continuation label at upper right if extra outer bags are needed. Not actual size.

Section 7
Riparian Habitat

E. William Schweiger[1] and Ted R. Angradi[2]

Interactions among aquatic and riparian ecosystem components are important in Great River ecosystem functioning and condition. Riparian ecosystems contribute to and moderate the flux of materials and energy between terrestrial and aquatic habitats within GREs. Because shoreline and riparian characteristics (vegetation, stability, human modifications) can affect channel form, water velocity, and substrate, they influence ecosystem condition at multiple spatial scales. This section includes procedures for evaluating riparian vegetation, land cover, bank morphology, and human influences at each site. Methods in this section are adapted, in part, from Peck et al. (Unpublished drafts) and Ringold et al. (2001).

7.1 Sample locations

Data collection for the main channel shoreline (MCS) and riparian habitat is conducted by the river-sampling crew at shoreline stations and riparian plots along the primary 500 m MCS transect (Figure 7-1; see also Figure 4-2). The MCS is defined in Section 4.2.1. MCS bank sample stations are located at transects A, E, and K, located 0, 200, and 500 m upriver from the start of the primary MCS transect (Figure 7-1). Shoreline and macrohabitat type is evaluated for each of the five intervening 100-m shoreline segments.

7.2 Riparian plots

7.2.1 Location on the main channel shoreline

Riparian plots are established at one of two positions in relation to the MCS (see Section 4.2.1): 1) at or just above the bankfull elevation, or 2) at the main-channel wetted margin. If there are no limitations to riparian plot access, the preferred location is at or just above the

1 National Park Service, 1201 Oakridge Drive, Fort Collins, CO 80525

2 U.S. Environmental Protection Agency, Office of Research and Development, National Health and Environmental Effects Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804

bankfull elevation on the MCS. Access is considered limited when reaching the MCS transect requires more than 10 minutes of walking, relocation of the boat (e.g., into or across a secondary channel), or crossing posted or other obviously private property with controlled access. The amount of effort expended to locate riparian plots at the bankfull level is at the discretion of the crew leader. No specific procedures are provided here for obtaining landowner permission to access riparian areas on posted or non-posted private property. It is also up to the crew leader to decide if the limited activities in riparian plots (15 - 30 minutes per station) warrant solicitation of landowner permission. Physical access may often be limited on inside bends where a point bar separates the active channel from a distant high bank. If access to a plot is incomplete, it is acceptable to only sample a subset of a plot (e.g., just subplot 1, described below) or to collect measurements remotely (i.e., by viewing vegetation from the boat). All data collected using non-standard methods should be flagged. An alternate riparian location should not be substituted for a plot that is not sampled.

The first step in locating the riparian plots is to establish the approximate bankfull elevation on the MCS. Estimating this elevation during base flow is somewhat subjective. Best judgement should be used in cases where the bankfull elevation on the MCS is difficult to locate. If possible, document the bankfull level with a description and site photos (see Section 4). Bankfull elevation corresponds to a flow stage with a recurrence interval of 1 to 2 years which generally does not inundate the floodplain. Bankfull elevations often correspond to a "greenline," or the line of first perennial vegetation above the wetted margin of the main channel. The bankfull elevation may also be detected from the presence of debris caught on overhanging vegetation. However, subsequent to large floods, this material may be well above the bankfull level. For braided streams where perennial vegetation may be established on bars between the channels or on islands, the bankfull elevation or greenline of interest is on the MCS. On steep banks where the bankfull elevation or greenline is part way up the slope, the edge of the riparian plots will be located at the top of the bank. If the MCS is gradually sloped, the bankfull elevation may be more difficult to locate. Often there is a small berm on the MCS at the bankfull elevation, with subtle changes in vegetation. If MCS bank station A, E, or K is on a jetty or other artificial structure projecting into the channel, the riparian plot should be positioned landward of the jetty on the closest point on the non-artificial MCS.

If access to a bankfull elevation location is physically limited (e.g., at an inside bend

where the high bank is distant from the river) but is not posted or otherwise restricted, riparian plots should be established just above the wetted margin of the main channel along the MCS.

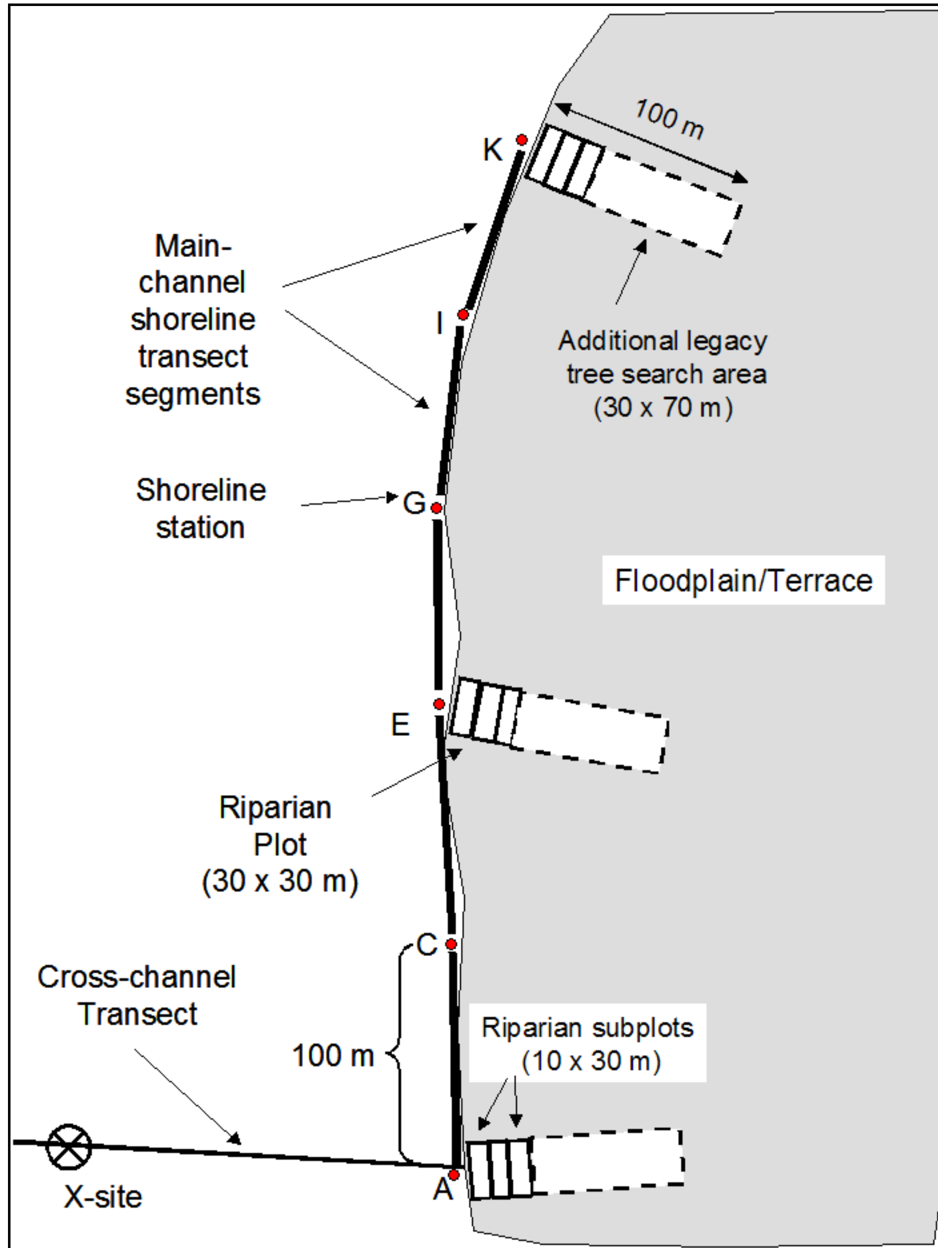

Figure 7-1. Sample layout for riparian habitat sampling.

7.2.2 Riparian plots

Riparian plots are centered on the corresponding MCS bank station (station A, E, or K; Figure 7-1). The plots extend landward 30 m perpendicular to the main channel and 15 meters upriver and downriver (Figure 7-2). Each plot is divided into three, 30 x 10 m subplots. Plot borders are established by stretching a tape 30 m into the riparian corridor orthogonal to the MCS (flags may be placed at 10 and 20 m). The upriver and downriver extent of each subplot are estimated by eye. The three subplots are numbered 1, 2, and 3 from the MCS edge landward.

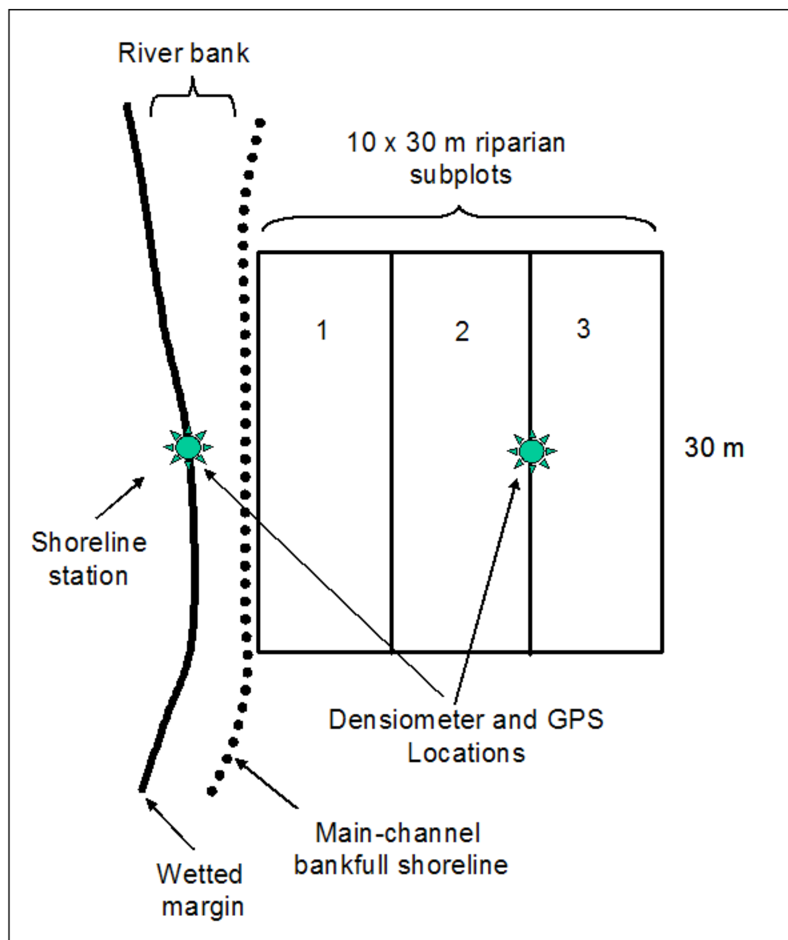

Figure 7-2. Closeup of 30 x 30 m riparian plot.

7.3 Bank and channel width measurements

MCS bank angle, height, and width and wetted channel width are determined at three shoreline stations (A, E, and K; Figure 7-1). Alternate MCS bank stations should not be substituted for a non-sampled station. MCS bank angles are estimated by viewing the bank from the wetted channel margin. Bankfull height on the MCS is measured with, in order of preference, a laser hypsometer, a surveyor's rod, or by visual estimation. If the MCS is established on a border fill or other similar habitat type, the bank angles and heights may be low. Channel widths for the main channel are measured from the MCS with a laser rangefinder sited across the channel to the wetted margin on the opposite bank. Table 7-3 describes procedures for bank measurements.

Macrohabitat and shoreline type are determined for the 100 m shoreline segments between MCS bank stations A, C, E, G, I, and K. Shoreline macrohabitat includes continuous and discrete habitat types (Table 7-1). Continuous macrohabitats (e.g., inside and outside bends) characterize every river shoreline and may lack a definitive beginning or end. Discrete macrohabitats (e.g., secondary channels, tributary mouths) are not necessarily found along every river shoreline. Macrohabitat types may overlap along a MCS. Shoreline type includes several natural and artificial categories (Table 7-2). Shoreline types are defined as mutually exclusive. Procedures for estimating the percentage of each shoreline type and macrohabitat type between each bank station are described in Table 7-3.

Table 7-1. Macrohabitat types (adapted from Sappington et al. 1998).

Macrohabitat	Description
Main channel crossover	The inflection point of the thalweg. Longitudinal length of the crossover area generally does not exceed 1.5-2 times the stream width. Shorelines are often relatively straight in crossover reaches.
Outside bend	The concave side of a river bend often characterized by rip rap or eroding cut banks
Inside bend	The convex side of a river bend; often characterized by sand bars
Pool	Low velocity area upriver of navigation locks or immediately downriver from dikes or jetties
Tributary mouth	Mouth of perennial tributary < 10 m across
Connected secondary channel	Lotic channel other than the main channel < 5 m across
Unconnected secondary channel	Channel < 5 m across, blocked at upriver end by a sand bar artificial closure, or otherwise; lentic
Other	Any other type of macrohabitat. Describe on form.

Table 7- 2. Shoreline types.

Shoreline type	Description
Stable bank	Naturally stabilized due to vegetation or substratum characteristics
Aggrading bank	Collecting sediment or extending into the channel; includes point bars and border fills
Eroding bank	Unstable, actively degrading banks
Blanket-rock revetment (riprap)	Stabilized by gravel, cobble or boulder-sized material placed at the river margin
Other revetment type	Includes car bodies, cement slabs, trash. Describe on form.
Other	Any other type of shoreline. Describe on form.

Table 7-3. Procedure for bank and shoreline measurements.

1. Proceed to shoreline station A (Figure 7-1). Fill in the site ID and date on the bank and shoreline macrohabitat form (Figure 7-3).

2. Record the coordinates of the shoreline station (Figure 7-2) using a hand-held GPS (applies to A, C, E, G, I, K).

3. At stations A, E, and K, stand at the edge of the wetted width on the MCS and site across the channel with a laser range finder and take a reading from an object judged to be at or near the wetted edge on the opposite bank. If the opposite bank of the main channel is obscured by an island, take the best reading possible and the flag the data.

4. At stations A, E, and K, estimate bankfull height on the MCS using one of the following methods: 1) use a laser hypsometer; 2) hold the surveyor's rod vertical, with its base at the water's edge and estimate (by eye) the height of the bankfull shoreline above the present water level; or 3) make a visual estimate. If the MCS is very gradual, a meter stick may be used. Depending on the hypsometer model, the minimum horizontal distance for a bank height reading may vary (minimum distance is 5 m for Opti-Logic laser hypsometers). Note the measurement method in comments.

5. At stations A, E, and K measure or estimate the bank width which is the horizontal distance (not slope distance) from the wetted edge to the bankfull level.

6. At stations A, E, and K, visually estimate the maximum bank angle from the wetted edge to the bankfull level. Ignore steep breaks in the slope that are <1 m long. Use the classes indicated on the bank and shoreline macrohabitat form (Figure 7-3).

7. At stations A, E, and K, additional riparian measurements are done before leaving the station (described later in this section).

8. Proceed to the next shoreline station. On the way, note the shoreline type (Table 7-2) and macrohabitat type (Table 7-1) in the 100-m MCS segment between stations. Estimate the percent of the segment in each shoreline and macrohabitat type. Ignore types < 5 m in length. The macrohabitat type may include discrete features (e.g., a tributary mouth) nested within continuous features (e.g., inside bend shorelines) and macrohabitat types may sum to more than 100%. Shoreline types are mutually exclusive and must sum to 100%.

9. Repeat steps 2 - 8 as appropriate at shoreline stations C, E, G, I, K.

7.4 Riparian measurements

EMAP-GRE riparian measurements include land cover/use classification, visual estimation of vegetative cover, canopy density measurements, presence of invasive plant species, legacy tree characteristics, and human activity and disturbance at the site.

7.4.1 Land cover classification

Land cover in each riparian subplot is classified by codes in Table 7-4 following procedures in Table 7-5. If a subplot appears atypical from the surrounding area, classification data for that subplot should be flagged (for example, if a road passes through a subplot). For riparian plots or subplots on posted land, an attempt should be made at classification without entering the property.

Table 7- 4. Land cover/land use classification codes.

Code	Description
B	Bare, unvegetated, non-forest, non-agricultural, undeveloped (less than 10% vegetative cover or no obvious vegetation; rock, sand, bare soil, etc.)
D	Developed/urban (pavement, buildings, residential yards, industrial/commercial areas, quarries, highways, etc.)
A	Intensive agriculture (row crop, cereal grain, grass seed, nursery, vineyard, orchard, Christmas trees, etc). Includes bare plowed fields and irrigated pasture.
X	Xeric (upland) shrubs or herbaceous. Includes non-irrigated pasture and hayfields.
HM	Mesic (riparian) herbaceous (grasses, forbs, etc.). Includes wet prairies or meadows, sedges, rushes, etc.
SM	Mesic (riparian) woody shrubs
F	Forested cover (> 10% tree [> 5 m tall] coverage)

Table 7-5. Procedures for classifying riparian subplots.

1.	Fill in site ID and date on both sides of the riparian classification and human influence form (Figure 7-4). Climb the MCS bank (if necessary) and establish the midpoint of the riverside edge of subplot 1 (Figure 7-2). The 30 x 30 meter plot will extend 15 m upriver and downriver and 30 meters landward from this point.
2.	If the land does not appear to be posted or if access is not otherwise restricted, stretch a tape 10 m perpendicular to the bank line landward, establishing the width of the first 10 x 30 m rectangular subplot (Figure 7-2). The upriver and downriver dimensions (15 m) are estimated by eye. If there is no access to any or all of the subplots, if it is unsafe, or the vegetation is impenetrable, all riparian estimates are flagged and done from the bank or closest location possible.
3.	From within riparian subplot 1, assign **only one** land cover/land use code (Table 7-4) for the subplot. If more than one code applies, use the code for the dominant land cover/land use and flag the data.
4.	Repeat the process in each subplot. Acquire and record the coordinates of the land edge of riparian subplot 2 using a handheld GPS (Figure 7-2).

7.4.2 Canopy density

Canopy density is estimated at the river's edge and the land-side edge of subplot 2 (Figure 7-2) using a Convex Spherical Densiometer (Lemmon, 1957). The densiometer must be taped exactly as shown in Figure 7-6 to limit the number of square grid intersections to 17. Densiometer readings can range from 0 (no canopy cover) to 17 (maximum canopy cover). Four measurements (upriver, downriver, landward, and toward the river) are obtained at each designated point. During measurements, hold the densiometer level approximately 1 m above the ground. The procedure is described in Table 7-6.

7.4.3 Riparian vegetation structure

Riparian vegetation areal cover is estimated by eye in three vertical layers in each subplot using methods adapted from Kaufmann and Robison (1998). Areal cover is analogous to the amount of shadow cast by a particular layer and just that layer when the sun is directly overhead. The type of woody vegetation (deciduous, coniferous, mixed, or none) is determined in each of the layers. Consider a layer "mixed" if deciduous and coniferous trees each comprise more than 25% of the areal coverage. The maximum cover in each layer is 100%, so areal

cover for the combined three layers could sum to 300%. Procedures for estimation of vegetation coverage are described in Table 7-6.

Table 7-6. Procedure for characterizing riparian vegetation structure.

1.	**Canopy density**. At river's edge (adjacent to MCS station A) hold the densiometer 1 m above the ground facing landward, away from the main channel. Position the densiometer so your face is just below the apex of the taped "V" (Figure 7-6). Count the number of intersections (0-17) that are covered by a leaf, stem, branch, or the bank. In heavy cover, it is usually easier to count the open intersections and subtract that number from 17. Record the count on the riparian classification and human influence form (Figure 7-4).
2.	Repeat step 1 three more times: facing upriver, downriver, and toward the river.
3.	**Riparian vegetation structure**. From within subplot 1, conceptually divide the riparian vegetation in into three layers: a canopy layer (>5 m [16 ft] tall), an understory layer (0.5 - 5 m tall), and a ground cover layer (<0.5 tall). Several vegetation types (e.g., grasses or woody shrubs) can potentially occur in more than one layer.
4.	Record the type of woody vegetation (deciduous, coniferous, mixed, or none) for each layer. Consider the layer "mixed" if both deciduous and coniferous trees comprise more than 25% of the areal coverage.
5.	Estimate the cover for each category (e.g., big trees, small trees) in each canopy layer (0 = absent, 1 = sparse [<10%], 2 = moderate [10 - 40%], 3 = heavy [40 - 75%], 4 = very heavy [>75%]). Estimate areal cover as the amount of shadow that would be cast by a particular layer alone if the sun were directly overhead.
6.	Estimate the percent of bare ground for the ground cover layer.
7.	Repeat Steps 3 - 6 for subplots 2 and 3.
8.	At the land-side edge of subplot 2 repeat steps 1 and 2 and record the GPS coordinates.
9.	Repeat steps 1 - 8 at each riparian plot.

7.4.4 Human influence

For each riparian subplot, the presence/absence and the proximity of human influences (i.e., activities, disturbance) are recorded. Each human influence is recorded as not present at the site; as present within subplot 1, 2, or 3; present between the river and subplot 1; present in the river; present on the opposite bank; or present on the target side of the river outside of riparian plots. Procedures for characterization of human activities and disturbances in the river and in the riparian area are described in Table 7-7.

Table 7- 7. Procedure for tallying human influence.

1.	From the edge of subplot 1 in riparian plot A, record any human influence categories between the subplot and the river.
2.	Examine the other subplots for human influence. Record any human influence present in any of the subplots (Figure 7-4). Other influences not included in the list on the form are likely to be present.
3.	From the corners of the riparian plot, search for human influences (on the river ecosystem) visible outside the plot and not already recorded from the subplots or from between subplot 1 and the river. Human influence outside the subplots may be in the river, on the opposite side of the river, or on the target side of the river. It is not necessary to circle “N” for every human influence not present.
4.	Repeat Steps 1 through 3 for riparian plots E and K.

7.4.5 Riparian "legacy" trees

Legacy tree data contribute to the assessment of "old growth" characteristics of riparian vegetation and can indicate historic riparian conditions and the potential for riparian tree growth. Legacy trees are defined as the largest tree of any species alive or dead in the riparian plot or within 100 m of the riverside edge of the riparian plot. The legacy tree may be on the floodplain or on the upland beyond the margin of the floodplain if the floodplain is <100 m wide. Procedures for identifying and recording the attributes of legacy trees are described in Table 7-8.

7.4.6 Invasive plant species

Invasive plant species may alter the structure and function of the riparian ecosystems which they invade. Stressed aquatic ecosystems are vulnerable to invasion by alien riparian plant species. Target invasive plant species are given in Table 7-9. These taxa were selected based on their degree of invasiveness in riparian systems, their capacity to alter ecosystem function, and ease of identification. Expected distributions, ID codes, and nomenclature are from the USDA Plants database (http://plants.usda.gov). Do not include species seen in another subplot as "outside of subplot" observations. Invasive species other than those in Table 7-9 may be noted in a comment. The procedures for tallying invasive plant species are described in Table 7-8.

Suspected invasive plan specimens may be informally collected for later identification or confirmation by a botanist. If necessary, these occurrences should be recorded on the field form before they are sent to the EMAP data manager. There are no formal invasive plant vouchering requirements for EMAP-GRE.

Table 7-8. Procedures for identifying riparian legacy trees and invasive plant species.

1.	**Legacy tree**. Search the three subplots and an area extending 70 m landward from subplot 3 (Figure 7-1). Identify the largest tree of any species, alive or dead (including snags), in the search area (one legacy tree for each riparian plot). If only small trees are present, select the largest example.
2.	Estimate the DBH and height of the legacy tree by category. Classify the tree as (D)eciduous, (C)oniferous, or (N)o trees. Estimate the distance from the tree to the wetted edge of the river. Record the information on the riparian legacy tree and invasive plants form (Figure 7-5).
3.	Assign a taxonomic category to the tree from the list on the form (Figure 7-5).
4.	**Invasive plant species**. Search each subplot for the presence of targeted invasive plant species (Table 7-9). Check the appropriate box on the form (Figure 7-5).
5.	Note the presence of any of the target species in the riparian areas outside riparian subplots.
6.	Repeat steps 1-6 at each riparian plot.
7.	If any other invasive plant species (not listed in Table 7-9) are observed at the site, note these in a comment.

Table 7-9. Target invasive plant species.

Common name	Genus species	USDA code	Expected occurrence by state (All = MT, ND, SD, NE, IA, KS, MO, IL, WI, MN, IN, KY, OH, WV, PA)
Canada thistle	*Cirsium arvense*	CIAR4	All
musk thistle	*Carduus nutans*	CANU4	All
leafy spurge	*Euphorbia esula*	EUES	MT, ND, SD, NE, IA, KS, MO, IL, WI, MN, IN, OH, WV, PA
Russian olive	*Elaeagnus angustifolia*	ELAN	MT, ND, SD, NE, IA, KS, MO, IL, WI, MN, KY, OH, PA
salt cedar	*Tamarix spp.*	TAMAR2	MT, ND, SD, NE, KS, MO, IL, OH, PA
buckthorn	*Rhamnus spp.*	RHAMN	All
reed canary grass	*Phalaris arundinacea*	PHAR3	All
giant reed	*Arundo donax*	ARDO4	KS, MO, IL, KY, WV
cheatgrass	*Bromus tectorum*	BRTE	All
teasel, Fuller's teasel	*Dipsacus fullonum*	DIFU2	MT, SD, NE, IA, KS, MO, IL, WI, IN, KY, OH, WV, PA
common, lesser burdock	*Arctium minus*	ARMI2	All
Japanese knotweed	*Polygonum cuspidatum*	POCU6	MT, ND, SD, NE, IA, KS, MO, IL, WI, MN, IN, KY, OH, WV, PA
mile-a-minute	*Polygonum perfoliatum*	POPE10	OH, WV, PA
garlic mustard	*Alliaria petiolata*	ALPE4	ND, NE, IA, KS, MO, IL, WI, MN, IN, KY, OH, WV, PA
purple loosestrife	*Lythrum salicaria*	LYSA2	All
knapweed	*Centaurea spp.*	CENTA	All
whitetop	*Cardaria draba*	CADR	All

7.5 General site assessment

After all bank and riparian data have been collected, an assessment of channel form and constrainment, and a general site assessment are conducted. Conditions on both shorelines in a 1000-m reach centered on the cross-channel transect, as well as conditions upriver that might influence the conditions at the site, are considered for the general assessment. The photographs in the site dossier (Section 4) are often useful for the general site assessment. Channel form and constrainment are evaluated using the procedures in Table 7-10. General assessment procedures are presented in Table 7-11.

Table 7-10. Channel form and constrainment.

1.	Fill in the site ID and data on both sides of the channel and general assessment form (Figure 7-7).
2.	Classify the channel pattern at the site as single, anastomosing, or braided. Anastomosing channels have relatively long major and minor channels branching and rejoining in a complex network. Braided channels also have multiple branching and rejoining channels, but these sub-channels are generally smaller, shorter, and more numerous, often with no obvious dominant channel(s) .
3.	Characterize channel constraint at the site. Evaluate whether the channel has a broad alluvial floodplain likely to flood. If not, determine if the channel is constrained by a V-shaped valley, an incised channel, or a narrow valley that necessarily limits the ability of the channel to migrate at high flows. For floodplain reaches, determine if the floodplain is protected wholly or partially by levees.
4.	For constrained channels, determine the constraining features. An unconstrained channel such as a floodplain river, can be locally constrained at non-flood flows by artificial revetment (riprap).
5.	Estimate the percent of the channel constrained (both banks) in the 1000-m reach centered on the cross channel transect (i.e., the 500 m primary and secondary transects). For unconstrained channels with no artificial revetments, percent constrained = 0%.
6.	Estimate valley width visually, if possible.

Watershed activities and human disturbance at the site are rated as not observed, low, moderate, or heavy. The distinction is subjective. For example, if there are only one or two houses visible from the river, the rating for "residences“ would be low. If one shoreline is

adjacent to a residential area that encroaches on the riparian zone, the rating for “residences” would be heavy. Similarly, a small patch of clear-cut logging on a hill overlooking the river would be rated as low disturbance. Logging activity in the riparian zone would be rated as heavy disturbance.

A subjective characterization of the level of development at the site and the overall aesthetic quality is required. Rate each of these attributes on a scale of 1 to 5. For development, assign a "5" rating if it is pristine, with no signs of any human development; assign a rating of "1" if the river at the site is totally developed (e.g., the river bank is lined with houses, or the riparian zone is otherwise completely developed). For aesthetic quality, base your decision on features of the site that reduce your enjoyment of the site (e.g., trash, foul odors, rip rapping, development). Also, rate the presence/absence of beaver activity, the dominant land use at the site, and the dominant age class of the riparian forest. Beaver activity may include bank lodges, felled trees, cuttings, dams in tributaries or backwaters, or actual sightings.

Table 7-11. Procedure for general site assessment.

1. Use your perceptions of the site obtained during the course of sampling to make a general assessment of the 1000-m reach centered on the cross-channel transect (i.e., the 500-m primary and secondary transects).

2. Rate each type of watershed activity or disturbance listed on the form as either not observed, low, moderate, or heavy on the channel and general assessment form (Figure 7-7). Ratings are subjective; extensive effort to quantify the presence and intensity of each disturbance is not required.

3. Assign a rating of 1 (highly disturbed) to 5 (pristine) based on your general impression of the intensity of impact from human disturbance. Also, assign a rating to the river based on overall aesthetic quality based on your opinion of how suitable the river water is for recreation and aesthetic enjoyment:

   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 Trashed; enjoyment nearly impossible.

4. Rate the presence of beaver activity at the site from 5 (intense) to 1 (absent). Beaver activity may include bank lodges, felled trees, cuttings, dams in tributaries or backwaters, or sightings.

5. Determine the dominant land use at the site. Pick **one** land use from among forest, agriculture, range, urban, and suburban/town. If there are other major land uses, make a note of them in the general assessment section of the form. If forest is the dominant land use, make a guess at the dominant age class of the forest (0-25, 25-75, or >75 years).

7.6 QA Considerations for riparian habitat sampling

A great variety of conditions among sites, plots, and subplots may be encountered in riparian sampling. Standardization, training, and crew specialization enhance the QA of riparian sampling. Some QA considerations for riparian habitat sampling are provided in Table 7-12.

Table 7-12. QA considerations for riparian habitat sampling.

- Attempt to expend equal effort for each plot and subplot despite variation in access, vegetation cover, and general sampling ease.
- Practice calibrating visual estimates of cover, distances, stem diameters, plot dimensions, etc. prior to actual data collection.
- Collect specimens of possible invasive species to show to a botanist when identification is uncertain.
- Use the "way-point averaging" function on the handheld GPS to acquire the most accurate site coordinates.
- To minimize intra-crew variability, crew members should specialize in tasks as much as possible (e.g, cover estimates, invasive species identification)
- For the shoreline type and macrohabitat estimates, be sure that all of the intervening shoreline between bank stations has been observed. Use the shoreline stations at 50-m intervals as a distance calibration for estimating percentages of shoreline in each type or habitat category.
- Conduct the general assessment for the whole site after all other sampling activities to insure a comprehensive perspective.

7.7 Equipment and supplies

Table 7-13 lists the equipment and supplies required to conduct all the activities described for characterizing riparian and bank physical habitat. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

Table 7-13. Equipment and supplies for sampling riparian habitat.

Qty	Item	
1	Surveyor's telescoping leveling rod	
1	Clinometer (or Abney level) with percent and degree scales (optional)	
1	Convex spherical canopy densiometer (Lemmon Model B), modified with taped "V"	
1	Bearing compass (backpacking type)	
2 rolls	Biodegradable surveyor's plastic flagging (2 colors)	
1	Digital camera	
1	Extra memory card for digital camera	
1	100 m fiberglass tape	
1	Meter stick for bank angle measurements	
1	Laser rangefinder with $\geq$1000-m range for channel width measurements	
1	Laser hypsometer (e.g., Opti-logic LH series) for bank and tree height measurements	
1 set	Laminated invasive-species reference guides	
1 set	Riparian habitat forms	

7.8 Literature cited

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Lemmon, P.E. 1957. A new instrument for measuring forest overstory density. Journal of Forestry 55:667-669.

Sappington, L., D. Dieterman, and D. Galat (editors). 1998. Missouri River Benthic Fish Consortium 1998 Standard Operating Procedures to Evaluate Population Structure and Habitat Use of Benthic Fishes along the Missouri and Lower Yellowstone Rivers. USGS Biological Resources Division, Columbia Environmental Research Center, Columbia, MO.

Ringold, P., S. Cline, M. Kentula, M. Bollman, T. Magee, S. Bryce, and P. Lattin. 2001. Draft Research Plan, EMAP Western Pilot Riparian Investigations in the John Day and Lower Deschutes Basins of Eastern Oregon. U.S. Environmental Protection Agency, Corvallis, OR. Draft.

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EMAP-GRE BANK AND SHORELINE MACROHABITAT FORM

Reviewed by (Initials): ________

SITE ID: GRW04449- ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 DATE: ___ / ___ / 2 0 0 _

Shoreline Station Location; Bank and Channel Width Characteristics							
Station	Latitude	Longitude	Wetted Width (m)	Bank Height (m)	Bank Width (m)	Bank Angle (circle one)	Flag
A	__ . ______	__ . ______				F G S V O	
C	__ . ______	__ . ______					
E	__ . ______	__ . ______				F G S V O	
G	__ . ______	__ . ______					
I	__ . ______	__ . ______					
K	__ . ______	__ . ______				F G S V O	

Shoreline Type Between Stations (% by type - sums to 100)					
	0-100 A - C	100-200 C - E	200-300 E - G	300-400 G - I	400-500 I - K
Stable Bank					
Aggrading Bank					
Eroding Bank					
Riprap					
Other revetment (Explain in comments)					
Other (Explain in comments)					
Flag					

Determining Bank Angle

F = Flat (<5°)
G = Gradual (5 - 30°)
S = Steep (30 - 75°)
V = Very steep (75 - 90°)
O = Overhung (>90°)

Macrohabitat Type Between Stations (% by type, may sum to > 100)					
	0-100 A - C	100-200 C - E	200-300 E - G	300-400 G - I	400-500 I - K
Channel cross-over (straightaway)					
Outside bend					
Inside bend					
Pool					
Tributary mouth < 5m wide					
Connected secondary channel < 5m wide					
Unconnected secondary channel (backwater) < 5m wide					
Other (Explain in comments)					
Flag					

Flag	Comments

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

21.

Figure 7-3. Bank and shoreline macrohabitat form (one per site).

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EMAP-GRE RIPARIAN CLASSIFICATION AND HUMAN INFLUENCE

Reviewed by (Initials): ______

SITE ID: GRW04449-______ DATE: ___/___/ 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 STATION: ☐ A ☐ E ☐ K

Land Use and Land Cover Classification

Subplot	Land Cover (circle one)	Flag
1	B D A X HM SM F OT	
2	B D A X HM SM F OT	
3	B D A X HM SM F OT	

Subplot 1, river edge (0 m) Distance to wetted edge (m)		Subplot 2, (20 m) land-side edge	

Canopy Densiometer (0-17)

	Landward	Downriver	Upriver	To. river	Flag
River edge (0 m)					
Subplot 2, land-side edge (20 m)					

Latitude	Longitude	Flag
___ . ______	___ . ______	

Human Influence (circle Yor N)

	Opposite side of river		In river		Between subplot 1 and river		Riparian Plot		Outside of plots, MCS side of river	
Dikes, riprap, revetments	Y N		Y N		Y N		Y N		Y N	
Locks, dams	Y N		Y N		Y N		Y N		Y N	
Buildings - industrial/commercial	Y N		Y N		Y N		Y N		Y N	
Buildings - residential	Y N		Y N		Y N		Y N		Y N	
Pavement	Y N		Y N		Y N		Y N		Y N	
Roads or rails	Y N		Y N		Y N		Y N		Y N	
Outlet pipes	Y N		Y N		Y N		Y N		Y N	
Docks/marina	Y N		Y N		Y N		Y N		Y N	
Trash	Y N		Y N		Y N		Y N		Y N	
Parks or lawns	Y N		Y N		Y N		Y N		Y N	
Row crops	Y N		Y N		Y N		Y N		Y N	
Grazing	Y N		Y N		Y N		Y N		Y N	
Recent logging	Y N		Y N		Y N		Y N		Y N	
Mining	Y N		Y N		Y N		Y N		Y N	
Other (Flag & explain)	Y N		Y N		Y N		Y N		Y N	
Other (Flag & explain)	Y N		Y N		Y N		Y N		Y N	

Land Cover Codes
B = Bare, un-vegetated, non-forest, non-agriculture, undeveloped
D = Developed, urban
A = Agriculture
X = Xeric (upland) shrubs or herbaceous
HM = Mesic (riparian) herbaceous
SM = Mesic (riparian) woody shrubs
F = >10% large tree cover
OT = Other (explain in comments)

Vegetation Coverage Codes
0 = Absent
1 = Sparse (<10%)
2 = Moderate (10-40%)_
3 = Heavy (40-75%)
4 = Very heavy (>75%)

Woody Vegetaton Type Codes
D = Deciduous
C = Coniferous
M = Mixed
N = No woody vegetation

Visual Estimates of Vegetation Coverage (circle one in each group)

	Subplot 1 (0 - 10 m)	Flag	Subplot 2 (10 - 20M)	Flag	Subplot 3 (20-30M)	Flag
Canopy (>5 m tall)						
Woody Vegetaton Type	D C M N		D C M N		D C M N	
Big trees (>0.3 m DBH)	0 1 2 3 4		0 1 2 3 4		0 1 2 3 4	
Small trees (<0.3 m DBH)	0 1 2 3 4		0 1 2 3 4		0 1 2 3 4	
Understory (0.5 to 5 m tall)						
Woody Vegetaton Type	D C M N		D C M N		D C M N	
Woody shrubs and seedlings (<0.1 m DBH)	0 1 2 3 4		0 1 2 3 4		0 1 2 3 4	
Herbaceous	0 1 2 3 4		0 1 2 3 4		0 1 2 3 4	
Ground Cover (<0.5 m tall)						
Woody Vegetaton Type	D C M N		D C M N		D C M N	
Woody shrubs and seedlings (<0.1 m DBH)	0 1 2 3 4		0 1 2 3 4		0 1 2 3 4	
Herbaceous	0 1 2 3 4		0 1 2 3 4		0 1 2 3 4	
Bare ground	0 1 2 3 4		0 1 2 3 4		0 1 2 3 4	

23.

Figure 7-4. Riparian classification and human influence form (one per station).

10457

EMAP-GRE RIPARIAN LEGACY TREE AND INVASIVE PLANTS

Reviewed by (Initials): ________

SITE ID: GRW04449-________ DATE: ___ / ___ / 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 STATION: ☐ A ☐ E ☐ K

Legacy Tree		Flag
Estimated DBH (m)	☐ <0.1 ☐ 0.1-0.3 ☐ 0.3-0.8 ☐ 0.8-2.0	
Estimated Height (m)	☐ <5 ☐ 5-15 ☐ 15-30	
Tree Type	☐ C ☐ D ☐ N	
Est. Distance to Main Channel Wetted Width (m)		

Cottonwood/poplar	☐	Unknown deciduous	☐
Ash	☐	Unknown conifer	☐
Maple/boxwood	☐	Unknown	☐
Sycamore	☐	Snag (dead tree)	☐
Willow	☐	Other (describe in comments)	☐
Pine	☐	Flag	

One legacy tree for each riparian plot, subplots combined; search area 100 meters from wetted channel.

Tree Type
C = Coniferous
D = Deciduous
N = No legacy tree

Invasive Plants: Common Name	Subplot 1	Subplot 2	Subplot 3	Outside Plots	Flag
Canada thistle	☐	☐	☐	☐	
Musk thistle	☐	☐	☐	☐	
Leafy spurge	☐	☐	☐	☐	
Russian-olive	☐	☐	☐	☐	
Salt cedar	☐	☐	☐	☐	
Buckthorn	☐	☐	☐	☐	
Reed canarygrass	☐	☐	☐	☐	
Giant reed	☐	☐	☐	☐	
Cheatgrass	☐	☐	☐	☐	
Teasel	☐	☐	☐	☐	
Common burdock	☐	☐	☐	☐	
Japanese knotweed	☐	☐	☐	☐	
Mile-a-minute	☐	☐	☐	☐	
Garlic mustard	☐	☐	☐	☐	
Purple loosestrife	☐	☐	☐	☐	
Knapweed	☐	☐	☐	☐	
Whitetop	☐	☐	☐	☐	
NO INVASIVE SPECIES	☐	☐	☐	☐	

Flag	Comment

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

24.

Figure 7-5. Riparian legacy tree and invasive plants form (one per station).

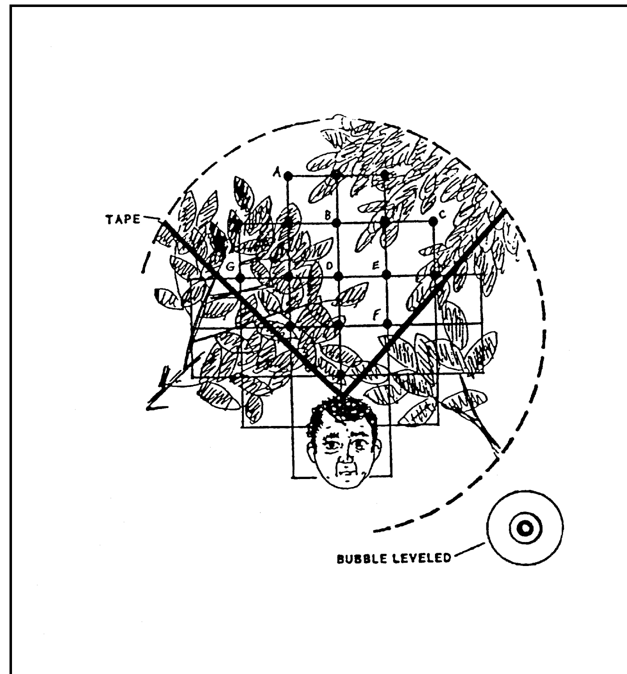

Figure 7-6. Use of the modified convex spherical canopy densiometer. In this example, 11 of the 17 intersections show canopy cover, giving a densiometer reading of 11. In heavy cover, it is easier to count the clear intersections and subtract from 17. Note proper positioning with the bubble leveled and face reflected at the apex of the "V."

EMAP-GRE CHANNEL AND GENERAL ASSESSMENT FORM (front)

SITE ID: GRW04449-______ DATE: ___/___/ 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 Reviewed by (Initials): ______

Channel Form and Constraint in Reach 500 Meters up and downriver from X-site

Channel Pattern (check one) FLAG

☐ Single Channel ☐ Anastomosing Channel (long 2ndary channels) ☐ Braided Channel (short 2ndary channels) ☐ Other (Flag and explain in comments)

Channel Constraint (check one) FLAG

☐ Channel very constrained in V-shaped valley (unlikely to flow overbank or cut new channels in flood) ☐ Channel in broad valley but incised (unlikely to flow overbank or cut new channels in flood) ☐ Channel in narrow valley but very constrained (valley width <10x bankfull width) ☐ Channel is unconstrained in broad valley (floodplain present) with levee flood control ☐ Channel is unconstrained in broad valley (floodplain present) with no levee flood control

Constraining Features (check one) FLAG

☐ Bedrock (channel in bedrock dominated gorge) ☐ Hillslope (channel in a narrow V-shaped valley) ☐ Terrace (channel constrained by incision) ☐ Modified (channel constrained by riprap, or other revetment) ☐ No constraining features (channel migration possible)

		FLAG				FLAG
Percent of channel constrained (0-100%)			Channel and Valley Width Measurements	Estimated valley width (m)		
Bankfull width (m)				Estimate of valley width not possible (check box)	☐	

Overall site characteristics in Reach 500 Meters up and downriver from X-site (check one in each row) FLAG

								FLAG
Development	Pristine	☐ 5	☐ 4	☐ 3	☐ 2	☐ 1	Highly Disturbed	
Aesthetics	Appealing	☐ 5	☐ 4	☐ 3	☐ 2	☐ 1	Unappealing	
Beaver Activity	Intense	☐ 5	☐ 4	☐ 3	☐ 2	☐ 1	Absent	
Dominant Land Use at MCS (pick one only)	☐ Forest	☐ Agriculture	☐ Range	☐ Urban	☐ Suburban			
Dominant Forest Age Class	☐ 0-25 years	☐ 25-75 years	☐ >75 years					

WATERSHED ACTIVITIES AND DISTURBANCES OBSERVED IN REACH 500 METERS UP AND DOWNRIVER FROM X-SITE FLAG

Residential	Recreational	Agricultural	Industrial	Stream Management
N L M H Residences	N L M H Hiking Trails	N L M H Cropland	N L M H Ind. Plants	N L M H Angling Pressure
N L M H Lawns	N L M H Parks	N L M H Pasture	N L M H Mines/Quarries	N L M H Dredging
N L M H Construction	N L M H Camping	N L M H Livestock Use	N L M H Oil/Gas Wells	N L M H Channelization
N L M H Pipes, Drains	N L M H Trash/Litter	N L M H Orchards	N L M H Pwr Plants	N L M H Water Level Fluc.
N L M H Dumping	N L M H Boats/PWC	N L M H Poultry	N L M H Recent logging	N L M H Fish Stocking
N L M H Roads	N L M H Swimming	N L M H Irrigation	N L M H Odors	N L M H Dams
N L M H Bridge/Culverts	N L M H Fishing	N L M H Water With.	N L M H Com. Use	N L M H Riprap
N L M H Sewage trt.	N L M H Other (Flag and explain)	N L M H Other (Flag and explain)	N L M H Other (Flag and explain)	N L M H Other (Flag and explain)
N L M H Other (Flag and explain)				

(Disturbance Intensity: N=Not observed, L=Low, M=Moderate, H=Heavy)

FLAG	COMMENTS

Flag codes: K=no measurement made, U=suspect measurement;F1, F2, etc=misc flags assigned by field crew. Explain in comments.

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22.

Figure 7-7. Channel and general site assessment form (one per site).

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Section 8
Fish

Erich B. Emery[1], Jeff A. Thomas[1], Mark Bagley[2], and Ted R. Angradi[3]

EMAP-GRE fish sampling methods are designed to collect all but the rarest fish inhabiting the near-shore habitat at a site. The sample collected is assumed to accurately represent the proportional abundance of the littoral fish assemblage at the site. Fish sample data include species composition, and the size and condition of individual fish. Other measures of assemblage structure and function can be calculated from the data and combined into indices of biotic condition potentially useful for assessing the condition of Great Rivers (Simon and Emery 1995, Emery et al. 2003).

Fish assemblage data are collected by electrofishing with a three-person crew during the day. A subsample of fish are retained for analysis of tissue contaminants (Section 9). After electrofishing, the crew collects fish habitat data. The procedures in this section are substantially revised from previous EMAP fish sampling methods (Peck et al., unpublished drafts). Habitat sampling methods are based on ORSANCO methods.

8.1 The electrofishing transects

Upon arriving at the site location, the fish-sampling crew flags the primary and secondary 500-m MCS transect at 100-m intervals (if not already flagged by the river-sampling crew). Fish are sampled by daytime electrofishing along the two 500-m shoreline transects. The primary transect extends upriver from the intersection of the cross-channel transect and the target shoreline (river right or river left) identified in the design file (see Figure 4-1). The secondary transect extends downriver from the intersection of the cross-channel transect and

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1 Ohio River Valley Water Sanitation Commission (ORSANCO), 5735 Kellogg Avenue, Cincinnati, OH 45228

2 U.S. Environmental Protection Agency, Office of Research and Development, National Exposure Research Laboratory, Ecological Research Division, 26 Martin Luther King Dr., Cincinnati, OH 45268

3 U.S. Environmental Protection Agency, Office of Research and Development, National Health and Environmental Effects Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804

the target shoreline (unless it has been adjusted as described in Section 4).

The shoreline electrofishing zone extends out from shore to a depth of 6 m (20 ft) or a distance of 30 m (100 ft), whichever is closer to the shore. Electrofishing is conducted for a minimum of 1800 seconds (0.5 h) of total shock time to collect fish from the designated zone. Increased shock time will be necessary to fish shorelines with abundant cover.

8.2 Electrofishing

8.2.1 Boat specifications and electrode configuration (recommended)

The standard EMAP-GRE electrofishing boat is a modified 5.5-m aluminum jon boat with a welded-aluminum hull. The boat is equipped with a 90-hp outboard motor for river navigation. An auxiliary 25-hp motor is mounted starboard of the main motor, just behind the driver. The smaller motor is used to maneuver the boat during electrofishing; it allows operation at slower speeds and in shallower water than the main motor. Other configurations are permissible.

A generator supplies power to a control box, which in turn controls the electrical field configuration. A single boom extends 2.5 - 3.0 m from the front of the boat with a single anode dropper affixed to the end of the boom. The boat’s hull serves as the cathode. There are 3 “kill” switches on board. There is a kill switch on the control box which shuts off all power coming from the box, there is a positive-pressure kill switch operated by a foot pedal mounted on the front deck, and there is a hand-held switch operated by the driver during operation of the electrofishing equipment. All 3 switches must be “on” in order to activate the electric field. This ensures redundancy within the electrical safety system: the driver and one crew member can both kill the electricity from the generator in an unsafe situation.

8.2.2 Electrofishing procedures

Before starting the electrofishing run, all safety "kill" switches should be tested by starting the generator, turning all switches to the "on" position, and then throwing each switch to the "off"' position to make sure each is working properly.

Electrofishing should not begin until 1000 h. Beginning at the upriver end of the primary 500-m MCS transect, the driver maneuvers the boat downriver parallel to the shoreline (Figure 8-1). The two other crew members stand in the bow of the boat and net all fish that are stunned. Stunned fish are placed in an aerated live well. Voltage and amperage adjustments may be necessary to ensure that a minimum of 3000 watts of output power are maintained at all times. It may be necessary to adjust power based on sampling effectiveness and incidental fish mortality. Trained crew members should be able to determine whether insufficient or excessive power is being used.

During the electrofishing run, the boat is navigated through the shoreline zone at a speed sufficiently slow to allow the fish netters to recover all stunned fish, including small fish, such as darters, which are difficult to see and do not always rise up off of the bottom when stunned. The boat should be moved in a serpentine fashion parallel to the shoreline, ensuring that the electrical field is passing over the shallow littoral areas as well as over the deeper channel margin, and ensuring that as much of the zone as possible is transected by the path of the field. The path of the boat (Figure 8-1) should be analogous to the motion of a person using a metal detector: a side-to-side path with complete lateral coverage and a slow forward pace.

Care should be taken to thoroughly work the electric field around objects such as snags, downed trees, piers, boulders, and other potential fish cover until each object yields no more fish. The field may have to be held over the structure for a few seconds to allow the fish to wriggle out of the cover and up into the field. The minimum electrofishing time for each transect is 1800 seconds of shock time. Along shorelines with swift current and/or little cover, it may be necessary to electrofish the transect twice to achieve the minimum shock time. There is no upper limit for electrofishing time. Electrofishing procedures are described in Table 8-1.

A large live well (≥ 300 L) should be used to ensure adequate holding capacity for all the fish collected in the 500-m transect. A strong and reliable aerator should be used to maintain oxygen levels in the tank. If an excessive number of fish are captured, it may be necessary to change the water in the live well during the run. Usually this is done after the electrofishing run has been completed, just prior to processing the fish. Fish that appear overly stressed as indicated by loss of righting response should be processed immediately and released. Individuals returned to the water during the electrofishing run should be released behind the boat and in deeper water, to ensure that they are not recaptured. At the completion of each 500-m electrofishing run, the crew leader records the end time and the total shock time, in seconds, on the fish sampling form (Figure 8-2).

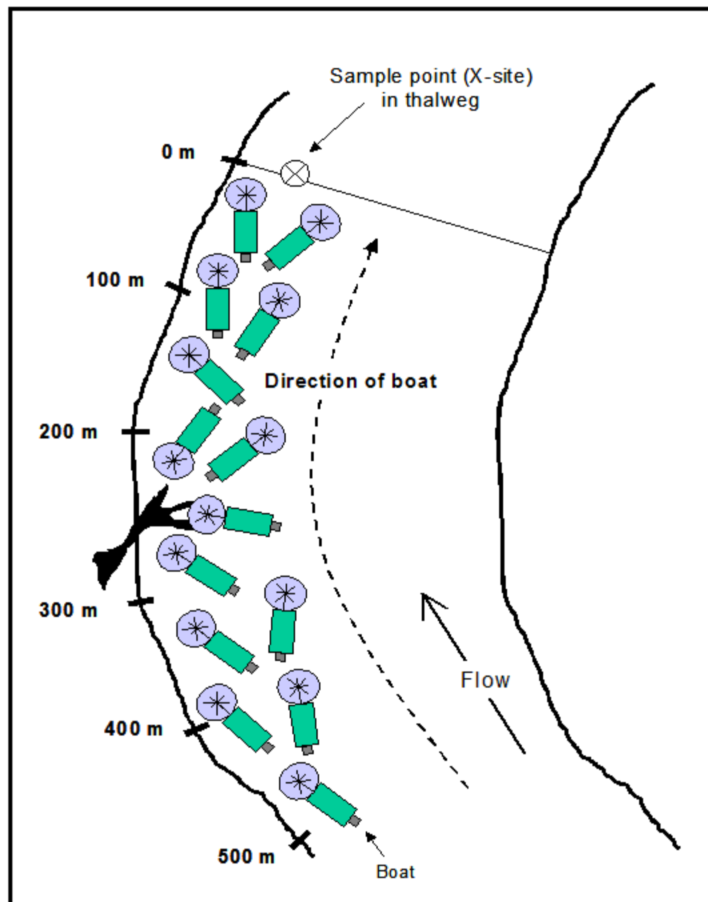

Figure 8-1. Recommended path of electrofishing boat showing equal coverage of shoreline and channel lateral margins as well as complete application of the field through, over or around cover objects. The zone should not extend greater than 30 m from shore, or to a depth greater than 6 m. Not to scale.

Table 8-1. Electrofishing procedures.

1.	Complete the header information on the fish sampling form (Figure 8-2) including site ID, date, coordinates of transect starting point at downriver end, transect (primary or secondary), target shoreline, and transect length. **Use a different fish sampling form for the primary and secondary transects**.
2.	Obtain the Secchi depth from the river-sampling crew, if possible. Otherwise measure Secchi depth (see Table 5-3)
3.	Navigate to the upriver end of the electrofishing transect and make all necessary electrical connections. Extend and secure the boom, fill the live well, and turn the aerator on.
4.	At a location outside the sampling zone, test the electrofishing unit and kill switches. Using pulsed DC, adjust voltage and amperage to maintain a minimum power output of 3000 watts. Make voltage and amperage adjustments to ensure that fish are being rolled easily, that smaller fish such as darters are effectively stunned, and that fish are not being injured. Record power output data on the form (volts, watts, amps, pulse rate, pulse width).
5.	Record the begin time on the fish sampling form and begin electrofishing. From the top of the zone, proceed slowly downriver, following a serpentine path parallel to the shoreline (Figure 8-1). Attempt to net all stunned fish. Avoid netting bias toward larger individuals. Do not attempt to fish in water deeper than 6 m (20 ft). Stay close to shore and fish the shallower margins. If the water is generally shallower than 6 m, the path of the boat should extend out into the channel no more than 30 m (100 ft) from shore. Carefully maneuver the boat around instream cover, fishing slowly to ensure that the cover is yielding no more fish before moving on.
6.	Attempt to fish the transect as thoroughly as possible, but do not place the crew in danger in order to fish particular habitats. Safety is the first concern. If part of the transect cannot be fished safely, note this in a comment on the form.
7.	At the end of the sample transect turn off the electrofishing gear and record end time and total shock time (Figure 8-2).
8.	The minimum electrofishing time for each transect is 1800 seconds of shock time. Along shorelines with swift current and/or little cover, it may be necessary to electrofish some or all of the transect twice to achieve the minimum shock time. There is no upper limit for electrofishing time.
9.	After processing the sample, repeat steps 1-7, as appropriate, for the secondary 500-m transect.

8.3 Sample processing

Sample processing includes identifying fish to species, examining them for external anomalies, measuring and weighing, preserving small specimens for later processing, photographing voucher specimens, and selecting specimens to be retained for tissue contaminant analysis. At two sites sampled in each year, fish-fin tissue is collected for DNA analysis. Fish are recorded by their complete American Fisheries Society (AFS) common name after Nelson et al. (2004). For processing, one crew member records data on the fish sampling form (Figure 8-2) while the other crew members sort, identify, weigh, and measure fish. After they are processed, fish from the primary transect should be released where they will not be recollected from the secondary transect.

All small specimens (<12 cm) and specimens that cannot be identified with certainty in the field should be preserved in formalin for later processing by the sampling crew. Preserved specimens will eventually be deposited in a museum collection. **With the exception of small fish collected for DNA analysis, which must be preserved in 75% ethanol, all fish will be preserved in 10% buffered formalin.** Fish sample processing procedures are described in Table 8-2.

Table 8-2. Fish sample processing procedures.

1. If the handling of threatened or endangered fishes is permitted under the collecting permit, they should be processed first in order to expedite their return to the water. Otherwise they should be released immediately.

2. For each larger specimen that can be identified to species and accurately weighed in the field, record the complete AFS common name (Nelson et al. 2004). All small specimens (<12 cm) and specimens that cannot be identified to species with certainty in the field should be preserved in one or more jars as a field composite and retained by the crew for later laboratory processing (fish voucher procedures described in Table 8-3). Do not record data for these specimens in the field. Potential small voucher specimens should be preserved in the same containers as the composite fish sample.

3. At the first or second and last site sampled in 2005, fish tissue for DNA analysis will be collected. These methods are described in Table 8-6.

4. Select and retain fish for tissue analysis (see Section 9).

5. Examine each fish for DELTs (deformities, erosions, lesions, and tumors). Record the presence of DELTS on an individual fish or among a batch of small fish using the codes in Table 8-4. Other abnormalities (e.g., blind eyes, pop-eye, fungus) can be recorded using flags.

6. Using a measuring board marked with 3-cm size classes, record fish length by size class (e.g., fish $\leq$ 3 cm long are in size class 1, fish >3 and $\leq$ 6 cm are in size class 2, etc.

7. Record the weight of each fish in kg (1g = 0.001kg). Fish too small to be weighed should be retained for laboratory processing. Be sure to release fish in a location where they will not be recollected during sampling of the secondary transect.

8. In the laboratory, sort the preserved composite sample to species. Refer unknowns to a taxonomic expert for identification (no expert is specified by EPA – crews may use any qualified ichthyologist). Process the specimens as in the field and record the data on the original field forms. Fish too small to weigh individually may be weighed as a batch. Extract and label individual species voucher specimens (Table 8-3). Do not discard the processed specimens. They should be retained in the original jars with fresh formalin as a composite voucher sample to be deposited in a museum collection.

9. Field forms should be completed by the field crews. All fish should be identified to species and weighed before the forms are sent to the EMAP data center.

8.3.1 Unknowns and voucher specimens

Each crew member should be familiar enough with the large-river fish assemblages in the region to identify most larger specimens. A professional ichthyologist familiar with the fish species of the region should perform final identifications of unknowns. Obtaining identifications of unknowns is the responsibility of the crews, but MED can facilitate identifications if necessary.

Vouchers, in the form of a photograph (Figure 8-6 is an example) or as a preserved specimen, should be retained as a reference for every species allowable under the collecting permits. Each fish-sampling crew should photograph or collect one voucher specimen for each different species encountered each year. **All questionable fish and all small fish (<12 cm) should be preserved at every site as a field composite voucher for later laboratory identification by the fish-sampling crew**. Table 8-3 describes preparation of photo vouchers and preserved specimen vouchers. Large species can usually be adequately documented by a digital photograph.

An effort should be made to document with a photo or by collection any known or suspected non-indigenous exotic or invasive fish species. The collecting permit may specify that certain species not be returned alive to the water. A spatially-referenced and frequently-updated database of non-indigenous fish species of the U.S. can be searched at http://nas.er.usgs.gov/. Fish-sampling crews should be familiar with the potential and reported non-indigenous species in the river and regions they are sampling. Collections of non-indigenous fishes made during EMAP-GRE sampling should be submitted by fish-crew leaders to this database via the above web address.

Table 8-3. Voucher specimens. Note: these methods are modified at fin-tissue collection sites.

1. There are three types of voucher samples: 1) preserved field composite samples that include questionable specimens and all small fish, 2) preserved individual species vouchers that are extracted from the field composites, and 3) photo vouchers for larger fish. A preserved individual species voucher or photo voucher should be retained for every species captured by every fish-sampling crew each year. All fish retained in the field composite are tracked with a single Sample ID in the header of the fish sampling form (Figure 8-2; multiple jars may be used).

2. **Preserved field composite voucher**. Small specimens (<12 cm) and unknowns to be retained for laboratory processing should be euthanized with a humane method. Euthanized specimens are placed in a leakproof plastic jar(s) with a screw top with 10% buffered formalin (see Table 3.1). Do not cram fish into jars so that they are fixed in a bent position; they should float freely in the jars. Fish biomass should not exceed 40% of the container contents by weight. Fish $\geq$12-cm long should be slit open in the lower right abdomen to promote preservation.

3. Fill out a fish voucher label for the jar (Figure 8-7) with EMAP-GRE site number, date, visit number and jar number. Circle “10% Formalin.” Transfer the sample ID number from the label to the header of field form (Figure 8-2) and the MED tracking form (Sample type CV; Figure 3-2). If >1 jar is needed, fill out a continuation label including the sample ID from the original fish voucher label. Add sufficient formalin to each jar to cover the fish. Place the voucher label(s) on the jar(s) and cover with clear tape.

4. **Preserved individual species vouchers** (optional). Specimens may be extracted from the preserved field composite to become individual species vouchers. Fish of the same species should be placed in a leakproof plastic jar(s) with a screw top. Do not cram fish into jars so that they are fixed in a bent position; they should float freely in the jars. Fish biomass should not exceed 40% of the container contents by weight. Fish $\geq$15-cm long should be slit open in the lower right abdomen to promote preservation.

5. Fill out a <u>new</u> fish voucher label for the jar (Figure 8-7) with EMAP-GRE site number, date, visit number and jar number. Circle “10% Formalin.” Transfer the sample ID number from the label to the MED tracking form (Sample type FS; Figure 3-2) and to the field form. Add sufficient formalin (See Table 3.1) to each jar to cover the fish. Place the voucher label(s) on jar(s) and cover with clear tape.

6. **Photo vouchers**. Place the fish on the measuring board. Place a completed Photo Fish Voucher Label (Figure 8-7) below the fish with the presumed common name of the fish, EMAP-GRE site number, transect, and date. Use a digital camera to take a high-resolution picture of the fish. Check the quality of the image before releasing the fish (Figure 8-6). Record the image file name on the form as a flag comment after the camera images have been downloaded from the camera. Save the image files to a folder named with the site number. Back up the image files as soon as possible. If you rename the image files in the office, be sure to provide the new name on the form.

8.3.2. External examination for anomalies

During processing in the field or in the laboratory, both sides of each fish should be examined for external anomalies. Readily-identified external anomalies include deformities; erosion of the fins, barbels, and gill covers; lesions; and tumors. Photographs of each type of anomaly are shown in Figure 8-5 (from Moulton et al. 2002). Smith et al. (2002) has additional guidance for assessing anomalies. Codes for each type of anomaly are given in Table 8-4.

Table 8-4. External anomaly codes (DELTs).

Category	Code	Description
Deformities	DE	Skeletal anomalies of the head, spine or body shape
Erosion	ER	Eroded barbels, fins, or gill covers; substantial fraying or reduction
Lesions	LE	Open sores or exposed tissue; raised warty outgrowths
Tumors	TU	Areas of irregular cell growth which are firm and cannot be broken open easily (masses caused by parasites can be broken open easily)
Other	OT	Flag and describe in a comment

8.3.3 Length and weight measurements

Procedures for recording length and weight measurements are presented in Table 8-2. Total length (Figure 8-6) is used to determine the 3-cm size class to which each fish belongs. Weights are taken using a spring-dial or digital scale and recorded to the nearest gram (0.001 kg). Lengths and weights are recorded on individual lines on the fish sampling form (Figure 8-2). Fish too small to be weighed individually can be grouped into 3-cm size classes and weighed as a batch.

8.3.4 Fish fin-tissue samples for DNA analysis

At two sites per year, fish-fin tissue is collected by caudal punch or scissor clip (Table 8-5). DNA will be extracted from this tissue for genetic analysis. The findings will be used to check identifications of specimens, examine genetic variation within species, and identify hybrids. At fin-tissue collection sites, the first 20 large specimens of each species are fin punched/clipped and photographed. All small fish at these sites will be preserved as a

composite sample in 75% ethanol, and processed (identified, examined for anomalies, measured, and weighed) in the lab as usual. The specimens are then be sorted into species and preserved in separate jars (one jar per species). The jars are labeled and shipped to NERL at the end of the season for fin-tissue sampling and DNA analysis.

In addition to the regular sampling of fin-tissue for DNA analysis, crews may collect additional fin-tissue samples of unknown or unusual specimens at any site and submit them for DNA analysis using the same sample tracking procedures. These specimens must be preserved in ethanol for DNA analysis. For questions specific to the DNA analysis, contact Mark Bagley (513-569-7455).

Table 8-5. Processing fish for fin-tissue DNA analysis.

1.	At two sites each year DNA tissue is check “DNA sample site?” box at the top of the fish sampling form (Figure 8-2). Do not use the same site (due to revisits) for both samples.
2.	**Large fish**. The first 20 large fish of each species (>12 cm; size class 5 or larger; **including** fish retained for fish tissue contaminants – Section 9) across both electrofishing transects at DNA sample sites should be identified and data should be recorded as usual (Table 8-2). For each fish, collect a punch or clip of the caudal (tail) fin that is 0.5-1.0 cm^2 in area. It may work best to punch the fish while it is held in the net. Keep a running count so that not more than 20 fish of each species (across transects) are punched. It is acceptable for all DNA samples to come from the first transect.
3.	Place the fin-tissue sample in a #1 paper coin envelope (provided by EPA). Fill out and affix an adhesive Fish Tissue DNA sample label (Figure 8-7) to the coin envelope. Pre-labeling the coin envelopes with blank sample labels is recommended.
4.	Place the labeled coin envelope next to the fish and take a digital photograph. Release the fish unless they are to be retained for a fish tissue contaminant sample.
5.	Repeat for each large fish of each species in the sample (maximum of 20 per species per site). Between fish, rinse hands and punch/scissors by vigorous agitation in river water.
6.	Transfer the sample ID from the label to the field form and the NERL tracking form for each sample (sample type DS). Use as many NERL tracking forms as necessary.
7.	Download and back up digital images which will be transferred to EPA at the end of the season. Record the image file name (and common name) on the NERL sample tracking form.

Continued

Table 8-5. Processing fish for fin-tissue DNA analysis, continued.

8.	Place all the sample envelopes for a site in a zip-lock plastic bag labeled with the site ID and refrigerate. Ship the samples weekly to NERL along with the fish tissue contaminant samples (Section 9).
9.	**Small fish**. All small fish (<12 cm; size class 4 or smaller; **excluding** fish retained for fish tissue contaminants – Section 9) should be preserved as a composite sample in 85% ethanol. In the lab, the fish should be processed as usual (Table 8-2). After the data are recorded, the fish should be sorted to species and each species placed in a separate jar with new 75% ethanol. **Note: If more than a week elapses between sample collection and lab processing, replace the ethanol in the jar(s) with new 75% ethanol**.
10.	Fill out and affix an adhesive Fish Tissue DNA label on each jar and cover with clear tape. Seal the jar with plastic electrician's tape. Transfer the sample ID to the fish sampling form (Figure 8-2). Record the ID number <u>next to the first record for that species</u> that does not already have a sample ID (because it was also included in the large fish DNA sample).
11.	Transfer the sample ID and species common name to the NERL sample tracking form (sample type DS) and retain samples until they can be transferred to NERL.
12.	Note: In addition to the sampling of fin-tissue for DNA analysis at two sites in 2005, crews may collect additional fin-tissue samples of unknown or unusual specimens at any other site and submit them for DNA analysis using the same sample tracking procedures outlined above.

8.4 Fish habitat

After electrofishing and fish processing, the fish-sampling crew records physical habitat data for each 500-m shoreline transect. At six of the points marked on the shoreline (0, 100, 200, 300, 400, and 500 m from the downstream end; Figure 8-8), the crew records substrate composition at 3-m intervals out from the shoreline, or as close as the boat can get to the shoreline, to a distance of 30 m from shore. In addition to recording depth and substrate composition, the crew estimates the amount of fish cover for each 100-m segment of shoreline (Figure 8-8). Methods for quantifying fish habitat are described in Table 8-6.

Table 8-6. Fish habitat data collection.

1. Navigate to the site using GPS. Communicate with the river-sampling crew to obtain specific site information that may already be available (e.g., new site coordinates, hazards, Secchi depth, conductivity).

2. If not already marked, locate the downriver end of the 500-m MCS transect using the GPS coordinates from the design file, and flag it. Use the trip odometer on a hand held GPS or other method to mark off the transect in 100-m intervals to 500 m. The 0-m point is located at the downriver end of the transect, but the habit measurements can be made working downriver as long as the field forms are filled in correctly.

3. Fill in the header information on the fish habitat form (Figure 8-9), including the site ID, date, transect start coordinates (at downriver end), whether it is the primary (upriver) or secondary (downriver) transect, and the transect length.

4. Determine the channel morphology at the site and circle the appropriate macrohabitat (see Table 7-1).

5. At each of the six points located along the shoreline for each zone (at 0, 100, 200, 300, 400, and 500 m from the transect starting point), one crew member drops the weighted end of a 30-m floating rope on the shore at the water's edge or as close as the boat can approach the shoreline (which represents the inside margin of the electrofishing zone). The driver then slowly backs the boat away from shore in a line perpendicular to the shoreline. The crew member holding the rope slowly feeds out the line, keeping the rope tight without dislodging the weight at the shore end.

6. Record the bank substrate (-3 m DFS [distance from shore]) at 3-m DFS intervals, as indicated by marks along the floating rope. The person operating the pole probes the river bed several times and announces the depth (to nearest 0.25 m) and substrate(s). Substrate and depth are recorded to a distance of 30 m from shore (or where the weight on the habitat rope is dropped).

7. In areas of high current velocity, a hand-held GPS or a laser rangefinder may work better than the habitat rope for locating substrate probe locations.

8. Substrate composition is recorded as boulder, cobble, gravel, sand, fines, or hardpan, or multiple for each of the 72 points (12 points at 6 sites).

9. For each 100-m zone along the shoreline (e.g. 0-100 m, 100-200 m etc.) determine the percentage coverage category of each type of fish cover in the electrofishing zone. Circle the percent cover category for each type present on the form. Fish cover is anything that could provide cover for a fish. A uniform sand or gravel beach with no overhanging vegetation or undercut banks would provide 0% cover. For linear cover features (e.g., undercut banks) estimate the percent of the 100 m with the cover feature.

10. Repeat steps 2-9, as appropriate, for the secondary transect.

8.5 QA considerations for fish sampling

Crew members should be properly trained in techniques for operating the boat and electrofishing equipment. Proper use of the equipment, including maintaining the electrical field and maneuvering of the boat to optimize capture of fish, is critical to ensure that a representative sample is collected. QA of fish sample processing depends on correct identification of specimens. Crew members should have sufficient training to identify most fish that are collected. Questionable fish should be retained as voucher specimens. Table 8.7 provides some QA considerations for fish sampling.

Table 8-7. QA considerations for fish sampling.

- Sampling should probably not take place if Secchi depth is < 15 cm (6 inches) or if river stage is elevated > 0.5 m (20 inches) above normal levels. The decision to sample or not is up to the crew leaders.
- The transects should be fished **before** starting habitat data collection so that fish are not spooked from the shoreline.
- Electrofishing should not begin until 1000 h.
- Use a digital camera on a high resolution setting for taking photo vouchers.
- All small fish (minnows and questionable small specimens) should be retained as a preserved field composite voucher sample and processed in the lab by the field crew.
- Do not cram fish voucher specimens into jars. They should be free floating so they are not fixed in a bent position.
- Replace the ethanol in sample jars with fresh 75% ethanol if more than one week elapses between initial preservation and lab processing.
- Netters should wear polarized sunglasses.
- When netting shocked fish, avoid size bias.
- Avoid shorthand or local common names for fish. Nelson et al. (2004) is the standard.

8.6 Safety considerations for fish sampling

These rivers are large, navigable systems and are often congested by barge and recreational traffic. Extreme precautions should be taken when electrofishing, crossing the channel, and navigating to and from sampling locations. Primary responsibility for safety rests with the crew leader. However, each member of the three-person crew should be alert, aware of safety considerations (Table 8-89), trained to recognize safety concerns, and trained in first aid and CPR.

Table 8-8. Safety considerations for fish sampling.

- The electrofishing unit has a high voltage output and is capable of delivering a fatal shock.

- Large (>10 kg) silver carp (*Hypophthalmichthys molitrix*) can jump >2 m out of the water. People have been seriously injured by carp collisions. Silver carp are present in the lower reaches of all three GRE rivers. Be alert for jumping fish while running the river and during electrofishing.

- Crew members should be able to swim, and should receive CPR, first-aid, and safe boating training.

- The rivers sampled for this project are subject to heavy barge and recreational boating traffic. When navigating at night, running lights and a spotlight are required so that other vessels are aware of the boat and so the driver can more easily detect obstacles in the water.

- If the generator is running, do not touch the anode or cathode (if a cathode other than the boat hull is used). Do not touch objects outside the boat. Do not reach into the water. If doing so, make sure all electricity to the water has been turned off by ensuring that all three switches are in the “off” position (unit, pedal, and hand switch).

- Do not electrofish in high waves or other conditions that may cause sudden motions of the boat that can cause someone to lose their balance.

- Do not fish in the rain. Excessive water running from the deck of the boat into the water may create a path for current to follow from the water, up onto the deck. Prior to each sampling event, all electrical “kill” switches should be checked to ensure they are working properly.

- All members of the electrofishing crew should wear USCG-approved PFDs whenever in the boat.

- Good line of sight and communication should be maintained among crew members at all times. The generator is loud and often drowns out verbal communication. Hand signals should be used to communicate boat direction, power on/off, and other vital information.

- All crew members should know the location of the nearest hospital.

- Use caution around onboard gas tanks. Never refill the generator when it is hot. The generator exhaust gets extremely hot while in use. Caution should be used to ensure that no item is touching the exhaust and that all items near the exhaust are secured to ensure they do not shift position while underway and possibly come in contact with the exhaust.

- All electrical connections should be checked prior to use to ensure that proper, tight connections are maintained. Loose connections can cause sparking and fire.

- All crew members should know the on-board location of the cell phone, first aid kit, fire extinguisher, and truck keys.

8.7 Equipment and supplies

Table 8-9 is a checklist of the equipment and supplies necessary for fish sampling. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

8.8 Literature cited

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Table 8-9. Equipment and supplies for fish sampling. Standard boat safety gear is not listed. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5

Qty	Item	
1	Electrofishing anode and boom	
1	Electrofishing control box	
2	Extra boat batteries	
1	Digital camera with macro function, extra memory card	
1	Chainman hip chain with extra string (optional)	
1	Laser rangefinder (optional)	
2	Habitat rope (floating nylon marked in 3 m increments) and anchor (1 L bottle filled with quick-setting concrete with a loop of rope pushed into the wet concrete)	
2	Habitat pole (2 3-m sections of 3/4" copper pipe with a threaded coupling and caps on both ends) marked at 0.25 m intervals	
2 pr	Rubber gloves	
2	Non-conducting dip nets with 1/4" mesh	
1	Minnow net for dipping small fish from live well	
2	Measuring board with 3 cm size classes (see Figure 8-6)	
1	1-Kg scale (spring or electronic)	
1	12-Kg scale (spring or electronic)	
1	25-Kg scale (spring or electronic)	
2	Plastic weighing trays	
5 L	10% borax-buffered formalin	
2 pr	Single hole punch for collecting fin tissue DNA sample (scissors may substitute)	
1	Scalpel for slitting open fish before preservation.	
Several	#1 paper coin envelopes (**provided by EPA**)	
5 L	75% ethanol for preserving fish after lab processing (**2005 DNA sites only**)	
5 L	85% ethanol for preserving fish in field (**2005 DNA sites only**)	
Several	Leak-proof HDPE jars for fish voucher and DNA samples specimens (various sizes from 250 mL - 4 L)	
1	Secchi disk and marked line	
1 set	Fish ID keys	
1 set	Fish habitat and fish sampling forms	
1 set	Sample labels	

EMAP-GRE FISH SAMPLING FORM (front)

Reviewed by (Initials): ______

SITE ID: GRW04449- ______ DATE: ___ / ___ / 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 PAGE: 1 OF ____

Coordinates of Transect Starting Point						DNA SAMPLE SITE?: Yes ☐ No ☐	
Begin time (24h)	End time (24 hr)	Composite Sample ID		Number of Jars	MCS transect	Reach Length (m)	Flag
:	:				☐ Primary ☐ Secondary		
Total shock time (s)	Secchi depth (cm)	Volts	Watts	Amps	Pulse rate	Pulse Width (ms)	Flag
					○ pps ○ Hz		

SPECIES	DELTS 1	DELTS 2	COUNT	SIZE CLASS	WEIGHT (kg)	SAMPLE ID	FLAG

FLAG	COMMENTS

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

Draft

28.

Figure 8-2. Fish sampling form (front).

EMAP-GRE FISH SAMPLING FORM (back)

Reviewed by (Initials): ______

SITE ID: GRW04449- DATE: ___ / ___ / 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 PAGE: ___ OF ___

Sample ID: ______ MCS transect ☐ Primary ☐ Secondary

SPECIES	DELTS 1	DELTS 2	COUNT	SIZE CLASS	WEIGHT (kg)	SAMPLE ID	FLAG

FLAG	COMMENTS

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

Draft

29.

Figure 8-3. Fish sampling form (back).

EMAP-GRE FISH SAMPLING FORM (cont.)

Reviewed by (Initials): ______

SITE ID: GRW04449-______ DATE: ___/___/2 0 0_ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2 PAGE: ___ OF ___

Sample ID: ______ MCS transect ☐ Primary ☐ Secondary

SPECIES	DELTS 1	DELTS 2	COUNT	SIZE CLASS	WEIGHT (kg)	SAMPLE ID	FLAG

FLAG	COMMENTS

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

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30.

Figure 8-4. Fish sampling continuation form.

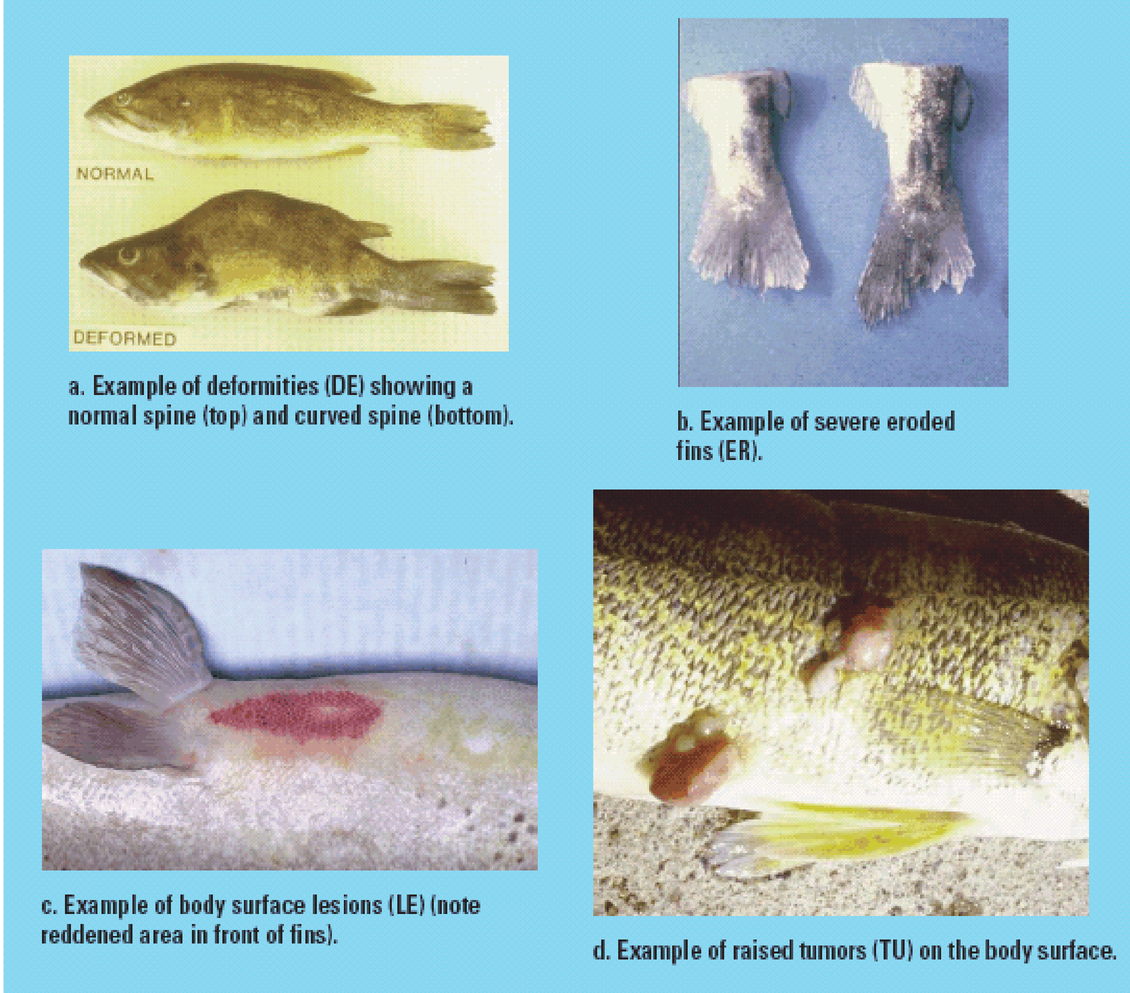


Figure 8-5. DELT anomalies (reprinted from Moulton et al. 2002).

Figure 8-6. Example of a voucher photo. The fish shown is in size class 4 (9-12 cm total length).

FISH VOUCHERS

75% Ethanol/10% Formalin

GRW04449-___ ___ ___

___/___/ 200__

Transect number 1 2

Site visit number 1 2

300177

____ of ____

FISH VOUCHERS

75% Ethanol/ 10% Formalin

GRW04449-___ ___ ___

___/___/ 200__

Transect number 1 2

Site visit number 1 2

Sample ID#___________

____ of ____

PHOTO FISH VOUCHER

GRW04449- ___ ___ ___

____/____/ 200__

Transect 1° 2°

____

Common name

FIN TISSUE DNA

GRW04449-___ ___ ___

___/___/ 200__

311255

____

Common name

FIN TISSUE DNA

GRW04449-___ ___ ___

___/___/ 200__

Sample ID______________

____

Common name

Figure 8-7. Fish voucher labels, photo voucher label, fish voucher tag, and fin tissue DNA sample label. The fish voucher label at upper left is placed on the jar holding the field composite of preserved specimens retained by the sampling crew for laboratory identification and individual species voucher samples. If multiple jars are needed, the continuation label at upper right is used. The fish voucher label is filled out and placed next to the fish specimen when it is photographed. The fish voucher tag label is placed in the jar of preserved specimens. The fin-tissue DNA label is placed on the coin envelope for larger fish (and photographed with the fish) and placed on the jar for smaller specimens. Use continuation label if multiple jars are needed for DNA sample. Not actual size.

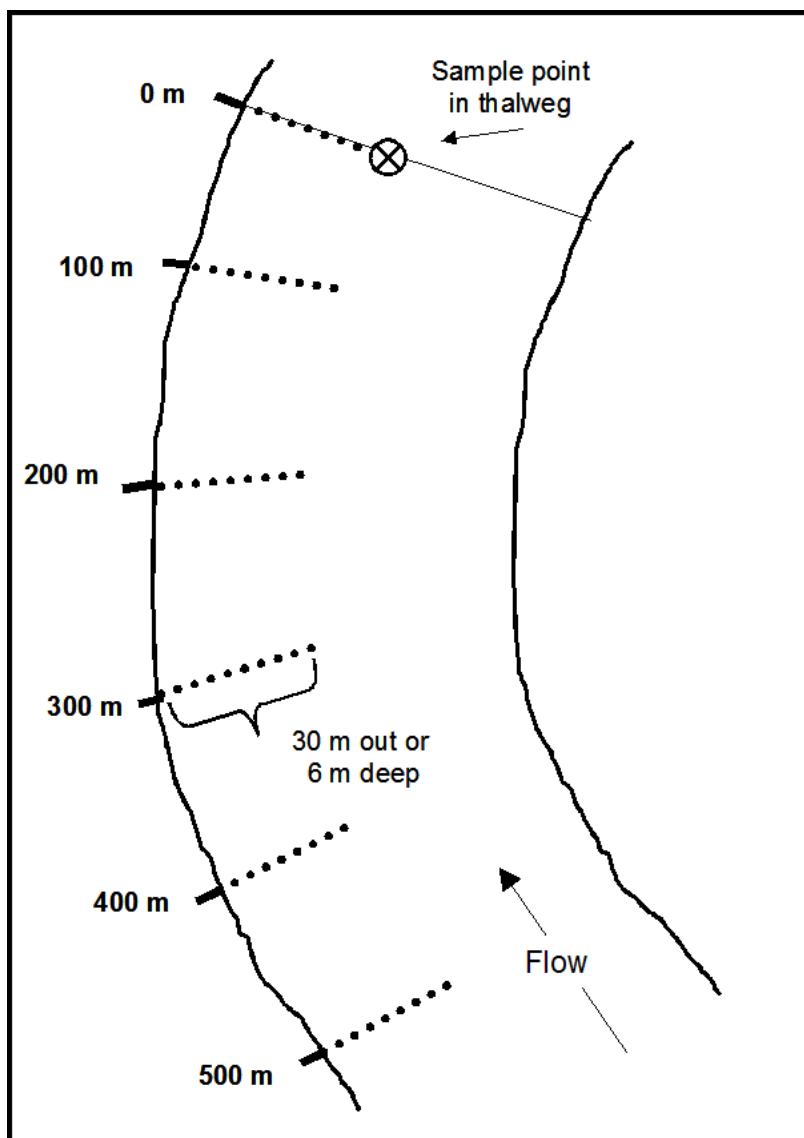
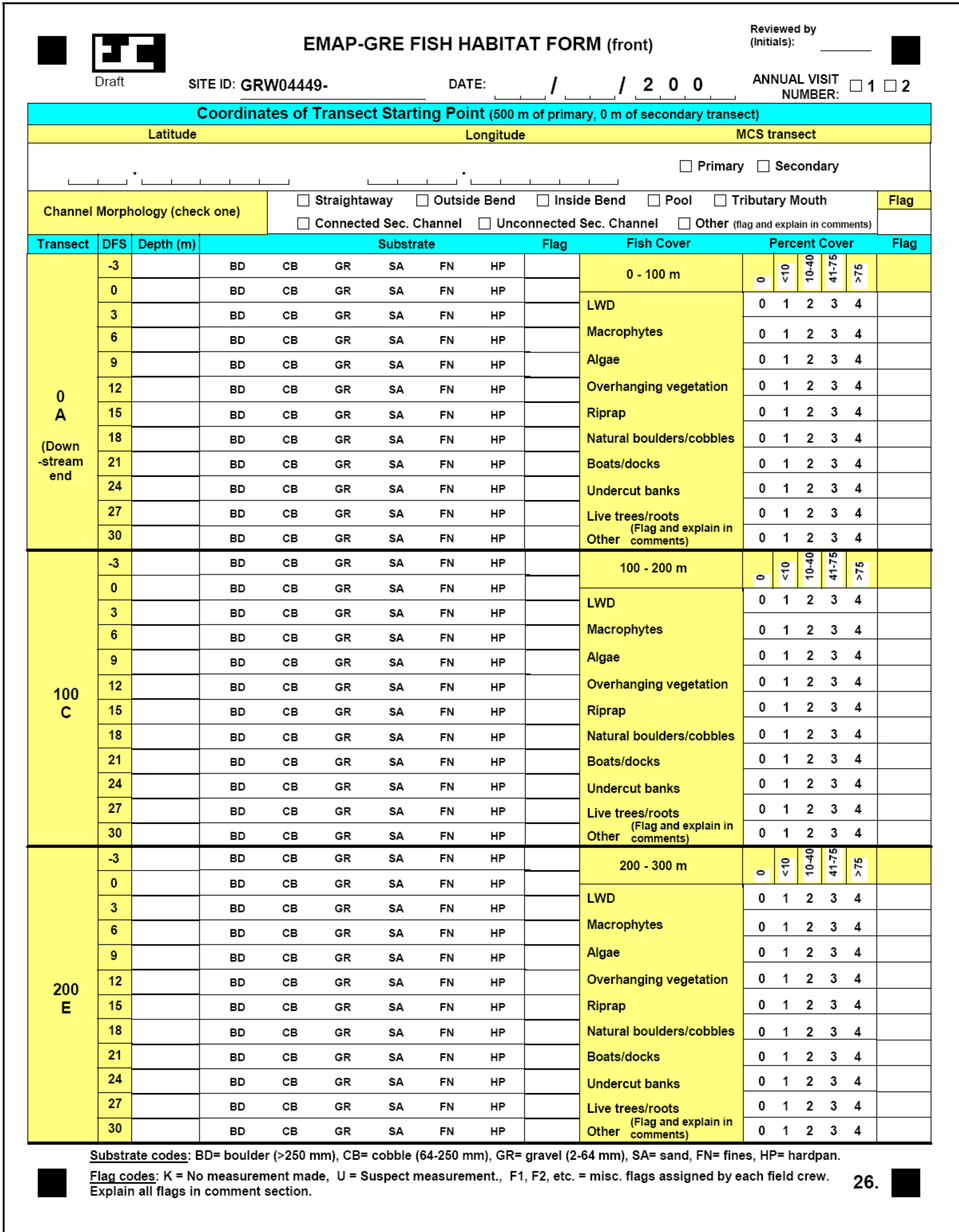


Figure 8-8. Layout of physical habitat measurement locations. At each transect into the channel, substrate is recorded every 3 m to a distance of 30 m or a depth of 6 m, whichever is closer to the bank. Depth is recorded out to a distance 30 m from shore (or wherever the weight on habitat rope is dropped) using the habitat pole or depth sounder. Each transect starts as close as the boat can get to the shoreline. Fish cover is quantified for the 100-m segments between substrate transects. Not to scale.



Draft

EMAP-GRE FISH HABITAT FORM (front)

Reviewed by (Initials): ______

SITE ID: GRW04449-______ DATE: ____ / ____ / 2 0 0 __ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2

Coordinates of Transect Starting Point (500 m of primary, 0 m of secondary transect)

Latitude	Longitude	MCS transect
___ . ______	___ . ______	☐ Primary ☐ Secondary

Channel Morphology (check one)	☐ Straightaway ☐ Outside Bend ☐ Inside Bend ☐ Pool ☐ Tributary Mouth ☐ Connected Sec. Channel ☐ Unconnected Sec. Channel ☐ Other (flag and explain in comments)	Flag

Transect	DFS	Depth (m)	Substrate	Flag	Fish Cover	Percent Cover					Flag
0 A (Down-stream end)	-3		BD CB GR SA FN HP		0 - 100 m	0	<10	10-40	41-75	>75	
	0		BD CB GR SA FN HP		LWD	0	1	2	3	4	
	3		BD CB GR SA FN HP		Macrophytes	0	1	2	3	4	
	6		BD CB GR SA FN HP		Algae	0	1	2	3	4	
	9		BD CB GR SA FN HP		Overhanging vegetation	0	1	2	3	4	
	12		BD CB GR SA FN HP		Riprap	0	1	2	3	4	
	15		BD CB GR SA FN HP		Natural boulders/cobbles	0	1	2	3	4	
	18		BD CB GR SA FN HP		Boats/docks	0	1	2	3	4	
	21		BD CB GR SA FN HP		Undercut banks	0	1	2	3	4	
	24		BD CB GR SA FN HP		Live trees/roots	0	1	2	3	4	
	27		BD CB GR SA FN HP		Other (Flag and explain in comments)	0	1	2	3	4	
	30		BD CB GR SA FN HP								
100 C	-3		BD CB GR SA FN HP		100 - 200 m	0	<10	10-40	41-75	>75	
	0		BD CB GR SA FN HP		LWD	0	1	2	3	4	
	3		BD CB GR SA FN HP		Macrophytes	0	1	2	3	4	
	6		BD CB GR SA FN HP		Algae	0	1	2	3	4	
	9		BD CB GR SA FN HP		Overhanging vegetation	0	1	2	3	4	
	12		BD CB GR SA FN HP		Riprap	0	1	2	3	4	
	15		BD CB GR SA FN HP		Natural boulders/cobbles	0	1	2	3	4	
	18		BD CB GR SA FN HP		Boats/docks	0	1	2	3	4	
	21		BD CB GR SA FN HP		Undercut banks	0	1	2	3	4	
	24		BD CB GR SA FN HP		Live trees/roots	0	1	2	3	4	
	27		BD CB GR SA FN HP		Other (Flag and explain in comments)	0	1	2	3	4	
	30		BD CB GR SA FN HP								
200 E	-3		BD CB GR SA FN HP		200 - 300 m	0	<10	10-40	41-75	>75	
	0		BD CB GR SA FN HP		LWD	0	1	2	3	4	
	3		BD CB GR SA FN HP		Macrophytes	0	1	2	3	4	
	6		BD CB GR SA FN HP		Algae	0	1	2	3	4	
	9		BD CB GR SA FN HP		Overhanging vegetation	0	1	2	3	4	
	12		BD CB GR SA FN HP		Riprap	0	1	2	3	4	
	15		BD CB GR SA FN HP		Natural boulders/cobbles	0	1	2	3	4	
	18		BD CB GR SA FN HP		Boats/docks	0	1	2	3	4	
	21		BD CB GR SA FN HP		Undercut banks	0	1	2	3	4	
	24		BD CB GR SA FN HP		Live trees/roots	0	1	2	3	4	
	27		BD CB GR SA FN HP		Other (Flag and explain in comments)	0	1	2	3	4	
	30		BD CB GR SA FN HP								

Substrate codes: BD= boulder (>250 mm), CB= cobble (64-250 mm), GR= gravel (2-64 mm), SA= sand, FN= fines, HP= hardpan.

Flag codes: K = No measurement made, U = Suspect measurement., F1, F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

26.

Figure 8-9. Fish habitat sampling form (front).

Draft

EMAP-GRE FISH HABITAT FORM (back)

Reviewed by (Initials): ________

SITE ID: GRW04449- DATE: ___ / ___ / 2 0 0

ANNUAL VISIT NUMBER: ☐ 1 ☐ 2

Substation	DFS	Depth (m)	Substrate						Flag
300 G	-3		BD	CB	GR	SA	FN	HP	
	0		BD	CB	GR	SA	FN	HP	
	3		BD	CB	GR	SA	FN	HP	
	6		BD	CB	GR	SA	FN	HP	
	9		BD	CB	GR	SA	FN	HP	
	12		BD	CB	GR	SA	FN	HP	
	15		BD	CB	GR	SA	FN	HP	
	18		BD	CB	GR	SA	FN	HP	
	21		BD	CB	GR	SA	FN	HP	
	24		BD	CB	GR	SA	FN	HP	
	27		BD	CB	GR	SA	FN	HP	
	30		BD	CB	GR	SA	FN	HP	
400 I	-3		BD	CB	GR	SA	FN	HP	
	0		BD	CB	GR	SA	FN	HP	
	3		BD	CB	GR	SA	FN	HP	
	6		BD	CB	GR	SA	FN	HP	
	9		BD	CB	GR	SA	FN	HP	
	12		BD	CB	GR	SA	FN	HP	
	15		BD	CB	GR	SA	FN	HP	
	18		BD	CB	GR	SA	FN	HP	
	21		BD	CB	GR	SA	FN	HP	
	24		BD	CB	GR	SA	FN	HP	
	27		BD	CB	GR	SA	FN	HP	
	30		BD	CB	GR	SA	FN	HP	
500 K Upstream end	-3		BD	CB	GR	SA	FN	HP	
	0		BD	CB	GR	SA	FN	HP	
	3		BD	CB	GR	SA	FN	HP	
	6		BD	CB	GR	SA	FN	HP	
	9		BD	CB	GR	SA	FN	HP	
	12		BD	CB	GR	SA	FN	HP	
	15		BD	CB	GR	SA	FN	HP	
	18		BD	CB	GR	SA	FN	HP	
	21		BD	CB	GR	SA	FN	HP	
	24		BD	CB	GR	SA	FN	HP	
	27		BD	CB	GR	SA	FN	HP	
	30		BD	CB	GR	SA	FN	HP	

Fish Cover	Percent Cover					Flag
300 - 400 m	0	<10	10-40	41-75	>75	
LWD	0	1	2	3	4	
Macrophytes	0	1	2	3	4	
Algae	0	1	2	3	4	
Overhanging vegetation	0	1	2	3	4	
Riprap	0	1	2	3	4	
Natural boulders/cobbles	0	1	2	3	4	
Boats/docks	0	1	2	3	4	
Undercut banks	0	1	2	3	4	
Live trees/roots	0	1	2	3	4	
Other (Flag and explain in comments)	0	1	2	3	4	
400 - 500 m	0	<10	10-40	41-75	>75	
LWD	0	1	2	3	4	
Macrophytes	0	1	2	3	4	
Algae	0	1	2	3	4	
Overhanging vegetation	0	1	2	3	4	
Riprap	0	1	2	3	4	
Natural boulders/cobbles	0	1	2	3	4	
Boats/docks	0	1	2	3	4	
Undercut banks	0	1	2	3	4	
Live trees/roots	0	1	2	3	4	
Other (Flag and explain in comments)	0	1	2	3	4	

COMMENTS	

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

27.

Figure 8-10. Fish habitat sampling form (back).

Section 9
Fish Tissue Contaminants

James M. Lazorchak[1], Erich B. Emery[2], David M. Walters[1], and Spence A. Peterson[3]

Fish tissue contaminants are an indicator of bioaccumulation of persistent toxic substances in the environment (Table 9-1), and can be used to estimate exposure to contaminants associated with fish consumption for higher trophic levels, including humans. Various studies have been done on fish tissue contaminants that have focused on different parts of the fish (whole fish, fillets, livers). EMAP-GRE will focus on whole fish because of its emphasis on the health of the ecosystem. Although whole-fish contamination is primarily an indicator of risk to piscivorous wildlife, whole-fish data are still relevant for estimating human exposure to contaminants through fish consumption. Use of whole fish reduces sample processing effort in the field because no gutting, skinning, or filleting of fish are necessary.

At every EMAP-GRE site, two composite fish samples are collected: a small-fish sample and a large-fish sample. The small-fish sample includes individuals of one species (if possible) whose adults are small. The large-fish sample includes individuals of one species (if possible) whose adults are larger. Both sizes of fish have advantages. Small fish are more ubiquitous than the larger fish, and therefore are more likely to be present in sufficient numbers at more sites. With small species, it may be possible to get a more representative sample of the contaminant load at the site by combining 20 - 200 individual small fish in a composite sample than by combining only a few larger fish. Small fish may be a more appropriate indicator for assessing ecological risk to wildlife because they are more likely to be prey for piscivores than are larger fish. Large fish are more mobile than small fish, and are more likely to partly reflect contaminant exposure away from the site. Larger, longer-lived fish may exhibit greater bioaccumulation and may be more sensitive indicators of contaminants in the environment.

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1 U.S. Environmental Protection Agency, Office of Research and Development, National Exposure Research Laboratory, Ecological Research Division, 26 W. Martin Luther King Dr., Cincinnati, OH 45268

2 Ohio River Valley Sanitation Commission, 5735 Kellogg Avenue, Cincinnati, OH 45228

3 U.S. Environmental Protection Agency, Office of Research and Development, National Health and Environmental Effects Laboratory, Western Ecology Division, 200 SW 35th St., Corvallis, OR 97333

9.1 Integration with fish sampling

Fish tissue samples are collected during electrofishing (Section 8). At each site, two separate 500-m reaches are electrofished. Composite fish samples for tissue sample analysis may be retained from either or both of the 500-m electrofishing reaches at the discretion of the crew leader. The origin of the fish from within the site is recorded on the fish tissue form (Figure 9-1).

9.2 Selecting fish tissue specimens

The fish-sampling crew should attempt to collect a sample of small fish, and, if possible, a sample of larger fish. Procedures for selecting fish for tissue specimens are described in Table 9-2.

9.3 Preparing composite samples

After the fish comprising the small-fish and large-fish composite have been selected (Table 9-3), they are recorded on the fish tissue form (Figure 9-1) and packaged for shipment to the laboratory. Procedures for preparing composite samples are described in Table 9-4.

Table 9-1. Target analytes for composite fish tissue samples. Detection limit for mercury is 0.01 ppm. Detection limit for all other analytes is 0.001 ppm. Number in parentheses is the CAS number. Number followed by a # is the Ballschmitter-Zell number.

Mercury (7439-97-6)
Aldrin (309-00-2)
Chlordane-cis (5103-71-9)
Chlordane-trans (5103-74-2)
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)
Hexachlorocyclohaxane [Gamma-BHC/Lindane] (58-89-9)
Mirex (2385-85-5)
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)
3,3',4,4' Tetrachlorobiphenyl, #77 (32598-13-3) (coplaner)
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)
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',5 Pentachlorobiphenyl, #126 (coplaner)
Polybrominated Diphenyl Ethers (PBDE) congeners 47, 99, 100, 153 and 154

Percent Moisture and Lipid content

Table 9-2. Procedure for selecting fish for tissue analysis.

1. **Small-fish-species composite**. Retain similar-sized specimens (smallest individual $\geq$ 75% of the total length of the largest individual) of a single species from the list in Table 9-3 in priority order. Select the first species on the list for which a sample of at least 50 g can be collected. Retain a total sample of the selected species of up to 400 g. The "similar-size" rule can be violated if necessary to obtain $\geq$ 50 g.

2. **Large-fish-species composite**. Retain at least three and preferably five similar-sized specimens (smallest individual $\geq$ 75% of the total length of the largest individual) **of a single species** from the list in Table 9-3 in priority order. Try not to retain fish larger than 2 kg. Large specimens present problems for storage and lab processing.

3. If at least three similar-sized or same-size-class specimens are not available, move down the list (Table 9-3) to the next large species. Collecting three individuals of the same species is preferable to collecting a mixture of species, even in they are higher priority species.

4. If at least three similar-sized or same-size-class specimens are not available for any large species, return to the top of the list (Table 9-3) and composite individuals across size classes to obtain at least three specimens of a single species.

5. If these criteria still cannot be met, use best judgement to collect small- and large-fish species composite samples comprised of as many specimens as possible. For small fish, at least 8 g of fish are needed for analysis. Collect a multispecies composite sample as a last resort.

Table 9-3. Fish target species list for tissue analysis. 3-cm size classes refer to demarcations on the measuring board. The same information appears on the back of the fish tissue form (Figure 9-1).

Priority	Common name	Size range (mm)	3-cm size classes
SMALL target species			
1	emerald shiner	< 120	1 - 4
2	river shiner	< 120	1 - 4
3	spotfin shiner	< 120	1 - 4
4	bullhead minnow	< 120	1 - 4
5	silver chub	< 120	1 - 4
6	another minnow species	< 120	1 - 4
7	gizzard shad	< 150	1 - 5
LARGE target species			
1	sauger	120 - 180	5 - 6
2	sauger	180 - 240	7 - 8
3	sauger	> 240	≥ 9
4	largemouth bass	180 - 240	7 - 8
5	largemouth bass	240 - 300	8 - 10
6	largemouth bass	> 300	≥ 11
7	other black bass	> 180	≥ 7
8	brown trout	> 120	≥ 5
9	rainbow trout	> 120	≥ 5
10	channel catfish	120 - 180	5 - 6
11	channel catfish	450 - 510	16 - 17
12	channel catfish	180 - 450	7 - 15
13	freshwater drum	>120	≥ 5
14	shorthead redhorse	>120	≥ 5
15	other redhorse species	>120	≥ 5
16	bluegill	>120	≥ 5
17	longear sunfish	>120	≥ 5
18	other sunfish species	>120	≥ 5
19	common carp	>180	≥ 7
20	smallmouth buffalo	>120	≥ 5
21	river carpsucker	>120	≥ 5
22	flathead catfish	>120	≥ 5
23	white bass/wiper	>120	≥ 5
24	quillback	>120	≥ 5

Table 9-4. Procedures for preparing composite fish tissue samples.

1.	Fill in site ID and date on the fish tissue form (Figure 9-1).
2.	**Small-fish-species composite**. Record the AFS common name (from Nelson et al. 2004) and count of individuals in the sample. From the composite collection location choices on the form, indicate where the fish were collected at the site. Fish may be collected from beyond the end of the transect if needed (mark “other” for collection location).
3.	Euthanize fish with a cervical/cranial blow or other humane method. Use clean hands to transfer specimens to aluminum foil. Keep hands, work surfaces, and foil clean and free of potential contaminants (mud, fuel, slime, formalin, sun screen, insect repellant, etc.)
4.	On a clean work surface, wrap all the fish in a single piece of aluminum foil, making sure that the dull side of the foil is in contact with the fish. Place the wrapped sample in a 1 gallon self-sealing plastic bag. Expel excess air from each bag and wrap each bag with clear tape to seal the sample. Go to step 8.
5.	**Large-fish-species composite**. Record the AFS common name (from Nelson et al. 2004) and size class of each individual in the sample. From composite collection location choices on the form, indicate where the fish were collected at the site. Fish may be collected from beyond the end of the transect if needed (mark “other” for collection location).
6.	Euthanize fish with a cranial blow or other humane method. Use clean hands to transfer specimens to aluminum foil. Keep hands, work surfaces, and foil clean and free of potential contaminants (mud, fuel, slime, formalin, sun screen, insect repellant, etc.).
7.	On a clean work surface, wrap the fish in a single piece of aluminum foil, if possible, making sure that the dull side of the foil is in contact with the fish. Fish may be wrapped individually if necessary. Place the wrapped sample in a 2 gallon self-sealing plastic bag (use a separate bag from the small fish sample). Expel excess air from each bag and wrap each bag with clear tape to seal the sample.
8.	Prepare a fish tissue label for each sample (Figure 9-2). Fill in the site ID and date and circle the sample type (small or large fish). Record the sample ID (from the labels) on the form.
9.	Affix the appropriate label on each bag and cover with clear tape. Place each sample in a second self-sealing plastic bag.
10.	Prepare a continuation label (Figure 9-2) for the outside of each bag. Transfer the sample ID from the inner label to the outer label. Affix the label to the outside of the outer bag and cover with clear tape.
11.	Place the double-bagged samples in a cooler with bagged ice. If possible, freeze the samples at the base location prior to shipment.

9.4 Equipment and supplies

Table 9-5 is a checklist of equipment and supplies required for collecting fish tissue samples. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

Table 9-5. Equipment and supplies for collecting fish tissue samples. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

Qty	Item	
1	Measuring board with 3 cm size classes (see Figure 8-6)	
1 roll	Heavy duty aluminum foil (18" size)	
2	1-gallon self-sealing plastic bags	
2	2-gallon self-sealing plastic bags	
1	Cooler with ice.	
1	Fish tissue form	
1 sets	Fish tissue sample labels	

9.5 Literature cited

Nelson, J.S, E.J. Crossman, H. Espinosa-Perez. L.T. Findlay, C.R. Gilbert, R.N. Lea., and J.D. Williams. 2004. Common and scientific names of fishes from the United States, Canada, and Mexico, Sixth edition. The American Fisheries Society. Bethesda, MD.

Draft

EMAP-GRE FISH TISSUE FORM

Reviewed by (Initials): ________

SITE ID: GRW04449-______ DATE: ___/___/ 2 0 __ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2

Small Fish Species Sample				
Sample ID		Composite Collection Location (Circle One)	DP DS DB OT	
Common Name		Size Class	Count	Flag

Large Fish Species Sample				
Sample ID		Composite Collection Location (Circle One)	DP DS DB OT	
Common Name		Size Class	Count	Flag

Composite Collection Location Codes

DP Distributed through the primary 500m transect
DS Distributed through the secondary 500m transect
DB Distributed through both transects
OT Other (describe in comments)

Flag	COMMENTS

Flag codes: K = No measurement made, U = Suspect measurement., F1,F2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

31.

Figure 9-1. Fish tissue form.

FISH TISSUE

Small fish Large fish

GRW04449-___ ___ ___

___/___/ 200__

Site visit number 1 2

300211

FISH TISSUE

Small fish Large fish

GRW04449___ ___ ___

___/___/ 200__

Site visit number 1 2

Sample ID__________

Figure 9-2. Sample labels for fish tissue contaminants. The number at the bottom of the label on the left is the unique sample ID. The label on the left is affixed to the inner bag holding the sample. The continuation label on the right is affixed to the outer bag or can be used for additional bags if needed. Not actual size.

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Section 10

Benthic Macroinvertebrates

Ted R. Angradi[1], Donald J. Klemm[2], Jim M. Lazorchak[2], and Brent R. Johnson[2]

Benthic macroinvertebrates inhabit river bed sediments and adhere to hard substrates in the water column. Macroinvertebrates have several advantages as indicators of ecological condition (Barbour et al. 1999, Klemm et al. 1990). They are ubiquitous in all GRE aquatic habitats and are relatively easy to collect in large numbers in most habitats. Macroinvertebrate assemblages are typically very diverse and sensitive to a variety of stressors. In some cases, macroinvertebrate assemblage composition can reveal the nature of the anthropogenic stress to which the assemblage has been exposed (Barbour et al. 1999).

In EMAP-GRE, benthic macroinvertebrates are collected by the river-sampling crew in two habitats: shallow (<1 m), near-shore littoral areas, and the surface of large woody debris (LWD) or “snags” in the main channel. In near-shore littoral areas, benthos samples are collected by kick sampling. Snags are sampled by boat using a modified kick net. The kick sampling and sample-processing procedures described herein are adapted, with significant modification, from Peck et al. (Unpublished drafts).

10.1 Near-shore kick sampling

At each site, two 500-m main channel shoreline (MCS) transects, starting at the intersection of the cross-channel transect and the MCS (Figure 4-1), are located and flagged by either the fish- or river-sampling crew, depending on which crew arrives at the site first. The primary transect is initially flagged at 100-m intervals; intermediate littoral stations at 50-m intervals are located and flagged using a handheld GPS or by visual estimation during littoral sampling (a different flag color than at the 100-m stations may be used). At each of the resulting 11 evenly-spaced points along the 500-m MCS transect (stations A-K; Figure 4-2), two

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1 U.S. Environmental Protection Agency, Office of Research and Development, National Health and Environmental Effects Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804

2 U.S. Environmental Protection Agency, Office of Research and Development, National Exposure Research Laboratory, Ecological Research Division, 26 W. Martin Luther King Dr., Cincinnati, OH 45268

30-second, 0.26-m^2 (2.8 ft^2) kick samples are collected in the near-shore littoral zone using a standard rectangular-frame kick net (335 x 508-mm frame; 500-µm mesh). The 22 kick samples at each site are combined into a single composite sample representing a total bottom area of 5.7 m^2. Sampling is restricted to the littoral habitat because deeper benthic habitats of the channel are much more difficult to sample, and benthic organisms are often present in very low abundance in non-littoral channel areas – especially in large, sand-bed rivers. Table 10-1 describes the kick sampling procedures in detail.

In some reaches that are extensively modified with wing-dams, spur-dikes, or other channel-training structures, vertical shorelines may prohibit safe kick sampling at some littoral stations. When this situation is encountered, search 5 m up- and down-river from the station for safe kick sample locations. If no safe location is available, do not collect kick samples at that station and note the missing samples with a flag on the form. Littoral sample locations in dike fields are located along the wetted edge of the natural shoreline contour unless the littoral station occurs opposite of the base of a dike (see Figure 10-1).

10.2 Snag sampling

In alluvial floodplain rivers, snags are massive pieces of large woody debris (LWD) that are imbedded in or resting on the river bottom (Angradi et al., 2004). The snag sample (Figure 10-2) is collected from the snag that is nearest to the intersection of the cross-channel and the MCS and which meets the suitability criteria. A specialized "snag net" is used to collect a 1-m-long sample from the up-current side of the snag. The snag net resembles a standard rectangular-frame kick net, but with the frame constructed so that the net fits over half the circumference of a snag (Figure 10-3). Two sizes of snag nets with mouth widths ("diameters") of 0.2 m (8 inches) and 0.33 m (13 inches) will be used. For larger snags, a standard rectangular-frame kick net (33 x 51-cm frame; 500-µm mesh) can be used to sample the surface of the snag. Snag sampling methods are described in Table 10-2.

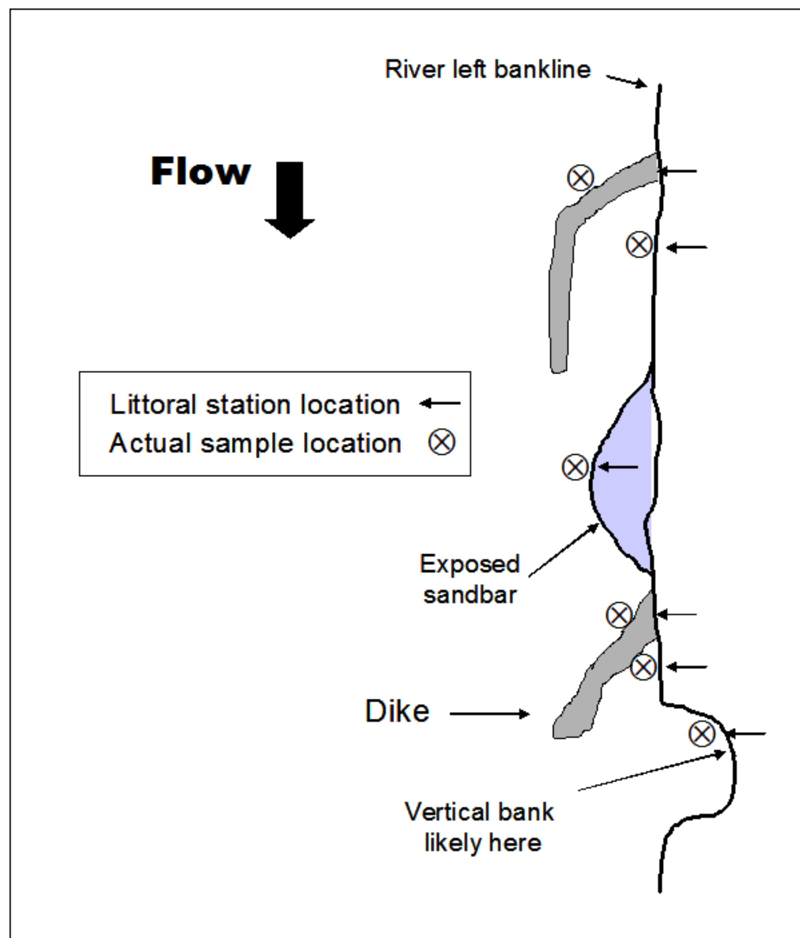

Figure 10-1. Guidance on sample location for littoral stations in dike fields. Littoral stations should follow the wetted edge of the natural contour of the shoreline unless the station occurs at the base of a dike. In this case, the sample is collected on the face of the dike. Scalloped shorelines behind some dikes may have unsampleable vertical banks.

Suitable snags for sampling are in water at least 0.6 m deep, are exposed to some current, and are at least 5 m long, the minimum length for a piece of LWD established for EMAP (Peck et al., unpublished drafts). Suitable snags are at least 15 cm (6 inches) in diameter where the snag breaks the surface or comes within 30 cm (12 inches) of the water surface. This definition of a suitable snag is restrictive to minimize among-sample variability and maximize the efficiency of the snag nets. Finding the nearest suitable snag may require some searching up- and down-river from the X-site. **Surrogate snag samples may be collected**

from man-made structures (pilings, navigation markers, etc.) if no natural snags are present. Ancillary data collected for each snag include depth, snag diameter at the water surface, snag surface characteristics, water velocity, and the distance of the snag from the shore. Take a picture of the sampled snag if possible.

Figure 10-2. Snag sampling on the Missouri River. The captain is holding the boat in position against the snag using the motors. The biologist at left is brushing the surface of the snag in front of the snag net.

10.3 Large woody debris abundance

At each site, large woody debris (LWD) meeting the minimum size criteria is tallied for a 500 m x 10 m near-shore quadrat adjacent to the MCS transect. This procedure is included in this section of the manual because the LWD is most efficiently tallied while searching for a suitable snag sampling location. Nearshore LWD is categorized by type, after Angradi et al. (2004), as below the wetted perimeter ("wet"), between the wetted perimeter and the bankfull stage but not having originated a its present location ("beached"), and partly below the wetted perimeter (providing aquatic habitat) but originating at the present location ("anchored"). A piece of LWD that is anchored may also be wet because it provides aquatic habitat, but it should be counted as "anchored." Anchored LWD that is not providing aquatic habitat is not counted. Density of LWD (number per hectare of channel) in the main channel including the 10-m-wide near-shore quadrat is visually estimated and recorded by density category. EMAP-GRE includes fewer LWD size categories than previous EMAP methods (Kaufmann 2003). Our experience on the Upper Missouri River in 2000-2003 showed that most LWD was ≤ 1 m diameter at the large end and ≤ 20 m in length. Furthermore, accurate estimation of piece length is difficult for LWD in the channel because usually only a portion of the LWD is visible. Procedures for quantifying LWD are described in Table 10-3.

10.4 Sample processing

After the composite kick or snag samples have each been distributed into one or more jars (with no jar filled more than 1/3 full with sample), the jars should be filled almost completely with 10% carbonate-buffered formalin. Each jar is then topped of with a concentrated rose bengal solution (Table 3-1). A label is filled out (Figure 10-6) and placed inside each jar and on the outside of each jar. If extra jars are needed, transfer the sample ID to a continuation label and place it on the outside of each jar. Each sample composite (not each jar) gets a unique sample ID. Details of the sample processing procedure are provided in Table 10-4.

Table 10-1. Procedures for collecting near-shore littoral kick samples (adapted, in part, from Klemm et al. 2003).

1.	Fill in the site ID and date on the littoral and snag sampling form (Figure 10-4). Go to MCS station A at the intersection of the cross-channel transect and the MCS (Figure 4-1 and 4-2) and record the latitude and longitude at the intersection using a hand-held GPS unit.
2.	At each of 11 evenly-spaced stations on the MCS transect (i.e., station A at 0 m, station B at 50 m, etc.; Figure 4-2) collect two kick samples. Locate stations that have not been pre-flagged (50 m, 150 m, 250 m, etc.) using a hand-held GPS or by estimating the halfway point between flagged stations.
3.	Locate kick samples in a zone bounded on the shore side by the apparent low-water mark from daily flow fluctuations (most relevant on the regulated Upper Missouri River) and bounded on the river side by the 0.6-m depth contour (recommended maximum sample depth; deeper sampling may be possible). The low-water mark at a site can often be detected by the presence of periphyton or attached filamentous algae just below the low water mark. If samples cannot be safely collected at a station due to vertical banks or other reason, search 5 m up- and down-river for a safe location. If a safe location is still not available, do not collect kick samples at that station and note the missing sample for the station with a flag on the form.
4.	If there is sufficient current to extend the net, go to step 5; if not go to step 16.
5.	**Method for kick sampling in current.** At each sample location, hold the net opening facing up-current, and position the net securely on the stream bottom. Avoid large rocks or debris that prevent the net from seating properly on the bottom.
6.	Visually define a square quadrat on the bottom just up-current from the net that is one net-width on every side (0.26 m^2). Check the quadrat for heavy organisms such as mussels and snails (if the bottom is visible). Place these organisms in the net by hand.
7.	Holding the net in place with your knees, pick up any loose cobbles or large pieces of gravel or debris from the quadrat and rub them with your hands or a small brush so that organisms wash into the net. Discard the rocks outside the quadrat. In water too deep to brush rocks in front of the net, place the rocks in the sample composite bucket.
8.	After scrubbing and removing the larger substrate particles, hold the net securely in position while stirring the substrate remaining within the quadrat to a depth of 10 cm for 30 seconds using either a gloved hand, or by kicking the substrate vigorously for 30 seconds.
9.	Remove the net from the water with a quick upstream motion to wash the organisms to the back of the net.

Continued

Table 10-1. Procedures for collecting near-shore littoral kick samples, continued.

10.	Kicking in a sand bottom can result in a very full net. Most of the sand will pass through the mesh if the net is gently agitated while the net mouth is held out of the water with one hand and the collecting bucket supported with the other hand.
11.	Sweep the net through clean water several times to consolidate net contents in the screened bucket at the cod end. Inspect the net for clinging organisms; using forceps, place any organisms found into the Dolphin bucket. Dump the contents of the Dolphin bucket into a sample composite bucket. In some cases it may be faster and easier to hold the first sample from a station in the net while the second kick sample is being taken.
12.	On the form, circle the appropriate dominant substrate size/type at the sample location.
13.	Repeat steps 5 -11 for a second kick sample location at the station. Be sure to move upstream at least 1 m before collecting the second kick sample.
14.	Repeat steps 5 -13 for the remaining littoral sample stations.
15.	Go to step 23.
16.	**Method for sweep sampling in slack water or pools**. At each sample location, hold the net opening facing upstream, position the net securely on the stream bottom. Avoid rocks or debris that prevent the net from seating properly on the bottom.
17.	Visually define a square quadrat on the bottom just up-current from the net that is one net width on every side (0.26 m^2). Check the quadrat for heavy organisms such as mussels and snails (if the bottom is visible). Place these organisms in the net by hand.
18.	Holding the net in place with your knees, pick up any loose cobbles or large pieces of gravel and debris and place them in the sample composite bucket.
19.	Vigorously kick the substrate remaining within the quadrat and then drag net through the disturbed area just above the bottom. Continue this for 30 seconds (counting “one Missouri, two Missouri, etc.” is sufficiently accurate). Keep the net moving so captured organisms cannot escape.
20.	Remove the net from the water with a quick upstream motion to wash the organisms to the back of the net.
21.	Sweep the net through clean water several times to consolidate net contents in the Dolphin bucket at the cod end. Inspect the net for clinging organisms; using forceps, place any organisms found into the Dolphin bucket. Dump the contents of the Dolphin bucket into the composite sample bucket.
22.	Repeat steps 16 - 21 for a second kick sample location at the station. Be sure to move upstream at least 1 m before collecting the second kick sample.

Continued

Table 10-1. Procedures for collecting near-shore littoral kick samples, continued.

23.	On the form, circle the appropriate dominant substrate size/type at the sample location.
24.	Repeat steps 15 - 23 for the remaining sweep sample replicates in slack water areas.
25.	Dump the contents of the composite bucket, including both “current” and “slack water” samples, into a 0.3-m diameter 500-µm mesh sieve and wash the sample gently (no nozzle) using the onboard washdown hose (the 0.3-m diameter sieve will fit over a 20-L bucket to catch wash water). Gravel, large organic particles, and macrophytes should be thoroughly washed, inspected for clinging organisms and discarded. If there is a large amount of coarse sand or small gravel in the sample, use a second 20-L bucket to elutriate the sample before sieving.
26.	Transfer the washed composite sample into a 500-mL jar using the wide-bore funnel and the wash bottle with minimal water. Use two or more jars if necessary. **Do not fill any jar more than 1/3 full with sample.**
27.	For sample jars that are not pre-labeled, the site number and sample type should be written directly on the jar(s) with a Sharpie (e.g., “kick 045") to avoid mixing up jars. This notation should be covered later with the sample label. Place the samples in a cooler.
28.	Go to Table 10-4 for procedures for labeling and preserving samples.

10.5 QA considerations for macroinvertebrate sampling

Standardization of effort and attention to detail are important for maintaining a high QA standard for field sampling. Several QA considerations for macroinvertebrate sampling are presented in Table 10-5.

10.6 Safety considerations for macroinvertebrate sampling

Safety is paramount. General and boat-related safety guidance is presented in Section 2. Safety considerations relevant to macroinvertebrate sampling are presented in Table 10-6.

10.7 Equipment and supplies

Table 10-7 is a checklist of equipment and supplies required for collecting macroinvertebrate samples. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

Table 10-2. Procedure for collecting snag macroinvertebrate samples.

1. Find the natural snag nearest to the intersection of the cross-channel transect and the MCS (littoral sample station A, Figure 4-2) which meets the snag suitability criteria. Search both shorelines 1 km up-river and down-river from the transect and on the way back to the ramp. If no suitable natural snag is found, a man-made snag substitute should be sampled (e.g., piling). If a snag is sampled that does not meet all the natural snag criteria in step 2, note this in the comments on the littoral and snag sampling form (Figure 10-4).

2. The snag must be in flowing waters at least 0.6 m (2 ft) deep, must be ≥5 m (16.5 ft) long, and with a diameter of ≥ 0.15 m (6 inches) where the snag breaks the water surface or comes within 0.3 m (1 ft) of the water surface. Select the proper snag net for the snag (small [0.20 m "diameter"] or large [0.35 m "diameter"]). For snags too large for the large snag net, use the rectangular kick net (Figure 10-3).

3. Navigate up to the snag. Approach the snag slowly. The boat driver should be able hold the boat in position using the motor(s) (Figure 10-2).

4. Place the net against the snag facing up-current just below where the snag breaks the surface or where the snag comes closest to the water surface. If debris is wrapped around the snag at the water surface, sample further down (up current) on the snag.

5. A second crew member should use a long-handled brush to scrub the snag to wash organisms into the net in a traveling sample down the snag (Figure 10-2). Attempt to sample ≥1 m of snag. Be sure to scrub the sides of the snag. This is a qualitative method; not all the organisms on the snag will be captured in the net.

6. Sweep the net through clean water to consolidate net contents in the screened bucket at the cod end. Inspect the net for clinging organisms; place any found into the screened bucket using a forceps.

7. Transfer the contents of the Dolphin bucket directly into a 250-mL jar and rinse organisms from the Dolphin bucket into the jar using a wash bottle and a minimum of water. Sieving the sample will probably be unnecessary. If sieving is necessary to reduce sample volume, use the procedures in Table 10-1, step 25. Use two jars if the sample fills the jar more than 1/3 full.

8. Record the depth under the sampled part of the snag using the boat's sonar (Figure 10-5). Estimate the snag diameter size class in cm at the water surface (or where the snag comes closest to the surface). Record the snag surface characteristics (e.g., smooth, rough, algae present). Determine the distance from the snag to the nearest shoreline using a laser rangefinder or other method (visual estimation is acceptable). Record the net used and the approximate length of the snag sample (1 m is the goal). Record the coordinates of the snag.

Continued

Table 10-2. Procedure for collecting snag macroinvertebrate samples, continued.

9.	Use a velocity meter to measure surface water-velocity (m/s) above the sampled area.
10.	Alternatively, navigate several boat lengths up-river of the snag, shift to neutral and allow the boat to drift past the snag as closely as possible and record the speed-over-ground from the boat-mounted GPS unit as the boat passes the snag. A speed-over-ground of 1 km/h ≈ 0.28 m/s (1 mile/h = 0.45 m/s). This method does not work if it is windy.
11.	For sample jars that are not pre-labeled, the site number and sample type should be written directly on the jar(s) with a Sharpie (e.g., “snag 045") to avoid confusion later. This notation should be covered later with the sample label. Place the samples in a cooler.
12.	Go to Table 10-4 for procedures for labeling and preserving samples.

Table 10-3. Procedures for quantifying large woody debris (LWD).

1.	LWD is defined as pieces ≥ 5 m long and ≥ 0.3 m in diameter at the large end. Pieces may or may not break the water surface. For LWD in deep water (also called snags), the diameter of the large end of the piece will have to be estimated. In most cases snags in the main channel that are exposed to current are necessarily massive (or they would not be there) and will have a large-end diameter > 0.3 m. For pieces that are not cylindrical at the large end, visually estimate what the diameter would be for a cylindrical piece of the same volume.
2.	**Near shore LWD tally**. While cruising along the target shoreline in the boat (or while on foot if there is a lot of LWD), tally each piece of LWD by type: "wet," "beached," or "anchored" in the 10 x 500 m littoral quadrat on the littoral and snag sampling form. Wet LWD is in the channel below the wetted perimeter. Beached LWD is between the wetted perimeter and the visually-estimated bankfull level. Anchored LWD is partly below the wetted perimeter, but is anchored in the bank where it originated. Anchored LWD is typically a tree that has been undermined by bank erosion and has toppled over into the river but which has not been washed away. Beaver-felled trees often become anchored LWD. **LWD that is anchored but does not provide aquatic habitat is not counted**. For log jams in which some pieces may not be visible, attempt to estimate the number of pieces. Sum the tally and record the total for each type on the form (Figure 10-5).
3.	**Channel LWD**. Note the presence of LWD in the channel outside the littoral quadrat. Estimate the density of LWD as number/hectare in the channel between the main channel banks for the 500-m MCS transect (including the wet LWD from the littoral transect). A hectare is equal to an area of 100 x 100 m. Mark the appropriate category on the form.

Table 10-4. Procedures for labeling and preserving macroinvertebrate samples.

1.	To avoid clutter in the boat, benthos samples can be transported to the ramp or base location (if it is close to the ramp) in a cooler to be preserved.
2.	Fill each jar almost to the top with 10% carbonate-buffered formalin. Top off each jar with concentrated rose bengal solution (Table 3-1). Prepare a label(s) (Figure 10-6) for inside each jar(s). Circle the sample type (kick or snag); fill in the site number; enter the sample date, and print the collectors name. Place the label(s) inside the jar(s).
3.	Cap the jar and gently invert and rotate the jar to distribute the preservative. The preservative in the sample jar should be pink.
4.	Prepare a label (Figure 10-6) for the outside of the jar. Circle the sample type (kick or snag); fill in the site number from the design file; enter the sample date, visit number, jar number and total number of jars. Place the label on the jar and cover it with clear tape. Record the total number of jars and the sample ID from the label on the littoral and snag sampling form.
5.	If the sample requires more than one jar use a continuation label (Figure 10-6). Use the sample ID number from step 4.
6.	Seal each jar with plastic electrician's tape by wrapping with the threads (clockwise). Store the preserved sample upright in a secondary container until transport or shipment to the laboratory.

Table 10-5. QA considerations for macroinvertebrate sampling.

- Attempt to expend equal effort for each kick sample replicate despite variation in current and substrate.
- Strive to avoid any bias when locating kick samples.
- A few drops of non-permanent “thread-lock” on the kick-net ferule threads will prevent the handle from twisting during sampling.
- Use a gentle wash (no nozzle) when sieving the sample to avoid damaging fragile organisms (e.g., worms).
- Avoid sampling when a barge is approaching.
- Always inspect the kick or snag net for invertebrates clinging to the mesh.
- Try to get a natural snag or snag substitute (e.g., piling, channel marker, floating dock) sample at every site.
- Remove large organic debris from samples and drain excessive water from jars before adding preservative to insure that final preservative strength is sufficient to preserve organisms.

Table 10-6. Safety considerations for macroinvertebrate sampling.

- Use extreme care walking on rip rap. Rocks can shift unexpectedly and serious falls are possible.
- Use caution when kick sampling in swift or deep water. Wear a suitable PFD and consider using a safety tether held by an assistant. For most people, conditions are rarely suitable for collecting a good kick sample in water deeper than 0.6 m.
- Do not attempt to kick sample vertical or near-vertical banks.
- Professional-quality breathable waders with a belt are recommended for kick sampling. Neoprene booties are an alternative, but should have sturdy, puncture-resistant soles.
- Avoid wet-wading in areas down-river from effluent discharge points.
- Use caution approaching and sampling snags. Good communication between the crew and captain is essential to avoid grounding and injury.
- Use safety glasses and gloves when handling formalin.

Table 10-7. Equipment and supply checklist for macroinvertebrate sampling. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-6.

Qty	Item	
1	Modified rectangular kick net ("also called Slack Sampler") with 500-µm mesh and handle (e.g., Wildco 425-M53 or equivalent)	
2	300-mL Dolphin plankton bucket with 500-µm mesh (e,g,. Wildco 47-D60 or equivalent)	
1	0.20-m diameter snag net with 500-µm mesh and 200-mL Dolphin bucket (Wildco 424-C56 or equivalent)	
1	0.33-m diameter snag net with 500-µm mesh and 200-mL Dolphin bucket (Wildco 424-A56 or equivalent)	
1	US standard 35 sieve (500-µm mesh) 30-cm diameter, stainless-steel mesh	
1	Wash bucket with (500-µm mesh) (e.g., Wildco 190-E25) (optional)	
2-3	20-L plastic bucket for transporting composite between stations and catching wastewater during when washing the sample	
1 roll	Biodegradable flagging (different color from tape use to lay out the site)	
1	Velocity meter	
2 pr.	Forceps for removing invertebrates from nets	
1	1-L wash bottle	
1	Long-handled scrub brush for snag sampling ("deck brush" style works best)	
1	Small brush for kick sampling	
1	Large bore funnel for transferring samples from sieve to jar	
at least 6	HDPE sample jars, wide mouth, leakproof, screw top, 1 L capacity (for kick samples)	
at least 4	HDPE sample jars, wide mouth, leakproof, screw top, 250-mL capacity (for snag samples) (Fisher Scientific 03-311-3D or equivalent)	
1 roll	Plastic electrician's tape for sealing sample jars	
1 set	Labels	
at least 3 L	10% carbonate-buffered formalin	
1 L	Concentrated rose bengal solution (see Table 3-1)	
1	Littoral and snag sampling form	

10.8 Literature cited

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Figure 10-3. Snag and kick nets. The two sizes of snag nets are 0.20 m (left) and 0.33 m diameter (middle). For snags >0.33 m in diameter, the standard D-frame kick net (right) is used.

Draft

EMAP-GRE LITTORAL AND SNAG SAMPLING FORM

Reviewed by (Initials): ________

SITE ID: GRW04449-________ DATE: ___ / ___ / 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2

Dominant Substrate at Kick Sample Location (Distance from start of transect in parentheses)					
	Substrate	Flag		Substrate	Flag
A (0)	XB SB CB GC GF SA FN WD HP OT		G (300)	XB SB CB GC GF SA FN WD HP OT	
B (50)	XB SB CB GC GF SA FN WD HP OT		H (350)	XB SB CB GC GF SA FN WD HP OT	
C (100)	XB SB CB GC GF SA FN WD HP OT		I (400)	XB SB CB GC GF SA FN WD HP OT	
D (150)	XB SB CB GC GF SA FN WD HP OT		J (450)	XB SB CB GC GF SA FN WD HP OT	
E (200)	XB SB CB GC GF SA FN WD HP OT		K (500)	XB SB CB GC GF SA FN WD HP OT	
F (250)	XB SB CB GC GF SA FN WD HP OT		Total number of kick samples:		Flag:

CODE	SIZE CLASS	SIZE RANGE (mm)	DESCRIPTION/COMMENTS
XB	Large boulders	>1000	Includes some riprap
SB	Small boulders	>250 to 1000	Basketball size and larger; Includes some riprap
CB	Cobbles	>64 to 250	Tennis ball to basketball size; includes some riprap
GC	Gravel (coarse)	>16 to 64	Marble to tennis ball size
GF	Gravel (fine)	>2 to 16	Ladybug to marble size
SA	Sand	>0.06 to 2	Gritty between fingers
FN	Fines	<0.06	Silt, clay, muck, not gritty between fingers
WD	Wood	Any size	Describe in comments
HP	Hard pan		Firm, consolidated fine substrate, packed clay
OT	Other		Describe in comments

Composite Kick Sample

Sample ID	No. of Jars	Comment

Periphyton Composite Sample	Sample ID	Comp. Vol. (mL)	# Stations Sampled	No. of Jars	Comment

Sediment Composite Sample	Sample ID	Comp. Vol. (L)	# Stations Sampled	Comment
		.		

LWD Tally (Record total in small box)

LWD = ≥ 5 m long ≥ 0.3 m diameter at larger end

	Wet LWD	Beached LWD	Anchored LWD	Flag
0.3 - 0.6 m (12 - 24")				
> 0.6 m (> 24")				

Channel LWD Density (pieces/ha)	☐ None ☐ 1 - 2 ☐ 2 - 5 ☐ 6 - 10 ☐ >10	Flag

Flag codes: K=no measurement made, U=suspect measurement; F1, F2, etc=misc flags assigned by field crew. Explain in comments.

32.

Figure 10-4. Littoral and snag sampling form (front).

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EMAP-GRE LITTORAL AND SNAG SAMPLING FORM (back)

Reviewed by (Initials): ______

SITE ID: GRW04449-______ DATE: ___ / ___ / 2 0 0 _ ANNUAL VISIT NUMBER: ☐ 1 ☐ 2

GPS Coordinates of Snag

Latitude ___ . ______

Longitude ___ . ______

Flag ___

Snag Characteristics

Net Used	Flag	Length of Sample (m)	Flag	Snag Surface	Flag
SS LS KN					

Depth (m)	Flag	Dist. from Shore (m)	Flag	Velocity m/s	Flag

Snag Diameter

- ☐ 15 - 25 cm
- ☐ 25 - 35 cm
- ☐ 35 - 45 cm
- ☐ 45 - 55 cm
- ☐ > 55 cm

Flag ___

Snag Surface Codes
SB = Smooth, no algae
SA = Smooth, algae
RB = Rough, no algae
RA = Rough, algae
BB = Bark on, no algae
BA = Bark on, algae

Velocity Conversions
1 k/h = 0.28 m/s
1 mile/h = 0.45 m/s

Snag nets
SS = Small snag
LS = Large Snag
KN = Kick Net

Snag Sample

Sample ID	No. of Jars	Comment

Flag	Comment

Flag codes: K=no measurement made, U=suspect measurement; F1, F2, etc=misc flags assigned by field crew. Explain in comments.

33.

Figure 10-5. Littoral and snag sampling form (back).

BENTHOS (10% formalin)
BK BS
GRW04449-___ ___ ___
___/___/ 200__
Site visit number 1 2
300011
Jar ___ of ___

BENTHOS (10% formalin)
BK BS
GRW04449- ___ ___ ___
___/___/ 200__
Site visit number 1 2
Sample ID __________
Jar ___ of ___

BENTHOS
BK BS
GRW04449-___ ___ ___
___/___/ 200__
Collector_______________

Figure 10-6. Labels for benthic macroinvertebrate samples. BK = kick sample; BS = snag sample. The label at upper left would be affixed to the outside of a jar. The continuation label at upper right is used if more than one jar is needed. The bottom label would be placed inside each jar. Not actual size.

Section 11
Periphyton and Sediment

Brian H. Hill[1] and James M. Lazorchak[2]

Periphyton includes algae, fungi, bacteria, protozoa, and associated organic matter on the surface of aquatic substrata. Periphyton assemblage composition is a useful indicator of environmental condition because it responds rapidly to a number of anthropogenic disturbances, including habitat alteration, excess nutrients, metals, herbicides, hydrocarbons, and acids (Pan et al. 1996, Hill et al. 2003).

Benthic organisms are in intimate contact with river sediments. Benthic assemblages are influenced by the physical and chemical properties of sediment. Sediment characteristics serve as exposure indicators for benthos, fish, and other wildlife (e.g., sediment toxicity) and as functional indicators of key ecosystem processes (e.g., sediment enzyme activity) (Sinsabaugh and Foreman 2001, Hill et al. 2002). Periphyton and sediment collection methods described herein are adapted from previous EMAP methods for wadeable and non-wadeable streams (Peck et al., unpublished drafts).

11.1 Periphyton sample collection

At each site, a 500-m main channel shoreline (MCS) transect, starting at the intersection of the cross-channel transect and the MCS (Figure 4-1), is laid out by either the fish- or river-sampling crew, depending on which crew arrives at the site first. The primary transect is initially flagged at 100-m intervals; intermediate littoral stations at 50-m intervals are located and flagged using a handheld GPS or by visual estimation during littoral sampling (use a different flag color than at the 100-m stations). At each of 11 evenly-spaced stations along the 500-m MCS transect (every 50 m; Figure 4-2), a 25-cm^2 littoral periphyton sample is collected from the dominant hard substrate at the station. If no hard substrates are present, fine substrates

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1 U.S. Environmental Protection Agency, Office of Research and Development, National Health and Environmental Effects Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804

2 U.S. Environmental Protection Agency, Office of Research and Development, National Exposure Research Laboratory, Ecological Research Division, 26 W. Martin Luther King Dr., Cincinnati, OH 45268

are sampled. For guidance on where to collect littoral samples in a dike field, see Figure 10-1. Table 11-1 describes the periphyton sample collection procedures in detail.

11.2 Sediment sample collection

At or near each of the 11 periphyton sampling locations, a fine-sediment sample is collected using either a hand scoop or a “petite Ponar” grab sampler. The objective is to collect a 4-L composite sample that is representative of MCS depositional areas at the site. The composite sample will be subsampled in the lab for multiple analyses. Table 11-2 describes the sediment sample collection procedures in detail.

11.3 QA Considerations for periphyton and sediment sampling

Standardization of effort and attention to detail are important for maintaining a high QA standard for field sampling. Several QA considerations for periphyton and sediment sampling are presented in Table 11-4.

11.4 Safety considerations for periphyton and sediment sampling

Safety is paramount. General and boat-related safety guidance is presented in Section 2. Safety considerations relevant to periphyton and sediment sampling are presented in Table 11-5.

11.5 Equipment and supplies

Table 11.6 is a checklist of equipment and supplies required for collecting periphyton and sediment samples. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

Table 11-1. Procedure for collecting periphyton samples.

1. Go to MCS station A at the intersection of the cross-channel transect and the main channel shoreline (Figure 4-2). Periphyton is collected at locations that correspond to macroinvertebrate kick sample locations (see Section 10).

2. Locate periphyton samples in a zone bounded on the shore side by the apparent low-water mark from daily flow fluctuations (most relevant on the regulated Upper Missouri River) and bounded on the river side by the 0.3-m (about mid-biceps) depth contour. This is the recommended maximum sample depth. The low-water mark at a site can often be detected by the presence of periphyton or attached filamentous algae just below the low-water mark. If samples cannot be safely collected at a station due to vertical banks or other reason, search 5 m up- and down-river for a safe location. If a safe location is still not available, do not collect a periphyton sample at that station and note the missing sample for the station with a flag on the form.

3. At each station, select a piece of hard substrate (coarse gravel, cobble, wood) that can be easily removed from the river bottom (usually <15 cm diameter). If substrate is heterogenous, select an example of the dominant substrate type at the station. Be sure to avoid the area that has just been kick sampled. Sampling just upriver from the kick sample location is recommended.

4. Place the substrate in a large funnel draining into a 500-mL sample composite bottle. Brush a 5 x 5 cm (2 x 2 inch) area on the upper surface of the substrate using a stiff tooth brush (supplied by EPA). Use of a delimiter or template is not required. Rinse the loosened periphyton and toothbrush bristles into the funnel using filtered river water in a wash bottle. Discard the brushed substrate.

5. If the substrate can be removed from the water but not held over the funnel (e.g., LWD), use a knife blade to scrape a 5 x 5 cm (2 x 2 inch) area from the upper surface and wash the knife blade into the funnel.

6. If there are no hard substrates at the sample station, use a 60-mL syringe to vacuum up 25 cm^2 of fine substrate (silty sand, silt, clay, muck) to a depth of approximately 2 cm. Alternatively, press a small petri plate into the substrate and slip a spatula under it and remove it from the substrate. In deep areas, this type of sampling might not be feasible. If no periphyton sample can be collected at a station, flag the data and note the reason in a comment on the littoral and snag sampling form (Figure 10-4).

7. If there are no sufficiently large substrates for a brush sample, or sufficiently fine substrates for a syringe sample (e.g., if the station is all hardpan or fine gravel), search 5 m up- and downriver for the nearest suitable substrate (coarse or fine) and sample it. If there are still no suitable substrates, do not collect a periphyton sample at that station and flag the data on the and note the reason in a comment on the littoral and snag sampling form.

Continued

Table 11-1. Procedure for collecting periphyton samples, continued.

8.	Repeat steps 2-7 at each of the 11 MCS stations. Record the total number of replicates (stations sampled) included in the composite.
9.	Fill the composite jar 2/3 full with river water, cap it, and place it in a cooler with ice (keep the sample dark until it is preserved). For sample jars that are not pre-labeled, the site number and sample type should be written directly on the jar with a Sharpie (e.g., "peri 045") to avoid confusion later. This notation should be covered later with the outside of the jar sample label.
10.	Go to Table 11-3 for procedures for labeling and preserving samples.

Table 11-2. Procedure for collecting sediment samples.

1.	At each of 11 evenly-spaced stations on the MCS (i.e., 0, 50, 100 m from the down-river end of the transect; Figure 4-2), that correspond to macroinvertebrate kick sample locations (see Section 10), collect a sediment sample.
2.	Locate sediment samples in areas or patches of fine substrate (silty sand, silt, clay, muck) in a zone bounded on the shore side by the apparent low-water mark from daily flow fluctuations (most relevant on regulated Upper Missouri River) and bounded on the river side by the 0.3-m (usually about mid biceps) depth contour (recommended maximum sample depth; deeper sampling may be possible). The low-water mark at a site can often be detected by the presence of periphyton or attached filamentous algae just below the low-water mark. If samples cannot be safely collected by wading at a station due to vertical banks or other reason go to step 5.
3.	Be sure to avoid the area that has just been kick sampled. Sampling up-river from the kick sample location is recommended. If fine substrates are not present within 5 m up- or downriver from the station, do not collect sediment at that station and flag the station on the form.
4.	If fine substrate is present, use a small scoop to collect a sample of about 225 cm^2 (≈ 15 x 15 cm [6 x 6 inches]) of the top 2 cm of substrate (this volume is approximately equal to six scoops). Place the sample in a clean bucket. Use gloves for handling sediment. Do not assume rip rapped shorelines lack fine sediment. Look for fines between the large rocks
5.	If wading is not possible, use a petite Ponar sampler or similar device deployed from the boat to collect a sediment sample adjacent to the station. Release the petite Ponar sample onto a tub and use the scoop to collect about 225 cm^2 (≈15 x 15 cm [6 x 6 inches]) of the top 2 cm of the sample. Estimate sample area visually. Place the subsample in the sediment composite bucket and discard the rest of the Ponar sample.
6.	Repeat steps 2-5 at each of the 11 littoral stations. Record the total number of replicates (stations) included in the composite. Note in a comment the stations at which sediment was collected using a non-wading method.
7.	It is important that a sufficient sediment (not less than 4 L) sample for analysis be collected. If multiple stations have no fine sediment, it is permissible to collect extra sample at stations that do have fine sediment or between stations. Be sure to note this in a comment.
8.	Using a large stainless steel spoon, thoroughly mix the composite sample in the bucket and transfer 4 L of the composite in a 30 x 50-cm 3-mil thick polyethylene bag. Try to limit the amount of sediment adhering to the inside of the bag near the top. Grasp the bag just above the sediment to express the air. Twist and knot the bag to seal it. Write the site number and date directly on the bag with a permanent marker and place it in a cooler with ice.
9.	Go to Table 11-3 for procedures for labeling and preserving samples.

Table 11-3. Procedures for labeling and preserving periphyton and sediment samples.

1.	To avoid clutter in the boat, periphyton and sediment samples may be transported to the ramp or base location (if it is close to the ramp) in a cooler with ice for final labeling and preservation.
2.	Periphyton. Add 20 mL of 100% borax-buffered formalin to the 500-mL periphyton composite, and top off the bottle with river or tap water (final formalin concentration of 4%). Prepare a label (Figure 11-1) for outside the jar. Using a fine-point permanent marker, fill in the site number, the sample date, and total preserved sample volume. Place the label on the jar and cover it with clear tape. Record the sample ID and other data on the littoral and snag sampling form (Figure 10-4)
3.	Seal each periphyton jar with plastic electrician's tape by wrapping with the threads (clockwise). Store the preserved samples upright in a secondary container to await transport or shipment to the laboratory.
4.	Sediment. Place the sediment sample inside a second 3-mil polyethylene bag, twist the top, and knot to seal. Prepare a label (Figure 11-1) for outside the outer bag. Using a fine-point permanent marker, fill in the site number and sample date. Place the label on the outer bag and cover it with clear tape. Record the sample ID and other data on the littoral and snag sampling form (Figure 10-4). Place the sample on ice or in a refrigerator. Do not freeze sediment samples.

Table 11-4. QA considerations for periphyton and sediment sampling.

- Try to make sure each of the 11 periphyton and sediment subsamples comprises an approximately equal portion of the total composite.
- It is permissible to collect sediment between stations to insure a composite volume of at least 4L. Note deviations from standard procedure in a comment.
- Do not assume rip rapped shorelines lack fine sediment. Look for fines between the large rocks.
- Mix the composite sediment sample thoroughly before extracting the final 4L composite.
- When sampling LWD pulled from the river, be sure to sample the upper surface of the LWD.
- Monitor sediment samples in your possession to insure they do not warm up or freeze.

Table 11-5. Safety considerations for periphyton and sediment sampling.

- Use extreme care walking on riprap. Rocks can shift unexpectedly and serious falls are possible.
- Use caution when sampling in swift or deep water. Wear a suitable PFD and consider using a safety tether held by an assistant. For most people, conditions are rarely suitable for collecting a periphyton or sediment sample in water deeper than 0.6 m.
- Do not attempt to collect periphyton or sediment from vertical or near vertical banks.
- Professional-quality breathable waders with a belt are recommended for littoral sampling. Neoprene boots are an alternative, but should have sturdy, puncture-resistant soles.
- Use caution using the Ponar-type samplers. The jaws are sharp and may close unexpectedly. Replace frayed lines and worn parts.
- Raise the Ponar sampler from and into a plastic tub rather than from the boat deck. This prevents feet from getting under the sampler.
- Don’t try to remove large pieces of LWD from the river by yourself.
- Use safety glasses and gloves when handling formalin.
- Avoid contact with sediment samples. Use gloves if necessary.

Table 11-6. Equipment and supply checklist for periphyton and sediment sampling. Generic supplies required for all EMAP-GRE field sampling are listed in Table 2-5.

Qty	Item	
1	Petite Ponar sampler (Wildco 1728-G30 or equivalent) with plastic tub, drop line, and spare pinch pin. A standard Ponar or similar device may substitute.	
1	Stiff-bristle tooth brush for brushing periphyton (Wildco 156-F40 or equivalent)	
1	Knife for scraping LWD	
1	60-mL syringe with 1-cm (3/8") hole bored into the end for sampling fine substrate	
1	Scoop for collecting sediment	
1	Graduated plastic bucket	
1	Large stainless steel spoon for mixing sediment composite	
1	1-L wash bottle	
1	Large bore funnel with $\geq$ 20-cm wide-opening	
1	HDPE sample jars, screw top, 500-mL capacity for periphyton samples (Fisher Scientific 03-311-3E or equivalent)	
2	30 x 50-cm, 3-mil polyethylene bags for sediment samples (Aquatic EcoSystems FSB5-10 "x 20" or equivalent)	
1 L	Borax-buffered formalin (100%)	
1	Pipette with 10-mL capacity or small plastic beaker for adding formalin to periphyton samples	
1 roll	Plastic electrician’s tape for sealing sample jars	
1 set	Sample labels	
1	Littoral and snag sampling form	

11.6 Literature cited

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SEDIMENT GRAB (SG)	PERIPHYTON (PA) (4% formalin)
GRW04449-___ ___ ___	GRW04449-___ ___ ___
___/___/ 200__	___/___/ 200__
Composite volume _____ L	Composite volume _____ mL
Site visit number 1 2	Site visit number 1 2
300255	300211

Figure 11-1. Set of sample labels for the site. Labels are affixed to the outside of the jar or bag (sediment). Bottom number is the unique sample ID. Not actual size.

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