Upper Mississippi River Clean Water Act Monitoring
Minnesota-Wisconsin Area Pilot Project
Field Operations Manual

Draft – September 11, 1015

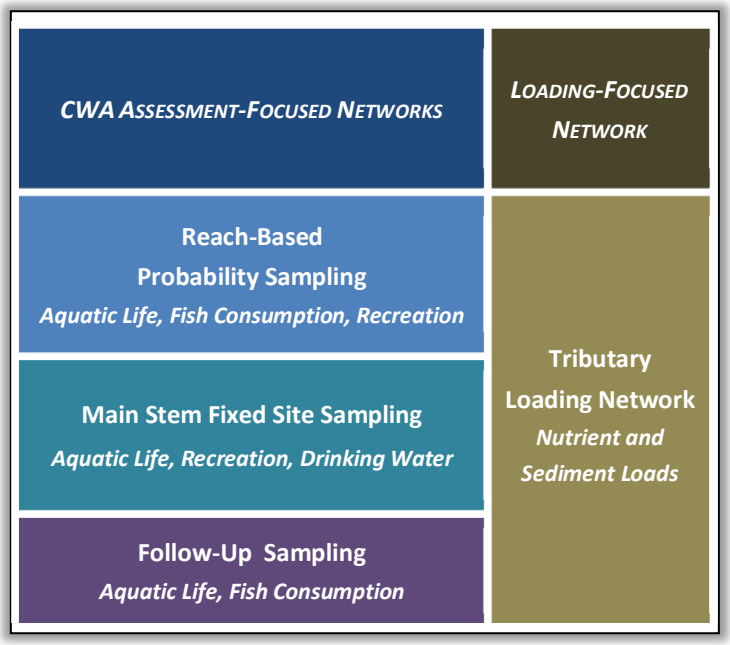
Table of Contents (currently also provides development status)

1. INTRODUCTION

1.1 UMR CWA Monitoring Plan Overview and Rationale

The *Upper Mississippi River Clean Water Act Recommended Monitoring Plan* (*UMR CWA Monitoring Plan*) was developed by the interagency Upper Mississippi River Basin Association Water Quality Task Force to address the lack of a coordinated, comprehensive Clean Water Act (CWA) monitoring approach on the Upper Mississippi River (UMR). The *UMR CWA Monitoring Plan* was adopted by the Upper Mississippi River Basin Association (UMRBA) Board in February 2014 and is structured as a series of networks that uniquely and comprehensively support assessment of aquatic life, fish consumption, recreation, and drinking water use attainment on the UMR; utilizing both newly collected data and existing data sets (Figure 1). This includes both fixed site and probabilistically-selected monitoring locations.

Figure 1: Illustration of UMR CWA Recommended Monitoring Plan, including constituent networks and designated uses which can be assessed utilizing data from these networks



1.2 Strategy Implementation and the Minnesota-Wisconsin Pilot Project

Following the UMRBA Board's approval, the states have moved forward to implement the *UMR CWA Monitoring Plan*. Implementation steps have included compiling existing water quality data in a "virtual pilot" effort, as well as planning for a field pilot to be conducted by the states of Minnesota and Wisconsin on a subset of the UMR's upper reaches, beginning in May 2016.

1.3 Field Operations Manual Scope and Applicability

This *UMR CWA Pilot Monitoring Project Field Operations Manual (Pilot Manual)* has been developed to provide the technical and procedural detail necessary for the states of Minnesota and Wisconsin, as well

as other partners, to implement UMR CWA pilot monitoring. While it will inform sampling procedures UMR-wide, the nature of the pilot is such that changes and improvements are expected to be made as a result of this initial effort. Therefore, it is likely that a revised operations manual will follow completion of pilot monitoring.

Further, the pilot monitoring project is somewhat reduced in scope from the "full" monitoring program described in the *UMR CWA Monitoring Plan*. Specifically, the following components are not part of the Minnesota-Wisconsin pilot monitoring project:

- There are no public water supply intakes on the shared Minnesota-Wisconsin portion of the UMR. As such, no monitoring will be done for drinking water use-only analytes (e.g., VOCs, SOCs, phenols, and fluoride).
- Metals will not be sampled at probabilistic sites during the pilot project.
- Algal toxins (microcystin and cylindrospermopsin) will not be sampled as part of the pilot project.
- (WI has indicated fish tissue monitoring may not be part of pilot in reaches 2-3. Need to confirm.)
- Follow-up sampling and monitoring for secondary indicators (e.g., sediment chemistry) is not explicitly addressed in the pilot, though such monitoring may occur at the discretion of the states.
- Index site monitoring will not be part of the pilot project.
- The tributary loading network will not be sampled as part of the pilot project.

Therefore, because this *Pilot Manual* is scoped only to describe sampling that will actually occur during the pilot, it does not necessarily provide all the information needed to implement full monitoring river-wide.

Additionally, this *Pilot Manual* is focused on field operations. As such, it does not provide extensive detail on sample site selection, laboratory analytical methods, data management, data analysis/score calculation, or CWA designated use assessment.

1.4 Acknowledgements

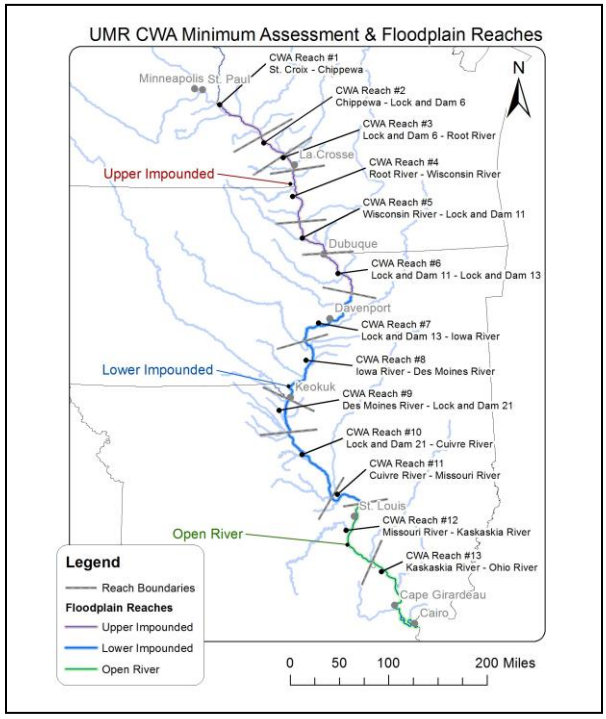
The content of this manual builds largely on the prior work of US EPA's EMAP-GRE, USACE's UMRR-LTRMP, state monitoring protocols, and the documents developed by the Midwest Biodiversity Institute (MBI) for the WQTF. These documents are referenced as appropriate throughout the specific sampling protocols. (Will add more, and more specific, acknowledgments here.)

2. MONITORING OVERVIEW AND GENERAL PROCEDURES

2.1 Geographic Extent

The *UMR CWA Recommended Monitoring Plan* organizes monitoring around the "minimum CWA assessment reaches" established via interstate Memorandum of Understanding in 2003. These reaches are illustrated in Figure 2. An additional reach internal to Minnesota (Reach 0) also falls under the *Plan*.

Figure 2: Minimum, interstate UMR CWA assessment reaches



The geographic extent of the pilot monitoring project, and hence of this *Pilot Manual*, is the main stem UMR from Upper St. Anthony Falls (USACE River Mile 854) to the Root River (River Mile 694). This includes the UMR CWA assessment reaches 0 through 3 (Table 1). All sampling takes place in the river's main channel and adjacent shoreline throughout the run of the river (i.e., Lake Pepin and other lake-like areas are included).

Table 1: Geographic extent of pilot monitoring program, UMR assessment reaches 0 through 3.

Reach Number	Reach Name (Description/8-digit HUC code)	River Miles	Segment Length (miles)
0	**Assessment Reach 0** (Upper St. Anthony Falls to St. Croix River)	**854-811.5**	**42.5**
1	**Assessment Reach 1 (Rush-Vermillion)** (St. Croix River to Chippewa River/ HUC 07040001)	**811.5-763.4**	**48.1**
2	**Assessment Reach 2 (Buffalo-Whitewater)** (Chippewa River to Lock and Dam 6/ HUC 07040003)	**763.4-714.2**	**49.2**
3	**Assessment Reach 3 (La Crosse-Pine)** (Lock and Dam 6 to Root River/HUC 07040006)	**714.2-693.7**	**20.5**

2.2 Scope of Indicators Monitored

The *UMR CWA Monitoring Plan* specifies sampling across a wide extent of parameters. As previously described, the pilot monitoring project will include most, but not all, of these parameters, as displayed in Table 2. In brief, the pilot focuses on fixed site and the probabilistic networks, while dropping some indicators (e.g., probabilistic network metals) that are specified in the full *Plan*.

Table 2: Scope of parameters monitored in pilot project, where: ***X****= sampled in pilot, shaded=not sampled in either pilot or full plan, shaded X = sampled in full plan, but not sampled in pilot*

Indicator Group	Indicators	Probabilistic Monitoring (15 sites per reach)	Mainstem Fixed Network (20 sites UMR-wide)	Tributary Loading Network (34 sites)
Biological Communities	Fish	**X**		
	Vegetation	**X**		
	Macroinvertebrates	**X**		
Fish Tissue	Mercury (Hg)	**X**		
	PCBs	**X**		
Field	Water Temperature	**X**	**X**	X
	DO (conc.& sat)	**X**	**X**	X
	pH	**X**	**X**	X
	Conductivity	**X**	**X**	X
	Turbidity	**X**	**X**	X
	Secchi Depth	**X**	**X**	
Nutrients	NO3+NO2	**X**	**X**	X
	TN	**X**	**X**	X
	NHx	**X**	**X**	X
	TP	**X**	**X**	X
	DP	**X**	**X**	X
	Chlorophyll a	**X**	**X**	X
Bacteria	*Escherichia coli*	**X**	**X**	
Algal Toxins	Microcystin		X	
	Cylindrospermopsin		X	
Miscelleanous	BOD	**X**	**X**	
	Chloride	**X**	**X**	
	Sulfate	**X**	**X**	
	TSS	**X**	**X**	X
	TOC or DOC		**X (TOC)**	
	Hardness (Ca & Mg)	**X**	**X**	X
	Alkalinity	**X**	**X**	
	Fluoride		X*	
Metals	Aluminum (Al)	X	**X**	
	Calcium (Ca)	X	**X**	
	Cadmium (Cd)	X	**X**	
	Chromium (Cr)	X	**X**	
	Copper (Cu)	X	**X**	
	Iron (Fe)	X	**X**	
	Lead (Pb)	X	**X**	
	Magnesium (Mg)	X	**X**	
	Potassium (K)	X	**X**	
	Sodium (Na)	X	**X**	
	Zinc (Zn)	X	**X**	
Other	Arsenic (As)	X	**X**	
	Mercury (Hg)	X	**X**	
	Selenium (Se)	X	**X**	
Organics	VOCs, Pesticides, Other		X*	
	Phenols		X*	
Physical Habitat and Characteristics	Substrate	**X**		
	Depth	**X**		
	Velocity	**X**		
	Discharge		**X**	X

*Only sampled at fixed sites in proximity to a drinking water intake.

2.3 Overview of Fixed-Site and Probabilistic Monitoring

As described previously, the pilot project focuses on the fixed site and probabilistic network, which are central to providing a robust characterization of the UMR's condition under the *UMR CWA Monitoring Plan*. A brief description of these networks and the sampling conducted under each of them follows. Further detail is provided under the indicator group-specific procedures later in this manual.

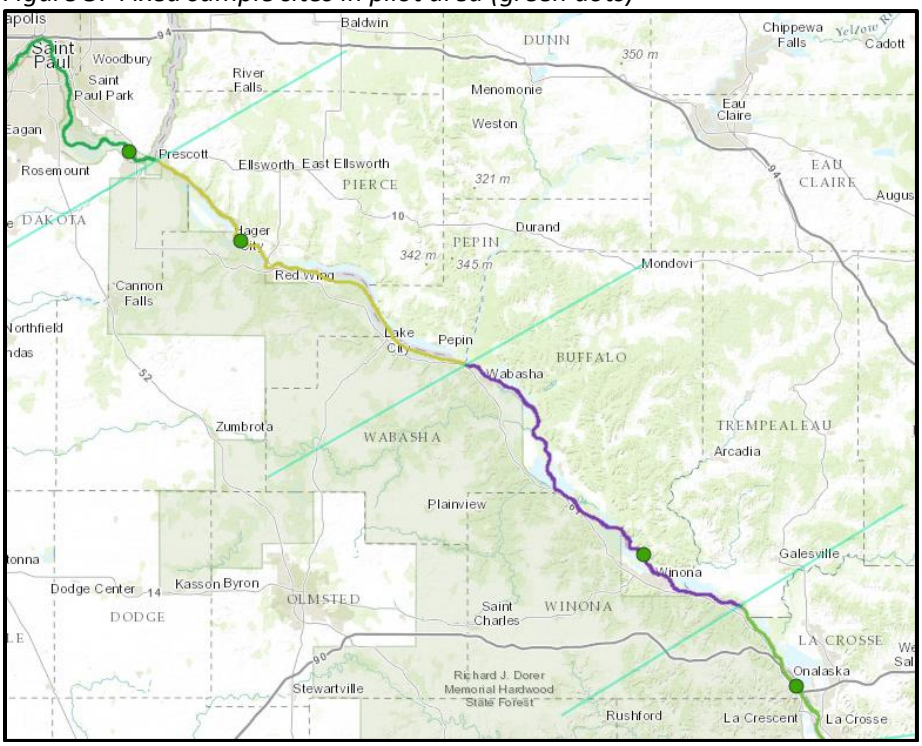
2.3.1 Fixed Site Network

Site Locations: The fixed site network is composed of 20 stations on the UMR, with at least one fixed site placed in each of the CWA assessment reaches and additional sites in reaches where a drinking water intake is present. There are no drinking water intakes in the pilot reaches. Therefore, in the area covered by the pilot monitoring project, there are just four fixed sites as shown in Table 3 and Figure 3. Sampling responsibilities for these fixed sites are also indicated in Table 3.

Table 3: Fixed sites in pilot monitoring area.

Reach	River Mile	Location Description	Agency Sampling During Pilot	Agency Previously Sampling Location (and Site dentifier)	Nearby USGS Gage	HUC-8	Latitude	Longitude
0	815.3	Lock & Dam 2	MCES	MCES (UMR 815.6)	05344500	7040001	44.76100	-92.86900
1	796.9	Lock & Dam 3	MCES	MCES/WDNR (UM 796.9)	05344980	7040002	44.61200	-92.61000
2	728	Winona	MPCA	MPCA (S000-096)	05378500	7040003	44.08200	-91.66400
3	698	Lock & Dam 7	WI DNR	MPCA (S000-067)	05386400	7040006	43.86634	-91.31008

Figure 3: Fixed sample sites in pilot area (green dots)



Indicators: Monitoring at fixed sites is conducted on a monthly basis year-round for field parameters, nutrients, discharge and other water chemistry. Bacteria *(E. coli)* is sampled monthly during the months of April through October. See Table 2 and Table 4 (below) for details regarding indicators monitored. As there are no drinking water intakes in the pilot reach, drinking-water only contaminants (e.g. VOCs, pesticides, phenols, fluoride) will not be sampled at fixed sites during the pilot.

Table 4: Fixed Site Monitoring Summary

Spatial Design	Index Period	Number of Sites	Media & Frequency (per index period)
Mainstem Fixed Network	Year Round (chemistry, discharge) April to October (bacteria)	15 UMR sites	Water Chemistry: 12x *E. coli*: 7x Discharge: 12x (from existing USGS and USACE gaging stations)

General Sampling Procedures: There are several important distinctions between fixed site and probabilistic site sampling. Perhaps most prominently, there is no biological sampling at fixed sites, so procedures are focused on water chemistry and other physical measurements. Therefore, it is expected that one sampling crew can collect all of the parameters needed at the fixed sites. (correct?)

Further, fixed sites are typically located at "hard structures" on the river (e.g., bridges, locks and dams) which do not require the use of a boat. Additionally, fixed site monitoring also extends year-round, so it will be necessary to consider any special techniques that apply during winter months at these sites (what happens in winter?). For these reasons, the sampling procedures for fixed sites will differ from those used at probabilistic sites.

By definition, site location is predetermined for fixed sites, so site selection process is straightforward and sites can be found on the online WQTF viewer (and site descriptions/site file/dossiers?). Further, each of the fixed sites in the pilot area corresponds to a location either currently or previously sampled by a state, federal, or local entity. As such, sampling crews may wish to communicate with other these agencies in advance regarding site access and other site considerations. This will also help in identifying any special equipment needed to reach the water surface, to be used in winter, etc. In general, samples should be taken at the river centerline (i.e., navigation sailing line), but this can be adjusted as needed in light of physical structures, access, etc.

Samples requiring lab analysis (e.g., those for metals, nutrients, miscellaneous, *E. coli,* and "other" parameters) will be collected as grab samples. Sample depth, like that of probabilistic samples, will be one meter. Other samples are field collected and results will be recorded on site. Please see the parameter-specific instructions for details on fixed site sampling for particular indicators.

(Any more to say here? More description of what a fixed site typically looks like/how it functions/equipment needed? Is there a standard reference for this? Or does that all go in the parameter-specific section? There are so few fixed sites we could maybe even append site dossiers for these to the manual/on web viewer.)

2.3.2 Probabilistic Network

Site Locations: Fifteen probabilistic sites are identified within each CWA assessment reach (i.e., a total of 60 sites in the pilot project area). These sites are allocated along the river's centerline (defined by

USACE navigation sailing line) using a site draw conducted by US EPA. An overdraw is part of this allocation, to be used as needed should initially identified sites not prove feasible for sampling. Sample sites are shown in Figure 4. Assignment of monitoring responsibilities for probabilistic sampling is described in Table 5. Note that vegetation is separated out as it requires a differing sampling design.

Figure 4: Probabilistic monitoring locations in pilot area (blue dots)

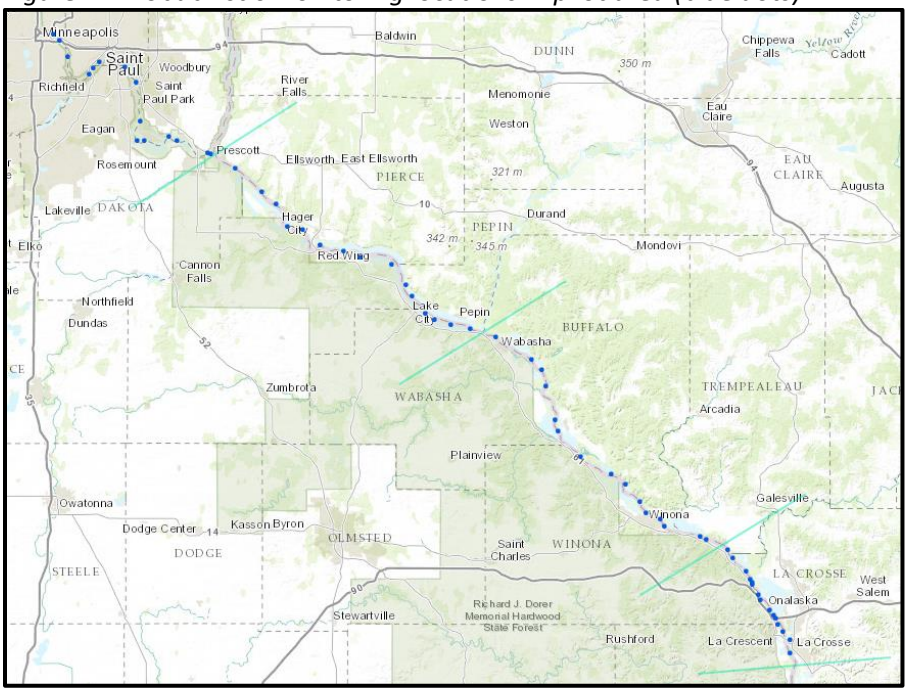


Table 5: Probabilistic Monitoring Responsibilities (need to confirm these)

Reach	Probabilistic-Chemistry, Fish Tissue, Fish Assemblage, Macroinvertebrates	Probabilistic -Vegetation
0	MPCA	MN DNR
1	MPCA	MN DNR
2	WI DNR	WI DNR
3	WI DNR	WI DNR

Indicators: Biology (fish and macroinvertebrates), fish tissue, water chemistry, field parameters, site characteristics, and discharge are all sampled at the probabilistic locations during a July to September index period. See Table 2 and Table 6 for details. Biological and fish tissue samples are only collected during one sampling event, but water chemistry and other parameters, including field parameters and discharge, are sampled three times during the index period. Vegetation is sampled separately.

Table 6: Probabilistic Monitoring Summary

Spatial Design	Index Period	Number of Sites	Media & Frequency (per index period)
Reach-Based Probabilistic	July to September	15 sites per reach	Fish and habitat: 1x Macroinvertebrates: 1x Water Chemistry: 3x
		100 sites per reach	Submersed Aquatic Vegetation (SAV): 1x

General Sampling Procedures: Unlike fixed sites, probabilistic sites will change in every round of sampling and will not typically fall at hard structures on the river. Additionally, biological sampling takes place at probabilistic sites. As such, probabilistic sampling is boat-based and more equipment-intensive. It will likely require more than one crew to complete all the sampling types necessary. (What do we think on crews? Separate crews or one does all? What is implication of E. coli holding time? How about fish tissue sampling?)

More detail on indicator-specific procedures is provided later in this document, but in general for probabilistic sampling:

- **Water chemistry sampling will be done via grab samples and take place at the centerline "x point."** Crews will sample at the centerline "x point" identified in the sample draw from USEPA. (confirm this choice – x point sits right in the navigation channel, is this a concern either in terms of safety or impact on water quality values?) Water quality samples will be collected at one meter depth using the method selected by the sampling entity (bucket, Van Dorn, etc.). (is it OK to leave this open or do we want to specify?)

- **Biological sampling utilizes 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 pre-designated during sample selection. (This is taken pretty much directly from EMAP FOM.) 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 5). Fish assemblages (and site characteristics?) are sampled in the near-shore littoral zone along both MCS transects. Eleven locations, spaced 50 m apart along the primary MCS transect (Figure 6), define sample locations for macroinvertebrates (and site/riparian measurements?) (This assumes only EMAP kick-net sampling. Would have to modify for inclusion of Hester-Dendy.)

- **If the specified <u>bank</u> cannot be sampled for safety, access, or other reasons** (Are there other reasons why a bank or site may not be sampled?) **then the opposite bank may be sampled.** This modification must be noted in sampling documentation.

- **If the specified <u>site</u> cannot be used (i.e., neither bank can be sampled), a replacement site from the overdraw pool should be used.** This modification must be noted in sampling documentation. (Anything else we want to say here?)

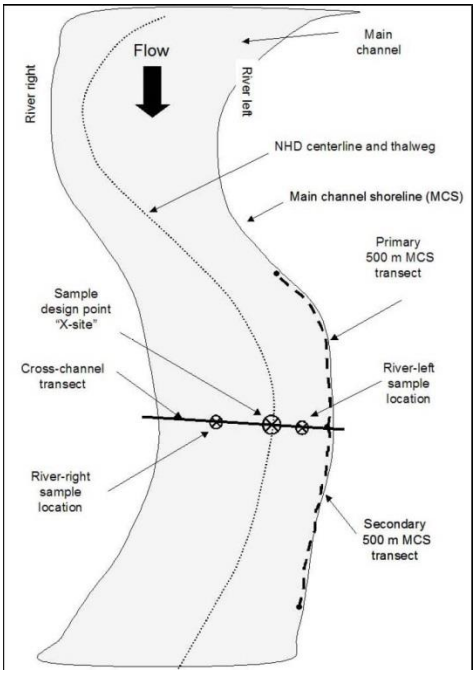


Figure 5. Idealized EMAP-GRE sampling site. (This is straight from EMAP, needs modification. How to best display for pilot sampling?)

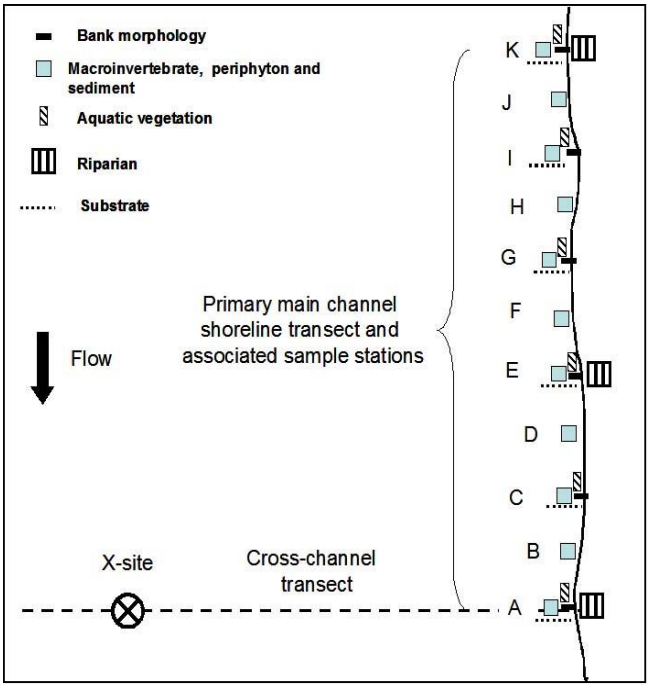


Figure 6: Main channel shoreline (MCS) sampling for EMAP-GRE sites (This is straight from EMAP, needs modification. How to best display?)

2.3.3 Probabilistic Network-Vegetation

(Note that this section is not as fully developed as previous ones, as we've not discussed much to date.)

Submersed aquatic vegetation monitoring is also implemented using a probabilistic design. However, due to the patchiness in vegetation occurrence on the river, a much more intensive spatial design is employed (i.e., 100 sites per reach). Individual sites are then aggregated into 15 "assessment samples" per reach (Maybe? This is just one option for aggregation.) Because of the differing spatial arrangement for vegetation, a separate sampling crew will carry out this monitoring. Vegetation samples will be taken in the same July-September index period as other probabilistic samples. See Table 6 as well as the indicator-specific sampling description later in this *Pilot Manual* for more details. (Note: There are plenty of sites already drawn for SMI, no need to ask for new draw)

(The following is text and figure excerpt from published SMI manuscript *Ecological Indicators 13(2012) 196-205*. It gives a general description of vegetation sampling for SMI. May want to adapt for use here.)

We placed a minimum of 300 random sampling stations (this was 100 per year for 3 years) in each of the seven HUCs (these are same as CWA reaches), and distributed the sampling stations into 15 sub-HUC sections called assessment samples. Each assessment sample contained aggregations of 18–23 stations, based on proximity to one another. In total, we surveyed 2131 stations that were aggregated into 105 assessment samples (15 per HUC), which had a mean length of 3.8 km (range = 0.6–10 km, median = 3.3 km). All stations within an assessment sample were located within a single navigation pool to minimize the direct influence of dams (Figure 8).

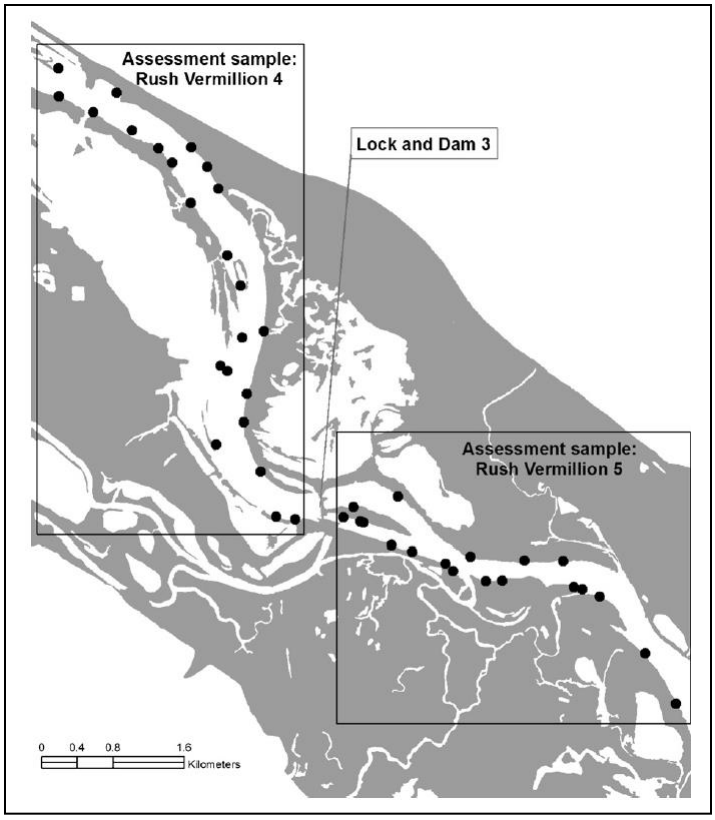


Figure 8: Example allocation of vegetation sites, including aggregation into assessment samples.

2.3.4 Site Dossiers

Site dossiers contain aerial imagery and other site attribute information used in site verification and sampling activities. An example is given in Figure 7. 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 import into GPS units. Crews may use a different local coordinate projection for navigation, but all data must reported as decimal degrees in NAD83 datum. (This is straight from EMAP FOM. Do we want to create site dossiers? If so, on paper? Or electronically such as via online viewer? Just for probabilistic or for all sites? If desired, who creates?)

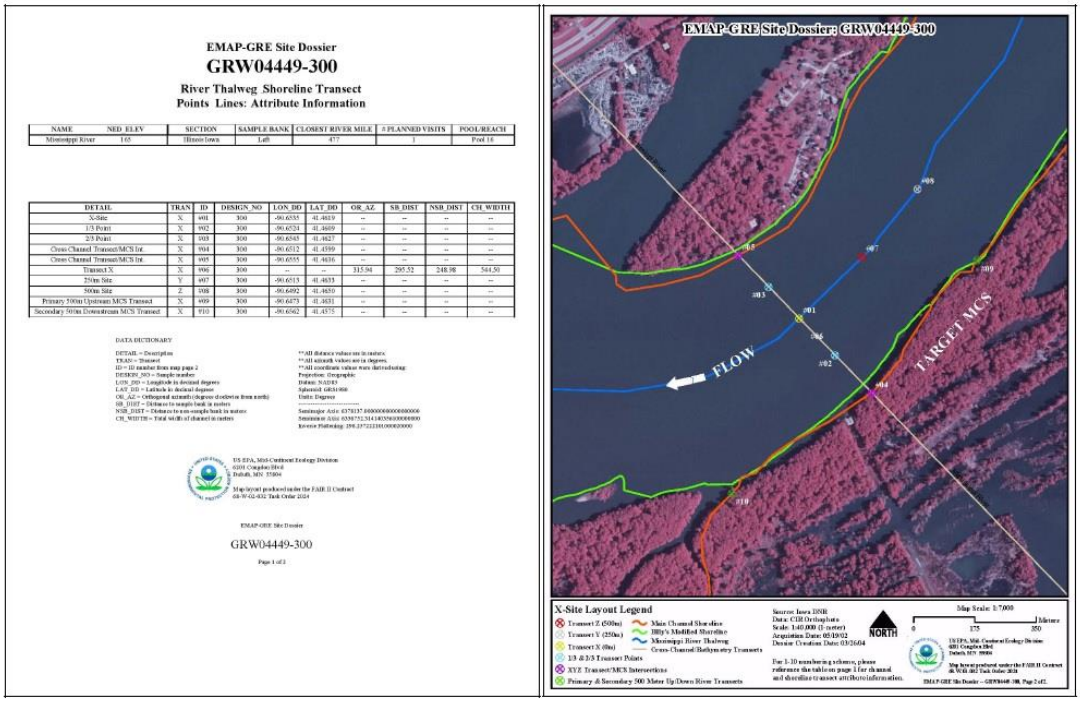


Figure 7: Example EMAP-GRE site dossier.

2.4 Laboratories

Three laboratories (?) will perform water chemistry analysis of samples collected in pilot monitoring as shown in Table 7.

Table 7: Water Chemistry Laboratories for Pilot Monitoring

Laboratory	Submitting Agency	Types of Samples Analyzed
Met Council Laboratory	Met Council Env. Services	Water chemistry, fixed sites.
Minnesota Dept. of Health	MPCA	Water chemistry, fixed and probabilistic sites.
Wisconsin State Laboratory	WI DNR	Water chemistry, fixed and probabilistic sites.

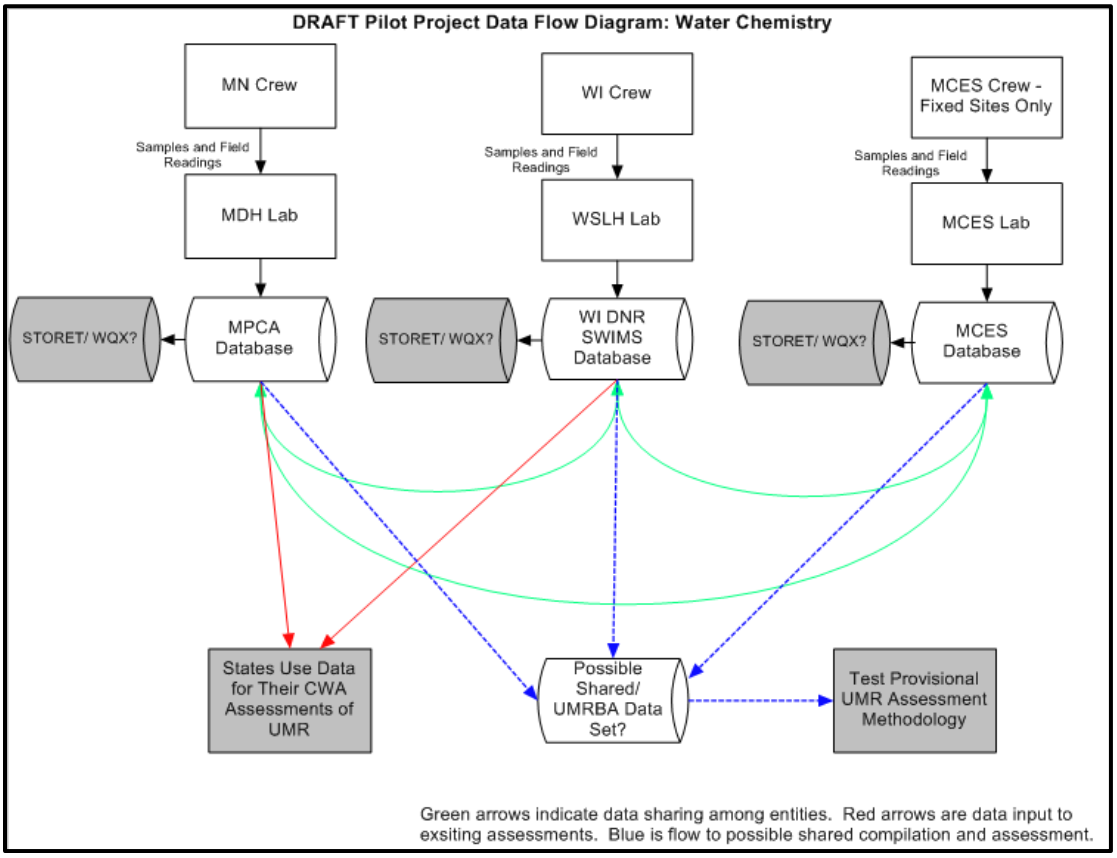
What about fish tissue? Macroinvertebrates? Fish (if needed)? What other labs to include?

2.4 QA/QC Procedures

We have only had some preliminary discussions of QA/QC procedures to date. Assume we will want to address in general here and then specifically under indicator-specific instructions, covering both inter-laboratory (splits) and field replicates, etc. Need to work this out further.

2.5 Data Management Procedures

Again, we'll presumably want to address data input/flow in general here and then also under individual indicator discussion as needed. In general, are we assuming this follows the typical flow for agencies augmented by sharing of data among the participating agencies. Figure below shows possible data flow for chemistry data in pilot project.



2.6 Overview of Field Operations

EMAP FOM had such a section covering issues including:

- Sampling crew makeup
- Daily setup/calibration/stock solution preparation/daily flow of sampling
- Safety and boat operations
- General equipment needs

Assuming we'll want some similar type of section in place.

3. INDICATOR GROUP-SPECIFIC PROCEDURES

3. 1 Water Chemistry and Physical Conditions

3.1.1 Indicators

The table below summarizes water chemistry and physical condition indicators. This includes field parameters, lab-submitted analytes, and physical habitat/characteristics (depth, flow velocity, etc.). (do these here or alongside fish?)

Table 8: Water Chemistry and Physical Condition Parameters

Indicator Group	Indicators	Probabilistic Monitoring (15 sites per reach)	Mainstem Fixed Network (20sites UMR-wide)	Notes
Field	Water Temperature	**X**	**X**	All these are field collected and recorded (what is mechanism for recording?)
	DO (conc.& sat)	**X**	**X**	
	pH	**X**	**X**	
	Conductivity	**X**	**X**	
	Turbidity	**X**	**X**	
	Secchi Depth	**X**	**X**	
Nutrients	NO3+NO2	**X**	**X**	Field/lab filtration required for many of these parameters, as is preservation by chemical and/or cooling means. See (?) for details.
	TN	**X**	**X**	
	NHx	**X**	**X**	
	TP	**X**	**X**	
	DP	**X**	**X**	
	Chlorophyll a	**X**	**X**	
Miscelleanous	BOD	**X**	**X**	Field/lab filtration required for some of these parameters, as is preservation by chemical and/or cooling means. See (?) for details.
	Chloride	**X**	**X**	
	Sulfate	**X**	**X**	
	TSS	**X**	**X**	
	TOC or DOC		**X (TOC)**	
	Hardness (Ca & Mg)	**X**	**X**	
	Alkalinity	**X**	**X**	
	Fluoride		X	
Metals	Aluminum (Al)	X	**X**	Preservation by chemical and/or cooling means required for these parameters. Field/lab filtration not required. See (?) for details.
	Calcium (Ca)	X	**X**	
	Cadmium (Cd)	X	**X**	
	Chromium (Cr)	X	**X**	
	Copper (Cu)	X	**X**	
	Iron (Fe)	X	**X**	
	Lead (Pb)	X	**X**	
	Magnesium (Mg)	X	**X**	
	Potassium (K)	X	**X**	
	Sodium (Na)	X	**X**	
	Zinc (Zn)	X	**X**	
Other	Arsenic (As)	X	**X**	
	Mercury (Hg)	X	**X**	
	Selenium (Se)	X	**X**	
Physical Habitat and Characteristics	Substrate	**X**		Exact means of measurement TBD, though discharge taken from nearest gage.
	Depth	**X**		
	Velocity	**X**		
	Discharge		**X**	

Will sampling procedures be the same or different for fixed and probabilistic sites? Beyond what is in the general summary in previous section, not sure what to put here. How much detail do folks need? How much can other protocols familiar to samplers simply be referenced?

3.1.2 Fixed Site Water Chemistry and Physical Conditions Sampling

Procedures TBD. Includes collection, field filtering & preservation, and lab sheet/data entry. Are there some existing procedures that we can reference/import here? Do we want to split out field parameters, lab analytes, and physical conditions when we describe procedures?

3.1.3 Probabilistic Water Chemistry and Physical Conditions Sampling

Procedures TBD. How different will probabilistic be from fixed site?

3.1.4 QA/QC for Water Chemistry Sampling

Will need to define some specifics here (replicates, etc.).

3.1.5 Equipment and Supplies

EMAP-GRE FOM had a section describing supplies needed for each indicator group. Do we want to do something similar or leave up to sampling entity? I've kept EMAP table in (below) as an example.

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 current
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-:m 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	200-mm Secchi disk affixed to a calibrated stick (0.01 m increments)
1	Plankton tow net with 63-:m 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-:m 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)
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 :m) 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

3.2 Fish Assemblage

Fish assemblage only sampled at probabilistic sites. **For now, have simply pulled in EMAP-GRE procedures in full. These procedures are excerpted over the next eight pages.** Given the likelihood of substantial changes, have not gone through and formatted with subheading numbering, etc.

Do we need to spell out up front the parameters we are looking for in order to support GRFIn calculation (e.g., CPUE-natives, proportion nonindigenous individuals, etc.) or just leave it to the lab sheet to get the raw data (species, DELTs, count, etc.) and go from there?

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 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 integrity 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 (Hughes and McCormick 2003). Habitat sampling methods are based on ORSANCO methods.

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 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.

Electrofishing

Boat specifications and electrode configuration (recommended): The standard EMAP-GRE electrofishing boat is a modified 5.5-m aluminum jon boat with an extra-thick puncture-resistant 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. All OK here? Do agencies have similar/compatible equipment available to them? Recall that we've also discussed net mesh size – ¼ inch (EMAP) vs. 1/8 inch (LTRMP).

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. 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.

For consistency, electrofishing should not begin until 1000h. Beginning at the upriver end of the 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 3,000 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) and the field 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.

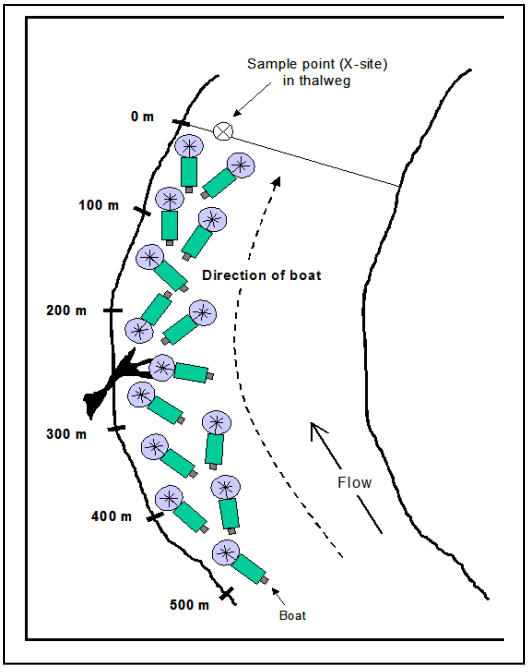


Figure X: Recommend path of electrofishing boat showing equal coverage of shoreline and channel later margins as well as complete application of the field through, over, or around cover objects. The zone should not extend greater than 30 m from shore, or to a depth greater than 6 meters.

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.

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 3,000 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 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.

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. (Note that we've said we'll only measure game fish, as length is not a GRFIn metric.) 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 handling 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 fish, record the AFS common name (Nelson et al. 2004). If a specimen is too small or otherwise cannot be easily identified to species in the field, it should be preserved and retained by the crew for later identification (Table 8-3). Do not record field data for fish that are retained as voucher specimens. During processing, select and retain fish for tissue analysis (see Section 9). Release all other fish. If needed, fill in the site ID and date on continuation forms (Figure 8-4). Be sure to release fish in a location where they will not be recollected during sampling of the secondary transect.
3	Examine each fish for DELTs (deformities, erosions, lesions, and tumors). Record the presence of DELTS on an individual fish or among fish in a batch using the codes in Table 8-4. Other abnormalities (e.g., blind eyes, pop-eye, fungus) can be recorded using flags.
4	The measuring board is divided into 3-cm size classes. Fish length is recorded by size class (e.g., fish < 3 cm long are in size class 1; fish >3 and < 6 cm are in size class 2, etc. Smaller fish may be sorted by species into 3-cm size-classes and weighed as a batch (count > 1).
5	Record the weight of each fish (or batch of small fish) in kg (1g = 0.001kg). Small fish in species batches too small to be weighed accurately with the smallest scale are not weighed and the data are flagged.
6	Preserve unknowns and voucher specimens as needed (Table 8-3). Note, in a comment, the approximate number of specimens preserved. A digital camera image is usually adequate for larger specimens (e.g., > 150 mm). Check the voucher box if a field-identified specimen has been preserved as a voucher specimen.

Unknowns and voucher specimens

Each crew member should become familiar enough with the large-river fish assemblages in the region to identify most species. Trained ichthyologists familiar with the fish species of the region should perform final taxonomic identifications of unknowns.

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 collect or photograph one voucher specimen for each different species encountered each year. It is strongly recommended that unknown or questionable small fish and all minnows should be preserved at every site for later identification by the fish-sampling crew. Table 8-3 describes preparation of photo vouchers and preserved specimen vouchers. Large or common species can usually be adequately documented by a photo.

An effort should be made to document with a photo or by collection any known or suspected non-indigenous exotic or invasive fish species that are captured. 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.

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 >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 leak-proof 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 >15-cm long should be slit open in the lower right abdomen to promote preservation.
5	Fill out a new 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, EMAPGRE 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.

External examination for anomalies

During processing, 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 comments

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 of larger fish are recorded on individual lines on the Fish Sampling Form (Figure 8-2). Smaller fishes of each species can be grouped into 3-cm size classes and weighed as a batch.

Fish habitat

(Not clear if we are discussing site characteristic data collection here or under chemistry. For now, leaving in the discussion of fish habitat. The following is excerpt from EMAP-GRE FOM.)

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 m, 100 m, 200 m, 300 m, 400 m, and 500 m from the downstream end; Figure 88), 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 3-5 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.
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 2-8, as appropriate, for the secondary transect.

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 ensuring 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 1000h.
Use a digital camera on a high resolution setting for taking photo vouchers and check image quality before releasing fish.
Do not cram fish voucher specimens into jars. They should be free floating so they are not fixed in a bent position.
Netters should wear polarized sunglasses.
When netting shocked fish, avoid size bias.
Avoid shorthand or colloquial common names for fish. Nelson et al. (2004) is the standard.

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, 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 so as 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.

3.3 Fish Tissue Contaminants

The following text is brought in from the fish tissue monitoring two-pager we've been working on, so does not exactly fit the format of other sections yet.)

Monitoring Purpose

Fish tissue monitoring under the UMR CWA plan is intended to have multiple purposes as follows:

- **To support characterization of fish consumption use status on the UMR at the "assessment reach" level**, pending the states' potential use of a shared, reach-based CWA assessment methodology.
- **To provide information to the UMR individual states** for purposes of their Section 305(b) assessments and Section 303(d) listings (i.e., their individual determinations of the attainment of fish consumption and/or human health uses).
- **To provide data useful in the UMR states' fish consumption advisory processes**, the outcomes of which may be incorporated into CWA assessment and listing decisions.
- **To help identify spatial patterns and trends over time** in levels of fish tissue contaminants.

Monitoring Components

Probabilistic Design**:** Under the *UMR CWA Recommended Monitoring Plan*, fish tissue samples will be collected as part of a probabilistic design, alongside the collection of fish (via electroshocking) for multimetric index score calculation. While this probabilistic design was adopted primarily for the purpose of aquatic life use assessment, it provides a platform wherein fish tissue samples can be efficiently collected and supports a consistent, reach-level method of characterizing fish consumption use status.

Index Period**:** The index period of fish tissue sampling is July to September.

Sampling Frequency**:** Under the *UMR CWA Recommended Monitoring Plan*, it is anticipated that each reach will be sampled once every five years. Additionally, follow-up samples (see below) may also be collected. As such, a subset of sites may be sampled more frequently than once every five years.

Number of Sites/Number of Samples: Under this probabilistic design, 15 sample sites will be assigned in each of the UMR assessment reaches. Across these fifteen sites, 10 fish per species group will be retained for tissue analysis in each reach (i.e., a 20 total samples per reach as there are two species groups). Discretion will be given to samplers to spread these samples to the greatest extent possible across the reach and across pools in the reach. This is a grand total of 280 samples for the UMR as a whole, reaches 0 to 13, over the course of a five year cycle. In instances where there is known and significant variation in tissue concentrations between pools within a reach (e.g., between Pools 1 and 2, and between Pools 5 and 5a), a total of 20 samples per species per reach may be collected.

Fish Species and Sizes Preferred: Fish will be sampled from a top predator group and a bottom-feeder group. In general, fish will be of a size that is representative of the "middle" of the age distribution for a particular species. The top predator species group to be utilized for purposes of the pilot is the black basses (smallmouth and largemouth bass), in the size range of 15-17" (4-6 years old). It is possible that different predator species groups will need to be utilized in areas of the river beyond the pilot project,

as black basses will not likely be available river-wide. For the bottom-feeder fish group, common carp will be sampled river-wide, with a size target of 18-21" (4-6 years old).

Sample Type**:** Skin-on fillets from individual fish will be prepared. (Do we need more detailed instructions here? Are the crews doing sampling familiar with tissue sample preparation?)

Analytes**:** All samples will be analyzed for mercury and total PCBs. Analytical methods used will be (TBD) for mercury and (TBD) for total PCBs. (What lab(s) will do the tissue analysis? Does this then determine the method to be used?).

Follow-up and Targeted Sampling**:** Once every five year probabilistic monitoring is considered "baseline" monitoring for the purposes of the UMR CWA plan. Additionally, samples may be taken in years subsequent to the baseline monitoring either to follow up on elevated results or at targeted locations as needed by states' fish consumption advisory programs.

Summary

Proposed fish tissue monitoring under the UMR CWA plan is summarized in the table below:

Sampling Type	**Index Period**	**Frequency**	**Number of Sites**	**Fish Species Group**	**Number of Fish Retained per Reach**	**Total Samples UMR-Wide**	**Sample Type**	**Analytes**
Baseline: Reach-Based Probabilistic	July to September	Once per five years	15 sites per reach (n=210 river-wide)	Top Predator (black basses)	10	280	Skin-on fillet	Mercury PCBs
				Bottom-Feeder (common carp)	10			
Follow-Up or State FCA Targeted	July to September	As needed, following baseline monitoring	As needed	As needed	As needed	TBD	Skin-on fillet	As needed

Sampling Logistics

The agency(s)/entity(s) involved in fish tissue sampling will be determined by the states as they choose in implementing the *UMR CWA Recommended Monitoring Plan*, keeping in mind that the initial sampling would be done by the same crew that is collecting fish assemblage information via electroshocking. Follow-up sampling may or may not be done by the same crew/agency that conducts initial sampling. States may also collaborate with each other to divide up monitoring along shared river reaches.

3.4 Benthic Macroinvertebrates

Applies only to probabilistic network. Specific method is TBD, pending outcomes of comparison study. For now, have simply inserted text from EMAP-GRE FOM. Due to likely changes, have not modified format to fit other sections of chapter.

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 (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 Klemm et al. (2003).

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 (use a different flag color than at the 100 m stations). At each of the resulting 11 evenly-spaced points along the 500-m MCS transect (stations A-K; Figure 4-2), two 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-:m 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).

Snag sampling (I am assuming we are not doing snag sampling, but leaving text in for now to confirm)

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., in press). Snags were historically the most important hard substrate in large alluvial rivers. In many reaches where extensive channel modification and bank

stabilization (riprapping) have not occurred, snags provide otherwise-rare stable substrate. 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 snag (33 x 51-cm frame; 500-:m mesh) can be used to sample the surface of the snag. Snag sampling methods are described in Table 10-2.

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 (Kaufmann 2003). 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. 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.

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 orginated 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.

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. The jar is then topped of with a concentrated rose bengal solution (Table 3-2). Fill out and place a label (Figure 10-6) 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, 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 sam ple 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.
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. 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 tim es 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.

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-:m m esh 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 sm all gravel in the sample, use a second 20-L bucket to elutriate the sam ple 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.

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.

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.

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 snag nearest to the intersection of the cross-channel transect and the MCS (littoral sam ple station A, Figure 4-2) and which meets the snag suitability criteria. Search both shorelines 1 km up river and down river from the transect. If no suitable snag is found, search on the way back to the ramp. If a piling or snag is sampled that does not m eet all the criteria in step 2, note this in the comments on the Littoral and Snag Sam pling 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 minim um 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 sam ple 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.
9	Use a velocity meter to measure surface water-velocity (m/s) above the sam pled 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 m ile/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 sam ple label. Place the sam ples in a cooler.
12	Go to Table 10-4 for procedures for labeling and preserving samples.

*Table 10-3. Procedures for quantifying LWD***.**

1	LW D 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 LW D tally. W hile 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. W et LWD is in the channel below the wetted perimeter. Beached LWD is between the wetted perimeter and the visually-estimated bankfull level. Anchored LW D 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 LW D. 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 Littoral and Snag Sampling Form (Figure 10-5).
3	Channel LWD. Note the presence of LW D 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. If there is > 0 and < 1 pieces of LWD/hectare, mark the 1-2 pieces/ha box.

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 3-2). 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 sam ple 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 or immediately after a barge has passed.
Always inspect the kick or snag net for invertebrates clinging to the mesh.
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 riprap. 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.
If a line is use to hold the boat against (abeam of) the snag for sampling in strong current, do not tie the line to the boat.
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-:m m esh and handle (e.g., Wildco 425-M53 or equivalent)	
2	300 m L Dolphin plankton bucket with 500-:m mesh (e,g,. Wildco 47-D60 or equivalent)	
1	0.20-m diameter snag net with 500-:m m esh and 200 mL Dolphin bucket (W ildco 424-C56 or equivalent)	
1	0.33-m diameter snag net with 500-:m m esh and 200 mL Dolphin bucket (W ildco 424-A56 or equivalent)	
1	US standard 35 sieve (500-:m m esh) 30-cm diameter, stainless-steel mesh	
1	W ash bucket with (500-:m mesh) (e.g., W ildco 190-E25) (optional)	
2-3	20 L plastic bucket for transporting composite between stations and catching wastewater during when washing the sam ple	
1 roll	Biodegradable flagging (different color from tape use to lay out the site)	
1	Velocity meter	
2 pr.	Forceps for rem oving invertebrates from nets	
1	1L 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 3-2)	
1	Littoral and Snag Sampling Form	

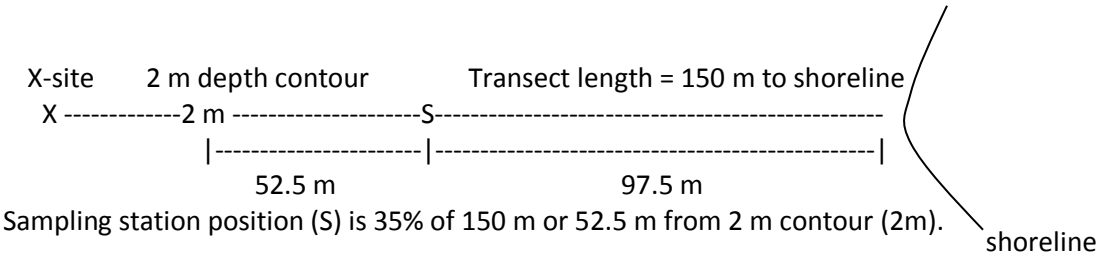
3.5 Aquatic Vegetation

The following is SMI sampling protocol as provided by Megan Moore, Minnesota DNR. Has not yet been modified to fit format of other sections in chapter.

1) **Office Verification**
   a) Once site assignments are obtained from EPA, evaluate safety and access concerns.
   b) Chose alternates (and evaluate) as necessary

2) **Field steps**
   a) Determine sampling site:
      i) Navigate to X-site coordinates designated by EPA.
      ii) Proceed towards sample bank along a line running perpendicular from X-site to sample bank until reaching 2.0 m depth.
      iii) If the 2 meter depth > than 10 meters from shore, record location on data form of the laptop computer. Determine "transect length" or distance from 2.0 m site to sample bank using GPS and/or rangefinder. If the 2.0 meter depth is within 10 meters of shore, sample at that point. This is done due to limitations in GPS accuracy.
      iv) Using "Station Position", determine the distance to sampling station along the transect. Enter the 2 meter depth coordinates into the GPS (either by entering the numbers or using the man overboard button). Navigate along the transect until the calculated distance is reached and anchor the boat. .
      v) Record sampling station coordinates on the data form.

Figure 1: Site Layout (Example – Station position=35%, Transect Length = 150 m)



   b) Vegetation Sampling:
      i) Sampling area is 2 m ring around the boat. Sampling follows Long Term Resource Monitoring Program procedures (Yin and Langrehr 2000) as detailed in items *b.ii-b.vii* below.
      ii) Search sampling area for any visual species and record on data form.
      iii) For non-rooted floating (NRF), rooted floating (RF), and emergent (EM) species, assign a cover of 1-5 based on the visual percent of the 2m ring occupied by each lifeform.
      iv) For non-rooted floating (NRF), rooted floating (RF), and emergent (EM) species, assign a cover of 1-5 based on the visual percent of the 2 m ring occupied by each individual species.
      v) Plants are collected from six sub-sampling areas - four off the corners of the boat and two off either side of middle of boat.
      vi) At each sub-sampling area:
         (1) Visually search for any species within the sub-sampling area and record
         (2) Extend rake out 1 m and lower to the sediment then drag along bottom back to the boat.

(3) Read and record the depth on the rake.
(4) Twist the rake 180 degrees and lift out of water.
(5) Record a 1 on the data form for any RF or EM species on the rake.
(6) Gently remove floating or emergent species while retaining submersed species and flip any submersed species touching the metal of the rake back onto the tines.
(7) Sweep rake through the water once to compact submersed vegetation
(8) Assign submersed vegetation a 1 to 5 based on amount on the rake and record on the data form.
(9) Identify individual species and assign a 1 to 5 based on amount on the rake and record on the data form.

vii) Repeat steps 1-9 above for each sub-sampling area.

c) Water Transparency Measurements (See Appendix 9 for complete SOP in Workplan):
   i) Determine water clarity with transparency tube. A transparency tube, also known as turbidity tube, is used to measure water clarity or provide estimates of turbidity and suspended particulate matter concentrations in water. The turbidity tube is a clear, graduated, plastic tube (4.5 cm diameter * 120 or 60 cm long) that has a Secchi disc image and drain tube at the bottom. A representative water sample is collected and poured into or drained from the tube slowly until the Secchi image disappears (filling) or appears (draining). The height of water above the disk at this is point is recorded. The process is repeated and the average of the two readings is recorded. Make sure that the sample is sufficiently mixed and that the reading is taken as quickly as possible in order to prevent sediment from accumulating on the Secchi disk image.

d) Water Velocity Measurements
   i) Determine water velocity per standard Long Term Resource Monitoring Protocols (Soballe and Fischer 2004) and record on data forms. Use a compass to determine and record the direction of the current.

3) Positioning the location of the Sampling Station relative to the X-site

a) As described in Figure 1, the sampling station is located in >0.2 m of water between the X-site and sample bank at a random distance from the 2.0 m contour. This situation assumes the sampling frame accurately depicts the secondary channels and the channel depths rise regularly from mid-channel to shore. Where these assumptions do not hold, the following rules should be followed to position the sampling station.

b) If the X-site is < 2.0 m deep AND the channel still qualifies as a secondary-channel, then the X-site becomes the sampling station. The rationale is that the design has successfully placed the station in the zone where vegetation may be present.

c) If the channel no longer qualifies as a secondary channel, the site is “non-target” and is replaced.

d) If the calculated sampling station is >2.0 m deep (e.g. the Station Position puts the presumed Station in a deeper channel), then the sampling station is moved to the closest point that is <2.0 m deep. The crew should first search along the line between the presumed sampling station and sample bank. If the boat ends up perpendicular to shore with the front end touching shore

and the back of the boat in greater than 2 meters of water, the crew should search along the transect from the presumed sampling station through the X-site and toward the opposite shore. If <2.0 m not found, search in channel area up to 100 m up and downstream of X-site. If <2.0 m depth not found, site is "non-target" because the channel does not have zone where vegetation is expected to be present. The unusual situation would only apply if the entire channel was >2.0 m deep and had such extremely steep bank angles as to preclude sampling. In most cases, if the 2.0 m contour is adjacent to the sample bank, then sample with the boat as close to shore as practical.

e) If the sampling station is <0.2 m deep, then move back towards X-site until reach >0.2 meters and sample there.

f) If there are hazards (or areas <0.2 m deep) between the X-site and sampling station, but the station is expected to be between 0.2 and 2.0 m deep, than navigate around hazards, return to the transect line, and proceed to sampling station. If station turns out to be <0.2 m or >2.0 m deep, refer to above rules.

4) If the site qualifies as "non-target", then an alternate site is sampled. A Site Data Form must be completed for the original site showing VERIFY FIELD is not "TSS", and SPECIES CODE = "NOSMPL". After all sampling is complete for the HUC, the alternate id of the replacement site is recorded in ALTERNATE ID

5) Non-target sites are in channels that are no longer connected to the main channel, a secondary channel, impounded areas, or Lake Pepin on either end OR is less than 30 meters wide OR is too shallow to navigate. All non-target sites must be replaced in list-order from the available alternates for the same unit. Alternate sites may only be used once in a given year (or once until the panel is complete).

3.6 Bacteriological Indicators

Fixed site and probabilistic network. *E. coli* is the only bacteriological indicator sampled under pilot. Any suggested text/procedure here? Definitely need to work on this section further.

Confirm that we want to do for probabilistic as well as fixed. How do we deal with the logistics of getting the sample to the laboratory within the hold time?

4. SUPPORTING MATERIALS

References

All reference/resource documents listed here.

Glossary/Acronyms

As needed.