Ogmcoal Dnr< ogmcoal@utah.gov>

Re: Mel Coonrad

1 message

Ingrid Campbell< ingridcampbell@utah.gov> Mon, Jun 3, 2013 at 10:28 AM
To: EIS Engineering <eisec@preciscom.net>
Cc: Mindi Lundberg <Mindilundberg@preciscom.net>, Ogmcoal Dnr <ogmcoal@utah.gov>

Mr. Coonrod-

First, let me apologize for misspelling your name. We may have gotten off on the wrong foot regarding this macroinvertebrate issue. I hope we can get beyond that and work together to get the information that we need to move forward.

We do want to ensure that the studies that are being done are providing the information needed to support the mine and your client. At your request I have copied this email to Mindi Lundberg so that she can be aware of the requirements. We look forward to working with her.

In order for the Division to determine the health of Crandall Creek, we need to have specific studies and surveys completed. I am including below a short discussion of the things that are expected.

Please see the attached files as an example of appropriate methodology to use for the macroinvertebrate survey required for Crandall Creek. This is the methodology used by the Forest Service and is recommended by the Division as a best technology currently available. Several issues must be included in this year's survey in order for us to accept the report for the mine's annual monitoring commitment. These include the following:

~The sampling site locations must be recorded and must be the same sampling sites used by JBR in previous studies. These sites were selected by myself, Joe Helfrich of UDOGM, and Pam Jewkes of the Manti La Sal National Forest.

~Samples must be sent to certified lab for analysis. This is a standard practice and considered by the Division as a Best Technology Currently Available and should also be done for the consistency with previous reports.

~The species should be identified down to the species level and a discussion about the tolerance characteristics of the species presence should be included. Family level is not adequate for determining the impact of mine water discharge on the macroinvertebrate community.

~typical macroinvertebrate indices such as the ones calculated in previous JBR reports should be reported.

~An analysis between this year's results and previous years should be completed as a discussion in the report.

A report that is not comparable to previous reports or is insufficient to address the current macroinvertebrate population in the stream and the impact from mine water discharge cannot be accepted. If you have any questions, please don't hesitate to call.

Ingrid Campbell
Environmental Scientist III
Coal Regulatory Program

Division of Oil Gas & Mining
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801-538-5318
Ingridcampbell@utah.gov

On Tue, May 21, 2013 at 10:29 AM, EIS Engineering <eisec@preciscom.net> wrote:

> Ms. Campbell,
>
> EIS implemented a riparian study of Crandall Creek. The study was done as outlined in the Crandall Canyon Mine "Scope of Work" and in cooperation with the Manti LaSal National Forest and UDWR. Comments from "JBR" either indicate their apparent lack of knowledge or their animosity towards EIS.
>
> It is important for you to understand the difference between a biotic community study (macro-invertebrates) and a riparian study. You may want to refer to "Fundamentals of Ecology" by Odum. In 1994 we were hired to evaluate the overall morphology of Crandall Creek; vegetation (obligate and facultative) soil primarily hydric, creek structure and a random sample of macro-invertebrates. In addition, we did a fishery inventory with UDWR.
>
> The only reason I am involved, is Jay Marshall is an old and dear associate and friend of mine. However, if you doubt my qualifications with only 30+ years of experience, a BS degree in Zoology, a Master's degree with post-graduate studies in Ecology and several years of teaching experience at major universities, I will gladly turn this project over to Mindi Lundberg, a professional Biologist, with our firm. Her resume' is attached.
>
> I would also like to know the qualifications of your staff who would review this report, at least as to what advanced degrees they hold and years of experience?
>
> Please refer all future communication and correspondence to Mindi Lundberg, EIS Environmental & Engineering Consulting, mindilundberg@preciscom.net.
>
> By the way, the correct spelling is "Coonrod".
>
> Thank you.
>
> Mel Coonrod
>
> President/Owner
>
> EIS Environmental & Engineering Consulting

From: Lqjulg#Fdp sehoc#^p dlbwr=lqjulgfdp sehoC xwdk1jry'#
Sent: P rqgd|/#P d|#53/#5346#5=3:#SP
To: hlvhfC suhflvfrp 1qhw
Subject: P hc#Frrqudg

Hi Mr. Coonrad- Could you please take a look at the attachment? This is a page from the first macroinvertebrate report done by JBR that questions your study done in 1994. I would like to speak with you over the phone about this to ensure that the study will be done by a professional qualified to do macroinvertebrate studies and will have a more thorough analysis of macroinvertebrates to the species level and resolve the other defiencies pointed out in the attachment. Please call me when you get a moment.

Thank you

Ingrid Campbell

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remacromethodology.zip
11392K

CHANNEL CONSTRAINT AND FIELD CHEMISTRY-WADEABLE STREAMS

Reviewed by (initial):_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ **DATE:___ ___/___ ___/ 2 0 ___ ___**

MULTI-PARAMEMTER UNIT MEASUREMENTS

	Comments
STREAM TEMP (°C): ___ ___.___	**RESIDUAL CHLORINE (optional):**
pH: ___ ___.___ ___	
SPECIFIC CONDUCTIVITY (µS/cm) ___ ___ ___ ___ ___ ___	
STREAM DO mg/L: ___ ___.___	
STREAM DO%: ___ ___ ___.___	
TIME OF DAY: ___ ___ ___ ___	

CHANNEL CONSTRAINT

CHANNEL PATTERN (Check one)

- ○ **One Channel**
- ○ **Anastomosing (complex) channel:** (Relatively long major and minor channels branching and rejoining)
- ○ **Braided channel:** (Multiple short channel branching and rejoining – mainly one channel broken up by numerous mid-channel bars)

CHANNEL CONSTRAINT (Check one)

- ○ **Channel very constrained in V-shaped valley** (i.e. it is very unlikely to spread out over valley or erode a new channel during flood)
- ○ **Channel is in Broad Valley** but channel movement by erosion during flood is **constrained by incision** (Flood flows do not commonly spread over valley floor or into multiple channels)
- ○ **Channel is in Narrow Valley but is not very constrained,** but limited in movement by relatively narrow valley floor
- ○ **Channel is Unconstrained in Broad Valley** (i.e. during flood it can fill off-channel areas and side channels, spread out over flood plain, or easily cut new channels by erosion)

CONSTRAINING FEATURE (Check all that apply)

- ○ **Bedrock/Sandstone** (i.e. channel is constrained by bare bedrock or sandstone)
- ○ **Hillslope** (i.e. channel constrained in narrow V-shaped valley)
- ○ **Terrace** (i.e. channel is constrained by its own incision into stream gravel/soil deposits)
- ○ **Human Bank Alterations** (i.e. constrained by rip-rap, landfill, dike, road, etc.)
- ○ **No constraining features**

PERCENT OF CHANNEL LENGTH WITH MARGIN IN CONTACT WITH EACH CONSTRAINING FEATURE (percentage fields should add up to 100%)**:**	**Constraining Feature 1** (primary)	**Constraining Feature 2** (if applicable)	**Constraining Feature 3** (if applicable)
	Type:_____	**Type:_____**	**Type:_____**
	Percentage: ___ ___ ___ %	**Percentage: ___ ___ ___ %**	**Percentage: ___ ___ ___ %**

Valley width (Visual Estimated Average): ___ ___ ___ ___(m)
Note: Be sure to include distances between both sides of valley border for valley width.

If you cannot see the valley borders, record the distance you can see and mark this circle ○

Comments:

TORRENT/FLASH FLOOD EVIDENCE ASSESSMENT FORM

Reviewed by (initial):_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ ___ **DATE:___ ___/___ ___/ 2 0 ___ ___**

TORRENT EVIDENCE

Please fill-in the circle if any of the following are evident while sampling.
Visit the following web-sites pre/post sampling to see if any recent major flow regimes occurred:

- **http://www.wrh.noaa.gov/slc/**
- **http://waterdata.usgs.gov/ut/nwis/current/?type=flow**

EVIDENCE OF TORRENT EVENTS/FLASH FLOODS:	
○	Stream channel has a devegetated corridor. This corridor lacks riparian vegetation due to a large flood event.
○	Vegetation along stream margin is depressed.
○	Riparian trees have fresh bark scars at many points along the stream at heights above bankfull level.
○	Riparian vegetation has fallen into the channel as a result of scouring near their roots.
○	Much of stream bed substrate is more loosely consolidated than expected (appears recently deposited).
○	There are locales along the reach where gravel or cobble deposits can be seen above bankfull level from a large flood event.
○	Isolated puddles of water are seen above bankfull level.
○	Deposits of debris (sticks, trash, gravel, vegetation, etc.) can be found above bankfull level.
○	Water appears more turbid than typically expected (considering the eco-region).
○	Overall abundance of benthos sample appears lower than otherwise expected, considering the eco-region, due to recent flood event.
○	Soil above bankfull height is damp.
○	Significant amounts of large woody debris within the stream or riparian corridor appear to have recently been moved.
MAGNITUDE/DESCRIPTION OF EVENT:	
○	Evidence of flooding present, but magnitude is common for this system on a yearly basis (i.e. desert streams after a monsoonal summer storm).
○	Evidence of a large flood present (10-20 year flood), but did not make massive changes to the system.
○	Evidence shows a massive event where signs like channel blow-outs, devegetation, and major changes to the stream are present (i.e. ≥100 year flood).
OTHER:	
○	Other evidence not covered in options above (explain observations in comments section)
NO EVIDENCE:	
○	No evidence of torrent or flash flood events.

COMMENTS
(if evidence of flooding is present, please explain when you think the event occurred, whether it be within the month, longer than a month, within the year, more than two years, etc.)

Reviewed by (initial): _____
Updated: 03/2011

DWQ Electro-shocking/Fish Tissue Collection Field Sheet

Please use a new sheet for each site (do not combine multiple sites on one sheet)

Site Name:					
STORET # (not applicable for DWR personnel):					
County:					
Date:					
GPS Coordinates		Degrees	Minutes	Seconds	Other (decimal degrees/UTM)
Datum:	Latitude				
	Longitude				
Reach Length (m):					
Shocker Settings					
Shocking Time (s):					
Volts:					
Pulse Rate (Hz):					
Pulse Width (ms):					

1. Tally final counts for each species and their appropriate sizes in the circles. 2. Species codes on backside of this sheet	**ELECTRO-SHOCKING DATA**				
	Size 1: 0-60mm (0-2.36 in)	**Size 2: 61-200mm** (2.40-7.87 in)	**Size 3: 201-300mm** (7.91-11.81 in)	**Size 4: 301-400mm** (11.85-15.74 in)	**Size 5: >401mm** (>15.78 in)
Species code:	○	○	○	○	○
Species code:	○	○	○	○	○
Species code:	○	○	○	○	○
Species code:	○	○	○	○	○
Species code:	○	○	○	○	○
Species code:	○	○	○	○	○
Species code:	○	○	○	○	○
Species code:	○	○	○	○	○

Comments:	Number of Netters:

Reviewed by (initial): _____
Updated: 03/2011

Fish Tissue Data (Hg)

Sample ID*	Length (mm)	Weight (g)	Comments
01			
02			
03			
04			
05			
06			
07			
08			
09			
10			
11			
12			
13			
14			
15			
16			
17			
18			
19			
20			
21			If there are more than 21 samples, use another sheet and staple all the sheets together.

*Sample ID=STORET-Fish Code (listed below)-Unique Sequence ID per Site (i.e. 4982100CTT01)

Collector(s) Names (for Hg collection only):

_____ _____ _____

_____ _____

Species Codes:

Black bullhead (BBH)	**Least chub (LEC)**	**Utah sucker (UTS)**
Black crappie (BLC)	**Leatherside chub (LSC)**	**Virgin spinedace (VSD)**
Bluehead sucker (BHS)	**Longnose dace (LND)**	**Walleye (WLE)**
Bluegill (BLG)	**Mountain whitefish (MWF)**	**Wiper (WIP)**
Bonneville whitefish (BWF)	**Mountain sucker (MTS)**	**White bass (WHB)**
Bonneville cisco (BCI)	**Mottled sculpin (MOS)**	**Woundfin (WFN)**
Brook trout (BKT)	**Paiute sculpin (PTS)**	**Yellow perch (YLP)**
Brown trout (BRT)	**Rainbow trout (RBT)**	
Channel catfish (CCF)	**Redside shiner (RSS)**	
Common carp (CMC)	**Red-shiner (RES)**	**Note: Not all spp found in Utah are listed here. If crew collects a spp that is not listed then hand write the spp name on the front side of the sheet under "Species code"**
Cutthroat trout (CTT)	**Roundtail chub (RTC)**	
Desert sucker (DSS)	**Smallmouth bass (SMB)**	
Flannelmouth sucker (FLS)	**Speckled dace (SPD)**	
Green sunfish (GSF)	**Splake trout (SPT)**	
Kokanee (KOK)	**Striped bass (STB)**	
Lake trout (LKT)	**Tiger muskie (TGM)**	
Largemouth bass (LMB)	**Tiger trout (TGT)**	

PHab: CHANNEL/RIPARIAN CROSS-SECTION FORM - STREAMS

Reviewed by (Initials): _____

STORET / SITE ID: _____ DATE: ____ / ____ / 2 0 ____ TRANSECT: ○A ○B ○C ○D ○E ○F ○G ○H ○I ○J ○K X-tra Side Channel ○

SUBSTRATE CROSS-SECTIONAL INFORMATION

	Dist LB XX.XX m	Depth XXX cm	Size Class Code	Embed. 0-100%	Flag
Left					
LCtr					
Ctr					
RCtr					
Right					

SUBSTRATE SIZE CLASS CODES	Embed. (%)
RS = Bedrock (Smooth) - (Larger than a car)	0
RR = Bedrock (Rough) - (Larger than a car)	0
RC = Concrete/Asphalt	
XB = Large Boulder (1000 to 4000 mm) - (Meterstick to car)	
SB = Small Boulder (250 to 1000 mm) - (Basketball to meterstick)	
CB = Cobble (64 to 250 mm) - (Tennis ball to Basketball)	
GC = Coarse Gravel (16 to 64 mm) - (Marble to Tennis ball)	
GF = Fine Gravel (2 to 16 mm) - (Ladybug to marble)	
SA = Sand (0.06 to 2 mm) - (Gritty - up to Ladybug size)	100
FN = Silt / Clay / Muck - (Not Gritty)	100
HP = Hardpan - (Firm, Consolidated Fine Substrate)	0
WD = Wood - (Any Size)	
OT = Other (Write comment below)	

FISH COVER/ OTHER

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

	Cover in Channel	Flag
Filamentous Algae	0 1 2 3 4	
Macrophytes	0 1 2 3 4	
Woody Debris >0.3 m (BIG)	0 1 2 3 4	
Brush/Woody Debris <0.3 m (SMALL)	0 1 2 3 4	
Live Trees or Roots	0 1 2 3 4	
Overhanging Veg. =<1 m of Surface	0 1 2 3 4	
Undercut Banks	0 1 2 3 4	
Boulders	0 1 2 3 4	
Artificial Structures	0 1 2 3 4	

Filamentous Algae Presence		%	Flag
Present coverage	0 1 2 3 4		
Potential for colonization	0 1 2 3 4		

VISUAL RIPARIAN ESTIMATES

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

D = Deciduous
C = Coniferous
E = Broadleaf Evergreen
M = Mixed
N = None

RIPARIAN VEGETATION COVER	Left Bank	Right Bank	Flag
Canopy (>5 m high)			
Vegetation Type	D C E M N	D C E M N	
BIG Trees (Trunk >0.3 m DBH)	0 1 2 3 4	0 1 2 3 4	
SMALL Trees (Trunk <0.3 m DBH)	0 1 2 3 4	0 1 2 3 4	
Understory (0.5 to 5 m high)			
Vegetation Type	D C E M N	D C E M N	
Woody Shrubs & Saplings	0 1 2 3 4	0 1 2 3 4	
Non-Woody Herbs, Grasses, & Forbs	0 1 2 3 4	0 1 2 3 4	
Ground Cover (<0.5 m high)			
Woody Shrubs & Saplings	0 1 2 3 4	0 1 2 3 4	
Non-Woody Herbs, Grasses and Forbs	0 1 2 3 4	0 1 2 3 4	
Barren, Bare Dirt or Duff	0 1 2 3 4	0 1 2 3 4	

HUMAN INFLUENCE	0 = Not Present P = >10 m C = Within 10 m B = On Bank		
	Left Bank	Right Bank	Flag
Wall/Dike/Revetment /Riprap/Dam	0 P C B	0 P C B	
Buildings	0 P C B	0 P C B	
Pavement/Cleared Lot	0 P C B	0 P C B	
Road/Railroad	0 P C B	0 P C B	
Pipes (Inlet/Outlet)	0 P C B	0 P C B	
Landfill/Trash	0 P C B	0 P C B	
Park/Lawn	0 P C B	0 P C B	
Row Crops	0 P C B	0 P C B	
Pasture/Range/Hay Field	0 P C B	0 P C B	
Logging Operations	0 P C B	0 P C B	
Mining Activity	0 P C B	0 P C B	

BANK MEASUREMENTS

	Bank Angle 0 - 360	Undercut Dist. (m)	Flag
Left			
Right			
Wetted Width XXX.X m			
Bar Width XX.X m			
Bankfull Width XXX.X m			
Bankfull Height XX.X m			
Incised Height XX.X m			

CANOPY COVER MEASUREMENTS

DENSIOMETER (0-17Max)

		Flag			Flag
CenUp			CenR		
CenL			Left		
CenDwn			Right		

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

Flag	Comments

PHAB: THALWEG PROFILE & WOODY DEBRIS FORM STREAMS

Reviewed by (initial):_____

STORET / SITE ID: _____ DATE: ___/___/20___ TRANSECT: ○A-B ○B-C ○C-D ○D-E ○E-F ○F-G ○G-H ○H-I ○I-J ○J-K

THALWEG PROFILE

For Transect A-B ONLY: Increment (m) X.X: _____ Total Reach Length (m): _____

STATION	THALWEG DEPTH (cm) (XXX)	WETTED WIDTH (m) (XXX.X)	BAR WIDTH[1] Present	BAR WIDTH[1] XX.X	SOFT /SMALL SEDIMENT	CHANNEL UNIT CODE	POOL FORM CODE	SIDE CHANNEL	BACK WATER	FLAG	COMMENTS
0			Y N		Y N			Y N	Y N		
1			Y N		Y N			Y N	Y N		
2			Y N		Y N			Y N	Y N		
3			Y N		Y N			Y N	Y N		
4			Y N		Y N			Y N	Y N		
*5			Y N		Y N			Y N	Y N		
6			Y N		Y N			Y N	Y N		
*7			Y N		Y N			Y N	Y N		
8			Y N		Y N			Y N	Y N		
9			Y N		Y N			Y N	Y N		
10			Y N		Y N			Y N	Y N		
11			Y N		Y N			Y N	Y N		
12			Y N		Y N			Y N	Y N		
13			Y N		Y N			Y N	Y N		
14			Y N		Y N			Y N	Y N		

SUBSTRATE

Station (5 or 7)	LFT	LCTR	CTR	RCTR	RGT	FLAG
*						

FLAG	COMMENTS (for SUBSTRATE and LWD)

LARGE WOODY DEBRIS

(≥10 cm small end diameter; ≥ 1.5 m length)

CHECK IF UNMARKED BOXES ARE ZERO ☐ FLAG _____

DIAMETER LARGE END	PIECES ALL/PART IN BANKFULL CHANNEL Length 1.5-5m	5-15m	>15m	PIECES BRIDGE ABOVE BANKFULL CHANNEL Length 1.5-5m	5-15m	>15m
0.1-<0.3 m						
0.3-0.6 m						
0.6-0.8 m						
>0.8 m						

SUBSTRATE SIZE CLASS CODES

RS = BEDROCK (SMOOTH) - (LARGER THAN A CAR)
RR = BEDROCK (ROUGH) - (LARGER THAN A CAR)
RC = CONCRETE/ASPHALT
XB = LG. BOULDER (1000 TO 4000 mm) - METERSTICK TO CAR)
SB = SM. BOULDER (260 TO 1000 mm) - BASKETBALL TO METERSTICK)
CB = COBBLE (64 TO 260 mm) - (TENNIS BALL TO BASKETBALL)
GC = COARSE GRAVEL (16 TO 64 mm) - (MARBLE TO TENNIS BALL)
GF = FINE GRAVEL (2 TO 16 mm) - (LADYBUG TO MARBLE)
SA = SAND (0.06 TO 2 mm) - (GRITTY - UP TO LADYBUG SIZE)
FN = SILT/ CLAY / MUCK - (NOT GRITTY)
HP = HARDPAN - (FIRM, CONSOLIDATED FINE SUBSTRATE)
WD = WOOD - (ANY SIZE)
OT = OTHER (COMMENT ON OTHER SIDE)

POOL FORM CODES

N = Not a pool
W = Large Woody Debris
R = Rootwad
B = Boulder or Bedrock
F = Unknown, fluvial

COMBINATIONS:
eg. WR, BR, WRB

CHANNEL UNIT CODES

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

Flag Codes: K = no measurement made; U = suspect measurement; F1, F2, ect. = flags assigned by each field crew; G1, G2, etc. for flags not specific to one station. Explain all flags in comments. 1 = Measure Bar Width at Station 0 and Mid-Station (5 or 7).

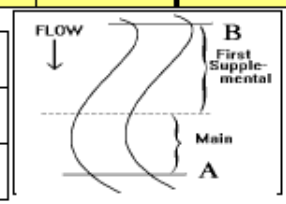
PHab: SLOPE AND BEARING FORM - STREAMS

Reviewed by (initial): _____

STORET / SITE ID: _____ **DATE:** ___ / ___ / 2 0 ___

TRANSECT & METHOD Mark method for every Transect	MAIN (always used) Slope(%) or Elev. Diff. (cm) Mark Units for every Transect	BEARING 0 - 359	PROPOR-TION %	FIRST SUPPLEMENTAL Slope(%) or Elev. Diff. (cm)	BEARING 0 - 359	PROPOR-TION %	SECOND SUPPLEMENTAL Slope(%) or Elev. Diff. (cm)	BEARING 0 - 359	PROPOR-TION %	FLAG
A < B ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
B < C ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
C < D ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
D < E ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
E < F ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
F < G ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
G < H ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
H < I ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
I < J ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
J < K ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									

FLAG	COMMENT



Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, M (M = Method - used for method comment only) = flag assigned by field crew. Explain all flags in comment sections

 CL=Clinometer; HL=Hand Level; LA=Laser rangefinder with electronic clinometer; TR=Transit, surveyors level or total station; WT=Water Tubing.

UCASE Reference Site/Site Rating Field Evaluation Form

Reviewed by (initials):_____
Updated: 04/2011

Stream/Site Name:_____

STORET:_____ **Date:**_____ **Crew Members:**_____

UCASE ID:_____**Lat/UTM:**_____**Long/UTM:**_____

Best Professional Judgement Grades: After answering all other site evaluations, please grade site on the relative reference quality of the site on a WATERSHED scale and a SITE SPECIFIC scale and provide supporting comments if desired:

WATERSHED SCALE (overall grade of watershed upstream of site; Macro-assessment) circle one:

A) Ideal - wilderness area or virtually no human disturbance upstream of site.

B) Good - minimal disturbances upstream of site. Some activities present upstream in entire watershed but impacts are minimal.

C) Marginal - human disturbances present above site but stream still shows signs of integrity.

D) Sub-marginal - Considerable human disturbances above site in entire watershed; impairment from activities is obvious.

E) Poor - Watershed is heavily manipulated where activities are highly impactful to site; minimal signs of integrity present.

F) Very Poor - Extensive human disturbances in watershed. Integrity of stream is minimal or absent due to activities.

SITE SCALE (grade of site on a reach-wide scale; Micro-assessment) circle one:

A) Ideal - wilderness area and virtually no human disturbance at reach.

B) Good - minimal disturbance at or near site. Some human-based activities present near or at reach but impacts are minimal.

C) Marginal - human activities present at site while sampling. Signs of impacts are present, but site still functions properly.

D) Sub-marginal - Considerable human disturbance near or at site. Activities are having direct impact to site, integrity of site is reduced.

E) Poor - Site is heavily manipulated or impacted from human activities; minimal signs of integrity present.

F) Very Poor - Extensive human disturbances at or near site. Integrity is very minimal or absent.

Comments on grades:

Description of Habitat Features at Site: Accounting for natural factors related to climate, geology, stream size, stream confinement, eco-region, etc., please grade the site 1 through 5 after sampling whether the site has the following features (1=feature indicative of poor habitat….5=feature indicative of ideal habitat/reference conditions). If 1 or 2 is given, comment on why feature does not meet your expectations for this type of stream. Base ratings on ECO-REGION (don't compare a mountain stream to a desert stream). If any categories are undetermined by crew, put a **?** in the grade box. If description does not apply, put an NA in the grade box.

Grades (1-5)	***Description of Habitat Features***	***Comments***
	Riparian vegetation and structure (if non-natives present do not score a 5).	
	Historic riparian vegetation (e.g. legacy trees).	
	Bank conditions (if rip-rap, vegetation removal, car bodies present, etc. do not score a 5)	
	Availability of in-channel habitat (look for a mixture of substrates, undercut banks, overhanging veg, mixture of channel units, etc.)	
	Channel morphology (if channel is been re-routed or manipulated by humans, do not score a 5).	
	Natural flow regime (score accordingly if site is affected by upstream diversions/manipulation structures).	
	Sedimentation and turbidity for stream-type (if stream is naturally turbid, put NA).	
	Influence of human caused sheens; odors; or scum	
	Other:	

Potential Stressor Identification Checklist. Indicate the relative effects (i.e., alterations to the chemical, physical, or biological components of the stream ecosystem) of all activities that are present **within the stream channel or riparian corridor** at both the reach or watershed scale (include all tributaries in watershed no matter the size). If YES, describe and indicate intensity as follows:

Reach: **1** = historical activity most likely in vicinity of site but does not appear to have current effects; **2** = activity present but appears to have minimal current effects; **3** = activity present at or near site where impacts are evident but site is not being entirely degraded; **4=** activity present at or near site and has a significant impact to site but some integrity still exists**; 5** = activity present on bank or within the stream channel creating obvious impairments to site.

Watershed: **1** = historical activity most likely above site but no signs of currents effects occur; **2** = activity present above the reach in a few limited locales and effects are minimal; **3** = activity is present upstream of the reach where impacts are evident but not creating a major impairment; **4 =** activity is present upstream and has a moderate impact to site but some integrity still exists **5** = extensive signs of activity throughout the watershed resulting in major impacts.
Mark a **'?'** in the NO column, if after all feasible field observations have been made you cannot confidently tell whether a stressor is present.

	Reach		***Watershed***		**Comments**
	Yes	**No**	**Yes**	**No**	
Point discharge present?	1 2 3 4 5		1 2 3 4 5		
Hazardous waste sites, landfills?	1 2 3 4 5		1 2 3 4 5		
Mines or oil fields?	1 2 3 4 5		1 2 3 4 5		
Feed lots, poultry farms, or hatcheries?	1 2 3 4 5		1 2 3 4 5		
Urban, industrial, commercial, or residential land use?	1 2 3 4 5		1 2 3 4 5		
Channelization?	1 2 3 4 5		1 2 3 4 5		
Dams or impoundments (not beaver)[1]?	1 2 3 4 5		1 2 3 4 5		
Diversions (i.e., canal head gates)[2]?	1 2 3 4 5		1 2 3 4 5		
Logged or burned forests?	1 2 3 4 5		1 2 3 4 5		
Grazed lands[3]?	1 2 3 4 5		1 2 3 4 5		
Cropped lands?	1 2 3 4 5		1 2 3 4 5		
Non-Motorized Recreation use?	1 2 3 4 5		1 2 3 4 5		
Roads/Motorized Trails?	1 2 3 4 5		1 2 3 4 5		

[1]Score watershed a 2 if there is an impoundment small enough to not largely change natural flow or temperature regimes; [2]Score watershed a 2 if no evidence of daily fluctuations in flow, 3 if water withdrawals are minimal, 4 if water withdrawals are <1/3 flow and large fluctuations are infrequent, 5 if withdrawals >1/3 flow or highly variable; [3]Score reach a 2 if some evidence exists but vegetation looks intact **and** there is no evidence of stream habitat damage (i.e., collapsed banks).

Flow Regime at Site (circle one): Perennial Intermittent Ephemeral

RIPARIAN "LEGACY" TREES AND INVASIVE ALIEN PLANTS

Reviewed by (initial): _____

STORET / SITE ID: _____ DATE: ___/___/20___

Tran	LARGEST POTENTIAL LEGACY TREE VISIBLE FROM THIS STATION						ALIEN PLANT SPECIES PRESENT IN LEFT AND RIGHT RIPARIAN PLOTS	
	Trees not Visible	DBH (m)	Height (m)	Dist. from wetted margin (m)	Type	Taxonomic Category	Check all that are present	
A	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☐ 15-30 ☐ >30	_____	☐ Deciduous ☐ Coniferous ☐ Broadleaf Evergreen	_____	☐ NONE	☐ RC Grass ☐ Salt Ced ☐ Hblack ☐ G Reed ☐ Engl Ivy ☐ CanThis ☐ Teasel ☐ C Burd ☐ Ch Grass ☐ M This ☐ Spurge ☐ Rus Ol
B	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☐ 15-30 ☐ >30	_____	☐ Deciduous ☐ Coniferous ☐ Broadleaf Evergreen	_____	☐ NONE	☐ RC Grass ☐ Salt Ced ☐ Hblack ☐ G Reed ☐ Engl Ivy ☐ CanThis ☐ Teasel ☐ C Burd ☐ Ch Grass ☐ M This ☐ Spurge ☐ Rus Ol
C	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☐ 15-30 ☐ >30	_____	☐ Deciduous ☐ Coniferous ☐ Broadleaf Evergreen	_____	☐ NONE	☐ RC Grass ☐ Salt Ced ☐ Hblack ☐ G Reed ☐ Engl Ivy ☐ CanThis ☐ Teasel ☐ C Burd ☐ Ch Grass ☐ M This ☐ Spurge ☐ Rus Ol

INSTRUCTIONS

Potential Legacy trees are defined as the largest tree within your search area, which is as far as you can see, but within maximum limits as follows:

Wadeable Streams: Confine search to no more than 50 m from left and right bank and extending upstream to next transect (for 'K' look upstream 4 channel widths)

Non-wadeable Rivers: Confine search to no more than 100 m from left and right bank and extending both upstream and downstream as far as you can see confidently.

Alien Plants: Confine search to riparian plots on left and right bank

Wadeable Streams: 10 m x 10 m

Non-wadeable Rivers: 10 m x 20 m

Not all aliens are to be identified in all states. See Field Manual and Plant Identification Guide.

TAXONOMIC CATEGORIES

Acacia/Mesquite
Alder/Birch
Ash
Maple/Boxelder
Oak
Poplar/Cottonwood
Sycamore
Willow
Unknown or Other Deciduous

Cedar/Cypress/Sequoia
Fir (including Douglas fir and hemlock)
Juniper
Pine
Spruce
Unknown or Other Conifer

Unknown or Other Broadleaf Evergreen

Snag (Dead tree of any species)

ALIEN SPECIES

RC Grass	Reed canarygrass	*Phalaris arundinacea*
Engl Ivy	English ivy	*Hedera helix*
ChGrass	Cheat grass	*Bromus tectorum*
Salt Ced	Salt Cedar	*Tamarix spp.*
Can This	Canada thistle	*Cirsium arvense*
M This	Musk thistle	*Carduus nutans*
Hblack	Himalayan blackberry	*Rubus discolor*
Teasel	Teasel	*Dipsacus fullonum*
Spurge	Leafy spurge	*Euphorbia esula*
G Reed	Giant reed	*Arundo donax*
C Burd	Common burdock	*Arctim minus*
Rus Ol	Russian-olive	*Elaeagnus angustifolia*

COMMENTS

Transects D to K continued on other side

RIPARIAN "LEGACY" TREES AND INVASIVE ALIEN PLANTS

Reviewed by (initial): _____

STORET / SITE ID: _____ DATE: ___ / ___ / 20 ___

Tran	LARGEST POTENTIAL LEGACY TREE VISIBLE FROM THIS STATION						ALIEN PLANT SPECIES PRESENT IN LEFT AND RIGHT RIPARIAN PLOTS				
	Trees not Visible	DBH (m)	Height (m)	Dist. from wetted margin (m)	Type	Taxonomic Category	Check all that are present				
D	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☐ 15-30 ☐ >30	_____	☐ Deciduous ☐ Coniferous ☐ Broadleaf Evergreen	_____	☐ NONE	☐ RC Grass ☐ Engl Ivy ☐ Ch Grass	☐ Salt Ced ☐ CanThis ☐ M This	☐ Hblack ☐ Teasel ☐ Spurge	☐ G Reed ☐ C Burd ☐ Rus Ol
E	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☐ 15-30 ☐ >30	_____	☐ Deciduous ☐ Coniferous ☐ Broadleaf Evergreen	_____	☐ NONE	☐ RC Grass ☐ Engl Ivy ☐ Ch Grass	☐ Salt Ced ☐ CanThis ☐ M This	☐ Hblack ☐ Teasel ☐ Spurge	☐ G Reed ☐ C Burd ☐ Rus Ol
F	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☐ 15-30 ☐ >30	_____	☐ Deciduous ☐ Coniferous ☐ Broadleaf Evergreen	_____	☐ NONE	☐ RC Grass ☐ Engl Ivy ☐ Ch Grass	☐ Salt Ced ☐ CanThis ☐ M This	☐ Hblack ☐ Teasel ☐ Spurge	☐ G Reed ☐ C Burd ☐ Rus Ol
G	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☐ 15-30 ☐ >30	_____	☐ Deciduous ☐ Coniferous ☐ Broadleaf Evergreen	_____	☐ NONE	☐ RC Grass ☐ Engl Ivy ☐ Ch Grass	☐ Salt Ced ☐ CanThis ☐ M This	☐ Hblack ☐ Teasel ☐ Spurge	☐ G Reed ☐ C Burd ☐ Rus Ol
H	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☐ 15-30 ☐ >30	_____	☐ Deciduous ☐ Coniferous ☐ Broadleaf Evergreen	_____	☐ NONE	☐ RC Grass ☐ Engl Ivy ☐ Ch Grass	☐ Salt Ced ☐ CanThis ☐ M This	☐ Hblack ☐ Teasel ☐ Spurge	☐ G Reed ☐ C Burd ☐ Rus Ol
I	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☐ 15-30 ☐ >30	_____	☐ Deciduous ☐ Coniferous ☐ Broadleaf Evergreen	_____	☐ NONE	☐ RC Grass ☐ Engl Ivy ☐ Ch Grass	☐ Salt Ced ☐ CanThis ☐ M This	☐ Hblack ☐ Teasel ☐ Spurge	☐ G Reed ☐ C Burd ☐ Rus Ol
J	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☐ 15-30 ☐ >30	_____	☐ Deciduous ☐ Coniferous ☐ Broadleaf Evergreen	_____	☐ NONE	☐ RC Grass ☐ Engl Ivy ☐ Ch Grass	☐ Salt Ced ☐ CanThis ☐ M This	☐ Hblack ☐ Teasel ☐ Spurge	☐ G Reed ☐ C Burd ☐ Rus Ol
K	☐	☐ 0-0.1 ☐ .75-2 ☐ .1-.3 ☐ >2 ☐ .3-.75	☐ <5 ☐ 5-15 ☐ 15-30 ☐ >30	_____	☐ Deciduous ☐ Coniferous ☐ Broadleaf Evergreen	_____	☐ NONE	☐ RC Grass ☐ Engl Ivy ☐ Ch Grass	☐ Salt Ced ☐ CanThis ☐ M This	☐ Hblack ☐ Teasel ☐ Spurge	☐ G Reed ☐ C Burd ☐ Rus Ol

SAMPLE COLLECTION FORM-WADEABLE STREAMS

Reviewed by (initial):_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ **DATE:___ ___/___ ___/ 2 0 ___ ___**

WATER CHEMISTRY

Sample Bottle	Sample Collected?	Sample Bottle	Sample Collected?	Process: See protocol Preservation: Place on ice. Lab: State of Utah Health Lab within two weeks of collection. Shop storage: Fridge	Comments/Flags
Total Chemistry	Y / N	Filtered Nutrient	Y / N		
Non-Filtered Nutrient	Y / N	Filtered Metals	Y / N		

TARGETED BENTHOS SAMPLE

No. of Jars (Primary)	Was a Replicate Sample Taken (if Yes, record no. of jars?	Collection Method (Choose one)	Comments/Flags
___ ___	Y / N ___ ___	○D-net ○Modified surber ○Other (indicate in comments)	

TRANSECT:		_____		_____		_____		_____	
Dom. Substrate	**Channel**	**Sub.**	**Chan.**	**Sub.**	**Chan.**	**Sub.**	**Chan.**	**Sub.**	**Chan.**
Fine/Sand	**Pool**	○**F**	○**P**	○**F**	○**P**	○**F**	○**P**	○**F**	○**P**
Gravel	**Glide**	○**G**	○**GL**	○**G**	○**GL**	○**G**	○**GL**	○**G**	○**GL**
Coarse	**Riffle**	○**C**	○**RI**	○**C**	○**RI**	○**C**	○**RI**	○**C**	○**RI**
Other: Note in Comments	**Rapid**	○**O**	○**RA**	○**O**	○**RA**	○**O**	○**RA**	○**O**	○**RA**
SUBSTRATE SIZE CLASSES F/S – ladybug or smaller (<2 mm) G – ladybug to tennis ball (2 to 64 mm) C – tennis ball to car sized (64 to 4000 mm) O – bedrock, hardpan, wood, vegetation, leaf litter, undercut, macrophyhtes, etc.		_____		_____		_____		_____	
		Sub.	**Chan.**	**Sub.**	**Chan.**	**Sub.**	**Chan.**	**Sub.**	**Chan.**
		○**F**	○**P**	○**F**	○**P**	○**F**	○**P**	○**F**	○**P**
		○**G**	○**GL**	○**G**	○**GL**	○**G**	○**GL**	○**G**	○**GL**
		○**C**	○**RI**	○**C**	○**RI**	○**C**	○**RI**	○**C**	○**RI**
		○**O**	○**RA**	○**O**	○**RA**	○**O**	○**RA**	○**O**	○**RA**

Note:

- Always perform 8 kicks at every site.

- Target riffle habitat primarily. If riffles are scarce or absent (e.g. low gradient, beaver ponds, etc.), target edge habitats (e.g. overhanging veg., undercut banks, etc.) and mark "O" in the substrate column and explain situation in comments.

- If riffles are present, but scarce, multiple kicks can be performed at the same riffles throughout the reach.

- If kicks are made in beaver influenced areas, please explain in the comments section.

Process: Fill a clean wide-mouth bottle with ~30-40% of composite sample (use multiple bottles if necessary).
Preservation: Fill rest of bottle with denatured alcohol (EtOH) and seal lid with electrical tape. Keep secure in upright position.
Lab: Utah State University Bug Lab; submit in large batch at end of field season.
Shop storage: Store on shelf with other bug samples.

BENTHOS COMMENTS:

PERIPHYTON COLLECTION FORM
STREAM DISCHARGE FORM-WADEABLE STREAMS

Reviewed by (initial):_______

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ **DATE:___ ___/___ ___/ _2_ _0_ ___ ___**

COMPOSITE PERIPHYTON SAMPLE

Composite Volume (mL) (**Quantitative**)	Number of transects sampled (0-11): ___ ___	**Qualitative** Collection	
___ ___ ___		Was a **qualitative** sample collected at this site: Y / N If yes, indicate # of vials submitted to the Rushforth Lab: ___ ___	
Species (Assemblage) ID	**Chlorophyll**	**Ash-Free Dry Mass (Biomass)**	**Comments/Flags**
Process: Fill vial with **50**mL of water from composite sample. **Preservation:** Seal cap with electrical-tape, place sample in a zip-top baggie; keep cool on ice. **Lab:** Rushforth Phycology within 2 weeks of collection. **Shop storage:** Fridge	**Process:** Filter **25** mL from composite sample using a pre-filter. **Preservation:** Fold filter in half and place in a lightless canister (e.g. film canister) and keep frozen (dry ice) in a zip-top baggie. **Lab:** State of Utah Health Lab within two weeks of collection. **Shop storage:** Freezer	**Process:** Filter **25** mL from composite sample using a pre-filter. **Preservation:** Fold filter in half and place in a lightless canister (e.g. film canister) and keep frozen (dry ice) in a zip-top baggie. **Lab:** State of Utah Health Lab within two weeks of collection. **Shop storage:** Freezer	Please indicate if any aquatic vegetation samples were collected for ID at Utah State University Herbarium
Indicate volume if not 50 mL ___ ___ **FLAG:** ________	Indicate volume filtered if not 25 mL ___ ___ **FLAG:** ________	Indicate volume filtered if not 25 mL ___ ___ **FLAG:** ________	

○ FLOW TRACKER

Discharge is determined directly in field, record value here:

Q=______________________ ○**cfs** ○**m³/s** **FLAG:** ________

○ USGS Gage

USGS gage is near site and flow was looked up online:

________________________ **cfs** **FLAG:** ________

○ VELOCITY AREA

Distance Units ○ft ○cm	**Depth Units** ○ft ○cm	**Velocity Units** ○ft/s XX.X ○m/s X.XX	**FLAG**
________	________	________	_____
________	________	________	_____
________	________	________	_____
________	________	________	_____
________	________	________	_____
________	________	________	_____
________	________	________	_____
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________	________	________	_____
________	________	________	_____

○ NEUTRAL BOUYANT OBJECT

	FLOAT 1	**FLOAT 2**	**FLOAT 3**
Float Dist. ○ft ○m	___ ___	___ ___	___ ___
Float Time (sec)	___ ___ ___	___ ___ ___	___ ___ ___
FLAG	___ ___ ___	___ ___ ___	___ ___ ___

Cross Sections on Float Reach

	Upper Section	**Middle Section**	**Lower Section**
Width ○ft ○m	___ ___.___	___ ___.___	___ ___.___
Depth 1 ○ft ○m	___ ___ ___	___ ___ ___	___ ___ ___
Depth 2	___ ___ ___	___ ___ ___	___ ___ ___
Depth 3	___ ___ ___	___ ___ ___	___ ___ ___
Depth 4	___ ___ ___	___ ___ ___	___ ___ ___
Depth 5	___ ___ ___	___ ___ ___	___ ___ ___

COMMENTS/FLAGS

If flow is estimated, please indicate value here:

STREAM ASSESSMENT FORM - STREAMS/RIVERS

Reviewed by (initial): _____

STORET/
SITE ID: _____ DATE: ___ / ___ / 2 0 ___

WATERSHED ACTIVITIES AND DISTURBANCES OBSERVED (Intensity: Blank=Not observed, L=Low, M=Moderate, H=Heavy)

Residential	Recreational	Agricultural	Industrial	Stream Management
L M H Residences	L M H Hiking Trails	L M H Cropland	L M H Industrial Plants	L M H Liming
L M H Maintained Lawns	L M H Parks, Campgrounds	L M H Pasture	L M H Mines/Quarries	L M H Chemical Treatment
L M H Construction	L M H Primitive Parks, Camping	L M H Livestock Use	L M H Oil/Gas Wells	L M H Angling Pressure
L M H Pipes, Drains	L M H Trash/Litter	L M H Orchards	L M H Power Plants	L M H Dredging
L M H Dumping	L M H Surface Films	L M H Poultry	L M H Logging	L M H Channelization
L M H Roads		L M H Irrigation Equip.	L M H Evidence of Fire	L M H Water Level Fluctuations
L M H Bridge/Culverts		L M H Water Withdrawal	L M H Odors	L M H Fish Stocking
L M H Sewage Treatment			L M H Commercial	L M H Dams

SITE CHARACTERISTICS (200 m radius)

Waterbody Character

Pristine ☐ 5 ☐ 4 ☐ 3 ☐ 2 ☐ 1 Highly Disturbed
Appealing ☐ 5 ☐ 4 ☐ 3 ☐ 2 ☐ 1 Unappealing

Beaver

Beaver Signs: ☐ Absent ☐ Rare ☐ Common
Beaver Flow Modifications: ☐ None ☐ Minor ☐ Major

Dominant Land Use

Dominant Land Use Around 'X' ☐ Forest ☐ Agriculture ☐ Range ☐ Urban ☐ Suburban/Town
If Forest, Dominant Age Class ☐ 0 - 25 yrs. ☐ 25 - 75 yrs. ☐ > 75 yrs.

WEATHER

GENERAL ASSESSMENT (Biotic integrity, Vegetation diversity, Local anecdotal information)

STREAM ASSESSMENT FORM - STREAM/RIVERS (cont.)

Reviewed by (Initial): _____

STORET/
SITE ID: _____ DATE: ____ / ____ / 2 0 ____

GENERAL ASSESSMENT (continued)

STREAM VERIFICATION FORM-WADEABLE STREAMS

Reviewed by (initial):_____

SITE NAME: **DATE:___ ___/___ ___/ 2 0 ___ ___ TEAM:_____**

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ UCASE ID (if applicable): UT___ ___ST-___ ___ ___ VISIT: 0 1 2 3

STREAM VERIFICATION INFORMATION

Stream Verified by (fill in all that apply): ○ **GPS** ○ **Local Contact** ○ **Signs** ○ **Roads** ○ **Topo Map** ○ **Crew Experience**

○ **Other (Describe here):** ○ **Not Verified (Explain in Comments)**

Coordinates	Latitude North	Longitude South	Datum: (Circle one)
DMS **Map/Defined pre-trip** **(fill in only one unit)** Decimal Degrees UTM	___ ___ ___ ___ ___ ___ .___ ___ ___.___ ___ ___ ___ ___ ___ ___ ___ ___ ___ ___ ___ ___	___ ___ ___ ___ ___ ___. ___ ___ ___ ___.___ ___ ___ ___ ___ ___ ___ ___ ___ ___ ___ ___	NAD 27 NAD 83
DMS **GPS/Actual at F(X)-SITE** **(fill in only one unit)** Decimal Degrees UTM	___ ___ ___ ___ ___ ___. ___ ___ ___.___ ___ ___ ___ ___ ___ ___ ___ ___ ___ ___ ___ ___	___ ___ ___ ___ ___ ___. ___ ___ ___ ___.___ ___ ___ ___ ___ ___ ___ ___ ___ ___ ___ ___	**Elevation (ft):** _____

DID YOU SAMPLE THIS SITE?

○ **YES** If YES, circle-in one below

SAMPLEABLE (Choose method used)

○ **Wadeable -** Continuous flowing water; 90%-100% wadeable

○ **Partial Wadeable -** Continuous flowing water; 50%-89% wadeable

○ **Wadeable Interrupted -** Flowing water not continuous along reach but ≥50% flowing

○ **Impounded by Beaver -** Beaver dam present at site but ≥50% of reach still in its channel and wadeable

○ **Altered Channel -** Stream present but not as on map

○ **NO** If NO, check one below

NON-SAMPLEABLE (Choose method used)

○ **Dry Channel -** Less than 50% site has flowing water

○ **Map Error -** No evidence channel/waterbody present

○ **Impounded by Beaver -** Too deep or water outside of its channel

○ **Impounded Stream** - Site underneath lake or pond (human caused or natural, discluding beaver)

○ **Wetland -** No definable channel

○ **Other -** Explain in comments

DIRECTIONS TO STREAM SITE:

STREAM VERIFICATION FORM-WADEABLE STREAMS (cont.)

Reviewed by (initial):_____

SITE NAME:	**DATE:___ ___/___ ___/ 2 0 ___ ___ TEAM:_____**
SITE/STORET ID:___ ___ ___ ___ ___ ___ ___	**UCASE ID (if applicable): UT___ ___ST-___ ___ ___ VISIT: 0 1 2 3**
LAND OWNERSHIP INFO/GENERAL COMMENTS ABOUT SITE LOCATION:	

STREAM REACH DETERMINATION

Channel Width Used to Define Reach (m)	Total Reach Length Intended (m)	COMMENTS
___ ___ ___	___ ___ ___ ___ ___	

SKETCH MAP - Arrow indicates North

PERSONNEL

NAME	DUTIES	
	Bio/Chem Sampling	Habitat
_____	○	○
_____	○	○
_____	○	○
_____	○	○
_____	○	○

United States
Environmental Protection
Agency

Office of Research and
Development
Washington, DC 20460

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October 2006

Surface Waters

Western Pilot Study:

Field Operations Manual for Wadeable Streams

Environmental Monitoring and Assessment Program

EPA/620/R-06/003
October 2006

ENVIRONMENTAL MONITORING AND ASSESSMENT PROGRAM-SURFACE WATERS

WESTERN PILOT STUDY:

FIELD OPERATIONS MANUAL FOR WADEABLE STREAMS

by

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ABSTRACT

This document describes field procedures that were used during the Environmental Monitoring and Assessment Program (EMAP) Western Pilot Study, conducted from 1999 through 2004. Objectives for EMAP involve developing appropriate scientific and technical tools for evaluating ecological condition on regional and national scales. The procedures in this document are based on previously published EMAP procedures, modified to adapt them to the specific requirements of the Western Pilot Study. Procedures are described for collecting field measurement data and/or acceptable samples for several response and stressor indicators, including aquatic vertebrate assemblages, benthic macroinvertebrate assemblages, periphyton assemblages, water chemistry, physical habitat, invasive riparian plants, and fish tissue contaminants. Additional information on data management, safety and health, and other logistical aspects are integrated into the procedures and overall operational scenario. Flowcharts and other graphic aids summarize specific field activities required to visit a stream site and collect data for these indicators. Tables give step-by-step instructions for each procedure. These figures and tables are designed to be extracted and bound separately to make a convenient reference guide for field teams. The manual also includes example field data forms for recording field measurements and sample tracking information. Checklists of all supplies and equipment needed for each field task are included to help ensure that these materials are available when required. These procedures should be considered for future use in field studies sponsored by EMAP, related research projects such as the Regional Environmental Monitoring and Assessment Program (R-EMAP), and possibly in other stream monitoring programs designed to assess the ecological condition of wadeable streams.

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ACRONYMS, ABBREVIATIONS, AND MEASUREMENT UNITS

Acronyms and Abbreviations

AFDM	Ash-free dry mass
APA	Acid/Alkaline phosphatase activity
BEAST	Benthic Assessment Sediment
BPJ	Best professional judgment
BOD	Biological Oxygen Demand
CENR	(White House) Committee on the Environment and Natural Resources
CFR	Code of Federal Regulations
CPR	Cardiopulmonary resuscitation
CWA	Clean Water Act
dbh	Diameter at breast height
DC	Direct Current
DD	Decimal degrees
DIC	Dissolved inorganic carbon
DLGs	Digital line graphs
DMS	Degrees, minutes, and seconds
DO	Dissolved oxygen
DOT	Department of Transportation
EERD	Ecological Exposure Research Division
EMAP	Environmental Monitoring and Assessment Program
EMAP-SW	Environmental Monitoring and Assessment Program-Surface Waters Resource Group
EMAP-W	Environmental Monitoring and Assessment Program-Western Pilot Study
EPA	U.S. Environmental Protection Agency
ERB	Ecosystems Research Branch
GIS	Geographic Information System
GPRA	Government Performance and Results Act
GPS	Global Positioning System
HDPE	High density polyethylene
IATA	International Air Transport Association
ID	Identification
LWD	Large woody debris
MAHA	Mid-Atlantic Highlands Assessment
MAIA	Mid-Atlantic Integrated Assessment
NAD	North American Datum
NAWQA	National Water-Quality Assessment Program
NERL	National Exposure Research Laboratory
NHD	National Hydrography Dataset

ACRONYMS, ABBREVIATIONS, AND MEASUREMENT UNITS (CONTINUED)

Acronyms and Abbreviations (continued)

NHEERL	National Health and Environmental Effects Research Laboratory
NOAA	National Oceanic and atmospheric Administration
NSWS	National Surface Water Survey
ORD	Office of Research and Development
OSHA	Occupational Safety and Health Administration
pdf	Portable document file
P-hab	Physical habitat
PVC	Polyvinyl chloride
QA	Quality assurance
QAPP	Quality assurance project plan
QC	Quality control
QCCS	Quality control check sample
RBP	Rapid Bioassessment Protocol
R-EMAP	Regional Environmental Monitoring and Assessment Program
RF3	U.S. EPA River Reach File
RIVPACS	River Invertebrate Prediction and Classification System
SL	Standard length
SOP	Standard Operating Procedure
TIME	Temporally Integrated Monitoring of Ecosystems
TL	Total length
UN	United Nations
USGS	United States Geological Survey
WED	Western Ecology Division
WRS	Willamette Research Station (Corvallis, OR)
WSA	Wadeable Streams Assessment
YOY	Young of year
YSI	Yellow Springs Instrument system

Measurement Units

A	Ampere
cm	Centimeter
ft	Foot
gal	Gallon
ha	Hectare
Hz	Hertz
in	Inches
L	Liter
m	Meter
m^2	Square meters
mg/L	Milligram per liter

ACRONYMS, ABBREVIATIONS, AND MEASUREMENT UNITS (CONTINUED)

Acronyms and Abbreviations (continued)

mm	Millimeter
µm	Micrometer
µS/cm	Microsiemens per centimeter
mS/cm	Millisiemens per centimeter
msec	Millisecond
N	Normal (equivalents per liter)
ppm	Parts per million
pps	Pulses per second
psi	Pounds per square inch
S/cm	Siemens per centimeter
V	Volts
VA	Volt-ampere

SECTION 1
INTRODUCTION

by
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This manual contains procedures for collecting samples and measurement data from various biotic and abiotic components of streams in the western United States for the U.S. Environmental Protection Agency (EPA) Environmental Monitoring and Assessment Program (EMAP) Western Pilot Study (EMAP-W). These procedures are designed for use during a one-day visit by a team of three or four persons to sampling sites located on wadeable streams or rivers (typically Strahler stream order 1 through 3 as depicted on 1:100,000 scale topographic maps, or higher orders for semiarid and arid regions of the western U.S.). The purposes of this manual are to: (1) document the procedures used in the collection of field data and various types of samples for EMAP-W and (2) provide these procedures for use by other groups implementing stream monitoring programs similar to EMAP.

These procedures were modified from those initially developed for EMAP research studies in the eastern U.S and published in Lazorchak et al. (1998). They were initially developed based on information gained from a workshop of academic, State, and Federal experts (Hughes 1993), and subsequent discussions between aquatic biologists and ecologists both within EMAP and various Federal and State scientists in the western U.S.

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1.1 OVERVIEW OF EMAP-SURFACE WATERS

The U.S. EPA implemented EMAP to develop the necessary monitoring tools to determine the current status, extent, changes and trends in the condition of our nation's ecological resources on regional and national scales (U.S. EPA 1998a, U.S. EPA 2002a). The EMAP Surface Waters Resource Group (EMAP-SW) is charged with developing the appropriate tools to assess the health of lakes, streams, and wetlands in the United States. These activities support two EPA strategic goals, *Clean and Safe Water* and *Healthy Communities and Ecosystems* (U.S. EPA 2003a, 2003b), to improve the EPA's abilities to execute its legislative mandates under both the Clean Water Act (CWA) and the Government Performance and Results Act (GPRA).

The tools being developed by EMAP-SW include appropriate indicators of ecological condition, statistical sampling designs to determine the status and extent of condition and to detect regional-scale trends in condition (e.g., Urquhart and Kincaid 1999), and approaches to characterizing the benchmark or *reference* conditions that are required to define aquatic assemblage or ecosystem condition (e.g., Bailey et al. 2004, Stoddard et al. 2006a). Collectively, these tools provide the science needed for a monitoring framework (e.g., Committee on Environment and Natural Resources 1997) to assess objectively whether the nation's ecological resources are responding positively, negatively, or not at all, to existing or future regulatory actions (Whittier and Paulsen 1992, Hughes et al. 2000).

Several EMAP-SW research studies were conducted prior to EMAP-W. These included 1) northeastern lakes (Paulsen et al. 1991, Larsen and Christie 1993), 2) wetlands in the Prairie Pothole region of North and South Dakota (Peterson et al. 1997), 3) lakes and streams of the northeastern and eastern U.S., designed specifically to assess the changes and trends in chemical condition in acid-sensitive surface waters resulting from the 1990 Clean Air Act amendments (Ford et al. 1993, Stoddard et al. 1996, 2003). EMAP-SW began stream sampling studies in 1993 by collaborating on the Mid-Atlantic Highlands Assessment (MAHA, Davis and Scott 2000, U.S. EPA 2000). This collaboration continued as the MAHA project evolved into the Mid-Atlantic Integrated Assessment (MAIA) project (Bradley and Landy 2000), which produced assessments of the condition of estuarine resources (U.S. EPA 1998b), landscapes (Jones et al. 1997, 2001), and surface waters (Stoddard et al. 2006b). Tools and procedures developed by EMAP-SW have also been adapted and used in projects sponsored by the Regional Environmental Monitoring and Assessment Program (R-EMAP, U.S. EPA 1993). In 2000, EMAP-SW assisted in research on adapting EMAP protocols for very large rivers such as the Ohio, Missouri, and Mississippi (Schweiger et al. 2005) and in 2004 helped develop a formal protocol for Great Rivers

(Angradi et al. 2006). Also in 2004, EMAP-SW collaborated with the U.S. EPA Office of Water and State water quality agencies on the Wadeable Streams Assessment (WSA), an initiative to produce a national-scale assessment of stream resources by the end of 2005 (U.S. EPA 2004a, 2006).

Field procedures developed for EMAP-SW research projects are available for lakes (Baker et al. 1993), for wadeable streams in the eastern U.S. (Lazorchak et al. 1998), and for nonwadeable streams and rivers in the eastern U.S (Lazorchak et al. 2000). Procedures used for the WSA (U.S. EPA 2004b) are slightly modified from those presented in this manual. Field procedures used for non-wadeable streams and rivers for EMAP-W are presented in a companion document to this manual (Peck et al. in press).

1.2 WESTERN PILOT STUDY

Details regarding EMAP-W are described in the peer-reviewed research plan (U.S. EPA 2000), the statistical summary report (Stoddard et al. 2005a), and the assessment report (Stoddard et al. 2005b). The primary goals of EMAP-W were to:

1. Develop the monitoring tools (ecological indicators, stream survey design, estimates of reference condition) necessary to produce unbiased estimates of the ecological condition of surface waters across a large geographic area (or areas) of the western U.S.; and
2. Demonstrate those tools in a large-scale assessment of the status and extent of ecological condition. Quantifying a regional trend in condition was not an objective, as it required a design that included a component to more specifically address objectives for trend detection (multiple status surveys and/or repeated sampling of sites across several years).

Measurable objectives of EMAP-W, expressed as three general assessment questions included:

1. What proportion of stream and river miles in the western U.S. are in acceptable (or poor) biological condition?
2. What is the relative importance of potential stressors (e.g., physical habitat modification, sedimentation, nutrients, etc.) in streams and rivers across the western U.S.?
3. What stressors are associated with streams and rivers having biological assemblages in poor condition?

EMAP-W advanced the science of monitoring with respect to the type of systems addressed (mountainous and arid systems) and the size of the region covered (essentially one third of the conterminous U.S.). EMAP-W also applied EMAP design and assessment tools to answer some urgent and practical assessment questions facing the western EPA Regional Offices, using a methodology that could be extended to the entire nation, as was demonstrated by the WSA for benthic macroinvertebrate assemblages, water chemistry, and physical habitat.

The survey design developed for EMAP-W selected sites that were statistically representative of the resource population of interest, termed the *target population* (Stevens and Olsen 2004, Stoddard et al. 2005a, 2005b). The survey design allows results for a comprehensive suite of ecological indicators (Section 1.3) to be extrapolated from the sites sampled to the entire target population, or to selected subpopulations (e.g., States, eco-regions, drainage basins) having an adequate sample size. The target population of interest for EMAP-W was all perennial streams and rivers as represented in the EPA's River Reach File (RF3) and the U.S. Geological Survey (USGS) Pacific Northwest River Reach file, with the exception of the Columbia River, the mainstem Missouri River, and the lower portions of the Snake and Colorado Rivers. Figure 1-1 presents the sites from the target population that were sampled, showing the various study components that were integrated into a single survey design. A detailed description of the survey design and the site selection process is presented in the EMAP-W statistical summary (Stoddard et al. 2005a). An important characteristic of the survey design is that not all sites initially identified for sampling turn out to be sampled; some do not meet the criteria for inclusion in the target population, and some target sites cannot be sampled for various reasons (e.g., physically inaccessible or access permission is not obtained). The survey design allows for estimation of these various types of errors, resulting in a more accurate and robust determination of the resource population that can be confidently assessed.

In addition to the sites that were sampled as part of the survey design, candidate reference sites (Figure 1-2) were selected by several different approaches to provide the capability for characterizing reference condition for the various ecological indicators. Details regarding candidate reference site selection, the final selection of reference sites, and the characterization of reference conditions are found in the EMAP-W statistical summary (Stoddard et al. 2005a) and Stoddard et al. (2006a).

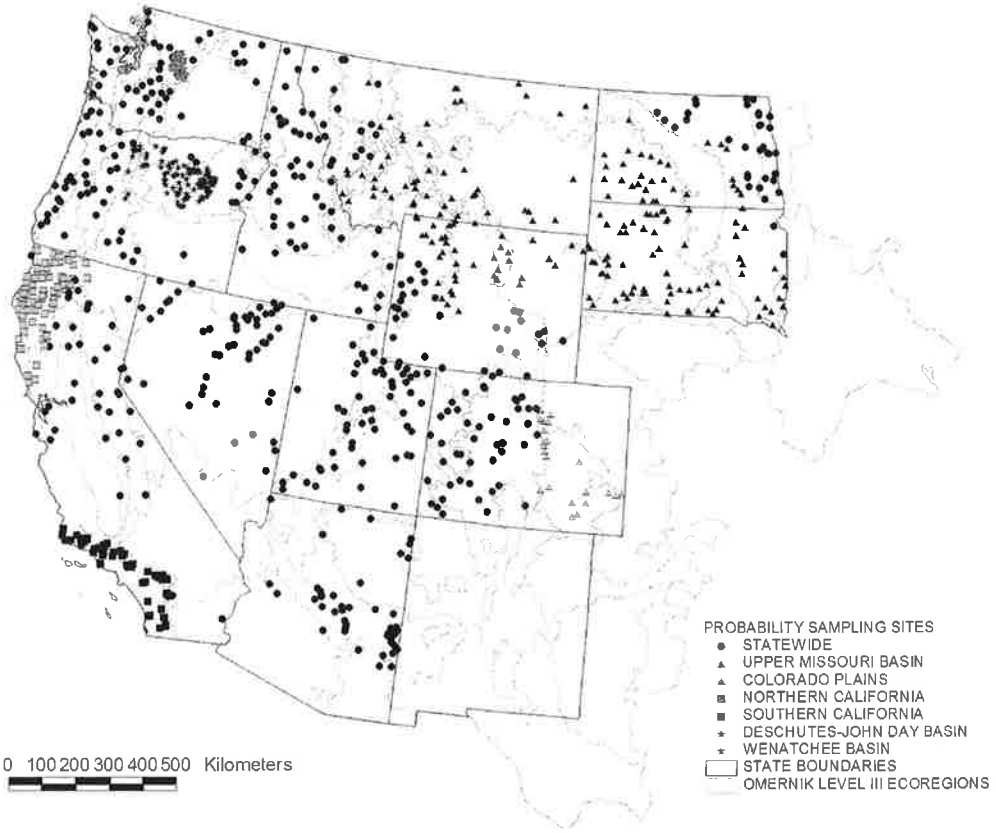

Figure 1-1. Study areas and locations of sites sampled for the EMAP Western Pilot Study. Modified from Stoddard et al. 2005a.

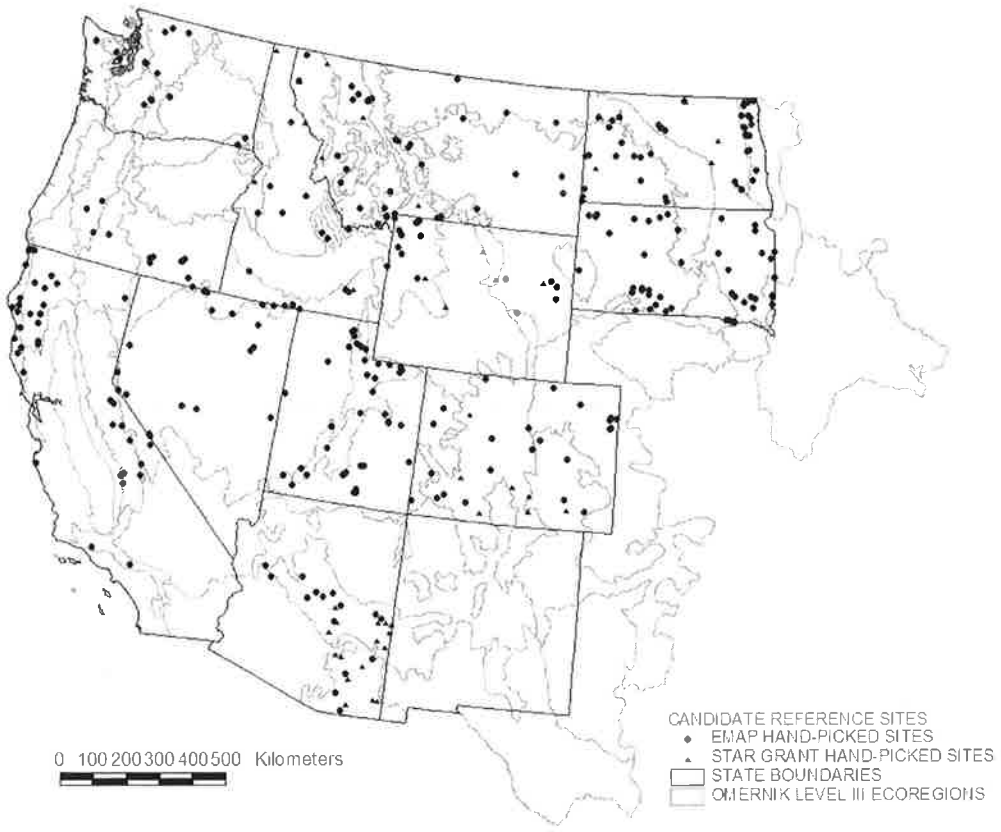

Figure 1-2. Candidate reference sites sampled for the EMAP Western Pilot Study. Modified from Stoddard et al. 2005a.

1.3 SUMMARY OF ECOLOGICAL INDICATORS

The following sections describe the rationale for each of the ecological indicators included in the stream sampling procedures presented in this manual. This information is presented to help users understand the significance of certain aspects of the field procedures.

EMAP-SW considers two principal types of indicators (Suter 1990), *condition* and *stressor* (U.S. EPA 1998a, Hughes et al. 2000). Condition indicators are biotic or abiotic characteristics of an ecosystem that can provide an estimate of the condition of an ecological resource with respect to some environmental value, such as biotic integrity. Stressor indicators are characteristics that are expected to change the condition of a resource if the intensity or magnitude is altered. Additional details are given in the relevant sections that follow. EMAP-SW has typically included condition indicators based on different biological assemblages, following Barbour et al. (1999), as each assemblage may exhibit different responses to stressor variables (in terms of type or severity).

1.3.1 Water Chemistry

Data are collected from each stream for a variety of physical and chemical constituents. Information and stressor indicators derived from these data are used to evaluate stream condition with respect to stressors such as acidic deposition, nutrient enrichment, and other inorganic contaminants. In addition, streams can be classified with respect to water chemistry type, water clarity, mass balance budgets of constituents, temperature regime, and presence of anoxic conditions. Chemical data are also important in characterizing reference conditions for biological assemblage indicators. Examples of relationships between stream chemistry and watershed-level land use data are described in Herlihy et al. (1998).

1.3.2 Physical Habitat

Naturally occurring differences among surface waters in physical habitat structure and associated hydraulic characteristics contribute to much of the observed variation in species composition and abundance within a zoogeographic province (Hynes 1970, Allen 1995). The structural complexity of aquatic habitats provides the variety of physical and chemical conditions to support diverse biotic assemblages and maintain long-term stability. Anthropogenic alterations of riparian areas and stream channels, wetland drainage, grazing and agricultural practices, and stream bank modifications such as revetments or develop-

ment, generally act to reduce the complexity of aquatic habitat and result in a loss of species and ecosystem degradation.

Stressor indicators derived from data collected about physical habitat quality are used to help explain or diagnose stream condition relative to various condition indicators. Important attributes of the physical habitat in streams include channel dimensions, gradient, substrate characteristics; habitat complexity and cover; riparian vegetation cover and structure; disturbance due to human activity, and channel-riparian interaction (Kaufmann 1993). Overall objectives for this suite of indicators are to develop quantitative and reproducible indices, using both multivariate and multimetric approaches, to classify streams and to monitor biologically relevant changes in habitat quality and intensity of disturbance. Kaufmann et al. (1999) discuss procedures for reducing EMAP field habitat measurements and observations to metrics that describe channel and riparian habitat at the reach scale.

1.3.3 Periphyton Assemblage

Periphyton are the algae, fungi, bacteria, and protozoa associated with substrates in aquatic habitats. These organisms exhibit high diversity and are a major component in energy flow and nutrient cycling in aquatic ecosystems. Many characteristics of periphyton assemblage structure and function can be used to develop indicators of ecological conditions in streams (Stevenson and Bahls 1999, Hill et al. 2000). Periphyton are sensitive to many environmental conditions, which can be detected by changes in species composition, cell density, ash free dry mass (AFDM), chlorophyll *a*, and enzyme activity (e.g., alkaline and acid phosphatase). Each of these characteristics may be used, singly or in concert, to assess condition with respect to societal values such as biological integrity and trophic condition.

Condition indicators developed from periphyton assemblage data involve the calculation of composite indices for biotic integrity, ecological sustainability, and trophic condition. The composite indices can be calculated from measured or derived first-order and second-order indices. The first-order indices include species composition (richness, diversity), cell density, AFDM, chlorophyll, and enzyme activity (e.g., Sayler et al. 1979), which individually are indicators of ecological condition in streams. Second-order indices can be calculated from periphyton assemblage characteristics, such as the autotrophic index (Weber 1973), assemblage similarity compared to reference sites, and autecological indices (e.g., Lowe 1974, Lange-Bertalot 1979, Charles 1985, Dixit et al. 1992, Potapova et al. 2004). Hill et al. (2000, 2003), Fore (2002), and Wang et al. (2005) describe the

development of multimetric indexes based on periphyton assemblages in wadeable streams.

1.3.4 Benthic Macroinvertebrate Assemblage

Benthic macroinvertebrates inhabit the sediments or live on the bottom substrates of streams. Monitoring these assemblages is useful in assessing the status of the water body and discerning trends in ecological condition. Benthic macroinvertebrate assemblages respond differently to a wide array of stressors. As a result, it is often possible to determine the type of stress that has affected a benthic macroinvertebrate assemblage (Plafkin et al. 1989; Klemm et al. 1990; Barbour et al. 1999). Because many macroinvertebrates have relatively long life cycles of a year or more and are relatively immobile, macroinvertebrate assemblage structure is a function of past conditions.

Rosenberg and Resh (1993) and Bonada et al. (2006) present and compare various approaches to biological assessment using benthic invertebrates, and Norris (1995) briefly summarizes and discusses approaches to analyzing benthic macroinvertebrate community data. For EMAP-W, two different approaches were used to develop ecological indicators based on benthic invertebrate assemblages. The first is a multimetric approach, where different structural and functional attributes of the assemblage are characterized as metrics. Individual metrics that respond to different types of stressors are scored against expectations under conditions of minimal human disturbance. The individual metric scores are then summed into an overall index value that is used to judge the overall level of impairment of an individual stream reach. Examples of multimetric indices based on benthic invertebrate assemblages include Kerans and Karr (1993), Fore et al. (1996) Barbour et al. (1995, 1996), Klemm et al. (2003), and Ode et al. (2005). The multimetric index developed for EMAP-W is described by Stoddard et al. (2005a).

The second approach was to develop indicators of biological condition based on multivariate analysis of benthic assemblages and predictive models based on associated abiotic variables (Hawkins et al. 2000, Van Sickle et al 2005, Mazor et al. 2006). Examples of this type of approach as applied to benthic invertebrate assemblages include RIVPACS (Wright 1995), BEAST (Reynoldson et al. 1995), and Hawkins et al. (2000). Hawkins (2006) compares the performance of different types of approaches, including multimetric and predictive models, used to assess condition at regional scales. The predictive model indicator developed for EMAP-W is described in Stoddard et al. (2005a).

1.3.5 Aquatic Vertebrate Assemblage

Aquatic vertebrate assemblages of interest to EMAP-W include fish and amphibians. The fish assemblage represents a critical component of biological condition from both an ecosystem function and a public interest perspective. Historically, fish assemblages have been used for biological monitoring in streams more often than in lakes (e.g., Plafkin et al. 1989, Karr 1991). Fish assemblages can serve as good indicators of ecological conditions because fish are long-lived and mobile, forage at different trophic levels, integrate effects of lower trophic levels, and are reasonably easy to identify in the field (Plafkin et al. 1989, Barbour et al. 1999). Amphibians comprise a substantial portion of vertebrate biomass in streams of many areas of the U.S. (Hairston 1987, Bury et al. 1991). Reports of dramatic declines in amphibian biodiversity (e.g., Blaustein and Wake 1990, Phillips 1990) have increased the level of interest in monitoring these assemblages. Amphibians may also provide more information about ecosystem condition in the headwater or intermittent streams in certain areas of the country than other biological response indicators (Hughes 1993). The objective of field sampling is to collect a representative sample of the aquatic vertebrate assemblage by methods designed to 1) collect all except very rare species in the assemblage and 2) provide a measure of the abundance of species in the assemblages (McCormick 1993, Reynolds et al. 2003). Information collected for EMAP that is related to vertebrate assemblages in streams includes assemblage attributes (e.g., species composition and relative abundance) and the incidence of external pathological conditions.

Indicators based on vertebrate assemblages were developed primarily using the multimetric approach described in Section 1.3.4 for benthic macroinvertebrates, and originally conceived by Karr and others (Karr et al. 1986). Simon and Lyons (1995) provide a recent review of multimetric indicators as applied to stream fish assemblages. McCormick et al. (2001) provide an example of a multimetric indicator developed for the Mid-Atlantic region using EMAP-SW data, based on an evaluation process described by Hughes et al. (1998). Hughes et al. (2004) present a multimetric indicator to assess condition in cold-water streams in the Pacific Northwest. The multimetric indicator developed for EMAP-W is described by Stoddard et al. (2005a).

1.3.6 Fish Tissue Contaminants

For EMAP-W, the primary purpose of determining contaminant levels in fish tissue is to provide a measure of the potential exposure of stream systems to toxic compounds. It is also meant to be used in conjunction with the other stressor indicators (physical habitat, water chemistry, land use, human population density, other records of relevant anthro-

pogenic stresses) and condition indicators (fish, macroinvertebrates, periphyton) to help diagnose whether the probable cause of stream degradation, when it is shown by the condition indicators to occur, is water quality, physical habitat, or both.

Whole fish are collected and analyzed because they present fewer logistical problems for field crews (no gutting required in the field) and the analytical lab (no filleting necessary). Yeardley et al. (1998), Peterson et al. (2002), and Lazorchak et al. (2003) provide examples of the use of fish tissue contaminant data in EMAP studies.

The relative bioaccumulation of contaminants by large and small stream fish is not known, so two different types of samples were prepared. One type of sample was prepared using a species whose adults are small (e.g., small minnows, sculpins, or darters). The second type of sample was prepared using a species whose adults are of larger size (e.g., suckers, bass, trout, sunfish, carp). In addition to being more ubiquitous than the larger fish (and therefore more likely to be collected in sufficient numbers to combine into a composite sample), small fish have other advantages over large fish. Most important, it may be possible to get a more representative sample of the contaminant load in that stream segment (although it could be at a lower level of bioaccumulation) by creating a composite sample from a larger number of small individuals than by compositing a few individuals of larger species (Peterson et al. 1996, Lazorchak et al. 2003). The major advantage that larger fish could potentially offer, whether predators (piscivores) or bottom feeders, is a higher level of bioaccumulation and thus greater sensitivity to detect contaminants.

1.4 RESPONSE DESIGN

For a given indicator, the *response design* is the process of deciding what to measure and how to measure it at a given sampling point (Stevens and Urquhart 2000). For EMAP-W, the response design for each indicator was integrated into an overall response design that was applied at every sampling site. This integrated response design addressed the sampling requirements for all indicators, minimized the level of disturbance to the stream during sampling, and minimized the time required to complete all the required activities at a site. For all indicators, a spring-summer baseflow period was defined as the appropriate *index period* to collect samples and measurements.

For water chemistry and discharge, spatial variability over relatively short distances is relatively small (barring anomalous inputs or withdrawals) in comparison to variability among sites, so a single sample at the sampling point adequately characterizes the general chemical and flow conditions present at the sampling point. For biological assemblages and

physical habitat, collection or characterization at a single point at a sampling site does not provide adequate representation of the assemblage or physical habitat conditions present (different habitat types generally yield different species), and so a *support reach* about each sampling point is necessary (Barbour et al. 1999). The overall length of the support reach is determined primarily by the requirements for the vertebrate assemblage indicator (which were the most mobile organisms sampled) to collect the majority of commonly occurring species in their rank abundance (Reynolds et al. 2003). This distance (40 times the mean wetted channel width at the sampling point) also closely coincides with approximately 4 riffle-pool sequences or 2 meander cycles, which were recommended as the length of reach to sample to adequately characterize the various components of physical habitat (Robison 1998, Kaufmann et al. 1999). Results from Li et al. (2001) also suggested a support reach length of 40 channel widths was adequate for assessing benthic macroinvertebrate assemblages.

To ensure allocation of sampling effort throughout the entire support reach, and maximize consistency in sampling across all sampling points and comparability of the results in the assessment of condition, a systematic approach to the collection of samples or field measurements was used, based on the establishment of 11 equidistant cross-sectional transects within the support reach. Habitat data, benthic macroinvertebrate samples, and periphyton samples were collected at each transect, while other habitat measurements and aquatic vertebrates were collected between each of the transects.

1.5 OBJECTIVES AND SCOPE OF THE FIELD OPERATIONS MANUAL

Only field-related sampling and data collection activities are presented in this manual. Laboratory procedures and methods (including sample processing and analytical methods) associated with each ecological indicator are summarized in Chaloud and Peck (1994); detailed procedures are the same (or nearly so) as those published for the WSA (U.S. EPA 2004c).

This manual is organized to follow the sequence of field activities during the 1-day site visit. Section 2 presents a general overview of all field activities. Section 3 presents those procedures that are conducted at a *base location* before and after a stream site visit. Section 4 presents the procedures for verifying the site location and defining a reach of the stream where subsequent sampling and data collection activities are conducted. The next nine sections (5 through 13) describe the procedures for collecting samples and field measurement data for various condition and stressor indicators. Specific procedures associated with each indicator are presented in tables that can be copied, laminated if

desired, and taken into the field for quick reference. Section 14 describes the final activities that are conducted before leaving a stream site. Appendix A contains a list of all equipment and supplies required by a crew to complete all field activities at a stream. Appendix B provides a summary of the changes from previously published field procedures for EMAP-SW, and minor modifications made each year during EMAP-W to clarify procedures and data forms. These modifications provide traceability to previous drafts of this manual that were used each year. Appendix C (on CD-ROM) contains a set of blank field data forms. Appendix D (on CD-ROM) contains photos and detailed taxonomic characteristics and habitat information about the invasive riparian plant species presented in Section 8.

1.6 QUALITY ASSURANCE

Quality assurance is a required element of all EPA-sponsored studies that involve the collection of environmental data (U.S. EPA 2001, 2002b). Large-scale and/or long-term monitoring programs such as those envisioned for EMAP require a comprehensive and rigorous quality assurance (QA) program that can be implemented consistently by all participants throughout the duration of the monitoring period. A QA program comprises various planning, implementation, assessment, reporting, and quality improvement activities that work collectively to provide confidence in the data or product from a project or study. A QA project plan (QAPP; e.g., Chaloud and Peck [1994] for EMAP-SW activities) contains more detailed information regarding QA and quality control (QC) activities and procedures associated with general field operations, sample collection, measurement data collection for specific indicators, laboratory operations, and data validation and reporting activities. The QAPP developed for EMAP-W will be published as a separate report and/or included with the metadata compiled for the database of results. The QA program for EMAP-W is similar to that implemented for the WSA (U.S. EPA 2004a).

Quality control activities serve to ensure data will meet the performance objectives established for each method or technique used for a project. Quality control activities associated with field operations are integrated into the field procedures. Important QA activities associated with field operations include a comprehensive training program that includes practice sampling visits, and the use of a qualified museum facility or laboratory to confirm any field identifications of biological specimens. The overall sampling design for EMAP-W includes a subset of sites (at least 1 per state per year) that are revisited twice per year for two years to obtain estimates of important components of variance for estimating status and trend (e.g., Larsen 1997, Urquhart et al., 1998) for the various ecological indicators.

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NOTES

SECTION 2
OVERVIEW OF FIELD OPERATIONS

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

2.1 DAILY OPERATIONAL SCENARIO

The field team is divided into two groups, termed the *Geomorphs* and the *Biomorphs*, that reflect their initial responsibilities more than their expertise. The geomorphs are primarily responsible for conducting the intensive physical habitat characterization. The biomorphs are primarily responsible for collecting biological samples. Table 2-1 provides the estimated time required to conduct various field activities. Figure 2-1 presents one scenario of the general sequence of activities conducted at each stream reach. For some wide, shallow streams, the required reach length and/or the larger area requiring sampling effort may necessitate two days be allocated to complete all required activities.

Upon arrival at a stream site, the geomorphs verify and document the site location, determine the length of stream reach to be sampled, and establish the required transects

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

Activity	Group	Est. Time Required
Site verification and establishing sampling reach and transects	Geomorphs (2 persons)	1 hr
Water chemistry sampling and stream discharge determination	Biomorphs (2 persons)	1 hr
Collecting and processing benthos and periphyton samples	Biomorphs (2 persons)	2 hr
Intensive physical habitat characterization, including legacy tree identification and presence of invasive plant taxa	Geomorphs (2 persons)	2 to 3 hr
Aquatic vertebrate sampling and processing	Geomorphs and Biomorphs (4 persons)	1 to 3 hr
Rapid habitat assessment Visual stream assessment	Biomorphs (2 persons)	0.5 hr
Sample tracking and packing	Geomorphs (2 persons)	1 hr
SUMMARY	19 to 27.5 person-hours	8.5 to 11.5 hrs per team[a]

[a] For wider wadeable streams (e.g., > 20 m), it may require more than one day to complete all required activities.

(Section 4). The biomorphs collect samples and (optional) field measurements for water chemistry (Section 5) and determine stream discharge (Section 6). The biomorphs also collect periphyton and benthos samples (Sections 9 and 10, respectively). The geomorphs conduct the intensive physical habitat characterization (Section 7). Both groups are involved with collecting aquatic vertebrates (Section 11) and preparing fish samples for analysis of toxic contaminants (Section 12). Finally, the biomorphs conduct a (optional) habitat characterization based on the Rapid Bioassessment Protocols (RBP; Plafkin et al. 1989, Barbour et al. 1999), and a visual assessment of the stream and its surrounding area (Section 13). All team members participate in the final activities at a site (Section 14), which include reviewing data forms, and preparing samples for transport and shipment (Section 3).

2.2 GUIDELINES FOR RECORDING DATA AND INFORMATION

During the one-day visit to a stream, a field team obtains and records a substantial amount of data and other information for all of the various ecological indicators described

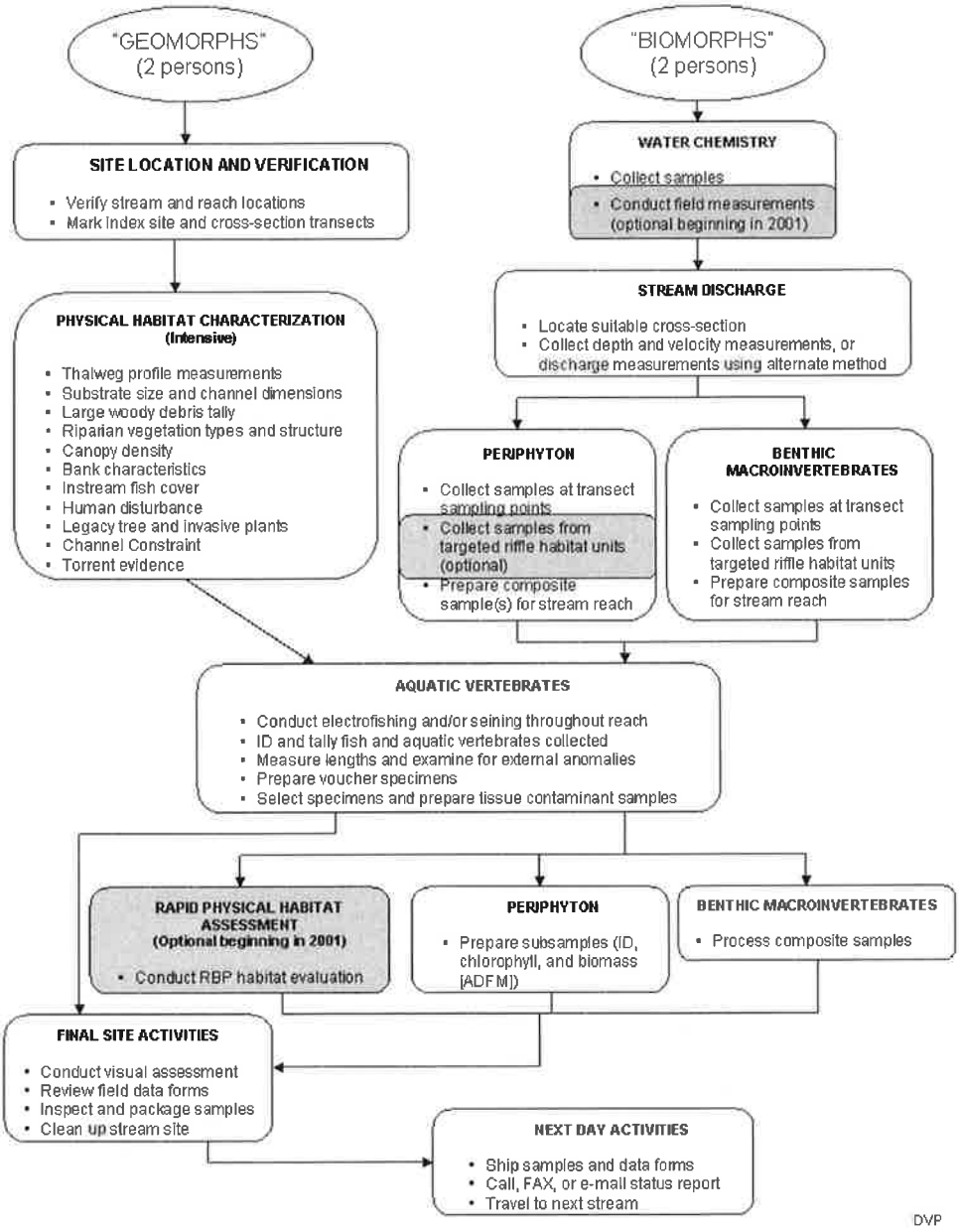


Figure 2-1. General sequence of stream sampling activities. Modified from Chaloud and Peck, (1994). Shaded activities are optional.

in Section 1.3. In addition, all the associated information for each sample collected must be recorded on labels and field data forms to ensure accurate tracking and subsequent linkage of other data with the results of sample analyses.

The field data forms developed for EMAP-W are compatible with an optical scanner system to allow rapid entry, review, and conversion of the information recorded on a printed form into one or more electronic files and reduce the need for manual data entry. While these forms facilitate data recording by the field crew, field and sample information must be recorded accurately, consistently, and legibly. Measurement data that cannot be accurately interpreted by others besides the field teams, and/or samples with incorrect or illegible information associated with them, are lost to the program. The cost of a sampling visit, coupled with the short index period, effectively prohibits resampling a stream when the initial information recorded was inaccurate or illegible. Some guidelines to assist field personnel with recording information are presented in Table 2-2. Examples of completed data forms and labels are presented in the sections describing field sampling and measurement procedures for different indicators.

2.3 HEALTH AND SAFETY

Collection and analysis of samples (e.g., benthic invertebrates, fish, periphyton, sediment) can involve significant risks to personal safety and health (drowning, electrical shock, pathogens, etc.). While safety is often not considered an integral part of field sampling routines, personnel must be aware of unsafe working conditions, hazards connected with the operation of sampling gear, boats, and other risks (Berry et al. 1983). Personnel safety and health are of the highest priority for all investigative activities and must be emphasized in safety and health plans for field, laboratory, and materials handling operations. Preventive safety measures and emergency actions must be emphasized. Management should assign health and safety responsibilities and establish a program for training in safety, accident reporting, and medical and first aid treatment. Safety documents and standard operating procedures (SOPs) containing necessary and specific safety precautions must be available to all field personnel. Additional sources of information regarding field and laboratory safety related to biomonitoring studies include Berry et al. (1983), U.S. EPA (1986), Ohio EPA (1990), and the U.S. Geological Survey (Yobbi et al. 1996). Note that the information provided in the following sections is general in nature, and *does not substitute for any specific health and safety requirements mandated by a specific organization.* Consult with your organization's health and safety official to determine the requirements associated with conducting the field activities described in this document.

TABLE 2-2. GUIDELINES FOR RECORDING FIELD DATA AND OTHER INFORMATION

Field Measurements:

Data Recording:

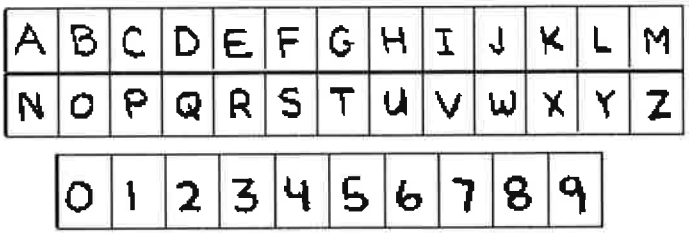
Record measurement values and/or observations on data forms preprinted on water-resistant paper.

Headers on the second pages of all forms link the data. Fill in all headers of all pages or data will be lost or linked to the wrong site record (this is a good one to review at the end of the day).

***DO NOT** mark on or around the cornerblocks or ID Box* (the squares in the corners and the funky box with the number over it). These markings are crucial to the scanning software and changing them in any way will affect performance.

Write legibly. Use a dark pencil lead that is at least a No. 2 for softness (HB), or use a dark permanent (waterproof) marking pen. Your writing must be dark enough to be picked up by the scanner. Erase mistakes completely and write the correct value whenever you can. NOTE: The scanner cannot read the number over an erasure If you must line out an incorrect value, place the correct value nearby in the appropriate box so the data entry operator can easily find it.

Use all caps when filling in the name fields on the forms. Clearly distinguish letters from numbers (e.g., *0* versus *O*, *2* versus *Z*, *7* versus *T* or *F*, etc.). Do not put lines through *7's*, *0's*, or *Z's*. Write *4's* with an open top rather than closed. Do not use slashes. Below are examples of lettering that are readable by the scanning software:



It is not necessary to write in all caps in the long comments sections on the stream verification and stream assessment forms, but write legibly (because the data entry operators still need to read it to type it in.) *Avoid marginal notes*, etc. Be concise, but avoid using abbreviations and/or shorthand notations. If you run out of space, attach a sheet of paper with the additional information, rather than trying to squeeze everything into the space provided on the form.

When you need to circle a choice, make a medium-sized circle around your choice.

(Yes) No 1 (2) 3 4

For square boxes, mark inside the box with an *X*. For circles (*bubbles*), fill in completely.

(Continued)

TABLE 2-2 (Continued)

Data Recording:

Record data and information so that all entries are obvious. Enter data completely in every field that you use. Follow the *comb guidelines*– print each number or letter in the individual space provided. Keep letters and numerals from overlapping. Record data to the number of decimal places provided on the forms. Illegible information is equivalent to no information.

If the measurement for a field is zero, enter zero. If left blank, it will be recorded as missing data. (There are parts of forms that are left blank when they are not being used. A typical example is The Stream Discharge Form. Usually only one type of velocity and discharge information is recorded and the unused areas of the form are left blank).

If the field calls for meters, write the answer in meters. Do not fill in a number and put (cm) for units. Do not add additional decimal places.

	Dist. from Bank	Velocity	Depth	Flag
1	0	0	0	
2	10	-0.1	0.6	
3	20	0.8	1.0	
4	30	1.3	1.3	

Record information on each line, even if it has to be recorded repeatedly on a series of lines (e.g., fish names or species codes, physical habitat characteristics). *Ditto marks* (") can be used if necessary and if they are clearly distinguishable from letters or numbers. ***Do not use*** a straight vertical line to indicate repeated entries.

Data Qualifiers (Flags):

Use only defined flag codes from the list below and record on data form in appropriate field. If the information is important enough to write on the page, use a *Fn* flag and put it in the comment section. If you have been instructed to collect a piece of information for which there is no space on the form, choose a flag and comment section, and use them consistently.

FLAG	COMMENT
F1, F2, etc.	Miscellaneous comments assigned by field team (e.g., Fish dead)
K	Sample not collected; No measurement or observation made
U	Suspect sample, measurement or observation, or collected using a non-standard procedure
Q	Unacceptable QC check associated with measurement
Z	Last interval sampled (e.g., stream discharge)

(Continued)

TABLE 2-2 (Continued)

Data Qualifiers (Flags):

If you cannot take a measurement, leave the measurement field blank and put the K flag in the Flag column.

	Dist. from Bank	Velocity	Depth	Flag
1	0	0	0	
2	10	-0.1	60	
3	20	0.8	100	
4	30		130	K

K Too deep and swift to obtain velocity measurement

Review of Data Forms:

Have someone who did not fill in the forms review them at the end of the day. Some information is duplicated. Sometimes, however, when one measurement is missing, many other metrics based on that measurement are also lost. Be thorough.

Examples: Site_ID--used in conjunction with Visit Date and Visit Number to merge data from different forms (unique record identifiers)
Visit Date–used in conjunction with Site ID and Visit Number to merge data from different forms (unique record identifiers)
Transect–Needed to merge different types of habitat data and to calculate many habitat metrics correctly
Increment distance (on the back of the thalweg form)-used in conjunction with all 100-150 thalweg depth measurements

Returning the Forms

Return the originals.
Make a photocopy and keep it, in case questions arise during data entry.
Try to keep the forms in their original order.
Do not staple the forms together.

Include a list of sites visited. Please include a summary list with Site ID and Visit Date with each batch of forms being returned.

(Continued)

TABLE 2-2 (Continued)

Sample Labels and Tracking

Sample Labels:

Sample Labels– Use adhesive labels with preprinted ID numbers and a standard recording format for each type of sample.

Record information on labels using a fine-point indelible marker. Cover completed labels with clear tape.

Sample Tracking Information:

Record sample ID number from the label and associated collection information on sample collection form. Use a dark pencil or pen.

Complete any sample tracking forms required. Include tracking forms with all sample shipments.

Sample Qualifiers (Flags):

Use only defined flag codes and record on sample collection form in appropriate field.

- K Sample not collected or lost before shipment; not possible to resample.
- U Suspect sample (e.g., possible contamination, does not meet minimum acceptability requirements, or collected using a nonstandard procedure)
- F*n* Miscellaneous flags (*n*=1, 2, etc.) assigned by a field team for a particular sample shipment.

Explain all flags in comments section on sample collection form.

Review of Labels and Collection Forms:

The field team compares information recorded on labels, sample collection forms, and tracking forms for accuracy before leaving a stream. Make sure Sample ID numbers match on all forms.

2.3.1 General Considerations

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

TABLE 2-3. GENERAL HEALTH AND SAFETY CONSIDERATIONS

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

Become familiar with the hazards involved and establish appropriate safety practices prior to using any sampling device. At least one individual involved in electrofishing should attend an approved electrofishing training course, such as those offered by the U.S. Fish and Wildlife Service. In many states, this is a requirement for obtaining a scientific collecting permit. Other personnel involved with electrofishing must be trained by a person experienced in this method. Reynolds (1983) and Ohio EPA (1990) provide additional information regarding electrofishing safety procedures and practices.

If boats are used to access sampling sites, consider and prepare for hazards associated with the operation of motor vehicles, boats, winches, tools, and other incidental equipment. Boat operators should be familiar with U.S. Coast Guard rules and regulations for safe boating contained in a pamphlet, *Federal Requirements for Recreational Boats*, available from a local U.S. Coast Guard Director or Auxiliary, or a state boating official (U.S.

Coast Guard 1987). In some states, a separate license may be required to operate a motorized watercraft. All boats with motors must have fire extinguishers, boat horns, life jackets or flotation cushions, and flares or communication devices.

A communications plan to address safety and emergency situations is essential. Be fully aware of all lines of communication. Field personnel should have a daily check-in procedure, and contacts for police, ambulance, fire departments, and search and rescue personnel.

Proper field clothing should be worn to prevent hypothermia, heat exhaustion, sunstroke, drowning, or other dangers. Field personnel should be able to swim. Chest waders made of rubberized or neoprene material and suitable footwear must always be worn with a belt to prevent them from filling with water in case of a fall. The use of a personal flotation device is advisable at dangerous wading stations if one is not a strong swimmer because of the possibility of sliding into deep water.

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

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

2.3.2 Safety Equipment and Facilities

Appropriate safety apparel such as waders, lab coats, gloves, safety glasses, etc. must be available and used when necessary. Bright colored caps or vests (e.g., hunter orange) should be worn during field activities. Provide cellular or satellite telephones or portable radios to field teams working in remote areas for use in case of an emergency. A first aid kit (including an emergency blanket) and a fire extinguisher should be taken to

every site. A shovel and/or a Pulaski axe may be required for sites in National or State Forest areas during fire season. Have soap and water, alcohol, or hand sanitizer solution available to clean exposed body parts that may have been contaminated by pollutants in the water. Also have skin cleanser available to prevent or reduce the effects of exposure to poison oak or sumac. Use a fume hood in the laboratory when working with carcinogenic chemicals (e.g., formaldehyde, formalin) that may produce dangerous fumes.

2.3.3 Safety Guidelines for Field Operations

General safety guidelines for field operations are presented in Table 2-4. Personnel participating in field activities on a regular or infrequent basis should be in sound physical condition and have a physical exam annually or in accordance with State or organizational requirements. Consider all surface waters and sediments as potential health hazards due to toxic substances or pathogens. Become familiar with the health hazards associated with using chemical fixing and/or preserving agents. Formaldehyde (or formalin) is highly allergenic, toxic, and dangerous to human health (carcinogenic) if utilized improperly. Chemical wastes can cause various hazards due to flammability, explosiveness, toxicity, causticity, or chemical reactivity. Dispose of all chemical wastes in accordance with approved procedures, (e.g., National Institute for Occupational Safety and Health 1981, U.S. EPA 1986).

During the course of field activities, a team may observe violations of environmental regulations, may discover improperly disposed hazardous materials, or may observe or be involved with an accidental spill or release of hazardous materials. In such cases, take the proper action and do not become exposed to something harmful. The following guidelines should be applied:

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

TABLE 2-4. GENERAL SAFETY GUIDELINES FOR FIELD OPERATIONS

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

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

- ***Never*** *disturb, or even worse, retrieve improperly disposed hazardous materials* from the field and bring them back to a facility for disposal.. To do so may worsen the impact to the area of the incident, incur personal or organizational liability, cause personal injury, or cause unbudgeted expenditures of time and money for proper treatment and disposal of the material. However, it is important not to ignore environmental incidents. You are required to notify the proper authorities of any incident of this type so they can take the necessary actions to respond properly to the incident.

- Provide the following emergency telephone numbers to all field teams to contact in the event of an environmental incident: State or Tribal department of environmental quality or protection, U.S. Coast Guard, and the U.S. EPA Regional Office. In the event of a major environmental incident, the National Response Center may need to be notified at 1-800-424-8802.

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NOTES

SECTION 3
BASE LOCATION ACTIVITIES

Donald J. Klemm[1], Brian H. Hill[2], Frank H. McCormick[3],
David V. Peck[4], and Marlys Cappaert[5]

Field teams conduct a number of activities at a *base location* before and after visiting each stream site. These activities are generally conducted on the same day as the sampling visit. These activities are required to ensure that the field teams know where they are going, that access to the stream site is possible and permissible, that all the necessary equipment and supplies are in good order to complete the sampling effort, and that samples are packaged and shipped correctly and promptly. Modifications to base location procedures from those described in the previous EMAP-SW field operations manual for wadeable streams (Klemm et al. 1998) and during the course of EMAP-W are summarized in Appendix B.

In some situations, field teams may have personnel available who are certified to ship preserved biological samples that constitute dangerous goods. Such samples must be transported and presented for shipment in accordance with State, Federal, and international regulations. Because of the large geographic area being sampled for EMAP-W, it is critical to minimize the potential for transferring exotic or nuisance species of plants and animals or waterborne pathogens such as salmonid whirling disease.

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

2 U.S. EPA, National Health and Ecological Effects Research Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804.

3 USDA Forest Service, Olympia Forestry Sciences Laboratory, Pacific Northwest Research Station, 3625 93rd Avenue SE, Olympia, WA 98512.

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

5 Computer Sciences Corporation, c/o U.S. EPA, 200 SW 35th St., Corvallis, OR 97333

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

3.1 ACTIVITIES BEFORE EACH STREAM VISIT

As part of the site selection process, each candidate site was evaluated to determine if it met the criteria for inclusion as part of the target population (see Section 1.2 or Stoddard et al. 2005) and could be accessed for sampling (including an initial contact of landowners to obtain permission). The initial evaluation process was often followed by a site reconnaissance visit prior to the start of sampling activities. For some sites, the evaluation and reconnaissance were conducted by a person who was also either a field team leader or member. For other sites, the evaluation and reconnaissance was conducted by someone other than a field tem member. For each site scheduled for sampling, the locational and access-related information acquired during the evaluation and reconnaissance process is provided to the field team leader as part of a *site dossier*.

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

3.1.1 Confirming Site Access

Before visiting a stream, review the contents of the specific stream dossier and identify any revisions to the information contained in the dossier since the evaluation was conducted or the reconnaissance visit. Contact the landowner(s) listed in the dossier well in advance of the scheduled visit to confirm permission to sample that was obtained earlier.

3.1.2 Daily Sampling Itinerary

Based upon the sampling schedule provided to each team, team leaders are responsible for developing daily itineraries. The team leader reviews each stream dossier to

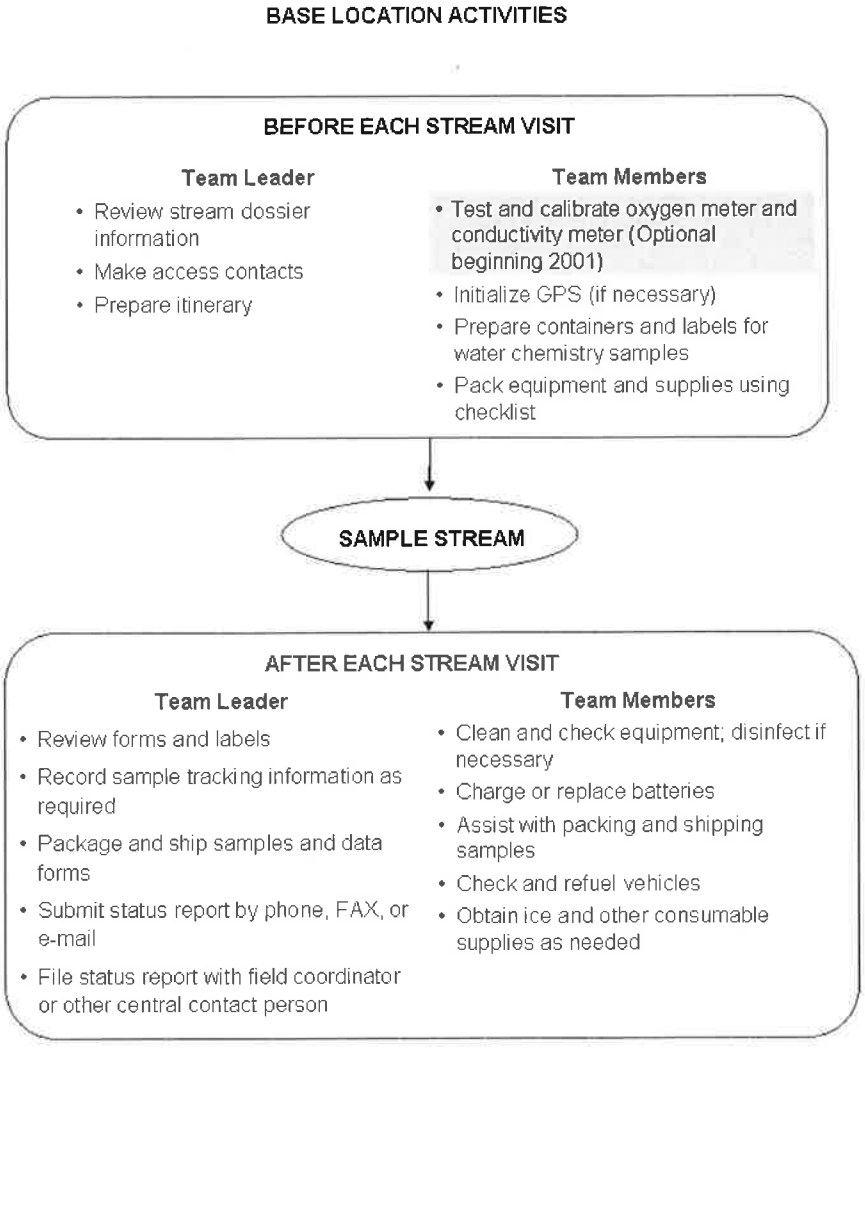


Figure 3-1. Base location activities. Shaded activities are optional.

ensure that it contains the appropriate maps, contact information, copies of permission letters, and access instructions. The team leader also determines the best access routes, confirms permission with landowners or local contacts, confirms lodging plans for the upcoming evening, and coordinates rendezvous locations with individuals who must meet with a field team prior to accessing a site. Use this information to develop an itinerary for the stream. Include anticipated departure time, routes of travel, location of any intermediate stops (e.g., to drop off samples, pick up supplies, etc.) and estimated time of arrival at the final destination after completing the stream visit. Provide the itinerary (and any changes that occur due to unforeseen circumstances) to the field coordinator or other central contact person identified for the specific field study. Failure to adhere to the reported itinerary can result in the initiation of expensive search and rescue procedures and disruption of carefully planned schedules. In addition, carry emergency medical and personal information for each team member to the field, possibly in the form of a safety logbook that remains in the vehicle (see Section 2).

3.1.3 Instrument Inspections and Performance Tests

Some instruments require testing and/or calibration prior to departure for the stream site. Field instruments include a global positioning system (GPS) receiver and a current velocity meter. Optional instruments (since 2001) include a conductivity meter and a dissolved oxygen (DO) meter. Backup instruments should be available if instruments fail the performance tests or calibrations described in the following subsections.

3.1.3.1 Global Positioning System Receiver–

Specific performance checks vary among different brands of GPS receivers. Follow the instructions in the receiver's operating manual to make sure the unit is functioning properly. Turn on the receiver and check the batteries. Replace batteries immediately if a battery warning is displayed. Make sure extra batteries are stored with the receiver and are available in the field. Follow the manufacturer's instructions for setting up the receiver when it becomes necessary (e.g., before first use, after replacing batteries, or if a new positional reference is required).

3.1.3.2 Dissolved Oxygen Meter–

As an initial performance test before use each year, test DO meters for accuracy against the Winkler titration method (individual test kits for field use are acceptable). In addition, inspect and test the dissolved oxygen meters periodically during the course of field sampling operations. At a minimum, check the instruments before the field season starts

and again after field sampling has been completed. The inspection and testing procedure, based on the use of a Yellow Springs Instruments (YSI) Model 53 oxygen meter, is summarized in Figure 3-2. Some modification to the procedure may be necessary for other models or types of dissolved oxygen meters. The procedure to use for newer DO meters (e.g., the YSI Model 85 or 95), is presented in Table 3-1.

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

After inspecting the meter and probe, attempt to calibrate it using the procedure presented in Table 3-1 (for YSI Models 85 or 95), or by following the instructions in the instrument operating manual. Do not record the calibration information obtained during the performance test. The meter is calibrated again at each stream site. If the meter cannot be successfully calibrated, check the temperature probe reading against a thermometer and/or replace the membrane, probe, or meter (if spare units are available). After the test, turn the meter off, and store the probe according to the manufacturer's instructions.

3.1.3.3 Conductivity Meters–

Follow the operating manual provided with the meter to check the batteries, the electronics, and to inspect the probe. New probes or probes that have been stored dry may require conditioning before use.

The operation of the conductivity meter is checked periodically at a base location using a standard solution of known conductivity. Prepare a quality control check sample (QCCS) as described in Table 3-2. The QCCS can be prepared as either of two dilutions of the stock standard, depending on the theoretical conductivity desired based on the anticipated range of conductivities in the field. A 1:100 dilution of the stock provides a QCCS with a conductivity of 75.3 µS/cm at 25 °C (Metcalf and Peck 1993). A 1:200 dilution results in a QCCS with a conductivity of 37.8 µS/cm at 25 °C (Peck and Metcalf 1991). Prepare a fresh lot of the QCCS every two weeks from the stock standard solution. For higher conductivity systems, a 0.01 N potassium chloride solution can be used as a QCCS (theoretical value = 1,413 µS/cm at 25 °C).

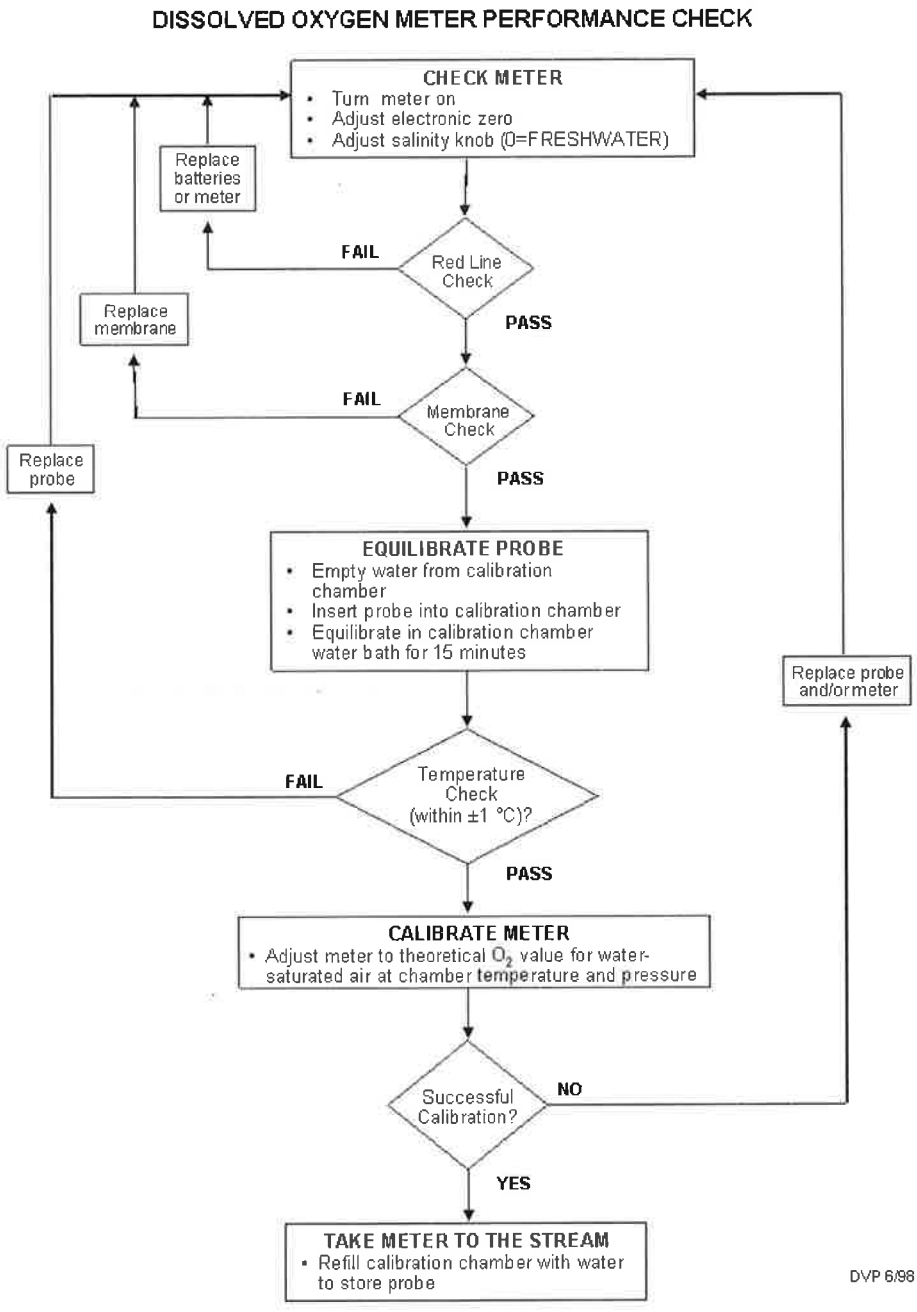


Figure 3-2. Performance test procedure for a dissolved oxygen meter. Based on a YSI Model 53 meter or comparable instrument.

TABLE 3-1. CHECKING THE CALIBRATION OF A DISSOLVED OXYGEN METER[a]

1. If a combination oxygen/temperature/conductivity meter is used, check the oxygen probe before checking the conductivity probe.

2. Periodically, check the temperature probe of the meter against a field thermometer. This can be done in a bucket of water at a base location or at a stream site. The displayed temperature should be within ± 1 °C of the thermometer reading.

3. At each location, obtain the approximate local altitude from a topographic map or other source (e.g., a local airport).

4. Inspect the DO probe membrane for wrinkles, cracks, bubbles, etc. Replace the membrane cap assembly if necessary.

5. Check the calibration chamber and fill it with cold tap water to dampen the sponge. Drain the chamber and insert the probe into the chamber.

6. Turn the meter on and make sure the meter passes all the internal electronics checks.

7. Press the MODE key until the dissolved oxygen reading inside the chamber is displayed in mg/L. Allow approximately 15 minutes for the readings to stabilize (i.e., a change of < 0.02 mg/L over a 1-minute period).

8. Press the UP ARROW and DOWN ARROW keys simultaneously to enter calibration mode.

9. Use the UP ARROW or DOWN ARROW key to enter the local altitude [to the nearest 100 feet (e.g., *15* equals 1500 ft)]. After the correct altitude is displayed, press the ENTER button.

10. In the lower part of the display, *CAL* should appear along with the theoretical value based on temperature and altitude.

11. Once the actual value displayed is stable, compare the actual and theoretical values. They should agree ± 0.5 mg/L. If not, check the temperature probe against a thermometer (Step 1), or install a new membrane cap assembly, then repeat the calibration procedure.

[a] For use with YSI Models 85 and 95. Modified from YSI Incorporated. 1986. *Model 85 handheld oxygen, conductivity, salinity, and temperature system operations manual.* YSI Incorporated, Yellow Springs, OH.

TABLE 3-2. STOCK SOLUTIONS, USES, AND INSTRUCTIONS FOR PREPARATION

SOLUTION	USE	PREPARATION
Bleach (10%)	Soaking solution to disinfect gear from spores of whirling disease, New Zealand mud snails, and amphibian chytrid fungus	Dilute 400 mL (13 oz) household chlorine bleach solution to 4 L (1 gal) with tap water.
Bleach (50%)	Wipe-on or spray-on solution to disinfect gear from spores of whirling disease, New Zealand mud snails, and amphibian chytrid fungus	Dilute 2-L (64 oz) household chlorine bleach solution to 4-L (1 gal) with tap water
Sparquat 256® germicidal disinfectant	Alternative soaking solution to disinfect gear from spores of whirling disease, New Zealand mud snails, and amphibian chytrid fungus	Dilute 120-180 mL (4-6 oz) to 4 L (1 gal) with tap water
Formula 409® cleaner (antibacterial version)	Alternative soaking solution to disinfect gear from spores of whirling disease, New Zealand mud snails, and amphibian chytrid fungus	Dilute 2-L (64 oz) to 4-L (1 gal) with tap water
Conductivity Standard Stock Solution[a] (optional)	To prepare conductivity quality control check sample solution	Dissolve 3.4022 g KH_2PO_4 and 3.5490 g Na_2HPO_4 (analytical grade; dried at 120 °C for 3 h and stored desiccated) in 1000.0 g (1.0018 L at 20 °C, 1.0029 L at 25 °C) reagent water.
Quality Control Check Sample for conductivity (optional)	To check operation of conductivity meter	1:100 dilution of standard stock solution with reagent water (theoretical conductivity = 75.3 µS/cm at 25 °C)[a] 1:200 dilution of standard stock solution with reagent water (theoretical conductivity = 37.6 µS/cm at 25 °C)[b]
Formalin, borax buffered[c] (pH 7-8)	Preservative for fish specimens and periphyton samples	Add 400 g borax detergent (e.g., *Twenty Mule Team®*) to each 20-L container of 100% formalin. Test with pH paper.
Ethanol (95%)	Preservative for benthic macroinvertebrate samples.	None.

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

If a YSI Model 85 meter is being used, check the performance of the probe by following the procedure presented in Table 3-3. Make sure the correct mode (temperature-compensated conductivity) is used for the check. Compare the displayed value of the QCCS to the theoretical value of the QCCS at 25 °C (75.3 µS/cm or 37.8 µS/cm).

If another model of a conductivity meter is used, refer to the procedure presented in Table 3-4. If the meter cannot display temperature-compensated conductivity, develop a table showing theoretical values of the QCCS solution at different temperatures.

3.1.3.4 Current Velocity Meters--

Field teams may be using one of three types of current velocity meters, a photo-optical impeller type meter (e.g., Swoffer Model 2100) a vertical axis meter (e.g., Price type AA), or an electromagnetic type meter (e.g., Marsh McBirney Model 201D). General guidelines regarding performance checks and inspection of current meters are presented in Table 3-5. Consult the operating manual for the specific meter and modify this information as necessary.

3.1.4 Preparation of Equipment and Supplies

Check the inventory of equipment and supplies prior to departure using the stream visit checklists presented in Appendix A. Pack meters, probes, and sampling gear to minimize physical shock and vibration during transport. Prepare stock preservative solutions as described in Table 3-2 if necessary. Follow Department of Transportation (DOT) and the Occupational Safety and Health Administration (OSHA) regulations for handling and transporting hazardous materials such as formalin and ethanol. Regulations pertaining to formalin are in the Code of Federal Regulations (CFR, specifically 29 CFR 1910.1048). These requirements should be summarized for all hazardous materials being used for the project and provided to field personnel. Transport formalin and ethanol in appropriate containers with absorbent material.

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

TABLE 3-3. PERFORMANCE CHECK OF NEWER CONDUCTIVITY METERS[a]

1. If using a combination DO/conductivity meter (e.g., the YSI Model 85), check the conductivity probe after completing the calibration check for the DO probe.

2. Inspect the probe for deposits or fouling.

3. Turn the meter on and make sure all internal electronics checks are completed successfully.
4. Use the *MODE* key to display "temperature compensated" conductivity (The "°C" symbol on the display will be flashing).

5. Swirl the conductivity probe for 3-5 seconds in a 250-mL bottle containing the daily QCCS solution labeled *RINSE*.

6. Transfer the probe from the *RINSE* bottle to a second 250-mL bottle of QCCS labeled *TEST*. Let stabilize for 20 seconds.

7. If the measured value of the QCCS is within ±10% or ±10 µS/cm of the theoretical value (whichever is greater at the theoretical value), rinse the probe in deionized water. Store as described in the operating manual and package the meter for transport to the stream site.
   If the measured value of the QCCS is not within ±10% or ±10 uS/cm of theoretical value, repeat Steps 5 through 7.

8. If the value is still unacceptable, replace the QCCS in both the *RINSE* and *TEST* bottles and repeat the measurement process.
   If the measured value is still not acceptable, clean the conductivity probe as described in the manual, check the batteries, soak in deionized water for 24 hours, and repeat Steps 1 through 7.
   If the measured value is still unacceptable, replace the meter.

[a] For use with YSI Models 85 and 95. Modified from YSI Incorporated. 1986. *Model 85 handheld oxygen, conductivity, salinity, and temperature system operations manual.* YSI Incorporated, Yellow Springs, OH.

TABLE 3-4. PERFORMANCE CHECK OF OLDER CONDUCTIVITY METERS[a]

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

2. Swirl the conductivity probe for 3-5 seconds in a 250-mL bottle containing the daily QCCS solution labeled *RINSE*.

3. Transfer the probe from the *RINSE* bottle to a second 250-mL bottle of QCCS labeled *TEST*. Let stabilize for 20 seconds.

4. If the measured value of the QCCS is within ±10% or ±10 µS/cm of the theoretical value (whichever is greater at the theoretical value), rinse the probe in deionized water. Store as described in the operating manual and package the meter for transport to the stream site.

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

   If the value is still unacceptable, replace the QCCS in both the *RINSE* and *TEST* bottles and repeat the measurement process.

   If the measured value is still not acceptable, clean the conductivity probe as described in the manual, check the batteries, soak in deionized water for 24 hours, and repeat Steps 1 through 3.

   If the measured value is still unacceptable, replace the meter.

[a] For use with older models of conductivity meters (e.g., YSI Model 33 S-C-T).

TABLE 3-5. GENERAL PERFORMANCE CHECKS FOR CURRENT VELOCITY METERS

Photoelectric Impeller Meters (e.g., Swoffer Model 2100)
• Check that the calibration adjustment cover screws are tightly fitted on the display case.
• Periodically check the condition of the connector fitting between the display unit and the sensor.
• Connect the sensor to the display unit and check the calibration value stored in memory. If this value is less than the correct value for the display unit-sensor rotor combination, replace the batteries.
• Periodically perform a spin test of the rotor assembly, following the instructions in the meter's operating manual. A displayed count value of 300 or greater is indicative of satisfactory performance at low current velocities.
• If a buzzing sound occurs when the rotor assembly is spun by hand, or if the shaft shows visible wear, replace the rotor assembly.
• Periodically examine the thrust-bearing nut on the rotor assembly. If a "cup" begins to form on the bottom surface of the nut, it should be replaced.
Vertical-axis Meters (e.g., Price Type AA; from Smoot and Novak 1968)
• Inspect the bucket and wheel hub assembly, yoke, cups, tailpiece, and the pivot point each day before use.
• Inspect the bearings and check the contact chamber for proper adjustment.
• Periodically conduct a spin test of the meter. The minimum spin time is 1.5 minutes, while the recommended time is between 3 and 4 minutes.
Electromagnetic Meters (e.g., Marsh-McBirney Model 2010)
• Check the meter calibration daily as part of morning routine. Calibration value should be 2.00 $\pm$ 0.05.
• Once per week, check the zero value using a bucket of quiescent water. Place the probe in the bucket and allow to sit for 30 minutes with no disturbance. The velocity value obtained should be 0.0 $\pm$ 0.1. Adjust the meter zero if the value is outside this range.

Sample containers for water chemistry can be labeled before departing from the base location. Figure 3-3 illustrates the preprinted labels. Prepare a set of three water chemistry sample containers all having the same ID number (one for the 4-L cubitainer and two for the 60-mL syringes) and labels completed with the appropriate information (described in Section 5). After labeling, place the syringes in their plastic container, and place the cubitainer and beakers in a clean self-sealing plastic bag to prevent contamination. ***Do not*** *prepare labels for other types of sample containers* (e.g., periphyton, benthos, vertebrates) before reaching the stream site. Problems in sample tracking will result if containers are labeled and then are not used at a stream.

3.2 ACTIVITIES AFTER EACH STREAM VISIT

Upon reaching a lodging location after sampling a stream, review all completed data forms and sample labels for accuracy, completeness, and legibility. Fill in missing or illegible information on forms or labels as accurately as possible. The team leader initials all data forms after review. The other team members should inspect and clean sampling equipment, check the inventory of supplies, make a final inspection of samples, and prepare samples for shipment. Other activities include shipping samples, submitting sampling status and tracking information to the EMAP-SW information management staff at the EPA Western Ecology Division (WED) in Corvallis, and communicating with the field coordinator or other central contact person.

3.2.1 Equipment Cleaning

All equipment and gear used at a stream site must be cleaned and disinfected between sites to reduce the risk of transferring nuisance species and pathogens. Three organisms of primary concern in the western U.S. are *Myxobolus cerebralis* (the sporozoan parasite that causes salmonid whirling disease), *Potamopyrgus antipodarum* (the nonnative New Zealand mudsnail), and *Batrachochytrium dendrobatidis* (a chytrid fungus that threatens amphibian populations).

The life cycle of *M. cerebralis* parasite includes both a resistant *myxospore* and a more free-floating, highly infective form (the *triactinomyxon).* Myxospores can remain dormant for 30 years or more, while the triactinomyxons reside at very high densities and concentrations in fish and fish parts. The New Zealand mud snail has an extremely high reproductive rate (it can reproduce asexually), can survive for extended periods in damp

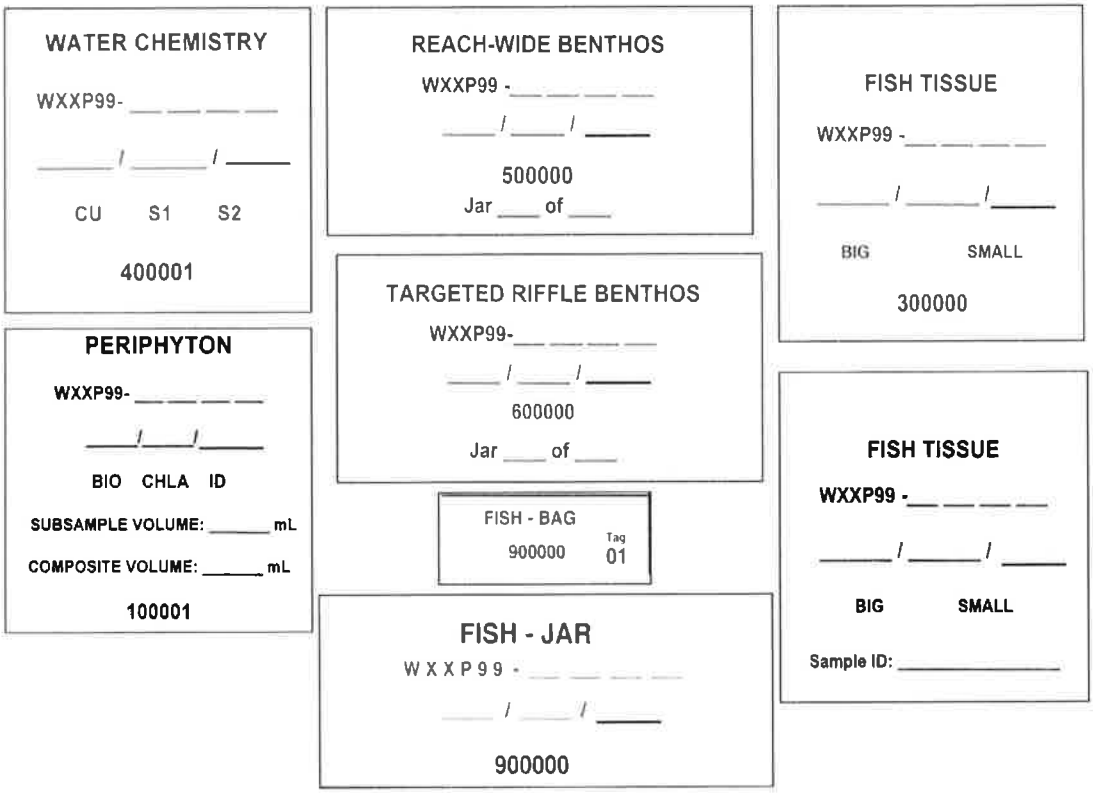


Figure 3-3. Sample container labels.

conditions, and has no natural predators (it passes through the digestive tract of fishes unharmed). Little is known about the amphibian chytrid fungus, but it may be associated with recent die-offs of several endangered frogs and toads, including the boreal toad in Colorado.

Field teams should be provided with the latest information (as part of the site dossier) regarding those streams, drainages, etc. that are known or suspected to be infested with whirling disease, New Zealand mudsnails, or amphibian chytrid fungus. This information is available from State or Federal fishery biologists or pathologists, or from State game and fish agency websites.

Equipment cleaning procedures are given in Table 3-6. These are based on guidelines established by the National Park Service (*http://www.nps.gov/archive/romo/downloads/aquatic_guidelines/aquatic_guidelines2006.pdf*), and are intended to work to

protect against all three organisms. Additional online sources of information regarding whirling disease, mud snails, or chytrid fungus, including information on cleaning and disinfecting gear, include the Whirling Disease Foundation (*www.whirling-disease.org*), the USDA Forest Service (*Preventing Accidental Introductions of Freshwater Invasive Species*, available from *http://www.fs.fed.us/invasivespecies/documents/Aquatic_is_prevention.pdf*), and the California Department of Fish and Game (Hosea and Finlayson 2005). General information about freshwater invasive species is available from the U.S. Geological Survey Nonindigenous Aquatic Species website (*http://nas.er.usgs.gov*), the *Protect Your Waters* website (*http://www.protectyourwaters.net/hitchhikers*) that is co-sponsored by the U.S. Fish and Wildlife Service, and the Sea Grant Program (*http://www.sgnis.org*). Note that many organizations now recommend ***against*** using felt-soled wading boots in affected areas due to the difficulty in removing myxospores and mudsnails. Handle and dispose of disinfectant solutions properly, and take care to avoid damage to lawns or other property.

3.2.2 Sample Packing, Shipment, and Tracking

Pack and ship samples from each stream visit as soon as possible after collection, normally the day following a stream visit. Field teams are provided with specific information for the shipping destinations, contact persons, and the required shipping schedule for each type of sample. Record sample tracking information (including sample types, sample ID numbers, and other field-related information that is required by the laboratory to conduct analyses and associate results to a specific sample and stream site) during the packing process. After each shipment, file a status report with the EMAP-SW information management staff at WED and with the field coordinator.

3.2.2.1 Unpreserved Samples–

Unpreserved samples include water chemistry, periphyton (including the preserved ID sample), and fish tissue samples. For unpreserved samples, record information onto a tracking form as shown in Figure 3-4. Fill out the tracking form for all samples taken. Use the standard codes provided on the form to record the type of sample and its condition. Record all *subsample types* (cubitainer and syringe for water chemistry, ID, chlorophyll, and biomass for periphyton) in the comments field. For fish tissue contaminants samples, record the common name, and for the *small* tissue sample, record the number of individuals in the comments field (this information is needed by the laboratory, and is not available on the sample label). Prepare *one additional copy* of the form (a photocopy is acceptable). Retain

TABLE 3-6. EQUIPMENT CARE AFTER EACH STREAM VISIT

1. General cleaning for biological contaminants (e.g., plant and animal material).
   - Prior to departing a stream, drain all water from live wells and buckets used to hold and process fish.
   - Inspect sampling gear and waders, boots, etc. for evidence of mud, snails, plant fragments, algae, animal remains, or other debris. Remove using brushes, screwdrivers, or other tools.
   - At the base location, inspect and rinse seines, dip nets, kick nets, waders, and boots with water and dry. Use one of the procedures below to disinfect gear if necessary.

2. Additional precautions to prevent transfer of Whirling Disease spores, New Zealand mudsnails, and amphibian chytrid fungus.[a]
   - Before visiting the stream, consult the site dossier and determine if the stream is in an area where whirling disease, New Zealand mud snails, or chytrid fungus are known to exist. Contact the local State fishery biologist to confirm the existence or absence of these organisms.
   - If the stream is listed as "positive" for any of the organisms, or no information is available, *avoid using felt-soled wading boots*, and, after sampling, disinfect **all** fish and benthos sampling gear and other equipment that came into contact with water or sediments (i.e., waders, boots, etc.) by one of the following procedures:

   Option A:
   1. Soak gear in a 10% household bleach solution for at least 10 minutes, **or** wipe or spray on a 50% household bleach solution and let stand for 5 minutes
   2. Rinse with clean water (**do not** use stream water), and remove any remaining debris
   3. Place gear in a freezer overnight **or** soak in a 50% solution of Formula 409® household cleaner (antibacterial version) for at least 10 minutes **or** soak gear in 120 °F (49 °C) water for at least 1 minute.
   4. Dry gear in direct sunlight (at least 84 °F) for at least 4 hours.

   Option B:
   1. Soak gear in a solution of Sparquat® (4-6 oz. per gallon of water) for at least 10 minutes (Sparquat is especially effective at inactivating whirling disease spores).
   2. Place gear in a freezer overnight **or** soak gear in 120 °F (49 °C) water for at least 1 minute.
   3. Dry gear in direct sunlight (at least 84 °F) for at least 4 hours.

3. Clean and dry other equipment prior to storage.
   - Rinse chlorophyll filtration chamber three times with distilled water after each use.
   - Rinse periphyton sampling equipment with tap water at the base location.
   - Rinse coolers with water to clean off any dirt or debris on the outside and inside.
   - Make sure conductivity meter probes are rinsed with deionized water and are stored moist.
   - Rinse all beakers used to collect water chemistry samples three times with deionized water to prevent contamination of the next stream sample. Place the beakers in a 1-gallon sealable plastic bag with a cubitainer for use at the next stream.

[a] Based on guidelines established for Rocky Mountain National Park (available from *http://www.nps.gov/archive/romo/downloads/aquatic_guidelines/aquatic_guidelines2006.pdf*).

(Continued)

TABLE 3-6 (Continued)

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

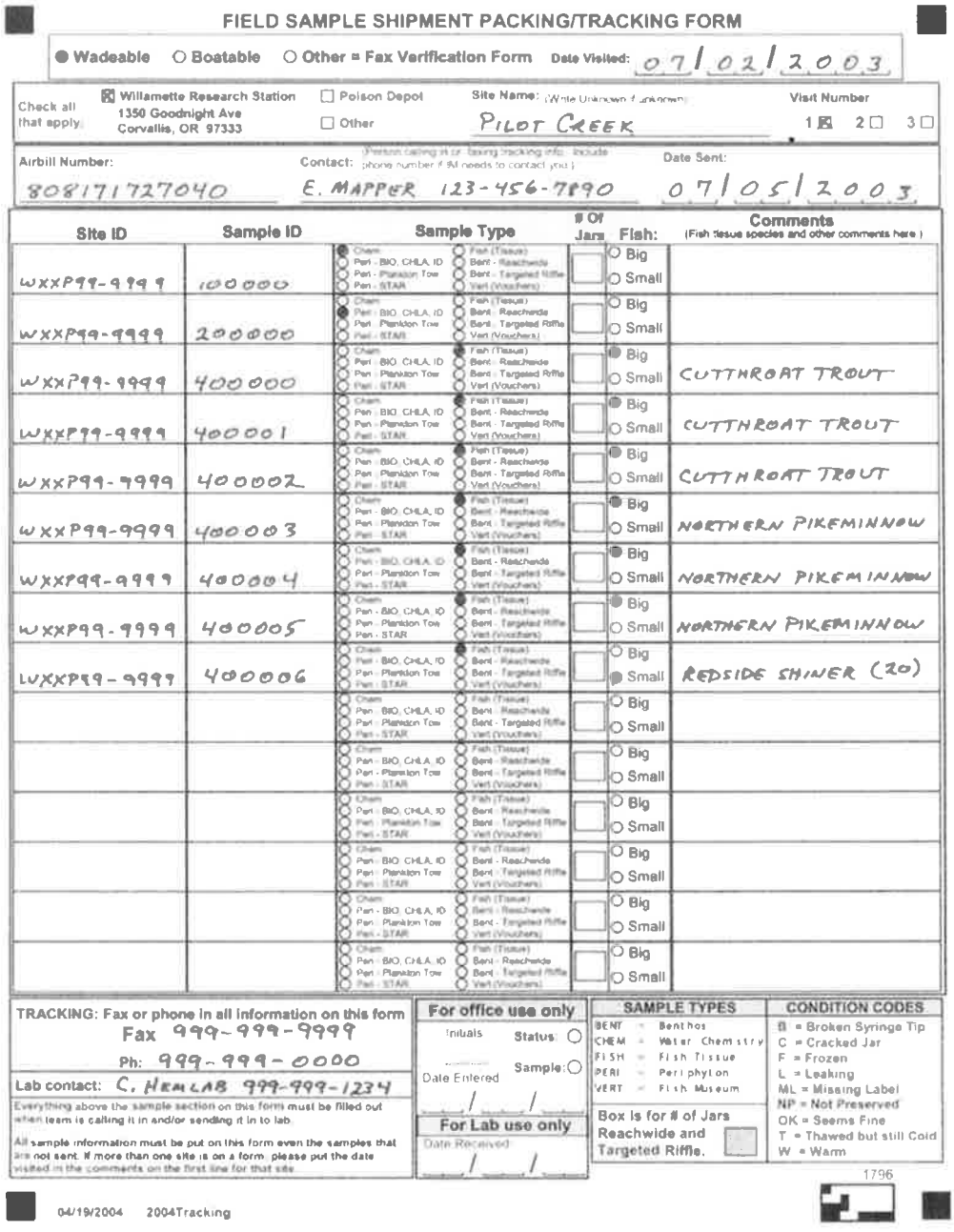
5. Inventory equipment and supply needs and relay orders to the Field Coordinator.

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

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

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

9. Review the field data forms for the site. Make corrections and completions where possible, and initial each form after review.



FIELD SAMPLE SHIPMENT PACKING/TRACKING FORM

● Wadeable ○ Boatable ○ Other = Fax Verification Form Date Visited: 07/02/2003

Check all that apply: ☒ Willamette Research Station, 1350 Goodnight Ave, Corvallis, OR 97333 ☐ Poison Depot ☐ Other

Site Name: PILOT CREEK Visit Number: 1☒ 2☐ 3☐

Airbill Number: 808171727040 Contact: E. MAPPER 123-456-7890 Date Sent: 07/05/2003

Site ID	Sample ID	Sample Type	# Of Jars	Fish:	Comments (Fish tissue species and other comments here.)
WXXP99-9999	100000	Perl - BIO, CHLA, ID		○ Big ○ Small	
WXXP99-9999	200000	Perl - BIO, CHLA, ID		○ Big ○ Small	
WXXP99-9999	400000	Fish (Tissue)		● Big ○ Small	CUTTHROAT TROUT
WXXP99-9999	400001	Fish (Tissue)		● Big ○ Small	CUTTHROAT TROUT
WXXP99-9999	400002	Fish (Tissue)		● Big ○ Small	CUTTHROAT TROUT
WXXP99-9999	400003	Fish (Tissue)		● Big ○ Small	NORTHERN PIKEMINNOW
WXXP99-9999	400004	Fish (Tissue)		● Big ○ Small	NORTHERN PIKEMINNOW
WXXP99-9999	400005	Fish (Tissue)		● Big ○ Small	NORTHERN PIKEMINNOW
WXXP99-9999	400006	Fish (Tissue)		○ Big ● Small	REDSIDE SHINER (20)
				○ Big ○ Small	
				○ Big ○ Small	
				○ Big ○ Small	
				○ Big ○ Small	
				○ Big ○ Small	
				○ Big ○ Small	

TRACKING: Fax or phone in all information on this form
Fax 999-999-9999
Ph: 999-999-0000
Lab contact: C. HEMLAB 999-999-1234

Everything above the sample section on this form must be filled out when team is calling it in and/or sending it in to lab.

All sample information must be put on this form even the samples that are not sent. If more than one site is on a form, please put the date visited in the comments on the first line for that site.

For office use only
Initials Status: ○
Sample: ○
Date Entered / /

For Lab use only
Date Received / /

SAMPLE TYPES
BENT = Benthos
CHEM = Water Chemistry
FISH = Fish Tissue
PERI = Periphyton
VERT = Fish Museum

Box is for # of Jars Reachwide and Targeted Riffle.

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

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Figure 3-4. Sample tracking form for unpreserved samples.

the original copy to prepare the status report for the site (Section 3.3), and then include it as part of the data forms packet for the site. Include the copy as a packing list in the shipment. Ship unpreserved samples to the analytical laboratory facility (the Willamette Research Station [WRS] laboratory at WED was used for EMAP-W).

General guidelines for packing and shipping the various types of unpreserved samples described in this manual are presented in Table 3-7. Use ice substitute packs whenever possible to avoid leakage due to melting ice. When shipping samples using ice, use fresh ice. Use block ice when available, sealed in large plastic bags. If block ice is not available, contain the ice in several self-sealing plastic bags. Label each bag of ice as *ICE* with an indelible marker to prevent any leakage of meltwater from being misidentified by couriers as a possible hazardous material spill. If ice substitute packs are used, place each pack into a self-sealing plastic bag before use.

Ship water chemistry samples as soon as possible after collection in order to meet holding time requirements for some laboratory analyses (especially nutrients). To ship water chemistry samples, use a large (30-gallon) plastic bag as a liner in an insulated shipping container (e.g., a plastic or metal cooler). Use clear tape to completely cover each sample label (cubitainer and syringes) to prevent damage from water or condensation during shipment. Put the syringes into a separate plastic container for shipment, then put the container and cubitainer into a second large plastic bag and close. Place the bag containing the samples inside the plastic bag lining the shipping container. Place bags of ice around the bag of samples, but inside the plastic bag lining the shipping container. Be sure to use sufficient quantities of ice to ensure samples will remain cold until arrival at the laboratory. *Typically, the total weight of each shipping container (samples plus ice) should be between **40 and 50 pounds** (more for shipments from hot locations).* Alternatively, at least one-half of the total weight of each shipping container should be ice.

Then close the outer plastic bag. Insert the copy of the completed tracking form (Figure 3-4) into a self-sealing plastic bag, and tape the bag to the inside of the lid, then close the container. Seal the container with shipping tape and affix any required shipping-related labels to the outside of the container. Attach an adhesive plastic sleeve to the lid of the container and insert any required shipping forms.

Samples requiring freezing (Table 3-7) are usually shipped with the refrigerated samples, but these may be stored in the field in a portable freezer or on dry ice for a short

TABLE 3-7. GENERAL GUIDELINES FOR PACKING AND SHIPPING UNPRESERVED SAMPLES

Sample Type (container)	Guidelines
Samples requiring refrigeration (4 °C)	
Water Chemistry (4-L cubitainer and 60-mL syringes)	Ship on day of collection or within 24 hr by overnight courier. Use fresh ice in labeled plastic bags for shipping. *Use enough ice so that total weight of each shipping container is* ***at least 40 lbs****., or the weight of ice is equal to or greater than the weight of the samples.* Line each shipping container with a large plastic bag. Place syringes in a plastic container. Place syringe container and cubitainer inside of a second plastic bag. Cover labels completely with clear tape. The cubitainer and syringes should have same sample ID number assigned. Confirm the sample ID assigned on the labels matches the ID number recorded on the field collection form and the sample tracking form.
Samples requiring freezing (-20 °C) within 24 hours of collection	
Periphyton chlorophyll (filter in aluminum foil)	If samples cannot be kept frozen in the field, ship on day of collection or within 24 h by overnight courier. Cover the label completely with clear tape. Protect samples from meltwater if ice is used by double bagging ice and placing samples in a plastic container. Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form (or other sample tracking report). If dry ice is used to transport or ship samples, special shipping containers, outside labeling, and shipping forms may be required.
Periphyton biomass (filter in aluminum foil)	
Periphyton activity (50-mL centrifuge tube)	
Fish Tissue (aluminum foil; two 30-gal plastic bags)	If samples cannot be kept frozen in the field, ship on day of collection or within 24 h by overnight courier. Cover labels completely with clear tape. Label on each bag should have identical Sample ID number assigned. Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form (or other sample tracking report). Protect samples from meltwater if ice is used by double bagging ice. Special shipping containers, outside labeling, and shipping forms may be required for shipments containing dry ice.

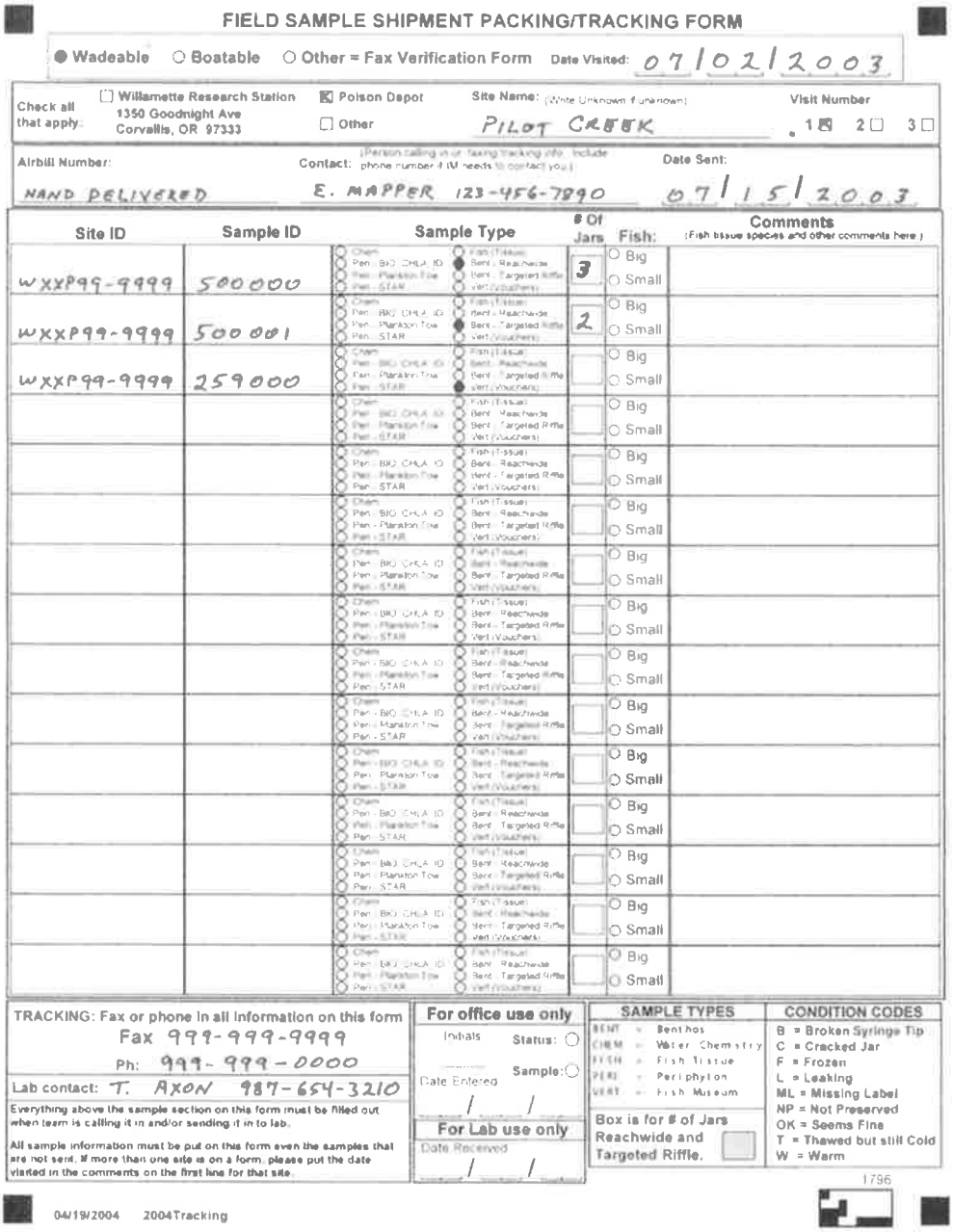
period (e.g., one week) if necessary. If only ice (or ice substitute packs) is available for field storage, ship the samples to the laboratory as soon as possible after collection, using fresh ice (or ice substitute packs) to keep them as cold as possible. When using ice, double bag the ice and tape the last bag shut to prevent contamination of samples by melting ice. If ice substitute packs are used, place each pack into a self-sealing plastic bag. If possible, place samples into a sealed plastic container to protect them from meltwater. Dry ice may also be used for shipping. Note that dry ice is considered a hazardous material, and requires special shipping containers, shipping labels, and shipping forms for ground or air transport. If dry ice is used, the requirements and directions for packing and shipping samples should be provided to each field team.

3.2.2.2 Preserved Samples–

Transport samples that are preserved in 10% buffered formalin (fish voucher specimens) or ethanol (benthic macroinvertebrate samples) in appropriate containers surrounded with some type of acceptable absorbent material (e.g., vermiculite). Preserved vertebrate voucher samples should be packaged as dangerous goods, but may be shipped in small quantities as a nonregulated chemical (but confirm this with your shipping service).

Complete a separate tracking form as shown in Figure 3-5 for all preserved samples (fish voucher specimens and benthos). These samples are likely to be retained by the field team and periodically transported to intermediate *storage depots*, where they will accumulate prior to shipment or delivery to the appropriate support laboratories. Again, you will need to make a copy of the completed form for each site. Retain the original copy to prepare the status report for the site (Section 3.3), and then include it as part of the data forms packet for the site. Include the copy as a packing list when you drop off samples at the storage depot.

Guidelines for packing, labeling, transporting, and shipping samples containing formalin or ethanol are presented in Table 3-8. Note these are guidelines only and do not necessarily conform to the latest regulations, so *confirm any requirements associated with dangerous goods shipments with your shipping service before offering any preserved samples for shipment.* It may be necessary to provide additional guidance to each field team. In order to offer dangerous goods for shipment, a person must be certified through participation in a training course to prepare dangerous goods for shipment. Most of this training deals with packaging, forms and labels that must be used. Dangerous goods shipments must always be presented to the shipper directly as either a pick-up by a driver or a drop-off at a shipping facility.



FIELD SAMPLE SHIPMENT PACKING/TRACKING FORM

● Wadeable ○ Boatable ○ Other = Fax Verification Form Date Visited: 07/02/2003

Check all that apply: ☐ Willamette Research Station 1350 Goodnight Ave Corvallis, OR 97333 ☒ Poison Depot ☐ Other

Site Name: (Write Unknown if unknown) PILOT CREEK

Visit Number 1☒ 2☐ 3☐

Airbill Number: HAND DELIVERED

Contact: (Person calling in or faxing tracking info. Include phone number if IM needs to contact you.) E. MAPPER 123-456-7890

Date Sent: 07/15/2003

Site ID	Sample ID	Sample Type	# Of Jars	Fish:	Comments (Fish tissue species and other comments here.)
WXXP99-9999	500000	Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)	3	○ Big ○ Small	
WXXP99-9999	500001	Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)	2	○ Big ○ Small	
WXXP99-9999	259000	Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)		○ Big ○ Small	
		Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)		○ Big ○ Small	
		Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)		○ Big ○ Small	
		Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)		○ Big ○ Small	
		Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)		○ Big ○ Small	
		Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)		○ Big ○ Small	
		Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)		○ Big ○ Small	
		Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)		○ Big ○ Small	
		Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)		○ Big ○ Small	
		Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)		○ Big ○ Small	
		Chem; Per - BIO CHLA ID; Per - Plankton Tow; Per - STAR; Fish (Tissue); Bent - Reachwide; Bent - Targeted Riffle; Vert (Vouchers)		○ Big ○ Small	

TRACKING: Fax or phone in all information on this form
Fax 999-999-9999
Ph: 999-999-0000
Lab contact: T. AXON 987-654-3210
Everything above the sample section on this form must be filled out when team is calling it in and/or sending it in to lab.
All sample information must be put on this form even the samples that are not sent. If more than one site is on a form, please put the date visited in the comments on the first line for that site.

For office use only
Initials Status: ○
Sample: ○
Date Entered / /

For Lab use only
Date Received / /

SAMPLE TYPES
BENT = Benthos
CHEM = Water Chemistry
FISH = Fish Tissue
PERI = Periphyton
VERT = Fish Museum
Box is for # of Jars Reachwide and Targeted Riffle.

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

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Figure 3-5. Sample tracking form for preserved samples.

TABLE 3-8. GENERAL GUIDELINES FOR PACKING AND SHIPPING PRESERVED SAMPLES

Sample Type (container)	Preservative	Guidelines
Samples requiring preservation with formalin		
Periphyton ID (50-mL centrifuge tube)[a]	4% buffered formalin (2 mL per 50-mL sample)	Labels or tags placed inside of the jar must be of water-resistant paper or 100% rag content paper. The label on outside of the container should be completely covered with clear tape. Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form and sample tracking form.
Fish voucher specimens (1-L and/or 4-L jars)	10 % buffered formalin	
Packaging and Shipping Guidance (International Air Transport Association [IATA] Instructions 914, no limit)		
Inside packaging	High-density polyethylene (HDPE) bottles with leakproof screw-top cap (must meet United Nations [UN] specification IP2). Apply a strip of plastic tape around the cap to seal each bottle securely. Each container should only be filled to the shoulder of the bottle to provide headspace.	
Outside packaging	Screw-top plastic pail (5-gal size) with ratcheted lid is recommended (UN specification 1H2). Line container with plastic bag meeting IP5 specifications.	
Absorbent material	Not required. Stabilize contents with packing "peanuts".	
Labeling	Outside package marked with UN shipping name and ID no.:"*Environmentally hazardous substance, liquid, n.o.s. (Formalin < 5%), UN3082*". A *Class 9 miscellaneous* label and at least two package orientation labels should be affixed to the container.	
Shipping forms	Include packing list with each container. Note total quantity of formalin in liters and the gross container weight in pounds. Prepare Shippers Manifest just prior to shipment.	

(Continued)

TABLE 3-8 (Continued)

Sample Type (container)	Preservative	Guidelines
Samples requiring preservation with ethanol		
Benthic Macro-invertebrates (500-mL or 1-L jars)	At least 70 % ethanol	Confirm the sample ID assigned on the label matches the ID number recorded on the field collection form and sample tracking from.
Packaging and shipping guidance (IATA Instructions 307, 60-L limit)		
Inside packaging	HDPE bottles with leakproof screw-top cap (must meet UN specification IP2). Apply a strip of plastic tape around the cap to seal each bottle securely. Each container should only be filled to the shoulder of the bottle to provide headspace.	
Outside packaging	Screw-top plastic pail (5-gal size) with ratcheted lid is recommended (UN specification 1H2). Line container with plastic bag meeting IP5 specifications. Each pail can hold no more than 5.0 L total liquid (= 8 500-mL bottles or 5 1-L bottles).	
Absorbent material	Sufficient volume of absorbent material (vermiculite, UN A100 Absorbent sheets [18"×18"] or equivalent) to absorb contents of all inner packaging. If necessary, stabilize contents with packing "peanuts".	
Labeling	Outside package marked with UN shipping name and ID no.:"*Alcohol, flammable, toxic, n.o.s. (Denatured alcohol), UN1986*". A *Class 3 flammable* label, a *Class 6 toxic* label, and at least two package orientation labels should be affixed to the container.	
Shipping Forms	Include packing list with each container. Note total quantity of alcohol in liters and the gross container weight in pounds. Prepare Shippers Manifest just prior to shipment. Note there is a 60-L limit for a single shipment.	

[a] Preserved periphyton samples may be shipped with unpreserved samples, as the concentration of formalin is below the level which requires handling and shipment as dangerous goods.

Depending on the requirements and constraints of the project, there are other ways to ship preserved samples to laboratories so that they do not constitute dangerous goods. These generally involve holding the preserved samples for several days, decanting the preservative, and shipping the samples to the laboratory without preservative. Preservative is added upon receipt at the laboratory. This is the method used for the Wadeable Streams Assessment (U.S. EPA 2004). Consult with the laboratory to determine if any alternative shipping procedures are possible.

3.3 STATUS REPORTS

After visiting and/or sampling a site, contact the field coordinator and the EMAP information staff at WED and provide a status report. File reports every day that unpreserved samples are shipped. File a status report for *every site visited* (even if not sampled). For EMAP-W, these status reports inform the laboratory staff in Corvallis of the anticipated delivery of samples (this is especially important when samples are shipped on a Friday for Saturday delivery), and allow the information management staff to better track sites and samples (especially preserved samples that are not delivered directly to a laboratory).

The procedure for preparing and submitting a status report to WED is presented in Table 3-9. The information needed for a status report comes from the stream verification form (see Section 4) and from the tracking forms prepared for both unpreserved and preserved samples. Submit status reports to WED by phone/voice mail or by FAX. Teams should also inventory their supplies after each stream visit. Submit requests for replenishment to the field coordinator well in advance of exhausting on-hand stocks.

3.4 EQUIPMENT AND SUPPLIES

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

TABLE 3-9. STATUS REPORTING

1. File a status report after **every site visit** (even if not sampled) on the day that you ship unpreserved samples (before shipment if practical). Submit reports to the field coordinator and to the EMAP-SW information management staff at WED (Corvallis, OR).

2. Complete two separate tracking forms for each site. One form is for unpreserved samples (water chemistry, periphyton [including the ID sample], and fish tissue samples), the other for preserved samples (fish voucher and benthos). Make a copy of each form (by hand or a photocopy). Include the copy with the sample shipment (unpreserved) or with the samples themselves (preserved samples).

3. Use the original copies of the tracking forms and the stream verification form from the site to prepare the status report.

4. Contact the EMAP-SW IM message center by telephone. You will be prompted to leave a voice mail message. NOTE: There is no need to leave a separate message with the analytical laboratory staff in Corvallis. They will be able to access this number and retrieve your report.

5. Include the following information in your report:
   - Your name and the date and time of the call
   - From the stream verification form:
     - Site ID number and visit number
     - Sampling status from the verification form (e.g., Sampleable/Wadeble, Non-sampleable-not wadeable, no access-access denied, etc.)
     - Date sampled or visited
   - From the tracking form for unpreserved samples:
     - Date shipped
     - Airbill number
     - Anticipated date of delivery to laboratory (usually the next day)
     - For each sample in shipment:
       - Sample ID
       - Sample type (chemistry, periphyton, etc.)
       - Comments regarding condition or missing subsamples (Note, you do not need to record the species information for tissue samples)
   - From the tracking form for preserved samples:
     - Sample ID
     - Sample Type (vertebrate voucher, reachwide benthos, etc.)
     - Comments regarding number of jars, condition, or missing samples

   Alternatively, you can FAX copies of the verification form and two tracking forms to the **EMAP-SW IM staff FAX machine.**

Return the original forms to the data forms packet for the site for later shipment to Corvallis.

BASE LOCATION ACTIVITIES

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

Figure 3-6. Equipment and supply checklist for base location activities.

3.5 LITERATURE CITED

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NOTES

NOTES

SECTION 4
INITIAL SITE PROCEDURES

by
Alan T. Herlihy[1]

Upon arrival at a stream site, first confirm you are at the correct site. Then determine if the stream meets certain criteria for sampling and data collection activities to occur. Decide whether the stream is unduly influenced by rain events which could affect the representativeness of field data and samples. Certain conditions at the time of the visit may warrant the collection of only a subset of field measurements and samples. Finally, if it is determined that the stream is to be sampled, lay out a defined reach of the stream within which all subsequent sampling and measurement activities are conducted. Modifications to procedures in this section from those published previously for EMAP-SW by Herlihy (1998), and over the course of the EMAP-W study are presented in Appendix B.

4.1 SITE VERIFICATION ACTIVITIES

4.1.1 Locating the Index Site

Stream sampling points were chosen from the "blue line" stream network represented on 1:100,000-scale USGS maps, following a systematic randomized selection process developed for EMAP stream sampling (Stevens and Olsen 2004). The latitude and longitude of a sampling point are provided on a site information sheet that is part of the dossier compiled for each sampling site (see Section 3). To help the field team locate the sampling point later in the field, mark the location of the sampling point with an *X* on a finer-resolution (1:24,000-scale) USGS map. A geographic information system [GIS] can also be used to develop maps with the sampling points marked on them. Site coordinates on the site information sheet are based on the North American Datum 1927 (NAD 1927), and have to be converted if another datum (such as NAD 1983) is used. This point is referred to as the *index site* or ***X-site***. Include the map with the X-site identified in the dossier compiled for the site.

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

Complete a Stream Verification Form for each stream visited (regardless of whether you end up sampling it), following the procedures described in Table 4-1. While traveling from a base location to a site, record a detailed description of the last part of the route taken (e.g., from a major road or other landmark) on page 1 of the Stream Verification Form (Figure 4-1). This information will allow others to find the site again in the future. Upon reaching the X-site for a stream, confirm its location and that you are at the correct stream. Use all available means to accomplish this, and record the information on page 1 of the Stream Verification Form (Figure 4-1).

4.1.2 Determining the Sampling Status of a Stream

In probability-based survey designs such as EMAP, not all selected stream sites turn out to be streams due to errors in the sampling frame used to select the sites. An important element of the EMAP survey design is it allows the estimation of the actual extent of stream length within the study area. After the stream and location of the X-site are confirmed, evaluate the stream reach surrounding the X-site and classify the stream into one of three major sampling status categories (Table 4-1). The primary distinction between *Sampleable* and *Non-Sampleable* streams is based on the presence of a defined stream channel and presence of water.

Even if there is no water present at the X-site coordinates, the site may still be sampleable as a stream with *interrupted flow* stream (Section 4.3.1). If the channel is dry at the X-site coordinates, determine if there is water present within 75 m upstream and downstream of the X-site. If there are isolated pools of water within the 150-m reach, classify the site as *Wadeable-Interrupted* and sample it using the modified procedures outlined in Section 4.3.1. If the entire reach is dry, classify the site as *Dry-visited* on the Stream Verification Form. NOTE: Do not *slide* the reach (Section 4.3) for the sole purpose of obtaining more areas of water to sample (e.g., the downstream portion of the reach has water, but the upstream portion does not).

In some areas of the western U.S., canals are numerous. Classify the site as *NON-SAMPLEABLE-Permanent, Other* on the Stream Verification Form and identify the site as a nontarget canal in the comments section if **both** of the following conditions are true:

1. The channel within the sampling reach is totally constructed at a location where there is no evidence or other information to suggest a natural channel has ever existed.

TABLE 4-1. SITE VERIFICATION PROCEDURES

1. Find the stream location in the field corresponding to the sampling point marked on a 1:24,000 scale topographic map (***X-site***) that is provided with the dossier for each site. Record the routes taken and other directions on the Stream Verification Form so that someone can visit the same location in the future.

2. Record the site coordinates provided in the dossier for the site in the MAP COORDINATES field on the Stream Verification Form. Once the X-site is located and marked at the stream site, use a GPS receiver (set GPS datum to *NAD 1927*) to obtain the actual X-site coordinates. Record these as the GPS COORDINATES on the verification form. If a GPS receiver cannot be used, estimate the X-site coordinates as best you can from the topographic map and record these as the GPS COORDINATES on the verification form.

   NOTE: If a datum other than NAD 1927 is used (e.g., NAD 1983), the site coordinates will require conversion before visiting the site.

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

4. Scan the stream channel upstream and downstream *from the X-site*, decide if the site is sampleable or not, and mark the appropriate box on the Stream Verification Form. If the channel is dry at the X-site, determine if water is present within 75 m upstream and downstream of the X-site. Assign one of the following sampling status categories to the stream. Record the category on the Stream Verification Form.

SAMPLEABLE Categories

Wadeable: The stream can be sampled with wadeable stream protocols, continuous water flow and > 50% of the sample reach is wadeable.

Boatable: The site can be sampled by boat following nonwadeable river protocols.

Partially Sampled by Wading (Boat): More than 50% of the reach cannot be safely sampled by wadeable protocols **and** the reach is inaccessible to boat sampling due to barriers or water velocity/depth. Sample using modified procedures.

Wadeable Interrupted: The flow of water is not continual, but there is water in the sample reach (e.g., isolated pools). Sample using modified procedures.

Altered Channel: There is a stream at the location marked as the X-site on the map, but the stream channel does not appear the way it is drawn on the map. An example would be a channel rerouting following a flood event that cut off a loop of the stream. Move as far as necessary from the original site coordinates (while staying in the same stream), and establish a new X-site at the same relative position in the altered channel. Make careful notes (including distance and direction from the original site coordinates) and sketches of the changes on the Stream Verification Form.

(Continued)

TABLE 4-1 (Continued)

NON-SAMPLEABLE Categories

PERMANENT:

Dry Channel: A discernible stream channel is present but there is no water anywhere within a 150-m reach centered on the X-site. If determined at the time of the sampling visit, record on the field form as *Dry-Visited*; if the site was determined to be dry (or otherwise nonperennial) from another source and/or field verified before the actual sampling visit, record as *Dry-Not visited*.

Wetland (No definable stream channel): There is standing water present, but no definable stream channel. In cases of wetlands surrounding a stream channel, record the site as *Sampleable-Wadeable* but restrict sampling to the stream channel.

Map Error: No evidence that a water body or stream channel was ever present at the coordinates provided for the X-site.

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

Other: The site is nontarget for reasons other than those above. Examples would include underground pipelines or a nontarget canal.

A sampling site must meet **both** of the following criteria to be classified as a nontarget canal:

1. The channel is constructed and there is no evidence at the site or other information to suggest a natural channel has ever existed.
2. The sole purpose/usage of the reach is to transfer water. There are no other uses of the waterbody by humans (e.g., fishing, swimming, boating).

TEMPORARY:

Not wadeable (boatable): A site that should be sampled but the crew did not have the right equipment (e.g., A boatable river visited by a wadeable stream crew without rafts).

Other: The site could not be sampled on that particular day, but is still a target site. Examples might include a recent precipitation event that has caused unrepresentative conditions.

NO ACCESS to site Categories

Access Permission Denied: You are denied access to the site by the landowner(s).

Permanently Inaccessible: The site is unlikely to be sampled by anyone due to physical barriers that prevent access to the site (e.g., cliffs) or the channel proper (e.g., deep muck or dense vegetation along or over the channel).

Temporarily Inaccessible: The site cannot be reached at the present time due to conditions that may not be present at some future date (e.g., forest fire, high water, roads temporarily closed, unsafe weather conditions)

5. Do not sample *NON-SAMPLEABLE* or *NO ACCESS* sites. Place an *X* in the NO box for "Did you sample this site?" and check the appropriate box in the NON-SAMPLEABLE or NO ACCESS section of the Stream Verification Form. Provide a detailed explanation in comments section.

STREAM VERIFICATION FORM - STREAMS/RIVERS

Reviewed by (Initial): DP

SITE NAME: PILOT CREEK DATE: 07/01/2001 VISIT: 0 (1) 2 3

SITE ID: WXXP99-9999 TEAM: XX-1

STREAM/RIVER VERIFICATION INFORMATION

Stream/River Verified by (X all that apply): ☒ GPS ☐ Local Contact ☐ Signs ☐ Roads ☒ Topo. Map
☐ Other (Describe Here): ☐ Not Verified (Explain in Comments)

Coordinates		Latitude North	Longitude West	Type of GPS Fix	Are GPS Coordinates w/i 10 Sec. of map?
MAP	Degrees, Minutes, and Seconds OR Decimal Degrees	38 10 25	114 25 10	☐ 2D ☒ 3D	☒ Yes ☐ No
GPS	Degrees, Minutes, and Seconds OR Decimal Degrees	38 10 26	114 25 15		

DID YOU SAMPLE THIS SITE?

☒ YES If YES, check one below

SAMPLEABLE (Choose method used)
- ☒ Wadeable - Continuous water, greater than 50% wadeable
- ☐ Boatable
- ☐ Partial - Sampled by wading (Explain in comments)
- ☐ Partial - Sampled by boat (Explain in comments)
- ☐ Wadeable Interrupted - Not continuous water along reach
- ☐ Boatable Interrupted - Not continuous water along reach
- ☐ Altered - Stream/River Present but not as on Map

☐ NO If NO, check one below

NON-SAMPLEABLE-PERMANENT
- ☐ Dry - Visited
- ☐ Dry - Not visited
- ☐ Wetland (No Definable Channel)
- ☐ Map Error - No evidence channel/waterbody ever present
- ☐ Impounded (Underneath Lake or Pond)
- ☐ Other (explain in comments)

NON-SAMPLEABLE-TEMPORARY
- ☐ Not boatable - Need a different crew
- ☐ Not wadeable - Need a different crew
- ☐ Other (Explain in comments)

NO ACCESS
- ☐ Access Permission Denied
- ☐ Permanently Inaccessible (Unable/Unsafe to Reach Site)
- ☐ Temporarily Inaccessible-Fire, etc. (Explain in comments)

GENERAL COMMENTS:

DIRECTIONS TO STREAM/RIVER SITE: From Barnesville, go East on County Road 996 ~5 miles to Smithtown Road. Turn South and travel 0.6 miles to gravel road on left. Turn onto gravel road and drive 0.5 miles to house on right side of road. Owner will unlock gate to road leading to stream near the X-site location.

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

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

B. The sole purpose and usage of the waterbody is to move water. There are no other human uses, such as fishing, swimming, or boating.

If you are in doubt about whether a site is a nontarget canal, or if you think the waterbody might represent an important resource for aquatic biota, then sample it if you have permission.

If the course of the stream is different from what appears on the map, and the site coordinates provided do not fall within the existing channel, the site is considered to be "altered." *Altered streams are the **only** time the X-site can be relocated from the original coordinates.* Move as far as necessary to relocate the X-site in the same relative position in the altered channel (make sure you are still in the same stream). Record the direction and distance the X-site was moved, and sketch the altered channel configuration on the Stream Verification Form.

Record the sampling status and pertinent site verification information on the Stream Verification Form (Figure 4-1). If the site is non-sampleable or inaccessible, the site visit is completed, and no further sampling activities are conducted.

4.1.3 Sampling During or After Rain Events

Avoid sampling during high flow rainstorm events. For one, it is often unsafe to be in the water during such times. In addition, biological and chemical conditions during episodes are often quite different from those during baseflow. On the other hand, sampling cannot always be restricted to only strict baseflow conditions. It would be next to impossible to define "strict baseflow" with any certainty at an unstudied site. Such a restriction would also greatly shorten the index period when sampling activities can be conducted. Thus, some compromise is necessary regarding whether to sample a given stream because of storm events. To a great extent, this decision is based on the judgment of the field team. Some guidelines to help make this decision are presented in Table 4-2. The major indicator of the influence of storm events will be the condition of the stream itself. If you decide a site is unduly influenced by a storm event, do not sample the site that day. Classify the site as *NO ACCESS-Temporarily Inaccessible* on the Stream Verification Form. Notify the field coordinator or other central contact person to reschedule the stream for another visit.

TABLE 4-2. GUIDELINES TO DETERMINE THE INFLUENCE OF RAIN EVENTS

- If it is running at bank full discharge or the water seems much more turbid than typical for the class of stream do not sample it that day.
- Do not sample the stream if it is unsafe to wade in the majority of the stream reach.
- Keep an eye on the weather reports and rainfall patterns. Do not sample a stream during periods of prolonged heavy rains.
- If the stream seems to be close to normal summer flows, and does not seem to be unduly influenced by storm events, go ahead and sample it, even if it has recently rained or is raining.

4.1.4 Site Photographs

Taking site photographs was an optional activity in EMAP-W. However, you should seriously consider taking photos at all sites, given the current capability of digital cameras and GPS and GIS technologies. If site photos are taken, develop procedures for labeling the image files and connecting the images to a GPS location. Take photos if the site has unusual natural or man-made features associated with it. Photographs of sites believed to be minimally disturbed by human activities can provide valuable information during interpretation and assessment activities. If you do take any photographs at a stream, start the sequence with one photograph of an 8.5 × 11 inch piece of non-glossy white paper (or a small "dry erase" board) with the site ID, stream name, and date printed in large, thick letters. After the photo of the site ID information, take at least two photographs at the X-site, one in the upstream direction and one downstream. Take any additional photos you find interesting after these first three pictures. For pictures of aquatic vertebrates (see Section 12) or other small objects, place the paper with the stream ID and date in each snapshot. Keep a separate log of your photographs in a small notebook (or a separate field data form) and briefly describe each one.

4.2 LAYING OUT THE SUPPORT REACH

At each sampling point (X-site location), the response designs (Section 1.4) for the various indicators must be implemented. Unlike chemistry, which can be measured at a point, the response designs for biological assemblage and physical habitat indicators require sampling a length of a stream to obtain a representative picture of the assemblages and habitat conditions to characterize the sampling point. The length of stream sampled at each sampling point (termed the *support reach*) represents the integration of the response

designs for the individual indicators. Based on several studies (Robison 1998, Li et al. 2001, Reynolds et al. 2003), a support reach with a length of 40 times the average channel width measured near the X-site is established at each X-site.

Establish the support reach about the X-site using the procedures described in Table 4-3. Reconnoiter the support reach to make sure it is clear of obstacles that would prohibit sampling and data collection activities. Record the channel width used to determine the support reach length, and the support reach length upstream and downstream of the X-site, on page 2 of the Stream Verification Form as shown in Figure 4-2. Figure 4-3 illustrates the principal features of the established support reach, including the location of 11 cross-section transects used for physical habitat characterization (Section 7), and specific sampling points on each cross-section transect for later collection of periphyton samples (Section 9) and benthic macroinvertebrate samples (Section 10).

There are some conditions that may require adjusting the support reach about the X-site (i.e., the X-site no longer is located at the midpoint of the support reach) to avoid features we do not wish to (or physically cannot) sample across. The support reach should be of the same stream order as the X-site. Do not proceed upstream into a stream reach if the stream order decreases, or downstream into a stream reach if the stream order increases (stream order is based on 1:100,000 scale maps). If you encounter an impoundment (lake, reservoir, or pond), or an impassible barrier (e.g., a waterfall, a cliff) while laying out the support reach, adjust the reach such that the lake/stream confluence is at one end. Note this is different from when the X-site itself falls within an impounded channel (in this case, the site is not sampled unless it is still wadeable [Table 4-1]). Adjusting, or *sliding* the support reach involves noting the distance of the confluence, barrier, or other restriction from the X-site, flagging the confluence, impoundment/stream confluence, or barrier as the endpoint of the reach, and adding the distance to the other end of the reach, such that the total support reach length remains the same, but it is no longer centered about the X-site. In cases where you are denied access permission to a portion of the support reach, you can adjust the reach to make it entirely accessible; use the point of access restriction as the endpoint of the reach.

Do not *slide the support reach so that the X-site falls outside of the reach boundaries.* In this case, do not sample, and classify the site as *NO ACCESS* or *NON-SAMPLEABLE-Permanent* on the Stream Verification Form, depending on the reason for

TABLE 4-3. LAYING OUT THE SUPPORT REACH

1. Use a surveyor's rod or tape measure to determine the wetted width of the channel at 3 to 5 places considered to be of *typical width* within approximately 5 channel widths upstream and downstream from the X-site. Average the readings together and round to the nearest 0.5 m. If the average width is less than 3.5 m's, use 150 m as a minimum support reach length. Record this width on page 2 of the Stream Verification Form.

   For channels with *interrupted flow*, estimate the width based on the unvegetated width of the channel (again, with a 150 m minimum).

2. Check the condition of the stream upstream and downstream of the X-site by having one team member go upstream and one downstream. Each person goes until they can see the stream to a distance of 20 times the average channel width (equal to one-half the support reach length, but a minimum of 75 m) determined in Step 1 from the X-site.

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

3. Determine if the support reach needs to be adjusted about the X-site due to confluences with higher order streams (downstream), lower order streams (upstream), impoundments (lakes, reservoirs, ponds), physical barriers (e.g., falls, cliffs), or because of access restrictions to a portion of the initially-determined support reach.

   If such a confluence, barrier, or access restriction is present, note the distance and flag the confluence, barrier, or the limit of access as the endpoint of the reach. Move the other endpoint of the support reach an equivalent distance away from the X-site. *The X-site must still be* ***within the support reach*** *after adjustment.* The total support reach length does not change, but the support reach is no longer centered on the X-site.

   NOTE: ***Do not*** *slide the support reach to avoid man-made obstacles such as bridges, culverts, rip-rap, or channelization, or in streams with interrupted flow to obtain more inundated areas to sample.*

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

(Continued)

TABLE 4-3 (Continued)

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

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

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

STREAM VERIFICATION FORM - STREAMS/RIVERS (cont.) Reviewed by (initial): DP

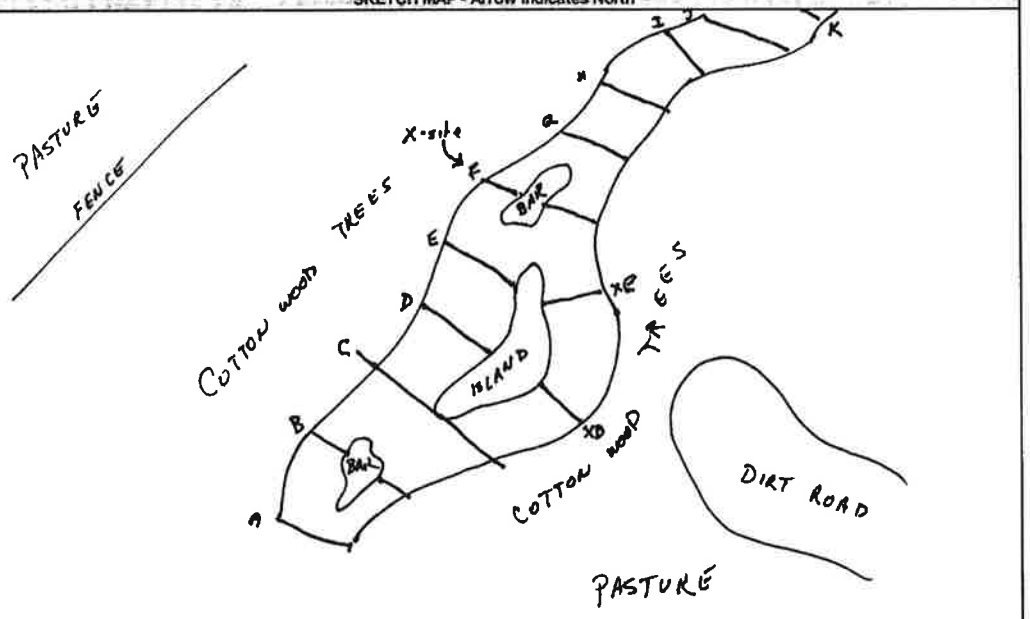
SITE NAME: PILOT CREEK DATE: 07 / 01 / 2001 VISIT: 0 (1) 2 3

SITE ID: WXXP99- TEAM: XX-1

STREAM/RIVER REACH DETERMINATION

Channel Width Used to Define Reach (m)	DISTANCE (m) FROM X-SITE		Comment
	Upstream Length	Downstream Length	
3	7.5	7.5	

SKETCH MAP - Arrow Indicates North



PERSONNEL Team Number: XX-1

NAME	DUTIES Biomorph	Geomorph	Forms
J. SNMOE	☒	☐	☐
G. WHIZ	☒	☐	☐
E. MAPPER	☐	☒	☒
S. TREAM	☐	☒	☐
	☐	☐	☐

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

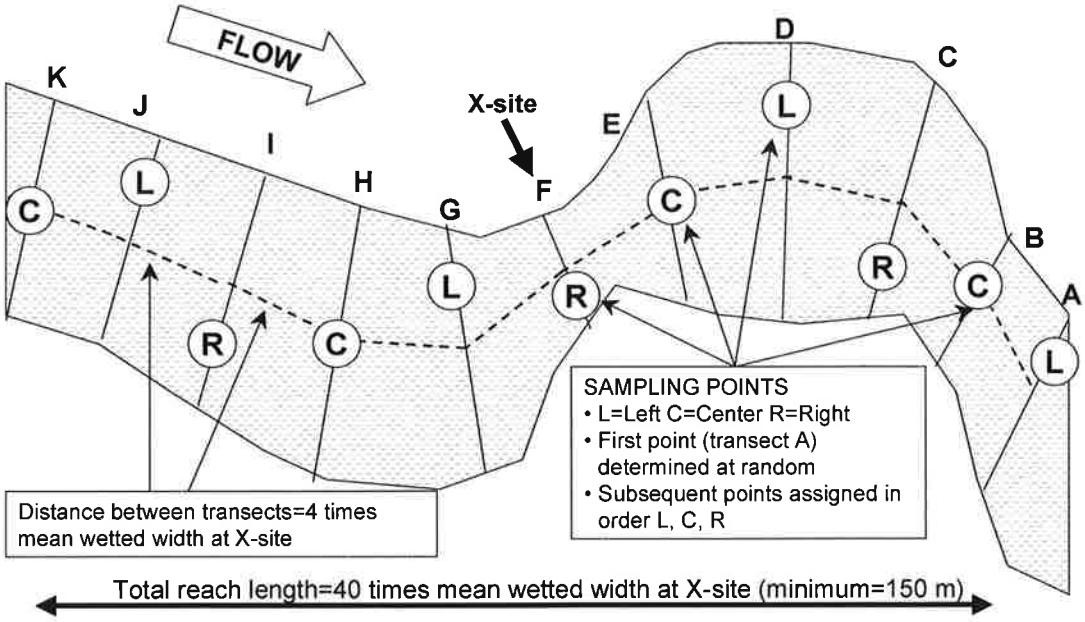

Figure 4-3. Support reach features.

sliding the reach. Do not slide a support reach to avoid man-made obstacles such as bridges, culverts, rip-rap, or channelization. These represent features and effects that EMAP is attempting to study. Do not slide the support reach to obtain more water to sample if the flow is interrupted (Section 4.3.1).

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

4.3 MODIFYING SAMPLE PROTOCOLS FOR HIGH OR LOW FLOWS

4.3.1 Streams with Interrupted Flow

The full complement of field data and samples may not be able to be collected from streams that are categorized as *Wadeable-Interrupted* (Table 4-1). Note that data are **not** collected from sampling points that are completely dry (as defined in Table 4-1). Interrupted streams will have some cross-sections with biological and habitat measurements and some with none. Modified procedures for interrupted streams are presented in Table 4-4. Collect the water chemistry samples and measurements(Section 5) at the X-site (even if the support reach has been adjusted by sliding it). If the X-site is dry and there is water elsewhere in the support reach, the collect the water chemistry sample and chemical measurements from a location having water with a surface area greater than 1 m^2 and a depth greater than 10 cm.

Collect data for the physical habitat indicator (Section 7) along the entire sample reach from interrupted streams, regardless of the amount of water present at each transect. Obtain depth measurements along the deepest part of the channel (the *thalweg*) along the entire sampling reach (even if dry) to provide a record of the *water status* of the stream for future comparisons (e.g., the percent of the support reach length with isolated pools or no water). Other measurements associated with characterizing riparian condition, substrate type, etc. are useful to help infer conditions in the stream when water is flowing continuously.

4.3.2 Partially Wadeable Sites

At some sites, it is not possible to safely wade the majority or all of the support reach because of excessive depth and/or current velocity, cascades or rapids, or extreme siltation. These sites are also not amenable to sampling by boat due to shallowness, barriers or low current velocity. In these reaches, it will be impossible to do all of the wadeable collection and measurement procedures, but wading the stream will provide more data than attempting to sample it by boat.

If more than 50% of the support reach cannot be safely sampled by wadeable protocols **and** the reach appears to be inaccessible to boat sampling due to barriers or water velocity/ depth conditions, classify the site as *SAMPLEABLE-Partially Wadeable* on the Stream Verification Form. Keeping safety in mind, attempt as much of the indicator

TABLE 4-4. MODIFICATIONS FOR SUPPORT REACHES WITH INTERRUPTED FLOW

Water Chemistry

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

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

Physical Habitat Characterization, Periphyton, Sediment, and Benthic Macroinvertebrates

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

At each of the transects (cross sections), sample the stream depending on flow status:

Dry Channel: No surface water anywhere in cross section;
Collect all physical habitat data. Use the unvegetated area of the channel to determine the channel width and the subsequent location of substrate sampling points. Record the wetted width as 0 m. Record substrate data at the sampling points located in the unvegetated, but dry, channel. Do not collect macroinvertebrates, sediment or periphyton from this transect.

Damp Channel: No flowing water at transect, only puddles of water < 10 cm deep;
Collect all physical habitat data.
Collect periphyton samples from the wet spots; these are great places for algae.
Do not collect a benthic macroinvertebrate or sediment sample.

Water Present: Transect has flow or pools > 10 cm deep;
Collect all data and measurements for physical habitat, periphyton, sediment, and benthic macroinvertebrate indicators, using standard procedures.

If at the end of sampling, there were more than 2 transects *Dry* or *Damp* so that there are missing macroinvertebrate or periphyton transects (< 10 transects) in the composite sample,, then take additional samples from other places in the stream reach that had sufficient water. Preferably, these samples would be taken at the mid-point between transects but may be taken anywhere in streams with only a small amount of water. Try to get an equivalent amount of material as you would from an 11 transect composite. Make detailed notes on the sample collection form for how and where you did sample.

Aquatic Vertebrates

- In interrupted streams, sample any wet areas within the sampling reach that are potential habitat for aquatic vertebrates. ***Do not sample*** *downstream of transect A or upstream of transect K, even if there appears to be good habitat present.*

sampling as you can. It may be impossible to do thalweg depth profiles, discharge measurements, and aquatic vertebrate sampling, but it should be possible to do the various assessments that don't require getting in the water (riparian zone characterization [Section 7], rapid habitat and/or visual assessments [Section 13]). It is also usually possible to collect a water sample for chemistry and perhaps to collect benthos and periphyton samples at transects by sampling near the stream margin or at suitable locations between established transects. The amount of sampling that can actually be done will depend on observed conditions; do only what can be done ***safely***. Be sure to make detailed comments on the Stream Verification Form, describing what the conditions were like and how much sampling could actually be done. Use the sketch map on the back of the Stream Verification Form to indicate problem areas and where samples were collected if you had to go off transect. If barriers to the site prohibit physically reaching the X-site, or if the entire support reach cannot be sampled due to unsafe conditions or extremely dense vegetation covering the channel, classify the site as *NO ACCESS-Inaccessible* on the Stream Verification Form.

4.3.3 Streams with Braided Channel Patterns

Depending upon the geographic area and/or the time of the sampling visit, you may encounter a stream having a *braided* channel pattern, which are characterized by numerous sub-channels that are generally small and short, often with no obvious dominant channel, separated by unvegetated bars (See Section 7.6.1). If you encounter a stream with a braided channel pattern, use the procedures presented in Table 4-5 to establish the support reach. The objective of this protocol modification is to avoid sampling an excessively long stretch of stream. Figuring the mean width of extensively braided systems for purposes of determining the support reach length is a bit of a challenge. For braided channels, calculate the mean width as the bankfull channel width as defined in the physical habitat protocol (Section 7). For relatively small streams (mean bankfull width near the X-site $\leq$ 15 m) the support reach is defined as 40 times the mean bankfull width near the X-site. For larger streams, (mean bankfull width near the X-site > 15 m), sum up the actual wetted width of all the braids and use that as the width for calculating the 40 channel width support reach length. If that seems too short for the system in question, lay out a longer support reach that includes 2 or 3 meander cycles, or about 6 riffle-pool sequences). Make detailed notes and sketches on the Stream Verification Form (Figure 4-2) about what you did. It's important to remember that the purpose of the 40 channel width support reach length is to sample enough stream to incorporate the variability in habitat types and obtain a representative sample of the biological assemblages. In a braided stream where there is a 100 m wide active channel (giving a 4 km reach length based on the standard procedure) and only

TABLE 4-5. MODIFICATIONS FOR STREAMS WITH BRAIDED CHANNEL PATTERNS

1. Estimate the mean width as the bankfull channel width as defined in the physical habitat protocol.

   A. If the mean width is less than or equal to 15 m, set up a 40 channel width sample reach in the normal manner.

   B. If more than 15 m, sum up the actual wetted width of all the braids and use that as the width for calculating the 40 channel width reach length. Remember the minimum reach length is always 150 m.

   C. If the reach length determined in 1B seems too short for the system in question, set up a longer sample reach, taking into consideration the objective is to sample a long enough stretch of a stream to include at least 2 to 3 meander cycles (or about 6 pool-riffle habitat sequences).

2. Make detailed notes and sketches on the Stream Verification Form about what you did.

10 m of wetted width (say five, 2-m wide braids), a 400 m long support reach length is likely to be sufficient, especially if the system has fairly homogenous habitat throughout its length.

4.4 EQUIPMENT AND SUPPLIES

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

4.5 LITERATURE CITED

Herlihy, A.T. 1998. Initial site procedures. Pages 45-56 in J.M. Lazorchak, D.J. Klemm, and D.V. Peck (Eds.). *Environmental Monitoring and Assessment Program-Surface Waters: field operations and methods for measuring the ecological condition of wadeable streams*. EPA/620/R-94/004F. U.S. Environmental Protection Agency, Washington, D.C.

Li, J., A.T. Herlihy, W. Gerth, P.R. Kaufmann, S.V. Gregory, S. Urquhart, and D.P. Larsen. 2001. Variability in stream macroinvertebrates at multiple spatial scales. *Freshwater Biology* 46:87-97.

EQUIPMENT AND SUPPLIES FOR INITIAL SITE ACTIVITIES

QTY.	Item	
1	Dossier of site and access information	
1	Topographic map with *X-site* marked	
1	Site information sheet with map coordinates and elevation of X-site	
1	GPS receiver and operating manual	
	Extra batteries for GPS receiver	
1	Stream Verification Form	
	Soft lead (#2) pencils	
1	Surveyor's telescoping leveling rod	
1	50-m fiberglass measuring tape with reel	
2 rolls	Surveyor's flagging tape (2 colors)	
	Fine-tipped indelible markers to write on flagging	
1	Waterproof camera and film (or digital camera)	
1	Sheet of cardstock or thin cardboard (white with non-glossy finish or light gray) with Site ID printed on it for use with site photographs	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for initial site activities	

Figure 4-4. Equipment and supplies checklist for initial site activities.

Reynolds, L., A.T. Herlihy, P.R. Kaufmann, S.V. Gregory, and R.M. Hughes. 2003. Electrofishing effort requirements for assessing species richness and biotic integrity in western Oregon streams. *North American Journal of Fisheries Management* 23:450-461.

Robison, E.G. 1998. *Reach scale sampling metrics and longitudinal pattern adjustments of small streams*. PhD Dissertation, Oregon State University, Corvallis, Oregon. Available from *http://www.humboldt.edu/%7Eegr2/WatershedTools.html*.

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NOTES

SECTION 5
WATER CHEMISTRY

by
Alan T. Herlihy[1]

Water chemistry data are used to characterize acid-base status, trophic condition (in terms of nutrient enrichment), chemical stressors (e.g., excessive conductivity, sulfate from mining activity, or chloride derived from human land use activities), and to classify steams based on their water chemistry. Water chemistry information includes measurements of the major cations and anions, nutrients, turbidity and color. Syringe samples are collected for laboratory analysis of pH and dissolved inorganic carbon (DIC). Syringes are used to protect samples from exposure to the atmosphere because the pH and DIC concentrations can change if the streamwater equilibrates with atmospheric CO_2.

At some sites in EMAP-W, additional *in situ* or streamside measurements of specific conductance, dissolved oxygen, and temperature were made. Specific conductance (or conductivity) is a measure of the ability of the water to pass an electrical current, which is related to the ionic strength of a solution. In natural waters, minimal concentrations of dissolved oxygen (DO) are essential for survival of most aquatic organisms. Dissolved oxygen concentration and water temperature are used to assess water quality and the potential for healthy aerobic organism populations.

The response design for water chemistry is based on the National Stream Survey (Kaufmann et al. 1988) and the Mid-Atlantic Integrated Assessment (Stoddard et al. 2006). All samples and optional field measurements are obtained at a single sampling location-below the surface at mid-channel (or an area of flowing water) at the X-site. Spatial variability across the channel of a single stream is expected to be minimal in wadeable streams as compared to the variability expected among sites, so a composite water chemistry sample is not required. Longitudinal variability at small scales (on the order of 0.5 km), appears to be minimal based on conductivity data taken systematically along several larger stream reaches in the Mid-Atlantic region. Water chemistry results from single point

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samples are associated with land use variables (Herlihy et al. 1998a), as well as biological assemblage metrics (McCormick et al. 2001, Klemm et al. 2003, Hill et al. 2003)

Most of the procedures outlined in this section are similar to the ones utilized in streams for the EPA National Surface Water Survey (NSWS; Kaufmann et al., 1988) and have been adapted from the NSWS field operations handbook (U.S. EPA, 1989). Activities and procedures presented here are essentially unchanged from those previously published for EMAP-SW (Herlihy, 1998b). Changes to collection and measurement procedures from Herlihy (1998b), and modifications made over the course of EMAP-W are summarized in Appendix B. At each stream, optional *in situ* and streamside measurements are made using field meters and recorded on standard data forms. Streamwater is collected in one 4-L container and two 60 mL syringes that are stored on ice in darkness and shipped or driven to the analytical laboratory within 24 hours of collection (see Section 3). Overnight express mail for these samples is required because the syringe samples need to be analyzed, and the 4-L bulk sample needs to be stabilized (by filtration and/or acidification) within a short period of time (72 hours) after collection.

5.1 SAMPLE COLLECTION

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

Collect a water chemistry sample as described in Table 5-1 from the middle of the stream channel at the X-site, unless no water is present at that location (see Section 4). Throughout the collection process, it is important to take precautions to avoid contaminating the sample. Rinse all sample containers three times with portions of stream water before filling them with the sample. Many streams have a very low ionic strength and can

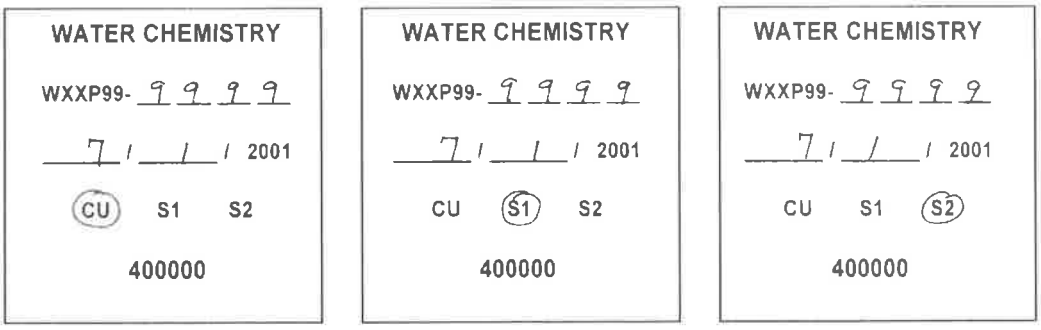


Figure 5-1. Completed sample labels for water chemistry.

be contaminated quite easily by perspiration from hands, sneezing, smoking, insect repellent, sunscreen, or other chemicals used when collecting other types of samples (e.g., formalin or ethanol). Thus, make sure that none of the water sample contacts your hands before going into the cubitainer or syringe. All of the chemical analyses conducted using the syringe samples can be affected by equilibration with atmospheric carbon dioxide; thus it is essential that no outside air contact the syringe samples during or after collection.

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

5.2 FIELD MEASUREMENTS

Table 5-2 presents the procedures for obtaining field measurement data for the water chemistry indicator. Check the conductivity and dissolved oxygen meters (if used) in the field using the same procedures as those used at a base location (Section 3). If field meters are not being used, determine stream temperature with a field thermometer (one that does not use mercury). Record the results of field checks of these meters, the transect where the measurement was made (usually the X-site), time of measurement, and the measured values for specific conductance, dissolved oxygen, and stream temperature on the Field Measurement Form as shown in Figure 5-3. If a combination dissolved oxygen-conductivity-temperature meter is being used to determine *in situ* conditions, the procedure presented in Table 5-3 may be more appropriate to use.

TABLE 5-1. SAMPLE COLLECTION PROCEDURES FOR WATER CHEMISTRY

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

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

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

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

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

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

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

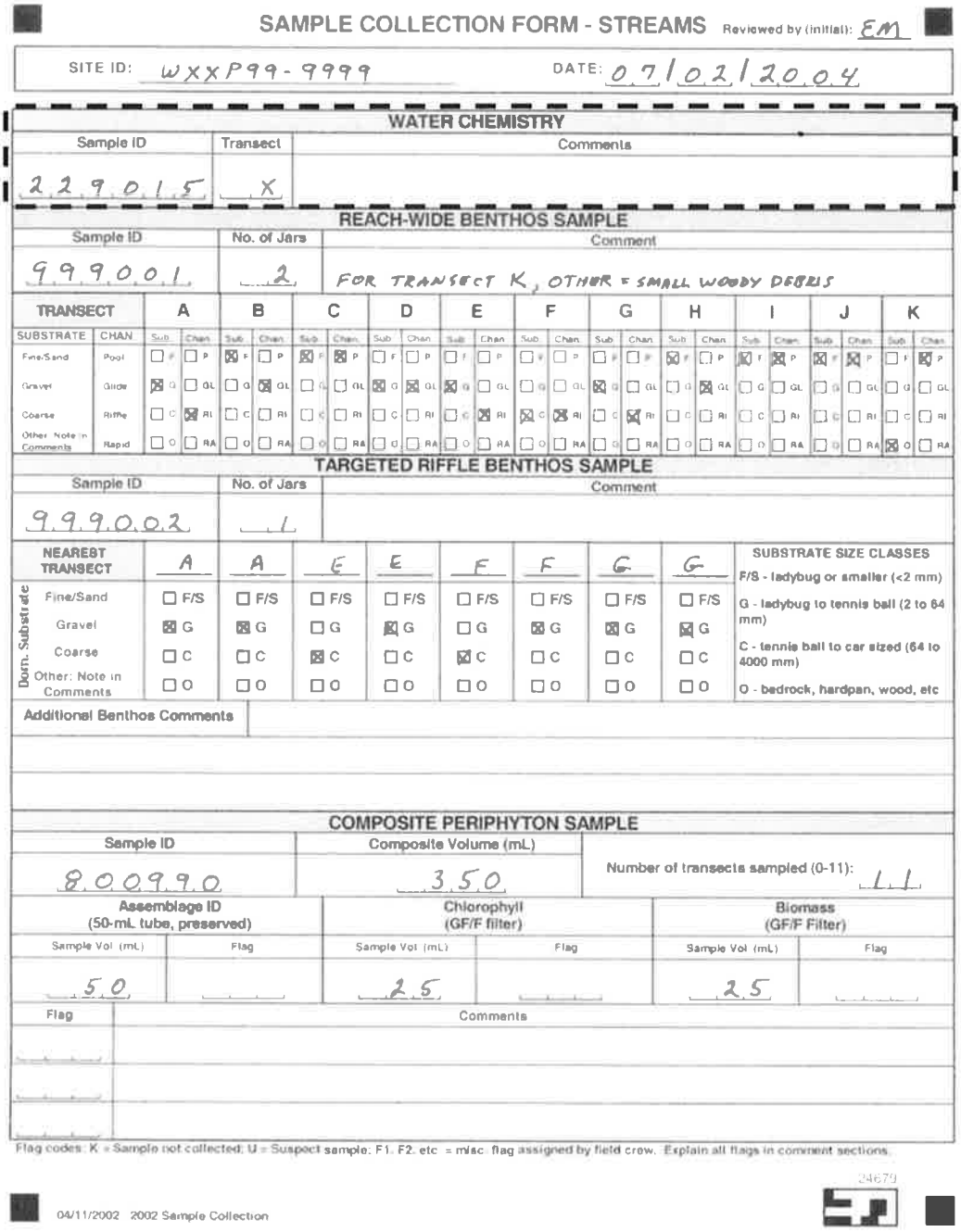
7. Submerge the syringe into the stream again and **slowly** fill the syringe with a fresh sample. Try not to get any air bubbles in the syringe. If more than 1-2 tiny bubbles are present, discard the sample and draw another one.

8. Invert the syringe (tip pointing up), and cap it with a syringe valve. Tap the syringe lightly to detach any trapped air bubbles. With the valve open, expel the air bubbles and a small volume of water, leaving between 50 and 60 mL of sample in the syringe. Close the syringe valve. If any air bubbles were drawn into the syringe during this process, discard the sample and fill the syringe again (Step 7).

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

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

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



SAMPLE COLLECTION FORM - STREAMS Reviewed by (initial): EM

SITE ID: WXXP99-9999 DATE: 07/02/2004

WATER CHEMISTRY

Sample ID	Transect	Comments
229015	X	

REACH-WIDE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999001	2	FOR TRANSECT K, OTHER = SMALL WOODY DEBRIS

TRANSECT	A		B		C		D		E		F		G		H		I		J		K	
SUBSTRATE	CHAN	Sub	Chan	Sub	Chan	Sub	Chan	Sub	Chan	Sub	Chan	Sub	Chan	Sub	Chan	Sub	Chan	Sub	Chan	Sub	Chan	Sub	Chan
Fine/Sand	Pool	☐ F	☐ P	☒ F	☐ P	☐ F	☒ P	☐ F	☐ P	☐ F	☐ P	☐ F	☐ P	☐ F	☐ P	☒ F	☐ P	☐ F	☒ P	☒ F	☒ P	☐ F	☒ P
Gravel	Glide	☒ G	☐ GL	☐ G	☒ GL	☐ G	☐ GL	☒ G	☒ GL	☒ G	☐ GL	☐ G	☐ GL	☒ G	☐ GL	☐ G	☒ GL	☐ G	☐ GL	☐ G	☐ GL	☐ G	☐ GL
Coarse	Riffle	☐ C	☒ RI	☐ C	☐ RI	☐ C	☐ RI	☐ C	☐ RI	☐ C	☒ RI	☒ C	☒ RI	☐ C	☒ RI	☐ C	☐ RI	☐ C	☐ RI	☐ C	☐ RI	☐ C	☐ RI
Other: Note in Comments	Rapid	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☐ O	☐ RA	☒ O	☐ RA

TARGETED RIFFLE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999002	1	

NEAREST TRANSECT	A	A	E	E	F	F	G	G
Dom. Substrate	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S
Fine/Sand	☒ G	☒ G	☐ G	☒ G	☐ G	☒ G	☒ G	☒ G
Gravel	☐ C	☐ C	☒ C	☐ C	☒ C	☐ C	☐ C	☐ C
Coarse	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O
Other: Note in Comments								

SUBSTRATE SIZE CLASSES
F/S - ladybug or smaller (<2 mm)
G - ladybug to tennis ball (2 to 64 mm)
C - tennis ball to car sized (64 to 4000 mm)
O - bedrock, hardpan, wood, etc

Additional Benthos Comments

COMPOSITE PERIPHYTON SAMPLE

Sample ID	Composite Volume (mL)	Number of transects sampled (0-11):
8009900	350	11

Assemblage ID (50-mL tube, preserved)		Chlorophyll (GF/F filter)		Biomass (GF/F Filter)	
Sample Vol (mL)	Flag	Sample Vol (mL)	Flag	Sample Vol (mL)	Flag
50		25		25	

Flag	Comments

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

24679

04/11/2002 2002 Sample Collection

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

TABLE 5-2. PROCEDURES FOR STREAMSIDE AND *IN SITU* CHEMISTRY MEASUREMENTS

Specific Conductance

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

2. If you haven't tested the meter at a base location recently, insert the probe into the *RINSE* container of the quality control check sample (QCCS) and swirl for 3 to 5 seconds. Remove the probe, shake it off gently, transfer it to the *TEST* container of QCCS, and let it stabilize for 20 seconds.

   If the measured conductivity is not within 10% or 10 µS/cm of theoretical value, repeat the measurement process. If the value is still unacceptable, do not use the meter until it can be inspected, diagnosed, and repaired.

3. Submerge the probe in and area of flowing water near the middle of the channel at the same location where the water chemistry sample is collected. Record the measured conductivity and any pertinent data comments about the measurement on the Field Measurement Form.

Dissolved Oxygen and Temperature

1. Inspect the probe for outward signs of fouling and for an intact membrane. Do not touch the electrodes inside the probe with any object. Always keep the probe moist by keeping it inside its calibration chamber.

2. Check the batteries and electronic functions of the meter as described in the operating manual.

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

3. After the calibration, submerge the probe in midstream at mid-depth at the same location where the water chemistry sample is collected. Face the membrane of the probe upstream, and allow the probe to equilibrate. Record the measured DO and stream temperature on the Field Measurement Form. Record the time the DO and temperature measurement was made in 24 hour units (e.g. 14:23) on the field form. If the DO meter is not functioning, measure the stream temperature with a field thermometer and record the reading on the Field Measurement Form along with any pertinent data comments.

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

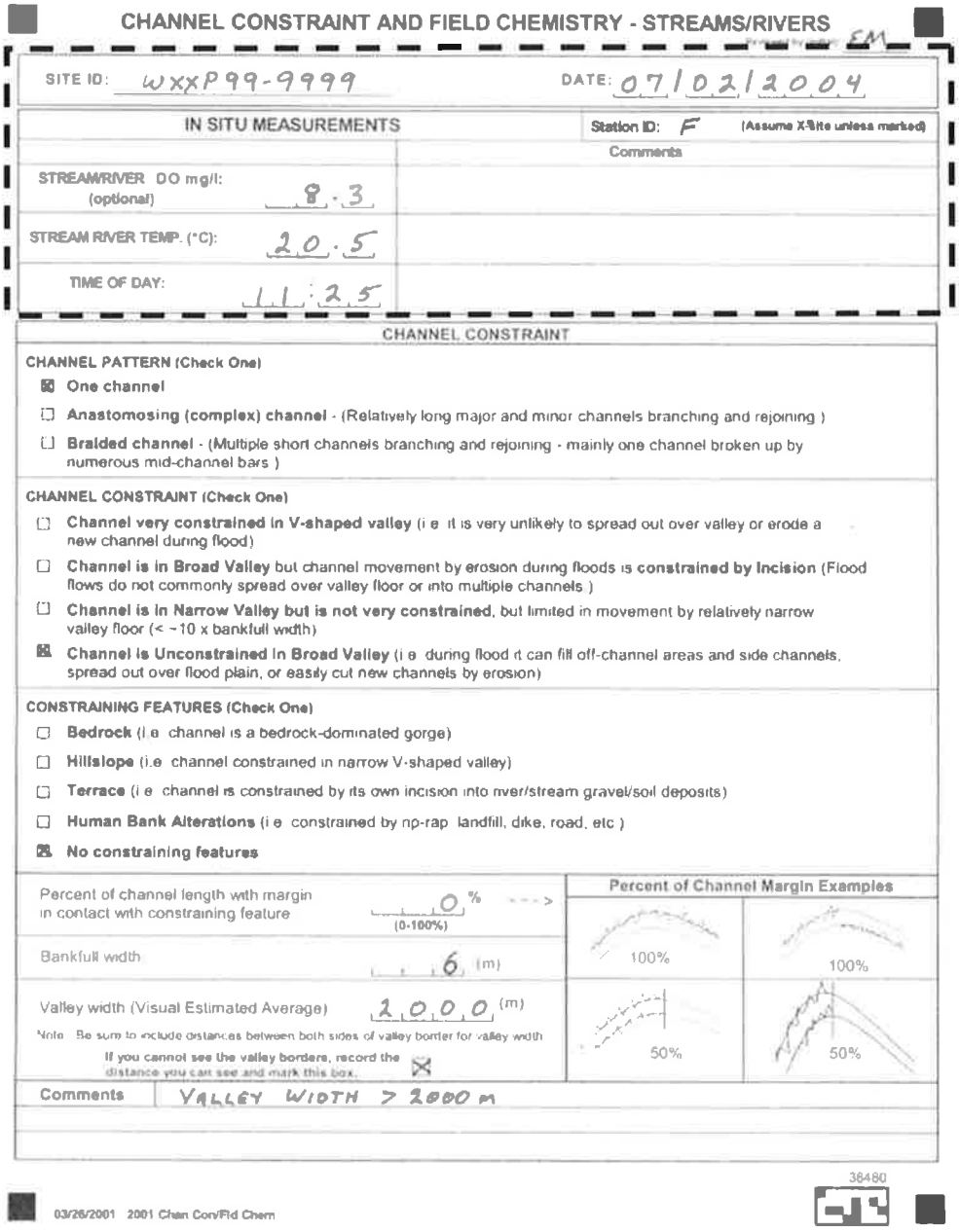
(Continued)

TABLE 5-2 (Continued)

Temperature Only (if no field meters are being used)

1. Place a field thermometer (±1 °C accuracy) beneath the surface of the stream in an area of flowing water at or near where the water chemistry samples were collected.

2. Record the stream temperature (estimated to the nearest 0.1 °C) on the Field Measurement Form. Record the time the temperature measurement was made in 24 hour units (e.g. *14:23*) on the field form, along with any pertinent data comments (e.g., measurement taken in sun or shade).



CHANNEL CONSTRAINT AND FIELD CHEMISTRY - STREAMS/RIVERS

SITE ID: WXXP99-9999 DATE: 07/02/2004

IN SITU MEASUREMENTS | Station ID: F (Assume X-Site unless marked)

	Comments
STREAM/RIVER DO mg/l: (optional) 8.3	
STREAM RIVER TEMP. (°C): 20.5	
TIME OF DAY: 11:25	

CHANNEL CONSTRAINT

CHANNEL PATTERN (Check One)

- [X] **One channel**
- [ ] **Anastomosing (complex) channel** - (Relatively long major and minor channels branching and rejoining)
- [ ] **Braided channel** - (Multiple short channels branching and rejoining - mainly one channel broken up by numerous mid-channel bars)

CHANNEL CONSTRAINT (Check One)

- [ ] **Channel very constrained in V-shaped valley** (i e it is very unlikely to spread out over valley or erode a new channel during flood)
- [ ] **Channel is in Broad Valley** but channel movement by erosion during floods is **constrained by incision** (Flood flows do not commonly spread over valley floor or into multiple channels)
- [ ] **Channel is in Narrow Valley but is not very constrained**, but limited in movement by relatively narrow valley floor (< ~10 x bankfull width)
- [X] **Channel is Unconstrained in Broad Valley** (i e during flood it can fill off-channel areas and side channels, spread out over flood plain, or easily cut new channels by erosion)

CONSTRAINING FEATURES (Check One)

- [ ] **Bedrock** (i e channel is a bedrock-dominated gorge)
- [ ] **Hillslope** (i.e channel constrained in narrow V-shaped valley)
- [ ] **Terrace** (i e channel is constrained by its own incision into river/stream gravel/soil deposits)
- [ ] **Human Bank Alterations** (i e constrained by rip-rap landfill, dike, road, etc)
- [X] **No constraining features**

		Percent of Channel Margin Examples
Percent of channel length with margin in contact with constraining feature (0-100%)	0 % --->	100% / 100%
Bankfull width	6 (m)	
Valley width (Visual Estimated Average)	2000 (m)	50% / 50%

Note Be sure to include distances between both sides of valley border for valley width

If you cannot see the valley borders, record the distance you can see and mark this box. [X]

Comments: VALLEY WIDTH > 2000 m

36480

03/26/2001 2001 Chan Con/Fld Chem

Figure 5-3. Channel Constraint and Field Measurement Form, showing data recorded for water chemistry.

TABLE 5-3. PROCEDURES FOR *IN SITU* MEASUREMENTS OF DISSOLVED OXYGEN, CONDUCTIVITY, AND TEMPERATURE USING A MULTI-FUNCTION METER[a]

Conductivity QCCS check:

1. Check the probe for fouling, intact and unwrinkled oxygen membrane, and bubble behind the membrane. Replace the electrolyte solution and membrane cap assembly if necessary.

2. Turn the meter on and allow the self-test sequence to finish (approx. 15 seconds).

3. Use the MODE key to display *temperature compensated conductivity* (The *°C* symbol on the display will be flashing).

4. Swirl the conductivity probe for 3-5 seconds in a 250-mL bottle containing the daily QCCS solution labeled *RINSE*.

5. Transfer the probe from the *RINSE* bottle to a second 250-mL bottle of QCCS labeled *TEST*. Let stabilize for 20 seconds.

6. If the measured value of the QCCS is within ±10% or ±10 µS/cm of the theoretical value (whichever is greater at the theoretical value), rinse the probe in deionized water and proceed to Step 8.
   If the measured value of the QCCS is not within ±10% or ±10 µS/cm of theoretical value, repeat Steps 4 through 6.

7. If the value is still unacceptable, replace the QCCS in both the *RINSE* and *TEST* bottles and repeat the measurement process.
   If the measured value is still not acceptable, clean the conductivity probe as described in the manual, check the batteries, and repeat Steps 1 through 6.
   If the measured value is still unacceptable, do not make any conductivity measurements. Note problems in the comments section of the field measurement form.

Dissolved oxygen calibration:

8. Check the calibration chamber and fill it with stream water to dampen the sponge and get the chamber temperature as close to stream temperature as possible. Drain the chamber and insert the probe into the chamber.

9. Press the MODE key until the dissolved oxygen reading inside the chamber is displayed in mg/L. Allow approximately 15 minutes for the readings to stabilize (i.e., a change of < 0.02 mg/L over a 1-minute period).

(Continued)

[a] For use with YSI Models 85 and 95. Modified from YSI Incorporated. 1986. *Model 85 handheld oxygen, conductivity, salinity, and temperature system operations manual.* YSI Incorporated, Yellow Springs, OH.

TABLE 5-3 (continued)

Dissolved oxygen calibration (cont.):

10. Press the UP ARROW and DOWN ARROW keys simultaneously to enter calibration mode.

11. Obtain the approximate local altitude from either the site dossier or a topographic map. Use the UP ARROW or DOWN ARROW key to enter the local altitude [to the nearest 100 feet (e.g., *15* equals 1500 ft)]. After the correct altitude is displayed, press the ENTER button.

12. In the lower part of the display, *CAL* should appear along with the theoretical value based on temperature and altitude.

13. Once the actual value displayed is stable for 10 seconds, press the ENTER button to save the calibration. NOTE: *make sure the display says* ***SAVE***. Do not turn the meter off after saving the calibration.

***In situ* measurements:**

14. Remove the probe from the calibration chamber and hold it in mid-channel and mid-depth at the X-site. Press the MODE button to cycle the display to DO in mg/L. Face the probe upstream and/or jiggle the probe up and down to ensure a continuous movement of water across the membrane surface. Unstable and inaccurate measurements will result if the flow of water across the membrane is < 0.1 m/s.

15. Wait at least 1 minute for the displayed readings to stabilize, and record the DO value and stream temperature on the Field Measurement Form.

16. Press the MODE button to cycle the display to temperature corrected specific conductance (the *°C* symbol will flash). Record the displayed conductivity value in µS/cm on the Field Measurement Form.

    NOTE: If the conductivity is high (> 999 µS/cm), the display will convert from µS/cm to mS/cm. *Be sure to check the units indicated on the display.* If mS/cm are displayed, multiply the value by 1000 to convert it to µS/cm before recording it on the data form (e.g., 9 mS/cm would be recorded as 9000 µS/cm). Extremely low values (< 10) are likely to be in mS/cm in most streams sampled in EMAP-W.

17. After completing all *in situ* measurements, rinse the probe with deionized water and store in the calibration chamber. Be sure to keep the sponge in the chamber moist at all times.

[a] For use with YSI Model 85 or equivalent. Modified from YSI Incorporated. 1986. *Model 85 handheld oxygen, conductivity, salinity, and temperature system operations manual.* YSI Incorporated, Yellow Springs, OH.

5.3 EQUIPMENT AND SUPPLIES

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

5.4 LITERATURE CITED

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EQUIPMENT AND SUPPLIES FOR WATER CHEMISTRY

QTY.	Item	
1	Dissolved oxygen/Temperature meter with probe (optional)	
1	DO repair kit containing additional membranes and probe filling solution (optional)	
1	Conductivity meter with probe (optional)	
1	250-mL or 500-mL plastic bottle of conductivity QCCS labeled *RINSE* (in plastic bag) (Optional)	
1	250 mL or 500-mL plastic bottle of conductivity QCCS labeled *TEST* (in plastic bag) (Optional)	
1	Field thermometer	
1	500 mL plastic beaker with handle (in clean plastic bag)	
1	4-L cubitainer with completed sample label attached (in clean plastic bag)	
2-4	60 mL plastic syringes (with Luer type tip) with completed sample labels attached	
1	Plastic container with snap-on lid to hold filled syringes	
2-4	Syringe valves (Mininert® with Luer type adapter, or equivalent, available from a chromatography supply company)	
1	Cooler with 4 to 6 plastic bags (1-gal) of ice **or** a medium or large opaque garbage bag to store the water sample at stream-side	
1	Sample Collection From	
1	Field Measurement Form	
1 set	Water Chemistry labels (if not filled out and attached at base site)	
	Soft-lead pencils for filling out field data forms	
	Fine-tipped indelible markers for filling out labels	
1 roll or box of tape strips	Clear packaging tape to cover labels	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for water chemistry	

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

NOTES

NOTES

SECTION 6
STREAM DISCHARGE

Philip R. Kaufmann[1]

Stream discharge is equal to the product of the mean current velocity and vertical cross sectional area of flowing water. Discharge measurements are important for assessing trends in streamwater acidity and other characteristics that are very sensitive to streamflow differences. Stream discharge information is also useful in interpreting the representativeness of water chemistry data and some physical habitat information.

The response design for stream discharge involves a single determination at a suitable location within the support reach to characterize conditions at the sampling point. The location selected should be as close as possible to the location where chemical samples are collected (typically the X-site; see Section 5), so that these data correspond. Variability in stream discharge within the support reach is expected to be small as compared to variability in stream discharge among streams, so multiple determinations at a site are not required. Discharge is usually determined after collecting water chemistry samples. Although discharge is part of the physical habitat indicator (Section 7), it is presented as a separate section because the "biomorphs" measure it while the "geomorphs" conduct the other habitat characterization procedures (see Section 2).

No single method for measuring discharge is applicable to all types of stream channels. The preferred procedure for obtaining discharge data is based on "velocity-area" methods (e.g., Rantz and others 1982, Linsley et al. 1982), and providing either the measurement data to calculate discharge (Section 6.1) or the calculated value for stream discharge (if the instrumentation has the capability; Section 6.4). For streams that are too small or too shallow to use the equipment required for the velocity-area procedure, two alternative procedures are presented. One procedure (Section 6.2) is based on timing the movement of a neutrally buoyant object (e.g., a plastic golf ball, a small rubber ball, or an orange) through a measured length of the channel, after measuring one or more cross-

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

sectional depth profiles within that length (adapted from Robins and Crawford 1954). The second procedure (Section 6.3) is based on timing the filling of a volume of water in a calibrated bucket.

Most of the procedures and activities presented here are unchanged from those previously published for EMAP-SW (Kaufmann 1998). Modifications made during the course of EMAP-W are presented in Appendix B.

6.1 VELOCITY-AREA PROCEDURE

Because velocity and depth typically vary greatly across a stream, accuracy in field measurements is achieved by measuring the mean velocity and flow cross-sectional area of many increments across a channel (Figure 6-1). Each increment gives a subtotal of the stream discharge, and the whole is calculated as the sum of these parts. Discharge measurements are made ***at only one*** *carefully chosen channel cross section within the support reach*. It is important to choose a channel cross section that is as much like a canal as possible. A glide area with a U-shaped channel cross section that is free of obstructions provides the best conditions for measuring discharge by the velocity-area method. You may remove rocks and other obstructions to improve the cross-section before any measurements are made. However, because removing obstacles from one part of a cross-section affects adjacent water velocities, you must not change the cross-section once you commence collecting the set of velocity and depth measurements.

The procedure for obtaining depth and velocity measurements is outlined in Table 6-1 (based on Rantz and others 1982). Record the data from each measurement on the Stream Discharge Form as shown in the left side of Figure 6-2. To reduce redundancy and to conserve space, Figure 6-2 shows measurement data recorded for all four procedures. In the field, use only one procedure to obtain discharge data. If you are using a current velocity meter that can compute discharge directly, refer to Section 6.4.

6.2 NEUTRALLY BUOYANT OBJECT PROCEDURE

In very small, shallow streams where the standard velocity-area method cannot be applied, obtain an estimate of discharge using the neutrally buoyant object method. The required pieces of information are the mean flow velocity in the channel and the cross-sectional area of the flow. Estimate the mean velocity measuring the time it takes for a

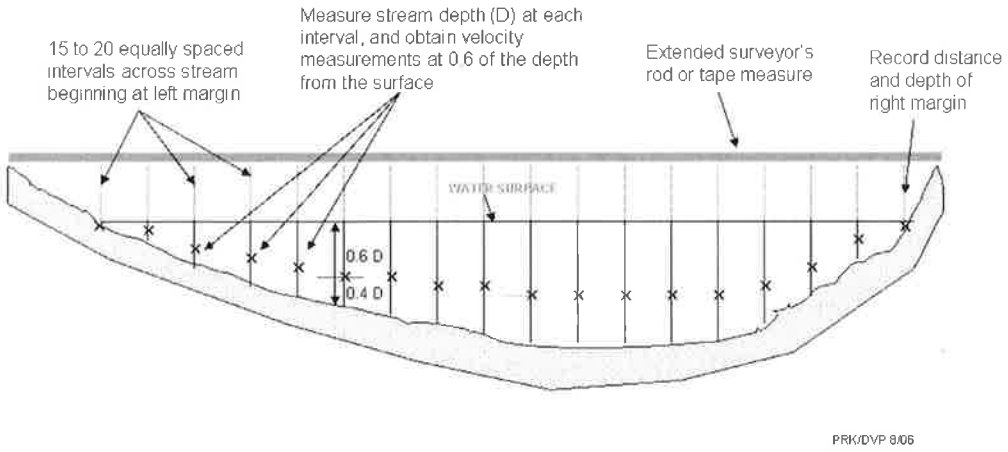

Figure 6-1. Layout of a channel cross-section for obtaining discharge data by the velocity-area procedure.

neutrally buoyant object to flow through a measured length of the channel. Determine the channel cross-sectional area from a series of depth measurements along one or more channel cross-sections. Since the discharge is the product of mean velocity and channel cross-sectional area, this method is conceptually very similar to the standard velocity-area method.

The neutrally buoyant object procedure is described in Table 6-2. Examples of suitable objects include small oranges, small sponge rubber balls, "wiffle-type" practice golf balls, or small sticks. The object must float very low in the water. It should also be small enough that it does not drag on the bottom. Choose a stream segment that is roughly uniform in cross-section, and that is long enough to require 10 to 30 seconds for an object to float through it. Select one to three cross-sections to represent the channel dimensions

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

1. Locate a cross-section of the stream channel for discharge determination that has most of the following qualities:

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

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

3. Attach the velocity meter probe to the calibrated wading rod. Check to ensure the meter is functioning properly and the correct calibration value is displayed. Calibrate (or check the calibration) the velocity meter and probe as directed in the meter's operating manual. Place an *X* in the VELOCITY AREA box on the Stream Discharge Form.

4. Divide the total wetted stream width into 15 to 20 equal-sized intervals. To determine interval width, divide the width by 20 and round up to a convenient number. Intervals should not be less than 10 cm wide, even if this results in less than 15 intervals. The first interval is located at the left margin of the stream (left when looking downstream), and the last interval is located at the right margin of the stream (right when looking downstream).

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

6. Place the wading rod in the stream at the interval point and adjust the probe or propeller so that it is at the water surface. Place an *X* in the appropriate DISTANCE UNITS and DEPTH UNITS boxes on the Stream Discharge Form. Record the distance from the left bank and the depth indicated on the wading rod on the Stream Discharge Form.

   Note for the first interval, distance equals 0 cm, and in many cases depth may also equal 0 cm. For the last interval, the distance will equal the wetted width (in cm) and depth may again equal 0 cm.

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

(Continued)

TABLE 6-1 (Continued)

8. Wait 20 seconds to allow the meter to equilibrate, then measure the velocity. Place an "X" in the appropriate VELOCITY UNITS box on the Stream Discharge Form. Record the value on the Stream Discharge Form. Note for the first interval, velocity may equal 0 because depth will equal 0. Note that negative velocity readings are possible, when recording negative values, assign a flag to denote they are indeed negative values.

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

9. Move to the next interval point and repeat Steps 6 through 8. Continue until depth and velocity measurements have been recorded for all intervals. Note for the last interval (at the right margin), depth and velocity values may equal 0.

10. At the last interval (the right margin), record a *Z* in the FLAG field on the field form to denote the last interval sampled.

STREAM DISCHARGE FORM

Reviewed by: EM

SITE ID: WXXP99-9999 DATE: 07/02/2004

☒ Velocity Area

Distance Units: ☐ ft ☒ cm | Depth Units: ☐ ft ☒ cm | Velocity Units: ☐ ft/s XX.X ☒ m/s X.XX

(Final measurement should be left here.)

	Dist. from Bank	Depth	Velocity	Flag
1	0	0	0	F1
2	20	6	-0.10	
3	40	6	0.30	
4	60	12	0.59	
5	80	15	0.37	
6	100	15	0.34	
7	120	24	0.43	
8	140	27	0.37	
9	160	40	0.43	
10	180	40	0.37	
11	200	46	0.30	
12	220	37	0.27	
13	240	30	0.25	
14	260	24	0.15	
15	280	15	0.10	
16	300	0	0	Z
17				
18				
19				
20				

☒ Timed Filling

Repeat	Volume (L)	Time (s)	Flag
1	4.0	10.5	F1
2	4.0	11.2	
3	4.0	10.8	
4	4.0	11.0	
5	4.0	10.7	

☒ Neutral Bouyant Object

	Float 1	Float 2	Float 3
Float Dist. ☐ ft ☒ m	5	5	5
Float Time (s)	10	10	12
Flag	F1		

Cross Sections on Float Reach

	Upper Section	Middle Section	Lower Section
Width ☐ ft ☒ m	2.5	1.8	3.0
Depth 1 ☐ ft ☒ cm	10	5	12
Depth 2	9	15	20
Depth 3	9	20	15
Depth 4	8	7	6
Depth 5	5	2	5

☒ Q Value

If discharge is determined directly in field, record value here: Q = 0.24 ☐ cfs ☒ m³/s FLAG F1

Flag	Comments
F1	DATA FOR ALL 4 METHODS ARE SHOWN.
Z	LAST INTERVAL MEASURED.

Flag Codes: K = No measurement or observation made; U = Suspect measurement or observation; Q = Unacceptable QC check associated with measurement; Z = Last station measured (if not Station 20); F1, F2, etc. = Miscellaneous flags assigned by each field crew. Explain all flags in comments section.

56248

02/19/2003 2003 Stream Discharge

Figure 6-2. Stream Discharge Form, showing data recorded for all four discharge measure ment procedures. Note that only one procedure is actually used at each stream site.

TABLE 6-2. NEUTRALLY BUOYANT OBJECT PROCEDURE FOR DETERMINING STREAM DISCHARGE

1. Place an *X* in the NEUTRALLY BUOYANT OBJECT box on the Stream Discharge Form.

2. Select a segment of the sampling reach that is deep enough to float the object freely, and long enough that it will take between 10 and 30 seconds for the object to travel. Mark the units used and record the length of the segment in the FLOAT DIST. field of the Stream Discharge Form.

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

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

4. At each cross section, measure the wetted width using a surveyor's rod or tape measure, and record both the units and the measured width on the Stream Discharge Form. Measure the stream depth using a wading rod or meter stick at points approximately equal to the following proportions of the total width: 0.1, 0.3, 0.5, 0.7, and 0.9. Record the units and the depth values (not the distances from the bank) on the Stream Discharge Form.

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

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

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

within the segment, depending on the variability of width and/or depth. Determine the stream depth at 5 equally spaced points at each cross-section. Measure the time required for the object to pass through the segment that includes all of the selected cross-sections. Repeat the timing two more times. Record data on the Stream Discharge Form as shown in the lower right portion of Figure 6-2.

6.3 TIMED FILLING PROCEDURE

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

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

The timed filling procedure is presented in Table 6-3. Make sure that the entire flow of the spillway is going into the bucket. Record the time it takes to fill a measured volume on the Stream Discharge Form as shown in the upper right portion of Figure 6-2. Repeat the procedure 5 times. If the cross-section contains multiple spillways, you will need to do separate determinations for each spillway. If so, clearly indicate which time and volume data replicates should be averaged together for each spillway; use an additional Stream Discharge Form if necessary. On the additional form, record a flag value (e.g., *F1*)

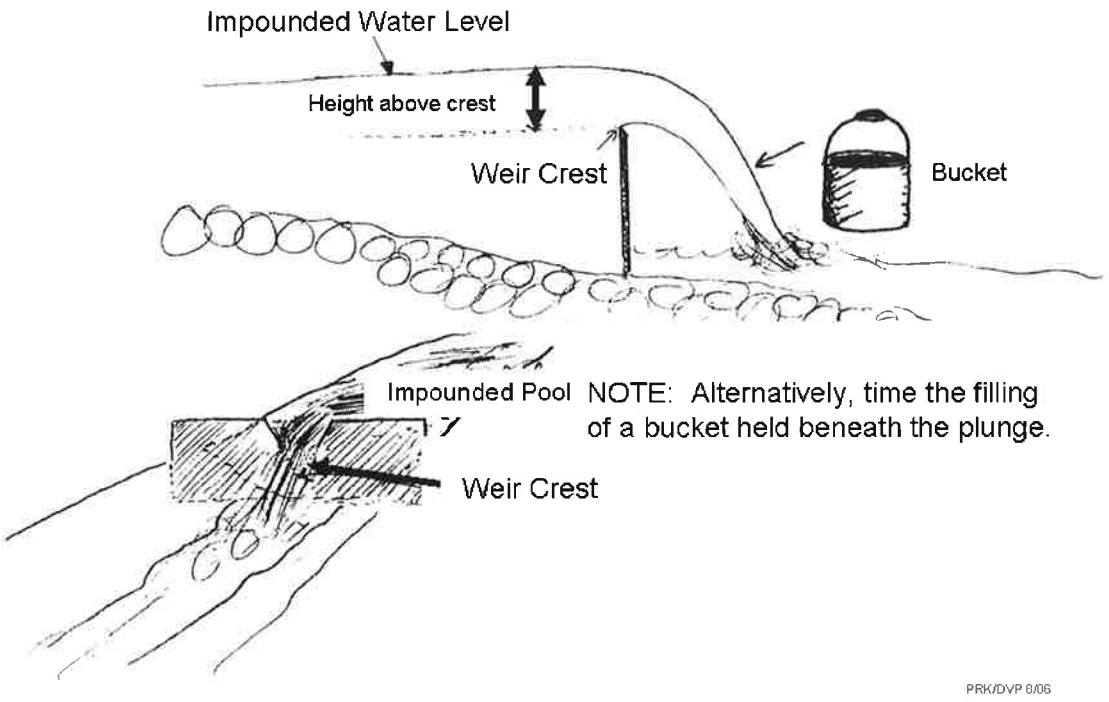

Figure 6-3. Use of a portable weir in conjunction with a calibrated bucket to obtain an estimate of stream discharge.

on all lines in the *Timed Filling* section, and explain the flag means an additional spillway was measured in the comment section.

6.4 DIRECT DETERMINATION OF DISCHARGE

The previous three procedures all provide data from which discharge (*Q*) is calculated by computer later. Some current velocity meters have the capability to calculate *Q* in the field immediately after taking all of the measurements. If you are using an instrument with this capability, record the *Q* value and associated units as shown in the lower portion of the Stream Discharge Form (Figure 6-2). Note that if the *Q Value* box is marked, do not record the raw measurement data on the form.

TABLE 6-3. TIMED FILLING PROCEDURE FOR DETERMINING STREAM DISCHARGE

NOTE: If measuring discharge by this procedure will result in significant channel disturbance or will stir up sediment, do not determine discharge until all biological and chemical measurement and sampling activities have been completed.

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

2. Place an *X* in the TIMED FILLING box in the stream discharge section of the Stream Discharge Form.

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

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

6.5 EQUIPMENT AND SUPPLIES

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

6.6 LITERATURE CITED

Kaufmann, P.R. 1998. Stream Discharge. Pages 67-76 in J.M. Lazorchak, D.J. Klemm, and D.V. Peck (eds.). *Environmental Monitoring and Assessment Program-Surface Waters: field operations and methods for measuring the ecological condition of wadeable streams.* EPA/620/R-94/004F. U.S. Environmental Protection Agency, Washington, D.C.

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NOTES

EQUIPMENT AND SUPPLIES FOR STREAM DISCHARGE

QTY.	ITEM	
1	Surveyor's telescoping leveling rod (7-.m long, metric scale, round cross-section)	
1	50-m fiberglass measuring tape and reel	
1	Current velocity meter, probe, and operating manual	
1-2	Extra batteries for velocity meter	
1	Top-set wading rod (metric or English scale) for use with current velocity meter	
1	Portable weir with 60° "V" notch (optional)	
1	Plastic sheeting to use with weir (optional)	
1	Plastic bucket (or similar container) with volume graduations	
1	Stopwatch	
1	Neutrally buoyant object (e.g., "wiffle-type" plastic golf ball, orange, small rubber ball,, or stick)	
1	Covered clipboard	
	Soft (#2) lead pencils	
	Stream Discharge Forms (1 per stream plus extras if needed for timed filling procedure or additional velocity-area intervals)	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for stream discharge	

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

SECTION 7
PHYSICAL HABITAT CHARACTERIZATION

Philip R. Kaufmann[1]

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

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

All these attributes may be altered directly or indirectly by anthropogenic activities. Nevertheless, their expected values tend to vary systematically with stream size (drainage area) and overall gradient (as measured from topographic maps). The relationships of specific physical habitat measurements described in this section to these seven attributes are discussed by Kaufmann (1993). Biological measures such as aquatic macrophytes, riparian vegetation, and large woody debris are included in this and other physical habitat assessments because of their role in modifying habitat structure and light inputs. The

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

measurements from this physical habitat characterization are used with water chemistry, temperature, and other data sources (e.g., remote sensing of basin land use and land cover). The combined data analyses will more comprehensively describe additional habitat attributes and larger scales of physical habitat or human disturbance than are evaluated by the field assessment alone. A comprehensive data analysis guide (Kaufmann et al. 1999) discusses the detailed procedures used to calculate metrics related to stream reach and riparian habitat quality from field data collected using the EMAP field protocols. This guide also discusses the precision associated with these measurements and metrics.

These procedures are intended for evaluating physical habitat in wadeable streams, and are designed for monitoring applications where robust, quantitative descriptions of the habitat at the reach scale are desired, but time is limited.. The EMAP field procedures are most efficiently applied during baseflow conditions and during times when terrestrial vegetation is active, but may be applied during other seasons and higher flows except as limited by safety considerations. The qualitative nature of the habitat quality rank scores produced by many currently available rapid habitat assessment methods (e.g., those described in Section 14, Plafkin et al. 1989, Rankin 1995, Barbour et al. 1999) have not as yet been demonstrated to meet the objectives of EMAP, where more quantitative assessment is needed for site classification, trend interpretation, and analysis of possible causes of biotic impairment.

The habitat characterization protocol developed for EMAP differs from other rapid habitat assessment approaches (e.g., Plafkin et al. 1989, Rankin 1995, Barbour et al., 1999) by employing a randomized, systematic spatial response design that allocates collection of data along the entire support reach, minimizes bias in the placement and positioning of measurements across different sites and field crews, and allows a consistent collection of data from all sites. Measures are taken over defined channel areas and these sampling areas or points are placed systematically at spacings that are proportional to baseflow channel width. This response design scales the support reach length and resolution in proportion to stream size (to represent approximately 3-4 meander cycles or 5-6 riffle-pool sequences at a given sampling point). It also allows statistical and series analyses of the data that are not possible under other designs. We strive to make the protocol objective and repeatable by using easily learned, repeatable measures of physical habitat in place of estimation techniques wherever possible. Where estimation is employed, we direct the sampling team to estimate attributes that are otherwise measurable, rather than estimating the quality or importance of the attribute to the biota or its impor-

tance as an indicator of disturbance. We have included the more traditional visual classification of channel unit scale habitat types because they have been useful in past studies and enhance comparability with other work (e.g., Bisson et al. 1982, Frissell et al. 1986, Hankie and Reeves 1988, Hawkins et al. 1993, Arend 1999).

Changes to previously published EMAP-SW procedures (Kaufmann and Robison 1998), as well as modifications implemented during EMAP-W are summarized in Appendix B. The procedures are employed on a support reach length 40 times its baseflow wetted width, as described in Section 4. Measurement points are systematically placed to statistically represent the entire reach. Stream depth and wetted width are measured at very tightly spaced intervals, whereas channel cross-section profiles, substrate, bank characteristics and riparian vegetation structure are measured at larger spacings. Woody debris is tallied along the full length of the sampling reach, and discharge is measured at one location (see Section 6). The tightly spaced depth and width measures allow calculation of indices of channel structural complexity, objective classification of channel units such as pools, and quantification of residual pool depth, pool volume, and total stream volume.

The time commitment to gain repeatability and precision is greater than that required for more qualitative methods. The additional substrate measurements (pebble count of 105 vs. 55 particles) add 20 to 30 minutes to the protocol described by Kaufmann and Robison (1998). In our field trials, two people typically complete the specified channel, riparian, and discharge measurements in about 2 to 3 hours of field time (see Section 2, Table 2-1). However, the time required can vary considerably with channel characteristics. On streams up to about 4 meters wide with sparse woody debris, measurements can be completed in about two hours. The current protocol, requiring 21 wetted width measurements, will require less than 4.5 hours for a well-practiced crew of two, even in large (>10 m wide), complex streams with abundant woody debris and deep water.

7.1 COMPONENTS OF THE HABITAT CHARACTERIZATION

There are five different components of the EMAP-W physical habitat characterization (Table 7-1), including stream discharge, which is described in Section 6. Measurements for the remaining four components are recorded on 11 copies of a two-sided field form, and separate forms for recording slope and bearing measurements, recording

TABLE 7-1. COMPONENTS OF PHYSICAL HABITAT CHARACTERIZATION

Component	Description
Thalweg Profile: (Section 7.4.1)	• Measure maximum depth, classify habitat and pool-forming features, and check presence of backwaters, side channels and loose, soft deposits of sediment particles at 10-15 equally spaced intervals between each of 11 channel cross-section transects (100 or 150 individual measurements along entire reach). • Measure wetted width and evaluate substrate particle size classes at 11 regular channel cross-section transects and midway between them (21 width measurements and substrate cross-sections).
Woody Debris Tally: (Section 7.4.2)	• Between each of the channel cross sections, tally large woody debris numbers within and above the bankfull channel according to specified length and diameter classes (10 separate tallies).
Channel and Riparian Characterization: (Section 7.5, Section 8)	• At 11 cross-section transects (21 for substrate size) placed at equal intervals along reach length: - Measure: channel cross section dimensions, bank height, bank undercut distance, bank angle, slope and compass bearing (back-sight), and riparian canopy density (densiometer). - Visually Estimate[a]: substrate size class and embeddedness; areal cover class and type (e.g., woody trees) of riparian vegetation in Canopy, Mid-Layer and Ground Cover; areal cover class of fish concealment features, aquatic macrophytes and filamentous algae. - Observe & Record[a]: Presence and proximity of human disturbances, presence of large trees, and presence of invasive riparian plants (Section 8).
Assessment of Channel Constraint, Debris Torrents, and Major Floods (Section 7.6)	• After completing thalweg and transect measurements and observations, identify features causing channel constraint, estimate the percentage of the channel margin that is constrained for the whole reach, and estimate the ratio of bankfull/valley width. Check evidence of recent major floods and debris torrent scour or deposition.
Discharge: (see Section 6)	• In medium and large streams (defined in Section 6) measure water depth and velocity at 0.6 depth at 15 to 20 equally spaced intervals across one carefully chosen channel cross-section. • In very small streams, measure discharge by timing the passage of a neutrally buoyant object through a segment whose cross-sectional area has been estimated or by timing the filling of a bucket.

[a] Substrate size class is estimated for a total of 105 particles taken at 5 equally-spaced points along each of 21 cross-sections. Depth is measured and embeddedness estimated for the 55 particles located along the 11 regular transects A through K. Cross-sections are defined by laying the surveyor's rod or tape to span the wetted channel. Woody debris is tallied over the distance between each cross-section and the next cross-section upstream. Riparian vegetation and human disturbances are observed 5m upstream and 5m downstream from the cross section transect. They extend shoreward 10m from left and right banks. Fish cover types, aquatic macrophytes, and algae are observed within the channel 5m upstream and 5m downstream from the cross section stations. These boundaries for visual observations are estimated by eye.

observations concerning riparian *legacy* (large) trees and alien invasive riparian plants (Section 8), assessing the degree of channel constraint, and recording evidence of debris torrents or recent major flooding. The *thalweg profile* is a longitudinal survey of depth, habitat class, presence of deposits of soft/small sediments, and presence of off-channel habitats at 100 equally spaced stations (150 in streams less than 2.5 m wide) along the centerline between the two ends of the sampling reach. *Thalweg* refers to the flow path of the deepest water in a stream channel. Wetted width is measured and substrate size is evaluated at 21 equally spaced cross-sections (at 11 regular transects [A through K], and 10 supplemental cross-sections spaced midway between each of these). Data for the second component, the *woody debris tally*, are recorded for each of 10 segments of stream located between the 11 regular transects. The third component, the *channel and riparian characterization*, includes measures and/or visual estimates of channel dimensions, substrate, fish cover, bank characteristics, riparian vegetation structure, presence of large (legacy) riparian trees, nonnative (alien) riparian plants (Section 8), and evidence of human disturbances. These data are obtained at each of the 11 equally-spaced transects established within the sampling reach. In addition, measurements of the stream slope and compass bearing between stations are obtained, providing information necessary for calculating reach gradient, residual pool volume, and channel sinuosity. The fourth component, *assessment of channel constraint, debris torrents, and major floods*, is an overall assessment of these characteristics for the whole reach, and is undertaken after the other components are completed.

7.2 HABITAT SAMPLING LOCATIONS WITHIN THE SUPPORT REACH

Measurements are made at two scales of resolution along the length of the support reach; the results are later aggregated and expressed for the entire reach, a third level of resolution. Figure 7-1 illustrates the locations within the support reach where data for the different components of the physical habitat characterization are obtained. We assess habitat over stream reach lengths that are approximately 40 times their average wetted width at baseflow, but not less than 150 m long. This allows us to adjust the support reach length to accommodate varying sizes of streams (see Sections 1.4 and 4.2). Many channel and riparian features are characterized on 11 cross-sections and pairs of riparian plots spaced at 4 channel-width intervals (i.e., transect spacing = 1/10th the total support reach length). The thalweg profile measurements must be spaced evenly over the entire support reach. In addition, they must be sufficiently close together that they do not miss deep areas and habitat units that are in a size range of about ⅓ to ½ of the average

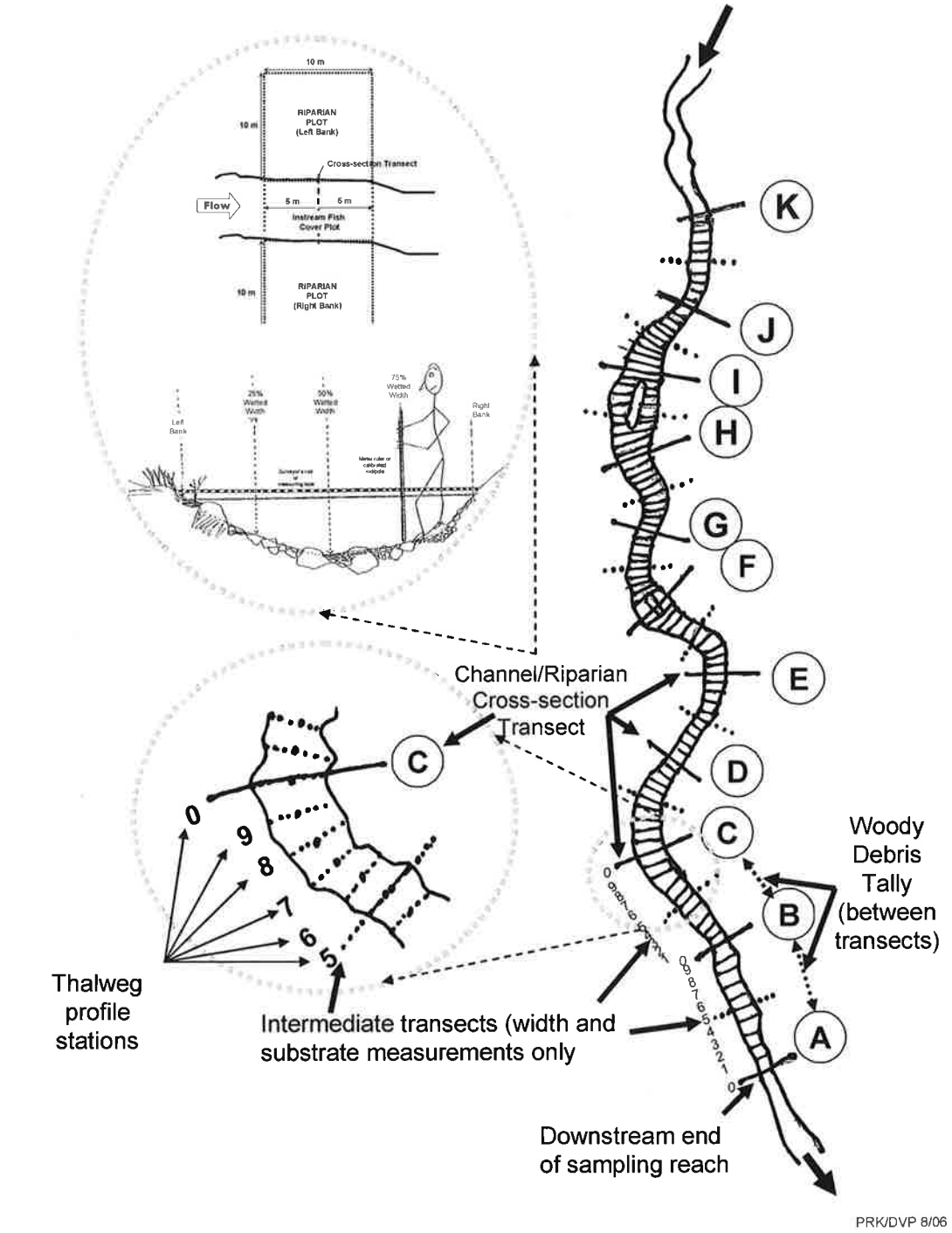


Figure 7-1. Support reach layout for physical habitat measurements (plan view).

channel width. Follow these guidelines for choosing the increment between thalweg profile measurements:

- Channel Width < 2.5 m — increment = 1.0 m
- Channel Width 2.5 to 3.5 m — increment = 1.5 m
- Channel Width > 3.5 m — increment = 0.01 × (support reach length)

Following these guidelines, you will make 150 evenly spaced thalweg profile measurements in the smallest category of streams, 15 between each detailed channel cross section. In all of the larger stream sizes, you will make 100 measurements, 10 between each cross section.

In contrast to Kaufmann and Robison (1998), we specify wetted width and substrate at 21 transects (the 11 regular transect cross-sections plus 10 supplemental cross-sections midway between regular transects), for a systematic pebble count of 105 (rather than 55) particles. If more resolution is desired based on the objectives of a particular monitoring study, width measurements may be made at all 100 or 150 thalweg profile locations.

7.3 LOGISTICS AND WORK FLOW

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

1. *Thalweg Profile and Large Woody Debris Tally (Section 7.4).* Two people (the "geomorphs") proceed upstream from the downstream end of the sampling reach (see Figure 7-1) making observations and measurements at the chosen increment spacing. One person is in the channel making width and depth measurements, and determining whether soft/small sediment deposits are present under his/her staff. The other person records these measurements, classifies the channel habitat, records presence/absence of side channels and off-channel habitats (e.g., backwater pools, sloughs, alcoves), and tallies large woody debris. Each time this team reaches a flag marking a new cross-section transect, they start filling out a new copy of the Thalweg Profile and Woody Debris Form. They interrupt the thalweg profile and woody debris tallying activities to complete data collection at each cross-section transect as it comes. When the crew member in the water makes a width measurement at channel locations midway between

regular transects (i.e., *A*, *B*, . . . *K*), she or he also locates and estimates the size class of the substrate particles on the left channel margin and at positions 25%, 50%, 75%, and 100% of the distance across the wetted channel. Procedures for this substrate tally are the same as for those at regular cross-sections, but data are recorded on the thalweg profile side of the field form.

2. *Channel/Riparian Cross-Sections (Section 7.5)*. One person proceeds with the channel cross-section dimension, substrate, bank, and canopy cover measurements. The second person records those measurements on the Channel/ Riparian Cross-section Form while making visual estimates of riparian vegetation structure, instream fish cover, and human disturbance specified on that form. They also make observations to complete the riparian "legacy" tree field form. Slope and bearing are determined together by backsighting to the previous transect. Supplementary points may need to be located and flagged (using a different color) if the stream is extremely brushy, sinuous, or steep to the point that you cannot sight for slope and bearing measures between two adjacent transects.

   The work flow for the thalweg profile and channel cross described above can be modified by delaying the backsighting measurements for slope and bearing and the woody debris tally until after reaching the upstream end of the reach. Backsighting and wood tallies can be done on the way back down (Note that in this case, the slope and bearing data form would have to be completed in reverse order).

3. *Channel Constraint and Torrent Evidence (Section 7.6)*. After completing observations and measurements along the thalweg and at all 11 transects, the field crew completes the overall reach assessments of channel constraint and evidence of debris torrents and major floods.

4. *Stream Discharge (Section 6)*. Discharge measurements are made after collecting the water chemistry sample. They are done at a chosen optimal cross section (but not necessarily at a transect) near the X-site. However, do not use the electromagnetic current meter close to where electrofishing is taking place. Furthermore, if a lot of channel disruption is necessary and sediment must be

stirred up, wait on this activity until all chemical and biological sampling has been completed.

7.4 THALWEG PROFILE AND LARGE WOODY DEBRIS MEASUREMENTS

7.4.1 Thalweg Profile

Thalweg refers to the flow path of the deepest water in a stream channel. The thalweg profile is a longitudinal survey of maximum flow path depth and several other selected characteristics at 100 or 150 equally spaced points (termed *stations*) along the length of the support reach measured along the centerline of the channel. Data from the thalweg profile allows calculation of indices of residual pool volume, stream size, channel complexity, and the relative proportions of habitat types such as riffles and pools. The EMAP-SW habitat assessment modifies traditional methods by measuring upstream distance in the **middle** of the channel, rather than along the thalweg itself (though each thalweg depth measurement is taken at the point of the deepest flow path at each station). One person walks upstream carrying a fiberglass telescoping (1.5 to 7.5 m) surveyor's rod and a 1-m metric ruler (or a calibrated rod or pole, such as a ski pole, shovel handle, wooden dowel, or old billiard cue). A second person on the bank or in the stream carries a clipboard with 11 copies of the field data form.

The procedure for obtaining thalweg profile measurements is presented in Table 7-2. Record data on the Thalweg Profile and Woody Debris Data Form as shown in Figure 7-2. Use the surveyor's rod and a metric ruler or calibrated rod or pole to make the required depth and width measurements at each station, and to measure off the distance between stations as you proceed upstream. The first thalweg measurement is taken at the transect (station *0*). Ideally, in streams 2.5 m wide or greater, every tenth thalweg measurement (station *9*) will bring you one increment spacing from the flag marking a new cross-section transect. The flag will have been set previously by carefully measuring along the channel, making the same bends that you do while measuring the thalweg profile (refer to Figure 7-1). However, you may still need to make minor adjustments to align each 10th measurement to be one increment short of the next cross section transect. In streams with average widths less than 2.5 m, make thalweg measurements at 1-meter increments. Because the minimum support reach length is set at 150 meters, there will be 15 measurements on a field data form: Station 0 at the transect plus 14 additional stations between it

TABLE 7-2. THALWEG PROFILE PROCEDURE

1. Determine the increment distance between measurement stations based on the wetted width used to determine the length of the support reach.

   For widths less than 2.5 m, establish stations every 1 m (150 total).
   For widths greater then 2.5 and less than or equal to 3.5 m, establish stations every 1.5 m (100 total).
   For widths greater than 3.5 m, establish stations at increments equal to 0.01 times the support reach length (100 total).

2. Complete the header information on the Thalweg Profile and Woody Debris Form, noting the transect pair (downstream to upstream). Record the increment distance determined in Step 1 in the *INCREMENT* field on the field data form.

3. Begin at the downstream end (*station 0*) of the first transect (transect *A*).

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

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

5. At station *5* or *7* (see above) classify the size of the bed surface particle at the tip of your depth measuring rod at the left wetted margin and at positions 25%, 50%, 75%, and 100% of the distance across the wetted width of the stream. This procedure is identical to the substrate size evaluation procedure described for regular channel cross-sections (transects *A* through *K*), except that for these midway supplemental cross-sections, substrate size is entered on the thalweg profile side of the field form.

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

   NOTE: For streams with interrupted flow, where no water is in the channel at a transect or station, record zeros for depth.

(Continued)

TABLE 7-2 (Continued)

NOTE: It is critical to obtain thalweg depths at all stations. *At stations where the thalweg is too deep to measure directly*, stand in shallower water and extend the surveyor's rod or calibrated rod or pole at an angle to reach the thalweg. Determine the rod angle by resting the clinometer on the upper surface of the rod and reading the angle on the external scale of the clinometer. Leave the depth reading for the station blank, and record a *U* flag to indicate a non-standard procedure was used. Record the water level on the rod and the rod angle in the comments section of the field data form. For even deeper depths, it is possible to use the same procedure with a taut string as the measuring device. Tie a weight to one end of a length of string or fishing line, and then toss the weight into the deepest channel location. Draw the string up tight and measure the length of the line that is under water. Measure the string angle with the clinometer exactly as done for the surveyor's rod.

If a direct measurement cannot be obtained, make the **best estimate** you can of the thalweg depth, and use a *U* flag to identify it as an estimated measurement.

7. At the point where the thalweg depth is determined, observe whether unconsolidated, loose (*soft*) deposits of small diameter (≤16mm), sediments are present directly beneath your ruler, rod, or pole. *Soft/small sediments* are defined here as fine gravel, sand, silt, clay or muck readily apparent by "feeling" the bottom with the staff. Record presence or absence in the *SOFT/SMALL SEDIMENT* field on the field data form. Note: A thin coating of fine sediment or silty algae coating the surface of cobbles should not be considered soft/small sediment for this assessment. However, fine sediment coatings should be identified in the comments section of the field form when determining substrate size and type.

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

9. If the station cross-section intersects a mid-channel bar, indicate the presence of the bar in the *BAR WIDTH* field on the field data form.

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

11. Record the presence or absence of quiescent off-channel aquatic habitats, including sloughs, alcoves and backwater pools in the *BACKWATER* column of the field form.

12. Proceed upstream to the next station, and repeat Steps 2 through 11.

13. Repeat Steps 2 through 12 until you reach the next transect. At this point complete Channel/Riparian measurements at the new transect (Section 7.5). Then prepare a new Thalweg Profile and Woody Debris Form and repeat Steps 2 through 12 for each of the reach segments, until you reach the upstream end of the sampling reach (transect *K*). At transect *K*, you will have completed 10 copies of the Thalweg Profile and Woody Debris Form, one for each segment (*A* to *B*, *B* to *C*, etc.).

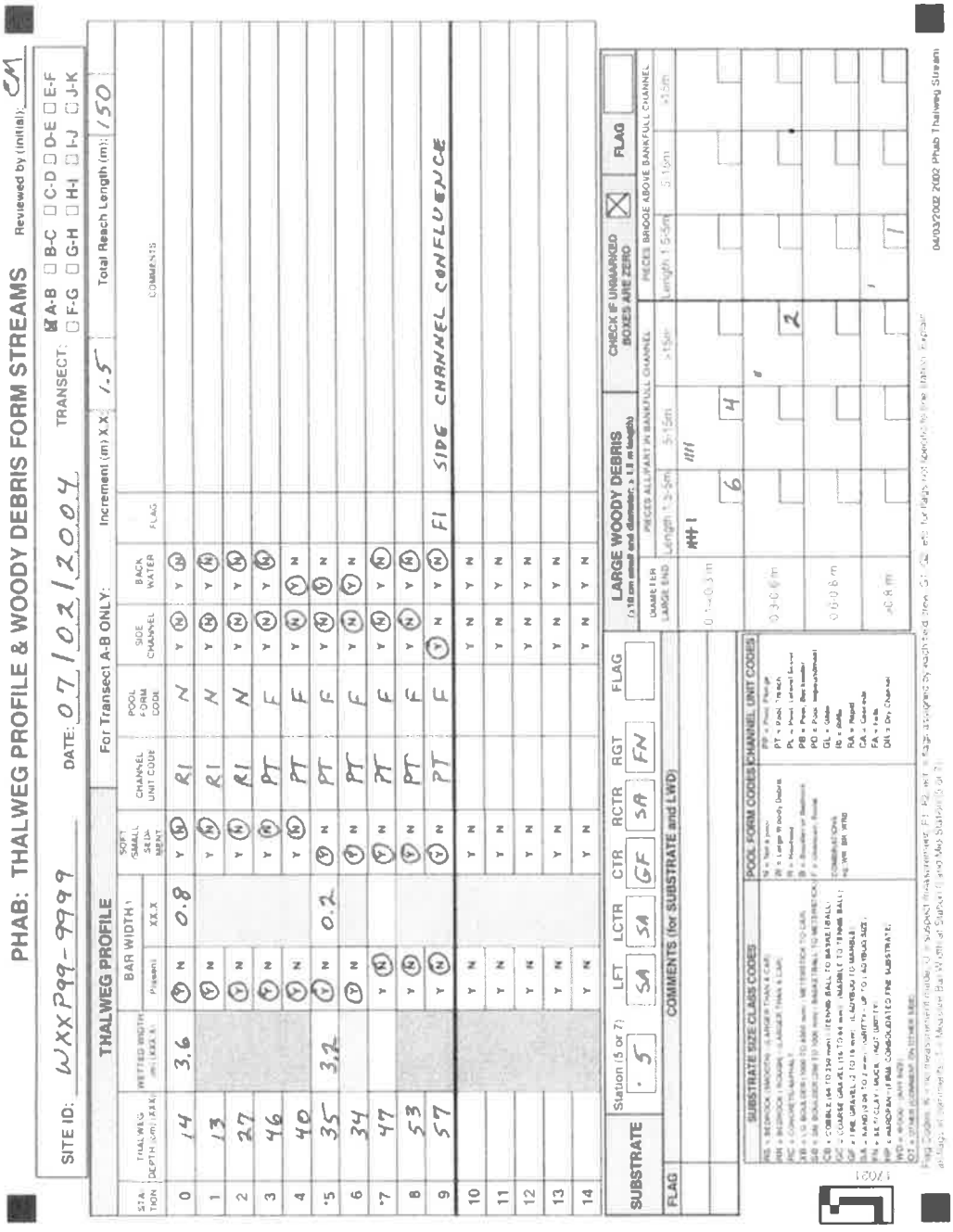


Figure 7-2. Thalweg Profile and Woody Debris Form.

and the next transect upstream. Use the five extra lines on the thalweg profile portion of the data form (Figure 7-2) to record these measurements.

It is very important that thalweg depths are obtained from **all** measurement stations. Missing depths at the ends of the support reach (e.g., due to the stream flowing into or out of a culvert or under a large pile of debris) can be tolerated, but those occurring in the middle of the support reach are more difficult to deal with. Flag these missing measurements using a *K* code and explain the reason for the missing measurements in the comments section of the field data form. At points where a direct depth measurement cannot be obtained, make your best estimate of the depth, record it on the field form, and flag the value using a *U* code (for a nonstandard measurement), explaining that it is an estimated value in the comments section of the field data form. *Where the thalweg points are too deep for wading*, measure the depth by extending the surveyor's rod at an angle to reach the thalweg point. Record the water level on the rod, and the rod angle, as determined using the external scale on the clinometer (vertical = 90°). This procedure can also be done with a taut string or fishing line (see Table 7-2). In analyzing this data we calculate the thalweg depth as the length of the rod (or string) under water multiplied by the trigonometric *sine* of the rod angle. (For example, if 3 meters of the rod are under water when the rod held at 30 degrees (*sine*=0.5), the actual thalweg depth is 1.5 meters.) These calculations are done after field forms are returned for data analysis. On the field form, crews are required only to record the wetted length of the rod under the water, a *U* code in the flag field (to indicate a nonstandard technique), and a comment to the right saying "*depth taken at an angle of xx degrees.*" If a direct measurement of the thalweg depth is not possible, make the best estimate you can of the depth, record it, and use a *U* flag and a comment to note it is an estimated value.

At every thalweg station, determine by sight or feel whether deposits of *soft/small sediments* are present on the channel bottom. These particles are defined as substrate equal to or smaller than fine gravel (≤ 16 mm diameter). These soft/small sediments are **different** from *Fines* described when determining the substrate particle sizes at the cross-section transects (Section 7.5.2). If the channel bottom is not visible, determine if soft/small sediment deposits are readily obvious by feeling the bottom with your boot, the surveyor's rod, or a calibrated rod or pole. (Note that a very thin coating of silt or algae on a cobble bottom substrate **does not** qualify as soft/small sediments for this purpose.)

Wetted width is measured at each transect (station *0*), and midway between transects (station *5* for larger streams having 100 measurement points, or station *7* for smaller streams having 150 measurement points). The wetted width boundary is the point at which substrate particles are no longer surrounded by free water. Substrate size is estimated for five particles evenly spaced across each midway cross-section using procedures identical to those described for substrate at regular cross-sections (Section 7.5.2), but at the supplemental cross-sections, only the size class (not the distance and depth) data are recorded in spaces provided on the thalweg profile form.

While recording the width and depth measurements and the presence of soft/small sediments, the second person evaluates and records the habitat class and the pool forming element (Table 7-3) applicable to each of the 100 (or 150) measurement points along the length of the reach. These channel unit habitat classifications and pool-forming elements are modified from those of Bisson et al. (1982) and Frissell et al. (1986). The resulting database of traditional visual habitat classifications provides a bridge of common understanding with other studies. Make channel unit scale habitat classifications at the thalweg of the cross section. The habitat unit itself must meet a minimum size criteria in addition to the qualitative criteria listed in Table 7-3. Before being considered large enough to be identified as a channel-unit scale habitat feature, the unit should be at least as long as the channel is wide. For instance, if there is a small deep (pool-like) area at the thalweg within a large riffle area, do not record it as a pool unless it occupies an area about as wide or long as the channel is wide. If a backwater pool **dominates the channel**, record *PB* as the dominant habitat unit class. If the backwater is a pool that **does not dominate** the main channel, or if it is an **off-channel** alcove or slough (large enough to offer refuge to small fishes), circle *Y* to indicate presence of a backwater in the *BACKWATER* column of the field form, but classify the main channel habitat unit type according to characteristics of the main channel. *Sloughs* are backwater areas having marsh-like characteristics such as vegetation, and *alcoves* (or *side pools*) are deeper areas off the main channel that are typically wide and shallow (Helm 1985, Bain and Stevenson 1999). When trying to identify the pool forming element for a particular pool, remember that most pools are formed at high flows, so you may need to look for elements that are dry at baseflow, but still within the bankfull channel (e.g., boulders or large woody debris).

Mid-channel bars, islands, and side channels pose some problems for the sampler conducting a thalweg profile and require some guidance. *Bars* are defined here as mid-channel features **below the bankfull flow** mark that are dry during baseflow conditions

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

Channel Unit Habitat Classes[a]	
Class (Code)	**Description**
Pools:	Still water, low velocity, a smooth, glassy surface, usually deep compared to other parts of the channel:
Plunge Pool (*PP*)	Pool at base of plunging cascade or falls.
Trench Pool (*PT*)	Pool-like trench in the center of the stream
Lateral Scour Pool (*PL*)	Pool scoured along a bank.
Backwater Pool (*PB*)	Pool separated from main flow off the side of the channel (large enough to offer refuge to small fishes). Includes sloughs (backwater with marsh characteristics such as vegetation), and alcoves (a deeper area off a wide and shallow main channel)
Impoundment Pool (*PD*)	Pool formed by impoundment above dam or constriction.
Pool (*P*)	Pool (unspecified type).
Glide (*GL*)	Water moving slowly, with a *smooth, unbroken surface*. Low turbulence.
Riffle (*RI*)	Water moving, with *small ripples, waves and eddies* -- waves not breaking, *surface tension not broken*. Sound: babbling, gurgling.
Rapid (*RA*)	Water movement rapid and turbulent, surface with *intermittent whitewater* with breaking waves. Sound: continuous rushing, but not as loud as cascade.
Cascade (*CA*)	Water movement rapid and very turbulent over steep channel bottom. Much of the water surface is broken in *short, irregular plunges, mostly whitewater*. Sound: roaring.
Falls (*FA*)	*Free falling water* over a vertical or near vertical drop into plunge, water turbulent and white over high falls. Sound: from splash to roar.
Dry Channel (*DR*)	No water in the channel, or flow is submerged under the substrate (*hyporheic flow*).

(Continued)

[a] Note that in order for a channel habitat unit to be distinguished, it must be at least as wide or long as the channel is wide (except for off channel backwater pools, which are noted as present regardless of size).

TABLE 7-3 (Continued)

Categories of Pool-forming Elements[b]

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

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

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

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

Less than 15%	Indicate the presence of a side channel on the thalweg profile form.
16 to 49%	Indicate the presence of a side channel on the thalweg profile form. If you reach a cross-section transect with an island and side channel present, establish a secondary transect across the side channel designated as *X* plus the primary transect letter; (e.g., *XA*), by checking boxes for both *X-TRY SIDE CHANNEL* and the appropriate transect letter (e.g., *A* through *K*) on a **separate copy** of the field data form. Complete the detailed channel and riparian cross-section measurements for the side channel on this form (see Section 7.5.8).

When a side channel occurs due to an island, indicate its presence with continuous entries in the *SIDE CHANNEL* field on the Thalweg Profile and Woody Debris Form (Figure 7-2). In addition, note the points of divergence and confluence of the side channel in the comments section of the thalweg profile form. Begin entries at the point where the side channel converges with the main channel, and continue noting the side channel presence until you reach the upstream point where it diverges. When doing width measures with a side channel separated by an island, include only the width of the main channel in the measures at the time and then measure the side channel width separately.

For streams with interrupted flow, where no water is in the channel at a thalweg station (or where flow is submerged beneath the substrate), record zeros for depth and wetted width. Record the habitat type as dry channel (*DR*).

7.4.2 Large Woody Debris Tally

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

The procedure for tallying LWD is presented in Table 7-4. The tally includes all pieces of LWD that are at least partially in the baseflow channel (Zone 1), in the *active channel* (Zone 2, flood channel up to bankfull stage), or spanning above the active channel (Zone 3), as shown in Figure 7-4. The *active* (or *bankfull*) *channel* is defined as the channel that is filled by moderate sized flood events that typically recur every one to two years. LWD in or above the active channel is tallied over the entire length of the reach, including the area between the channel cross-section transects. Pieces of LWD that are not at least partially within Zones 1, 2, or 3 are not tallied.

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

TABLE 7-4. PROCEDURE FOR TALLYING LARGE WOODY DEBRIS

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

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

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

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

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

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

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

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

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

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

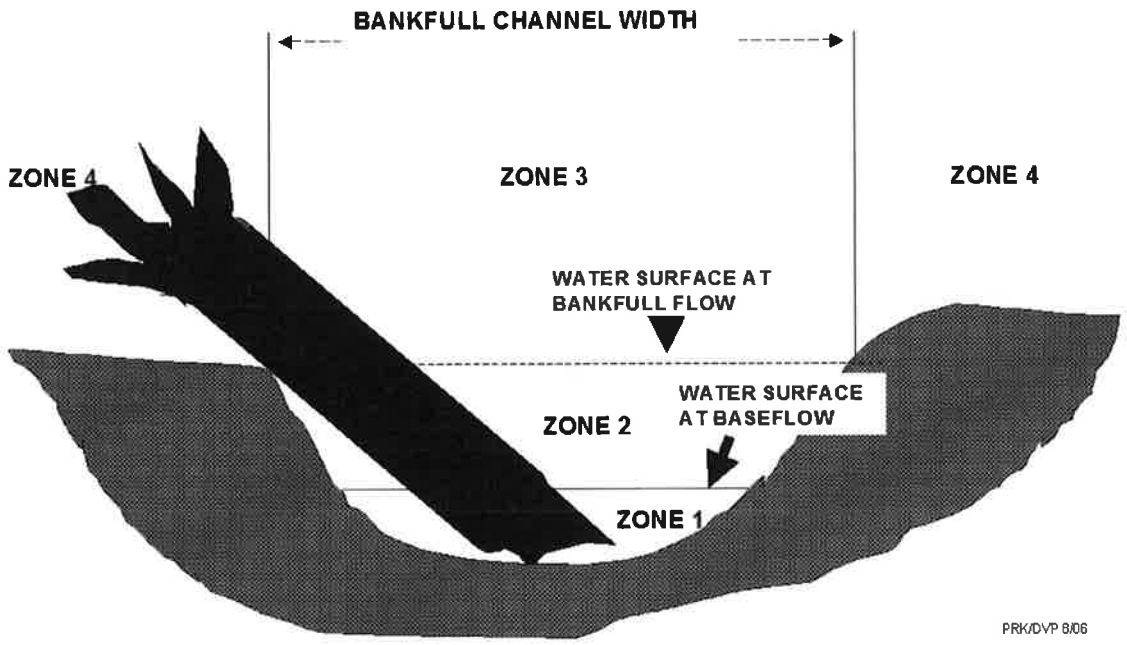


Figure 7-3. Large woody debris influence zones (modified from Robison and Beschta, 1990)

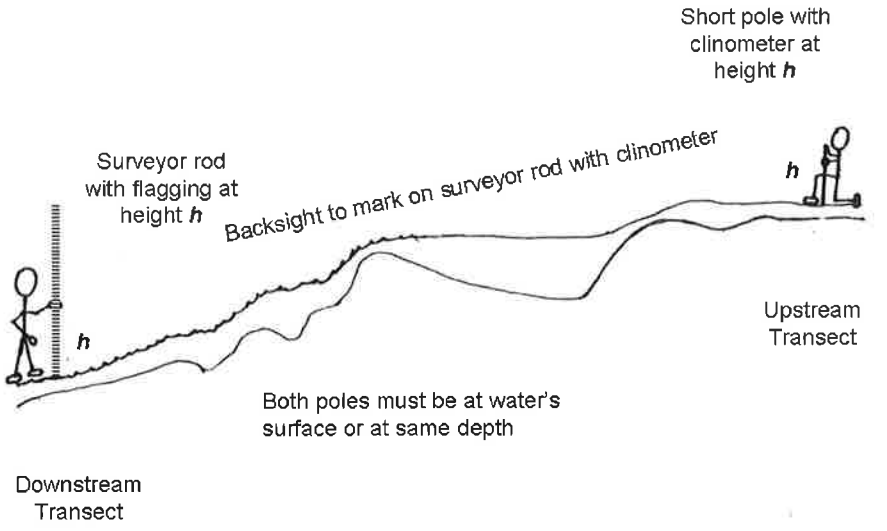
7.5 CHANNEL AND RIPARIAN MEASUREMENTS AT CROSS-SECTION TRANSECTS

7.5.1 Slope and Bearing

The slope, or gradient, of the stream reach is useful in four different ways. First, the overall stream gradient is one of the major stream classification variables, giving an indication of potential water velocities and stream power, which are in turn important controls on aquatic habitat and sediment transport within the reach. Second, the spatial variability of stream gradient is a measure of habitat complexity, as reflected in the diversity of water velocities and sediment sizes within the stream reach. Third, using methods described by Stack (1989) and Robison and Kaufmann (1994), the water surface slope, in conjunction with the multiple depth and width measurements taken in the thalweg profile (Section 7.4.1), is used to compute residual pool depths, volumes, and numbers. Finally, slope, in combination with measurements of channel dimensions, wood, and substrate particle size, is necessary for calculating relative bed stability (Kaufmann et al. 1999). Compass bearing between cross-section transects, along with the distance between transects, is used to estimate the sinuosity of the channel (ratio of the length of the reach divided by the straight line distance between the two reach ends).

Measure slope and bearing by *backsighting* downstream between transects (e.g., transect *B* to *A*, *C* to *B*, etc.) as shown in Figure 7-4. To measure the slope and bearing between adjacent transects, follow the procedure presented in Table 7-5. Record slope and bearing data on the Slope and Bearing Form as shown in Figure 7-5. An alternative procedure, where slope and bearing measurements are taken in reverse order (upstream to downstream) is presented in Table 7-6. Note that in this case, the Slope and Bearing Form is completed in **reverse order** (right to left, bottom to top), so be careful to record the measurements in the correct order if you choose to follow this procedure.

Slope is typically measured by two people, each having a pole that is marked at the same height. Alternatively (but much less precise), the second person can be marked at the eye level of the person doing the backsighting. *Be sure that you mark your eye level on the other person or on a separate pole beforehand while standing on level ground.* Sight to **your eye level** when backsighting on your co-worker. *We recommend that field crews use poles marked at* ***exactly the same height*** *for sighting slope, particularly in streams with slopes* ***less than 3%***. When two poles are used, sight from the mark on one pole to the mark on the other. Also, be sure that the second person is standing (or holding



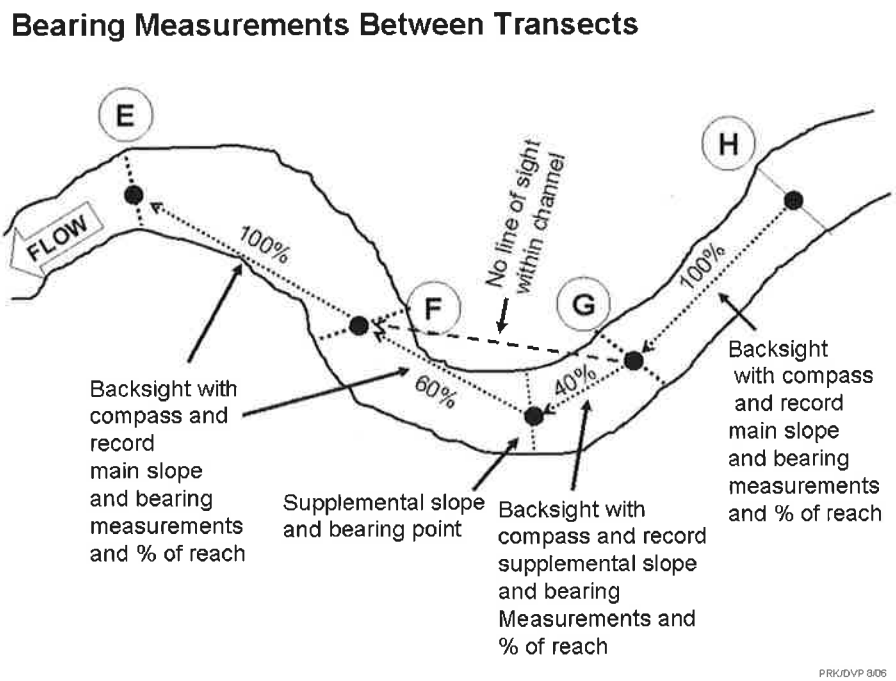


Figure 7-4. Channel slope and bearing measurements.

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

1. Stand in the center of the channel at the downstream cross-section transect. Determine if you can see the center of the channel at the next cross-section transect upstream without sighting across land (i.e., do not "short-circuit" a meander bend). If not, you will have to take supplementary slope and bearing measurements.

2. Mark a surveyor's rod and a calibrated rod (or meter ruler) at the same height. If a shorter pole or ruler is used, measure the height from the ground to the opening of the clinometer when it is resting on top.

3. Have one person take the marked surveyor's rod to the downstream transect. Hold the rod vertical with the bottom at the same level as the water surface. If no suitable location is available at the stream margin, position the rod in the water and note the depth.

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

4. Walk upstream to the next cross-section transect. Place the base of the calibrated rod at the level as the surveyor's rod (either at the water surface or at the same depth in the water).

   - If you have determined in Step 1 that supplemental measurements are required for this segment, walk upstream to the furthest point where you can stand in the center of the channel and still see the center of the channel at the downstream cross-section transect. Remember that your line of sight cannot "cross land." Mark this location with a different color flagging than that marking the cross-section transects.

5. Place the clinometer on the calibrated rod at the height determined in Step 2. With the clinometer, sight back downstream to the flagged height on the surveyor's rod at the downstream transect (or at the supplementary point). Mark the transect and method (*CL*) on the Slope and Bearing Form. Read and record the **percent** slope in the *MAIN* section on the Slope and Bearing Form. Record the *PROPORTION* as 100%. Mark the *%* box below the recorded value on the form. NOTE: if using a method where elevation difference is measured, mark the appropriate method box[a] and the *CM* box on the form.

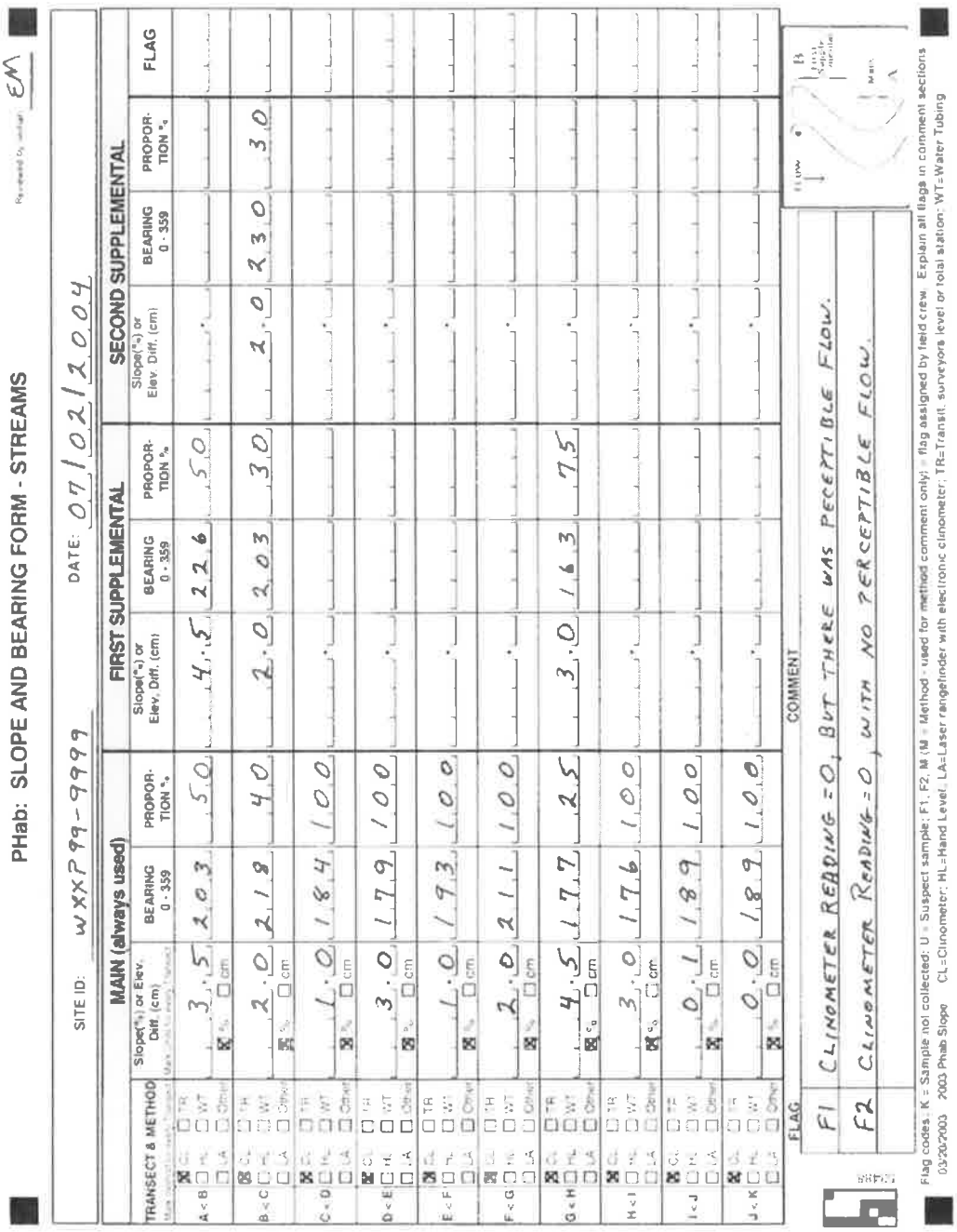
   - If you are backsighting from a supplemental point, record the slope (%) and proportion (%) of the stream segment that is included in the measurement in the appropriate *SUPPLEMENTAL* section of the Slope and Bearing Form.

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

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

7. Proceed to the next cross-section transect (or to a supplementary point), and repeat Steps 3 through 5 above.

[a] Method codes are: *CL*=clinometer, *TR*=transit, *HL*=hand level, *WT*=Water tube, *LA*=laser level, *OTHER*=method not listed (describe in comments section of form).



PHab: SLOPE AND BEARING FORM - STREAMS

SITE ID: WXP99-9999 DATE: 07/02/2004

TRANSECT & METHOD	MAIN (always used) Slope(%) or Elev. Diff. (cm)	BEARING 0 - 359	PROPORTION %	FIRST SUPPLEMENTAL Slope(%) or Elev. Diff. (cm)	BEARING 0 - 359	PROPORTION %	SECOND SUPPLEMENTAL Slope(%) or Elev. Diff. (cm)	BEARING 0 - 359	PROPORTION %	FLAG
A < B ☒ TR ☐ WT ☐ LA ☐ Other	3.5 ☒ % ☐ cm	203	50	4.5	226	50				
B < C ☒ TR ☐ WT ☐ LA ☐ Other	2.0 ☒ % ☐ cm	218	40	2.0	203	30	2.0	230	30	
C < D ☒ TR ☐ WT ☐ LA ☐ Other	1.0 ☒ % ☐ cm	184	100							
D < E ☒ TR ☐ WT ☐ LA ☐ Other	3.0 ☒ % ☐ cm	179	100							
E < F ☒ TR ☐ WT ☐ LA ☐ Other	1.0 ☒ % ☐ cm	193	100							
F < G ☐ TH ☐ WT ☐ LA ☐ Other	2.0 ☒ % ☐ cm	211	100							
G < H ☒ TR ☐ WT ☐ LA ☐ Other	4.5 ☒ % ☐ cm	177	25	3.0	163	75				
H < I ☒ TR ☐ WT ☐ LA ☐ Other	3.0 ☒ % ☐ cm	176	100							
I < J ☒ TR ☐ WT ☐ LA ☐ Other	0.1 ☒ % ☐ cm	189	100							
J < K ☒ TR ☐ WT ☐ LA ☐ Other	0.0 ☒ % ☐ cm	189	100							

COMMENT

FLAG

F1 CLINOMETER READING = 0, BUT THERE WAS PERCEPTIBLE FLOW.

F2 CLINOMETER READING = 0, WITH NO PERCEPTIBLE FLOW.

Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, M (M = Method - used for method comment only) = flag assigned by field crew. Explain all flags in comment sections
03/20/2003 2003 Phab Slope CL=Clinometer; HL=Hand Level; LA=Laser rangefinder with electronic clinometer; TR=Transit, surveyors level or total station; WT=Water Tubing

Figure 7-5. Slope and Bearing Form.

TABLE 7-6. MODIFIED PROCEDURE FOR OBTAINING SLOPE AND BEARING DATA

Use this procedure if you are starting at the **upstream transect** (*K*), after completing the thalweg profile and other cross-section measurements at transects *A* through *K*.

1. Stand in the center of the channel at the upstream cross-section transect. Determine if you can see the center of the channel at the next cross-section transect downstream without sighting across land (i.e., do not "short-circuit" a meander bend). If not, you will have to take supplementary slope and bearing measurements.

2. Mark a surveyor's rod and a calibrated rod (or meter ruler) at the same height. If a shorter pole or ruler is used, measure the height from the ground to the opening of the clinometer when it is resting on top.

3. Have one person take the marked surveyor's rod to the downstream transect. Hold the rod vertical with the bottom at the same level as the water surface. If no suitable location is available at the stream margin, position the rod in the water and note the depth.

    - If you have determined in Step 1 that supplemental measurements are required for this segment, walk downstream to the furthest point where you can stand in the center of the channel and still see the center of the channel at the upstream cross-section transect . Remember that your line of sight cannot "cross land." Mark this location with a different color flagging than that marking the cross-section transects.

4. Place the base of the calibrated rod at the level as the surveyor's rod (either at the water surface or at the same depth in the water).

5. Place the clinometer on the calibrated rod at the height determined in Step 2. With the clinometer, sight back downstream to the flagged height on the surveyor's rod at the downstream transect (or at the supplementary point).

    - If you are sighting to the next downstream transect, read and record the **percent** slope in the *MAIN* section on the Slope and Bearing Form for the **downstream transect** (e.g., *J* < *K*), *which is at the **bottom** of the form* (i.e., you are completing the form in reverse order). Record the *PROPORTION* as 100%.

    - If you are backsighting from a supplemental point, record the slope (%) and proportion (%) of the stream segment that is included in the measurement in the appropriate *SUPPLEMENTAL* section of the Slope and Bearing Form. The last sighting to a downstream transect (from either the upstream transect or the nearest upstream supplemental point) is always recorded as the *MAIN* reading.

6. Stand in the middle of the channel at upstream transect (or at a supplemental point), and sight with your compass to the middle of the channel at the downstream transect (or at a supplemental point). Record the bearing (degrees) in the same section of the Slope and Bearing form (Supplemental or Main) as you recorded the slope in Step 6.

7. Proceed to the next cross-section transect (or to a supplementary point), and repeat Steps 3 through 7 above.

the marked pole) at the water's edge or in the same depth of water as you are. The intent is to get a measure of the *water surface* slope, which may not necessarily be the same as the bottom slope.

The clinometer reads both percent slope and degrees of the slope angle; be careful to read and record percent slope. Percent slope is the scale on the **right-hand side** as you look through most clinometers. If using an Abney level, insure that you are reading the scale marked *PERCENT*. With the clinometer or the Abney level, verify this by comparing the two scales. Percent slope is always a higher number than degrees of slope angle (e.g., 100% slope=45° angle). For slopes > 2%, read the clinometer to the nearest 0.5%. For slopes < 2%, read to the nearest 0.25%. If the clinometer reading is 0%, but water is moving, record the slope as 0.1%. If the clinometer reading is 0% and water is not moving, record the slope as 0%.

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

As stated earlier, it may be necessary to set up intermediate (supplemental) slope and bearing points between a pair of cross-section transects if you do not have direct line-of-sight along (and within) the channel between stations (see Figure 7-4). This can happen if brush is too heavy, or if there are sharp slope breaks or tight meander bends. *If you would have to sight across land to measure slope or bearing between two transects, then you need to make one or more supplemental measurements* (i.e., do not "short-circuit" a meander bend). Mark these supplemental locations with a different color of plastic flagging than used for the cross-section transects to avoid confusion. Record these supplemental slope and bearing measurements, along with the proportion of the stream segment between transects included in each supplemental measurement, in the appropriate sections of the Slope and Bearing Form (Figure 7-5). Note that the main slope and bearing observations are always downstream of supplemental observations (i.e., from or to the

downstream transect). Similarly, first supplemental observations are always downstream of second supplemental observations.

Because of ease of use, portability, and cost, EMAP-W used hand-held clinometers to determine slope. Relative precision and accuracy of clinometer measurements decreases when the gradient is low (< 1%). We recognize other organizations may have access to more sophisticated instrumentation (e.g., laser levels), and have field personnel who are experienced in the use of these instruments. The Slope and Bearing Form (Figure 7-5) is designed to allow for different methods and/or different units of measuring slope. Mark the appropriate method box (instead of *CL*; method codes are identified in Tables 7-5 and 7-6), and mark the *CM* box (instead of the % box) if the method or instrument measures the change in elevation rather than the percent slope.

7.5.2 Substrate Size and Channel Dimensions

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

In the EMAP protocol, substrate size and embeddedness are evaluated at each of the 11 cross-section transects (refer to Figure 7-1) using a combination of methods adapted from those described by Wolman (1954), Bain et al. (1985), Platts et al. (1983), and Plafkin et al. (1989). Substrate size is also evaluated at 10 additional cross-sections located midway between each of the 11 regular transects (*A-K*). The basis of the protocol is a systematic selection of 5 substrate particles from each of 21 cross-section transects (Figure 7-6). In the process of measuring substrate particle sizes at each channel cross section, the wetted width of the channel and the water depth at each substrate sample point are measured (at the 10 midway cross-sections, only substrate size and wetted width are recorded). If the wetted channel is split by a mid-channel bar (see Section 7.4.1), the

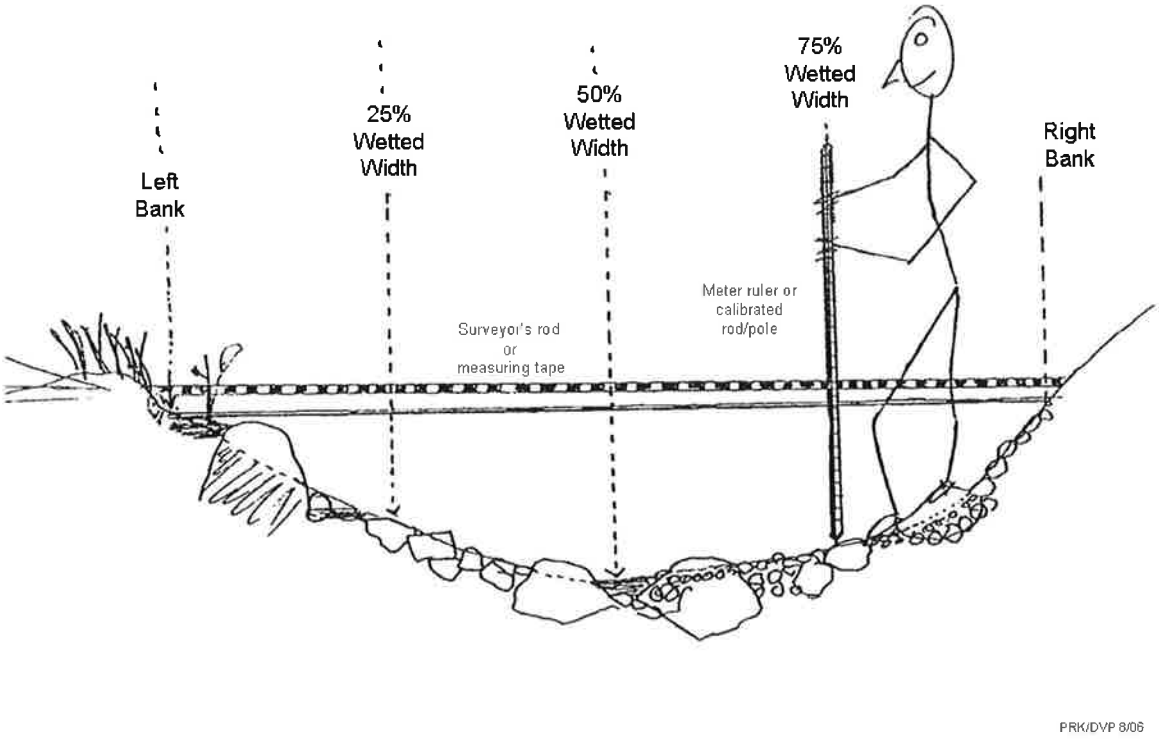


Figure 7-6. Substrate sampling cross-section.

five substrate points are centered between the wetted width boundaries regardless of the mid-channel bar in between. Consequently, substrate particles selected in some cross-sections may be "high and dry". *For cross-sections that are entirely dry, make measurements across the* ***unvegetated portion*** *of the channel.*

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

The procedure for obtaining substrate measurements is described in Table 7-7 (including all particle size classifications). Record these measurements on the Channel/ Riparian Cross-section side of the field form, as shown in Figure 7-7. For the supplemental cross-sections midway between regular transects, record substrate size and wetted width data on the thalweg profile side of the field form. To minimize bias in selecting a substrate particle for size classification, it is important to concentrate on correct placement of the measuring stick along the cross-section, and to select the particle right at the bottom of the stick (not, for example, a more noticeable large particle that is just to the side of the stick). Classify the particle into one of the size classes listed on the field data form (Figure 7-7) based on the middle dimension of its length, width, and depth. This *median dimension* determines the sieve size through which the particle can pass. Always distinguish *hardpan* from *fines*, coding hardpan as *HP*. Similarly, always distinguish concrete or asphalt from bedrock; denote these artificial substrates as *RC* and record their size class in the comments section of the field data form. Code and describe other artificial substrates (including metal, tires, car bodies, etc.) as *Other* (*OT*) on the field form. When you record the size class as *Other*, assign an *Fn* flag on the field data form and describe the substrate type in the comments section of the field form, as shown in Figure 7-7.

At substrate sampling locations on the 11 regular transects (*A-K*), examine particles larger than sand for surface stains, markings, and algal coatings to estimate embeddedness of all particles in the 10 cm diameter circle around the substrate sampling point. Embeddedness is the fraction of a particle's volume that is surrounded by (embedded in) sand or finer sediments on the stream bottom. By definition, record the embeddedness of sand and fines (silt, clay, and muck) as *100 percent*, and record the embeddedness of hardpan and bedrock as *0 percent*.

7.5.3 Bank Characteristics

The procedure for obtaining bank and channel dimension measurements is presented in Table 7-8. Data are recorded in the *BANK MEASUREMENTS* section of the Channel/Riparian Cross-section Form as shown in Figure 7-7. Bank angle and bank undercut distance are determined on the left and right banks at each cross section transect. Figure 7-8 illustrates how bank angle is determined for several different situations. The scale at which bank angle is characterized is approximately 0.5 m. A short (approx. 1-m long) pole is used to determine bank angle. The angle is determined based on the pole resting on the ground for about 0.5 m. Other features include the wetted width

TABLE 7-7. SUBSTRATE MEASUREMENT PROCEDURE

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

   NOTE: If a side channel is present, and contains between 16 and 49% of the total flow, establish a secondary cross-section transect. Use a separate field data form to record data for the side channel, designating it as a secondary transect by marking both the *X-TRY SIDE CHANNEL* box and the associated primary transect letter (e.g., *XA*, *XB*, etc.). Collect all channel and riparian cross-section measurements from the side channel.

2. Divide the wetted channel width channel by 4 to locate substrate measurement points on the cross-section. In the *DISTLB* fields of the form, record the distances corresponding to 0% (*LFT*), 25% (*LCTR*), 50% (*CTR*), 75% (*RCTR*), and 100% (*RGT*) of the measured wetted width. Record these distances at Transects *A-K*, but just the wetted width at midway cross-sections.

3. Place your sharp-ended meter stick or calibrated pole at the *LFT* location (0 m). Measure the depth and record it on the field data form. (Cross-section depths are measured only at regular transects *A-K*, not at the 10 midway cross-sections).

   - Depth entries at the left and right banks may be 0 (zero) if the banks are gradual.
   - If the bank is nearly vertical, let the base of the measuring stick fall to the bottom (i.e., the depth at the bank will be > 0 cm), rather than holding it suspended at the water surface.

4. Pick up the substrate particle that is at the base of the meter stick (unless it is bedrock or boulder), and visually *estimate its particle size*, according to the following table. Classify the particle according to its **median diameter** (the middle dimension of its length, width, and depth). Record the size class code on the field data form. (Cross-section side of form for transects *A-K*; special entry boxes on Thalweg Profile side of form for midway cross-sections.)

Code	Size Class	Size Range (mm)	Description
RS	Bedrock (Smooth)	>4000	Smooth surface rock bigger than a car
RR	Bedrock (Rough)	>4000	Rough surface rock bigger than a car
HP	Hardpan	>4000	Firm, consolidated fine substrate
LB	Boulders (large)	>1000 to 4000	Yard/meter stick to car size
SB	Boulders (small)	>250 to 1000	Basketball to yard/meter stick size
CB	Cobbles	>64 to 250	Tennis ball to basketball size
GC	Gravel (Coarse)	>16 to 64	Marble to tennis ball size
GF	Gravel (Fine)	> 2 to 16	Ladybug to marble size
SA	Sand	>0.06 to 2	Smaller than ladybug size, but visible as particles - gritty between fingers
FN	Fines	≤0.06	Silt Clay Muck (not gritty between fingers)
WD	Wood	Regardless of Size	Wood & other organic particles
RC	Concrete	Regardless of size	Record size class in comment field
OT	Other	Regardless of Size	Metal, tires, car bodies etc. (describe in comments)

(Continued)

TABLE 7-7 (Continued)

5. Evaluate substrate embeddedness as follows at 11 transects *A-K*. For particles larger than sand, examine the surface for stains, markings, and algae. Estimate the average percentage embeddedness of particles in the 10 cm circle around the measuring rod. Record this value on the field data form. By definition, sand and fines are embedded *100%*; bedrock and hardpan are embedded *0%*.

6. Move successively to the next location along the cross section. Repeat Steps 4 through 6 at each location. Repeat Steps 1 through 6 at each new cross section transect, (including any additional side channel transects established when islands are present).

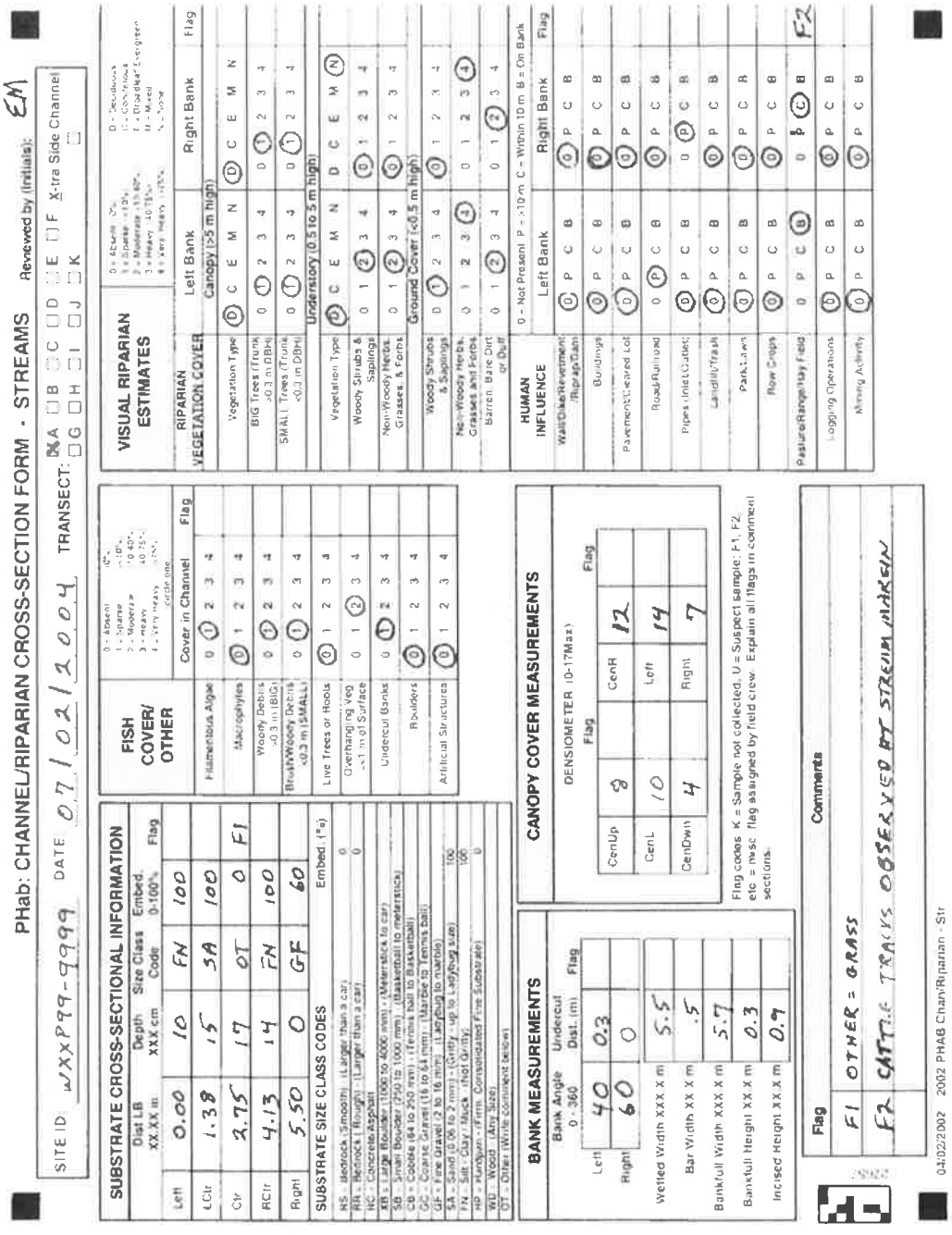


Figure 7-7. Channel/Riparian Cross-section Form for a main channel transect.

TABLE 7-8. PROCEDURE FOR MEASURING BANK CHARACTERISTICS

1. To measure *bank angle*, lay a meter ruler or a short (approx. 1-m long) rod down against the left bank (determined as you face downstream), with one end at the water's edge. At least 0.5 m of the ruler or rod should be *resting comfortably* on the ground to determine bank angle. Lay the clinometer on the rod, and read the bank angle in degrees from the external scale on the clinometer. Record the angle in the field for the left bank in the *BANK MEASUREMENT* section of the Channel/Riparian Cross-section Form.

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

   - If there is a large boulder or log present at the transect, measure bank angle at a nearby point where conditions are more representative.

2. If the bank is *undercut*, measure the horizontal distance of the undercutting to the nearest 0.01 m. The undercut distance is the distance from the water's edge out to the point where a vertical plumb line from the bank would hit the water's surface. Record the distance on the field data form. Measure submerged undercuts by thrusting the rod into the undercut and reading the length of the rod that is hidden by the undercutting.

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

4. Hold the surveyor's rod vertical, with its base planted at the water's edge. Examine both banks, then determine the channel *incision* as the *height up from the water surface to elevation of the* ***first terrace*** *of the valley floodplain* (Note this is at or above the bankfull channel height). Whenever possible, use the clinometer as a level (positioned so it reads 0% slope) to measure this height by transferring (back-sighting) it onto the surveyor's rod. Record this value in the *INCISED HEIGHT* field of the bank measurement section on the field data form.

5. While still holding the surveyor's rod as a guide, and sighting with the clinometer as a level, examine both banks to measure and record the *height of bankfull flow above the present water level*. Look for evidence on one or both banks such as:

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

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

7. Repeat Steps 1 through 6 at each cross-section transect, (including any additional side channel transects established when islands are present). Record data for each transect on a separate field data form.

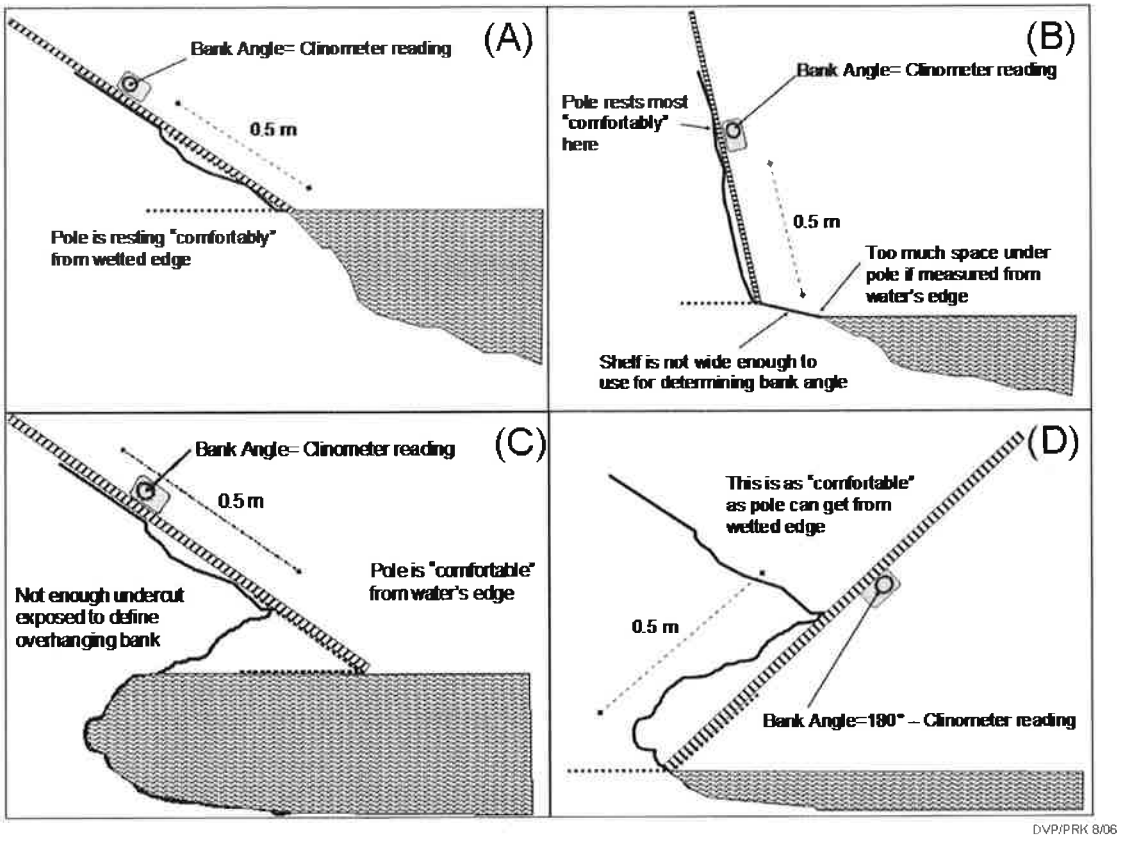


Figure 7-8. Determining bank angle under different types of bank conditions. (A) typical, (B) incised channel, 9C) undercut bank, and (D) overhanging bank.

of the channel (as determined in Section 7.5.2), the width of exposed mid-channel bars of gravel or sand, estimated incision height, and the estimated height and width of the channel at bankfull stage as described in Table 7-8. *Bankfull height* and *incised height* are both measured relative to the present water surface. In other words, both are measured up from the level of the wetted edge of the stream. This is done by placing the base of the small measuring rod at the bankfull elevation and sighting back to the survey rod placed at the water's edge using the clinometer as a level (i.e., positioned so the slope reading is *0%*.). The height of the clinometer above the base of the smaller rod is subtracted from the elevation sighted on the surveyor's rod.

Bankfull flows are large enough to erode the stream bottom and banks, but frequent enough (every 1 to 2 years) to not allow substantial growth of upland terrestrial vegetation.

Consequently, in many regions, it is these flows that have determined the width and depth of the channel. Estimates of the bankfull dimensions of stream channels are extremely important in EMAP surveys. They are used to calculate shear stress and bed stability (see Kaufmann et al., 1999). Unfortunately, we have to depend upon evidence visible during the low-flow sampling season. If available, consult published rating curves relating expected bankfull channel dimensions to stream drainage area within the region of interest. Graphs of these rating curves can help you get a rough idea of where to look for field evidence to determine the level of bankfull flows. Curves such as these are available from the USGS for streams in most regions of the U.S. (e.g., Dunne and Leopold 1978; Harrelson et al. 1994, Leopold 1994). To use them, you need to know the contributing drainage area to your sample site. Interpret the expected bankfull levels from these curves as a height above the streambed in a riffle, but remember that your field measurement will be a height above the present water surface of the stream. Useful resources to aid your determination of bankfull flow levels in streams in the United States are video presentations produced by the USDA Forest Service for western streams (USDA Forest Service 1995) and eastern streams (USDA Forest Service 2002).

After consulting rating curves that show where to expect bankfull levels in a given size of stream, estimate the bankfull flow level by looking at the following indicators:

- First look at the stream and its valley to determine the active floodplain. This is a depositional surface that frequently is flooded and experiences sediment deposition under the current climate and hydrological regime.
- Then look specifically for:
  - An obvious break in the slope of the banks.
  - A change from water-loving and scour-tolerant vegetation to more drought-tolerant vegetation.
  - A change from well-sorted stream sediments to unsorted soil materials.

In the absence of clear bankfull indications, consider the previous season's flooding as the best evidence available (note: you could be wrong if very large floods or prolonged droughts have occurred in recent years.). Look for:

- Drift debris ("sticky wickets" left by the previous seasons flooding).
- The level where deciduous leaf-fall is absent on the ground (carried away by previous winter flooding).

- Unvegetated sand, gravel or mud deposits from previous year's flooding.

In years that have experienced large floods, drift material and other recent high flow markers may be much higher than other bankfull indicators. In such cases, base your determination on less-transient indicators such as channel form, perennial vegetation, and depositional features. In these cases, flag your data entry and also record the height of drift material in the comments section of the field data form.

We use the vertical distance (height) from the observed water surface up to the level of the first major valley depositional surface (Figure 7-9) as a measure of the degree of *incision* or *downcutting* of the stream below the general level of its valley. This value is recorded in the *INCISED HEIGHT* field. (Note: In analyzing these data, we actually add the mean thalweg depth to incision heights to yield a flow-independent measure– we do the same thing for bankfull heights). If the first depositional surface is no longer active (i.e., frequently inundated by floods), it is called a *terrace* or an *abandoned floodplain*. Streams incise when their rate of sediment transport exceeds the supply of new sediment from upstream and from their banks. Conversely, *aggradation* occurs when sediment supplies exceed the capacity of the stream to transport sediment. Human activities can change the balance between sediment transport and supply in a number of ways. The power of the stream to transport sediment may be increased by human activities that in-crease flood flows (e.g., increases in the impervious area of a watershed), or remove large roughness elements like woody debris that dissipate stream power that might otherwise transport sediment. The supply of sediment may be increased by upslope erosion, or de-creased when, for example, upstream impoundments trap bedload sediments. It may not be evident at the time of sampling whether the channel is downcutting, stable, or aggrading (raising its bed by depositing sediment). However, by recording incision heights measured in this way and monitoring them over time, we will be able to tell if streams are incising or aggrading.

If the channel is not greatly incised, bankfull channel height and incision height will be the same (i.e., the first valley depositional surface is the active floodplain). However, if the channel is incised greatly, the bankfull level will be below the level of the first terrace of the valley floodplain, making bankfull channel height less than incision height (Figure 7-10). *Bankfull height is **never** greater than incision height.* You may need to look for evidence of recent flows (within about one year) to distinguish bankfull and incision heights. In cases where the channel is cutting a valley sideslope and has oversteepened and destabilized

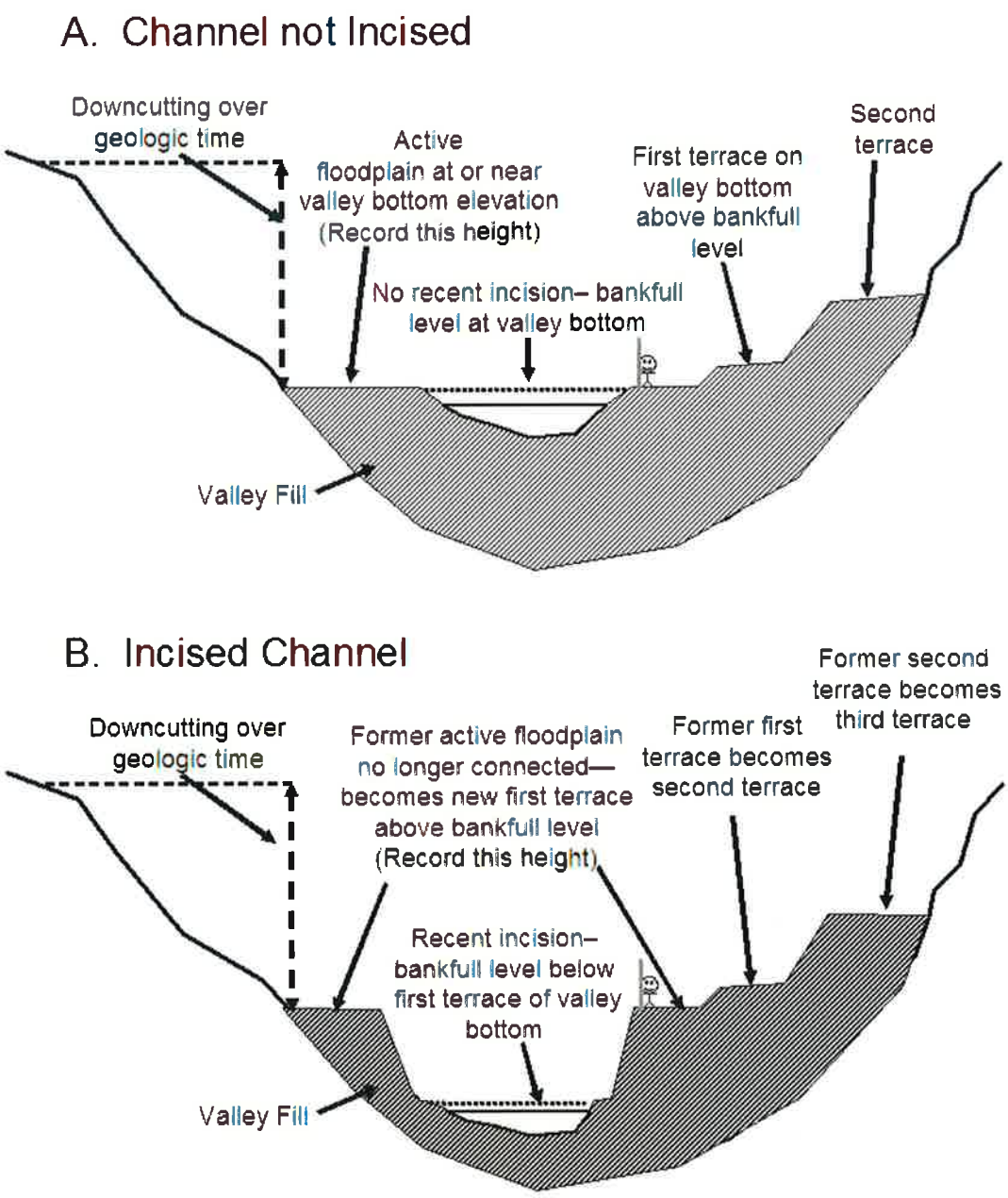


Figure 7-9. Schematic showing relationship between bankfull channel and incision. (A) not recently incised, and (B) recently incised into valley bottom. Note level of bankfull stage relative to elevation of first terrace (abandoned floodplain)on valley bottom. (Stick figure included for scale).

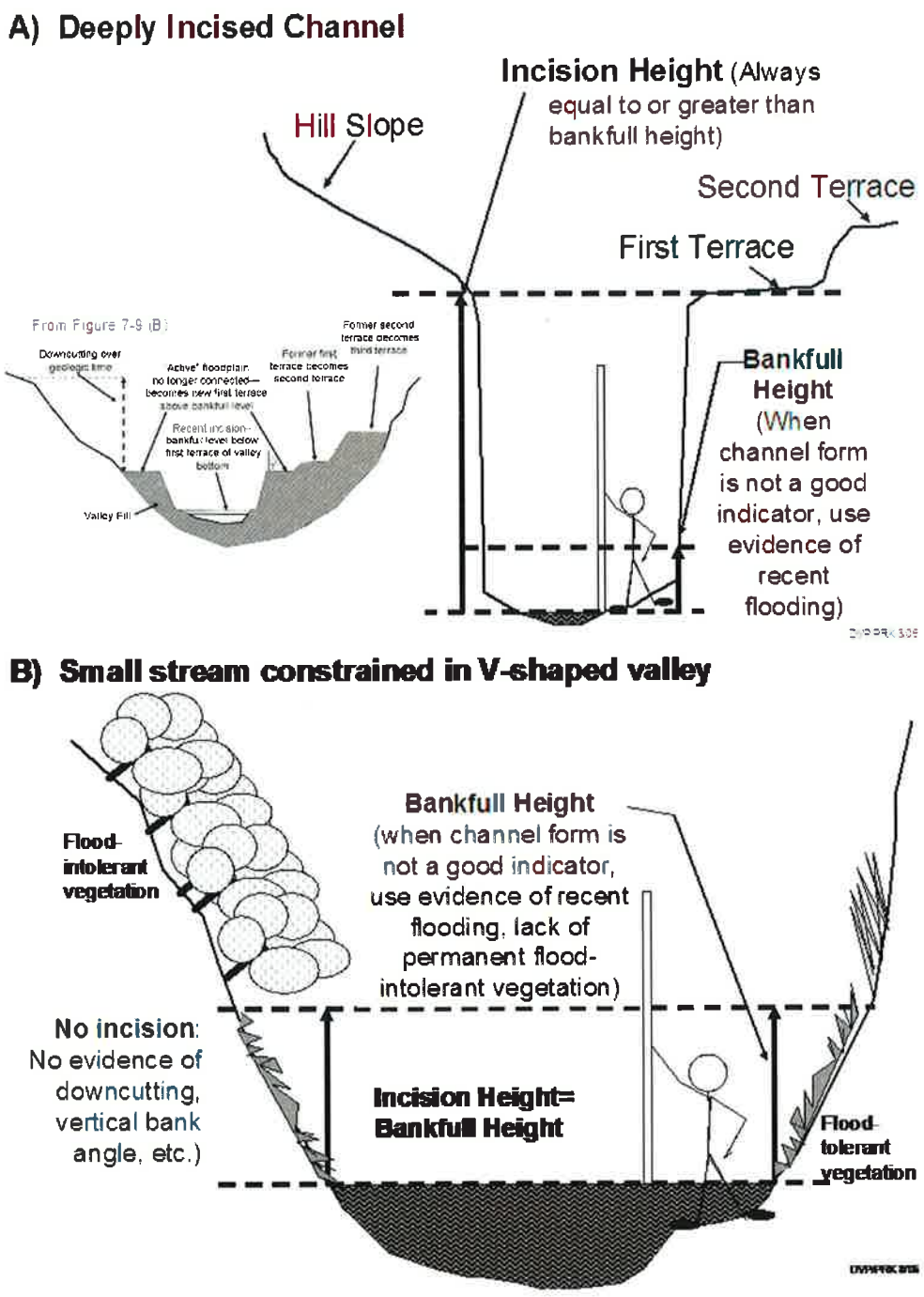


Figure 7-10. Determining bankfull and incision heights for (A) deeply incised channels, and (B) streams in deep V-shaped valleys. (Stick figure included for scale).

that slope, the bare "cutbank" against the steep hillside at the edge of the valley is not necessarily an indication of recent incision. In such a case, the opposite bank may be lower, with a more obvious terrace above bankfull height; choose that bank for your measurement of incised height. Examine both banks to more accurately determine incision height and bankfull height. Remember that incision height is measured as *the vertical distance to the first major depositional surface above bankfull* (whether or not it is an active floodplain or a terrace. If terrace heights differ on left and right banks (both are above bankfull), choose the **lower** of the two terraces. In many cases your sample reach may be in a "V" shaped valley or gorge formed over eons, and the slope of the channel banks simply extends uphill indefinitely, not reaching a terrace before reaching the top of a ridge (Figure 7-10). In such cases, record incision height values equal to bankfull values and make appropriate comment that no terrace is evident. Similarly, when the stream has extremely incised into an ancient terrace, (e.g., the Colorado River in the Grand Canyon), you may crudely estimate the terrace height if it is the first one above bankfull level. If you cannot estimate the terrace height, make appropriate comments describing the situation.

7.5.4 Canopy Cover Measurements

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

Canopy cover over the stream is determined at each of the 11 cross-section transects. A spherical densiometer (model *A*- **convex type**) is used (Lemmon 1957). Mark the densiometer with a permanent marker or tape exactly as shown in Figure 7-11 to limit the number of square grid intersections read to 17. Densiometer readings can range from 0 (no canopy cover) to 17 (maximum canopy cover). Six measurements are obtained at each cross-section transect (four measurements in each of four directions at mid-channel and one at each bank). The mid-channel measurements are used to estimate canopy cover over the channel. The two bank measurements complement your visual estimates of vegetation structure and cover within the riparian zone itself (Section 7.5.5), and are particularly important in wide streams, where the riparian canopy may not be detected by the densiometer when standing midstream.

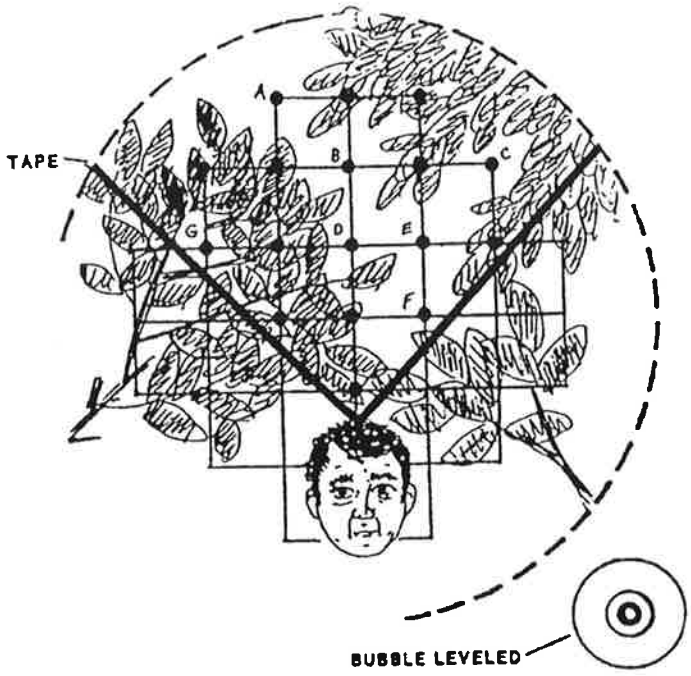


Figure 7-11. Schematic of modified convex spherical canopy densiometer. From Mulvey et al. (1992). Note proper positioning with the bubble leveled and face reflected at the apex of the "V". In this example, 10 of the 17 intersections show canopy cover, giving a densiometer reading of *10*.

The procedure for obtaining canopy cover data is presented in Table 7-9. Densiometer measurements are taken at 0.3 m (1 ft) above the water surface, rather than at waist level, to (1) avoid errors because people differ in height; (2) avoid errors from standing in water of varying depths; and (3) include low overhanging vegetation more consistently in the estimates of cover. Hold the densiometer level (using the bubble level) 0.3 m above the water surface with your face reflected just below the apex of the taped "V", as shown in Figure 7-11. Concentrate on the 17 points of grid intersection on the densiometer that lie within the taped "V". If the reflection of a tree or high branch or leaf overlies any of the intersection points, that particular intersection is counted as having cover. For each of the six measurement points, record the number of intersection points (maximum=17) that have vegetation covering them in the *CANOPY COVER MEASUREMENT* section of the Channel/ Riparian Cross-section Form as shown in Figure 7-7.

TABLE 7-9. PROCEDURE FOR CANOPY COVER MEASUREMENTS

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

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

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

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

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

6. Move to the water's edge (either the left or right bank). Repeat Steps 2 and 3 again, this time facing the bank . Record the value in the *LFT* or *RGT* fields of the field data form. Move to the opposite bank and repeat.

7. Repeat Steps 1 through 6 at each cross-section transect (including any additional side channel transects established when islands are present). Record data for each transect on a separate field data form.

7.5.5 Riparian Vegetation Structure

The previous section (7.5.4) described methods for quantifying the cover of canopy over the stream channel. The following visual estimation procedures supplement those measurements with a semi-quantitative evaluation of the type and amount of various types of riparian vegetation. These data are used to evaluate the health and level of disturbance of the stream corridor. They also provide an indication of the present and future potential for various types of organic inputs and shading. Additional measures within the riparian zone (legacy trees and invasive riparian plants) are described in Sections 7.5.9 and 8, respectively.

Riparian vegetation observations apply to the riparian area upstream 5 meters and downstream 5 meters from each of the 11 cross-section transects (refer to Figure 7-1). They include the visible area from the stream back a distance of 10m (~30 ft) shoreward from both the left and right banks, creating a 10 m × 10 m riparian plot on each side of the stream (Figure 7-12). The riparian plot dimensions are estimated, not measured. On steeply sloping channel margins, the 10 m × 10 m plot boundaries are defined as if they were projected down from an aerial view.

Table 7-10 presents the procedure for characterizing riparian vegetation structure and composition. Figure 7-7 illustrates how measurement data are recorded in the *VISUAL RIPARIAN ESTIMATES* section of the Channel/Riparian Cross-section Form. Conceptually divide the riparian vegetation into three layers: the *Canopy* layer (> 5 m high), the *Understory* layer (0.5 to 5 m high), and the *Ground cover* layer (< 0.5 m high). Note that several vegetation types (e.g., grasses or woody shrubs) can potentially occur in more than one layer. Similarly note that some things other than vegetation are possible entries for the *Ground cover* layer (e.g., barren ground).

Before estimating the areal coverage of the vegetation layers, record the type of *woody* vegetation (*broadleaf **D**eciduous*, ***C**oniferous*, *broadleaf **E**vergreen*, ***M**ixed*, or ***N**one*) in each of the two taller layers (Canopy and Understory). Consider the layer *Mixed* if more than 10% of the areal coverage is made up of the alternate vegetation type. If there is no woody vegetation in the understory layer, record the type as *None*.

Estimate the areal cover separately in each of the three vegetation layers. Note that the areal cover can be thought of as the amount of shadow cast by a particular layer

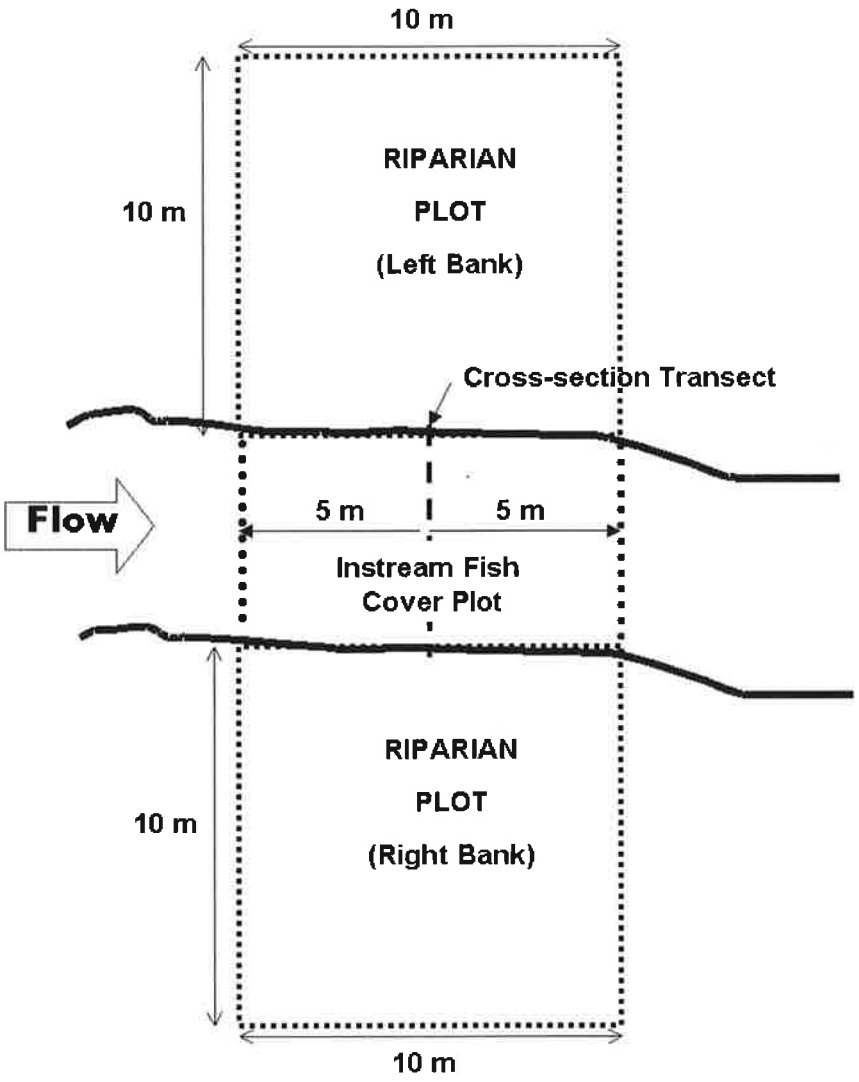


Figure 7-12. Riparian zone and instream fish cover plots for a stream cross-section transect..

TABLE 7-10. PROCEDURE FOR CHARACTERIZING RIPARIAN VEGETATION STRUCTURE

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

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

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

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

4. Within this 10 m × 10 m area, determine the dominant vegetation type for the *CANOPY LAYER* (vegetation >5 m high) as either ***D**eciduous*, ***C**oniferous*, *broadleaf **E**vergreen*, ***M**ixed*, or ***N**one*. Consider the layer *Mixed* if more than 10% of the areal coverage is made up of the alternate vegetation type. Indicate the appropriate vegetation type in the *VISUAL RIPARIAN ESTIMATES* section of the Channel/Riparian Cross-section Form.

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

6. Look at the *UNDERSTORY* layer (vegetation between 0.5 and 5 m high). Determine the dominant **woody** vegetation type for the understory layer as described in Step 4 for the canopy layer. If there is no woody vegetation in the understory layer, record the type as ***N**one*.

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

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

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

10. Repeat Steps 1 through 9 for all cross-section transects (including any additional side channel transects established when islands are present). Use a separate field data form for each transect.

alone when the sun is directly overhead. *The maximum cover in each layer is 100%, so the sum of the areal covers for the combined three layers could add up to 300%.* The four areal cover classes are *Absent*, *Sparse* (<10%), *Moderate* (10 to 40%), *Heavy* (40 to 75%), and *Very Heavy* (>75%). These cover classes and their corresponding codes are shown on the field data form (Figure 7-7). When rating vegetation cover types for a single vegetation layer, mixtures of two or more subdominant classes might all be given *Sparse* (*1*), *Moderate* (*2*), or *Heavy* (*3*) ratings. One *Very Heavy* cover class with no clear sub-dominant class might be rated *4* with all the remaining classes rated as either *Moderate* (*2*), *Sparse* (*1*) or *Absent* (*0*). Note that within a given vegetation layer, two cover types with 40-75% cover can both be rated *3*, but no more than one cover type could receive a rating of *4*.

7.5.5 Instream Fish Cover, Algae, and Aquatic Macrophytes

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

The procedure to estimate the types and amounts of instream fish cover is outlined in Table 7-11. Data are recorded in the *FISH COVER/OTHER* section of the Channel/ Riparian Cross-section Form as shown in Figure 7-7. Estimate the areal cover of all of the fish cover and other listed features that are in the water and on the banks 5 m upstream and downstream of the cross-section (see Figure 7-12). The areal cover classes of fish concealment and other features are the same as those described for riparian vegetation (Section 7.5.5).

The entry *FILAMENTOUS ALGAE* refers to long streaming algae that often occur in slow moving waters. *AQUATIC MACROPHYTES* are water-loving plants, including mosses, in the stream that could provide cover for fish or macroinvertebrates. If the stream channel contains live wetland grasses, include these as aquatic macrophytes. *WOODY DEBRIS* are the larger pieces of wood that can influence cover and stream morphology (i.e., those pieces that would be included in the large woody debris tally [Section 7.4]). *BRUSH/WOODY DEBRIS* refers to smaller wood pieces that primarily affect cover but not morphology. *LIVE TREES OR ROOTS* are living trees that are within the channel – estimate the areal cover provided by the parts of these trees or roots that are inundated. *OVERHANGING VEGETATION*

TABLE 7-11. PROCEDURE FOR ESTIMATING INSTREAM FISH COVER

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

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

3. For each cover type, estimate the areal cover. Record the appropriate cover class in the *FISH COVER/OTHER* section of the Channel/Riparian Cross-section Form:

   0=absent: zero cover,
   1=sparse: <10%,
   2=moderate: 10-40%,
   3=heavy: >40-75%, or
   4=very heavy: >75%).

4. Repeat Steps 1 through 3 at each cross-section transect (including any additional side channel transects established when islands are present). Record data from each transect on a separate field data form.

includes tree branches, brush, twigs, or other small debris that is not in the water but is close to the stream (within 1 m of the surface) and provides potential cover. *BOULDERS* are typically basketball- to car-sized particles. *ARTIFICIAL STRUCTURES* include those designed for fish habitat enhancement, as well as in-channel structures that have been discarded (e.g., concrete, asphalt, cars, or tires) or deliberately placed for diversion, impoundment, channel stabilization, or other purposes.

7.5.7 Human Influence

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

For the left and right banks at each of the 11 detailed Channel and Riparian Cross-sections, evaluate the presence/absence and the proximity of 11 categories of human influences with the procedure outlined in Table 7-12. Relate your observations and proximity evaluations to the stream and riparian area within 5 m upstream and 5 m downstream from the station (Figure 7-12). Four proximity classes are used: In the stream or on the bank within 5 m upstream or downstream of the cross-section transect, present within the 10 m × 10 m riparian plot but not in the stream or on the bank, present outside of the riparian plot, and absent. Record data on the Channel/Riparian Cross-section Form as shown in Figure 7-7. If a disturbance is within more than one proximity class, record the one that is closest to the stream (e.g., *C* takes precedence over *P*).

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

7.5.8 Cross-section Transects on Side Channels

If the wetted channel is split by an island, and the estimated flow in the side channel is less than or equal to 15% of the total flow, the bank and riparian measurements are made at each side of the main channel (the minor side channel is ignored other than to note its presence on the thalweg profile form), so one riparian plot is established on the island as shown in Figure 7-13. If an island is present that creates a major side channel

TABLE 7-12. PROCEDURE FOR ESTIMATING HUMAN INFLUENCE

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

2. Examine the channel, bank and riparian plot area adjacent to the defined stream segment for the following human influences: (1) *walls, dikes, revetments, riprap, and dams*; (2) *buildings*; (3) *pavement/cleared lots* (e.g., paved, gravelled, dirt parking lot, foundation); (4) *roads or railroads*, (5) *inlet or outlet pipes*; (6) *landfills or trash* (e.g., cans, bottles, trash heaps); (7) *parks or maintained lawns*; (8) *row crops*; (9) *pastures, rangeland, hay fields*, or *evidence of livestock*; (10) *logging*; and (11) *mining* (including gravel mining).

3. For each type of influence, determine if it is present and what its proximity is to the stream and riparian plot area. Consider human disturbance items as present if you can see them from the cross-section transect. Do not include them if you have to sight through another transect or its 10 m ×10 m riparian plot.

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

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

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

6. Repeat Steps 1 through 5 for each cross-section transect, (including any additional side channel transects established when islands are present). Record data for each transect on a separate field form.

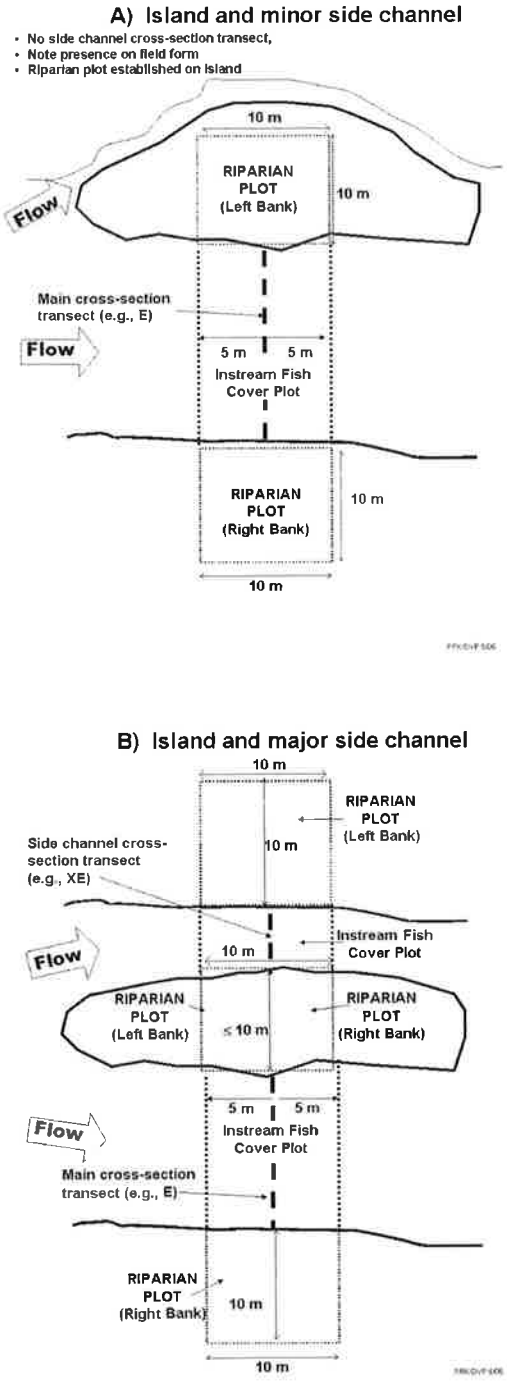


Figure 7-13. Riparian and instream fish cover plots for a streams with minor and major side channels.

containing **more than 15%** of the total flow (Section 7.5.1), an additional cross-section transect is established for the side channel as shown in Figure 7-13. Separate substrate, bank and riparian measurements are made for side channel transects. Data from the additional side channel transect are recorded on a separate Channel/Riparian Cross-section Form as shown in Figure 7-14. Riparian plots established on the island for each transect may overlap (and be < 10 m shoreward) if the island is less than 10 m wide at the transect.

7.5.9 Riparian "Legacy" Trees

The Riparian "Legacy" Tree procedure contributes to the assessment of "old growth" characteristics of riparian vegetation, and aids the determination of possible historic conditions and the potential for riparian tree growth. Follow the procedures presented in Table 7-13 to locate the **largest** tree associated with each transect. The tree you choose may not truly be an old *legacy* tree – just choose the largest you see. We use these data to determine if there are true legacy trees somewhere within the support reach. Note that only one tree is identified for each transect between that transect and the next one upstream; at transect *K*, look upstream a distance of 4 channel widths. Record the type of tree, and, if possible, the taxonomic group (using the list provided in Table 7-13) on the left-hand column of the Riparian "Legacy" Trees and Invasive Alien Plants form (Figure 7-15). Estimate the height of the tree and the diameter at breast height (dbh), and mark the appropriate height and dbh classes on the form. Estimate and record the distance of the legacy tree from the wetted margin of the stream.

7.6 CHANNEL CONSTRAINT, DEBRIS TORRENTS, AND RECENT FLOODS

7.6.1 Channel Constraint

Whether natural or the result of human activities, the presence of immovable or non-erodable river margins constrains the degree to which the stream can form its own channel and banks through scour and deposition. The degree of channel constraint can strongly influence the quantity and quality of habitat for aquatic organisms. Constraint also influences the type and degree of stream channel adjustment to anthropogenic alterations in flow and sediment supply, or to direct channel manipulations (e.g., dredging, revetments, impoundment). To assess overall reach channel constraint, we have modified methods

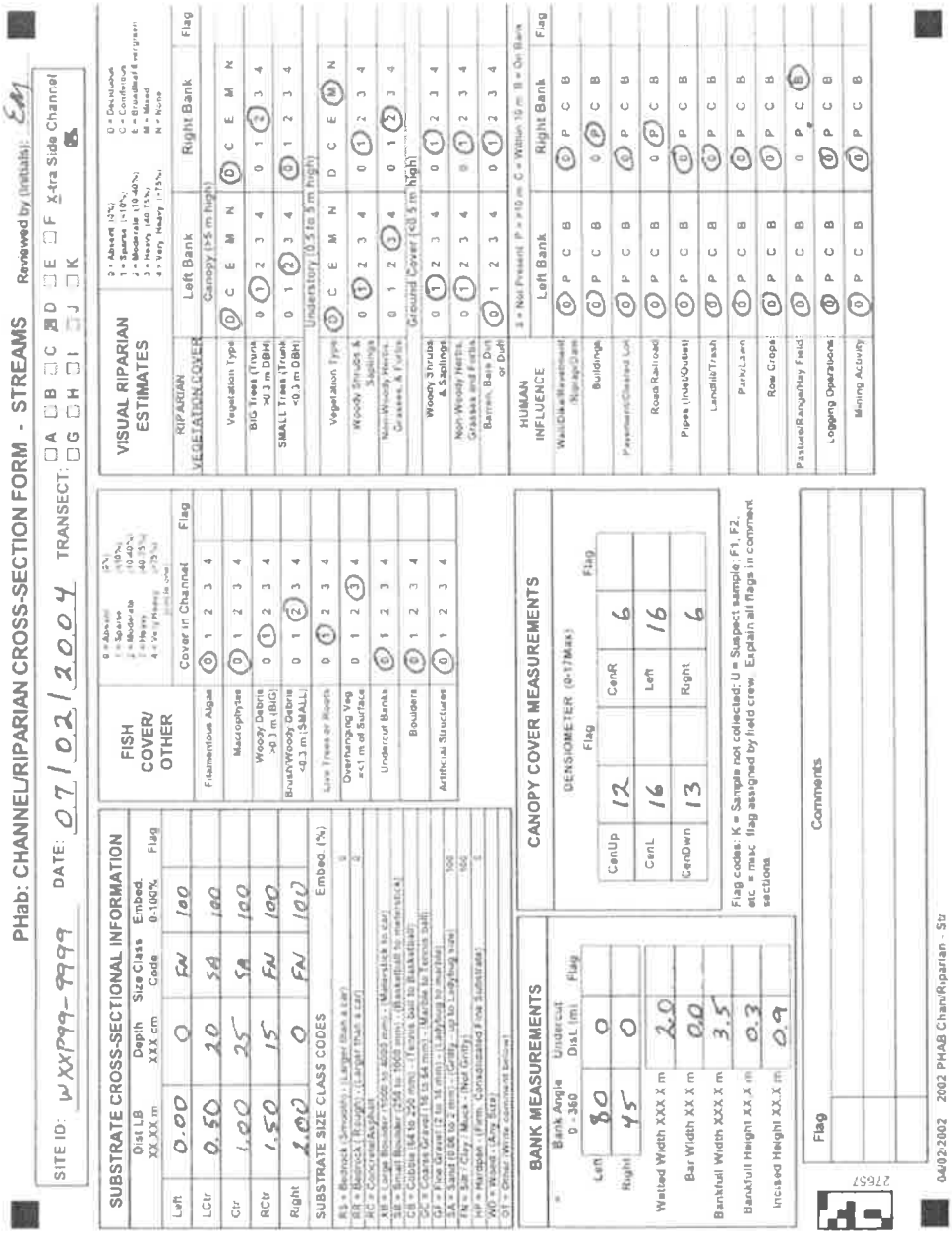


Figure 7-14. Channel/Riparian Cross-section Form for an additional major side channel transect.

TABLE 7-13. PROCEDURE FOR IDENTIFYING RIPARIAN LEGACY TREES

1. Beginning at Transect *A*, look upstream. Search both sides of the stream upstream to the next transect. At Transect *K*, look upstream for a distance of 4 channel widths. Locate the largest tree visible within 50 m (or as far as you can see, if less) from the wetted bank (note the tree you identify may be outside the current riparian zone).

2. Classify this tree as deciduous, coniferous, or broadleaf evergreen (classify western larch as coniferous). Identify, if possible, the species or the taxonomic group of this tree from the list below.

    1. Acacia/Mesquite
    2. Alder/Birch
    3. Ash
    4. Cedar/Cypress/Sequoia
    5. Fir (including Douglas Fir, Hemlock)
    6. Juniper
    7. Maple/Boxelder
    8. Oak
    9. Pine
    10. Poplar/Cottonwood
    11. Snag (Dead Tree of Any Species)
    12. Spruce
    13. Sycamore
    14. Willow
    15. Unknown or Other Broadleaf Evergreen
    16. Unknown or Other Conifer
    17. Unknown or Other Deciduous

    NOTE: If the largest tree is a dead snag, enter *Snag* as the taxonomic group.

3. Estimate the height of the potential legacy tree, its diameter at breast height (dbh) and its distance from the wetted margin of the stream. Enter this information on the left-hand column of the Riparian "Legacy" Trees and Invasive Alien Plants field form.

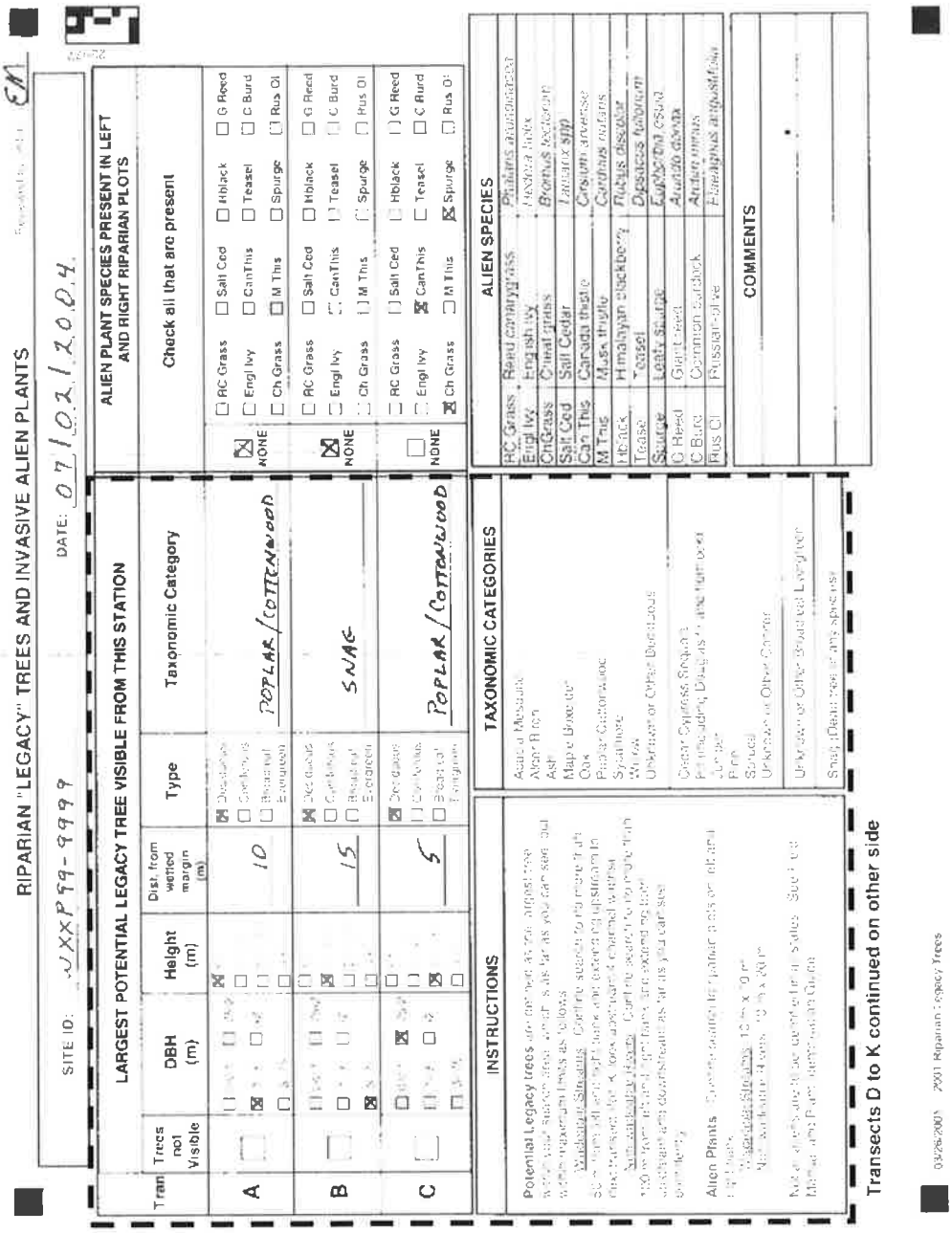


Figure 7-15. Riparian "Legacy" Tree and Invasive Alien Plants Form (Page 1), showing data for riparian legacy trees.

used by Oregon Department of Fish and Wildlife in their Aquatic Inventories (Moore et al. 1993).

After completing the thalweg profile and riparian/channel cross-section measurements and observations, envision the stream at bankfull flow and evaluate the degree, extent and type of channel constraint, using the procedures presented in Table 7-14. Record data on the Channel Constraint Assessment Form (Figure 7-16). First, classify the stream reach channel pattern as predominantly a *single channel*, an *anastomosing channel*, or a *braided channel* (Figure 7-17):

1. *Single channels* may have occasional in-channel bars or islands with side channels, but feature a *predominant single channel*, or a *dominant main channel with a subordinate side channel*.

2. *Anastomosing channels* have relatively long *major and minor channels* (but no predominant channel) in a complex network, diverging and converging around many *vegetated islands*. Complex channel pattern remains even during major floods.

3. *Braided channels* also have multiple branching and rejoining channels, (but no predominant channel) *separated by unvegetated bars*. Channels are generally smaller, shorter, and more numerous, often with no obvious dominant channel. During major floods, a single continuous channel may develop

After classifying the channel pattern, determine whether the channel is constrained within a narrow valley, constrained by local features within a broad valley, unconstrained and free to move about within a broad floodplain, or free to move about, but within a relatively narrow valley floor. Then examine the channel to ascertain the bank and valley features that constrain the stream. Entry choices for the type of constraining features are bedrock, hillslopes, terraces/alluvial fans, and human land use (e.g., a road, a dike, landfill, rip-rap, etc.). Estimate the percent of the channel margin in contact with constraining features (for unconstrained channels, this is 0%). To aid in this estimate, you may wish to refer to the individual transect assessments of incision and constraint. Finally, estimate the "typical" bankfull channel width and visually estimate the average width of the valley floor. If you cannot directly estimate the valley width (e.g., it is further than you can see, or if your

TABLE 7-14. PROCEDURES FOR ASSESSING CHANNEL CONSTRAINT

NOTE: These activities are conducted after completing the thalweg profile and littoral-riparian measurements and observations, and represent an evaluation of the entire stream reach.

CHANNEL CONSTRAINT: Determine the degree, extent, and type of channel constraint based on envisioning the stream at **bankfull flow**.

1. Classify the stream reach channel pattern as predominantly a **single** channel, an **anastomosing** channel, or a **braided** channel.

   Single channels *may have occasional in-channel bars or islands with side channels*, but feature a predominant single channel, or a dominant main channel with a subordinate side channel.

   Anastomosing channels *have relatively long major and minor channels* branching and rejoining in a complex network separated by vegetated islands, with no obvious dominant channel.

   Braided channels *also have multiple branching and rejoining channels,* separated by unvegetated bars. Subchannels are generally small, short, and numerous, often with no obvious dominant channel.

2. After classifying the channel pattern, determine whether the channel is constrained within a narrow valley, constrained by local features within a broad valley, unconstrained and free to move about within a broad floodplain, or free to move about, but within a relatively narrow valley floor.

3. Then examine the channel to ascertain the bank and valley features that constrain the stream. Entry choices for the type of constraining features are bedrock, hillslopes, terraces/alluvial fans, and human land use (e.g., a road, a dike, landfill, rip-rap, etc.).

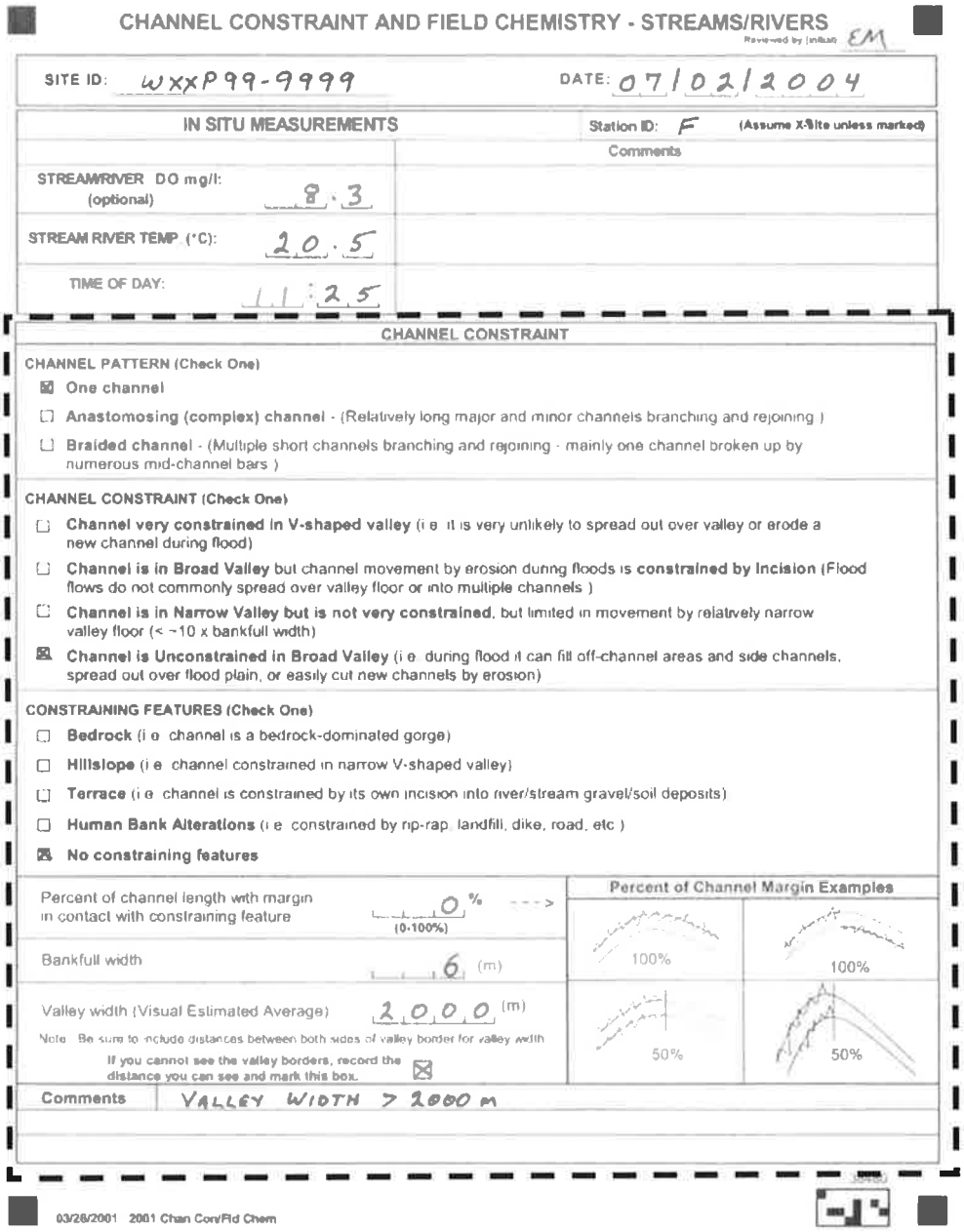
4. Based on your determinations from Steps 1 through 3, select and record one of the constraint classes shown on the Channel Constraint Form.

5. Estimate the percent of the channel margin in contact with constraining features (for unconstrained channels, this is 0%). Record this value on the Channel Constraint Form.

6. Finally, estimate the "typical" bankfull channel width, and visually estimate the average width of the valley floor. Record these values on the Channel Constraint Form.

   NOTE: To aid in this estimate, you may wish to refer to the individual transect assessments of incision and constraint that were recorded on the Channel/Riparian Cross-Section Forms.

   NOTE: If the valley is wider than you can directly estimate, record the distance you can see and mark the box on the field form.



CHANNEL CONSTRAINT AND FIELD CHEMISTRY - STREAMS/RIVERS

Reviewed by (initials) EM

SITE ID: WXXP99-9999 DATE: 07/02/2004

IN SITU MEASUREMENTS Station ID: F (Assume X-Site unless marked)

		Comments
STREAM/RIVER DO mg/l: (optional)	8.3	
STREAM RIVER TEMP (°C):	20.5	
TIME OF DAY:	11:25	

CHANNEL CONSTRAINT

CHANNEL PATTERN (Check One)

- [x] **One channel**
- [ ] **Anastomosing (complex) channel** - (Relatively long major and minor channels branching and rejoining)
- [ ] **Braided channel** - (Multiple short channels branching and rejoining - mainly one channel broken up by numerous mid-channel bars)

CHANNEL CONSTRAINT (Check One)

- [ ] **Channel very constrained in V-shaped valley** (i e it is very unlikely to spread out over valley or erode a new channel during flood)
- [ ] **Channel is in Broad Valley** but channel movement by erosion during floods is **constrained by incision** (Flood flows do not commonly spread over valley floor or into multiple channels)
- [ ] **Channel is in Narrow Valley but is not very constrained**, but limited in movement by relatively narrow valley floor (< ~10 x bankfull width)
- [x] **Channel is Unconstrained in Broad Valley** (i e during flood it can fill off-channel areas and side channels, spread out over flood plain, or easily cut new channels by erosion)

CONSTRAINING FEATURES (Check One)

- [ ] **Bedrock** (i e channel is a bedrock-dominated gorge)
- [ ] **Hillslope** (i e channel constrained in narrow V-shaped valley)
- [ ] **Terrace** (i e channel is constrained by its own incision into river/stream gravel/soil deposits)
- [ ] **Human Bank Alterations** (i e constrained by rip-rap, landfill, dike, road, etc)
- [x] **No constraining features**

		Percent of Channel Margin Examples
Percent of channel length with margin in contact with constraining feature (0-100%)	0 % --->	100% / 100%
Bankfull width	6 (m)	
Valley width (Visual Estimated Average)	2000 (m)	50% / 50%

Note Be sure to include distances between both sides of valley border for valley width

If you cannot see the valley borders, record the distance you can see and mark this box. [x]

Comments: VALLEY WIDTH > 2000 m

03/26/2001 2001 Chan Con/Fld Chem

Figure 7-16. Channel Constraint and Field Chemistry Form, showing data for channel constraint.

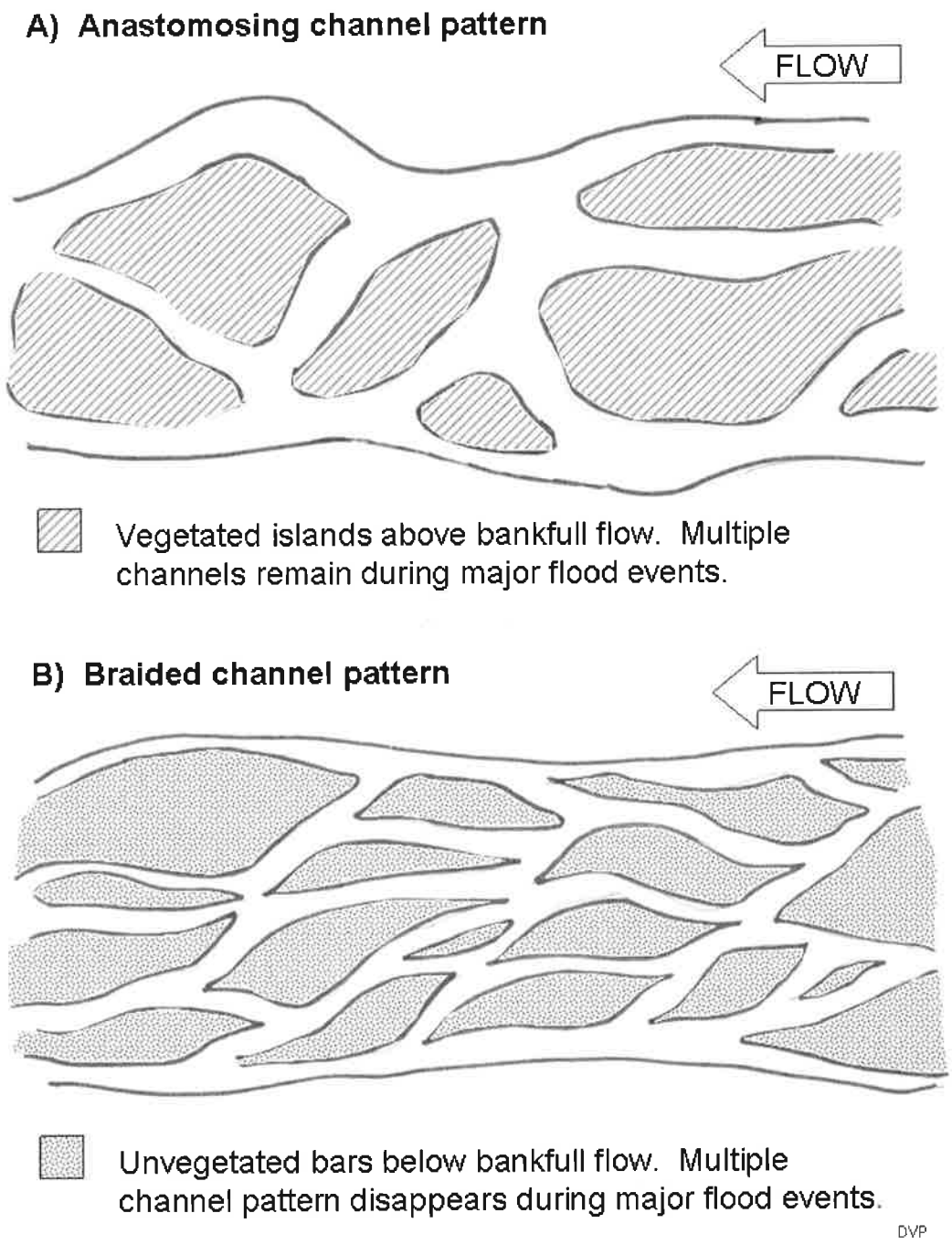


Figure 7-17. Types of multiple channel patterns.

view is blocked by vegetation), record the distance you can see and mark the appropriate box on the field form.

7.6.2 Debris Torrents and Recent Major Floods

Major floods are those that substantially overtop the banks of streams and occur with an average frequency of less than once every five years. Major floods may scour away or damage riparian vegetation on banks and gravel bars that are not frequently inundated. They typically cause movement of large woody debris, transport of bedload sediment, and changes in the streambed and banks through scouring and deposition. While they may kill aquatic organisms and temporarily suppress their populations, floods are an important natural resetting mechanism that maintains habitat volume, clean substrates, and riparian productivity.

Debris torrents, or *lahars*, differ from conventional floods in that they are flood waves of higher magnitude and shorter duration, and their flow consists of a dense mixture of water and debris. Their high flows of dense material exert tremendous scouring forces on streambeds. For example, in the Pacific Northwest, flood waves from debris torrents can exceed 5 meters deep in small streams normally 3 m wide and 15 cm deep. These torrents move boulders in excess of 1 m diameter and logs >1 m diameter and >10 m long. In temperate regions, debris torrents occur primarily in steep drainages and are relatively infrequent, occurring typically less than once in several centuries. They are usually set into motion by the sudden release of large volumes of water upon the breaching of a natural or human-constructed impoundment, a process often initiated by mass hillslope failures (landslides) during high intensity rainfall or snowmelt. Debris torrents course downstream until the slope of the stream channel can no longer keep their viscous sediment suspension in motion (typically <3% for small streams); at this point, they "set up", depositing large amounts of sediment, boulders, logs, and whatever else they were transporting. Upstream, the *torrent track* is severely scoured, often reduced in channel complexity and devoid of near-bank riparian vegetation. As with floods, the massive disruption of the stream channel and its biota are transient, and these intense, infrequent events will often lead to a high-quality complex habitat within years or decades, as long as natural delivery of large wood and sediment from riparian and upland areas remains intact.

In arid areas with high runoff potential, debris torrents can occur in conjunction with flash flooding from extremely high-intensity rainfall. They may be nearly annual events in

some steep ephemeral channels where drainage area is sufficient to guarantee isolated thunderstorms somewhere within their boundaries, but small enough that the effect of such storms is not dampened out by the portion of the watershed not receiving rainfall during a given storm.

Because they may alter habitat and biota substantially, infrequent major floods and torrents can confuse the interpretation of measurements of stream biota and habitat in regional surveys and monitoring programs. Therefore, it is important to determine if a debris torrent or major flood has occurred within the recent past. After completing the thalweg profile and channel/riparian measurements and observations, examine the stream channel along the entire sample reach, including its substrate, banks, and riparian corridor, checking the presence of features described on the Torrent Evidence Assessment Form (Figure 7-18). It may be advantageous to look at the channel upstream and downstream of the actual sample reach to look for areas of torrent scour and massive deposition to answer some of the questions on the field form. For example, you may more clearly recognize the sample reach as a torrent deposition area if you find extensive channel scouring upstream. Conversely, you may more clearly recognize the sample reach as a torrent scour reach if you see massive deposits of sediment, logs, and other debris downstream.

7.7 EQUIPMENT AND SUPPLIES

Figure 7-19 lists the equipment and supplies required to conduct all the activities described for characterizing physical habitat. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the stream. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

7.8 LITERATURE CITED

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EM

TORRENT EVIDENCE ASSESSMENT FORM - STREAMS

SITE ID: WXXP99-9999 DATE: 07/02/2004

TORRENT EVIDENCE	
Please X any of the following that are evident.	
EVIDENCE OF TORRENT SCOURING:	
☐	01 - Stream channel has a recently devegetated corridor two or more times the width of the low flow channel. This corridor lacks riparian vegetation with possible exception of fireweed, even-aged alder or cottonwood seedlings, grasses, or other herbaceous plants.
☐	02 - Stream substrate cobbles or large gravel particles are NOT IMBRICATED. (Imbricated means that they lie with flat sides horizontal and that they are stacked like roof shingles – imagine the upstream direction as the top of the "roof.") In a torrent scour or deposition channel, the stones are laying in unorganized patterns, lying "every which way." In addition many of the substrate particles are angular (not "water-worn.")
☐	03 - Channel has little evidence of pool-riffle structure. (For example, could you ride a mountain bike down the channel?)
☐	04 - The stream channel is scoured down to bedrock for substantial portion of reach.
☐	05 - There are gravel or cobble berms (little levees) above bankfull level.
☐	06 - Downstream of the scoured reach (possibly several miles), there are massive deposits of sediment, logs, and other debris.
☐	07 - Riparian trees have fresh bark scars at many points along the stream at seemingly unbelievable heights above the channel bed.
☐	08 - Riparian trees have fallen into the channel as a result of scouring near their roots.
EVIDENCE OF TORRENT DEPOSITS:	
☐	09 - There are massive deposits of sediment, logs, and other debris in the reach. They may contain wood and boulders that, in your judgement, could not have been moved by the stream at even extreme flood stage.
☐	10 - If the stream has begun to erode newly laid deposits, it is evident that these deposits are "MATRIX SUPPORTED." This means that the large particles, like boulders and cobbles, are often not touching each other, but have silt, sand, and other fine particles between them (their weight is supported by these fine particles -- in contrast to a normal stream deposit, where fines, if present, normally "fill-in" the interstices between coarser particles.)
NO EVIDENCE:	
☒	11 - No evidence of torrent scouring or torrent deposits.

COMMENTS

03/26/2001 2001 Torrent Evidence

Figure 7-18. Torrent Evidence Assessment Form.

EQUIPMENT AND SUPPLIES FOR PHYSICAL HABITAT

QTY.	Item	
1	Surveyor's telescoping leveling rod (round profile, metric scale, 7.5 m extended)	
1	50-m fiberglass measuring tape & reel	
1	Hip chain (metric) for measuring reach lengths (Optional)	
1	Clinometer (or Abney level) with percent and degree scales.	
1	Lightweight telescoping camera tripod (necessary only if slope measurements are being determined by one person)	
2	½-inch diameter PVC pipe, 2-3 m long: Two of these, each marked at the same height (for use in slope determinations involving two persons)	
1	Meter stick. Alternatively, a short (1-2 m) rod or pole (e.g., a ski pole) with cm markings for thalweg measurements, or the PVC pipe described for slope determinations can be marked in cm and used.	
1 roll ea.	Colored surveyor's plastic flagging (2 colors)	
1	Convex spherical canopy densiometer (Lemmon Model A), modified with taped "V"	
1	Bearing compass (Backpacking type)	
1 or 2	Fisherman's vest with lots of pockets and snap fittings. Used at least by person conducting the in-channel measurements to hold the various measurement equipment (densiometer, clinometer, compass, etc.). Useful for both team members involved with physical habitat characterization.	
2 pair	Chest waders with felt-soled boots for safety and speed (unless nuisance organisms like whirling disease or New Zealand mud snails are present) if waders are the neoprene "stocking" type. Hip waders can be used in shallower streams.	
	Covered clipboards (lightweight, with strap or lanyard to hang around your neck)	
	Soft (#2) lead pencils (mechanical are acceptable)	
11 plus extras	Channel/Riparian Cross-section & Thalweg Profile and Woody Forms	
1 plus extras	Slope and Bearing Form; Riparian "Legacy" Tree and Invasive Alien Plant Form; Channel Constraint Assessment Form; Torrent Evidence Form.	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for physical habitat characterization	

Figure 7-19. Checklist of equipment and supplies for physical habitat.

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NOTES

NOTES

SECTION 8
INVASIVE RIPARIAN PLANTS

Paul L. Ringold[1], Teresa Magee[2], and Philip R. Kaufmann[1]

A trend of increasing concern along streams in many parts of the Western U.S. is the invasion of alien (nonnative) tree, shrub, and grass species. The plant assemblages that result from such invasions can substantially alter stream ecosystems and/or negatively influence the economic value of the land they occupy (Pimentel et al. 2000, 2005). Riparian areas are particularly susceptible to invasions because of both natural and human-induced disturbance regimes, and the connectivity of the stream network (Nilsson and Svedmark 2002, Brown and Peet 2003). Determining the extent and severity of invasions provides some perspective on how different *least disturbed* (best of what is left) and *minimally disturbed* (unaltered) aquatic systems are in the western U.S.

The response design at the reach scale for invasive riparian plants is identical to that described in Section 7 for riparian vegetation structure. Riparian plots established in conjunction with the eleven cross-section transects along the support reach are examined for the presence of selected species of invasive plants. Twelve species were selected for inclusion in the invasive plant protocol as *target species*. Selection of these plants was based on ease of field identification by non-botanists, degree of economic or ecological impact at a regional (rather than local) scale, preference for riparian habitats, lack of toxicity, and ecological variety within the list of target species. Different combinations of the twelve species were developed for each individual State. Because each of the target species are aggressive invaders (they can spread extensively along a stream reach in a short period of time), presence anywhere in the support reach is the principal variable for this indicator (Stoddard et al. 2005).

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

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

8.1 DETERMINING THE PRESENCE OF INVASIVE RIPARIAN PLANTS

This procedure was previously included as part of the overall physical habitat characterization (Section 7), and uses the same field data form as the "legacy" tree component of the physical habitat characterization (described in Section 7.5.9). The procedure for determining the presence of invasive plant taxa at a site is presented in Table 8-1. At each transect, examine the area within the 10 m x 10 m riparian plots on each bank (Figure 8-1). Do not spend more than about 5 minutes in each plot looking for invasive riparian plants. Record the presence of any listed plant taxon on the Riparian "Legacy" Trees and Invasive Alien Plants field form (Figure 8-2). Appendix D (on the CD-ROM) includes color photographs and text descriptions of all target taxa to help determine if any of them are present within the plot. Note that the list of target plant taxa varies from state to state. Record only the presence of plants that are target taxa for your state, even though you may observe other invasive riparian taxa. If you notice the presence of other invasive species not on the list, you can note their presence in the comments section of the form. If no listed taxa for your state are present within the plot, mark the *NONE* box on the form.

8.2 EQUIPMENT AND SUPPLIES

Figure 8-3 lists the equipment and supplies required to conduct all the activities described for determining the presence of invasive riparian plants. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the stream. Use this checklist to ensure that equipment and supplies are organized and available at the stream site to conduct the activities efficiently.

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TABLE 8-1. PROCEDURE FOR IDENTIFYING INVASIVE RIPARIAN PLANT SPECIES

1. Examine the 10m x 10m riparian plots on both banks for the presence of invasive riparian plant species. Do not spend more than about 5 min in each plot looking for invasive plants. Look for those species from the following table listed as *target species* for your State.

Name to Check on Form	Common Name	Binomial: Genus species	CA	OR	WA	ID	ND	SD	WY	CO	AZ	UT	MT	NV
Can This	Canada Thistle	*Cirsium arvense*	X	X	X	X	X	X	X	X			X	
G Reed	Giant Reed	*Arundo donax*	X								X	X		X
Hblack	Himalayan Blackberry	*Rubus discolor*	X	X	X	X								
Spurge	Leafy Spurge	*Euphorbia esula*					X	X	X	X			X	
M This	Musk Thistle	*Carduus nutans*	X	X	X	X	X	X	X	X			X	
Englvy	English Ivy	*Hedera helix*	X	X	X	X	X	X	X	X	X	X	X	X
RCGrass	Reed Canarygrass	*Phalaris arundinacea*	X	X	X	X								
Rus Ol	Russian-olive	*Elaeagnus angustifolia*	X				X	X	X	X	X	X	X	X
SaltCed	Salt Cedar	*Tamarix spp.*	X				X	X	X	X	X	X	X	X
ChGrass	Cheatgrass	*Bromus tectorum*	X	X	X	X	X	X	X	X	X	X	X	X
Teasel	Teasel	*Dipsacus fullonum*	X	X	X					X			X	
C Burd	Common Burdock	*Arctium minus*	X	X	X	X	X	X	X	X	X	X	X	X

X On the list for this state
■ Not on the list for this state

2. Record the presence of any species listed for your State within the plot on either the left or right bank by marking the appropriate box(es) on the right hand column of the Riparian "Legacy" Trees and Invasive Alien Plants field form. Use the identification materials provided in Appendix D. If none of the species listed for your state is present in either of the plots at a given transect check the box labeled *NONE* for this transect.

3. Repeat Steps 1 through 5 for each remaining transect (*B* through *K*). At transect *K*, look upstream a distance of 4 channel widths).

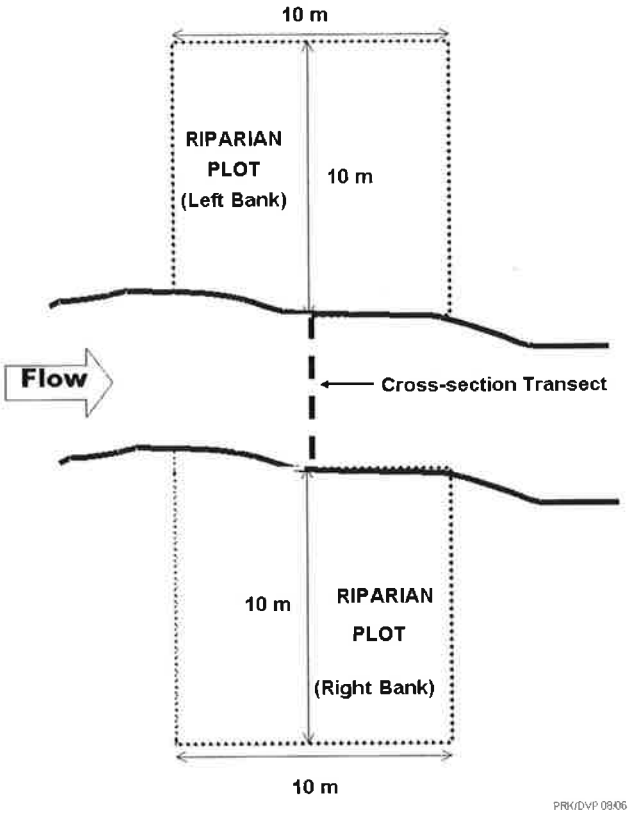

Figure 8-1. Boundaries for visual estimation of invasive riparian plants.

Pimentel, D., R. Zuniga, and D. Morrison. 2005. Update on the environmental and economic costs associated with alien-invasive species in the United states. *Ecological Economics* 52:273-288.

Stoddard, J.L., D.V. Peck, A.R. Olsen, D.P. Larsen, J. Van Sickle, C.P. Hawkins, R.M. Hughes, T.R. Whittier, G. Lomnicky, A.T. Herlihy, P.R. Kaufmann, S.A. Peterson, P.L. Ringold, S.G. Paulsen, and R. Blair. 2005. *Environmental Monitoring and Assessment Program: western streams and rivers statistical summary.* EPA 620/R-05/006. U.S. Environmental Protection Agency, Washington, D.C.

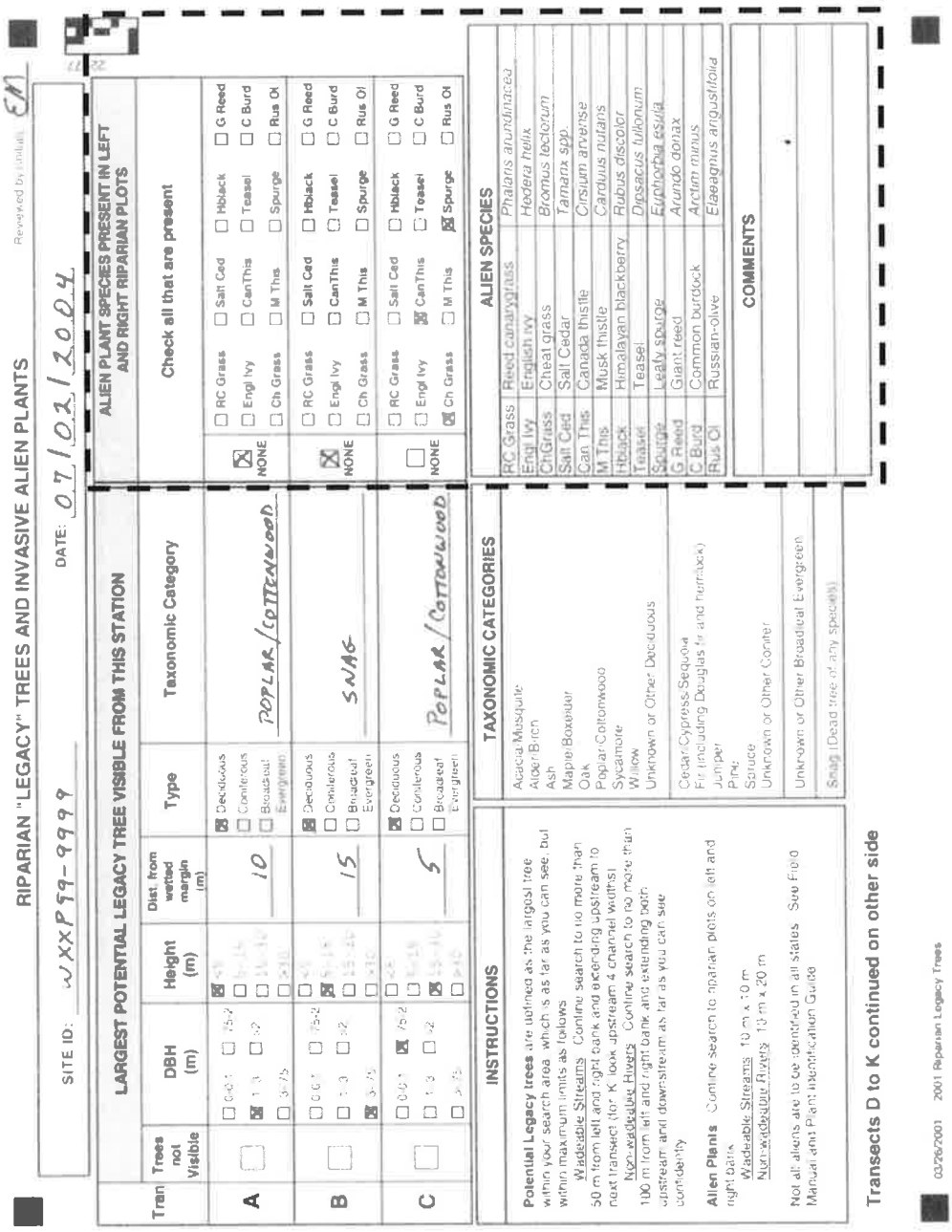

Figure 8-2. Riparian "Legacy" Tree and Invasive Alien Plant Form (Page 1), showing data for invasive riparian plants.

EQUIPMENT AND SUPPLIES FOR INVASIVE RIPARIAN PLANT DETERMINATION

QTY.	Item	
1	Covered clipboards (lightweight, with strap or lanyard to hang around neck)	
several	Soft (#2) lead pencils (mechanical are acceptable)	
1 plus extras	Riparian "Legacy" Tree and Invasive Alien Plant Form	
1 copy	Field operations and methods manual	
1 set	Laminated sheet of procedure table and quick reference guide (including color pictures) for target species of invasive plants (available as Appendix D from CD-ROM)	

Figure 8-3. Checklist of equipment and supplies for determining the presence of invasive riparian plants.

NOTES

SECTION 9
PERIPHYTON

Brian H. Hill[1] and David V. Peck[2]

Periphyton are algae, fungi, bacteria, protozoa, and associated organic matter associated with channel substrates. Periphyton are useful indicators of environmental condition because they respond rapidly and are sensitive to many anthropogenic disturbances, including habitat destruction, contamination by nutrients, metals, herbicides, hydrocarbons, and acidification (e.g., Bahls 1993, Pan et al. 1999, Hill et al. 2000, Griffith et al. 2002).

Collection procedures for periphyton are similar to the *multihabitat* procedure of the Rapid Bioassessment Protocol (RBP; Barbour et al. 1999). A representative sample of the periphyton assemblage cannot be collected from a single point. Therefore, the response design is based on the collection of multiple samples from throughout the support reach. A fixed number (and thus area) of collection points is allocated systematically throughout the support reach ensure a distribution of samples between faster water and slower water habitats, eliminate individual sampler bias, and provide a comparable and consistent sample from every site. To save time, periphyton sampling points are associated with benthic macroinvertebrate sampling points. All samples collected at a site are combined into a single composite sample to characterize the sampling point (Barbour et al. 1999) and to reduce the cost and effort in processing and analysis. The number of individual samples is expected to provide a composite sample having a sufficient number of individuals (attached algae and/or diatoms) to characterize the taxonomic composition and relative abundance of the assemblage.

Changes to the periphyton sampling procedures from the previous published EMAP-SW field operations manual (Hill 1998), and modifications made during EMAP-W

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1 U.S. EPA, National Health and Ecological Effects Research Laboratory, Mid-Continent Ecology Division, 6201 Congdon Blvd, Duluth, MN 55804.

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are summarized in Appendix B. The *biomorphs* (refer to Figure 2-1) collect periphyton samples at the same time as benthic macroinvertebrate samples (Section 10). At the completion of the day's sampling activities, but before leaving the stream, three types of laboratory samples are prepared from each composite index sample.

An optional procedure to collect an additional *targeted habitat* sample from a single habitat type is described in Section 9.3. This procedure is modified from the RBP procedure (Stevenson and Bahls 1999) and Hawkins et al. (2001). The targeted habitat sample allowed for a comparison of assessments of the periphyton assemblage based on the standard EMAP-W sampling procedure and those based on a targeted habitat procedure used in a separate study focusing on reference sites in the western U.S.

9.1 SAMPLE COLLECTION

The general scheme for collecting and processing periphyton samples from the support reach is illustrated in Figure 9-1. The procedure for collecting periphyton samples is presented in Table 9-1. At each transect, collect a sample from an assigned sampling point (left, center, or right as you face downstream). Locate the sampling point *1 m downstream of the transect* to avoid disturbing substrates that are enumerated and classified as part of the physical habitat characterization (Section 7). Sampling points at each transect may have been assigned when the support reach was laid out (Figure 9-1; refer also to Section 4; Table 4-3). If not, assign the sampling point at transect *A* at random using a digital watch or other suitable means. Once the first sampling point is determined, collect either an *erosional* or *depositional* sample, depending on whether the sampling point is in a flowing water (e.g., a riffle or run) *erosional habitat* with coarser substrate, or in a slow water (e.g., a pool) *depositional habitat* with finer substrate. Combine the individual transect samples into a single plastic bottle to create a composite sample for the reach. Record the total volume of the composite sample on the Sample Collection Form as shown in Figure 9-2. Note that optional targeted habitat periphyton samples (Section 9.4) are collected in conjunction with benthic macroinvertebrate samples from a targeted habitat (riffles), rather than at each transect.

9.2 PREPARATION OF LABORATORY SAMPLES

Three laboratory samples are prepared from the composite sample: an ID/enumeration sample (to determine taxonomic composition and relative abundances), a

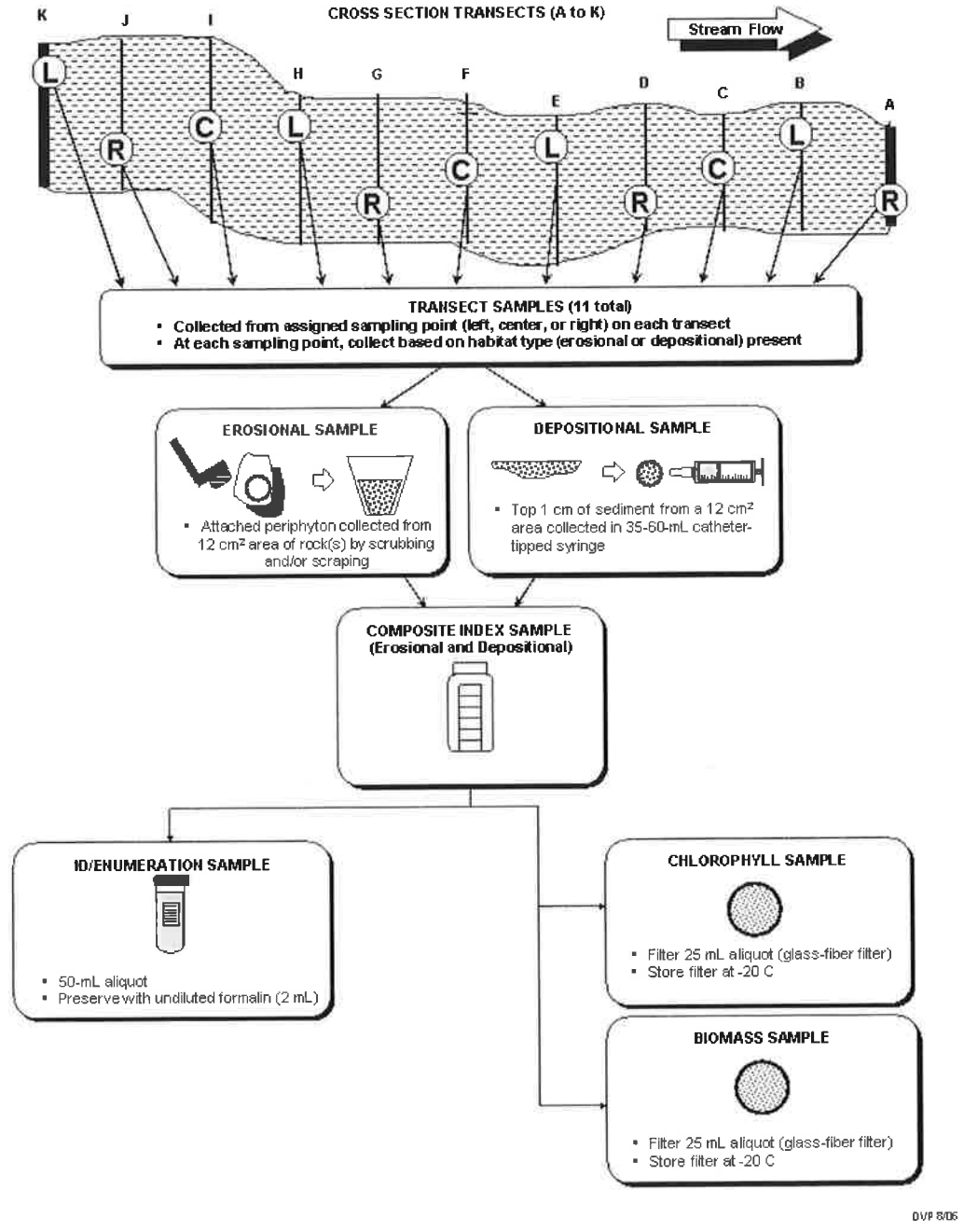


Figure 9-1. Response design for collecting periphyton samples.

TABLE 9-1. PROCEDURE FOR COLLECTING COMPOSITE SAMPLES OF PERIPHYTON

1. Starting at Transect *A*, determine if the assigned sampling point (*Left*, *Center*, or *Right*) is located in an erosional (riffle) habitat or a depositional (pool) habitat. Collect a single sample at the point using the appropriate procedure in Step 2 below. NOTE: to avoid disturbing the substrate on a transect, locate each sampling point **1 m downstream** of the actual transect.

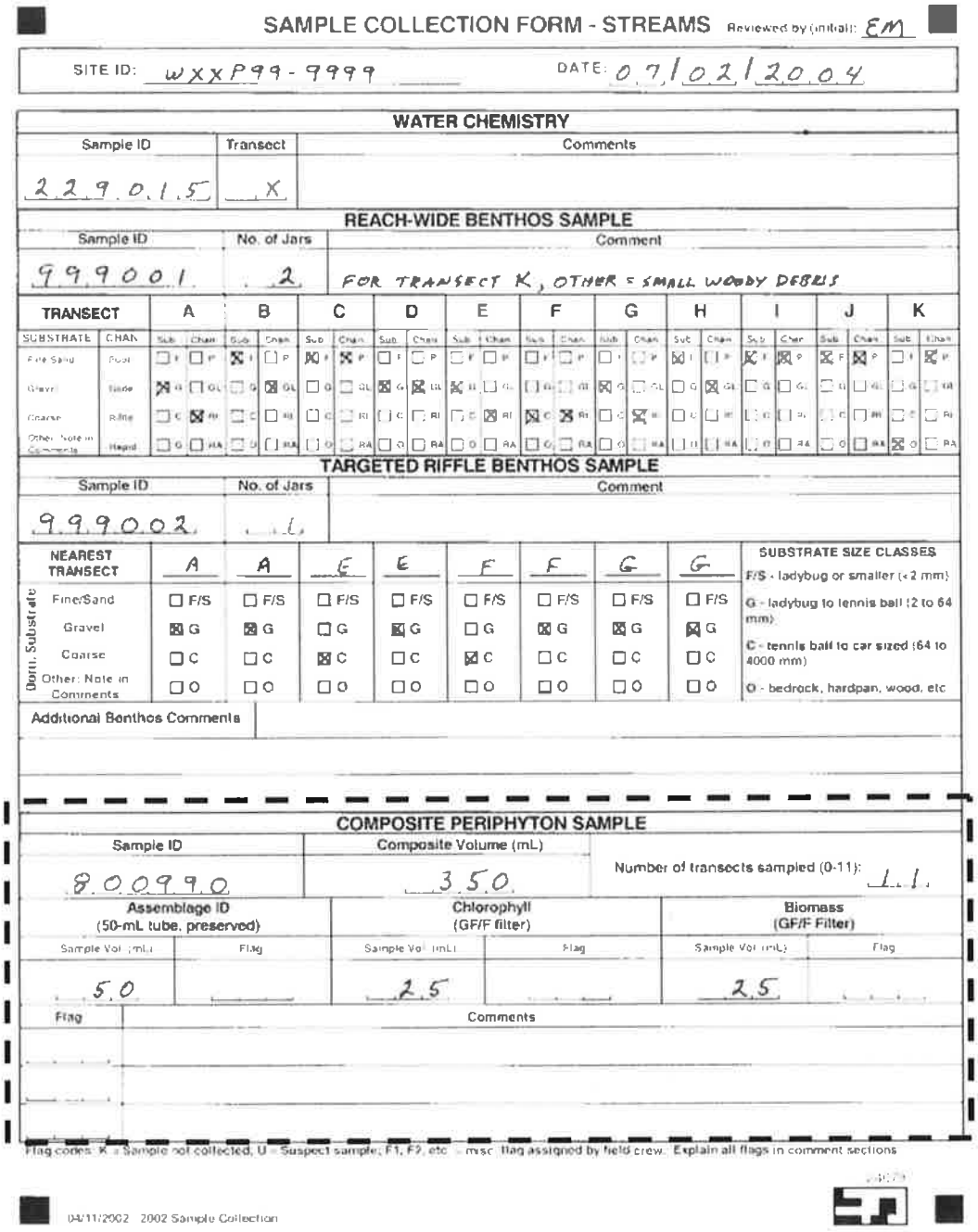
   If the sampling points were not assigned previously when laying out the sampling reach, proceed to Transect *A*. Randomly determine if it is a left (*L*), center (*C*), or right (*R*) sampling point for collecting periphyton and benthic macroinvertebrate samples. For example, use a digital wristwatch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

2A. *Erosional habitats* (coarser substrate present):
   (1) Collect a sample of substrate (rock or wood) that is small enough (< 15 cm diameter) and can be easily removed from the stream. Place the substrate in a plastic funnel which drains into a 500-mL plastic bottle with volume graduations marked on it and labeled *PERIPHYTON*.
   (2) Use the area delimiter to define a 12-cm^2 area on the **upper** surface of the substrate. Dislodge attached periphyton from the substrate within the delimiter into the funnel by brushing with a stiff-bristled toothbrush for 30 seconds. *Take care to ensure that the **upper surface** of the substrate is the surface that is being scrubbed, and that the **entire** surface within the delimiter is scrubbed.*
   (3) Fill a wash bottle with stream water. Using a **minimal** volume of water from this bottle, wash the dislodged periphyton from the rock, delimiter, and funnel into the 500-mL bottle.

2B. *Depositional habitats* (finer substrate present):
   (1) Use the area delimiter to confine a 12-cm^2 area of soft sediments.
   (2) Vacuum the top 1 cm of sediments from within the delimited area into a catheter-tipped syringe (35-60-mL size).
   (3) Empty the syringe into the 500-mL *PERIPHYTON* bottle (combining it with samples collected from erosional habitats).

3. Repeat Steps 1 and 2 for transects *B* through *K* to produce the composite sample for the support reach. Keep the collection bottle out of direct sunlight as much as possible to minimize degradation of chlorophyll.

4. After samples have been collected from all 11 transects, add sufficient stream water from the wash bottle to bring the total volume to the nearest 50-mL mark (final volume can be less than 500 mL). Mix the 500-mL bottle by gently swirling the sample to help separate periphyton from heavier sediments. Record the total volume of the composite sample in the periphyton section of the Sample Collection Form. Also record the number of transects where you obtained a periphyton sample.



SAMPLE COLLECTION FORM - STREAMS Reviewed by (initial): EM

SITE ID: WXXP99-9999 DATE: 07/02/2004

WATER CHEMISTRY

Sample ID	Transect	Comments
229015	X	

REACH-WIDE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999001	2	FOR TRANSECT K, OTHER = SMALL WOODY DEBRIS

TRANSECT	A	B	C	D	E	F	G	H	I	J	K
SUBSTRATE / CHAN	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan
Fine Sand / Pool	☐ F / ☐ P	☒ F / ☐ P	☒ F / ☒ P	☐ F / ☐ P	☐ F / ☐ P	☐ F / ☐ P	☐ F / ☐ P	☒ F / ☐ P	☒ F / ☒ P	☒ F / ☒ P	☐ F / ☒ P
Gravel / Glide	☒ G / ☐ GL	☐ G / ☒ GL	☐ G / ☐ GL	☒ G / ☒ GL	☒ G / ☐ GL	☐ G / ☐ GL	☒ G / ☐ GL	☐ G / ☒ GL	☐ G / ☐ GL	☐ G / ☐ GL	☐ G / ☐ GL
Coarse / Riffle	☐ C / ☒ RI	☐ C / ☐ RI	☐ C / ☐ RI	☐ C / ☒ RI	☐ C / ☒ RI	☒ C / ☒ RI	☐ C / ☒ RI	☐ C / ☐ RI	☐ C / ☐ RI	☐ C / ☐ RI	☐ C / ☐ RI
Other: Note in Comments / Rapid	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☒ O / ☐ RA

TARGETED RIFFLE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999002	11	

NEAREST TRANSECT	A	A	E	E	F	F	G	G
Dom. Substrate: Fine/Sand	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S
Gravel	☒ G	☒ G	☐ G	☒ G	☐ G	☒ G	☒ G	☒ G
Coarse	☐ C	☐ C	☒ C	☐ C	☒ C	☐ C	☐ C	☐ C
Other: Note in Comments	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O

SUBSTRATE SIZE CLASSES
F/S - ladybug or smaller (< 2 mm)
G - ladybug to tennis ball (2 to 64 mm)
C - tennis ball to car sized (64 to 4000 mm)
O - bedrock, hardpan, wood, etc

Additional Benthos Comments

COMPOSITE PERIPHYTON SAMPLE

Sample ID	Composite Volume (mL)	Number of transects sampled (0-11):
800990	350	11

Assemblage ID (50-mL tube, preserved)		Chlorophyll (GF/F filter)		Biomass (GF/F Filter)	
Sample Vol. (mL)	Flag	Sample Vol. (mL)	Flag	Sample Vol. (mL)	Flag
50		25		25	

Flag	Comments

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

04/11/2002 2002 Sample Collection

Figure 9-2. Sample Collection Form, showing data recorded for periphyton samples.

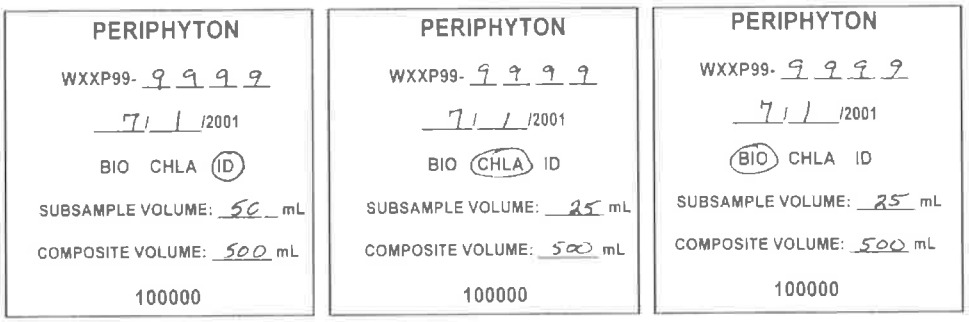


Figure 9-3. Set of completed periphyton sample labels.

chlorophyll sample, a biomass sample (for ash-free dry mass). Keep all the sample containers required for an individual stream in sealed in plastic bags until use (see Section 3) to avoid external sources of contamination (e.g., dust, dirt, or mud) that are present at streamside.

A set of completed periphyton sample labels is shown in Figure 9-3. All labels in a set have the same sample ID number. Circle the appropriate type of sample (chlorophyll, biomass, etc.) on each label. Attach the completed labels to their appropriate containers and cover with clear tape. When attaching the completed labels, do not cover any volume graduations and markings on the container.

9.2.1 ID/Enumeration Sample

Prepare the ID/Enumeration sample as a 50-mL aliquot from the composite sample, following the procedure presented in Table 9-2. Preserve each sample with 2 mL of undiluted formalin (37% formaldehyde), observing all safety precautions associated with handling formalin solution. *If you are using **diluted** formalin (e.g., 10-12%) as a preservative, adjust the subsample volume to **40 mL** and add **10 mL** of preservative.* Record the ID number (barcode) from the sample container label and the total volume of the sample (50 mL) in the appropriate fields on the Sample Collection Form as shown in Figure 9-2. Explain any deviations from the target volume in the comments field of the collection form. Store the preserved samples upright in a container containing absorbent material, according to the guidelines provided for handling formalin-preserved samples. Ship the ID/Enumeration sample together with the other periphyton samples.

TABLE 9-2. PREPARATION OF ID/ENUMERATION SAMPLE FOR PERIPHYTON

1. Thoroughly mix the bottle containing the composite periphyton sample.

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

3. Record the sample ID number of the label and the total volume of the composite index sample on the Sample Collection Form. Explain any deviations from the target volume in the comments section of the form.

4. Pour 50 mL of the composite sample into the labeled 50-mL centrifuge tube.

5. Wear gloves and safety glasses. Use a syringe or bulb pipette to add 2 mL of undiluted formalin solution (37% formaldehyde) to the sample. Cap the tube tightly and seal with plastic electrical tape. Shake gently to distribute the preservative.

   NOTE: If using previously diluted formalin (e.g., 10-12%) as a preservative, *adjust the subsample volume to **40 mL** and add **10 mL** of preservative solution.* Note the use of diluted preservative in the comments.

6. Record the volume of the sample (typically 50 mL; exclude the volume of preservative added to the ID sample) on the Sample Collection Form. Double check that the volume recorded on the collection form matches the total volume recorded on the corresponding sample label, especially if you are using diluted formalin as a preservative.

9.2.2 Chlorophyll Sample

Prepare a chlorophyll sample by filtering a 25-mL aliquot of the composite index sample through a glass fiber filter (Whatman GF/F or equivalent) as described in Table 9-3. Chlorophyll can degrade rapidly when exposed to bright light, so filter the samples in subdued light (or shade) as quickly as possible after collection to minimize degradation. The filtration apparatus is illustrated in Figure 9-4. Rinse the filtration chamber with de-ionized water each day at the base site and seal it in a plastic bag until use at the stream (see Section 3). Keep the glass fiber filters in a dispenser inside a sealed plastic bag until use.

It is important to measure the volume of the sample being filtered accurately (±1 mL) with a graduated cylinder. During filtration, do not exceed 7 pounds per square inch (psi) to avoid rupturing cells. If the vacuum pressure exceeds 7 psi, prepare a new sample. If the filter clogs completely before the volume of sample in the chamber has been filtered, discard the sample and filter, and prepare a new sample using a smaller volume of sample.

After filtering the sample, fold the filter paper in half and place it in a 50-mL centrifuge tube. Complete a sample label (Figure 9-3) and check it to ensure that all written information is complete and legible. Affix the label to the centrifuge tube and cover it completely with a strip of clear tape. Record the sample ID number printed on the label on the Sample Collection Form (Figure 9-2). Make sure the volume recorded on the sample label matches the corresponding volume recorded on the Sample Collection Form. Record a flag and provide comments on the Sample Collection Form if there are any problems in collecting the sample or if conditions occur that may affect sample integrity. Wrap the centrifuge tube in aluminum foil and store it in a resealable plastic bag in darkness. Store the sample frozen until shipment to the laboratory (Section 3).

9.2.3 Biomass Sample

Prepare the biomass sample from a 25-mL aliquot of the composite sample. Prepare the sample according to the procedure presented in Table 9-3. As with the chlorophyll sample, measure the volume to be filtered accurately (±1 mL). Rinse the filter chamber components (Figure 9-4) and the graduated cylinder thoroughly between the chlorophyll and biomass samples with deionized water.

TABLE 9-3. PROCEDURE FOR PREPARING CHLOROPHYLL AND BIOMASS SAMPLES FOR PERIPHYTON

1. Mix the composite sample thoroughly.

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

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

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

   - NOTE: For composite samples containing fine sediment, allow grit to settle before pouring the sample into the graduated cylinder.

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

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

6. Remove both plugs from the filtration chamber and pour out the filtered water in the chamber. Remove the filter funnel from the filter holder. Remove the filter from the holder with clean forceps. Avoid touching the colored portion of the filter. Fold the filter in half, with the colored side folded in on itself. Place the folded filter paper into a 50-mL centrifuge tube.

7. Complete a periphyton sample label for chlorophyll, including the volume filtered, and attach it to the centrifuge tube. Cover the label completely with a strip of clear tape.

8. Record the sample ID number (barcode) of the label and the total volume of the composite index sample on the Sample Collection Form. Record the volume filtered in the *CHLOROPHYLL* field on the Sample Collection Form. Double check that the volume recorded on the collection form matches the total volume recorded on the sample label.

9. Wrap the tube completely with aluminum foil to protect it from light. Place the centrifuge tube into a resealable plastic bag. Place the plastic bag into a portable freezer, a cooler containing dry ice, or between two sealed plastic bags of ice in a cooler.

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

11. Repeat Steps 1 through 11 to prepare the biomass sample, completing a periphyton sample label for biomass and recording sample information in the *BIOMASS* section of the Sample Collection Form.

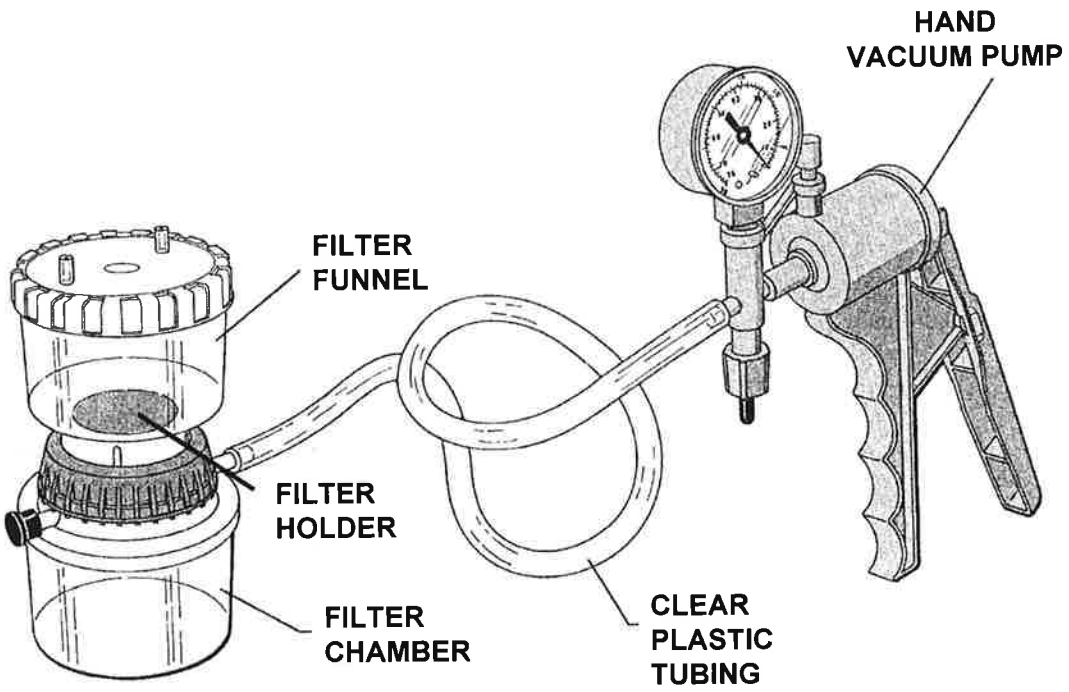


Figure 9-4. Filtration apparatus for preparing chlorophyll and biomass subsamples for periphyton. Modified from Chaloud et al. (1989).

After filtering the sample, complete a biomass sample label as shown in Figure 9-3. Check the sample label to ensure that all written information is complete and legible. Affix the label to the 50-mL centrifuge tube and cover it completely with clear tape. Record the sample ID number printed on the label and the volume filtered on the Sample Collection Form as shown in Figure 9-2. Make sure the information recorded on the sample label matches the corresponding information recorded on the Sample Collection Form. Record a flag and provide comments on the Sample Collection Form if there are any problems in collecting the sample or if conditions occur that may affect sample integrity. Wrap each tube in aluminum foil, place them in a resealable plastic bag. and store them frozen until shipment to the laboratory (Section 3).

9.3 COLLECTING PERIPHYTON FROM A TARGETED HABITAT

This is an optional procedure that involves the collection of a separate composite periphyton sample and the preparation of an additional ID/enumeration subsample. Collect a series of individual samples from **one** type of targeted habitat as described in Table 9-5. The preferred targeted habitat is riffle, with periphyton scraped from individual rocks collected at the same locations as the *targeted riffle* benthos sample (Section 10). If riffle habitat is not available within the stream reach, scrape periphyton from individual pieces of wood collected from throughout the stream reach. If a support reach lacks both riffle and wood, collect an additional *Sand/silt* sample at each transect where you collect a depositional periphyton sample. Combine all individual samples from the targeted habitat into a single 1-L container marked in 250-mL increments, and add stream water to bring the total volume of the composite sample to the nearest 250-mL mark. Prepare an ID/enumeration subsample having a total volume of 40 mL and preserve with formalin (2 mL of undiluted formalin, or 10 mL of dilute formalin solution). Complete a sample label as shown in Figure 9-5, and record the sample information on the separate field data form for the targeted habitat periphyton sample (Figure 9-6).

After becoming familiar with the sampling reach, determine the channel classification and dominant habitat type and record these on the field data form. Figure 9-7 provides guidance for determining the channel classification and dominant habitat type. This information allows data from EMAP-W sites to be combined with data from other sites where the EMAP physical habitat procedures were not used.

9.4 EQUIPMENT AND SUPPLIES

Figure 9-8 is a checklist of equipment and supplies required to conduct periphyton sample collection and processing activities. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure that all of the required equipment is brought to the stream. Use this checklist to ensure that equipment and supplies are organized and available at the stream site in order to conduct the activities efficiently.

TABLE 9-4. COLLECTING A TARGETED HABITAT PERIPHYTON SAMPLE

NOTE: This is an optional procedure.

1. Collect **one** composite periphyton sample at each site (listed in order of preference):
   A. If you collect an EMAP targeted riffle benthic sample, then collect a *ROCK* periphyton sample (Step 2).
   B. If you do not collect an EMAP targeted riffle sample, but there is sufficient submerged wood in the stream to obtain 16 individual samples, then collect a *SNAG* periphyton sample (Step 3).
   C. If you do not collect an EMAP targeted riffle sample, and there is not enough wood pieces available to collect a *SNAG* sample, then collect a *SAND/SILT* periphyton sample (Step 4).

2. *ROCK sample*:
   A. At each *riffle unit* where you collect an EMAP targeted riffle kicknet sample (Section 10), select **TWO** pieces of substrate for each kicknet sample (i.e., if you collect two kicknet samples from a single riffle unit, then you would collect 4 pieces of substrate from that riffle unit.).
      Collect the pieces from throughout the riffle unit (i.e., they do not have to be at or near the kicknet sample locations). Each piece must be large enough to use with the periphyton area delimiter.
      Collect the substrate pieces from a depth of 15-20 cm (or deepest area if stream is < 15 cm deep).
   B. Collect a sample from each piece of substrate using the EMAP procedure for erosional samples. Rinse the sample into a 1-L jar, marked at 250, 500, 750, and 1000 mL, labeled *TARGETED HABITAT PERIPHYTON SAMPLE*.
   C. Repeat Steps 2A and 2B for each piece of substrate and each targeted riffle kicknet sample. You will end up preparing 16 pieces of substrate total.
   D. Go to Step 5.

3. *SNAG sample*:
   A. As you proceed up the support reach, look for submerged pieces of wood. Each piece must be large enough for the periphyton area delimiter to completely fit on.
      Collect the wood pieces from a depth of 15-20 cm (or the deepest area if the stream is < 15 cm deep).
   B. Collect a sample from each piece of wood using the EMAP procedure for erosional samples. Rinse the sample into a 1-L jar, marked at 250, 500, 750, and 1000 mL, labeled *TARGETED HABITAT PERIPHYTON SAMPLE*.
   C. Repeat Steps 3A and 3B for each piece of wood until you have collected 16 individual samples.
   D. Go to Step 5.

(Continued)

TABLE 9-4 (Continued)

4. *SAND/SILT sample*:
   A. At each transect where you collected a standard EMAP depositional periphyton sample (i.e., using a syringe), collect **two** additional depositional samples from a depth of 15-20 cm (or the deepest part if the stream is < 15 cm deep).
      NOTE: Skip over transects where you collected standard EMAP erosional periphyton samples.
      Collect the samples from anywhere within the depositional habitat unit that the transect is passing through (i.e., they do not have to be the transect sample locations or on the transect itself).
   B. Use the EMAP procedure for the additional depositional samples (area delimiter and catheter-tipped syringe). Place the contents of each syringe sample into a 1-L jar, marked at 250, 500, 750, and 1000 mL, labeled *TARGETED HABITAT PERIPHYTON SAMPLE*.
   C. Repeat Steps 4A and 4B for each depositional transect, until you have collected a total of 16 additional depositional samples.
   D. Go to Step 5.

5. Prepare a single ID/Enumeration subsample from the targeted habitat composite sample (either *ROCK*, *SNAG* or *SAND/SILT*) as follows:

   A. Dilute the composite sample to the nearest 250 mL mark using stream water from a wash bottle.
   B. Cap the composite sample bottle tightly. Shake the bottle vigorously until all material is fully suspended and homogenized (clumps of algae are broken up).
   C. Pour about 10 mL of suspended sample into a 50 mL centrifuge tube.
   D. Repeat Steps 4B and 4C three more times, so the final subsample volume will be 40 mL.

6. Complete a sample label. Circle the sample type (*ROCK/GRAVEL* or *SAND/SILT*), and record the number of substrate pieces used (for rock samples) or the number of syringe samples collected. Circle the composite sample volume, and record the subsample volume.

   NOTE: For *SNAG* samples, write in *SNAG* on the sample label.

7. Add 2 mL of buffered formalin to the subsample. Cap tightly and seal with plastic tape.
   NOTE: *If using dilute (10-12%) formalin, add **10 mL** to the subsample* (or fill it to the 50-mL mark if the volume is greater than 40 mL).

8. Attach the completed label to the centrifuge tube.

9. Record the header information on the field data form. Make sure the site ID and date collected match those on the label.

(Continued)

TABLE 9-4 (Continued)

10. Record the sample collection information from the label onto the form.

    NOTE: If you collect a *SNAG* sample, write in *SNAG* in the SAMPLE TYPE field on the collection form.

11. *CHANNEL CLASSIFICATION*: Complete this part of the field form after you have completed sampling and have a good idea of the channel characteristics within the support reach.
    A. Establish the valley segment characteristics as either *colluvial*, *bedrock*, or *alluvial*.
       Colluvial: Generally, colluvial valleys are those in which colluvial fills (i.e., material from hill slopes) accumulate and are periodically excavated by the stream.
       Bedrock: There is no contiguous alluvial bed. Some alluvial material may be temporarily stored in scour holes, or behind flow obstructions, but in general the bedrock valley channel bed lacks an alluvial cover, and there is little, if any, channel fill.
       Alluvial: Channel is capable of sorting and transporting the load supplied to it from upstream channels, but the transport capacity is not sufficient to scour it to bedrock.
    B. Use the table on the back side of the field data form to classify the channel as either colluvial, bedrock, or one of the following subcategories of alluvial channels: *cascade channel*, *step-pool channel*, *plane-bed channel*, *pool-riffle channel*, *regime channel*, or *braided channel*.
12. *DOMINANT HABITAT TYPES*:
    A. Complete this part of the field form after you have completed sampling and thus have surveyed the entire support reach, select what you feel are the **dominant** erosional and depositional habitat types for the support reach.
    B. Use the descriptions on the back side of the field data form to select the dominant class of **both** erosional and depositional habitats. Circle **one class** in each category.

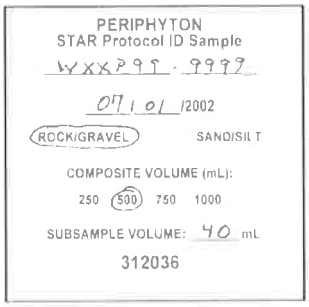

PERIPHYTON
STAR Protocol ID Sample
WXXP99-9999
07/01/2002
(ROCK/GRAVEL) SAND/SILT
COMPOSITE VOLUME (mL):
250 (500) 750 1000
SUBSAMPLE VOLUME: 40 mL
312036

Figure 9-5. Completed label for the targeted habitat periphyton sample.

9.5 LITERATURE CITED

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Griffith, M.B. B.H. Hill, A.T. Herlihy, and P.R. Kaufmann. 2002. Myltivariate analysis of periphyton assemblages in relation to environmental gradients in Colorado Rocky Mountain streams. *Journal of Phycology* 38:83-95.

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Hill, B.H. 1998. Periphyton. Pages 199-132 in J.M. Lazorchak, D.J. Klemm, and D.V. Peck (editors). *Environmental Monitoring and Assessment Program-Surface Waters: field operations and methods for measuring the ecological condition of wadeable streams.* EPA/620/R-94/004F. U.S. Environmental Protection Agency, Washington, D.C.

Hill, B.A., A.T. Herlihy, P.R. Kaufmann, R.J. Stevenson, F.H. McCormick, and C. Burch-Johnson. 2000. Use of periphyton assemblage data as an index of biotic integrity. *Journal of the North American Benthological Society* 19(1):50-67.

Pan, Y., R.J. Stevenson, B.H. Hill, P.R. Kaufmann, and A.T. Herlihy. 1999. Spatial patterns and ecological determinations of benthic algal assemblages in mid-Atlantic Highland streams, USA. *Journal of Phycology* 35:460-468.

MODIFIED STAR PROTOCOL PERIPHYTON DATA

Reviewed by (initial): EM

SITE NAME: PILOT CREEK DATE: 07/02/2004 VISIT: 0 (1) 2 3

SITE ID: WXXP99-9999 TEAM: XX-1

Sample ID	Sample Type (Choose one)	Composite Sample Volume (mL) (Choose one)	Subsample Volume (mL)
3,1,2,0,3,6,	● Rock ○ Snag ○ Sand/Silt	○ 250 ○ 750 ● 500 ○ 1000	4.0

Channel Classification (Choose one)

○ Colluvial ○ Bedrock ○ Cascade ○ Step-Pool ○ Plane-Bed ● Pool-Riffle ○ Regime ○ Braided

Dominant Habitat Type (Choose one from each category)

Erosional (Choose one)				Depositional (Choose one)			
○ Rapid	● Riffle	○ Run	○ Steprun	● Lateral	○ Scour	○ Plunge	○ Dammed

COMMENTS:

04/28/2003 2003 Star Periphyton

Figure 9-6. Field data form for targeted habitat periphyton samples.

Classification of Stream Channels[1]

VALLEY TYPE	Colluvial	Bedrock	Alluvial					
CHARACTERISTIC			Cascade	Step-pool	Plane-bed	Pool-riffle	Regime	Braided
Predominant bed material	Variable	Bedrock	Boulder	Cobble/ boulder	Gravel/ cobble	Gravel	Sand	Variable
Bedform pattern	Variable	Variable	None	Vertical oscillatory	None	Laterally oscillatory	Multilayered	Laterally oscillatory
Dominant roughness elements	Boulders large woody debris	Streambed banks	Boulders banks	Bedforms (steps pools) boulders large woody debris banks	Boulders and cobbles banks	Bedforms (bars pools) boulders and cobbles large woody debris sinuosity banks	Sinuosity bedforms (dunes riffles bars) banks	Bedforms (bars pools)
Dominant sediment sources	Hillslope debris flows	Fluvial hillslope debris flows	Fluvial hillslope debris flows	Fluvial hillslope debris flows	Fluvial bank erosion debris flows	Fluvial bank erosion inactive channels debris flows	Fluvial bank erosion inactive channels	Fluvial bank erosion debris flows
Typical slope (%)	>20	Variable	8 - 30	4 - 8	1 - 4	0.1 - 2	<0.1	< 3
Typical confinement	Strongly confined	Strongly confined	Strongly confined	Moderately confined	Variable	Unconfined	Unconfined	Unconfined
Pool spacing (channel widths)	Variable	Variable	<1	1 - 4	None	5 - 7	5 - 7	Variable

[1]Adapted from Montgomery and Buffington, 1993, *Channel classification, prediction of channel response, and assessment of channel condition*, Report TFW-SH10-93-002, Washington State Timber/Fish/Wildlife.

EROSIONAL HABITAT DESCRIPTIONS		
Name	Code	Description
Rapid	RA	Gradient >4% with swiftly flowing water and considerable surface turbulence. Rapids tend to have larger substrate sizes than riffles.
Riffle	RI	Gradient <4% and shallow with moderate velocity and moderate surface turbulence.
Run	RU	Runs are long, usually straight, low-gradient stream units without flow obstructions. The stream bottom is usually even and the water does not "pool".
Step Run	SR	Step runs are a series of runs that are separated by short (~3m) riffles or flow obstructions.

DEPOSITIONAL HABITAT DESCRIPTIONS		
Name	Code	Description
Lateral	LAT	As streams go around bends they scour laterally and lateral scour pools are created along the banks. The deepest portion is the stream margin.
Scour	SCO	Pool shape is often like a bowl. The deepest part of pool is in the center of the channel.
Plunge	PLU	Plunge pools are formed as water drops over an object. The deepest portion of the pool is directly underneath the falling point.
Dammed	DAM	Water backs up against an obstruction in the stream channel. The deepest portion will be against the object forming the obstruction.

Figure 9-7. Descriptions of channel classes and habitat types associated with targeted habitat periphyton samples. From Hawkins et al. (2001).

EQUIPMENT AND SUPPLIES FOR PERIPHYTON

QTY.	Item	
1	Large funnel (15-20 cm diameter)	
1	12-cm^2 area delimiter (a 3 cm slice of 3.8 cm [1.5 in] inside diameter Schedule 40 PVC pipe); with no modifications for sealing it to a substrate surface)	
1	Stiff-bristle toothbrush with handle bent at 90° angle	
1	1-L wash bottle labeled *STREAM WATER*	
1	1-L wash bottle labeled for and containing deionized water	
1	500-mL plastic bottle (with volume markings every 50-mL), labeled *PERIPHYTON COMPOSITE SAMPLE*	
1	35-60 mL catheter-tipped plastic syringe	
1	Small "fanny pack" or vest with pockets to carry funnel, delimiter, brush, wash bottles, collection bottle, and syringe	
3	50-mL screw-top centrifuge tubes with volume markings	
1 box	Glass-fiber filters for chlorophyll and biomass samples	
1 pair	Forceps for filter handling.	
1	25-mL or 50-mL graduated cylinder, or 50-mL screw-top centrifuge tube with volume markings	
1	Filtration unit, including filter funnel, cap, filter holder, and receiving chamber	
1	Hand-operated vacuum pump and clear plastic vacuum tubing	
1	Aluminum foil for wrapping containers holding chlorophyll and biomass filters	
2	Resealable plastic bags for chlorophyll and biomass samples	
2 mL	Undiluted formalin solution for ID/Enumeration samples (10 mL if using diluted formalin).	
1	Small syringe or bulb pipette for dispensing formalin	
1 pair ea.	Safety glasses and chemical-resistant gloves for handling formalin	
2 sets	Sample labels (3 per set) with the same barcode ID number	
1	Sample Collection Form for stream	
	Soft (#2) lead pencils for recording data on field forms	
	Fine-tipped indelible markers for filling out sample labels	
1 pkg.	Clear tape strips for covering labels	

(Continued)

Figure 9-8. Checklist of equipment and supplies for periphyton.

EQUIPMENT AND SUPPLIES FOR PERIPHYTON (Continued)

QTY.	Item	
1	Portable freezer, cooler with dry ice, or cooler with bags of ice to store frozen samples	
1 copy	Field operations and method manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for periphyton	
For the optional targeted habitat periphyton sample, the following additional items are needed:		
1	1-L plastic bottle, with volume markings at 250-, 500-, 750-, and 1000-mL, labeled *TARGETED HABITAT PERIPHYTON SAMPLE*	
1	50-mL screw-top centrifuge tube with volume markings	
2 mL	Undiluted formalin solution for ID/Enumeration samples (10 mL if using diluted formalin).	
1 ea	Sample label and field data form for targeted habitat periphyton sample	

Figure 9-8. (Continued).

Montgomery, D.R. and J.M. Buffington, 1993. *Channel classification, prediction of channel response, and assessment of channel condition.* TFW-SH10-93-002. Prepared for the SHAMW committee of the Washington State Timber/Fish/Wildlife Agreement. Seattle, Washington.

Stevenson, R.J. and L.L. Bahls. 1999. Periphyton protocols. Pages 6-1 to 6-23 in M.D. Barbour et al. *Rapid bioassessment protocols for use in streams and wadeable rivers: periphyton, benthic macroinvertebrates, and fish.* 2nd edition. EPA 841/B-99/002. U.S. Environmental Protection Agency, Washington, D.C.

NOTES

SECTION 10
BENTHIC MACROINVERTEBRATES

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

Benthic macroinvertebrates inhabit the bottom substrates of streams and reflect overall biological condition (Kerans and Karr 1994, Barbour et al. 1999, Reynoldson et al. 2001, Klemm et al. 2002, 2003, Clarke et al. 2003, Bailey et al. 2004, Griffith et al. 2005). Monitoring these assemblages is useful in assessing the status of the water body and detecting trend in ecological condition. Benthic assemblages respond to a wide array of stressors in different ways, and it is often possible to determine the type of stress that has affected the macroinvertebrate assemblage (e.g., Klemm et al. 1990). Because many macroinvertebrates have life cycles of a year or more and are relatively immobile, macroinvertebrate assemblage structure is a function of present and past conditions (Barbour et al. 1999).

The EMAP-SW benthic macroinvertebrate protocol is intended to evaluate the biological condition of wadeable streams in the United States for the purpose of detecting stresses on assemblage structure and assessing the relative severity of these stresses. It is based on the Level III procedure for benthic macroinvertebrates of the Rapid Bioassessment Protocol (RBP; Plafkin et al. 1989, Barbour et al. 1999), which has been adopted for use by many states.

A representative sample of the benthic assemblage cannot be collected from a single point. Therefore, the response design is based on the collection of multiple kicknet samples from throughout the support reach. Results from Li et al. (2001) suggest the length of the support reach (40 channel widths) provides an adequate sample of the assemblage in terms of taxa richness and relative abundance. A fixed number (and thus

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

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

area) of collection points is allocated systematically throughout the support reach ensure distribution of samples among available major habitat types, eliminate individual sampler bias, and provide a comparable and consistent sample from every site. All samples collected at a site are combined into a single composite sample to characterize the support reach (and thus the sampling point) and reduce the cost and effort in processing and analysis (Patil et al. 1994, Barbour et al. 1999, Roth et al. 2002). The number of individual samples (11) is expected to provide a composite sample having a sufficient number of individuals to characterize the taxonomic composition and relative abundance of the assemblage (e.g., Larsen and Herlihy 1998).

Changes to the previously published protocol for EMAP-SW (Klemm et al. 1998) and modifications made during EMAP-W are summarized in Appendix B. Samples are collected from each support reach with a D-frame kick net that can be used by one person (Figure 10-1). Kick net samples are collected at the same time as periphyton samples (Section 9). The *biomorphs* (refer to Figure 2-1) collect kick net samples for benthic macro-invertebrates at sampling points at each cross-section transect. Typically, one person will collect the kick net samples, while a second person times the collection of samples and records information on the field data form. However, in swift waters, two persons may be necessary to collect the samples. All kick net samples collected at transects are combined into a single composite sample (termed the *reachwide* sample).

When sufficient riffle habitats are available at a site, additional kick net samples are collected from available riffle habitats found within the support reach. These kick net samples are combined to create a separate composite sample (termed the *targeted riffle* sample).

Note that any crayfish collected as part of the aquatic vertebrate sampling (Section 11) are ***not*** combined with any of the benthic macroinvertebrate samples described in this section.

10.1 SAMPLE COLLECTION

10.1.1 Reachwide Sample

Collect each kick net sample **1-m downstream** of each of the 11 cross-section transects (transects *A* through *K*) at an assigned sampling point (*Left*, *Center*, or *Right*).

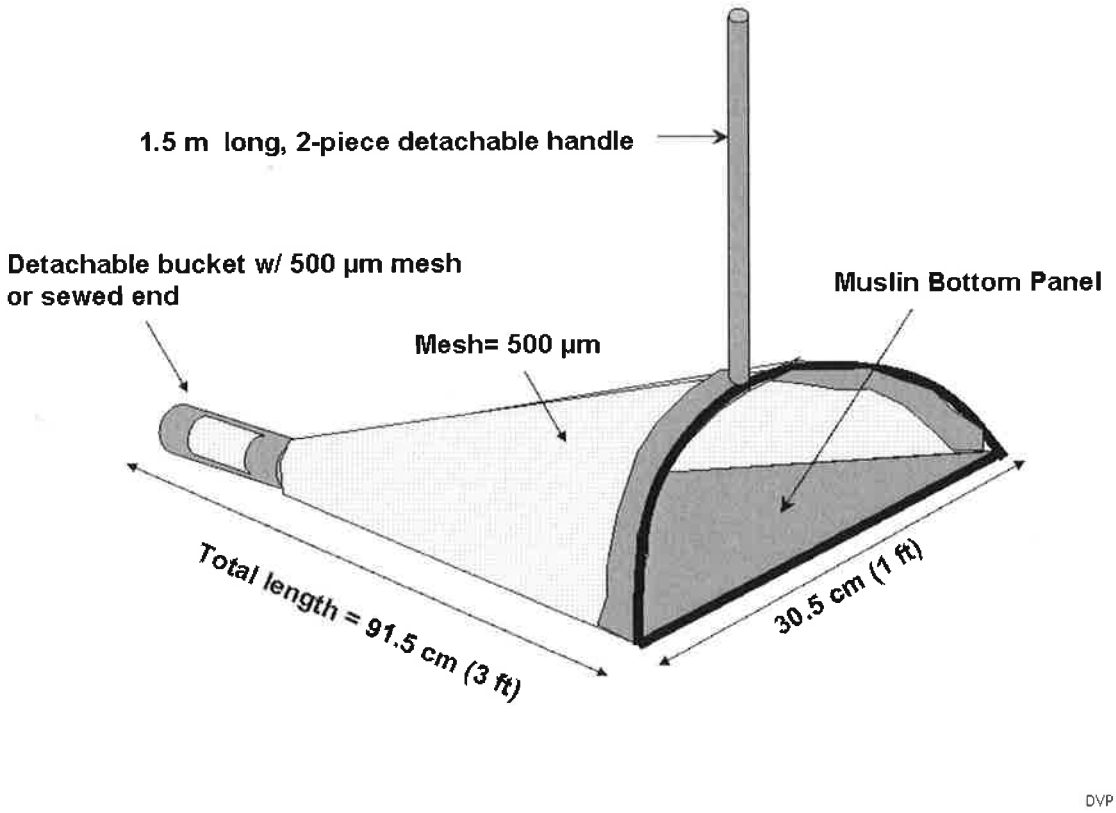


Figure 10-1. Modified D-frame kick net. (Not drawn to scale.)

as illustrated in Figure 10-2. These points may have been assigned when the support reach was laid out (refer also to Section 4; Table 4-3). If not, assign the sampling point at transect A at random using a digital watch or other suitable means. Once the first sampling point is determined, assign points at successive transects in order (*Left*, *Center*, *Right*). These are the same sampling points as those used for periphyton samples (Section 9). At transects assigned a *Center* sampling point where the stream width is between one and two net widths wide, pick either the *Left* or *Right* sampling point instead. If the stream is only one net width wide at a transect, place the net across the entire stream width and

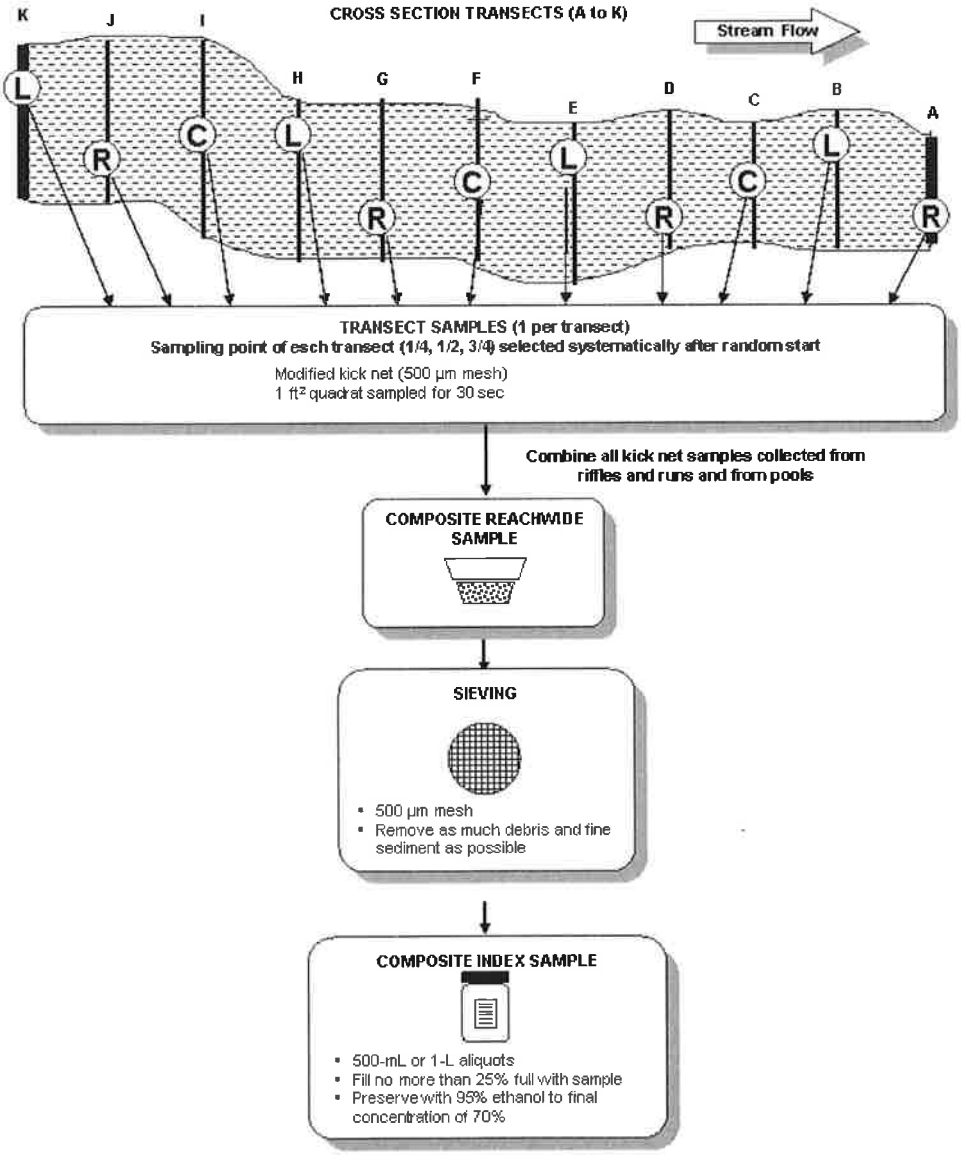


Figure 10-2. Response design for the reachwide benthic macroinvertebrate sample.

consider the sampling point to be *Center*. If a sampling point is located in water that is too deep or otherwise unsafe to wade, select an alternate sampling point on the transect at random.

Collect a kick net sample at each transect as described in Table 10-1. At each sampling point, determine if the *flowing water* or the *slack water* procedure is used based on whether or not there is enough current to extend the net. For each kick net sample, record the dominant substrate type (fine/sand, gravel, coarse substrate [coarse gravel or larger] or other [e.g., bedrock, hardpan, wood, aquatic vegetation, etc.]) and the habitat type (pool, glide, riffle, or rapid) on the Sample Collection Form as shown in Figure 10-3. Collect only from the upper 4 to 5 cm (1.5 to 2 in) of the substrate. As you go upstream from transect to transect, combine all the kick net samples into a container labeled *REACHWIDE*, no matter whether they were collected using the flowing water or slack water procedure.

If the kick net cannot be used to collect a sample at a flowing water sampling point, select 10 rocks from the area near the sampling point (but within the area of flowing water). Inspect and remove any organisms found on each rock and place them into the *REACH-WIDE* sampling container. If the kick net cannot be used at a slack water habitat due to an insufficient depth of water, spend about 30 seconds picking up pieces of substrate from a 0.09 m^2 (1 ft^2) area at the sampling point. Inspect and remove any organisms found on each piece of substrate and place them into the *REACHWIDE* sampling container. At vegetation-choked sampling points where neither procedure can be used, sweep the net through the vegetation for 30 seconds, then place the contents into the *REACHWIDE* sampling container.

10.1.2 Targeted Riffle Sample

Figure 10-4 illustrates the response design for the targeted riffle sample. Table 10-2 presents the procedure for selecting individual sampling points from the available riffle macrohabitat units found within the support reach. Note that if the total available area of riffle habitats is less than 8 ft^2 (i.e., such that eight non-overlapping kick net samples cannot be collected), do not collect a targeted riffle sample. There may be support reaches where more than one kick net sample is collected from a single riffle unit (but never from

TABLE 10-1. PROCEDURE TO COLLECT KICK NET SAMPLES FOR THE REACHWIDE COMPOSITE SAMPLE

1. At **1 m downstream** of each cross-section transect, beginning with transect *A*, locate the assigned sampling point (Left, Center, or Right as you face downstream) as 25%, 50%, and 75% of the wetted width, respectively. If you cannot collect a sample at the designated point because of deep water or unsafe conditions, relocate the point nearby on the same transect.

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

3. Determine if there is sufficient current in the area at the sampling point to extend the net fully. If so, use the *flowing water* procedure (go to Step 4). If not, use the *slack water* procedure (go to Step 10).
   For vegetation-choked sampling points where neither procedure can be used, sweep the net through the vegetation within a 0.09 m^2 (1 ft^2) quadrat for 30 seconds. Place the contents of this hand-picked sample into the *REACHWIDE* sampling container. Go to Step 15.

Flowing Water Procedure:

4. With the net opening facing upstream, position the net quickly and securely on the stream bottom to eliminate gaps under the frame. Avoid large rocks that prevent the sampler from seating properly on the stream bottom.
   NOTE: If there is too little water to collect the sample with the kick net, randomly pick up 10 rocks from the riffle and pick and wash the organisms off them into a bucket labeled *REACHWIDE* which is half-full of water.

5. Holding the net in position on the substrate, visually define a rectangular quadrat that is one net width wide and one net width long upstream of the net opening. The area within this quadrat is ~0.09 m^2 (1 ft^2). Alternatively, place a wire frame of the correct dimensions in front of the net to help delineate the quadrat to be sampled.

6. Hold the net in place with your knees. Check the quadrat for heavy organisms, such as mussels and snails. Remove these organisms from the substrate by hand and place them into the net. Pick up any loose rocks or other larger substrate particles in the quadrat. Use your hands or a small scrub brush to dislodge organisms so that they are washed into the net. Scrub all rocks that are golf ball-sized or larger and which are over halfway into the quadrat. Large rocks that are less than halfway into the sampling area are pushed aside. After scrubbing, place the substrate particles outside of the quadrat.

7. Keep holding the sampler securely in position. Start at the upstream end of the quadrat, use your foot and toes to **vigorously** kick the **upper 4 to 5 cm (1.5 to 2 in**) of the remaining finer substrate within the quadrat for 30 seconds (use a stopwatch). Avoid going too deep into the substrate with your kicking.

   NOTE: For samples located within dense beds of long, filamentous aquatic vegetation (e.g., algae or moss), kicking within the quadrat may not be sufficient to dislodge organisms in the vegetation. Usually these types of vegetation are lying flat against the substrate due to current. Use a knife or scissors to remove *only the vegetation that lies **within** the quadrat* (i.e., not entire strands that are rooted within the quadrat) and place it into the net.

(Continued)

TABLE 10-1 (Continued)

8. Pull the net up out of the water. Immerse the net in the stream several times to remove fine sediments and to concentrate organisms at the end of the net. Avoid having any water or material enter the mouth of the net during this operation.

9. Go to Step 14.

Slack Water Procedure:

10. Visually define a rectangular quadrat that is one net width wide and one net width long at the sampling point. The area within this quadrat is ~0.09 m^2 (1 ft^2). Alternatively, lay a wire frame of the correct dimensions in front of the net at the sampling point to help delineate the quadrat.

    NOTE: If there is not enough water present to use the net, spend 30 seconds collecting and examining pieces of substrate from about 0.09 m^2 (1 ft^2) of substrate at the sampling point.

11. Inspect the stream bottom within the quadrat for any heavy organisms, such as mussels and snails. Remove these organisms by hand and place them into the net or into a bucket labeled *REACHWIDE*. Pick up any loose rocks or other larger substrate particles within the quadrat and hold them in front of the net. Use your hands (or a scrub brush) to rub any clinging organisms off of rocks or other pieces of larger substrate (especially those covered with algae or other debris) into the net. After scrubbing, place the larger substrate particles outside of the quadrat.

12. Use your foot and toes to **vigorously** kick the **upper 4 to 5 cm (1.5 to 2 in)** of the remaining finer substrate within the quadrat while dragging the net repeatedly through the disturbed area just above the bottom. Keep moving the net all the time so that the organisms trapped in the net will not escape. Continue kicking the substrate and moving the net for 30 seconds. NOTE: If there is too little water to use the kick net, **vigorously** stir up the substrate with your gloved hands and use a sieve with 500 µm mesh size to collect the organisms from the water in the same way the net is used in larger pools.

11. After 30 seconds, remove the net from the water with a quick upstream motion to wash the organisms to the bottom of the net.

All samples:

12. Invert the net and transfer the contents into a bucket or wide-mouthed container with a lid marked *REACHWIDE*. Inspect the net for any residual organisms clinging to the net and deposit them into the *REACHWIDE* container. Use a squirt bottle with ethanol and watchmakers' forceps if necessary to remove organisms from the net. Carefully inspect any large objects (such as rocks, sticks, and leaves) in the bucket and wash any organisms found off of the objects and into the bucket before discarding the object. Remove as much detritus as possible without losing any organisms. Replace the lid on the bucket or container.

(Continued)

TABLE 10-1 (Continued)

All samples:

13. Determine the **predominant** substrate size/type you observed within the sampling quadrat. Place an *X* in the appropriate substrate type box for the transect on the Sample Collection Form. NOTE: If there is/are co-dominant substrate type(s), you may check more than one box; note the co-dominants in the comments section of the form.

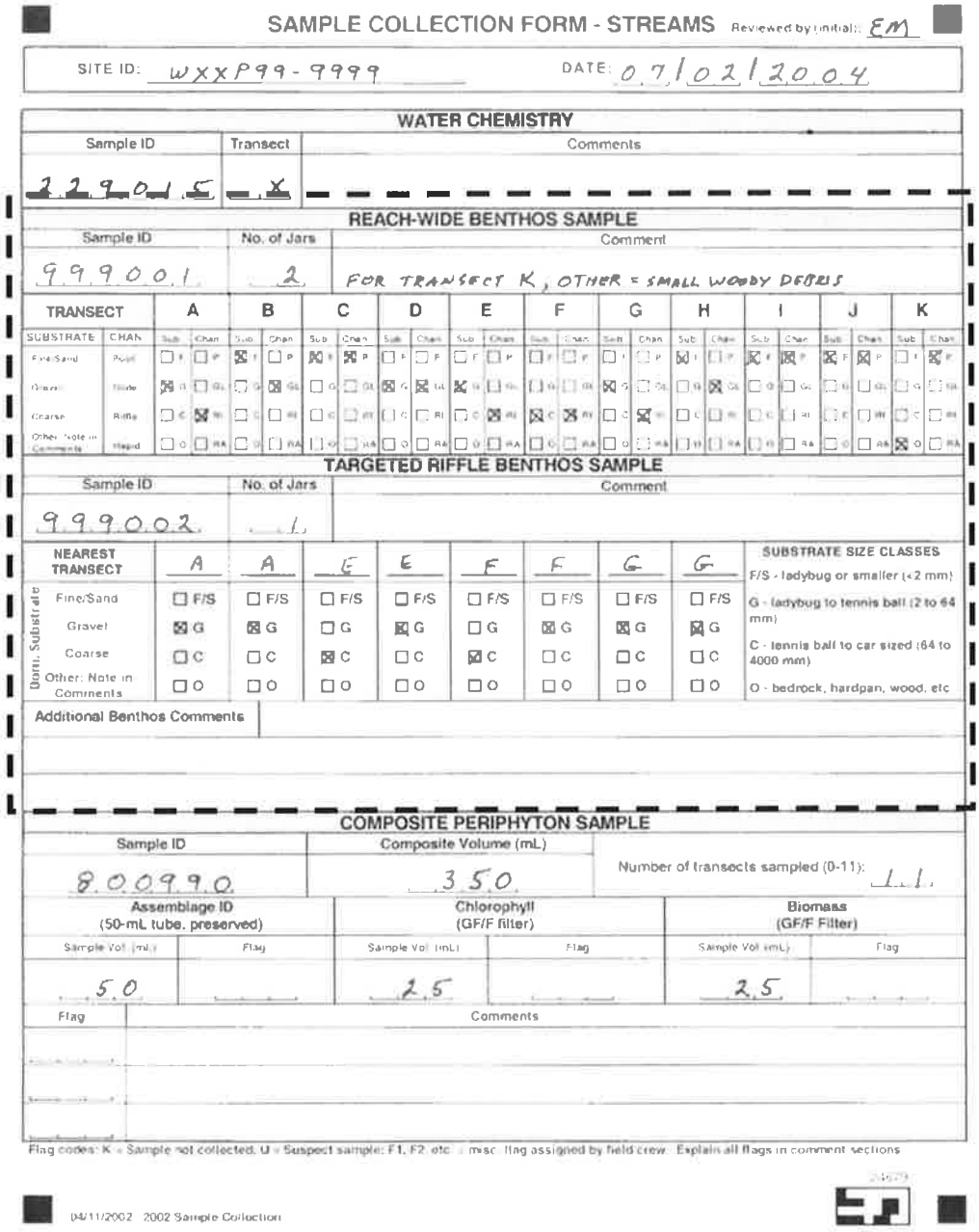
    Fine/sand: not gritty (silt/clay/muck < 0.06 mm diam.) to gritty, up to ladybug sized (2 mm diam.)
    Gravel: fine to coarse gravel (ladybug to tennis ball sized; 2 mm to 64 mm diam.)
    Coarse: Cobble to boulder (tennis ball to car sized; 64 mm to 4000 mm)
    Other: bedrock (larger than car sized; > 4000 mm), hardpan (firm, consolidated fine substrate), wood of any size, aquatic vegetation, etc.). Note type of "other" substrate in comments on field form.

14. Identify the habitat type where the sampling quadrat was located. Place an "X" in the appropriate channel habitat type box for the transect on the Sample collection Form.

    Pool; Still water; low velocity; with smooth, glassy surface; usually deep compared to other parts of the channel.
    GLide: Water moving slowly, with smooth, unbroken surface; low turbulence.
    RIffle: Water moving, with small ripples, waves, and eddies; waves not breaking, and surface tension is not broken; "babbling" or "gurgling" sound.
    RApid: Water movement is rapid and turbulent; surface with intermittent "white water" with breaking waves; continuous rushing sound.

    NOTE: This habitat type may be different from that recorded during the thalweg profile (Section 7), which is based on a channel width scale, rather than a quadrat scale. Also, this habitat may be different from the operational definition used to select the sampling procedure (flowing water vs. slack water), which is based solely on current velocity

15. Thoroughly rinse the net before proceeding to the next sampling location. Proceed upstream to the next transect (including transect *K*, the upstream end of the support reach) and repeat Steps 1 through 9. Combine all kick net samples from flowing water and slack water habitats into the *REACHWIDE* container.



SAMPLE COLLECTION FORM - STREAMS Reviewed by (initial): EM

SITE ID: WXXP99-9999 DATE: 07/02/2004

WATER CHEMISTRY

Sample ID	Transect	Comments
229015	X	

REACH-WIDE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999001	2	FOR TRANSECT K, OTHER = SMALL WOODY DEBRIS

TRANSECT	A	B	C	D	E	F	G	H	I	J	K
SUBSTRATE / CHAN	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan	Sub / Chan
Fine/Sand — Pool	☐ F / ☐ P	☒ F / ☐ P	☒ F / ☒ P	☐ F / ☐ P	☐ F / ☐ P	☐ F / ☐ P	☐ F / ☐ P	☒ F / ☐ P	☒ F / ☒ P	☒ F / ☒ P	☐ F / ☒ P
Gravel — Glide	☒ G / ☐ GL	☐ G / ☒ GL	☐ G / ☐ GL	☒ G / ☒ GL	☒ G / ☐ GL	☐ G / ☐ GL	☒ G / ☐ GL	☐ G / ☒ GL	☐ G / ☐ GL	☐ G / ☐ GL	☐ G / ☐ GL
Coarse — Riffle	☐ C / ☒ RI	☐ C / ☐ RI	☐ C / ☐ RI	☐ C / ☐ RI	☐ C / ☒ RI	☒ C / ☒ RI	☐ C / ☒ RI	☐ C / ☐ RI	☐ C / ☐ RI	☐ C / ☐ RI	☐ C / ☐ RI
Other: Note in Comments — Rapid	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☐ O / ☐ RA	☒ O / ☐ RA

TARGETED RIFFLE BENTHOS SAMPLE

Sample ID	No. of Jars	Comment
999002	1	

NEAREST TRANSECT	A	A	E	E	F	F	G	G
Dom. Substrate: Fine/Sand	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S	☐ F/S
Gravel	☒ G	☒ G	☐ G	☒ G	☐ G	☒ G	☒ G	☒ G
Coarse	☐ C	☐ C	☒ C	☐ C	☒ C	☐ C	☐ C	☐ C
Other: Note in Comments	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O	☐ O

SUBSTRATE SIZE CLASSES
F/S - ladybug or smaller (<2 mm)
G - ladybug to tennis ball (2 to 64 mm)
C - tennis ball to car sized (64 to 4000 mm)
O - bedrock, hardpan, wood, etc

Additional Benthos Comments

COMPOSITE PERIPHYTON SAMPLE

Sample ID	Composite Volume (mL)	Number of transects sampled (0-11):
800990	350	11

Assemblage ID (50-mL tube, preserved)		Chlorophyll (GF/F filter)		Biomass (GF/F Filter)	
Sample Vol (mL)	Flag	Sample Vol (mL)	Flag	Sample Vol (mL)	Flag
5.0		2.5		2.5	

Flag	Comments

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

04/11/2002 2002 Sample Collection

Figure 10-3. Sample Collection Form (page 1), showing information for the reachwide and targeted riffle benthic macroinvertebrate samples.

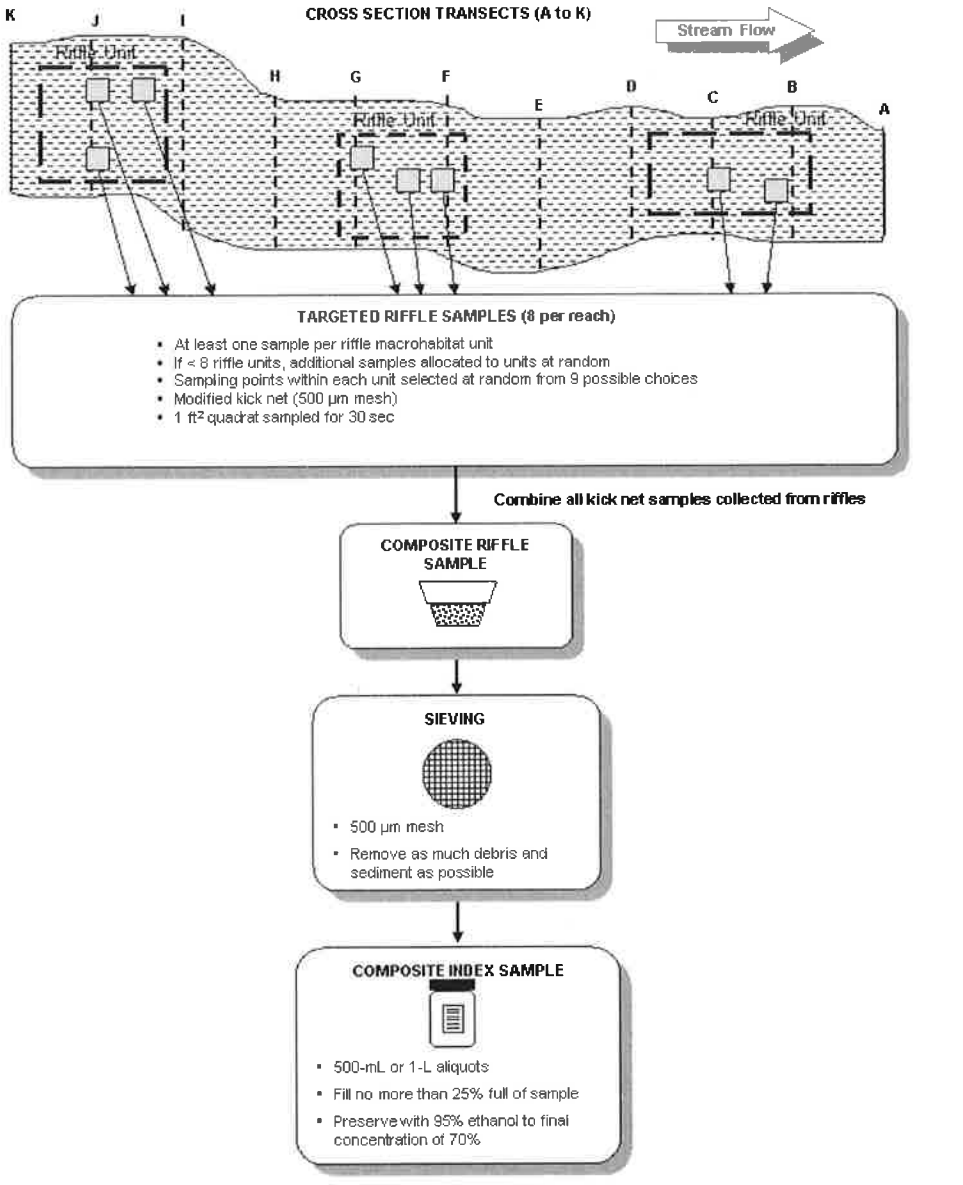


Figure 10-4. Response design for the targeted riffle benthic macroinvertebrate sample.

TABLE 10-2. LOCATING SAMPLING POINTS FOR KICK NET SAMPLES: TARGETED RIFFLE SAMPLE

1. Before sampling, survey the stream reach to visually estimate the total number (and area) of riffle macrohabitat "units" contained in the defined support reach. To be considered as a unit, the area of the riffle must be greater than 1 ft^2 (0.09 m^2).

   A. **Do not** collect a targeted riffle sample if the support reach contains less than **8 ft^2** of riffle macrohabitat.

   B. If the support reach contains more than one distinct riffle macrohabitat unit but fewer than eight units, allocate the eight sampling points among the units so as to spread the effort throughout the support reach as much as possible. You may need to collect more than one kick sample from a given riffle unit.

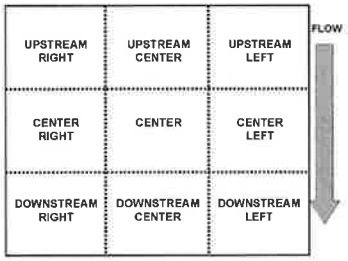
   C. If the number of riffle macrohabitat units is greater than eight, skip one or more habitat units at random as you work upstream, again attempting to spread the sampling points throughout the support reach.

   NOTE: The collection of targeted riffle samples can be conducted in conjunction with the collection of reachwide samples to minimize the number of passes up the reach. However, extreme care must be taken to ensure each type of sample is placed into the correct sample container (*REACHWIDE* or *TARGETED RIFFLE*) to avoid compromising both samples.

2. To minimize instream disturbance, begin sampling at the most downstream riffle unit, and sample riffle macrohabitat units as they are encountered.

3. At each unit, constrain the potential sampling area to exclude *margin habitats*. Margin habitats are edges (e.g., along channel margins or upstream or downstream ends) of the riffle macrohabitat unit. Define a core area for each riffle unit as the central portion, visually estimating a buffer strip circumscribing the identified unit. In some cases, the macrohabitat unit may be so small that it will not be feasible to define a core area and avoid an edge.

4. Visually lay out the core area of the unit sampled into 9 equal plots (i.e., a 3 × 3 grid) as shown below, and select a plot for sampling at random



5. Collect the kick net sample in the center of the selected plot as described in Table 10-3.

6. If one or more additional samples are required from a single macrohabitat unit, select additional plots at random as described in Step 4.

within the same plot in the 3 × 3 grid as described in Table 10-2). The objective for selecting sampling points within the available riffle macrohabitat units is to allocate points throughout the support reach as much as possible.

Procedures for collecting a kick net sample from riffle macrohabitat units are presented in Table 10-3. At each sampling point, a 1 ft^2 (0.09 m^2) quadrat is sampled. If you are collecting both reachwide and targeted riffle samples in the order they are encountered during a single pass through the reach, it is very important to rinse the kick net thoroughly between samples to avoid cross-contamination of the targeted riffle sample and the reachwide sample. Also, make sure you put the contents from a kick sample into the correct container.

10.2 SAMPLE PROCESSING

After collecting kick net samples for both the reachwide and targeted riffle samples, prepare two composite index samples from the contents of the *REACHWIDE* and *TARGETED RIFFLE* containers as described in Table 10-4. Record tracking information for each composite sample on the Sample Collection Form as shown in Figure 10-3. ***Do not*** *fill out the collection form until you have confirmed that you will collect samples at the site.* If the sample collection form is filled out before you arrive at the site, and then no samples are collected, a lot of time is wasted later by others searching for samples that do not exist.

A set of completed sample labels, including the label that is used if more than one jar is required for a single composite sample, is shown in Figure 10-5. Note that each composite sample (reachwide and targeted riffle) has a different sample ID number (barcode). The ID number is also recorded on a waterproof label that is placed inside each jar (Figure 10-5, lower right). If more than one jar is used for a composite sample, use a special label (Figure 10-5, lower left) to record the ID number assigned to the sample. ***Do not*** *use two different barcode numbers on two jars containing one single sample.* Blank labels for use inside of sample jars are presented in Figure 10-6. These can be copied onto waterproof paper. If a sample requires more than one jar, make sure the correct number of jars for the sample is recorded on the Sample Collection Form. Again, accurate record keeping in the field saves substantial amounts of time later.

TABLE 10-3. COLLECTING A KICK NET SAMPLE FOR THE TARGETED RIFFLE COMPOSITE SAMPLE

1. Beginning at the most downstream riffle unit within the sampling reach, locate the sampling point within the macrohabitat unit as described in Table 10-2. ***Do not*** *collect samples on established transects to avoid disturbing substrates that are part of the physical habitat characterization. Move the sampling point* ***downstream*** *of the transect.*

2. Position the kick net quickly and securely on the stream bottom so as to eliminate gaps between the frame and the stream bottom.

3. Hold the sampler firmly in position on the substrate. Define a quadrat immediately upstream from the mouth of the net having a width equal to the width of the net frame (total area = 1 ft^2 [0.09 m^2]).

4 Hold the kick net in place with your knees and pick up any loose rocks or other larger substrate particles in the quadrat. Use your hands or a small scrub brush to dislodge organisms so that they are washed into the kick net. Scrub all rocks that are golf ball-sized or larger and which are over halfway into the quadrat. Large rocks that are less than halfway into the sampling area are pushed aside. After scrubbing, place the substrate particles outside of the quadrat.

6. Keep holding the kick net securely in position. Starting at the upstream end of the quadrat, use your foot and toes to **vigorously** kick the **upper 4 to 5 cm (1.5 to 2 in)** of the remaining finer substrate within the quadrat for 30 seconds (use a stopwatch).

   NOTE: For samples located within dense beds of long, filamentous aquatic vegetation (e.g., algae or moss), kicking within the quadrat may not be sufficient to dislodge organisms in the vegetation. Usually these types of vegetation are lying flat against the substrate due to current. Use a knife or scissors to remove *only the vegetation that lies* ***within*** *the quadrat* (i.e., not entire strands that are rooted within the quadrat) and place it into the kick net.

7. Pull the kick net up out of the water. Immerse the kick net in the stream several times to remove fine sediments and to concentrate organisms at the end of the net. Avoid having any water or material enter the mouth of the kick net during this operation.

8. Invert the kick net into a wide-mouthed container or a plastic bucket with a lid marked *TARGETED RIFFLE* and transfer the sample. Inspect the kick net for any residual organisms clinging to the net and deposit them into the *TARGETED RIFFLE* container. Use a squirt bottle filled with ethanol and watchmakers' forceps if necessary to remove organisms from the net and place them into the container. Replace the lid on the container.

(Continued)

TABLE 10-3. (Continued)

9. Record the nearest transect location in the box for the sample on the Sample Collection Form. Also note the dominant substrate type by checking the appropriate box on the Sample Collection Form. Identify the **predominant** substrate size/type you observed within the sampling quadrat.

   Fine/sand: not gritty (silt/clay/muck < 0.06 mm diam.) to gritty, up to ladybug sized (2 mm diam.)
   Gravel: fine to coarse gravel (ladybug to tennis ball sized; 92 mm to 64 mm diam.)
   Coarse: Cobble to boulder (tennis ball to car sized; 64 mm to 4000 mm)
   Other: bedrock (larger than car sized; > 4000 mm), hardpan (firm, consolidated fine substrate), wood of any size, aquatic vegetation, etc.). Note type of "other" substrate in comments on field form.

10. Rinse the kick net thoroughly before proceeding to the next sampling location (either the next riffle unit or a different quadrat location within the same riffle unit).

11. Repeat Steps 1-10 at subsequent riffle sampling points until 8 kick net samples have been collected and placed into the *TARGETED RIFFLE* container.

TABLE 10-4. PROCEDURE FOR PREPARING COMPOSITE SAMPLES FOR BENTHIC MACROINVERTEBRATES

1. Pour off the water from the *REACHWIDE* (or *TARGETED RIFFLE*) bucket through a sieve (or sieve bucket) with 500 µm mesh size. Remove any large objects such as sticks, rocks, or plant material from the bucket or container. Inspect these objects carefully and dislodge any clinging organisms back into the bucket or container before discarding.

2. Estimate the total volume of the sample in the sieve and determine the size (500-mL or 1-L) and number of jars that will be needed for the sample. Avoid using more than one jar for each of the composite samples if possible (but don't overfill the jar with sample either).

3. Fill in a *REACHWIDE* (or *TARGETED RIFFLE*) sample label with the stream ID and date of collection. Attach the completed label to the jar and cover it with a strip of clear tape. Record the sample ID number for the composite sample on the Sample Collection Form. For each composite sample, make sure the number on the form matches the number on the label. ***Do not*** *record an ID number on the form until you have collected the sample!*

4. Wash the contents of the bucket or container to one side. Transfer the sample from the bucket or container into a jar, using a large-bore funnel if necessary. Use as little water from the wash bottle as possible to help transfer material. If the jar becomes too full of liquid, carefully pour off the water through the sieve. Continue to transfer sample material to the jar until it ***is not*** *more than ¼ full of solid material.* Use additional jars for the remaining sample. Carefully examine the bucket or container for any remaining organisms and use watchmakers' forceps to place them into the sample jar.

    - If a second jar is needed, fill in a sample label that does not have a pre-printed ID number on it. Record the ID number from the pre-printed label prepared in Step 4 in the *SAMPLE ID* field of the label. Attach the label to the second jar and cover it with a strip of clear tape. Record the number of jars required for the sample on the Sample Collection Form. *Make sure the number you record matches the actual number of jars used.* If possible, write *Jar N of X* on each sample label using a waterproof marker (*N* is the individual jar number, and *X* is the total number of jars for the sample).

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

    - Stream Number
    - Type of sampler and mesh size used
    - Sample type (*Reachwide* or *Targeted riffle*)
    - Name of stream
    - Date of collection
    - Collector's initials
    - Number of kick net samples collected

(Continued)

TABLE 10-4 (Continued)

6. Remove as much water as you can from each sample jar without including any sample material by pouring it through the sieve. **Completely fill** each jar with 95% ethanol (no head-space) so that the final concentration of ethanol is between 75 and 90%. It is very important that sufficient ethanol be used, or the organisms will not be properly preserved.
    - NOTE: Prepared composite samples can be transported back to the vehicle before adding ethanol if necessary. Fill each jar with stream water to cushion the sample from the grinding action of non-biological material in the sample. Replace the water with ethanol at the vehicle. Others (G. Lester, personal communication) suggest samples can be transported without water with no damage to specimens, or a minimal amount of ethanol can be added in the field to start the preservation process.
7. Replace the lid on each jar. Slowly tip the jar to a horizontal position, then gently rotate the jar to mix the preservative. Do not invert or shake the jar. After mixing, seal each jar with plastic tape.
8. Repeat Steps 1 through 8 for the *TARGETED RIFFLE* container.
9. Store the labeled sample jars in a container with absorbent material that is suitable for use with 95% ethanol until transport or shipment to the laboratory.

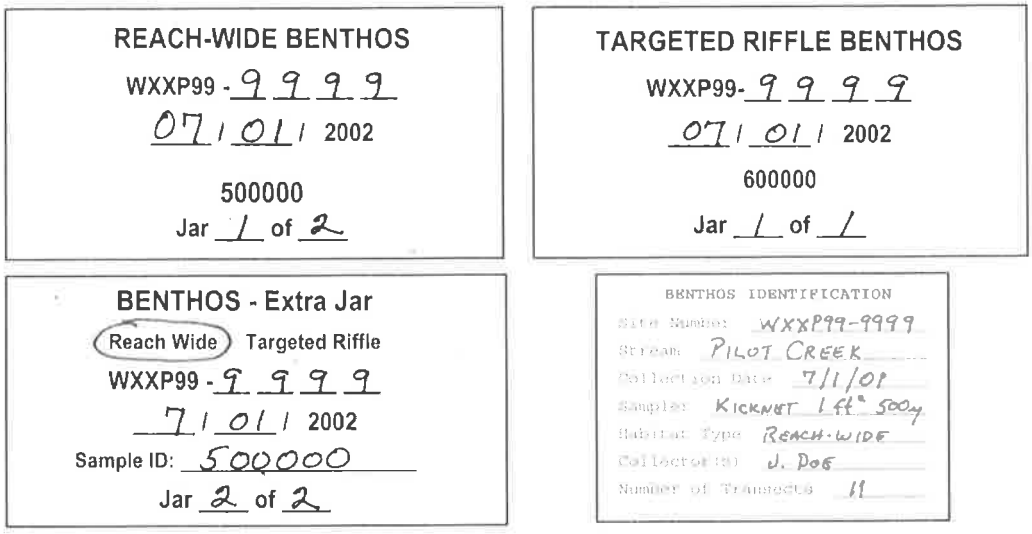


Figure 10-5. Completed labels for benthic macroinvertebrate samples. The label at lower left is used if more than one jar is required for a composite sample. The label at lower right is placed inside each sample container.

Check to be sure that the completed label on each jar is covered with clear tape, and a waterproof label is in each jar and filled in properly. Confirm that the inside and outside labels describe the same sample. Replace the lid on each jar and seal with plastic electrical tape. It is helpful to mark the lid of each jar with the site number and habitat type (*Reachwide* or *Targeted Riffle*); use a permanent marker or write on a piece of light-colored tape (or a small blank address label) and attach it to the lid. Place the sample jars in a cooler or other secure container for transporting and/or shipping the laboratory (see Section 3). The container and absorbent material should both be suitable for transporting ethanol. Check to see that all equipment is in the vehicle.

10.3 EQUIPMENT AND SUPPLIES

Figure 10-7 shows the checklist of equipment and supplies required to complete the collection of benthic macroinvertebrates from streams. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3) to ensure

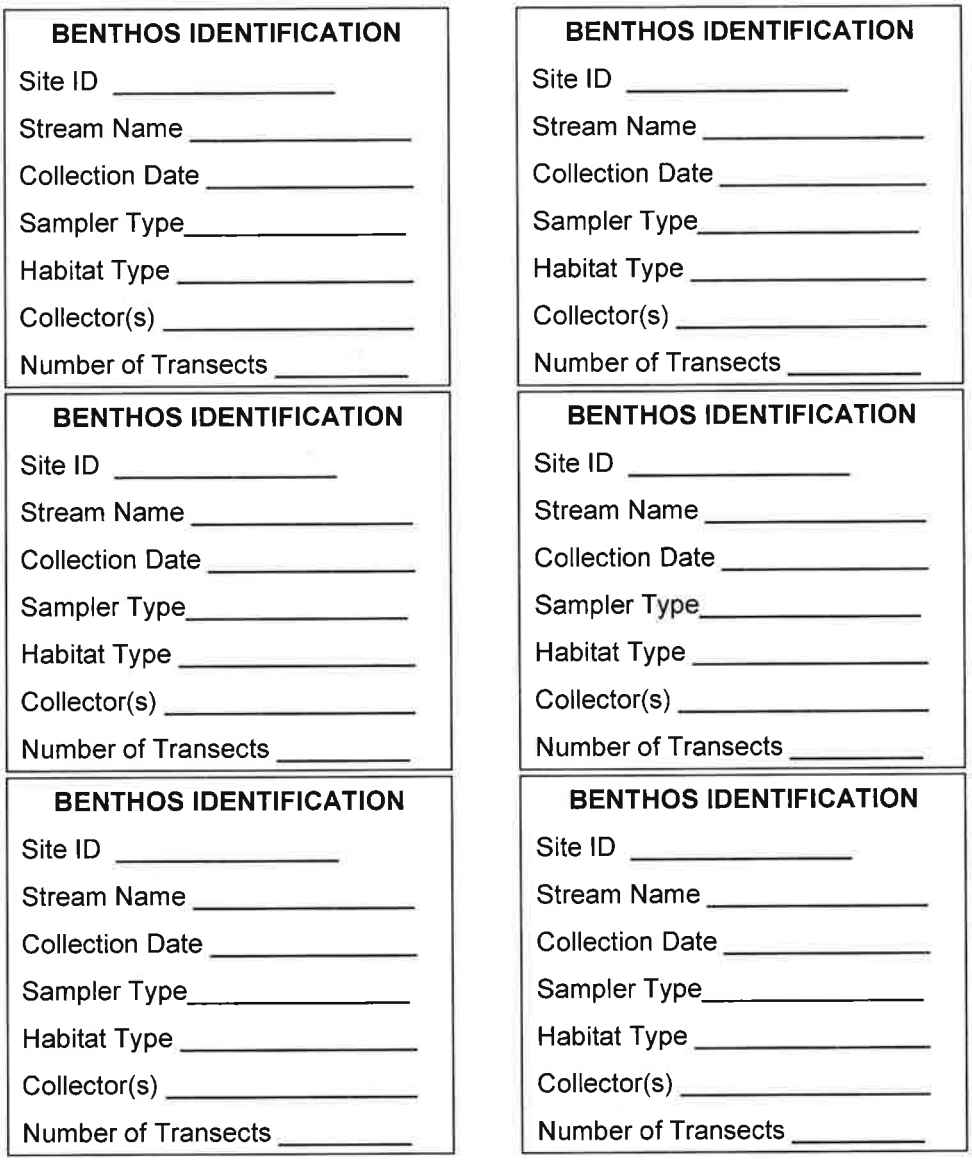


Figure 10-6. Blank labels for benthic invertebrate samples. This label is placed inside of each sample container.

EQUIPMENT AND SUPPLIES FOR BENTHIC MACROINVERTEBRATES

QTY.	ITEM	
1	Modified kick net (D-frame with 500 µm mesh) and 4-ft handle (Wildco #425-C50)	
	Spare net(s) and/or spare bucket assembly for end of net	
1	Watch with timer or a stopwatch	
2	Buckets, plastic, 8- to 10-qt capacity, labeled *REACHWIDE* and *TARGETED RIFFLE*	
1	Sieve with 500 µm mesh openings or Sieve-bottomed bucket, 500 µm mesh openings	
2 pr.	Watchmakers' forceps (straight and curved)	
1	Wash bottle, 1-L capacity labeled *STREAM WATER*	
1	Small spatula, spoon, or scoop to transfer sample	
1	Funnel, with large bore spout	
4 to 6 each	Sample jars, HDPE plastic with leakproof screw caps, 500-mL and/or 1-L capacity, suitable for use with ethanol	
2 gal	95% ethanol, in a proper container	
2 pr.	Rubber gloves suitable for use with ethanol	
1	Cooler (with suitable absorbent material) for transporting ethanol and samples	
2	Preprinted benthic sample labels with sample ID numbers	
4	Preprinted benthic sample labels without sample ID numbers	
6	Blank labels on waterproof paper for inside of jars	
1	Sample Collection Form for site	
	Soft (#2) lead pencils	
	Fine-tip indelible markers	
1 pkg.	Clear tape strips	
4 rolls	Plastic electrical tape	
1	Knife, pocket, with at least two blades	
1	Scissors	
1	Pocket-sized field notebook (optional)	
1 pkg.	Kim wipes in small resealable plastic bag	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for benthic macroinvertebrates	

Figure 10-7. Equipment and supply checklist for benthic macroinvertebrates.

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

10.4 LITERATURE CITED

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NOTES

SECTION 11
AQUATIC VERTEBRATES

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

Freshwater fish have long been a focus of bioassessment activities (Klemm et al. 1993), and were the subject of the first attempts to develop multimetric indicators of ecological condition based on assemblage characteristics (Gammon 1980, Karr 1981, Karr et al. 1986, 1991). Fish assemblages offer several unique advantages to assess ecological condition from other biological assemblages, based on their mobility, longevity, and trophic relationships (Plafkin et al. 1989, Barbour et al. 1999). Other aquatic vertebrates, most notably amphibians, are important components of aquatic communities in many parts of the U.S. (Hairston 1987, Bury et al. 1991), and provide information regarding ecological condition in streams where fish are rare or absent (e.g., high-elevation streams, headwater streams, and streams above natural colonization barriers).

The response design for aquatic vertebrates is similar to that developed for other bioassessment programs such as the RBP (Barbour et al. 1999). The objective is to collect a representative sample of all except very rare species in the assemblage. A sample from a single point does not provide an adequate representation of the vertebrate assemblage present. Sampling effort is allocated along the entire length of the support reach established at each sampling point. The length of the support reach (40 times the mean wetted width) is expected to allow collection of 90 percent of the species present based on several different studies (Patton et al. 2000, Cao et al. 2001, 2002, Reynolds et al. 2003).

Changes to previously published EMAP-SW procedures (McCormick and Hughes 1998), and modifications made during EMAP-W are summarized in Appendix B. Sampling fish, amphibian, and aquatic reptile species to estimate their proportionate abundances

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and the presence of external anomalies is conducted after all other field sampling and measurement activities are completed. All team members are involved in collecting aquatic vertebrates. Backpack electrofishers are used as the principal sampling gear (Section 11.1.1.1). Bank or towed electrofishers are recommended for wide but shallow streams (Section 11.1.1.2), and seining (Section 11.1.1.3) is used in habitats where high conductivity or turbidity preclude electrofishing. In addition to gathering data on the assemblage, fish specimens are retained for analysis of tissue contaminants (Section 12). Data collected by these procedures are used to assess fish (or aquatic vertebrate) assemblage condition across the region of interest (e.g., McCormick et al. 2001, Hughes et al. 1998, 2004, Bramblett et al. 2005).

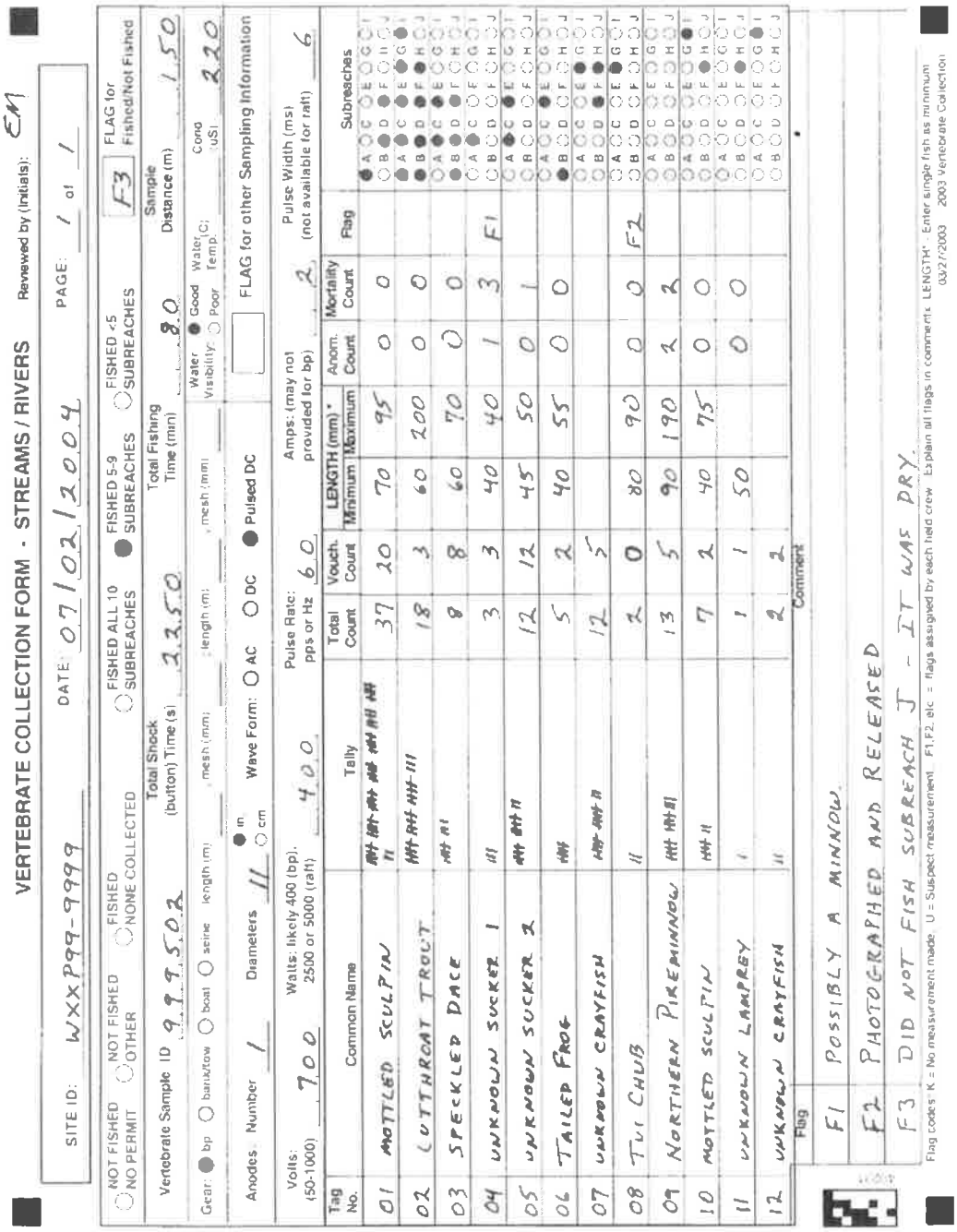
In addition, any crayfish specimens collected incidentally during aquatic vertebrate sampling are retained for identification, but are not combined with the benthic macroinvertebrate sample (Section 10). Crayfish collection data are used to estimate the potential extent of nonnative crayfish in the western U.S.

11.1 SAMPLE COLLECTION

The entire channel within the support reach is allocated among 10 *subreaches* (i.e., areas between transects established as described in Section 4, and equivalent to *segments* described in Section 7) to distribute effort along the entire reach. Total collection time for a site should be 45 minutes to 3 hours (Section 4) to obtain a representative sample. If a support reach is very wide, however, it may take two days to sample it effectively. If it appears that the support reach is wide enough that three hours of sampling will only allow you to sample 50% or less of the available surface area of the reach effectively, you should plan to spend two days to sample, and/or use the modified procedure for bank/towed electrofishing (Section 11.1.1.2). Record sampling data and general comments (perceived fishing efficiency, missed fish, gear operation, suggestions) on the Vertebrate Collection Form (Figure 11-1).

11.1.1 Electrofishing

The use of electrofishing gear to collect aquatic vertebrates requires training to do it safely and correctly to reduce the potential for injury or mortality to both humans and



VERTEBRATE COLLECTION FORM - STREAMS / RIVERS

Reviewed by (initials): EM

SITE ID: WXXP99-9999 DATE: 07/02/2004 PAGE: 1 of 1

○ NOT FISHED ○ NO PERMIT ○ OTHER ○ FISHED ○ NONE COLLECTED ○ FISHED ALL 10 SUBREACHES ● FISHED 5-9 SUBREACHES ○ FISHED <5 SUBREACHES F3 FLAG for Fished/Not Fished

Vertebrate Sample ID 991502 Total Shock (button) Time (s) 2350 Total Fishing Time (min) 30 Sample Distance (m) 150

Gear: ● bp ○ barkdow ○ boat ○ seine length (m): ___ mesh (mm): ___ length (m): ___ mesh (mm): ___ Water Visibility: ● Good ○ Poor Water Temp (C): ___ Cond uS: 220

Anodes: Number 1 Diameters 11 ● in ○ cm Wave Form: ○ AC ○ DC ● Pulsed DC ☐ FLAG for other Sampling Information

Volts: (50-1000) 700 Watts: likely 400 (bp), 2500 or 5000 (raft) 400 Pulse Rate: pps or Hz 60 Amps: (may not provided for bp) 2 Pulse Width (ms) (not available for raft) 6

Tag No.	Common Name	Tally	Total Count	Vouch. Count	LENGTH (mm)* Minimum	LENGTH (mm)* Maximum	Anom. Count	Mortality Count	Flag	Subreaches
01	MOTTLED SCULPIN	37	37	20	70	95	0	0		A B C D E F G H I J
02	CUTTHROAT TROUT	18	18	3	60	200	0	0		A B C D E F G H I J
03	SPECKLED DACE	8	8	8	60	70	0	0		A B C D E F G H I J
04	UNKNOWN SUCKER 1	3	3	3	40	40	1	3	F1	A B C D E F G H I J
05	UNKNOWN SUCKER 2	12	12	12	45	50	0	1		A B C D E F G H I J
06	TAILED FROG	5	5	2	40	55	0	0		A B C D E F G H I J
07	UNKNOWN CRAYFISH	12	12	5						A B C D E F G H I J
08	TUI CHUB	2	2	0	80	90	0	0	F2	A B C D E F G H I J
09	NORTHERN PIKEMINNOW	13	13	5	90	190	2	2		A B C D E F G H I J
10	MOTTLED SCULPIN	7	7	2	40	75	0	0		A B C D E F G H I J
11	UNKNOWN LAMPREY	1	1	1	50		0	0		A B C D E F G H I J
12	UNKNOWN CRAYFISH	2	2	2						A B C D E F G H I J

Flag	Comment
F1	POSSIBLY A MINNOW
F2	PHOTOGRAPHED AND RELEASED
F3	DID NOT FISH SUBREACH J - IT WAS DRY

Flag codes: K = No measurement made, U = Suspect measurement, F1,F2, etc = flags assigned by each field crew. Explain all flags in comments. LENGTH* - Enter single fish as minimum

03/27/2003 2003 Vertebrate Collection

Figure 11-1. Vertebrate Collection Form (page1).

aquatic vertebrates. Such training is becoming a prerequisite for obtaining a scientific collection permit from many State and Federal agencies (e.g., National Oceanic and Atmospheric Administration [NOAA] National Marine Fisheries Service). Information on training programs is available from the U.S. Fish and Wildlife Service National Conservation Training Center website (*http://training.fws.gov/BART/courses.html*), training courses may also be offered by commercial organizations or through local educational institutions. Texts such as Kolz et al. (1998) also provide information on training course content.

Primary responsibility for safety while electrofishing rests with the sampling team leader (Section 2). Electrofishing units can deliver a fatal electrical shock. While electrofishing, avoid contact with the water unless sufficiently insulated against electrical shock. Use chest waders with nonslip soles and heavy rubber "linesmen" gloves (*NOTE: some types of "breathable" waders* ***do not*** *provide adequate insulation against electric current when wet*). If waders become wet inside, stop electrofishing until they are thoroughly dry or use a dry pair. ***Always*** *avoid contact with the anode and cathode due to the potential shock hazard.* If you perspire heavily, wear polypropylene or other wicking and insulating clothing instead of cotton. If it is necessary for a team member to reach into the water to pick up a fish or something that was dropped, do so only after the electrical current is off and the anode is removed from the water. Do not resume electrofishing until all individuals are clear of the electroshock hazard. The electrofishing equipment is equipped with a 45° tilt switch that interrupts the current. Do not make any modifications to the electrofishing unit that would hinder turning off the electricity. Avoid electrofishing near unprotected people, pets, or livestock. Discontinue activity during thunderstorms or heavy rain. Team members should keep each other in constant view or communication while electrofishing. For each site, know the location of the nearest emergency care facility. Although the team leader has authority, each team member has the responsibility to question and modify an operation or to decline participation if it is unsafe.

11.1.1.1 Backpack Electrofishing--

The backpack electrofishing procedure is presented in Table 11-1; record information on the Vertebrate Collection Form (Figure 11-1). If the support reach cannot be completely sampled by either electrofishing or seining, fill in the appropriate "bubble" on the form, and explain why in the comments. If the stream is too wide to sample the width of the channel over the entire support reach in the 3-hour time limit effectively (i.e., you can only sample 50% or less of the available surface area), plan on two days of sampling

TABLE 11-1. BACKPACK ELECTROFISHING PROCEDURES

1. Allocate the total fishing time (45-180 min) among all subreaches based on stream size and complexity. If you cannot effectively sample at least 50% of the available area in the 180-minute (3 h) time limit, you should spend two days sampling, and consider using the modified procedures for bank/towed electrofishing.

2. Complete header information on the Vertebrate Collection Form.

3. ***Review all collecting permits*** *to determine if any sampling restrictions are in effect for the reach. In some cases, you may have to cease sampling if you encounter certain listed species, or use alternate gear types.*

   If you cannot sample a reach because of permit restrictions, mark the *NOT FISHED-NO PERMIT* bubble on the Vertebrate Collection Form.

   Search and sample for aquatic vertebrates and crayfish even if the stream is extremely small, and it appears that sampling may produce no specimens. If no individuals are collected, mark the *FISHED-NONE COLLECTED* bubble on the Vertebrate Collection Form. Explain why in comments section.

   If conductivity, turbidity, or depth precludes backpack electrofishing, sample by seining or bank/towed electrofishing if possible, otherwise do not sample. If you do not sample, mark the *NOT FISHED-OTHER* bubble on the Vertebrate Collection Form and explain why in the comments section of the form.

4. Set the unit to *pulsed DC*. Select initial voltage setting (150-400 V for high conductivity [>300 S/cm]; 500-800 V for medium conductivity [100 to 300 S/cm]; 900-1100 V for low conductivity [< 100 S/cm] waters). In waters with strong-swimming fish (length >200 mm), use a pulse rate of 30 Hz with a pulse width of 2 msec. If mostly small fish are expected, use a pulse rate of 60-70 Hz. Start the electrofisher, set the timer, and depress the switch to begin fishing. If fishing success is poor, increase the pulse width first and then the voltage. Increase the pulse rate last to minimize mortality or injury to large fish. If mortalities occur, first decrease the pulse rate, then the voltage, then the pulse width. Start cleared clocks. Note: some electrofishers do not meter all the requested header data; provide what you can.

5. Once the settings on the electrofisher are adjusted properly to sample effectively and minimize injury and mortality, record them on the Vertebrate Collection Form. Also, record the water temperature and conductivity.

6. Begin sampling at the downstream end of the support reach (subreach *A*), and fish in an upstream direction. Depress the switch and slowly sweep the electrode from side to side in the water in riffles and pools. Sample available cut-bank and snag habitats as well. Move the anode wand into cover with the current on, then remove the wand quickly to draw fish out. In fast, shallow water, sweep the anode and fish downstream into a net. In extremely wide reaches, work from the midline of the stream channel to the banks. Be sure to sample all habitats (deep, shallow, fast, slow, complex, and simple). In stretches with deep pools, fish the margins of the pool as much as possible, being extremely careful not to step or slide into deep water. Keep the cathode near the anode if fish catch is low.

(Continued)

TABLE 11-1 (Continued)

7. The netter, with the net 1 to 2 ft from the anode, follows the operator, nets stunned individuals, and places them in a bucket.

8. Continue upstream until the next transect is reached. Process fish and/or change water after each subreach to reduce mortality and track sampling effort (i.e., all subreaches where a species was collected).

9. Repeat Steps 6 through 8 until subreach *J* (between transects *J* and *K*) is finished. Record the water visibility, and the number of subreaches sampled (all 10, 5-9, or < 5) on the collection form; note which subreaches were not sampled and why in the comments section of the form. *SAMPLE DISTANCE* is the total reach length actually fished (i.e., it will be equal to or less than the support reach length).

and/or use the procedures for bank/towed electrofishing (Section 11.1.1.2) in conjunction with the backpack electrofishing units.

Determine that all team members are wearing waders and gloves and are clear of both electrodes. Wear polarized sunglasses and visors or caps to aid vision. The back-pack unit is equipped with an audio alarm that sounds when the output voltage exceeds 300 volts (V). It also serves as an input current indicator for pulse cycles greater than 5 Hertz (Hz). It begins as a strong continuous tone and begins to beep slowly at currents of 1.25 amperes (A), beeping faster as input current increases. In case of an overload (greater than 3 A), the beep becomes very rapid and the overload indicator comes on. Release the anode switch, adjust voltage and waveform, and resume fishing.

The anode is fitted with a net and a second netter uses an insulated dip net to retrieve stunned individuals, which are immediately deposited into a bucket for later processing (Section 11.3). If individuals show signs of stress (loss of righting response, gaping, gulping air, excessive mucus), change water or stop fishing and process them. This should only be necessary on very warm days, in long subreaches, or if a large number or biomass of aquatic vertebrates is collected. Stop electrofishing to process and release listed threatened or endangered species or large game fish as they are netted (see Section 11.2). If periodic processing is required, be sure to release individuals downstream to reduce the likelihood of collecting them again.

11.1.1.2 Bank/towed Electrofishing--

Bank/towed electrofishing sampling procedures are presented in Table 11-2. The primary electrofishing gear is either a small (9 ft) inflatable kayak modified to carry all fishing equipment, or an electrofishing unit that is set up on the bank with one or more anodes. The kayak configuration consists of a frame mounted generator and electrofishing control box, port and starboard cathodes, and one or two hand-held anodes fitted with netting. Alternatively, the generator and control box may remain on the riverbank connected to the electrodes by a 100-m long heavily insulated wire. The kayak is maneuvered by one or two persons, and the vertebrates are collected by the others wading in the water. Wear chest waders and linesman gloves to avoid electric shock, and polarized sunglasses and caps to reduce the glare. Use ear protection and hand signals to communicate direction and to have the power turned on or off when using generators. Adjust voltage and output according to sampling effectiveness and minimizing incidental mortality.

TABLE 11-2. BANK/TOWED ELECTROFISHING PROCEDURES

NOTE: On very wide, shallow reaches, it may be necessary to use this procedure in conjunction with backpack electrofisher units if larger units are not available.

1. Select a river bank for initial fishing (left for odd-numbered sites [e.g., WXXP99-0503], right for even) unless immediate hazards or obstructions preclude this. Stay along the selected bank for two subreaches to the degree it is safely wadeable. Switch to the opposite bank for the next two subreaches, alternating in this manner until the entire site is fished or hazards prevent it. Using a rangefinder, determine a downstream point that is 4 mean channel widths distant (this is the subreach length). Record this distance on the Vertebrate Collection Form.

   NOTE: The alternating bank approach is designed to ensure that shaded and unshaded portions of the reach will be sampled. If it appears that alternating banks after every two subreaches will result in sampling the same type of habitat throughout the reach (e.g., inside bends or outside bends), increase the number of subreaches sampled initially on one bank to three, then switch banks after every second subreach.

2. Fill the generator fuel tank with gas, check all electrical connections and potential conductors, and place the anodes and cathodes in the water. Fill live well and put on linesman gloves. Verify that all electrical switches are off, that cathodes are submerged, that all nontarget organisms are clear of the water or 20 ft away, and that barge (or kayak) surfaces are dry.

3. Complete header information on the Vertebrate Collection Form.

4. ***Review all collecting permits*** *to determine if any sampling restrictions are in effect for the reach. In some cases, you may have to cease sampling if you encounter certain listed species, or use alternate gear types.*

   If you cannot sample a reach because of permit restrictions, mark the *NOT FISHED-NO PERMIT* bubble on the Vertebrate Collection Form.

   If no individuals are collected, mark the *FISHED-NONE COLLECTED* bubble on the Vertebrate Collection Form. Explain why in the comments section.

   If conductivity, turbidity, or depth precludes electrofishing, sample by seining if possible, otherwise do not sample. If you do not sample, mark the *NOT FISHED-OTHER* bubble on the Vertebrate Collection Form and explain why in the comments section.

5. Start the generator, switch to *pulsed DC*, a pulse rate of 30 pps, low range (duty cycle) and 40%. Increase % (voltage) as needed to roll fish. If success is poor, reduce %, switch to high range, and again increase % as needed. If effectiveness is still low, switch to 60 pps and repeat the process. If the current (amperage needle) is reduced, switch back to low range to avoid overloading the generator. *Switching should occur when power to the control box is* ***off***. If the conductivity of the river is > 1700 µS/cm, use a larger generator or seine. Activate the thumb switches on each anode and confirm the current ceases when the switch is off. Crew members towing the barge (or kayak) activate the generator and pulsator switches. Verify that fish are rolled and relaxed but not rigid before beginning subreach. Record settings on the Vertebrate Collection Form and clear clocks. Record the conductivity and water temperature on the form as well.

(Continued)

TABLE 11-2 (Continued)

6. Zero the timer, and depress the thumb switch to begin fishing. With the system activated and safety switches on, fish upstream near the shore. Maneuver the anode(s) to cover a swath 3-4 meters wide, near cover, and at depths less than 1 meter wherever possible. Do not place yourself or the gear in danger to fish particular habitats; cut the generator and stow the gear before negotiating hazards.

7. Place fish directly in livewell; do not hold them in the electrical field. Pay special attention to netting small and benthic fishes as well as fishes that respond differently to the current--not just the big fish that move to the surface. Try to net all fish seen, but in productive areas this is impossible. Do not chase individual fish or place yourselves in unbalanced positions to net them. If benthic fish are not being collected, occasionally hold a net in the current and along the bottom, then sweep the anode downstream into the net. Draw fish from cover by thrusting the anode into the cover with the power on, then quickly removing it.

8. Cease sampling at the end of the subreach. Process the fish quickly and carefully, returning them to the water unless they are vouchered or saved for tissue.

9. Return to step 1 for each of the subsequent nine subreaches, but begin upstream from where fish were released and alternate banks on every other subreach. Record the water visibility, and the number of subreaches sampled (all 10, 5-9, or < 5) on the collection form; note which subreaches were not sampled and why in the comments section of the form. *SAMPLE DISTANCE* is the total reach length actually fished (i.e., it will be equal to or less than the support reach length).

Start at the downriver end of the support reach and along the designated shoreline, and fish upriver. The netters use a dip net and an insulated anode with a net ring to retrieve stunned individuals and deposit them into a live well in the kayak for later processing (Section 11.2). *Note, to avoid shocking crew members, insure that the thumb switch is* ***off*** *anytime the anode leaves the water.* Change the water in the live well at each transect to minimize mortality. If individuals show signs of stress (loss of righting response, gaping, gulping air, excessive mucus), stop fishing and process them. This should only be necessary on very warm days, in long subreaches, or if a very large number or biomass of aquatic vertebrates is collected. Cease electrofishing immediately to process and release specimens of listed species or large game fish as they are netted (Section 11.2). After processing at each transect, release individuals downriver and away from the shoreline to reduce the likelihood of collecting them again. At the completion of electrofishing each subreach, record information on the Vertebrate Collection Form (Figure 11-1).

Gasoline is extremely volatile and flammable. Its vapors readily ignite on contact with heat, spark or flame. Never attempt to refill the generator while it is running or near the water. Always allow the generator to cool before refilling. Keep gasoline out of direct sunlight to reduce volatilization and vapor release. Keep gasoline only in approved, tightly closed plastic containers.

11.1.2 Seining

Seining is used when the conductivity of the stream is too high and/or in streams with extremely high turbidity where electrofishing is ineffective. Avoid high mortality rates by employing multiple short seine hauls at each of the 10 subreaches by using either the *riffle habitat* or *pool habitat* method (Table 11-3). Allocate the total sampling time (up to 180 min) among the 10 subreaches (i.e., from 2 to 18 min per subreach). If no aquatic vertebrates are collected, indicate this on the form (Figure 11-1). Record the seine length, mesh size, the time spent seining (*SAMPLING TIME*) and the **combined length** of all seine hauls (*SAMPLING DISTANCE*) on the Vertebrate Collection Form (Figure 11-1). If more than two sizes or types of seines are required, record the information for the primary and secondary seines used on the collection form, and note any additional types used in the comments section of the form.

TABLE 11-3. SEINING PROCEDURES

1. Allocate the sampling effort throughout the reach so that the total fishing time will be between 45 min (small stream) and 3 hours (large stream). If you cannot effectively sample at least 50% of the available area in the 180-min time limit, spend two days to sample the reach.

2. Complete header information on the Vertebrate Collection Form. Record the conductivity and water temperature on the from as well.

3. ***Review all collecting permits*** *to determine if any sampling restrictions are in effect for the site. In some cases, you may have to cease sampling if you encounter certain listed species.*

   If you cannot sample a reach because of permit restrictions, mark the *NOT FISHED-NO PERMIT* bubble on the Vertebrate Collection Form.

   Search for aquatic vertebrates and crayfish even if the reach is extremely small, and it appears that sampling may produce no specimens. If no individuals are collected, mark the *FISHED-NONE COLLECTED* bubble on the Vertebrate Collection Form. Explain why in comments section.

   If you cannot sample by any method (electrofishing or seining), mark the *NOT FISHED-OTHER* bubble on the Vertebrate Collection Form and comment why.

4. Begin at the downstream end of the sampling reach (subreach *A*). Proceed along the reach, sampling available habitats using the appropriate methods below:

   4A. *Riffle habitats*: Use a seine 2 m long × 1.25 m high with 0.6 cm mesh.

   1. Place the seine perpendicular to the current across the downstream end of the riffle. Ensure that the lead line is on the bottom. Tilt the net slightly downstream to form a pocket to trap aquatic vertebrates.
   2. Starting about 2 m upstream, kick the substrate and overturn rocks, proceeding downstream toward the net.
   3. Raise the net quickly and examine it carefully for aquatic vertebrates (and crayfish).
   4. The length of a seine haul in this case is the distance upstream from the net where you started to kick. Keep a tally of the lengths of the individual hauls you conduct.

   4B. *Pool habitats*: Use a seine 3-9 m long × 2 m high with 0.6 cm mesh size.

   1. Two people pull the seine across the pool, using the shore or riffles as barriers.
   2. In areas with current, pull the net downstream and then sweep toward the shore with one or both poles, or keep one end of the seine near shore and sweep the other end in a wide arc from midstream to the same shore.
   3. Pull the net onto the shore and examine it carefully for aquatic vertebrates (and crayfish).
   4. The length of a seine haul in this case is the distance the net moved. Keep a tally of the lengths of the individual hauls you conduct.

(Continued)

TABLE 11-3 (Continued)

4C. *Snags and undercut banks*: Use a seine 2 m long × 1.25 m high; 0.6 cm mesh size.

1. Jab the seine under the cover and near the stream bottom, then quickly lift it above the stream surface.

5. Place individuals in buckets for processing, and continue upstream to the next habitat area.

6. Complete the header information on the Vertebrate Collection Form.

7. Repeat Steps 4 through 6 for successive habitat areas until subreach *J* (between transects *J* and *K* is finished. Record the water visibility, and the number of subreaches sampled (all 10, 5-9, or < 5) on the collection form; note which subreaches were not sampled and why in the comments section of the form. *SAMPLE DISTANCE* is the total length of all seine hauls conducted within the entire support reach (i.e., it will usually be less than the support reach length).

11.2 SAMPLE PROCESSING

Sample processing involves tallying and identifying fish, crayfish and amphibians, examining individual specimens for the presence of external anomalies (which are not classified further), obtaining length measurements from selected specimens, preparing voucher specimens for taxonomic confirmation and archival at a museum, and selecting specimens to prepare samples for fish tissue contaminants (Section 12). Process collections as quickly as possible at each transect to reduce stress to live specimens. One person can process fish from one bucket while the other team members continue to collect fish and deposit them into a second bucket. One person can identify, measure, and examine individuals while another person records information on the field data forms.

11.2.1 Taxonomic Identification and Tally

Table 11-4 presents the procedure for identifying and tallying aquatic vertebrates. Record identification, tally data, and comments for each species on the Vertebrate Collection Form (Figure 11-1). It is important to note all subreaches where a species is collected, as this is information is needed to develop estimates of sampling efficiency. Use common names from Nelson et al. (2004). Taxonomic identification should be performed only by trained ichthyologists familiar with the fish species and other aquatic vertebrate taxa of the region. Use taxonomic reference books and other materials that contain species descriptions, ranges, and identification keys to make species identifications in the field. Where there are many individuals of easily identified species, processing is facilitated by keeping a tally count of the number of individuals of each species and totaling the tally once processing is complete. If protected fish have died, voucher them. Notify the appropriate state officials as soon as possible.

After the entire reach has been sampled, estimate the sampling adequacy based on Cao et al. (2001). Use the presence/absence of vertebrate species (***do not*** *include crayfish*) between the "odd" and "even" groups of subreaches (i.e., the top and bottom rows of subreaches on the form) to calculate a Jaccard coefficient of similarity. An example calculation is given below and in Table 11-4. Treat unknowns suspected of being different taxa (e.g., *unknown sucker 1* and *unknown sucker 2* in Figure 11-1) as separate taxa. In cases where a species has more than one tag number assigned (e.g., mottled sculpin in Figure 11-1), count it only once in the total species list and use the total number of different

TABLE 11-4. PROCEDURE TO IDENTIFY, TALLY, AND EXAMINE AQUATIC VERTEBRATES

1. Complete all header information accurately and completely on the Vertebrate Collection Form. If no vertebrates or crayfish were collected, fill in the *FISHED-NONE COLLECTED* bubble on the form, and explain why in the comments.

2. Identify and process each individual completely, ideally handling it only once. When first collected, record the common name (***print** using capital letters*) on the first blank line of the Vertebrate Collection Form. If a species cannot be positively identified, assign it as *UNKNOWN* followed by its common family name (e.g., *UNKNOWN SCULPIN A*). Note every subreach where a species is collected (letters represent the downstream transect), by filling in the appropriate bubble in the *SUBREACHES* section of the form.

3. Process species listed as threatened and endangered first and release individuals immediately into the stream. Photograph specimens for voucher purposes if conditions permit and stress to individuals will be minimal. Indicate if photographed on Vertebrate Collection Form. If individuals have died, prepare them as voucher specimens and preserve in formalin.

   Remember that the photographs you take must provide sufficient detail to allow for someone else to confirm your identification. Fill the frame with each specimen and ensure that key characters (e.g., mouth, fin rays, etc.), are visible. Use more than one photo if necessary (e.g., one lateral view and one of the mouth if this is a key character).

4. Keep voucher specimens (up to 20) of smaller individuals of each species. If no smaller individuals are collected, photograph each species and indicate so on the data form. Large, questionable species may be placed on ice and then frozen.

   We retain 20 voucher specimens to allow us to adjust count data when one apparent species turns out to be more than one. For example, if you voucher 20 individuals of *Species A*, and five of those turn out to be *Species B*, we can adjust the total number of individuals of *Species A* collected by assigning 25% of them to *Species B* and the remaining 75% to *Species A*.

5. Tally the number of individuals of each species collected in the *TALLY* box on the Vertebrate Collection Form and record the total number in the *COUNT* field on the form.

6. For fish, measure the ***total length*** (from the nose to the distal end of caudal fin) of the largest and smallest individual to provide a size range for the species. For amphibians, measure body length (tip of the snout to the vent). Do not measure lengths for crayfish. Record these values in the *LENGTH* area of the Vertebrate Collection Form.

(Continued)

TABLE 11-4 (Continued)

7. Examine each individual for external anomalies and tally those observed. Readily identified external anomalies include missing organs (eye, fin), skeletal deformities, shortened operculum, eroded fins, irregular fin rays or scales, tumors, lesions, ulcerous sores, blisters, cysts, blackening, white spots, bleeding or reddening, excessive mucus, and fungus. After all of the individuals of a species have been processed, record the total number of individuals affected in the *ANOMALIES* area of the Vertebrate Collection Form. Photograph specimens with especially extreme anomalies.

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

9. Follow the appropriate procedure to prepare voucher specimens and/or to select specimens for tissue samples. Release all remaining individuals downstream of your location to avoid recapturing them.

10. For any line with a vertebrate name, ensure that all spaces on that line are filled in with a number, even if it is zero.

11. Repeat Steps 1 through 10 for all other vertebrate species.

12. After processing the vertebrates from all 10 subreaches (do not include crayfish), calculate a Jaccard Coefficient (*JC*) to assess sample adequacy. To help randomize the calculation, subreaches *A*, *C*, *E*, *G*, and *I* are in the "odd" group (the top row of sites on the form) and subreaches *B*, *D*, *F*, *H*, and *J* are in the "even" group (the bottom row of sites on the form). Calculate *JC* as:

$$JC = \frac{S}{T}$$

where ***S*** is the number of species shared by both groups of subreaches (the number of species having marks in both rows on the form), and ***T*** is the total number of species collected from the entire reach. Treat unknown taxa suspected of being different (e.g., *UNKNOWN SUCKER 1*, *UNKNOWN SUCKER 2*) as different taxa. For species where more than one tag number is assigned, count them only once in computing of ***T*** and use the total number of subreaches recorded across all tag numbers in computing ***S***. Record the value of *JC* in the comments section; if $JC < 0.7$, sample two additional subreaches (one "odd", one "even"). List the species in the appropriate group of subreaches ("odd" vs. "even") and recalculate *JC*. Continue until $JC \geq 0.7$ or there is insufficient time or space to sample.

EXAMPLE: You have collected a total of seven species. Six different species were collected from the "odd" group of subreaches (top row on form), and four different species from the "even" group of subreaches (bottom row on form). Of these, three species were collected from both groups of subreaches (both rows on form).

$JC = 3/7$

$JC \approx 0.4$ In this case, sample two additional subreaches and recalculate.

subreaches across all tag numbers in calculating the Jaccard coefficient. Calculate the Jaccard coefficient (*JC*) as:

$$JC = \frac{S}{T}$$

where *S* is the number of species having marks in both rows on the form, and *T* is the total number of species collected from the entire reach. If the calculated Jaccard value is < 0.7 and there is sufficient time available, sample an additional two subreaches (eight channel widths) upstream of the reach. Continue with additional pairs of subreaches until the calculated Jaccard value is ≥ 0.7, or until there is insufficient time or space to sample (e.g., the reach moves through a confluence and changes to a lower stream order, or an impoundment or other barrier is encountered. For the data presented in Figure 11-1, a total of nine vertebrate species were collected (note mottled sculpin is recorded twice). Six species have marks in both rows of the form, i.e., they were collected from both groups of subreaches. Calculate the Jaccard coefficient as:

$$JC = \frac{6}{9} = 0.67 \approx 0.7$$

For this site, the sampling effort is adequate, and no additional subreaches would be sampled.

11.2.2 External Examination and Length Measurements

During the tallying procedure for each species (Table 11-4), examine each individual for the presence of external anomalies. Smith et al. (2002) present examples of different types of external anomalies. Record the number of individuals affected on the Vertebrate Collection Form (Figure 11-1). Blackening and exopthalmia (popeye) may occasionally result from electrofishing. Do not include injuries due to sampling in the tally of external anomalies, but note these in the comments section of the Vertebrate Collection Form (Figure 11-1), as this information may be required for collection permit reports. Blackening from electrofishing usually follows the myomeres or looks like a bruise. If fish die due to the effects of sampling or processing, record the number of mortalities for each

species on the Vertebrate Collection Form (Figure 11-1). If not too large, prepare all dead individuals as voucher specimens (Section 11.2.3).

For each species, use a measuring board or ruler to determine the length of the largest and smallest individuals from all the individuals of that species collected at a site. Measure *total length* for fish (from the nose to the distal end of the caudal fin) and *body length* for amphibians (from the tip of the snout to the vent) on the Vertebrate Collection Form (Figure 11-1). No length measurements are taken for crayfish.

11.2.3 Preparing Voucher Specimens

Except for very large individuals or easily identified species, up to 20 individuals of each species are vouchered from each stream site to provide a permanent, archived, historical record of fish collections. Retaining 20 voucher specimens also enables us to correct tally data and more rigorously infer relative abundances when one apparent species in the field is found to be two or more species upon closer examination at the museum. *You should retain* ***at least one voucher specimen of all species*** *collected at a site.* This provides evidence for independent confirmation of taxonomic identifications when there is a possibility of a range extension. Prepare the voucher sample for a site according to the procedure presented in Table 11-5. Retain additional specimens of the appropriate species for the fish tissue contaminants samples (Section 13). *For each species, voucher specimens take priority over specimens for the tissue contaminant samples.*

The number of voucher specimens and the method of vouchering varies with species. Large, easily identified species, larger species that are difficult to identify in the field, or species that are uncommon in the region require a few specimens of both adults and juveniles, if both were collected. Very large specimens, especially of easily identified game fish, are vouchered by photographing them and then releasing them alive. More voucher specimens are required for smaller species, which are typically more difficult to identify in the field. Any *listed species* of special concern (state and federally protected species), are processed first, vouchered by photographs, and released alive. Include any individuals of listed species that die before they can be released as part of the preserved voucher sample for the stream.

TABLE 11-5. GUIDELINES AND PROCEDURES FOR PREPARING AQUATIC VERTEBRATE VOUCHER SPECIMENS

1. *Obtain **at least one** voucher specimen (preserved or photograph) of **every species** collected at a site.* Determine the voucher class of a species and the number of specimens to include in the voucher sample based on the following guidelines. *Process **Class 1** species first.* Voucher samples take priority over tissue contaminant samples.

   Class 1--State or federally listed species. Photograph and **release immediately**. Photographs should include (1) a card with the stream ID and common name (2) an object of known length (e.g., a ruler) with the specimen. If specimens have died, proceed to Step 2 and include them in the voucher sample. Assign an *Fn* flag for the species on the Vertebrate Collection Form and note it is a listed species in the comments section of the form. Notify the appropriate state officials as soon as possible.

   Class 2--Large easily identified species **OR** adults that are difficult to identify **OR** species that are uncommon in that region (e.g., sunfish, suckers, bullheads, trout, crayfish). Preserve 1-2 small (<150 mm total length) adult individuals per site plus 2-5 juveniles. If only large adults are collected, reserve smallest individuals until voucher procedure is complete and preserve **only** if space is available. Individuals with a total length > 160 mm should be slit on the lower abdomen of the **right** side before placing them into the container. Photograph if considered too large for the jar or place in a bag on ice for freezing (***Do not** voucher large gamefish*). Retain additional individuals for tissue contaminant samples.

   Class 3--Small to moderate-sized fish **OR** difficult to identify species (e.g., lampreys, juvenile salmonids, minnows, sculpins). Preserve up to 20 adults and juveniles (several per transect). If fewer than 20 individuals are collected, voucher them all. Retain additional individuals for the tissue contaminant sample.

2. Euthanize voucher specimens in a bucket with two carbon dioxide tablets and a small volume of water or by cranial concussion[a], then transfer them to a nylon mesh bag. Tally, then record the number of individuals included in the bag in the *VOUCHERED COUNT* field for the species on the Vertebrate Collection Form.

   NOTE: Do not pack specimens too tightly into a single mesh bag, as they will not be fixed and preserved properly. If necessary, use additional bags, each with a separate tag number. Record each tag separately on the collection form, even though they all represent the same species. Record tally values separately for each bag.

3. Select a *FISH-BAG* tag with the same ID number as the voucher sample jar (Step 6). Record the tag number in the *TAG NO.* field on the corresponding line for the species on the Vertebrate Collection Form. Place the tag into the mesh bag and close. *This bagging, tagging, and recording is crucial, as it enables us to estimate species proportionate abundances in the assemblage even when one suspected species turns out to be multiple species.*

4. Immediately place the bag into a container large enough to hold all voucher specimens loosely and half-filled with 10% formalin. *Use additional jars if necessary to avoid close packing and bending of voucher specimens.*

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

[a] Acceptable or conditionally acceptable methods of euthanization (AVMA Panel on Euthanasia 2001). **(Continued)**

TABLE 11-5 (Continued)

6. Prepare two *FISH-JAR* labels (each having the same sample ID number) by filling in the stream ID and the date of collection. Place one label into the sample jar. Cap tightly and seal with plastic electrical tape. Attach the second label to the outside of the sample container by covering it with a strip of clear tape. Record the voucher sample ID number on page 1 of the Vertebrate Collection Form. Record general comments (perceived fishing efficiency, missed fish, gear operation, suggestions) in blank lines of the form, and make sure all header information on the form is completed. NOTE: If more than one jar is required, use labels that have the same ID number printed on them and flag.

7. Place the preserved sample in a suitable container with absorbent material. Store the container in a well-ventilated area during transport. Follow all rules and regulations concerning the transport and shipment of samples containing 10% formalin.

[a] Acceptable or conditionally acceptable methods of euthanization (AVMA Panel on Euthanasia 2001).

Figure 11-2. Completed voucher sample label and specimen bag tag for aquatic vertebrates. Note that two voucher sample labels (left) are filled out– one is placed inside the jar, the second is taped to the outside.

For taking photographs, use a film or digital camera with sufficient macro capability to take clear, close-up photographs of small fish or other aquatic vertebrates. The photographs must be of sufficient magnification, clarity, and resolution so that important external identifying characteristics (e.g., mouths, fin positions and rays, coloration, scale patterns, etc.) can be distinguished. One purpose of a voucher photograph is to allow someone else to confirm your field identification without an actual specimen to examine. For each photograph, include a card with the site ID and the common name printed on it, and a measuring board, ruler, or other object to provide a length reference. If necessary, take more than one photo of a particular specimen (e.g., one lateral view, one close-up of the mouth). Provide a log (either an electronic file or hard copy) describing each photo or digital image, including date taken, file name (for images), site ID, tag number recorded on collection form, common name, and description or other notes associated with the photo or image when you submit the photos for archiving.

Voucher specimens for each species are euthanized by carbon dioxide or cranial concussion (American Veterinary Medical Association Panel on Euthanasia 2001), counted, and placed in nylon mesh bags or plastic jars (one or more bags per species). Each bag contains a numbered tag (Figure 11-2). Single specimens of easily identified and distinct species (e.g., sandroller, smallmouth bass) and crayfish may be placed directly in the jar with the tag. Record the tag number and the number of individuals vouchered for each species on the Vertebrate Collection Form (Figure 11-1). Preserve vouchers of sculpins, minnows, lampreys and other difficult species from throughout the support reach. *Note you can have more than one bag of specimens that you think are the same species.* This bagging, tagging, and recording procedure is crucial, as it enables us to estimate species proportionate abundances in the assemblage even when one apparent species turns out to be multiple species.

Place specimen bags into a large sample jar containing 10% buffered formalin (Section 3). The final volume of 10% formalin in the sample container should equal or exceed the total volume of specimens. Use additional containers if necessary and avoid tight packing of specimen bags or bending of specimens. *Delays in carrying out the preservation procedure, overpacking a bag or sample container, or an inadequate volume of preservative will result in unidentifiable (worthless) specimens.* Formalin vapors and solution are extremely caustic and may cause severe irritation on contact with skin, eyes or mucus membranes. Formaldehyde is a potential carcinogen, and contact with it should be avoided. Wear gloves and safety glasses and always work in a well-ventilated area. In case of contact with skin or eyes, rinse immediately with large quantities of water. Store stock solution in sealed containers in a safety cabinet or cooler lined with vermiculite or other absorbent material. If possible, transport outside the passenger compartment of a vehicle. Complete a set of two sample labels for each sample container as shown in Figure 11-2. Place one label inside each sample container, and attach the second label to the outside of the jar with clear tape. Record the sample ID number on the Vertebrate Collection Form (Figure 11-1).

11.3 EQUIPMENT AND SUPPLIES

Figure 11-3 is a checklist of equipment and supplies required to conduct protocols described in this section. This checklist may differ from the checklists presented in Appendix A, which are used at a base site to ensure that all equipment and supplies are brought to the stream site. Use the checklist presented in this section to ensure that equipment and supplies are organized and available to conduct the protocols efficiently.

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EQUIPMENT AND SUPPLIES FOR AQUATIC VERTEBRATE SAMPLING

QTY.	Item	
1	Gasoline or battery-powered backpack electrofishing unit with netted anode (electrode wand)	
	Extra battery (charged) or gasoline	
4 pr	Heavy-duty rubber gloves	
3 pr	Chest waders with non-slip soles & patch kit	
3 pr	Polarized sunglasses	
2	Long-handled dip nets (0.6 cm mesh) with insulated handles	
1	Watch or stopwatch to track elapsed fishing time	
4	Collapsible buckets for holding and processing aquatic vertebrates	
1	Minnow seine (2m X 1.25 m, 0.6 cm mesh) with poles	
1	Large seine (9 m X 2 m, 0.6 cm mesh) with poles	
1	Aquarium net	
1 set	Taxonomic reference books and keys for fishes and amphibians of the region	
1	Camera and film (or digital camera) with macro capability for photographing vouchers	
1-2	Fish measuring board & small plastic rulers (2)	
5-20	Small nylon mesh bags or stockings for holding voucher specimens	
1	Jackknife for preparing larger voucher specimens for preservation	
1 ea.	1, 2, and/or 4-L screw-top plastic jars (leakproof) for voucher samples	
2 L	10% (buffered) formalin or voucher sample jar half full of 10% formalin	
1	Container with absorbent material to hold formalin solution and preserved voucher sample jars	
1 pr	Safety glasses	
1 pr	Chemical-resistant gloves	
1	Covered clipboard	
1 box	Carbon dioxide tablets	
1	Sheet of pre-printed jar labels (4) and voucher bag tags (36), all with same preprinted sample ID number (barcode)	
1 pr	Scissors for cutting jar labels and tags	
1 roll	Plastic electrical tape	
1 pkg.	Clear tape strips	
2	Soft lead pencils for recording data and completing tags and labels	
2	Vertebrate Collection Forms	
3	Plastic safety whistles & ear protection if generators are used	
1	Field operations manual	
1 set	Laminated sheets of aquatic vertebrate procedure tables	
1 ea.	Vertebrate collection permits (State, Federal, Tribal)	

Figure 11-3. Equipment and supplies checklist for aquatic vertebrates.

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NOTES

NOTES

SECTION 12
FISH TISSUE CONTAMINANTS

Robert M. Hughes[1], Spencer A. Peterson[2], Frank H. McCormick[3],
and James M. Lazorchak[4]

In addition to gathering data on the aquatic vertebrate assemblage (Section 11), fish are retained for analysis of selected tissue contaminants (principally mercury). The fish tissue contaminants indicator is used to evaluate the potential burden of toxic chemicals at a site (e.g., Yeardley et al. 1998a, Peterson et al. 2002, 2005, Lazorchak et al. 2003). The focus is on fish species that commonly occur throughout the region of interest that are sufficiently abundant within a support reach.

The response design (see Section 1.3.7) for this indicator is based in part on that used for the aquatic vertebrates indicator (Section 11). Fish are collected from throughout the sampling reach. To maximize the potential of obtaining a sample from every site, two types of fish tissue contaminant samples are prepared for each site (if possible). *Small fish* tissue contaminant samples are composite samples of individuals <100 mm long. *Big fish* tissue contaminant samples are individuals that are >120 mm long. Whole fish are collected and analyzed because they present fewer logistical problems and integrate all fish parts (Yeardley et al. 1998a). Peterson et al. (2005) suggest that non-lethal biopsy samples collected from individual fish could be used to predict filet and whole fish concentrations of mercury, but did not determine it this were true for other contaminants.

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

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

3 USDA Forest Service, Olympia Forestry Sciences Laboratory, Pacific Northwest Research Station, 3625 93rd Avenue SE, Olympia, WA 98512.

4 U.S. EPA, National Exposure Research Laboratory, Ecological Exposure Research Division, 26 Marrtin Luther King Dr., Cincinnati, OH 45268.

Procedures for obtaining fish tissue contaminant samples are adapted from those previously published for EMAP-SW (Yeardley et al. 1998b). Minor modifications made during EMAP-W are summarized in Appendix B. These included changing the species list (no target species priority list for small species and adjusting the larger species list by replacing eastern species with their western counterparts) and clarifying labeling and shipping instructions.

12.1 PREPARING TISSUE CONTAMINANT SAMPLES

Prepare tissue contaminant samples as described in Table 12-1. To determine the proper quantity for each sample, use weight for the *small fish* composite sample and total length for each *big fish* sample. Use similar-sized individuals if possible (size difference between smallest and largest < 25%) to prepare the *small fish* composite sample. If a sufficient number of similar-sized individuals are not available, send as many fish of a single species as possible up to 400 g. Getting a sufficient sample in terms of weight is a higher priority than getting similar-sized individuals. If there is no single species with enough individuals available, prepare a composite sample using individuals of multiple species. For the *big fish* sample, send as many fish as possible, up to three fish for each of three species. For each species, *big fish* samples should represent a wide range in size, yet be as large as possible. Note that *voucher specimens have higher priority than tissue contaminant samples.*

Record information for the fish tissue contaminant samples on page 2 of the Vertebrate Collection Form (Figure 12-1). Examples of completed sample labels are presented in Figure 12-2. Use a permanent marker to complete the labels. Each individual comprising the *big fish* sample is wrapped, labeled, and bagged separately, while the individuals comprising the *small fish* composite sample are wrapped together. Thus, up to 10 different sample labels may be required (nine *big fish* and one *small fish*). Each sample is double bagged. Store tissue samples in a cooler with several bags of ice (or ice substitute packs). Place ice inside two bags and tape the outside bag shut to prevent melting ice from contaminating the samples. Store tissue contaminant samples on ice (freeze them if possible) until they can be shipped (Section 3). Tissue contaminant samples can be stored and shipped with other samples requiring ice or freezing (water chemistry and periphyton samples). *NOTE: It is critical that tissue contaminant samples are shipped with sufficient ice or ice substitute packs to ensure they arrive in good condition at the laboratory. Shipping containers with fish tissue contaminant samples should include*

TABLE 12-1. PROCEDURE TO PREPARE FISH TISSUE CONTAMINANT SAMPLES

NOTE: Use your best judgement to collect some type of fish tissue sample from every site.

***SMALL FISH* sample**: After voucher specimens have been prepared, choose a small fish species that has enough similarly sized individuals (ideally the difference between the smallest and largest individuals is < 25%) to equal 400 g (14 oz).

***BIG FISH* samples**. After considering voucher specimens, select three individuals >120 mm total length with a wide size range for each of three species. Preference order for Pacific coast and Rocky Mountain drainages: bass, pikeminnow, trout, catfish, sucker; preference order for Mississippi R. drainage: bass, walleye (or sauger), pike, trout, catfish, sucker. Preference is based on top carnivores vs. bottom feeders.

1. Euthanize fish with two carbon dioxide tablets and a small volume of water or by cranial concussion. Keep hands, foil, & bags clean and free of potential contaminants (mud, fuel, formalin, sunscreen, insect repellant, soap, etc.)

2. Record the standard common name of the species (in CAPITAL LETTERS) on the Vertebrate Collection Form. If more than one species is required to make up the *small fish* sample, record the common name of the most abundant species in the *COMMON NAME* column, and record the other species name in the *COMMENT* section.

3. For the *small fish* composite sample, record the total number of individuals in the *NUMBER OF SMALL* field. If the small sample consists of more than one species, record the number of individuals for each species in the *COMMENT* section.

4. For each *big fish* sample, record the total length of each individual in the *TOTAL LENGTH* column of the Vertebrate Collection Form.

5. Indicate the sample type by filling in the appropriate bubble in the *SAMPLE TYPE* column on form.

6. For the *small fish* composite sample, wrap all fish together in a single piece of aluminum foil, *with the **dull side** of the foil in contact with fish*. Place the sample in a resealable plastic bag.

7. Wrap each *big fish* sample **separately** in aluminum foil, with the **dull side** of the foil in contact with the fish. Place each individual in a single resealable plastic bag.

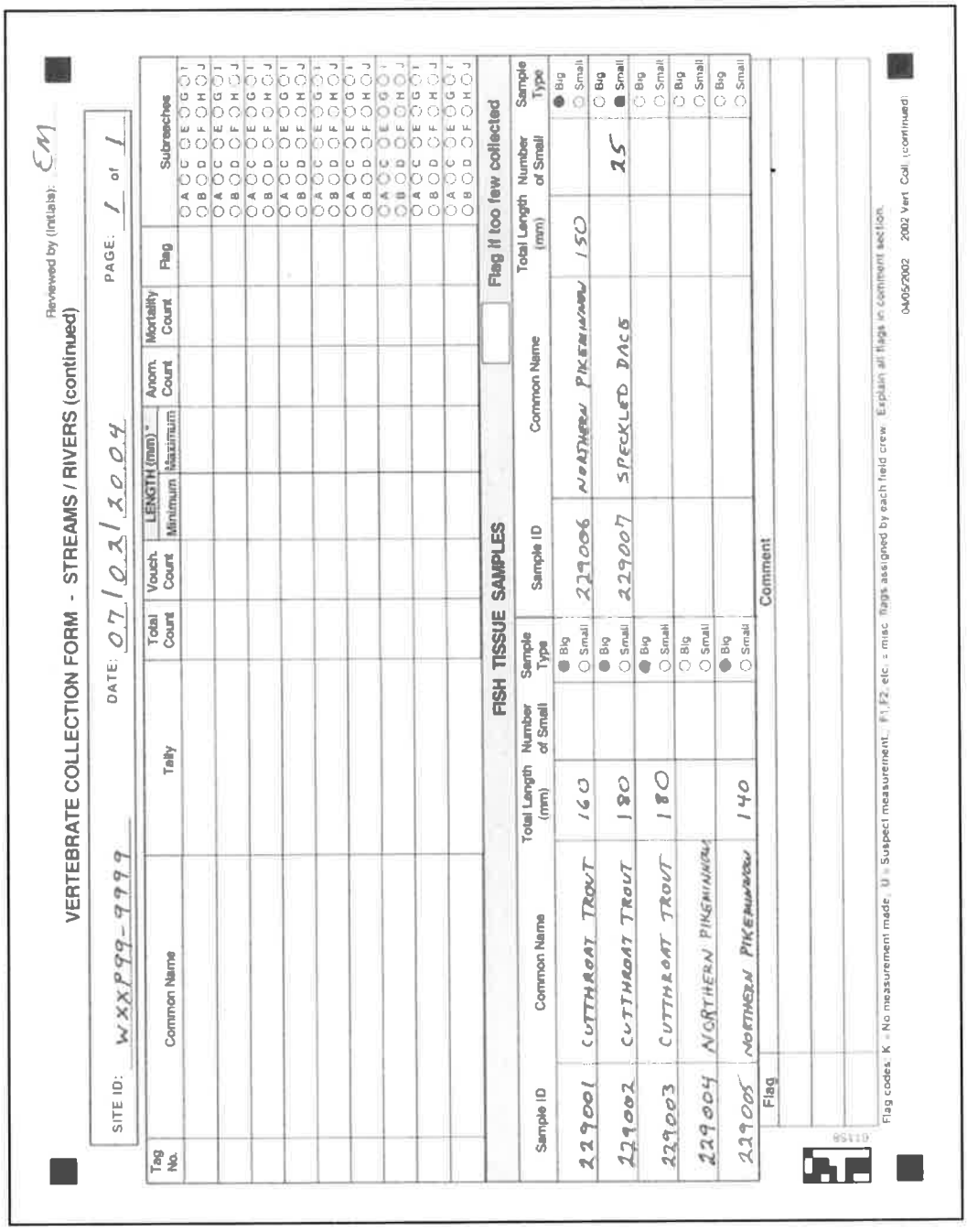
8. Expel excess air and seal each bag.

9. Prepare a sample label for each bag by filling in the site ID, the sample type (big, small) and the collection date on each label. Record the sample ID for each bag on the Vertebrate Collection Form.

10. Attach the appropriate label to each bag. Cover the label with a strip of clear tape. Place the labeled bag into a second resealable plastic bag, and label and tape the second bag.

11. Keep the double bagged samples **on ice** (or frozen if possible) until shipment.

[a] Acceptable or conditionally acceptable methods of euthanization (AVMA Panel on Euthanasia 2001).



VERTEBRATE COLLECTION FORM - STREAMS / RIVERS (continued)

SITE ID: WXP99-9999 DATE: 07/23/2004 Reviewed by (Initials): EM PAGE: 1 of 1

Tag No.	Common Name	Tally	Total Count	Vouch. Count	LENGTH (mm)* Minimum	LENGTH (mm)* Maximum	Anom. Count	Mortality Count	Flag	Subreaches
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J
										○A ○C ○E ○G ○I ○B ○D ○F ○H ○J

FISH TISSUE SAMPLES — Flag if too few collected

Sample ID	Common Name	Total Length (mm)	Number of Small	Sample Type	Sample ID	Common Name	Total Length (mm)	Number of Small	Sample Type
229001	CUTTHROAT TROUT	160		● Big ○ Small	229006	NORTHERN PIKEMINNOW	150		● Big ○ Small
229002	CUTTHROAT TROUT	180		● Big ○ Small	229007	SPECKLED DACE		25	○ Big ● Small
229003	CUTTHROAT TROUT	180		● Big ○ Small					○ Big ○ Small
229004	NORTHERN PIKEMINNOW			○ Big ○ Small					○ Big ○ Small
229005	NORTHERN PIKEMINNOW	140		● Big ○ Small					○ Big ○ Small

Flag

Comment

Flag codes: K = No measurement made, U = Suspect measurement, #1, #2, etc. = misc. flags assigned by each field crew. Explain all flags in comment section.

04/05/2002 2002 Vert Coll (continued)

Figure 12-1. Vertebrate Collection Form, showing information recorded for fish tissue contaminant samples.

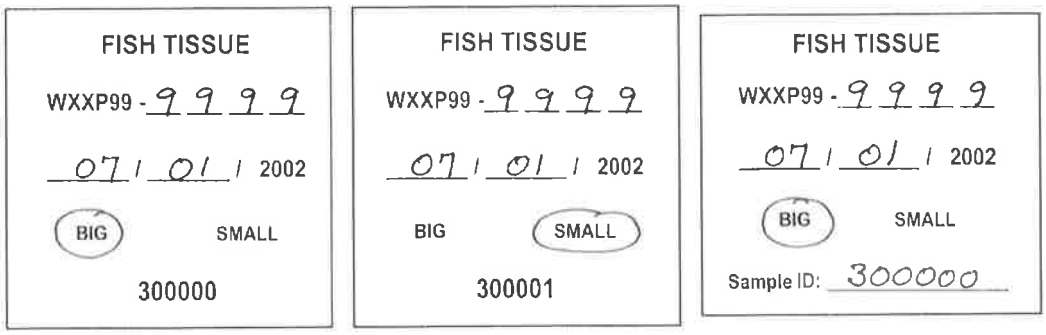


Figure 12-2. Completed labels for fish tissue contaminant samples. Note that a different label (i.e., ID number) is prepared for each individual used for *big fish* samples, and up to 10 different ID numbers could be used at a site. The label on the right is used on the outside of the sample container (or bag).

enough ice to have a total weight (samples plus ice) of ***at least 40-50 lbs****.* The volume of ice should equal or exceed the sample volume.

12.2 EQUIPMENT AND SUPPLIES

Figure 12-3 is a checklist of equipment and supplies required to conduct protocols described in this section. This checklist may differ from the checklists presented in Appendix A, which are used at a base site to ensure that all equipment and supplies are brought to the stream site. Use the checklist presented in this section to ensure that equipment and supplies are organized and available to conduct the protocols efficiently.

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EQUIPMENT AND SUPPLIES FOR FISH TISSUE CONTAMINANTS

QTY.	ITEM	
1	Bucket for anesthetization	
4	Carbon dioxide tablets (Alka-Seltzer® or equivalent)	
1 roll	Aluminum foil (heavy duty) (or 10 16 18" x 11" rectangles) for wrapping fish)	
32	½ - 2 -gallon resealable plastic bags, or heavy duty garbage bags	
2	Soft (#2) lead pencils or eversharps to record data	
2	Fine-point indelible markers to fill out labels	
1 pkg.	Clear tape strips	
10 pr.	Fish tissue labels (each pair with different ID numbers)	
2	Vertebrate Collection forms	
1 set	Laminated procedure tables for fish tissue contaminants	
1	Cooler with ice (double-bagged and taped)	

Figure 12-3. Equipment and supplies checklist for fish tissue contaminants.

Peterson, S.A., J. VanSickle, R.M. Hughes, J. Schacher, and S. Echols. 2005. A biopsy procedure for determining filet and predicting whole fish mercury concentration. *Archives of Environmental Contamination and Toxicology* 48:99-107.

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SECTION 13
RAPID HABITAT AND GENERAL VISUAL STREAM ASSESSMENTS

by
Alan T. Herlihy[1] and James M. Lazorchak[2]

After all other samples and field data have been collected, the field team conducts a visual-based *rapid habitat assessment* of the support reach, makes a *general visual assessment* of the stream and adjacent area, and makes a final check of the data forms and samples before leaving the stream site (see Section 15). The rapid habitat assessment procedures used are based on those developed as part of the EPA Rapid Bioassessment Protocol (RBP; Barbour et al. 1999). This habitat assessment is similar to, but not redundant with, the overall channel and habitat assessment associated with the targeted habitat periphyton sample (Section 9.4). The rapid habitat assessment focuses on integrating information from specific parameters on the structure of the physical habitat.

The general visual assessment of catchment and stream characteristics is based on the collective observations and impressions of field personnel while at a sampling site. This assessment is very valuable for data review and validation, future data interpretation, and selection of reference sites. Changes in the rapid habitat and general visual assessment procedures from those previously published for EMAP-SW (Lazorchak et al. 1998), and modifications made during EMAP-W, are summarized in Appendix B.

13.1 RAPID HABITAT ASSESSMENT

The rapid habitat assessment is separated into two basic approaches—one designed for high-gradient streams and one designed for low-gradient streams. Based on your visual impression of the dominant habitat type from collecting samples and measurements from throughout the sampling reach, classify the stream as either *Riffle/run preva-*

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

lent or *Glide/pool prevalent*. Choose the prevalent habitat type based on which habitat type occupies most of the length of the support reach. Landscapes of moderate to high-gradient typically contain riffle/run prevalent streams. Under natural conditions, riffle/run prevalent streams contain primarily coarse substrates (i.e., coarse gravel or larger; refer to Section 7) or many areas dominated by coarse substrates along a stream reach (Barbour et al. 1999). Landscapes of low to moderate gradient are characterized by glide/pool prevalent streams. These streambeds are dominated by finer substrates (fine gravel or smaller) or occasional areas of coarser sediments along a stream reach (Barbour et al. 1999). The entire support reach is evaluated for each parameter.

A different field data form is completed depending upon the prevalent habitat type. For each type of stream, 10 *parameters* of the physical habitat (Table 13-1) are considered and evaluated. Most of the parameters are evaluated similarly for both types of prevalent habitats. In three cases, a parameter is evaluated differently, or a different (but ecologically equivalent) parameter is evaluated for riffle/run prevalent versus glide/pool prevalent streams. Substrate embeddedness is evaluated in riffle/run prevalent streams, while pool substrate composition is evaluated in glide/pool prevalent streams. The presence of four potential microhabitat types based on combinations of depth and current velocity is evaluated in riffle/run prevalent streams, while the presence of four types of pool microhabitat based on depth and area are evaluated in glide/pool prevalent streams. The frequency of riffles is evaluated in riffle/run prevalent streams, while channel sinuosity is evaluated in glide/pool prevalent streams. For three parameters, each bank is evaluated separately and the cumulative score (right and left) is used for the reach.

Conduct the rapid habitat assessment using the procedure presented in Table 13-2. For each of the 10 parameters, rate the overall quality of the support reach on a scale of 0 to 20. For riffle/run prevalent streams, record your scores for each parameter on the *Riffle/Run* version of the Rapid Habitat Assessment Form as shown in Figures 13-1 and 13-2. If the stream is classified as a pool/glide prevalent stream, record your scores for each parameter on the *Glide/Pool* version of the Rapid Habitat Assessment Form as shown in Figures 13-3 and 13-4. Transfer the scores assigned for each parameter to the box in the left-hand column of the form.

TABLE 13-1. DESCRIPTIONS OF PARAMETERS USED IN THE RAPID HABITAT ASSESSMENT OF STREAMS[a]

Habitat Parameter (Prevalent Habitat Type R=Riffle/run G=Glide/ pool)	Description and Rationale
	Parameters Evaluated within the Support Reach
1. Epifaunal Substrate/ Available Cover (R, G)	Includes the relative quantity and variety of natural structures in the stream, such as cobble (riffle/run), large rocks, fallen trees, logs and branches, and undercut banks, available as refugia, feeding, or sites for spawning and nursery functions of aquatic macrofauna. A wide variety and/or abundance of submerged structures in the stream provides macroinvertebrates and fish with a large number of niches, thus increasing habitat diversity. As variety and abundance of cover decreases, habitat structure becomes monotonous, diversity decreases, and the potential for recovery following disturbance decreases. Riffles and runs are critical for maintaining a variety and abundance of insects in most high-gradient streams and serving as spawning and feeding refugia for certain fish. The extent and quality of the riffle is an important factor in the support of a healthy biological condition in high-gradient streams. Riffles and runs offer a diversity of habitat through variety of particle size, and, in many small high-gradient streams, will provide the most stable habitat. Snags and submerged logs are among the most productive habitat structure for macroinvertebrate colonization and fish refugia in low-gradient streams. However, "new fall" will not yet be suitable for colonization.
2A. Embeddedness (R)	Refers to the extent to which rocks (gravel, cobble, and boulders) and snags are covered or sunken into the silt, sand, or mud of the stream bottom. Generally, as rocks become embedded, the surface area available to macroinvertebrates and fish (shelter, spawning, and egg incubation) is decreased. Embeddedness is a result of large-scale sediment movement and deposition, and is a parameter evaluated in the riffles and runs of high-gradient streams. The rating of this parameter may be variable depending on where the observations are taken. To avoid confusion with sediment deposition (another habitat parameter), observations of embeddedness should be taken in the upstream and central portions of riffles and cobble substrate areas.
2B. Pool Substrate Characterization (G)	Evaluates the type and condition of bottom substrates found in pools. Firmer sediment types (e.g., gravel, sand) and rooted aquatic plants support a wider variety of organisms than a pool substrate dominated by mud or bedrock and no plants. In addition, a stream that has a uniform substrate in its pools will support far fewer types of organisms than a stream that has a variety of substrate types.
3A. Velocity and Depth Regimes (R)	Patterns of velocity and depth are included for high-gradient streams under this parameter as an important feature of habitat diversity. The best streams in most high-gradient regions will have all 4 patterns present: (1) slow-deep, (2) slow-shallow, (3) fast-deep, and (4) fast-shallow. The general guidelines are 0.5 m depth to separate shallow from deep, and 0.3 m/sec to separate fast from slow. The occurrence of these 4 patterns relates to the stream's ability to provide and maintain a stable aquatic environment.

[a] Modified from Barbour et al. (1999)

(Continued)

TABLE 13-1[a] (Continued)

Habitat Parameter (Prevalent Habitat Type R=Riffle/ run G=Glide/ pool)	Description and Rationale
	Parameters Evaluated within the Support Reach
3B. Pool Variability (G)	Rates the overall mixture of pool types found in streams, according to size and depth. The 4 basic types of pools are large-shallow, large-deep, small-shallow, and small-deep. A stream with many pool types will support a wide variety of aquatic species. Rivers with low sinuosity (few bends) and monotonous pool characteristics do not have sufficient quantities and types of habitat to support a diverse aquatic community. General guidelines are any pool dimension (i.e., length, width, oblique) greater than half the cross-section of the stream for separating large from small and 1 m depth separating shallow and deep.
4. Sediment Deposition (R, G)	Measures the amount of sediment that has accumulated in pools and the changes that have occurred to the stream bottom as a result of deposition. Deposition occurs from large-scale movement of sediment. Sediment deposition may cause the formation of islands, point bars (areas of increased deposition usually at the beginning of a meander that increase in size as the channel is diverted toward the outer bank) or shoals, or result in the filling of runs and pools. Usually deposition is evident in areas that are obstructed by natural or manmade debris and areas where the stream flow decreases, such as bends. High levels of sediment deposition are symptoms of an unstable and continually changing environment that becomes unsuitable for many organisms.
5. Channel Flow Status (R, G)	The degree to which the channel is filled with water. The flow status will change as the channel enlarges (e.g., aggrading stream beds with actively widening channels) or as flow decreases as a result of dams and other obstructions, diversions for irrigation, or drought. When water does not cover much of the streambed, the amount of suitable substrate for aquatic organisms is limited. In high-gradient streams, riffles and cobble substrate are exposed; in low-gradient streams, the decrease in water level exposes logs and snags, thereby reducing the areas of good habitat. Channel flow is especially useful for interpreting biological condition under abnormal or lowered flow conditions. This parameter becomes important when more than one biological index period is used for surveys or the timing of sampling is inconsistent among sites or annual periodicity.
	Parameters Evaluated Broader than the Support Reach
6. Channel Alteration (R, G)	Is a measure of large-scale changes in the shape of the stream channel. Many streams in urban and agricultural areas have been straightened, deepened, or diverted into concrete channels, often for flood control or irrigation purposes. Such streams have far fewer natural habitats for fish, macroinvertebrates, and plants than do naturally meandering streams. Channel alteration is present when artificial embankments, riprap, and other forms of artificial bank stabilization or structures are present; when the stream is very straight for significant distances; when dams and bridges are present; and when other such changes have occurred. Scouring is often associated with channel alteration.

[a] Modified from Barbour et al. (1999)

(Continued)

TABLE 13-1[a] (Continued)

Habitat Parameter (Prevalent Habitat Type R=Riffle/run G=Glide/ pool)	Description and Rationale
	Parameters Evaluated Broader than the Support Reach
7A. Frequency of Riffles (or Bends) (R)	Is a way to measure the sequence of riffles and thus the heterogeneity occurring in a stream. Riffles are a source of high-quality habitat and diverse fauna, therefore, an increased frequency of occurrence greatly enhances the diversity of the stream community. For high gradient streams where distinct riffles are uncommon, a run/bend ratio can be used as a measure of meandering or sinuosity (see 7b). A high degree of sinuosity provides for diverse habitat and fauna, and the stream is better able to handle surges when the stream fluctuates as a result of storms. The absorption of this energy by bends protects the stream from excessive erosion and flooding and provides refugia for benthic invertebrates and fish during storm events. To gain an appreciation of this parameter in some streams, a longer segment or reach than that designated for sampling should be incorporated into the evaluation. In some situations, this parameter may be rated from viewing accurate topographical maps. The "sequencing" pattern of the stream morphology is important in rating this parameter. In headwaters, riffles are usually continuous and the presence of cascades or boulders provides a form of sinuosity and enhances the structure of the stream. A stable channel is one that does not exhibit progressive changes in slope, shape, or dimensions, although short-term variations may occur during floods (Gordon et al. 1992).
7B. Channel Sinuosity (G)	Evaluates the meandering or sinuosity of the stream. A high degree of sinuosity provides for diverse habitat and fauna, and the stream is better able to handle surges when the stream fluctuates as a result of storms. The absorption of this energy by bends protects the stream from excessive erosion and flooding and provides refugia for benthic invertebrates and fish during storm events. To gain an appreciation of this parameter in low gradient streams, a longer segment or reach than that designated for sampling may be incorporated into the evaluation. In some situations, this parameter may be rated from viewing accurate topographical maps. The "sequencing" pattern of the stream morphology is important in rating this parameter. In "oxbow" streams of coastal areas and deltas, meanders are highly exaggerated and transient. Natural conditions in these streams are shifting channels and bends, and alteration is usually in the form of flow regulation and diversion. A stable channel is one that does not exhibit progressive changes in slope, shape, or dimensions, although short-term variations may occur during floods (Gordon et al. 1992).

[a] Modified from Barbour et al. (1999) **(Continued)**

TABLE 13-1[a] (Continued)

Habitat Parameter (Prevalent Habitat Type R=Riffle/run G=Glide/ pool)	Description and Rationale
	Parameters Evaluated Broader than the Support Reach
8. Bank Stability (Condition of Banks) (R, G)	Measures whether the stream banks are eroded (or have the potential for erosion). Steep banks are more likely to collapse and suffer from erosion than are gently sloping banks, and are therefore considered to be unstable. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil. Eroded banks indicate a problem of sediment movement and deposition, and suggest a scarcity of cover and organic input to streams. Each bank is evaluated separately and the cumulative score (right and left) is used for this parameter.
9. Bank Vegetative Protection (R, G)	Measures the amount of vegetative protection afforded to the stream bank and the near-stream portion of the riparian zone. The root systems of plants growing on stream banks help hold soil in place, thereby reducing the amount of erosion that is likely to occur. This parameter supplies information on the ability of the bank to resist erosion as well as some additional information on the uptake of nutrients by the plants, the control of instream scouring, and stream shading. Banks that have full, natural plant growth are better for fish and macroinvertebrates than are banks without vegetative protection or those shored up with concrete or riprap. This parameter is made more effective by defining the native vegetation for the region and stream type (i.e., shrubs, trees, etc.). In some regions, the introduction of exotics has virtually replaced all native vegetation. The value of exotic vegetation to the quality of the habitat structure and contribution to the stream ecosystem must be considered in this parameter. In areas of high grazing pressure from livestock or where residential and urban development activities disrupt the riparian zone, the growth of a natural plant community is impeded and can extend to the bank vegetative protection zone. Each bank is evaluated separately and the cumulative score (right and left) is used for this parameter.
10. Riparian Vegetated Zone Width (R, G)	Measures the width of natural vegetation from the edge of the stream bank out through the riparian zone. The vegetative zone serves as a buffer to pollutants entering a stream from runoff, controls erosion, and provides habitat and nutrient input into the stream. A relatively undisturbed riparian zone supports a robust stream system; narrow riparian zones occur when roads, parking lots, fields, lawns, bare soil, rocks, or buildings are near the stream bank. Residential developments, urban centers, golf courses, and rangeland are the common causes of anthropogenic degradation of the riparian zone. Conversely, the presence of "old field" (i.e., a previously developed field not currently in use), paths, and walkways in an otherwise undisturbed riparian zone may be judged to be inconsequential to altering the riparian zone and may be given relatively high scores. For variable size streams, the specified width of a desirable riparian zone may also be variable and may be best determined by some multiple of stream width (e.g., 4 x wetted stream width). Each bank is evaluated separately and the cumulative score (right and left) is used for this parameter.

[a] Modified from Barbour et al. (1999)

TABLE 13-2. PROCEDURE FOR CONDUCTING THE RAPID HABITAT ASSESSMENT

1. Based on observations during the day's sample collection and field measurement activities, classify the entire support reach as predominantly flowing water habitat (*Riffle/run*) or slow water habitat (*Glide/pool*). Choose the prevalent habitat type based on which habitat type occupies the majority of the length of the sampling reach.

2. Select the appropriate version of the Rapid Habitat Assessment Form (*Riffle/Run* or *Glide/Pool*) based on the classification in Step 1.

3. For each of the 10 habitat parameters, determine the general *quality category* (*POOR*, *MARGINAL*, *SUB-OPTIMAL*, or *OPTIMAL*) of the entire support reach. Use the descriptions provided on the data form to assign a score from the values available within each quality category. Circle the assigned score for each parameter on the form.

   NOTE: For parameters 1 through 7, the sampling reach can be scored from 0 (worst) to 20 (best). For parameters 8 through 10, each bank is evaluated separately (from 0 to 10), and the cumulative score for both right and left banks are used.

4. After the support reach has been scored for all parameters, transfer the score circled for each parameter to the corresponding *SCORE* box in the *HABITAT PARAMETER* column of the assessment form.

Reviewed by (initials): EM

RAPID HABITAT ASSESSMENT FORM: RIFFLE/RUN - STREAM

SITE ID: WXXP99-9999 DATE: 07/02/2004

HABITAT PARAMETER	CONDITION CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
1. Epifaunal Substrate/ Available Cover	Greater than 70% of substrate favorable for epifaunal colonization and fish cover; mix of snags, submerged logs, undercut banks, cobble or other stable habitat and at stage to allow full colonization potential; (i.e., logs/snags that are NOT new fall and NOT transient.)	40-70% mix of stable habitat; well-suited for full colonization potential; adequate habitat for maintenance of populations; presence of additional substrate in the form of newfall, but not yet prepared for colonization (may rate at high end of scale)	20-40% mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed.	Less than 20% stable habitat; lack of habitat is obvious; substrate unstable or lacking.
Score: 12	20 19 18 17 16	15 14 13 (12) 11	10 9 8 7 6	5 4 3 2 1 0
2. Embeddedness	Gravel, cobble, and boulder particles are 0-25% surrounded by fine sediment. Layering of cobble provides diversity of niche space.	Gravel, cobble, and boulder particles are 25-50% surrounded by fine sediment.	Gravel, cobble, and boulder particles are 50-75% surrounded by fine sediment.	Gravel, cobble, and boulder particles are more than 75% surrounded by fine sediment.
Score: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
3. Velocity/Depth Regime	All four velocity/depth regimes present (slow-deep, slow-shallow, fast-deep, fast-shallow). (Slow is less than 0.3 m/s, deep is greater than 0.5 m.)	Only 3 of the 4 regimes present (if fast-shallow is missing, score lower than if missing other regimes).	Only 2 of the 4 habitat regimes present (if fast-shallow or slow-shallow are missing, score low).	Dominated by 1 velocity/depth regime (usually slow-deep).
Score: 15	20 19 18 17 16	(15) 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
4. Sediment Deposition	Little or no enlargement of islands or point bars and less than 5% of the bottom affected by sediment deposition.	Some new increases in bar formation, mostly from gravel, sand or fine sediment; 5-30% of the bottom affected; slight deposition in pools.	Moderate deposition of new gravel, sand or fine sediment on old and new bars; 30-50% of the bottom affected; sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools prevalent.	Heavy deposits of fine material; increased bar development; more than 50% of the bottom changing frequently; pools almost absent due to substantial sediment deposition.
Score: 14	20 19 18 17 16	15 (14) 13 12 11	10 9 8 7 6	5 4 3 2 1 0
5. Channel Flow Status	Water reaches base of both lower banks, and minimal amount of channel substrate is exposed.	Water fills over 75% of the available channel; or less than 25% of channel substrate is exposed.	Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed.	Very little water in channel and mostly present as standing pools.
Score: 12	20 19 18 17 16	15 14 13 (12) 11	10 9 8 7 6	5 4 3 2 1 0
6. Channel Alteration	Channelization or dredging absent or minimal; stream with normal pattern.	Some channelization present, usually in areas of bridge abutments; evidence of past channelization, i.e., dredging, (greater than past 20 yr) may be present, but recent channelization is not present.	Channelization may be extensive; embankments or shoring structures present on both banks; and 40 to 80% of stream reach channelized and disrupted.	Banks shored with gabion or cement; over 80% of the stream reach channelized and disrupted. Instream habitat greatly altered or removed entirely.
Score: 18	20 19 (18) 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

3099

04/14/2000 2000 Riffle Run

Figure 13-1. Rapid Habitat Assessment Form for riffle/run prevalent streams (page 1).

Reviewed by (initials): EM

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

SITE ID: WXXP99-9999 DATE: 07/02/2004

HABITAT PARAMETER	CONDITION CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
7. Frequency of Riffles (or bends)	Occurrence of riffles relatively frequent; ratio of distance between riffles divided by width of the stream greater than 7:1 (generally 5 to 7); variety of habitat is key. In streams where riffles are continuous, placement of boulders or other large, natural obstruction is important.	Occurrence of riffles infrequent; distance between riffles divided by width of stream is between 7 to 15.	Occasional riffle or bend; bottom contours provide some habitat; distance between riffles divided by width of stream is between 15 to 25.	Generally all flat water or shallow riffles; poor habitat; distance between riffles divided by width of stream is a ratio of over 25.
Score: 13	20 19 18 17 16	15 14 (13) 12 11	10 9 8 7 6	5 4 3 2 1 0
8. Bank Stability (score each bank) NOTE: Determine left or right side by facing downstream.	Banks stable; evidence of erosion or bank failure absent or minimal; little potential for future problems. Less than 5% of bank affected.	Moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion.	Moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; obvious bank sloughing; 60-100% of bank has erosional scars.
Left Bank Score: 7	Left Bank: 10 9	8 (7) 6	5 4 3	2 1 0
Right Bank Score: 5	Right Bank: 10 9	8 7 6	(5) 4 3	2 1 0
9. Vegetative Protection (score each bank)	More than 90% of the streambank surfaces and immediate riparian zone covered by native vegetation, including trees, understory shrubs, or nonwoody macrophytes; vegetative disruption through grazing or mowing minimal or not evident; almost all plants allowed to grow naturally.	70-90% of the streambank surfaces covered by native vegetation; but one class of plants is not well represented; disruption evident but not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	50-70% of the streambank surfaces covered by vegetation; disruptions obvious; patches of bare soil or closely cropped vegetation common; less than one-half of the potential plant stubble height remaining.	Less than 50% of the streambank surfaces covered by vegetation; disruption of streambank vegetation is very high; vegetation has been removed to 5 centimeters or less in average stubble height.
Left Bank Score: 8	Left Bank: 10 9	(8) 7 6	5 4 3	2 1 0
Right Bank Score: 7	Right Bank: 10 9	8 (7) 6	5 4 3	2 1 0
10. Riparian Vegetative Zone Width (score each bank)	Width of riparian zone greater than 18 meters; human activities (i.e., parking lots, roadbeds, clear-cuts, lawns, or crops) have not impacted the zone.	Width of riparian zone 12-18 meters; human activities have impacted zone only minimally.	Width of riparian zone 6-12 meters; human activities have impacted zone a great deal.	Width of riparian zone less than 6 meters; little or no riparian vegetation due to human activities.
Left Bank Score: 6	Left Bank: 10 9	8 7 (6)	5 4 3	2 1 0
Right Bank Score: 5	Right Bank: 10 9	8 7 6	(5) 4 3	2 1 0

3099

04/14/2000 2000 Riffle Run

Figure 13-2. Rapid Habitat Assessment Form for riffle/run prevalent streams (page 2).

Reviewed by (Initials): EM

RAPID HABITAT ASSESSMENT FORM: GLIDE/POOL - STREAMS

SITE ID: WXXP99-9999 DATE: 07/02/2004

HABITAT PARAMETER	CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
1. Epifaunal Substrate/ Available Cover	Greater than 50% of substrate favorable for epifaunal colonization and fish cover; mix of snags, submerged logs, undercut banks, cobble or other stable habitat and at stage to allow full colonization potential (i.e. logs/snags that are NOT new fall and NOT transient.)	30-50% mix of stable habitat; well-suited for full colonization potential; adequate habitat for maintenance of populations; presence of additional substrate in the form of newfall, but not yet prepared for colonization (may rate at high end of scale)	10-30% mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed	Less than 10% stable habitat; lack of habitat is obvious; substrate unstable or lacking.
Score: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
2. Pool Substrate Characterization	Mixture of substrate materials, with gravel and firm sand prevalent; root mats and submerged vegetation common.	Mixture of soft sand, mud, or clay; mud may be dominant; some root mats and submerged vegetation present.	All mud or clay or sand bottom; little or no root mat; no submerged vegetation.	Hard-pan clay or bedrock; no root mat or vegetation.
Score: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
3. Pool Variability	Even mix of large-shallow, large-deep, small shallow, small-deep pools present.	Majority of pools large-deep; very few shallows.	Shallow pools much more prevalent than deep pools.	Majority of pools small-shallow or absent.
Score: 8	20 19 18 17 16	15 14 13 12 11	10 9 (8) 7 6	5 4 3 2 1 0
4. Sediment Deposition	Little or no enlargement of islands or point bars and less than 20% of the bottom affected by sediment deposition.	Some new increases in bar formation, mostly from gravel, sand or fine sediment; 20-50% of the bottom affected; slight deposition in pools.	Moderate deposition of new gravel, sand or fine sediment on old and new bars; 50-80% of the bottom affected; sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools prevalent.	Heavy deposits of fine material; increased bar development; more than 80% of the bottom changing frequently; pools almost absent due to substantial sediment deposition.
Score: 7	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
5. Channel Flow Status	Water reaches base of both lower banks, and minimal amount of channel substrate is exposed.	Water fills over 75% of the available channel; or less than 25% of channel substrate is exposed.	Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed.	Very little water in channel and mostly present as standing pools.
Score: 18	20 19 (18) 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
6. Channel Alteration	Channelization or dredging absent or minimal; stream with normal pattern.	Some channelization present, usually in areas of bridge abutments; evidence of past channelization, i.e., dredging, (greater than past 20 yr) may be present, but recent channelization is not present.	Channelization may be extensive; embankments or shoring structures present on both banks; and 40 to 80% of stream reach channelized and disrupted.	Banks shored with gabion or cement; over 80% of the stream reach channelized and disrupted. Instream habitat greatly altered or removed entirely.
Score: 16	20 19 18 17 (16)	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

1475

04/14/2000 2000 Glide Pool

Figure 13-3. Rapid Habitat Assessment Form for glide/pool prevalent streams (page 1).

Reviewed by (initials): EM

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

SITE ID: WXXP99-9999 DATE: 07/02/2004

HABITAT PARAMETER	CATEGORY			
	OPTIMAL	SUB-OPTIMAL	MARGINAL	POOR
7. Channel Sinuosity	The bends in the stream increase the stream length 3 to 4 times longer than if it was in a straight line. (Note: channel braiding is considered normal in coastal plains and other low-lying areas. This parameter is not easily rated in these areas.)	The bends in the stream increase the stream length 2 to 3 times longer than if it was in a straight line.	The bends in the stream increase the stream length 1 to 2 times longer than if it was in a straight line.	Channel straight; waterway has been channelized for a long distance.
Score: 13	20 19 18 17 16	15 14 (13) 12 11	10 9 8 7 6	5 4 3 2 1 0
8. Bank Stability (score each bank) NOTE: Determine left or right side by facing downstream.	Banks stable; evidence of erosion or bank failure absent or minimal; little potential for future problems. Less than 5% of bank affected.	Moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion.	Moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; obvious bank sloughing; 60-100% of bank has erosional scars.
Left Bank Score: 9	Left Bank: 10 (9)	8 7 6	5 4 3	2 1 0
Right Bank Score: 10	Right Bank: (10) 9	8 7 6	5 4 3	2 1 0
9. Vegetative Protection (score each bank)	More than 90% of the streambank surfaces and immediate riparian zone covered by native vegetation, including trees, understory shrubs, or nonwoody macrophytes; vegetative disruption through grazing or mowing minimal or not evident; almost all plants allowed to grow naturally.	70-90% of the streambank surfaces covered by native vegetation; but one class of plants is not well represented; disruption evident but not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	50-70% of the streambank surfaces covered by vegetation; disruptions obvious; patches of bare soil or closely cropped vegetation common; less than one-half of the potential plant stubble height remaining.	Less than 50% of the streambank surfaces covered by vegetation; disruption of streambank vegetation is very high; vegetation has been removed to 5 centimeters or less in average stubble height.
Left Bank Score: 4	Left Bank: 10 9	8 7 6	5 (4) 3	2 1 0
Right Bank Score: 6	Right Bank: 10 9	8 7 (6)	5 4 3	2 1 0
10. Riparian Vegetation Zone Width (score each bank)	Width of riparian zone greater than 18 meters; human activities (i.e., parking lots, roadbeds, clear-cuts, lawns, or crops) have not impacted the zone.	Width of riparian zone 12-18 meters; human activities have impacted zone only minimally.	Width of riparian zone 6-12 meters; human activities have impacted zone a great deal.	Width of riparian zone less than 6 meters; little or no riparian vegetation due to human activities.
Left Bank Score: 5	Left Bank: 10 9	8 7 6	(5) 4 3	2 1 0
Right Bank Score: 7	Right Bank: 10 9	8 (7) 6	5 4 3	2 1 0

1476

04/14/2000 2000 Glide Pool

Figure 13-4. Rapid Habitat Assessment Form for glide/pool prevalent streams (page 2).

13.2 GENERAL VISUAL STREAM ASSESSMENT

The visual assessment form is designed as a template for recording pertinent field observations. It is not comprehensive, so record any additional observations in the *General Assessment* section of the form. Fill out the assessment form after all other sampling and measurement activities have been completed. Consider only things at or upstream of the X-site (i.e., things that may influence the sampling point). Take into account all observations the sampling team has made while at the site. The assessment includes the following components: watershed activities and observed disturbances, site characteristics, weather during sampling, and a general assessment. Conduct the general visual assessment of the support reach as described in Table 13-3. Record data and observations for each component of the assessment on the Assessment Form as shown in Figure 13-5. The visual assessment form is used in place of a more detailed field journal.

Each watershed activity or disturbance is rated into one of four categories of abundance or influence: not observed, low, medium, or high. Leave the line blank for any activity or disturbance type not observed. The distinction between low, medium, and high will be subjective. For example, if there are 2-3 houses away from the stream, the rating for *Houses* may be *LOW*. If the stream is in a suburban housing development, rate it as *HIGH*. Similarly, a small patch of clear cut logging on a hill overlooking the stream would be rated as *LOW*. Logging activity right on the stream shore, however, would be rated as *HIGH*.

When assessing site characteristics, consider the land use and other activities within a circle with a 200 m radius around the X-site (400 m diameter). Waterbody character is defined as *the physical habitat integrity of the water body, largely a function of riparian and littoral habitat structure, volume change, trash, turbidity, slicks, scums, color, and odors.* Assess two attributes of waterbody character: the degree of human development, and aesthetics. Rate each of these attributes on a scale of 1 to 5. For development, give the stream a rating of *5* if it is *pristine*, with no signs of any human development or activity. A rating of *1* indicates a stream is in an area that is totally developed (e.g., the entire stream channel is lined with houses, or the riparian zone has been removed). For aesthetics, base your decision on any factor about the stream that bothers you (e.g., trash, algal growth, weed abundance, overcrowding). Also, rate the presence/absence of beaver activity and the dominant land use within this circle according to the classes listed on the form.

TABLE 13-3. PROCEDURE FOR CONDUCTING THE GENERAL VISUAL ASSESSMENT OF A STREAM

1. After all other sampling and measurement activities are completed, fill out the header section of an Assessment Form. Use your perceptions obtained throughout the day, while at the stream or driving/walking through the catchment to complete the remainder of the form. *Consider only things* ***at or upstream*** *of the X-site.*

2. ***Watershed Activities and Disturbances Observed***: Rate each type of activity or disturbance listed on the form as either *NOT OBSERVED*, *LOW*, *MEDIUM*, or *HIGH*, and record the rating on the Assessment Form. Keep in mind that ratings will be somewhat subjective and that an extensive effort to quantify the presence and intensity of each type of stressor is not required. General categories of activities and types of disturbance are described below:

   - *RESIDENTIAL*: The presence of any of the listed disturbances next to or near the stream.
   - *RECREATIONAL*: The presence of organized public or private parks, campgrounds, beaches or other recreation areas around the stream. If there are signs of informal areas of camping, swimming or boating around the stream (e.g., a swimming hole), record them as "primitive" parks, camping.
   - *AGRICULTURE*: The presence of cropland, pasture, range, orchards, poultry, and/or livestock. Also note any evidence of water withdrawals for agriculture.
   - *INDUSTRIAL*: Any industrial activity (e.g., canning, chemical, pulp), commercial activity (stores, businesses) or logging/mining activities around the stream or in the catchment. Describe in more detail in the comments section.
   - *MANAGEMENT*: Any evidence of water treatment, dredging or channelization, flow control structures, fish stocking, dams or other management activities.

   Any oddities, or further elaboration should be recorded in the COMMENTS section.

3. ***Site Characteristics***: (based on a circle with a *200 m radius* around the X-site)

   - *WATERBODY CHARACTER*: Assign a rating of *1* (highly disturbed) to *5* (pristine) based on your general impression of the intensity of impact from human disturbance. Mark the box next to the assigned rating on the Assessment Form. Assign a rating to the stream based on overall aesthetic quality, based on your opinion of how suitable the stream water is for recreation and aesthetic enjoyment today. Mark the box next to the assigned rating on the Assessment Form.

     5 Beautiful, could not be any nicer.
     4 Very minor aesthetic problems; excellent for swimming, boating, enjoyment.
     3 Enjoyment impaired.
     2 Level of enjoyment substantially reduced.
     1 Enjoyment nearly impossible.

(Continued)

TABLE 13-3 (Continued)

- *BEAVER*: If you noticed any signs of beaver presence in the stream (chewed sticks, trees, dams, lodges) rate the beaver presence as either *RARE* or *COMMON*. If no beaver signs were present, mark the *ABSENT* box. Also rate the amount of flow modification caused by any beaver activity as *NONE*, *MINOR*, or *MAJOR*.

- *DOMINANT LAND USE:* Make one estimate of the dominant land use in the circle around the x-site. Pick just one land use from among *FOREST*, *AGRICULTURE*, *RANGE*, *URBAN*, or *SUBURBAN/TOWN*. If there are other major land uses, make note of them in the *GENERAL ASSESSMENT* section of the form. If forest is the dominant land use, make a guess as to the dominant age class of the forest (0-25, 25-75, or > 75 years).

3. *WEATHER*: record a very brief description of the weather conditions during stream sampling (e.g., sunny, fair, partly cloudy, overcast, light rain, unseasonably warm, cold, or hot, etc.). Any unusual weather right before sampling (e.g., heavy rain, 6 inches of snow) is also worth noting here.

4. *GENERAL ASSESSMENT*: record comments on wildlife observed, perceived diversity of terrestrial/riparian vegetation, or overall biotic integrity on the Assessment Form. Record any information regarding the past or present characteristics or condition of the stream provided by local residents here as well.

STREAM ASSESSMENT FORM - STREAMS/RIVERS

Reviewed by (initial): EM

SITE ID: WXXP99-9999 DATE: 07/02/2004

WATERSHED ACTIVITIES AND DISTURBANCES OBSERVED (Intensity: Blank=Not observed, L=Low, M=Moderate, H=Heavy)

Residential	Recreational	Agricultural	Industrial	Stream Management
(L) M H Residences	L M H Hiking Trails	L M H Cropland	L M H Industrial Plants	L M H Levees
(L) M H Maintained Lawns	L M H Parks, Campgrounds	L M (H) Pasture	L M H Mines/Quarries	L M H Chemical Treatment
L M H Construction	L M H Primitive Parks, Camping	L (M) H Livestock Use	L M H Oil/Gas Wells	L M H Angling Pressure
L M H Pipes, Drains	L M H Trash/Litter	L M H Orchards	L M H Power Plants	L M H Dredging
L M H Dumping	L M H Surface Ferns	L M H Poultry	L M H Logging	L M H Channelization
(L) M H Roads		L M H Irrigation Equip.	L M H Evidence of Fire	L M H Water Level Fluctuations
L M H Bridge/Culverts		L M H Water Withdrawal	L M H Others	L M H Fish Stocking
L M H Sewage Treatment			L M H Commercial	L M H Dams

SITE CHARACTERISTICS (200 m radius)

Waterbody Character	Pristine	☐ 5	☐ 4	☒ 3	☐ 2	☐ 1	Highly Disturbed
	Appealing	☐ 5	☐ 4	☐ 3	☒ 2	☐ 1	Unappealing
Beaver	Beaver Signs:	☒ Absent		☐ Rare		☐ Common	
	Beaver Flow Modifications:	☒ None		☐ Minor		☐ Major	
Dominant Land Use	Dominant Land Use Around X	☐ Forest	☐ Agriculture	☒ Range	☐ Urban	☐ Suburban/Town	
	If Forest, Dominant Age Class	☐ 0 - 25 yrs	☐ 25 - 75 yrs	☐ > 75 yrs.			

WEATHER CLEAR, WITH LIGHT RAIN IN THE PREVIOUS 24 HOURS. AIR TEMP WAS 28 °C AT 11 AM.

GENERAL ASSESSMENT (Biotic integrity, Vegetation diversity, Local anecdotal information)

RIPARIAN TREES ARE 25-75 YRS. OLD. RIPARIAN TREE DENSITY IS LOW. LOCAL CONTACT REMEMBERS A DAM LOCATED JUST DOWNSTREAM OF THE X-SITE LOCATION. DAM WAS WASHED AWAY 10 YRS. AGO DURING A LARGE FLOOD EVENT. NO BIRDS OR OTHER WILDLIFE WERE OBSERVED DURING THE SITE VISIT.

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03/26/2001 2001 Stream Assessment

Figure 13-5. Stream Assessment Form (page 1).

The weather and general assessment components include any observations that will help in data interpretation. The weather component is just a place to record a brief description of the weather during sampling or just before sampling. General assessment comments can include comments on wildlife observed, diversity of terrestrial/riparian vegetation, overall biotic integrity, or any other observation. Comments from locals about current or past conditions are often useful and should be recorded in this section as well. The back side of the form (Figure 13-6) is available for additional general comments.

13.3 EQUIPMENT AND SUPPLIES

Figure 13-7 is a checklist of the supplies required to complete the rapid habitat and general visual stream assessments. This checklist may differ from the checklists presented in Appendix A, which are used at a base site to ensure that all equipment and supplies are brought to and are available at the stream site. Field teams are required to use the checklist presented in this section to ensure that equipment and supplies are organized and available to conduct the protocols efficiently.

13.3 LITERATURE CITED

Barbour, M.T., J. Gerritsen, B.D. Snyder, and J.B. Stribling. 1999. *Rapid bioassessment protocols for use in streams and wadeable rivers: periphyton, benthic macroinvertebrates, and fish.* 2nd edition. EPA/841-B-99-002. U.S. Environmental Protection Agency, Office of Water, Assessment and Watershed Protection Division, Washington, D.C.

Gordon, N.D., T.A. McMahon, and B.L. Finlayson. 1992. *Stream hydrology: an introduction for ecologists.* John Wiley and Sons, Inc., West Sussex, England.

Lazorchak, J.M., A.T. Herlihy, and J. Green. 1998. Rapid habitat and visual stream assessments. Pages 193-209 in J.M. Lazorchak, D.J. Klemm, and D.V. Peck (editors). *Environmental Monitoring and Assessment Program-Surface Waters: field operations and methods for measuring the ecological condition of wadeable streams.* EPA/620/R-94/004F. U.S. Environmental Protection Agency, Washington, D.C.

Plafkin, J.L., M.T. Barbour, K.D. Porter, S.K. Gross, and R.M. Hughes. 1989. *Rapid bioassessment protocols for use in streams and rivers: benthic macroinvertebrates and fish.* EPA/440/4-89/001. U.S. Environmental Protection Agency, Assessment and Watershed Protection Division, Washington, D.C.

STREAM ASSESSMENT FORM - STREAM/RIVERS (cont.) Reviewed by (initial): EM

SITE ID: WXXP99-9999 DATE: 07/02/2004

GENERAL ASSESSMENT (continued)

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03/26/2001 2001 Stream Assessment

Figure 13-6. Stream Assessment Form (page 2).

EQUIPMENT AND SUPPLIES FOR RAPID HABITAT AND GENERAL VISUAL STREAM ASSESSMENTS

QTY.	Item	
1	Rapid Habitat Assessment Form for Riffle/run prevalent streams	
1	Rapid Habitat Assessment Form for Pool/glide prevalent streams	
1	Assessment Form for visual stream assessment	
6	Soft (#2) lead pencils	
1	Covered clipboard or forms holder	
1 copy	Field operations and methods manual	
1 set	Laminated sheets of procedure tables and/or quick reference guides for rapid habitat and visual assessments	

Figure 13-7. Checklist of equipment and supplies required for rapid habitat and general visual stream assessments.

NOTES

SECTION 14
FINAL SITE ACTIVITIES

James M. Lazorchak[1]

Before leaving a stream site, the team leader reviews all of the data forms and sample labels for accuracy, completeness, and legibility. Use the checklist presented in Figure 14-1 to help with the review. A second team member inspects all sample containers and packages them in preparation for transport, storage, or shipment. Refer to Section 3 for details on preparing and shipping samples.

When reviewing field data forms, ensure that all required data forms for the stream have been completed. Confirm that the stream identification code, the year, the visit number, and the date of the visit are correct on all forms. On each form, verify that all information has been recorded accurately, the recorded information is legible, and any flags are explained in the comments section. Ensure that written comments are legible and do not use shorthand or abbreviations. Make sure the header information is completed on all pages of each form. After reviewing each form, initial the upper right corner of each page.

When inspecting samples, ensure that each sample is labeled, all labels are completely filled in and legible, and each label is covered with clear plastic tape. Compare sample label information with the information recorded on the corresponding field data forms (e.g., the Sample Collection Form) to ensure accuracy.

The other team members should return all of the equipment and supplies to the vehicle for transport and clean up the stream site. Pack all equipment and supplies in the vehicle for transport. Keep them organized so they can be inventoried using the equipment and supply checklists presented in Appendix A. Clean up and dispose of all waste material at the stream site. Transport it out of the area if necessary.

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

FORM AND LABEL CHECKLIST					
SITE ID:			DATE:		
FORM	Are Site ID, Date, and Visit Number recorded?	Is header information recorded on both sides?	Is information complete, correct, and legible?	Are flags assigned correctly and explained?	Initials on Form?
Stream Verification					
Sample Collection					
Targeted Habitat Periphyton (optional)					
Discharge Measurement					
Channel/Riparian Cross-section and Thalweg Profile (11 copies)					
Slope and Bearing					
Legacy Tree and Alien Invasive Plant					
Channel Constraint and Field Measurement					
Torrent Evidence					
Vertebrate Collection					
Rapid Habitat Assessment (either riffle/run or pool/glide) (optional)					
Visual Assessment					

LABEL	Is Site ID and Date recorded?	Do all subsamples (or extra containers) have the same sample ID?	Is the Sample ID on the collection form correct?	Is there a label inside the sample container?	Are labels covered with clear tape?
Water Chemistry					
Periphyton					
Targeted Habitat Periphyton (optional)					
Benthos (Reachwide)					
Benthos (Targeted Riffle)					
Vertebrates (Jar)					
Fish Tissue (Big) (up to 9)					
Fish Tissue (Small)					

Figure 14-1. Checklist for reviewing field data forms and sample labels.

APPENDIX A
EQUIPMENT AND SUPPLY CHECKLISTS

The checklists in this section represent a master list that can be used when loading field vehicles for a sampling visit. Each item is listed only once, with lists being organized to some extent by storage requirements rather than by individual indicator. Use the checklists presented at the end of each indicator section to organize yourself for various sampling and data acquisition activities at the stream site.

FIELD DATA FORMS AND SAMPLE LABELS A-2

OFFICE SUPPLIES AND TOOLS A-3

SITE-RELATED INFORMATION A-3

PERSONAL EQUIPMENT AND SUPPLIES A-4

CHEMICALS A-5

PACKING AND SHIPPING SUPPLIES A-5

SITE VERIFICATION AND SAMPLING REACH LAYOUT A-6

WATER CHEMISTRY A-6

STREAM DISCHARGE A-7

PHYSICAL HABITAT A-7

PERIPHYTON A-8

BENTHIC MACROINVERTEBRATES A-9

AQUATIC VERTEBRATES AND FISH TISSUE CONTAMINANTS A-10

FIELD DATA FORMS AND SAMPLE LABELS

Number per site	Item	
1	Stream Verification Form	
1	Sample Collection Form and Stream Discharge Form	
1 (optional)	Modified STAR Protocol Periphyton Data Form	
11 + extras	Channel/Riparian Cross-section and Thalweg Profile Form	
1	Slope and Bearing Form	
1	Riparian "Legacy" Tree and Invasive Alien Plants Form	
1	Channel Constraint and Field Measurement Form and Torrent Evidence Assessment Form	
2-3	Vertebrate Collection Form	
1 (optional)	Rapid Habitat Assessment Form for Riffle/run prevalent streams	
1 (optional)	Rapid Habitat Assessment Form for Glide/pool prevalent streams	
1	Assessment Form for visual stream assessment	
2 + extras	Sample Tracking Form	
3	Water chemistry labels (same ID number)	
3	Periphyton labels (same ID number)	
1 (optional)	Periphyton STAR Protocol ID label	
1	Reachwide Benthic sample labels, with preprinted ID numbers	
1	Targeted Riffle Benthic sample labels with preprinted ID numbers	
1 sheet	Benthic labels for extra containers (no preprinted ID number)	
1 sheet	Blank benthic sample labels on waterproof paper for inside of jars	
1 sheet	Pre-printed aquatic vertebrate jar labels (4) and voucher bag tags (36), all with same preprinted sample ID number	
1 sheet	Fish tissue sample labels (up to 10 different sample ID numbers)	
2 copies	Field operations and methods manual	
2 sets	Laminated sheets of procedure tables and/or quick reference guides	

OFFICE SUPPLIES AND TOOLS

Number per site	Item	
4	Covered clipboards or forms holders	
1 (optional)	Field notebook	
12	Soft (#2) lead pencils	
6	Fine-tip indelible markers	
1 roll	Duct tape	
1 pair	Scissors for cutting labels	
1	Pocket knife or multipurpose tool	
1	Battery charger (if needed for electrofishing unit)	
1	Toolbox with basic tools needed to maintain/repair sampling gear	

SITE-RELATED INFORMATION

Number per site	Item	
1	Dossier of access information for scheduled stream site	
1	Topographic map with *X-site* marked	
1	Site information sheet with map coordinates and elevation of *X-site*	
1	Sampling itinerary form or notebook	
1	Safety log and/or personal safety information for each team member	
1	Sheet of cardstock or thin cardboard (white with non-glossy finish or light gray) with Site ID printed on it for use with site photographs	
1	List of protected aquatic taxa for watershed	
1 ea.	All required scientific collection permits (State, Federal, Tribal)	

PERSONAL EQUIPMENT AND SUPPLIES

Number per site	Item	
1 pair per person	Chest waders with nonslip boots if waders are the "stocking" type. Hip waders can be used in shallower streams (except for electrofishing). Note felt-soled boots should not be used in areas where nuisance organisms are present.	
1 per person	Life vests	
3 pair	Polarized sunglasses	
1	First aid kit (including eye wash solution)	
1 per person	Rain gear	
1 or 2	Fisherman's vest or fanny pack for physical habitat characterization.	
1 per person	Safety whistles	
1 pair per person	Earplugs (if gas-powered generators are used)	
1 per person	Day packs, backs, fanny packs, and/or dry bags for personal gear	
1 ea.	Insect repellent, sunscreen, barrier ointment or cleanser for poison oak, hand sanitizer, water purifier unit	
1	Patch kit for waders	

CHEMICALS

Number per site	Item	
1 pair	Safety glasses	
2 pair	Chemical-resistant gloves	
1	Laboratory apron, resistant to ethanol and formalin	
1	Cooler (with suitable absorbent material) for transporting ethanol and samples	
2 gal	95% ethanol	
1	Cooler (with suitable absorbent material) for transporting formaldehyde/formalin	
2 gal	10% (buffered) formalin solution *OR* 0.2 gal (0.75 L) buffered undiluted formalin solution	
1 gal	Disinfectant solution for whirling disease, New Zealand mud snails, and amphibian chytrid fungus	
	Gasoline for electrofishing unit in approved container	

PACKING AND SHIPPING SUPPLIES

Number per site	Item	
	Ice (also dry ice if it is used to ship frozen samples) or ice substitute packs	
1 box	1-gal heavy-duty resealable (e.g., zipper-type closure) plastic bags	
1-box	30-gal plastic garbage bags for lining shipping containers	
1 roll	Clear tape for sealing shipping containers	
2 pkg.	Clear tape strips for covering labels	
4 rolls	Plastic electrical tape	
3	Insulated shipping containers for samples	
1	Portable freezer, cooler with dry ice, or cooler with bags of ice (or substitute ice packs) to store frozen samples (special containers may be needed if dry ice is used)	
2	Containers and absorbent material suitable to transport and/or ship samples preserved in formalin or ethanol	
6	Shipping airbills and adhesive plastic sleeves	
1 roll	Aluminum foil for periphyton and fish tissue contaminant samples	

SITE VERIFICATION AND SAMPLING REACH LAYOUT

Number per site	Item	
1	GPS receiver and operating manual	
	Extra batteries for GPS receiver	
1	Surveyor's telescoping leveling rod (round profile, metric scale, 7.5 m extended)	
1	50-m fiberglass measuring tape with reel	
2 rolls	Surveyor's flagging tape (2 colors)	
1 (optional)	Waterproof camera and film (or digital camera)	

WATER CHEMISTRY

Number per site	Item	
1 (optional)	Dissolved oxygen/Conductivity/Temperature meter with probe and operating manual	
1 (optional)	DO repair kit with additional membranes and probe filling solution	
1 (optional)	Conductivity meter, probe, and operating manual (if not integrated with DO/Temp meter	
4-8 (Optional)	Extra batteries for dissolved oxygen and conductivity meters	
1 (optional)	500-mL plastic bottle of conductivity QCCS labeled "Rinse" (in plastic bag)	
1 (optional)	500-mL plastic bottle of conductivity QCCS labeled "Test" (in plastic bag)	
1 (optional)	500-mL plastic bottle of deionized water to store conductivity probe	
1	Field thermometer	
1	500 mL plastic beaker with handle (in clean plastic bag)	
1	4-L cubitainer	
2	60 mL plastic syringes	
1	½ gal. size plastic container with snap-on lid to hold filled syringes	
2	Syringe valves	

STREAM DISCHARGE

Number per site	Item	
1	Current velocity meter and probe, with operating manual (e.g. Marsh-McBirney Model 201, Swoffer Model 2100, or equivalent)	
1	Top-set wading rod (metric scale) for use with current velocity meter	
1 (optional)	Portable Weir with 60° "V" notch	
1 (optional)	Plastic sheeting to use with weir	
1	Plastic bucket (or similar container) with volume graduations	
1	Stopwatch	
1	Neutrally buoyant object (e.g., wiffle-type plastic golf ball, orange, small rubber ball, stick, bobber)	

PHYSICAL HABITAT

Number per site	Item	
1	Fisherman's vest with lots of pockets and snap fittings (or fanny pack with pockets).	
1	50-m tape measure	
1	Clinometer with percent and degree scales. Alternate instruments (hand level, laser level, surveyor's level, or roofer's level [water tube]) can be used for slope measurements	
1	Lightweight telescoping camera tripod, (necessary only if slope measurements are being determined by only one person)	
1	½-inch diameter PVC pipe, 2-3 m long, each marked at the same height (for use in slope determinations involving two persons)	
1	Spherical **convex** canopy densiometer, modified with taped "V"	
1	Bearing compass (backpacking type)	
1	Meter stick. Alternatively, a short (1-2 m) rod or pole (e.g., a ski pole or wooden shovel handle) with cm markings for thalweg measurements	

PERIPHYTON

Number per site	Item	
1	Large funnel (15-20 cm diameter)	
1	12-cm^2 area delimiter (3.8 cm diameter pipe, 3 cm tall)	
1	Stiff-bristle toothbrush with handle bent at 90° angle	
1	1-L wash bottle for stream water	
1	1-L wash bottle containing deionized water	
1	500-mL plastic bottle (marked in 50-mL increments) labeled *PERIPHYTON* for composite sample	
1 (optional)	1-L plastic bottle (marked in 250-mL increments) labeled *TARGETED HABITAT PERIPHYTON* for targeted habitat composite sample	
1	35-60 mL plastic syringe (catheter-tip)	
3 (plus 1 optional)	50-mL screw-top centrifuge tubes (or similar sample vials). *Add an additional tube if collecting the optional targeted habitat periphyton sample.*	
1 box	Glass-fiber filters for chlorophyll and biomass samples	
1 pair	Forceps for filter handling.	
1	25-mL or 50-mL graduated cylinder (or use a 50-mL centrifuge tube with volume markings)	
1	Filtration unit, including filter funnel, cap, filter holder, and receiving chamber	
1	Hand-operated vacuum pump with length of flexible plastic vacuum tubing	
1	Small syringe or bulb pipette for dispensing formalin	

BENTHIC MACROINVERTEBRATES

Number per site	Item	
1	Modified kick net (500 µm mesh) and 4-ft handle	
1	Spare collection bucket for the kick net sampler (or an extra sampler)	
2	Buckets with screw-top lids, plastic, 8- to 10-qt capacity, labeled *REACHWIDE* and *TARGETED RIFFLE*	
1	Sieve, U.S. Std. No. 35 (500 µm mesh), or sieve bucket with 500-µm mesh openings	
2 pair ea.	Watchmakers' and curved tip forceps	
1	Small spatula, spoon, or scoop to transfer sample	
1	Funnel, with large bore spout	
4 to 6 ea.	Sample jars, HDPE plastic with leakproof screw caps, 500-mL and 1-L capacity, suitable for use with ethanol	
1 pkg.	Kim wipes in small self-sealing plastic bag	
1	Screw-top pail with absorbent material to transport preserved samples	

AQUATIC VERTEBRATES AND FISH TISSUE CONTAMINANTS

Number per site	Item	
1	Gasoline or battery-powered backpack electrofishing unit with electrode wand	
	Extra battery	
4 pr	heavy-duty insulated rubber "Linesmen" gloves for electrofishing	
2	Long-handled dip nets (0.6 cm mesh) with insulated handles	
1	Minnow seine (2m × 1.25 m, 0.6 cm mesh) with brailles	
1	Large seine (3 m × 2 m, 0.6 cm mesh) with brailles	
4	Collapsible buckets for holding and processing aquatic vertebrates	
1 set	Taxonomic reference books and keys for fishes and amphibians of the region	
1-2	Fish measuring board	
5-20	Small nylon mesh bags or nylon stockings for holding voucher specimens	
1	Jackknife for preparing larger voucher specimens for preservation	
1 ea.	1 and 2-L HDPE plastic jars with leakproof screw-top caps for voucher samples	
4	carbon dioxide tablets (Alka-Seltzer® or equivalent).	
1	Cooler to hold preserved voucher sample jars	
2	Aquarium nets (large and small)	

APPENDIX B
CHANGES AND MODIFICATIONS TO EMAP-SURFACE WATERS FIELD PROCEDURES

This section describes the changes made in EMAP field procedures for EMAP-W from those previously published for EMAP-Surface Waters (Lazorchak et al. 1998a). It also documents the minor modifications and clarifications made during the course of EMAP-W. These modifications were based in large part to comments received from EMAP-W field crews after every field season. This section is organized to follow the sequence of sections in the main body of this document:

B.1 BASE LOCATION ACTIVITIES B-2

B.2 INITIAL SITE PROCEDURES B-2

B.3 WATER CHEMISTRY B-3

B.4 DISCHARGE B-5

B.5 PHYSICAL HABITAT CHARACTERIZATION AND INVASIVE RIPARIAN PLANTS B-5

B.6 PERIPHYTON B-7

B.7 BENTHIC MACROINVERTEBRATES B-7

B.8 AQUATIC VERTEBRATES B-8

B.9 FISH TISSUE CONTAMINANTS B-9

B.10 RAPID HABITAT AND GENERAL VISUAL ASSESSMENTS B-10

B.11 LITERATURE CITED B-11

B.1 BASE LOCATION ACTIVITIES

Modifications to base location procedures (Section 3) described in the previous EMAP-SW field operations manual for wadeable streams (Klemm et al. 1998a) are summarized in Table B-1. Conductivity pens were not used in EMAP-W. Sediment samples for metabolism and sediment toxicity were not collected for EMAP-W. Performance evaluation procedures for field meters were modified to reflect updated instrumentation.

Modifications implemented during EMAP-W are also summarized in Table B-1. Field measurements of conductivity and dissolved oxygen became optional (2001), and the frequency of inspection and evaluation of field meters was reduced. All periphyton samples were treated as "unpreserved" samples for packing and shipment (2002). Guidelines for how much ice (or ice substitutes) to include in sample shipments, a more formalized procedure for filing status reports after each site visit, and additional information regarding the packing of preserved samples were also provided (2002).

For the final revision of this manual, updated material regarding cleaning and disinfecting equipment against the spread of whirling disease, New Zealand mud snail, and amphibian chytrid fungus was added, along with information regarding alternative procedures for shipping preserved samples.

B.2 INITIAL SITE PROCEDURES

Table B-2 summarizes changes to initial site procedures (Section 4) from those published previously for EMAP-SW by Herlihy (1998a), and modifications made during EMAP-W. Changes from Herlihy (1998a) include providing guidance for sampling streams that are partially wadeable, and for wide streams with a braided channel pattern. During EMAP-W, field data were no longer collected at stream sites having completely dry reaches. The field data form was modified for use with streams that were either determined to be nontarget before a field visit, or that were nontarget when visited. Additional criteria for determining nontarget sites were added, and clarifications were made to (1) procedures for sites having *interrupted flow* at the time of sampling, or which were dry at the time of the sampling visit, and 2) instructions for adjusting the initial support reach about the X-site.

TABLE B-1. SUMMARY OF CHANGES IN BASE LOCATION ACTIVITIES FOR THE WESTERN PILOT STUDY

Changes from Klemm et al. (1998a):
1. Removed reference to conductivity pens.
2. Removed procedures and information related to sediment metabolism and sediment toxicity sampling.
3. Performance evaluation procedures for field instrumentation were modified or added.
4. Added procedures for preparing dangerous goods samples for shipment.
5. Included cleaning procedures and solutions to prevent transfer of whirling disease spores between streams.
Changes from EMAP-Western Pilot Study Year 2000 activities:
1. The frequency of performance evaluation checks for field conductivity and dissolved oxygen meters was reduced from before each stream site to before and after the field season.
2. The use of ice substitute packs whenever possible to ship samples was strongly recommended to avoid problems associated with melted ice during shipment.
Changes from EMAP-Western Pilot Study Year 2001 activities:
1. Emphasized the use of more ice in shipments.
2. Revised shipping guidelines to treat periphyton ID sample the same as unpreserved samples.
3. Provided procedure for filing status reports after each site visit.
4. Provided additional detail regarding packaging and transport of preserved samples.
Changes from EMAP-Western Pilot Study Year 2002 activities:
1. Tracking form was reformatted to be easier to fill out and more compatible with optical scanner technology.

B.3 WATER CHEMISTRY

Changes in procedures for collecting water chemistry samples and field data (Section 5) from Herlihy (1998b) and modifications made during EMAP-W are summarized in Table B-3. The minimum volume of the bulk water sample required was reduced from 4 L to approximately 3 L, and procedures for measuring *in situ* DO and conductivity using a combination meter were included. In 2001, field measurements of dissolved oxygen and conductivity became optional, the frequency of QCCS checks of the conductivity meter was reduced, and the time when field measurements and samples were collected was recorded.

TABLE B-2. SUMMARY OF CHANGES IN INITIAL SITE PROCEDURES FOR THE WESTERN PILOT STUDY

Changes from Herlihy (1998a)

1. Developed guidance for sampling streams that are partially wadeable.
2. Developed guidance for how to sample streams that have a braided channel pattern.

Changes from Year 2000 Western Pilot Study Activities

1. Field data are no longer collected at sites where the entire sampling reach is dry when visited. They are now classified as non-sampleable.
2. The field data form was revised to deal more clearly with sites that are determined to be nontarget either before a field visit or at the time of the visit, including those that are temporarily inaccessible and can be visited again in a future year.
3. The field data form was revised so that site coordinates could be recorded in degree-minute-second (DMS) or decimal degree (DD) format. This was done to accommodate different types of GPS units or other data recording requirements of EMAP-W participants.

Changes from Year 2001 Western Pilot Study Activities

1. Included criteria for determining *nontarget canals* that are not sampled.
2. Clarified the determination for dry sites when visited.
3. Clarified instructions for sites with *interrupted flow* in regards to "sliding" the stream reach.
4. Included instructions for "sliding" the reach when access is denied to a portion of the reach.

TABLE B-3. SUMMARY OF CHANGES IN WATER CHEMISTRY PROCEDURES FOR THE WESTERN PILOT STUDY

Changes from Herlihy (1998b)

1. The minimum volume of the bulk water sample was reduced from 4-L to 3-L.
2. Procedures for using combination oxygen/conductivity/temperature meters were included.

Changes from Year 2000 Western Pilot Study Activities

1. Dissolved oxygen and conductivity measurements became optional.
2. The frequency of performance evaluation checks for field conductivity meters was reduced from before each field measurement to less frequent checks at base sites or home offices/ laboratories. If used, meters should be subjected to QCCS checks at a minimum frequency of before and after the field season.
3. The field form where DO and temperature measurements are recorded was eliminated. Data were recorded on the channel constraint form.
4. If field measurements are taken, the time of the measurements was recorded on the field data form.

B.4 DISCHARGE

Procedures for determining discharge (Section 6) were essentially unchanged from those previously published for EMAP-SW (Kaufmann 1998). In 2001, the field data forms were modified to allow a calculated value for discharge to be recorded (a fourth procedure), and to provide the option to record data for more than 20 intervals (using a supplemental form).

B.5 PHYSICAL HABITAT CHARACTERIZATION AND INVASIVE RIPARIAN PLANTS

Changes to various procedures for characterizing physical habitat (Section 7) previously published for EMAP-SW (Kaufmann and Robison 1998) are summarized in Table B-4. Four procedures (substrate particle size, instream fish cover, human influence, and thalweg habitat classification) are modified slightly from previous versions. The number of particles to be included in the systematic pebble count was increased (from 55 particles to 105) to provide improved precision of substrate characterizations such as %fines. To obtain the additional particles, 10 *supplemental* cross-sections are located midway between successive *regular* transects. Procedures for locating and estimating the size of particles on each cross-section remain unchanged, for regular and supplemental cross-sections, except that only the substrate size class and the wetted width data are recorded at the 10 supplemental cross-sections. Logistically, the supplemental substrate cross-section procedures are accomplished as part of the thalweg profile that is undertaken between regular transects (Section 7.4.1). However, the details of the actual measurements and observations are described in Section 7.5.2. The instream fish cover (Section 7.5.6) and human influence procedures (Section 7.5.7) now include additional or modified features. The thalweg habitat classification (Section 7.4.1) now includes the tallying of presence/absence of off-channel backwater habitats, (e.g., sloughs, alcoves, backwater pools). Backwater pools are included in this tally, but if they are the dominant channel habitat classification, they are also identified by a channel unit classification, as in previous versions of this field procedure.

Four new procedures are included for EMAP-W. The first (Section 7.5.8) is added to provide additional data on the size and proximity of large, old riparian trees. The second (Section 7.6.1), is added to classify the general degree of geomorphic channel constraint. This is an overall assessment of reach characteristics that is done after completing the thalweg profile and other measurements at the 11 cross-section transects. The third

TABLE B-4. SUMMARY OF CHANGES IN PHYSICAL HABITAT AND INVASIVE RIPARIAN PLANT PROCEDURES FOR THE WESTERN PILOT STUDY

Changes from Kaufmann and Robison (1998):

1. *Substrate*: The systematic pebble count is augmented from 55 particles (5 particles in each of 11 cross-sections) to 105 particles (5 particles in each of 21 cross-sections). Ten additional cross-sections are located midway between each regular transects. Only the substrate size class and the wetted width data are recorded at each supplemental cross-section.
2. *Instream Fish Cover*: Fish concealment features now include in-channel live trees or roots.
3. *Human Influence*: The human influence category *PAVEMENT* is modified to include cleared barren areas and renamed *PAVEMENT/CLEARED LOT*.
4. *Riparian "Legacy" Trees*: New protocol to obtain information on the size and proximity of large, old riparian trees.
5. *Channel Constraint*: New protocol to classify the general degree of geomorphic channel constraint. This is an overall assessment of reach characteristics that is done after completing the thalweg profile and other measurements at the 11 Cross-section Transects.
6. *Debris torrents*: New protocol to identify evidence of major floods or debris torrents (*lahars*). This is an overall assessment for the reach as a whole, and is done after completing the other measurements.
7. *Invasive Riparian Plants*: New protocol to obtain information on the occurrence of nonnative invasive riparian tree, shrub and grass species (presented in Section 8).

Modifications from Year 2000 Western Pilot Study Activities:

1. *Dry Streams*: Physical habitat data are no longer collected at streams reaches that are completely dry at the time of the field visit.
2. *Off-Channel Backwater Habitat*: The thalweg habitat classification now includes the tallying of presence/absence of off-channel backwater habitats, (e.g., sloughs, alcoves, backwater pools). If a backwater pool dominates the main channel habitat, *PB* is also entered as the channel unit classification code, as in previous versions of this field protocol.
3. *Riparian "Legacy" Trees*: Additional details regarding these procedures are included.
4. *Channel Constraint*: Additional detail regarding procedure is included; the number of constraint classes is reduced.
5. *Invasive Riparian Plants*: Additional details regarding these procedures are included. Target species of nonnative invasive tree, shrub and grass species is modified for some areas of the western U.S.

Modifications from Year 2001 Western Pilot Study Activities:

1. *Bank Characteristics*: Revised section dealing with determining bankfull and incision heights, adding a figure showing examples.
2. *Substrate*: The *BOULDER* class is divided into *Large* (> 1m median diameter) and *Small* (0.25 to 1m median diameter). Added a separate class for concrete.
3. *Riparian "Legacy" Trees*: Selection criteria for potential legacy trees are included.

Modifications from Year 2002 Western Pilot Study Activities:

1. *Slope and Bearing*: Form modified for use with other instruments/methods besides a clinometer.
2. *Riparian Vegetation Characterization*: Clarified that vegetation type in canopy and understory refers only to **woody** vegetation.

procedure (Section 7.6.2) identifies evidence of major floods or debris torrents (lahars). This is an overall assessment for the reach as a whole, and is done after completing the other measurements. The field form and procedures for assessing debris torrent evidence have been applied in Oregon and Washington research and R-EMAP surveys since 1994. The fourth procedure is used to determine the occurrence of nonnative invasive riparian tree, shrub and grass species (see Section 8).

Modifications to various habitat characterization procedures that occurred during EMAP-W are also summarized in Table B-4. In addition, the final revision of this manual includes additional figures showing a data form for a side channel transect and examples of bank angle measurements. A modified procedure table is now included that describes how to make slope and bearing measurements when starting at the most upstream transect and moving downstream.

B.6 PERIPHYTON

Changes to the periphyton sampling procedures (Section 9) from those previously published for EMAP-SW (Hill 1998), and modifications made during EMAP-W are summarized in Table B-5. Changes included increasing the number of transects where samples are collected, and reducing the number of composite samples from two to one per site. Also, preleached and preweighed glass-fiber filters were no longer required. An optional procedure to collect an additional ID/enumeration sample from a single "targeted" habitat type at each stream was developed in 2002. This procedure (see Section 9.4), is modified from one developed by Hawkins et al. (2001) for use in western streams and rivers, which in turn was adapted from the U.S. EPA Rapid Bioassessment Protocol (Stevenson and Bahls 1999). The objective of this additional sample was to allow for a comparison of the assessment of the periphyton assemblage based on the standard EMAP sampling procedure and the assessment based on a targeted habitat procedure used in a separate study focusing on reference sites in the western U.S.

B.7 BENTHIC MACROINVERTEBRATES

Changes to the benthic macroinvertebrate sampling procedures (Section 10) from those previously published for EMAP-SW (Klemm et al. 1998b), and modifications made during EMAP-W are summarized in Table B-6. Changes included using a different net and preparing a single composite sample for each site, as opposed to keeping flowing water

TABLE B-5. SUMMARY OF CHANGES IN PERIPHYTON PROCEDURES FOR THE WESTERN PILOT STUDY

Changes from Hill (1998)
1. The number of transects where periphyton samples are collected is increased from nine to eleven.
2. A single composite sample is prepared from the 11 cross-section samples, rather than preparing separating samples for flowing water (erosional) and slow water (depositional) transect samples.
3. The same glass-fiber filters are now used for both chlorophyll and biomass samples. Previously a pretreated and preweighed filter was provided to use for the biomass sample.
Modifications from Year 2000 EMAP-W Activities
1. Filters for chlorophyll and biomass are no longer wrapped in foil, but are folded and placed in separate 50-mL centrifuge tubes, which are labeled and then placed in a black plastic bag.
2. Samples for acid/alkaline phosphatase activity (APA) were no longer collected beginning in 2001.
Modifications from Year 2001 EMAP-W Activities
1. Clarified that sampling points should be located 1 m downstream of each transect to avoid disturbing substrates that are part of the physical habitat characterization.
2. Determined the preserved ID/Enumeration sample could be shipped with the rest of the periphyton samples.
Modifications from Year 2002 EMAP-W Activities
1. Volumes of subsample and preservative were adjusted for use with diluted formalin solutions.
2. Centrifuge tubes holding filters were wrapped in aluminum foil before storage to protect them from light.
3. Developed optional collection procedure to obtain an additional periphyton sample from a targeted habitat.

(*riffle*) and slack water (*pool*) samples separate. Modifications during EMAP-W were minor, involving clarifications to existing procedures.

B.8 AQUATIC VERTEBRATES

Changes to the aquatic vertebrate sampling procedures (Section 11) from those previously published for EMAP-SW (McCormick et al. 1998), and modifications made during EMAP-W are summarized in Table B-7. Changes included the specific inclusion of other aquatic vertebrates besides fish, eliminating recording different types of external anomalies and reducing the number of total length measurements to just the largest and smallest individual of each species. Most of the modifications focused on clarifications to

the vouchering procedures and recording information required by scientific collecting permits.

TABLE B-6. SUMMARY OF CHANGES TO BENTHIC MACROINVERTEBRATE PROCEDURES FOR THE WESTERN PILOT STUDY

Changes from Klemm et al. (1998)
1. Two types of samples are collected, a *Reachwide* sample and a *Targeted Riffle* sample. The *Reachwide* sample is collected from transects spaced evenly throughout the reach, as described in Klemm et al. (1998). The *Targeted Riffle* sample is collected from riffle areas only (i.e., if no riffle areas are present, the sample is not collected).
2. The number of kick samples in the *Reachwide* sample was increased from 9 (transects *B* through *J*) to 11 (Transects *A* through *K*). The number of kick samples in the *Targeted Riffle* sample is 8.
3. Each sample type is prepared as a single composite sample. For the *Reachwide* sample, all kick samples are combined into a single composite sample, replacing the *RIFFLE* composite and the *POOL* composite samples.
4. The sampling device was changed from a rectangular kick net to a D-Frame design. Mesh size decreased from 595 µm to 500 µm. The width of the net decreased from 18 in. to 12 in. (50 cm to 30 cm).
5. The area of each kick net sample was reduced from 0.5 m^2 to 0.09 m^2 (1 ft^2).
6. The time for each kick net sample was increased from 20 seconds to 30 seconds.
Modifications from EMAP-W Year 2000 Activities:
1. Clarified procedure for collecting at sampling points choked with vegetation.
2. Field form was modified to record the microhabitat type (pool, glide, riffle, rapid) for each *Reachwide* kick net sample.
Modifications from EMAP-W Year 2001 Activities:
1. Clarified that sampling points should be located 1 m downstream of each transect to avoid disturbing substrates that are part of the physical habitat characterization.
2. Emphasized accurate record keeping in terms of samples actually being collected and the correct number of jars for each sample recorded.
3. Provided approach for collecting samples located within beds of long, filamentous vegetation.
4. Clarified the identification of substrate types within a sampling quadrat.
5. Clarified option of waiting to preserve samples until arrival back at the vehicle.

B.9 FISH TISSUE CONTAMINANTS

Procedures for obtaining fish tissue samples (Section 12) are adapted from Yeardley et al. (1998). For EMAP-W, a priority list of target species for small fish was not developed because many smaller species are regionally endemic relative to the scale of EMAP-W. The expectation was that for a given site, the most common small fish species in the region would be collected and retained as the small fish sample. This species would probably be a cyprinid most places, and a sculpin in others, which matched up fairly well with the MAHA priority list (Yeardley et al. 1998). The target list for larger fish was modified to replace eastern species with western counterparts. In 2001, procedures of preparing, labeling, and

TABLE B-7. SUMMARY OF CHANGES IN AQUATIC VERTEBRATE PROCEDURES FOR THE WESTERN PILOT STUDY

Changes from McCormick and Hughes (1998)
1. Aquatic vertebrates collected from each *subreach* (i.e., between transects) were now tallied and recorded on separate field data forms.
2. Crayfish collected during aquatic vertebrate sampling were now counted and retained as part of the aquatic vertebrate sample.
3. Recording the occurrence of specific types of external anomalies was no longer required.
4. Determination of total lengths of 30 individual fish of each dominant species collected was no longer required.
Modifications from Year 2000 EMAP-W Activities
1. Aquatic vertebrates (and crayfish) were now tallied and recorded on a single data form for each reach; all subreaches where a species was collected were noted on the form.
2. Procedures for dealing with wide (>20 m) yet wadeable reaches were clarified and/or included.
Modifications from Year 2001 EMAP-W Activities
1. Procedures and field forms were revised to keep better track of sites where collection permits restricted or prohibited sampling for vertebrates, and to identify sites where all subreaches were not sampled.
2. The rationale for vouchering was clarified to provide additional detail regarding photographing voucher specimens.
Modifications from Year 2002 EMAP-W Activities
1. Field forms were revised to include conductivity and water temperature to facilitate collection permit reports.
2. Vouchering procedure was revised to emphasize that at least one voucher specimen of each species be retained (preserved or photographed) to allow for confirmation of range extensions.

tracking individual fish samples were clarified. In 2002, the use of more ice in shipments was emphasized.

B.10 RAPID HABITAT AND GENERAL VISUAL ASSESSMENTS

Changes in the rapid habitat assessment procedure (Barbour et al. 1999) from those published previously for EMAP-SW (Lazorchak et al. 1998b), and the original RBP procedures (Plafkin et al. 1989), involve additional assessment parameters for high gradient streams and a more appropriate set of parameters for low gradient streams, based on refinements from various applications across the country. Modifications during EMAP-W included making the visual habitat assessment an optional activity after the first year (2000). Modifications to the visual assessment procedure during EMAP-W included

adding a general description of weather conditions at a site and adding evidence of fire as a disturbance type.

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APPENDIX C
FIELD DATA FORMS

Two files containing the field data forms illustrated in this manual are provided on the CD-ROM:

17A_blank_stream_1_each.pdf:	Provides a single copy of each form.
17*B_blank_stream_all.pdf*:	Provides a complete set of blank forms (including multiple copies where required) that would be used at a site.

The files are in Adobe *portable document file* (pdf) format. Software to read and print these files (Adobe Acrobat Reader®) can be obtained free from *www.adobe.com*. Note that Acrobat Reader cannot be used to edit the forms. The files are formatted for double-sided printing.

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APPENDIX D
INVASIVE RIPARIAN PLANT IDENTIFICATION GUIDES

The following two files on the CD-ROM contains color images and associated information to identify the 12 plants selected as target species in EMAP-W in the field:

18A_Appendixd_Invasive_Riparian_Plant_ID_Guide_Low_Resolution_Version.pdf
18A_Appendixd_Invasive_Riparian_Plant_ID_Guide_Full_Resolution_Version.pdf

The first file contains low-resolution images, suitable for viewing on a computer screen. This file may also produce suitable hard copy output on many printers. The second file contains high-resolution images, and can be used if the if the low-resolution version does not produce a satisfactory hard copy. The files are in Adobe *portable document file* (pdf) format. Software to read and print this file (Adobe Acrobat Reader®) can be obtained free from *www.adobe.com*. Note that Acrobat Reader cannot be used to edit the forms. The files are formatted for double-sided printing.

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