APPENDIX A:

BABCOCK CHAIN-OF-CUSTODY, DRIVING DIRECTIONS, AND LABORATORY PROCEDURES
(Procedures provided electronically only)

E.S. Babcock & Sons, Inc.
6100 Quail Valley Court Riverside, CA 92507
(909) 653-3351 • FAX (909) 653-1662

Chain of Custody & Sample Information Record

Client: | Contact: | Phone No.

Project Name: _____
Project Location:

Turn Around Time: Routine | 3-5 Days | 48 Hours | 24 Hours (*Rushes Require Approval, Additional Charges May Apply)*

Sampler Information

Name: _____
Employer: _____
Signature: _____

ESB #	Sample ID	Date	Time	# of Containers & Preservatives: Unpreserved	H_2SO_4	HCl	HNO_3	$Na_2S_2O_3$	NaOH			Total # of Containers	Analysis Requested	Matrix	Notes
														DW = Drinking Water WW = Wastewater GW = Groundwater S = Soil SG = Sludge L = Liquid M = Miscellaneous	

Relinquished By (sign)	Print Name / Company	Date / Time	Received By (Sign)	Print Name / Company

(For Lab Use Only) — *Sample Integrity Upon Receipt*

Sample(s) Submitted on Ice? Yes No — Temperature
Custody Seal(s) Intact? Yes No N/A — ºC
Sample(s) Intact? Yes No

Lab No. _____

Page _____ of _____

Chain of Custody Form Instructions

Example filled-out Chain of Custody Forms are available in the monitoring location-specific binders at David Ortega's (951-955-4390) office. Additional, site specific instructions are provided for most CMP stations.

1. **Client**: **RCFC&WCD**

2. **Contact**: [name of monitoring program manager] **Rebekah Guill ~~Arlene Chun~~**

3. **Phone number**: The Contact's phone number **951-955-2901 ~~951-955-1330~~**

4. **Project Name**: SAR/SMR/WWR (note which one) or Complaint Response

5. **Project Location**: Brief description

6. **Turn Around Time**: Usually Routine.

7. **Sampler Name**: [name of lead sampler]

8. **Employer**: RCFC&WCD or the company name if other than a District employee collected the sample.

9. **Signature**: Signature of the lead sampler.

10. **Sample ID**: If at an existing MS4 monitoring station, note the station number here. If at a different location, use station number 823 for WWR complaints, 809 for SAR complaints, and 825 for SMR complaints. This number is used to enter the analysis into the database; **do not use other than the station number and a sequence number, if needed**. For example, the first of a sequence for a complaint sample collected in the Santa Ana Region would be labeled as "809-01".

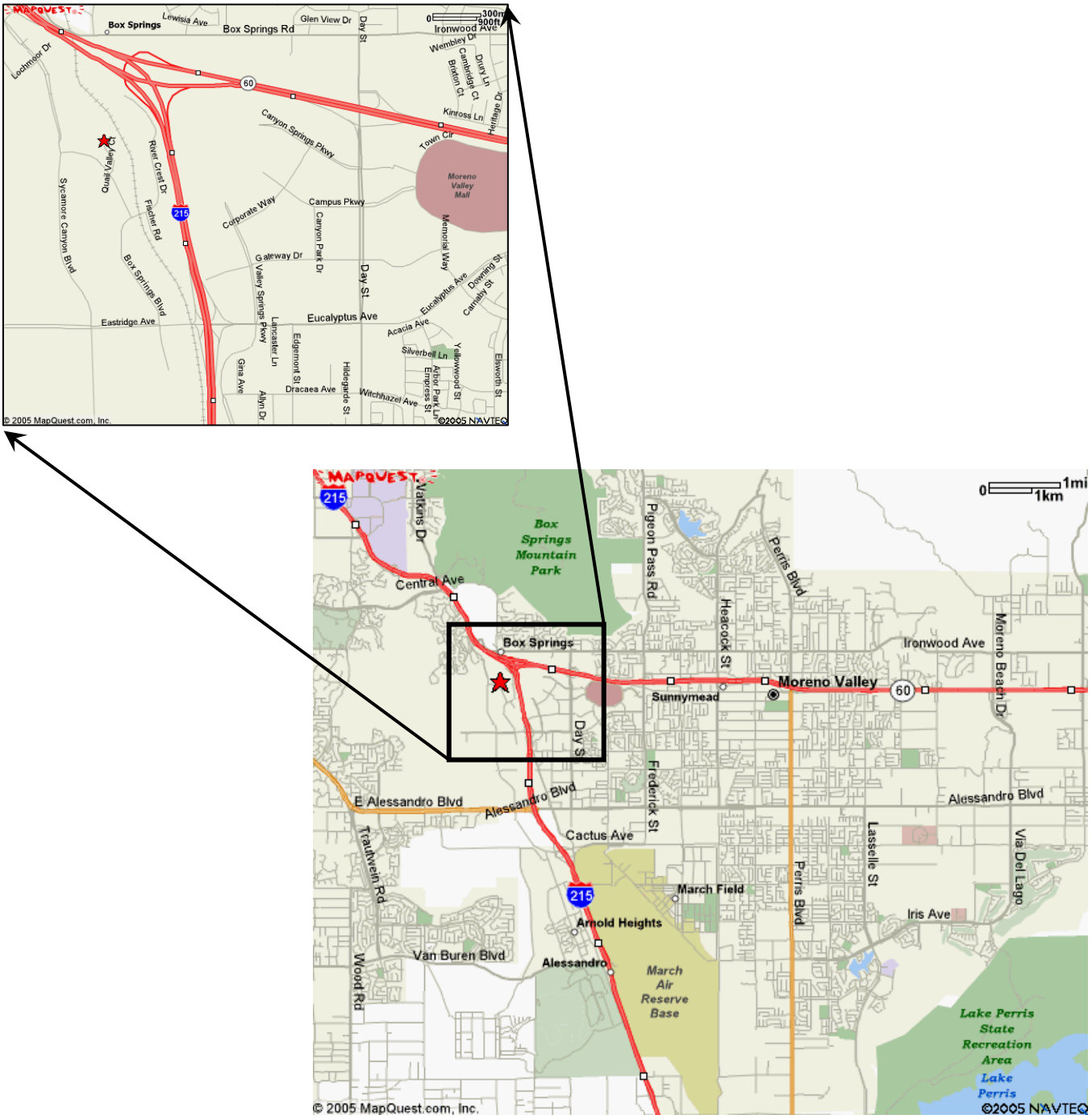
11. **# of Containers & Preservatives**: Write the number of bottles with a specific preservative under the preservative's column. Write the number of unpreserved bottles under the "Unpreserved" column. Write the total number of containers under the appropriate column as a check.

12. **Analysis Requested**: Indicate the pre-determined analyses for the samples or attach the specific watershed analyses sheet.

13. **Matrix**: Usually wastewater (WW) or soil (S)

14. **Relinquished By**: This area is important and must be filled out. When a person hands the sample to another person, the date and time the transfer took place and the signature of both parties involved in the transfer must be included. For example, when the samples are delivered to the lab, the District and lab staff people will sign and date the *Chain of Custody*. If someone outside of the District collects the sample, that person and you will be the first signatories.

Map to Babcock Laboratories



Driving directions to Babcock (taken from Mapquest)

From the South (e.g., Temecula):

Merge onto I-15 N	
Take I-215 N toward Riverside/San Bernardino	28.8 miles
Take the EUCALYPTUS AVE / EASTRIDGE AVE exit.	0.2 miles
Turn SLIGHT LEFT to take the EASTRIDGE AVE ramp.	<0.1 miles
Turn LEFT onto EUCALYPTUS AVE / EASTRIDGE AVE. Continue to follow EASTRIDGE AVE.	0.1 miles
Turn RIGHT onto BOX SPRINGS BLVD.	0.4 miles
Turn LEFT to stay on BOX SPRINGS BLVD.	0.1 miles
Turn RIGHT onto QUAIL VALLEY CT.	0.1 miles
End at **6100 Quail Valley Ct**	

From the West (e.g., Corona)

Take the CA-91 E toward SAN BERNARDINO	
Merge onto I-215 S / CA-60 E	4.2 miles
Take the BOX SPRINGS exit toward FAIR ISLE DR.	0.2 miles
Turn RIGHT onto BOX SPRINGS RD.	<0.1 miles
Turn LEFT onto SYCAMORE CANYON BLVD	0.5 miles
Turn LEFT onto BOX SPRINGS BLVD	0.2 miles
Turn LEFT onto QUAIL VALLEY CT.	0.1 miles
End at **6100 Quail Valley Ct**	

From the North (e.g., Norco)

Merge onto I-15 S toward SAN DIEGO	
Merge onto CA-60 E toward RIVERSIDE	16.5 miles
Take the BOX SPRINGS exit toward FAIR ISLE DR	0.2 miles
Turn RIGHT onto BOX SPRINGS RD	<0.1 miles
Turn LEFT onto SYCAMORE CANYON BLVD	0.5 miles
Turn LEFT onto BOX SPRINGS BLVD	0.2 miles
Turn LEFT onto QUAIL VALLEY CT.	0.1 miles
End at **6100 Quail Valley Ct**	

From the East (e.g., Banning)

Merge onto I-10 W toward LOS ANGELES	
Merge onto CA-60 W via the exit on the LEFT toward RIVERSIDE	18.4 miles
Take the BOX SPRINGS exit toward FAIR ISLE DR	0.1 miles
Turn LEFT onto BOX SPRINGS RD.	0.1 miles
Turn LEFT onto SYCAMORE CANYON BLVD.	0.5 miles
Turn LEFT onto BOX SPRINGS BLVD	0.2 miles
Turn LEFT onto QUAIL VALLEY CT	0.1 miles
End at **6100 Quail Valley Ct**	

APPENDIX B:

FIELD DATASHEET

WATER POLLUTION PREVENTION
FUNDED BY THE CITIES AND COUNTY OF RIVERSIDE

FIELD DATA SHEET

Rev 07/16/2013

STATION ID: Click and choose Station ID **SAMPLE DATE** (MM/DD/YYYY): _____

STATION NAME: Click and choose Station Name WATERSHED: ☐SAR ☐SMR ☐WWR

LOCATION (if not standard site):_____ Within: ☐Unincorp. or ☐City of_____

CONVEYANCE TYPE:_____ ☐Receiving Water ☐Within IAH

GPS INFO: Lat _____ Long _____ GPS Unit: _____ ☐Outfall, Owner:_____

PRINTED NAMES of Sampling Team:_____ ☐Other:_____

SIGNATURE of lead sampler: _____ Sampling AGENCY: _____

SAMPLE INFORMATION

☐ **VISITED, NOT SAMPLED (TIME:_____)**

EVENT CATEGORY:

☐ Wet Weather (Storm) OR
☐ Dry Weather
☐1st ☐2nd ☐3rd ☐4th
☐ Recon, IC/ID, or Complaint
☐ Other _____

No. of Samples: _____ **SAMPLE ID(s): 1213-XX-XXXX-01, 1213-XX-XXXX-XX**

STREAM FLOW:

Dry: ☐Yes ☐No Ponded: ☐Yes ☐No
Rising Groundwater: ☐Yes ☐No
Connects to Surface Receiving Water ☐Yes ☐No
Dry weather event u/s influence: ☐Yes ☐No

TYPE (check all that apply):

☐ **Grab** [SAMPLE TIME: _____]
☐ **Composite** TIME: see below
☐ Travel Blank ☐ Field Blank ☐ Field DUP
☐ Other:

FIELD PARAMETERS

Time Measured: _____

	Result	Units	Meter	Calibration Date
☐ **Water Temp**				
☐ **pH**				
☐ **EC**				
☐ **Turbidity**				
☐ **DO**				
☐				
☐				
☐				

FLOW ESTIMATION:

☐ USGS Gauge height/stage _____ ft Q (cfs) = _____

[Gauge Name/No.: _____]

☐ Calculation by visual measurement: Q (cfs) = _____

= [Coef(1,$^2/_3$,_____)]*[depth_____ft]*[width_____ft]*[vel_____fps]

Circular pipe: [vel_____fps][depth_____ft][width_____ft][R=_____ft]

SITE CONDITIONS

PRECIPITATION:

NOW: ☐ None ☐ Fog ☐ Drizzle ☐ Sprinkle ☐ Rain ☐ Hail/Snow

Last 24 hrs: ☐None ☐ <1" ☐ >1" ☐ _____

Last 72 hrs: ☐None ☐ <1" ☐ >1" ☐ _____

ODOR: ☐ None ☐ Sulfides ☐ Sewage ☐ Smoke ☐ Petroleum ☐ Other: _____

☐ Floatables_____ ☐ Settleables _____

☐ Vegetation_____ ☐ Staining _____

COLOR: ☐ Colorless ☐ Green ☐ Yellow ☐ Brown ☐ Other

CLARITY: ☐ Clear (see bottom) ☐ Cloudy ☐ Murky

Sheen Present: ☐ Yes ☐ No

TRASH: ☐ Yes ☐ No

From: ☐ Flows ☐ Dumping ☐Other:_____

COMPOSITE Samples: Auto/Grab, Flow/Time Weighted, ____Hrs

	Time	H(in.)	Flow(cfs)	%		Time	H(in.)	Flow(cfs)	%
1					13				
2					14				
3					15				
4					16				
5					17				
6					18				
7					19				
8					20				
9					21				
10					22				
11					23				
12					24				

Observations/Notes ☐ Photograph(s)

☐ **Additional sample(s) taken u/s, d/s** (circle one or both and complete separate FDS(s)) **at:**

- Field Data Sheet (FDS) Instructions -

Station ID – Enter station number and sampling date. **NOTE: These Station ID numbers must match bottle labels & COCs.** Remaining fields self-explanatory. Major land uses can be estimated in the field or back in the office.

Sample Information –Self-explanatory.

- Bottles must be stored on ice from when the sample is collected to when they reach the laboratory.
- Make sure the bottles are properly sealed (caps on snug).
- **Note all Sample IDs taken at this station.** The total number of containers refers to those in the primary sample set **ONLY** (XXXX-XX-XXX-**01**). **If field DUPLICATES are taken, a 2nd FDS is required as field duplicates require their own set of field parameters.** The FDS of the duplicate sample would reflect the number of bottles in the duplicate set (Sample ID XXXX-XX-XXX-**02**).
- A grab sample is a single sample collected at one time. Also indicate whether toxicity or bacteria samples were taken, if applicable. **This must match bottle labels and chain of custody.**
- A composite sample is a series of grab samples collected over a period of time. The separate samples are combined into a single flow-proportioned sample prior to analysis, usually at the laboratory. Ensure that the sample times, flow heights and flow estimation columns are filled out at the bottom of the page.

Field Parameters – Note the time at which field parameters were measured.
Field parameters must be measured, **bolded items** are the minimum. **Always include units!** Enter name (not type) of field meter used and its last calibration date in corresponding field.

Flow Estimation

- If there is a USGS gauge, enter the gauge height. Discharge can be retrieved when back at the office. Note the name or gauge number in space provided.
- If needed, note the channel shape parameters so flow can be calculated. Flow speed can be measured with a leaf or stick and a stopwatch. Do not use trash as a floatable in estimating velocity!
  - Coef = 1 (straight wall/rectangular channel), $^2/_3$ (trapezoidal channel), ½ (triangular channel), 0.8 (rough bottom), 0.9 (smooth bottom).
  - Width: Use top width of flow
- ***Flow in circular pipes.*** Record velocity (vel, fps), depth of flow @ center of pipe (ft), top width of flow (ft), and Radius of pipe (R, ft). Flow will be calculated at the office using Table .

Site Conditions – Self-explanatory.
The date(s) of last rain/storm may be filled out at the office.
Use the categories below to assist in describing the flow and the surroundings and stream flow:

Odor: None, Musty, Sewage, Rotten Egg, Sour Milk, Fishy, Petroleum, Ammonia, Chlorine, Decaying Organisms, Other (describe)

Floatables: None, Oil, Foam, Animal Waste, Green Waste, Algae, Food, Paper, Plastic, Other (describe). Include estimated percentage and character of the floatables in observations field at bottom of sheet. (e.g., foam with approx. 1" high bubbles, trash is approx. 75% paper, light sheen oil, etc.)

Settleables: None, Minor, Major, Other (describe)

Vegetation: None, Normal, Excessive, Dying, Color, Other (describe)

Staining: None, Salt, Clay, Oil, Rust, Microbes, Other (describe)

Color: None, Yellow, Brown, Grey, Red, Green, Amber, Blue, Olive Brown, Other (describe)

Sheen: Gasoline, Oil or other fuel

SMR (Only) Action Levels:
Notify Monitoring Manager if the following are exceeded.

Parameter	NAL (Dry)	SAL (Wet)
Turbidity	20 NTU	126 NTU
pH	6.5 – 8.5	--
DO	< 5.0 mg/L (Warm Waters)	--
	< 6.0 mg/L (Cold Waters)	--

Composite Samples (cross out if not used)

Circle method(s): Auto or Grab, Flow or Time and provide duration (#hrs). Note the time and the flow for each sample collected. If available at the sampling site, note the height reading (H, feet) at the staff gauge. The composited sample will be flow-proportioned in the laboratory.

Notes/Observations – Check box if photographs were taken
Note any observations regarding the sampling location, *including any sampling procedural variances* that occurred (i.e., sampling upstream, instrument malfunction, wildlife in area, safety, etc.). Make note of other conditions: *Sediments* [None, Normal, Excessive, Other (describe)], *Structural* [Normal, Cracking, Spauling, Other (describe)], *Biological* [Algal Bloom, Larvae, Crawfish, Frogs, Fish, Water Fowl, Other (describe)].

If additional samples are to be taken at another location, check the box and circle one or both u/s and d/s and provide the Station ID(s) for the station(s) and complete a separate FDS for each station. *Example: SMR MS4 Outfalls IAH sampling requires sampling at the Outfall site and at both the upstream and downstream proximate Receiving Water sites. See below:*

EXAMPLE: ☒ Additional sample(s) taken (u/s, d/s) at: 902RW4020, 902RW4021

APPENDIX C:

MONITORING LOCATIONS BY PERMIT

Table C-1 Monitoring Locations by Permit

Watershed	Station Name	Station Number	Station Type	Latitude	Longitude
Santa Ana	Corona Storm Drain	801CRN040	Outfall	33°53'7.18"	-117°34'7.62"
Santa Ana	Sunnymead Channel	802SNY316	Outfall	33°55' 3.47"	-117°14' 36.43"
Santa Ana	Hemet Channel	802HMT318	Outfall	33°44' 4.71"	-117°00'22.23"
Santa Ana	Magnolia Center Storm Drain	801MAG364	Outfall	33°57'56.60"	-117°24'56.08"
Santa Ana	University Wash	801UNV702	Outfall	33°59' 49.94"	-117°22' 22.07"
Santa Ana	North Norco Channel	801NNR707	Outfall	33°54' 26.93"	-117°34' 59.80"
Santa Ana	Perris Line J	802PLJ752	Outfall	33°48'16.45"	-117°12'31.54"
Santa Ana	Temescal Channel at Main	801TMS746	Receiving Water (Wet)	33° 53' 22.27"N	117° 33' 9.74"W
Santa Ana	Perris Valley Channel at Nuevo	802NVO325	Receiving Water (Wet & Dry)/ Bioassessment	33° 48' 4.29"N	117° 12' 2.14"W
Santa Ana	Santa Ana River at Highgrove	801AHG857	Receiving Water (Dry)/ Bioassessment	34° 01' 3.06"N	117° 22' 7.37"W
Santa Ana	Temescal Creek near Temescal Canyon Road	801TSC887	Bioassessment	33° 45' 6.84"N	117° 27' 2.85"W
Santa Margarita	Adobe Creek	902ADB848	Reference Station (Dry Weather)	33°30'48.10" N	117°16'04.40" W
Santa Margarita	Cole Creek	902COL188	Alternate Reference Station (Wet Weather)	33°31'41.75" N	117°16'18.44" W
Santa Margarita	Lower Temecula Creek	902LTC777	Receiving Water	33°28’26.4” N	117°07’46.1” W
Santa Margarita	Lower Murrieta Creek	902LMC778	Receiving Water	33°28’47.0” N	117°08’34.8” W
Santa Margarita	Upper Santa Margarita River	902USM828	Receiving Water - Stream Assessment	33° 28' 19.2" N	117° 08' 42.6" W
Santa Margarita	Redhawk Channel	902RDH768	Receiving Water - Stream Assessment	33° 28’ 34’6” N	117° 05’ 40.8” W
Santa Margarita	PENDING Recon of Cottonwood Creek	PENDING	Reference - Stream Assessment	PENDING	PENDING

Santa Margarita	Murieta Cr u/s Line E	902RW4020	Upstream RW (IAH)	33°32'48.94"	-117°12'52.54"
Santa Margarita	Line E Murrieta Cr MDP at Murrieta Cr	902MS4021	Outfall (IAH)	33° 32’ 48.5”	-117°12’49.6”
Santa Margarita	Murieta Cr d/s Line E	902RW4022	Downstream RW (IAH)	33°32'46.28"	-117°12'49.81"
Santa Margarita	Warm Springs Creek d/s of M.H.S. Rd. u/s of outlet	902RW4033	Upstream RW (IAH)	33°32'50.68"	-117°10'17.46"
Santa Margarita	RCP outlet to Warm Springs Creek d/s of M.H.S. Rd.	902MS4034	Outfall (IAH)	33:32:51.10	-117:10:18.79
Santa Margarita	Warm Springs Creek d/s of M.H.S. Rd. d/s of outlet	902RW4035	Downstream RW (IAH)	33°32'50.24"	-117°10'17.91"
Santa Margarita	Temecula Creek u/s of outlet at Butterfield Stage Rd	902RW4143	Upstream RW (IAH)	33°28'59.16"	-117°4'37.48"
Santa Margarita	Outlet into Temecula Creek @ Butterfield Stage Rd.	902MS4144	Outfall (IAH)	33:28:58.4	-117:04:37.9
Santa Margarita	Temecula Creek d/s of outlet at Butterfield Stage Rd	902RW4145	Downstream RW (IAH)	33°28'59.50"	-117°4'43.24"
Santa Margarita	Outlet to Long Canyon u/s of Commerce Ctr. Dr.	902MS4192	Outfall	33:30:52.52	-117:09:41.42
Santa Margarita	Long Canyon u/s of Commerce Ctr. Dr. d/s of outlet	902RW4193	Downstream RW	33°30'52.16"	-117°9'41.83"
Santa Margarita	Wildomar Channel u/s of Union St	902MS4243	Outfall	33:36:32.29	-117:17:25.13
Santa Margarita	Wildomar Channel d/s of Union St	902RW4244	Downstream RW	33°36'32.15"	-117°17'18.04"
Santa Margarita	Browning St Strom Drain at Santa Gertrudis	902MS4263	Outfall	33:34:05.1	-117:06:37.5
Santa Margarita	Santa Gertrudis d/s Browning St SD	902RW4264	Downstream RW	33°34'9.18"	-117°6'43.48"

Santa Margarita	Outlet to Empire Creek @ Butterfield Stage Rd	902MS4289	Outfall	33:31:08.3	-117:05:39.4
Santa Margarita	Empire Cr d/s outlet at Butterfield St Rd	902RW4290	Downstream RW	33° 31' 7.49"	-117° 5' 40.51"
Santa Margarita	Euclid loop storm drain at warm springs ch	902MS4309	Outfall	33:36:18.5	-117:06:48.4
Santa Margarita	Warm Spg Ch d/s Euclid Lp Storm Drain	902RW4310	Downstream RW	33° 36' 14.86"	-117° 6' 53.75"
White Water	Ramsey Street	719RMS782	Storm Drain	33°48’35.0” N	116°51’31.5” W
White Water	Portola Avenue	719POR817	Outfall	33°44’16.8” N	116°22’24.6” W
White Water	Avenue 52 Outfall	719AVE785	Outfall	33°40’17.4” N	116°08’56.4” W
White Water	CVSC at Avenue 52 Bridge	719CVS884	Receiving Water	33°40’20.9” N	116°08’57.8” W
White Water	Upper White Water	719WWT813	Receiving Water	33°59’01.5” N	116°08’57.8” W

APPENDIX D:

CLEAN HANDS/ DIRTY HANDS SOP

▶ Recommendation: Clean-sampling techniques are recommended when collecting samples for analysis of trace-organic compounds and major inorganic elements, particularly when the target analyte could be subject to contamination from field or laboratory procedures at a level that could exceed data-quality requirements.

Table 4-2. Clean Hands/Dirty Hands techniques for water-quality sampling

- Clean Hands/Dirty Hands techniques require two or more people working together.
- At the field site, one person is designated as Clean Hands (*CH*) and a second person as Dirty Hands (*DH*). Although specific tasks are assigned at the start to *CH* or *DH*, some tasks overlap and can be handled by either, as long as the prescribed care is taken to prevent contaminating the sample.
- Both *CH* and *DH* wear appropriate disposable, powderless gloves during the entire sampling operation and change gloves frequently, usually with each change in task. (Wearing multiple layers of gloves allows rapid glove changes.) Gloves must be appropriate to withstand any acid, solvent, or other chemical substance that will be used or contacted.
- *CH* takes care of all operations involving equipment that contacts the sample; for example, *CH*
  - Handles the surface-water sampler bottle.
  - Handles the discharge end of the surface-water or ground-water sample tubing.
  - Transfers sample to churn or cone splitter.
  - Prepares a clean work space (inside vehicle).
  - Sets up processing and preservation chambers.
  - Places equipment inside chambers (for example, sample bottles, filtration and preservation equipment).
  - Works exclusively inside chambers during collection/processing and preservation.
  - Changes chamber covers, as needed.
  - Sets up field-cleaning equipment and cleans equipment.
- *DH* takes care of all operations involving contact with potential sources of contamination; for example, *DH*
  - Works exclusively exterior to processing and preservation chambers.
  - Prepares and operates sampling equipment, including pumps and discrete samplers, peristaltic pump switch, pump controller, manifold system.
  - Operates cranes, tripods, drill rigs, vehicles, or other support equipment.
  - Handles the compressor or other power supply for samplers.
  - Handles tools such as hammers, wrenches, keys, locks, and sample-flow manifolds.
  - Handles single or multiparameter instruments for field measurements.
  - Handles the churn carrier, including outer protective bags.
  - Handles stream-gaging or water-level equipment.
  - Sets up and calibrates field-measurement instruments.
  - Measures and records water levels and field measurements.

APPENDIX E:

SOPS FOR CONDUCTING FIELD MEASUREMENTS AND FIELD COLLECTION OF WATER AND BED SEDIMENT SAMPLES IN SWAMP

Marine Pollution Studies Laboratory – Department of Fish and Game (MPSL-DFG) Standard Operating Procedures (SOPs) for Conducting Field Measurements and Field Collections of Water and Bed Sediment Samples in the Surface Water Ambient Monitoring Program (SWAMP)

The SOPs below are for reference and information purposes only, the documents are not required by the Surface Water Ambient Monitoring Program (SWAMP). Please see the SWAMP Quality Assurance Management Plan (http://www.swrcb.ca.gov/swamp/qamp.html) for more information regarding SWAMP QA/QC requirements.

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Field Measurements

Field Data Sheets

Field data sheets are used to record field observations, probe measurements, and water and sediment chemistry sampling. Field data sheets are provided through the Marine Pollution Studies Laboratory website at:
http://mpsl.mlml.calstate.edu/swdwnlds.htm
Click on the *Field Data Sheets* for the most recent versions. There are guidelines provided below to standardize what is recorded on all data sheets and that should be helpful in completing each form. The Beaufort Scale (see at the end of this document) is also used for specifications and equivalent wind speeds for water conditions. The entries discussed below and on the field data sheets are recorded at each sampling site.

Notes to Standardize SWAMP Field Data Sheets (For in the field use)

Upon arrival at a sampling site, record visual observations on the appearance of the water and other information related to water quality and water use.

Key Reminders to identify samples:

1. **Sample Time** is the SAME for all samples (Water, Sediment, & Probe) taken at the sampling event. Use time of FIRST sample as it is important for the chain of custody (COC).
2. **Left Bank/Right Bank**
   Left bank is defined as the bank to the left of the observer when facing downstream, and the *right bank* is to the right of the observer when facing downstream

FIELD OBSERVATIONS: (each one of these observations has a *Comment* field in the database so use comment space on data sheet to add information about an observation if necessary)

1. **DOMINANT SUBSTRATE**: if possible; describe DOMINANT substrate type; use UNK if you cannot see the dominant substrate type
2. **WADEABILITY**: in general, is the water body being sampled wadeable to the average person AT the POINT of SAMPLE
3. **BEAUFORT SCALE**: use scale 0-12; refer to scales listed at the end of this document.
4. **WIND DIRECTION**: records the direction from which the wind is blowing
5. **PICTURES:** Digital photos are taken to help document the actual sampling site. The convention is to take photos facing DOWNSTREAM, overlooking the site. Right bank and left bank are thus defined in this downstream-facing direction. Document any discrepancies from this convention. Only one photo is necessary, if both, left and right

bank, fit into one frame. Record all photos in the field data sheet space to record picture numbers given by camera; be sure to rename accordingly back in the office. All photos should be renamed and saved with the StationCode_yyyy_mm_dd_uniquecode (e.g. 123ABC123_2007_07_01_BBDS).

6. **SITE ODOR**: Note if hydrogen sulfide odor, musty odor, sewage odor, etc. is in the sampling reach
7. **SKY CODE**: Note recent meteorological events that may have impacted water quality
8. **OTHER PRESENCE**: VASCULAR refers to terrestrial plants or submerged aquatic vegetation (SAV) and NONVASCULAR refers to plankton, periphyton etc.
9. **PRECIPITATION**: Note if any precipitation is occurring during sampling
10. **PRECIPITATION LAST 24 HOURS**: Note how much precipitation has occurred within the last 24-h of sampling
11. **WATER ODOR**: Note if the sample water being collected has odor
12. **WATER CLARITY**: this describes the clarity of the water while standing creek side; clear represents water that is clear to the bottom, cloudy may not be clear to bottom but greater than 4" can be seen through the water column.
13. **WATER COLOR**: This is the color of the water from standing creek side
14. **OBSERVED FLOW**: Visual estimates in cubic ft/s.

SAMPLE DETAILS:

1. **EVENT TYPE**: Note the event type based which type of media is being collected
2. **SAMPLE TYPE**: GRAB samples are when bottles are filled from a single depth; INTEGRATED sample are taken from MULTIPLE depths and combined.
   a. GRAB: use 0.1 for subsurface samples; if too shallow to submerge bottle; depth =0
   b. INTEGRATED: -88 in depth sampled, record depths combined in sample comments
3. **SAMPLING CREW**: J. Smith, S. Ride (first person listed is crew leader)
4. **STARTING BANK**: Which side of the stream was accessed first. Bearings are always recorded looking downstream
5. **OCCUPATION METHOD**: What media was used to access the site
6. **TARGET LAT/LONG**: Refers to the existing station location that the sampling crew is trying to achieve; can be filled out prior to sampling
7. **ACTUAL LAT/ LONG**: is the location of the current sample event.
8. **SAMPLE LOCATION**: describes from where IN water body sample was taken: Can be combined; ex: bank/thalweg or midchannel /thalweg
9. **HYDROMODIFICATION:** Describe existing hydromodifications such as a grade control, drainage pipes, bridge, culvert
10. **HYDROMOD LOC**: if there was an IMMEDIATE (with in range potentially effecting sample) hydromodification; was sample taken upstream or downstream of modification; if there is no hydromodification, NA is appropriate
11. **STREAM DEPTH, WIDTH & DISTANCE FROM BANK**: describe in meters at point of sample. Distance from bank should be recorded from the starting bank

Field Data Logbook

A Field Data Logbook or a Field Folder is taken into the field on each sampling trip. The use of bound or loose-leaf notebooks is left up to the entity conducting the monitoring. A good safety precaution against the loss of a bound field data logbook is to photocopy the current pages upon returning from the field. These pages are kept on file at the specific sample collection entity's office. If a loose-leaf notebook is used, take care to remove original field data log sheets from the notebook and file in the office. Copies of the field data log sheets may be left in the notebook for future reference.

Field Data Logbooks (bound or loose leaf sheets) are maintained on file indefinitely in each regional office or contract laboratory office. They are never discarded, since the logbook may be the only written record of field measurements. Field Data Logbooks are reviewed periodically during SWAMP QA site visits. At this point, these field notes are not inclusive of the information that would be collected for biological assessment work, and several other data measurement types.

Flow

Sampling crews should be notified on reconnaissance forms if it is known that there is an operational United States Geological Survey (USGS) gage is located at or nearby a sampling site. If there is a USGS gage nearby, a gage height in feet is recorded and later converted to an instantaneous flow value and recorded in the logbook. The gage height is always to be reported to the USGS for conversion to flow. If a USGS gage is not available, a flow measurement should be taken, if requested. See Instantaneous Flow Measurement information starting on page 13 in this document. In addition, it is recommended that a flow severity value is recorded at each stream or river station that is not tidally influenced. See the Flow Severity section starting on page 13 of this document. Centroid velocity measurements may also be taken as a minimum acceptable rough characterization of the stream flow as requested, although this measurement is not to be recorded as a flow, since it is only a velocity measurement.

Record of Samples Collected for Purposes of Chemical Analysis

The general types of chemical samples to be collected are listed for each site, since this may vary from site-to-site (e.g., metals-in-water, pesticides-in-sediments, routine water quality). Analyses authorization forms are recommended since different authorized laboratories perform different chemical analyses. The method of preservation for each chemical sample is recorded, as appropriate.

Record of Data Submission

The *Logbook* field must indicate in some manner whether data recorded in the logbook has been transcribed onto data forms and submitted to the SWAMP data management staff.

Other Observations

Water Appearance Note general appearance (e.g., color, unusual amount of suspended matter, debris or foam)

Sediment Appearance

Color, Odor and sediment composition should be noted.

Weather Note recent meteorological events that may have impacted water quality; (e.g., heavy rains, cold front, very dry, very wet)

Biological Activity Note excessive macrophyte, phytoplankton or periphyton growth. The observation of water color and excessive algal growth is very important in explaining high chlorophyll a values. Other observations such as presence of fish, birds and spawning fish are noted.

Watershed or Instream Activities Note instream or drainage basin activities or events that are impacting water quality (e.g., bridge construction, shoreline mowing, livestock watering upstream).

Record of Pertinent Observations Related to Water Quality and Stream Uses If the water quality conditions are exceptionally poor, note that standards are not met in the observations, (e.g., dissolved oxygen is below minimum criteria). Note uses (e.g., swimming, wading, boating, fishing, irrigation pumps, navigation). Eventually, for setting water quality standards, the level of use will be based on comments related to the level of fishing and swimming activities observed at a station.

Specific Sample Information Note specific comments about the sample itself that may be useful in interpreting the results of the analysis (e.g., number of sediment grabs, or type and number of fish in a tissue sample). If the sample was collected for a complaint or fish kill, make a note of this in the observation section.

Missing Parameters If a scheduled parameter or group of parameters is not collected, make some note of this in the comments.

Field Data Measurements

While collecting water samples (see Field Collection Procedures for Water Samples section), record appropriate field measurements. When field measurements are made with a multiparameter instrument, it is preferable to place the sonde in the body of water to be sampled and allow it to equilibrate in the dissolved oxygen (D.O.) mode while water samples are collected. Field measurements are made at the centroid of flow, if the stream visually appears to be completely mixed from shore to shore. *Centroid* is defined as the midpoint of that portion of the stream width which contains 50% of the total flow. For routine field measurements, the date, time and depth are reported as a grab. Measure Quality Objectives (MQO's) for field measurements are listed in appendix C of the SWAMP QAMP.

Recommended Depths for Conducting Field Data Measurements

Water Depth Less than 5 ft (<1.5 m) If the water depth is less than 5 ft (1.5 m), grab samples for water are taken at approximately 0.1 m (4 in.), and multi-probe measurements are taken at approximately 0.2 m (8 in.). This is because all sensors have to be submerged, so 0.1 m would not be deep enough. But taking a grab sample at 0.2 m is not always feasible, as it is difficult to submerge bottles to that depth, and in many cases the bottle will hit the stream bottom.

Water Depth Greater than 5 ft (>1.5 m) If the water depth at the sampling point exceeds 5 ft (1.5 m) in depth, a vertical profile of dissolved oxygen, temperature, pH and specific conductance are made using the multiparameter probe equipment. The depth of the sonde at the time of measurement is most accurately determined from the depth sensor on the multiparameter sonde rather than depth labels on the cable.

Vertical Depth Profiles and Depth-Integrated Sample Collection If depth integration sampling is being conducted, or if vertical profile measurements are requested, multi-probe measurements are made starting at a depth of 0.2 m, and are then conducted at 1.0, 2.0, 3.0, 4.0, and 5.0 m depths after that until 5.0 m depth is reached. Beginning at 5.0 m, measurements are made every 5.0 m through depth profile.

Field data for multiparameter vertical depth profiles are recorded in final form on the SWAMP Field Data Sheets and submitted to the SWAMP data management staff.
Go to http://mpsl.mlml.calstate.edu/swdwnlds.htm for detailed information on data reporting.

Water Temperature (^{O}C)

Water temperature data are recorded for each SWAMP visit in final form in a Field Data Logbook and submitted to the SWAMP data management staff.
See http://mpsl.mlml.calstate.edu/swdwnlds.htm for detailed information on data reporting.

Temperature Sampling Procedures

Temperature is measured in-stream at the depth(s) specified above. Measuring temperature directly from the stream by immersing a multiprobe instrument or thermometer is preferred.

Hand Held Centigrade Thermometer

If an electronic meter is not available, the temperature is measured with a hand-held, centigrade thermometer (Rawson, 1982).

- In wadeable streams, stand so that a shadow is cast upon the site for temperature measurement.

- Hold the thermometer by its top and immerse it in the water. Position the thermometer so that the scale can be read.
- Allow the thermometer to stabilize for at least one minute, then without removing the thermometer from the water, read the temperature to the nearest 0.1^{o} C and record.
- Do not read temperature with the thermometer out of the water. Temperature readings made with modern digital instruments are accurate to within $\pm$ 0.1^{o} C.

Temperature Measurement from a Bucket

When temperature cannot be measured in-stream, it can be measured in a bucket-Nalgene or plastic. Care must be taken to insure a measurement representative of in-stream conditions.

The following conditions must be met when measuring temperature from a bucket:

- The bucket must be large enough to allow full immersion of the probe or thermometer.
- The bucket must be brought to the same temperature as the water before it is filled.
- The probe must be placed in the bucket immediately, before the temperature changes.
- The bucket must be shaded from direct sunlight and strong breezes prior to and during temperature measurement.
- The probe is allowed to equilibrate for at least one minute before temperature is recorded.
- After these measurements are made, this water is discarded and another sample is drawn for water samples which are sent to the laboratory.

pH (standard units)

pH data is recorded for each SWAMP visit in final form on the Field Data Sheets and submitted to the SWAMP data management staff. See http://mpsl.mlml.calstate.edu/swdwnlds.htm for detailed information on data reporting.

pH Sampling Equipment

The pH meter should be calibrated according to the recommended procedures for calibration and maintenance of SWAMP field equipment. Calibration directions are listed in the manufactures field equipment operations manual. The pH function is pre and post calibrated every 24 h of use for multiparameter instruments.

pH Sampling Procedures

In-stream Method

Preferably, pH is measured directly in-stream at the depth(s) specified earlier in this document. Allow the pH probe to equilibrate for at least one minute before pH is recorded to the nearest 0.1 pH unit.

pH Measurement from a Bucket

When pH cannot be measured in-stream, it can be measured in a bucket-Nalgene or plastic. The following precautions are outlined above; “Temperature Measurement from a Bucket”.

Potential Problems

- If the pH meter value does not stabilize in several minutes, out gassing of carbon dioxide or hydrogen sulfide, or the settling of charged clay particles may be occurring (Rawson, 1982).
- If out gassing is suspected as the cause of meter drift, collect a fresh sample, immerse the pH probe and read pH at one minute.
- If suspended clay particles are the suspected cause of meter drift, allow the sample to settle for 10 min, then read the pH in the upper layer of sample without agitating the sample.
- With care, pH measurements can be accurately measured to the nearest 0.1 pH unit.

Dissolved Oxygen (mg/L)

Dissolved oxygen (D.O.) data is recorded for each SWAMP visit in final form on a Field Data Sheet and submitted to the SWAMP data management staff.
See http://mpsl.mlml.calstate.edu/swdwnlds.htm for detailed information on data reporting.

Dissolved Oxygen Sampling Equipment

The dissolved oxygen meter should be calibrated according to the recommended procedures for calibration and maintenance of SWAMP field equipment. Calibration directions are listed in the manufactures field equipment operations manual.

Multiprobe Instrument

Pre and post calibrate the D.O. sensor every 24 h and for elevations greater than 500 ft on the multiprobe instrument. Preferably, D.O. is measured directly in-stream at the depth(s) specified in the Field Measurements section above. The D.O. probe must equilibrate for at least 90 s before D.O. is recorded to the nearest 0.1 % saturation or mg/L. Care must be taken at profile stations to insure that the reading is stable for each depth. Since dissolved oxygen takes the longest to stabilize, record this parameter after temperature, conductivity and pH. If the D.O. probe has an operable, automatic stirrer attached, the D.O. probe does not have to be manually stirred. However, if the probe is not equipped with an automatic stirrer, manual stirring must be provided by raising and lowering the probe at a rate of 1 ft/s (0.3m/s) without agitating the water surface. If the stream velocity at the sampling point exceeds 1 ft/s, the probe membrane can be pointed upstream into the flow and manual stirring can be avoided (Rawson, 1982).

D.O. Measurement from a Bucket

When D.O. cannot be measured in-stream, it can be measured in a bucket-Nalgene or plastic, following precautions outlined in the Temperature Measurement from a Bucket listed above. During equilibration and reading, water should be moved past the membrane surface at a velocity of 1 ft/s (0.3 m/sec), either by automatic stirrer or manual stirring. If stirred manually in a bucket, the water surface is not agitated (Rawson, 1982).

24-Hour Average D.O. (if requested in special study)

Unattended 24-Hour D.O. Data Collection

Why Collect 24-Hour Data

Dissolved oxygen sampling for standards compliance is targeted to water bodies where low instantaneous D.O. levels indicate partial or nonsupport of designated aquatic life uses. Intensive monitoring is conducted with automated equipment that is preset to record and store field measurements hourly over one 24-h period. Four or more dissolved oxygen measurements may also be made manually at 4-6-h intervals over one 24-h period, as long as one is made near sunrise (0500-0900 h) to approximate the daily minimum. However, data collected with automated equipment is preferred.

When to Take Measurements

All 24-h D.O. monitoring events must be spaced over an index period representing warm-weather seasons of the year (approx March 15-October 15), with between one-half to two-thirds of the measurements occurring during the critical period (July 1-September 30). The ***critical period*** of the year is when minimum stream flows, maximum temperatures, and minimum dissolved oxygen concentrations typically occur in area streams. **A flow measurement must be taken at the time of deployment.** In a perennial stream, a 24-h data for standards compliance can not be used if the flow is less than the 7Q2. In perennial streams, the D.O. criterion to do not apply for flows under the 7Q2. A period of about one month must separate each 24-h sampling event. Additional samples may be collected outside the index period to further characterize a water body, but that information is generally not used for assessing standards compliance.

Frequency of Measurements

The measurement interval should be no more than once per 15 min and no less than once per hour.

Where to Take Measurements

For purposes of determining standards compliance with the 24-h average criteria, samples collected near the surface will be considered representative of the mixed surface layer. In deep streams, reservoirs, and tidally influenced water bodies, automated equipment is positioned between 1 foot (from the surface) to one-half the depth of the mixed surface layer. At least 10 24-h monitoring events (using the 24-h criteria and/or absolute minimum criteria) at each site within a 5-year period are recommended to provide adequate data for assessment.

When to Collect Other Routine Samples, if doing 24-hour D.O. measurements

Other routine field measurements and water samples should be collect at either the time of deployment, at the reference check, or when the multiprobe recording 24-h data is retrieved. When ever possible, flow must be measured at the 24-h site.

Priority for Scheduling 24-Hour Sampling Events

- < 303d listed waterbodies
- < Waterbodies with Concerns for DO problems (too few samples available for full use assessment).

- Occurrence of low D.O. concentrations observed during the day
- Waterbodies with trends indicating declining D.O. concentrations
- Waterbodies which would contribute to an Ecoregion data set

Data Reporting for 24-hour D.O. measurements
Dissolved oxygen values recorded over the 24-h period are summed and divided by the number of measurements to determine the average concentration, which is compared to the 24-h criterion. The lowest D.O. value from each 24-h set is compared to the minimum criterion. There will be occasions when a complete 24-h data set won't be possible. For example, if there are 18 measurements instead of 24, a time weighted diurnal average needs to be calculated. This can be easily done using GW Basic.

Support of assigned aquatic life use is based on 24-h D.O. average and minimum criteria for each monitoring event. Report the 24-h average D.O. value, number of measurements over a 24-h period, and the minimum, and maximum values. Report data as a time composite sample with a beginning and ending date and time, covering the 24-h period measured.

Specific Conductance (µS/cm)

Specific conductance should be recorded for each SWAMP visit in final form on a Field Data Sheet and submitted to the SWAMP data management staff.
See http://mpsl.mlml.calstate.edu/swdwnlds.htm for detailed information on data reporting.

Specific Conductance Sampling Equipment
The conductivity meter should be calibrated according to the recommended procedures for calibration and maintenance of SWAMP field equipment. Calibration directions are listed in the manufactures field equipment operations manual.

Specific Conductance Sampling Procedure
Preferably, conductivity is measured directly in-stream at the depth(s) specified earlier in this document. Allow the conductivity probe to equilibrate for at least one minute before specific conductance is recorded to three significant figures (if the value exceeds 100). The primary physical problem in using a specific conductance meter is entrapment of air in the conductivity probe chambers. The presence of air in the probe is indicated by unstable specific conductance values fluctuating up to $\pm$100 µS/cm. The entrainment of air can be minimized by slowly, carefully placing the probe into the water; and when the probe is completely submerged, quickly move it through the water to release any air bubbles.

If specific conductance cannot be measured in-stream, it should be measured in the container it can be measured in a bucket-Nalgene or plastic. The following precautions are outlined above; "Temperature Measurement from a Bucket".

Salinity (parts per thousand--ppt, or ‰)
The value for salinity is computed from chloride concentration or specific conductance. The calculation assumes a nearly constant ratio for major ions in an estuary when seawater is diluted

by river water. This assumption does not hold for cases where salinity is less than about three parts per thousand. Salinity determinations at such low values are only approximate. In estuarine waters, salinity is a relevant and meaningful parameter. Often the salinity may be low, approaching that of freshwater. Nevertheless, this is useful information. Determine if a station is estuarine from historical records (i.e., experiences cases where salinity is >2.0 ppt) and always report salinity at this station, regardless of the salinity during periods of high flow.

Salinity is measured directly in-stream at the depth(s) specified earlier in this document. Salinity data should be recorded for each SWAMP visit in final form on a Field Data Sheet and submitted to the SWAMP data management staff. See http://mpsl.mlml.calstate.edu/swdwnlds.htm for detailed information on data reporting.

Values between 2.0 ppt and 1.0 ppt should be reported as <2.0 ppt rather than the actual value and values <1.0 ppt should be reported as <1.0 ppt. The field instruments compute salinity from specific conductance and temperature, and display the value in parts per thousand. Report salinity values above 2.0 ppt to the nearest 0.1 ppt.

Secchi Disc Transparency (meters)--if requested in special study

Secchi disk transparency should be recorded for each SWAMP visit in final form on a Field Data Sheet and submitted to the SWAMP data management staff. See http://mpsl.mlml.calstate.edu/swdwnlds.htm for detailed information on data reporting.

Secchi Disk Sampling Equipment

- Secchi disk, 20 cm in diameter
- Measuring tape

Secchi Disk Transparency Sampling Procedures

Preferably, Secchi disk transparency is measured directly in-stream wherever conditions allow. The Secchi disk should be clean, weighted and suspended with chain, wire, or Dacron line (the line used to suspend the Secchi disk should not be nylon or cotton; stretching may cause erroneous readings). Another option is to attach the Secchi disk to a metal rod calibrated in metric units.

Average Turbidity

The Secchi disk should be lowered vertically in a location shielded from direct sunlight. Glare from the water's surface will affect the accuracy of the measurement. Don't wear sunglasses.

Slowly lower the disk until it disappears from view. The person viewing the disk should maintain an eye level of less than two meters above the water's surface. Note the depth at which the disk disappears from view.

Slowly raise the disk until it becomes visible. Note the depth at which the disk reappears.

Compute the mathematical average of the two depths noted and record the average value to two significant figures in the field logbook. The recorded average value is the Secchi disk transparency.

High Turbidity (Muddy Water)

In streams with very high turbidity, high velocity, and/or poor access, it may be necessary to measure Secchi disk transparency in a bucket. Fill the bucket from the centroid of flow being careful not to disturb the substrate.

Follow steps above for measuring the Secchi disk depth within 30 s after raising the filled bucket from the water's surface. Or, re-suspend the solids by stirring, then quickly make the measurement.

Record Secchi disk transparency to two significant figures.

Low Turbidity (Clear Water)

Some bodies of water will be so clear and shallow that it will not be possible to lower the Secchi disk until it disappears from view.

Measure and record the depth at the deepest point accessible. Report Secchi disk transparency as greater than the deepest depth measured.

Example (Low Turbidity): South Fork Rocky Creek is a small (<1 ft^3/s) clear stream. The stream in the vicinity of the sampling site was less than 1 m deep and the bottom was clearly visible everywhere. However, a pool was located in the stream next to a bridge. The maximum depth of the pool was 2.6 m at which depth the Secchi disk was still visible. Therefore, Secchi disk transparency for South Fork Rocky Creek was recorded as > 2.6 m.

Importance of Secchi Disk Data

Eutrophication, the natural aging process in reservoirs and lakes is accelerated by human activities which add nutrients to lakes, reservoirs, and the surrounding watersheds. Section 314 of the Clean Water Act (CWA) of 1987 requires all states to classify lakes and reservoirs according to trophic state. Although chlorophyll a is the most direct measure of algal biomass, other indices and programs utilize Secchi disk depth as the primary factor.

Turbidity Measurement with Turbidity Meter

Nephelometric Turbidity can be determined by measuring the amount of scatter when light is passed through a sample using a turbidity meter. The LaMotte 2020 Turbidity meter is a suitable instrument for example.

Meters should be calibrated using a standard close to the expected sample value.

For instructions on how to operate the instruments refer to the manufacturer's manual. Turbidity measurements can be executed together with water sampling. The turbidity sample has to be representative for the sampled water mass. Make sure that no gas bubbles are trapped in the vial for the reading and that the outside of the vial is wiped completely clean (i.e., meaning free of moisture, lint and fingerprints). Take several measurements to assure an accurate reading. Do not record values that vary greatly. If variations are small, record an average. If settling particles are present, record a reading before and one after settling. The meter might have to be recalibrated with a different standard, if the sample water readings are outside of the calibration standard limits.

Days Since Last Significant Precipitation

Significant precipitation is defined as any amount that visibly influences water quality. Water quality in small to medium streams and in the headwaters of many reservoirs is influenced by runoff during and immediately after rainfall events. This influence is site specific and poorly studied. As part of a new initiative to understand and regulate the adverse effects of runoff, SWAMP would like to associate recent rains or melted snow with ambient water quality, using a parameter defined as "days since last significant precipitation". Record the number of days, rounded to the nearest whole number, since a rain has occurred that, in the best professional judgment of monitoring personnel, may have influenced water quality. If it is raining when the sample is collected, or has rained within the last 24-h, report a value of <1. If it has been a long time since a significant rain, record this as greater than that particular value, for example >7 days. If confidence about the recent history of precipitation is low, draw a line through the space on the data form.

Flow Severity -- recommended new parameter

Flow severity should be noted for each SWAMP visit to non-tidally influenced flowing streams and submitted in the comments on the SWAMP Field Data Sheet. It should be recorded even if flow is visible but not measurable on that sampling visit. There are no numerical flow guidelines associated with flow severity. This is an observational measurement that is highly dependent on the knowledge of monitoring personnel. It is a simple but useful piece of information when assessing water quality data. For example, a bacteria value of 10,000 with a flow severity of 1 would represent something entirely different than the same value with a flow severity of 5. The six flow severity values are; 1=No Flow, 2= Low Flow, 3 = Normal Flow, 4 = Flood, 5 = High Flow, and 6 = Dry. The following are detailed descriptions of severity values:

1 **No Flow** When a flow severity of one (1 = no flow) is recorded for a sampling visit, then a flow value of zero ft^3/s should also be recorded for that sampling visit. **A flow severity of one (1) (no flow) describes situations where the stream has water visible in isolated pools.** There should be no obvious shallow subsurface flow in sand or gravel beds between isolated pools. Low flow does not only apply

to streams with pools. It also applies to long reaches of bayous and streams that have no detectable flow but may have water from bank to bank.

2 **Low Flow** When stream flow is considered low a flow severity value of two (2) is recorded for the visit and the corresponding flow measurement is also recorded for that visit. In streams too shallow for a flow measurement but with detected water movement, record a value of < 0.10 cfs. Note: Use a stick or other light object to verified the direction of water movement (i.e., movement is downstream and not the affect of wind.) What is low for one stream could be high for another.

3 **Normal Flow** When stream flow is considered normal, a flow severity value of three (3) is recorded for the visit and the corresponding flow measurement is also be recorded for that visit. Normal is highly dependent on the stream. Like low flow, what is normal for one could be high or low for another stream.

4 and 5 **Flood and High Flow** Flow severity values for high and flood flows have long been established by EPA and are not sequential. Flood flow is reported as a flow severity of four (4) and high flows are reported as a flow severity of five (5). High flows would be characterized by flows that leave the normal stream channel but stay within the stream banks. Flood flows are those which leave the confines of the normal stream channel and move out on to the flood plain.

6 **Dry** When the stream is dry a flow severity value of six (6 = dry) is recorded for the sampling visit. In this case the flow is not reported. This will indicate that the stream is completely dry with no visible pools.

Flow information for over 200 USGS sites is available on the Internet. The address is **http://water.usgs.gov/index.html**. This is useful information in determining flow conditions prior to sampling. This information may be included in general observations.

Flow Measurement Method (Reporting)

The method (or instrument) used to measure flow is noted by reporting a method number. The method numbers are:

1- Flow Gage Station (USGS/IBWC)	3- Electric (ex. Marsh-McBirney)
2- Mechanical (ex. Pigmy meter)	4- Weir/Flume
5- Other (orange peel, etc.)	

Flow (ft^3/s)

If requested, flow data should be recorded for each monitoring visit to non-tidal, flowing streams. Flow data should be recorded in final form on a Field Data Sheet and submitted to the SWAMP data management staff. See http://mpsl.mlml.calstate.edu/swdwnlds.htm for detailed information on data reporting. The following are two exceptions to the flow reporting requirement:

No Flow/ Pools — If there is no flow at a stream site and accessible, isolated pools remain in the stream bed, collect and report the required field data and laboratory samples from the pools and report instantaneous flow. Under these conditions, flow (ft^3/s) should be reported as zero. The reported flow severity value should be one. Pools may represent natural low-flow conditions in some streams and the chemistry of these pools will reveal natural background conditions.

Dry — If the stream bed holds no water, the sampling visit is finished. Report that the stream was "dry" in the observations and record a value of six (meaning "dry") for flow severity. No value is reported for flow since there is no water.

Flow Measurement

If a flow measurement is required at a site, measure and record flow after recording visual observations. The intent of measuring flow first is to delay collection of chemical and biological water samples with limited holding times. Care must be taken not to collect water samples in the area disturbed during flow measurement. There are several acceptable flow measurement methods that can be used.

U.S. Geological Survey (USGS) Gaging Station

Some SWAMP Stations are sampled at sites where the USGS maintains flow gaging equipment. On any type of sampling visit to a site that has a USGS flow gage, observe and record the gage height to the nearest hundredth of a foot in the field logbook. Upon return to the office, contact the USGS office responsible for maintaining the gage. USGS personnel can provide the flow value in cubic feet per second (ft^3/s) that corresponds to the gage height. Although SWAMP personnel may have a rating curve available to them, shifts associated with changes in the stream bed may occur over time. Always call the USGS to determine the shift. At some sites the shift changes frequently. At others, the relation between stream flow and gage height is almost unchanging. If a gage is no longer maintained by USGS, cross out the recorded gage height and be prepared to measure flow by another method on the return visit to that site.

Several factors may influence the accuracy of the USGS rating curves that are used to convert gage height to flow. If there is any doubt about the accuracy of a USGS gage height reading or flow rating curve, sampling personnel should measure the flow if possible.

Gage height may be indicated at a USGS gage by one of three methods:

Staff Gage

Staff gages are enameled steel plates (with the appearance of large measuring tapes) bolted to some stable structure. For example, staff gages may be bolted to concrete bridge abutments, pillars, or docks. The staff gage face is white with black lettering and gradations. The gradations shown are feet, tenths of a foot, and 0.02 of a foot. The point at which the water level crosses the staff gage should be recorded to the nearest hundredth of a foot.

Wire Weight Gage

Wire weight gages are locked, metal boxes with approximate dimensions of 15 in. long x 12 in. tall x 12 in. deep. Wire weight gages are usually affixed to bridge rails near mid-stream. They must be unlocked with a USGS key. The wire weight gages house a weight attached by wire cable to a graduated reel (gradations are tenths and hundredths of feet) with a counter at one end.

When the reel is released the weight can be gradually lowered until the bottom of the weight contacts the water surface. At the point of contact, the weight causes the water surface to ripple slightly. Maintaining the weight in that position, record the counter value to the nearest whole number and the point indicated by the stylus on the graduated reel to the nearest hundredth of a foot. Determine if the gage is the movable type that can be moved to multiple locations on the bridge. This type is common on braided streams. A correction value is stamped on the bridge near each point that the gage can be attached. Record the corrected value as the gage height in feet.

Bubble Gage

Bubble gages are locked in metal sheds that are approximately 4 ft wide x 4 ft deep x 6.5 ft tall. The gage houses are most frequently located on the shore near a bridge but sometimes are attached to bridge pillars near mid-stream or established on the stream bank far from any bridge. The gage house must be unlocked with a USGS key. Bubble gages in gage houses usually indicate the gage height in two or three locations. A counter attached to the manometer system indicates gage height in feet. Some gage houses have stilling wells that can be entered. Often there is a staff gage on the inside wall.

Most bubble gages are also equipped with digital recorders. Digital recorders consist of two white, coded discs, approximately 4 in. in diameter with a punch tape overlapping a portion of each disc. The discs are marked with 100 gradations. As the front of the digital recorder is viewed, the stylus at the disc on the left indicates height in feet. The stylus at the disc on the right indicates gage height in hundredths of feet. The gage height from both discs should be added and the number recorded in the field logbook as gage height to the nearest hundredth of a foot.

Many USGS metal sheds also contain a surface level recorder. This devise can be opened to determine how stable stream flow has been prior to the sampling event. Record observations concerning the flow hydrograph.

Instantaneous Flow Measurement

Water quality monitoring visits to sites where there are no nearby USGS flow gauges will require water quality monitoring personnel to measure flow, when requested by Regional Water Quality Control Boards (Regional Boards).

Flow Measurement Equipment

Flow meter

One of the following or an equivalent:

- Marsh-McBirney Electronic meter
- Montedoro-Whitney Electronic meter
- Price Pigmy meter (with timer and beeper)
- Price meter, Type AA (with Columbus weight)

Additional Equipment

- Top-setting wading rod (preferably measured in tenths of feet)(see Figure 1).
- Tape measure (with gradations every tenth of a foot).

Flow Measurement Procedure (USGS, 1969)

Select a stream reach with the following characteristics:

- Straight reach with laminar flow (threads of velocity parallel to each other) and bank to bank. These conditions are typically found immediately upstream of riffle areas or places where the stream channel is constricted.
- The site should have an even streambed free of large rocks, weeds, and protruding obstructions that create turbulence. The site should not have dead water areas near the banks, and a minimum amount of turbulence or back eddies.

Flat Streambed Profile (cross section)

Stretch the measuring tape across the stream at right angles to the direction of flow. When using an electronic flow meter, the tape does not have to be exactly perpendicular to the bank (direction of flow). When using a propeller or pigmy type meter, however, corrections for deviation from perpendicular must be made.

If necessary and possible, modify the measuring cross section to provide acceptable conditions by building dikes to cut off dead water and shallow flows, remove rocks, weeds, and debris in the reach of stream one or two meters upstream from the measurement cross section. After modifying a streambed, allow the flow to stabilize before starting the flow measurement.

Record the following information on the flow measurement form (see example Flow Measurement Forms at end of this document):

- Station Location and Station ID
- Date
- Time measurement is initiated and ended
- Name of person(s) measuring flow
- Note if measurements are in feet or meters
- Total stream width and width of each measurement section
- For each cross section, record the mid-point, section depth and flow velocity

Measuring the Stream Width

Measure and record the stream width between the points where the tape is stretched (waters edge to waters edge).

Determining the Number of Flow Cross Sections

Determine the spacing and location of flow measurement sections. Some judgment is required depending on the shape of the stream bed. Measurements must be representative of the velocity within the cross-section. If the stream banks are straight and the depth is nearly constant and the bottom is free of large obstructions, fewer measurements are needed, because the flow is homogeneous over a large section. Flow measurement sections do not have to be equal width. However, they should be unless an obstacle or other obstruction prevents an accurate velocity measurement at that point. ***No flow measurement section should have greater than 10% of the total flow.***

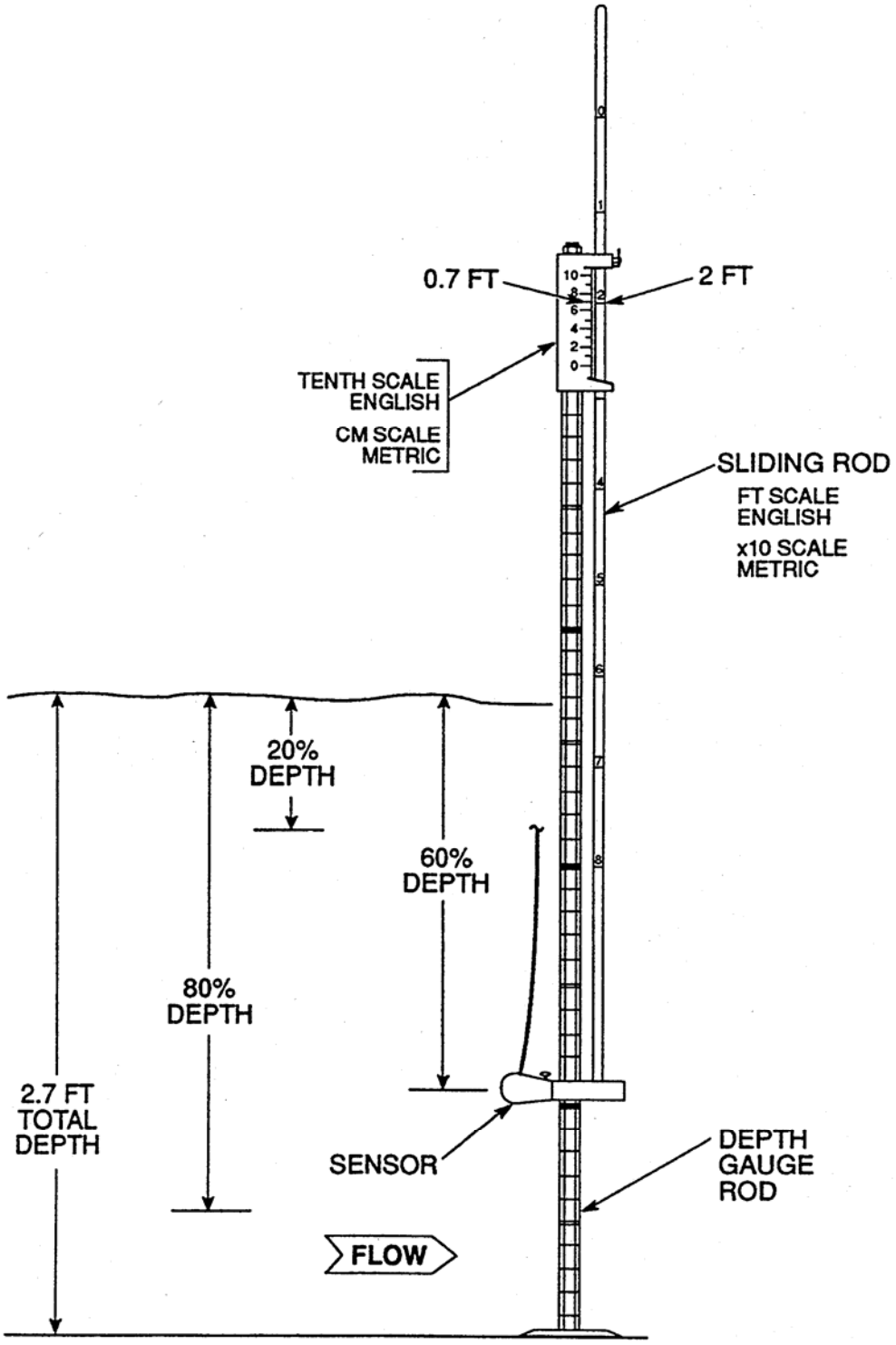
If the *stream width is less than 5 ft*, use flow sections with a width of 0.5 ft (See example 1 on page 23 of this document). If the *stream width is greater than 5 ft*, the minimum number of flow measurements is 10. The preferred number of flow measurement cross sections is 20-30 (See Example 2 on page 24 on this document). The total stream width is 26 ft with 20 measurements, section widths will be 1.3 ft (26/20 = 1.3).

Determining the Mid-Point of the Cross Section

To find the mid-point of a cross section, divide the cross section width in half. Using Example 2 (see forms at end of document);

- The total stream width is 26 ft with 20 cross sections and each cross section width is equal to 1.3 ft.
- Divide 1.3 ft in half and the mid-point of the first section is 0.65 ft. In this example the tape at waters edge is set at zero (0) ft.
- By adding 0.65 to zero the mid-point of the first section is 0.65 ft.
- Each subsequent mid-point is found by adding the section width (1.3 ft) to the previous mid-point. For example; MIDPOINT #1 is 0.65 + 0.0 = 0.65; MIDPOINT #2 is 0.65 + 1.3= 1.95 ft; MIDPOINT #3 is 1.95 + 1.3 = 3.25 ft and ....MIDPOINT # 20 is 24.05 +1.3.
- Place the top setting wading rod at 0.65 ft for the first measurement.
- Using a top setting wading rod, measure the depth at the mid-point of the first flow measurement section and record to the nearest 0.01 ft.

Figure 1. Top-Setting Wading Rod (Marsh-McBirney)



Adjusting the Sensor Depth at a Cross Section

Adjust the position of the sensor to the correct depth at each mid-point. The purpose of the top setting wading rod is to allow the user to easily set the sensor at 20%, 60%, and 80% of the total depth. The total depth can be measured with the *depth gage rod*. Each single mark represents 0.10 foot, each double mark represents 0.50 foot, and each triple mark represents 1.00 foot (see Figure 2).

For Depths < 2.5 Ft	If the depth is less than 2.5 ft, only one measurement is required at each measurement section. To set the sensor at 60% of the depth, line up the foot scale on the *sliding rod* with the *tenth scale*, located on top of the depth gage rod. If, for example, the total depth is 2.7 ft (as shown on Figure 2), then line up the 2 on the foot scale with the 7 on the tenth scale (Marsh-McBirney 1990).
For Depths > 2.5 Ft	If the depth is greater than 2.5 ft, two measurements should be taken at 20% and 80% of the total depth. To set the sensor at 20% of the depth, multiply the total depth by two. For example, if the total depth is 2.7 ft, the rod would be set at 5.4 ft (2.7 x 2). Line up the 5 on the sliding rod with the 4 on the tenth scale.
For Depths > 2.5 Ft (cont)	To set the sensor at 80% of the depth, divide the total depth by two. For example, the total depth is 2.7 ft the rod would be set at 1.35 ft (2.7/2). Line up the 1 on the sliding rod with the 0.35 on the tenth scale. The average of the two velocity measurements is used in the flow calculation. See page 2-36 for an example of a flow form recording measurements for depths greater than 2.5 ft. NOTE: The point where the rod is set for 20 and 80% of the depth will not equal values derived by calculating 20 and 80% of the total depth.

Measuring Velocity (this has typically been measured at 6/10 of the total depth, for velocity-only measurements)

- Position the meter at the correct depth and place at the mid-point of the flow measurement section. Measure and record the velocity and depth. The wading rod is kept vertical and the flow sensor kept perpendicular to the tape rather than perpendicular to the flow while measuring velocity with an electronic flow meter. When using a propeller or pigmy-type meter, however, the instrument should be perpendicular to the flow.
- Permit the meter to adjust to the current for a few seconds. Measure the velocity for a minimum of 20 s with the Marsh-McBirney and Montedoro-Whitney meters. Measure velocity for a minimum of 40 s (preferably 2 min with the Price and pigmy meters).
- When measuring the flow by wading, stand in the position that least affects the velocity of the water passing the current meter. The person wading stands a minimum of 1.5 ft downstream and off to the side of the flow sensor.

- A flow sensor, equipped with cable and weight may be used to measure flows where the water is too deep to wade. Follow the procedure involving meters attached to wading rods.
- Report flow values less than 10 ft^2/s to two significant figures. Report flow values greater than 10 ft^3/s to the nearest whole number, but no more than three significant figures.
- In cases where the flow is low and falling over an obstruction, it may be possible to measure the flow by timing how long it takes to fill a bucket of known volume.

Avoid measuring flow in areas with back eddies. The first choice would be to select a site with no back eddy development. However, this can not be avoided in certain situations. Measure the negative flows in the areas with back eddies. These negative values will be included in the final flow calculation.

Calculating Flow

To calculate flow, multiply the width x depth (ft^2) to derive the area of the flow measurement section. The area of the section is then multiplied by the velocity (ft/s) to calculate the flow in cubic feet per second (cfs or ft^3/sec) for that flow measurement section. When flow is calculated for all of the measurement sections, they are added together for the total stream flow (see Figure 2).

Q=Total Flow (or discharge), W=Width, D=Depth, V=Velocity.

$$Q = (W_1 * D_1 * V_1) + (W_2 * D_2 * V_2) + ... (W_n * D_n * V_n)$$

What to Do with Negative Values

Do not treat cross sections with negative flow values as zero. Negative values obtained from areas with back eddies should be subtracted during the summation of the flow for a site.

Flow Estimate (ft^3/s)

Flow estimate data may be recorded for a non-tidally influenced stream when it is not possible to measure flows by one of the methods described above. Flow estimates are subjective measures based on field personnel's experience and ability to estimate distances, depths, and velocities. If flow can not be measured at a routine non-tidal station, a new site should be selected where flow can be measured.

Flow Estimate Procedure

- Observe the stream and choose a reach of the stream where it is possible to estimate the stream cross section and velocity.
- Estimate stream width (ft) at that reach and record.
- Estimate average stream depth (ft) at that reach and record. Estimate stream velocity (ft/s) at that reach and record. A good way to do this is to time the travel of a piece of floating debris. If doing this method from a bridge, measure the width of the bridge. Have one person drop a floating object (something that can be distinguished from other

floating material) at the upstream side of the bridge and say start. The person on the downstream side of the bridge will stop the clock when the floating object reaches the downstream side of the bridge. Divide the bridge width by the number of seconds to calculate the velocity. The velocity can be measured at multiple locations along the bridge. These velocities are averaged. If this is done alone, watch for road traffic.

< Multiply stream width (ft) times average stream depth (ft) to determine the cross sectional area (in ft^2) which when multiplied by the stream velocity (in ft/s) and a correction constant, gives an estimated flow (ft^3/s).

Example: A stream sampler conducted a sampling visit to a stream while the flow meter was being repaired. The sampler looked at the creek downstream from the bridge and saw a good place to estimate flow. The stream width was around 15 ft. It appeared the average depth on this reach was about 0.75 ft. The sampler timed a piece of floating debris as it moved a distance of 10 ft in 25 s downstream over the reach. An estimated flow with a smooth bottom was calculated using the following formula.

Width x Depth x Velocity x A (correction factor)= estimated flow
15 ft (width) x 0.75 ft (depth) x 2.5 ft/s (velocity) x A =25 ft^3/s (cfs)

A is a correction constant: 0.8 for rough bottom and 0.9 for smooth bottom

Estimated flow should be reported to one or two significant figures.

Experienced field personnel are able to estimate flow to within 20% of actual flow for total flows less than 50 ft^3/s. The best way to develop this skill is to practice estimating flow before making measurements at all monitoring visits to non-tidally influenced flowing streams and then compare estimated flows with those obtained from USGS gages or from instantaneous flow measurements

Figure 2. Stream Flow (Discharge) Measurement

Example 1.
Stream Flow (Discharge) Measurement
Small Stream < 5 Ft Wide and #2.5 Ft Deep

Stream:___**_OAK CREEK**_____Date:__**5/29/91**___
Station Description:_____**at US Hwy 90A**_____
Time Begin:__**1545**_____Time End:__**1630**_____Meter Type:_**_Marsh-McBirney**_____
Observers:_____**BK/MK**_____Stream Width*:____**5 ft**_____ Section Width:____**0.5 ft**_____
Observations:_____

Section Midpoint (ft)	Section Depth (ft)	Observational Depth** Ft	Velocity		Area W x D (ft^2)	Discharge (Q) V x A (ft^3/s)
			At Point (ft/s)	Average (ft/s)		
0.25	**0.55**			**0.05**		**0.01375**
0.75	**0.80**			**0.11**		**0.044**
1.25	**0.85**			**0.27**		**0.42635**
1.75	**0.90**			**0.49**		**0.2205**
2.25	**1.10**			**0.58**		**0.275**
2.75	**1.50**			**0.72**		**0.540**
3.25	**1.20**			**0.76**		**0.456**
3.75	**0.90**			**0.76**		**0.342**
4.25	**0.75**			**0.44**		**0.165**
4.75	**0.30**			**0.00**		**0.00**
m^3/s x 35.3 = ft^3/s			**Total Discharge (3Q) (ft^3/s)**			**2.4826**

Example 2.

Stream Discharge Measurement Example (Larger Stream > 5 Ft and #2.5 Ft Deep)

Stream:___**_RED RIVER**_____Date:__**5/28/91**___

Station Description:_____**Post Oak Creek 40 m Below Sherman WWTP Outfall**_____

Time Begin:__**1542**_____Time End:__**1601**_____Meter Type:_**Marsh-McBirney**_____

Observers:_____**CM, EW, DO**_____Stream Width*:____**26 ft**___ Section Width:___**1.3 ft**_____

Observations:_____

Section Midpoint (ft)	Section Depth (ft)	Observational Depth** ft	Velocity		Area W x D (ft^2)	Discharge (Q) V x A (ft^3/s)
			At Point (ft/s)	Average (ft/s)		
0.65	0.55			2.03	0.715	1.451
1.95	0.40			2.04	0.520	1.061
3.25	0.42			2.02	0.546	1.103
4.55	0.38			1.77	0.494	0.874
5.25	0.40			1.75	0.520	0.910
7.15	0.42			1.93	0.546	1.054
8.45	0.40			1.99	0.52	1.035
9.75	0.37			1.92	0.481	0.924
11.05	0.37			1.56	0.481	0.750
12.35	0.43			1.32	0.559	0.738
13.65	0.40			1.36	0.520	0.707
14.95	0.42			1.33	0.546	0.726
16.25	0.40			1.35	0.520	0.702
17.55	0.45			1.64	0.585	0.959
18.85	0.48			1.70	0.624	1.061
20.15	0.48			2.00	0.624	1.248
21.45	0.50			1.95	0.650	1.268
22.75	0.40			2.18	0.520	1.134
24.05	0.48			1.71	0.624	1.067
25.35	0.50			0.60	0.650	0.390
				Total Discharge (3Q) (ft^3/s)		**19.162**

m^3/s x 35.3 = ft^3/s

Example 3.

Stream Flow (Discharge) Measurement (Larger Stream > 5 Ft and >2.5 Ft Deep)

Stream:____ARROYO COLORADO_____Date:__**6/16/98**__

Station Description:_____Downstream of Harlingen WWTP_____

Time Begin:__**1400**_____Time End:__**1445**_____Meter Type:__**Marsh-McBirney**_____

Observers:_____**JD, CK**_____Stream Width*:___**47.5 ft**___ Section Width:___**2.375 ft**_____

Observations: *Note that the starting point is at 4.7 ft on the measuring tape and not zero.

Section Midpoint (ft)	Section Depth (ft)	Observational Depth** ft	Velocity		Area W x D (ft^2)	Discharge (Q) V x A (ft^3/s)
			At Point (ft/sec)	Average (ft/sec)		
4.70	0.73			0.65	1.73	1.127
7.08	1.10			1.08	2.61	2.822
9.45	1.85			0.90	4.39	3.954
11.83	2.20			1.05	5.23	5.486
14.20	2.20			1.44	5.23	7.531
16.58	2.45			1.09	5.82	6.342
18.95	2.55	0.20	1.75	1.76	6.06	10.659
		0.80	1.76			
21.33	2.60	0.20	1.79	1.56	6.18	9.633
		0.80	1.32			
23.70	2.70	0.20	1.63	1.45	6.41	9.298
		0.80	1.26			
26.10	3.05	0.20	1.68	1.42	7.24	10.286
		0.80	1.15			
28.48	3.10	0.20	1.23	0.96	7.36	7.068
		0.80	0.69			
30.85	2.90	0.20	1.22	1.06	6.89	7.301
		0.80	0.89			
33.23	2.84	0.20	0.60	0.49	6.75	3.305
		0.80	0.37			
35.60	2.65	0.20	0.80	0.51	6.29	3.210
		0.80	0.21			
37.98	2.65	0.20	0.85	0.91	6.29	5.727
		0.80	0.96			
40.35	2.20			0.28	5.23	1.464
42.73	2.30			0.16	5.46	0.874
45.10	2.05			0.51	4.87	2.483
47.48	1.10			0.49	2.61	1.280
49.86	0.65			0.62	1.54	0.957

m^3/s x 35.3 = ft^3/s | **Total Discharge (3Q) (ft^3/s)**

Stream Flow (Discharge) Measurement Form

Stream:_____Date:_____
Station Description:_____
Time Begin:_____ Time End:_____ Meter Type:_____
Observers:_____Stream Width*:_____ Section Width:_____
Observations:

Section Midpoint (ft) (m)	Section Depth (ft) (m) (cm)	Observational Depth** ft-m-cm	Velocity		Area W x D (ft^2) (m^2)	Flow (Q) V x A (m^3/s) (ft^3/s)
			At Point (ft/s) (m/s)	Average (ft/s)(m/s)		

Summary of Significant Figures for Reporting Field Parameters

Parameter	Field Data Reporting Requirements
Water Temperature (^{o}C)	Report temperature to the nearest tenth of a degree. Round insignificant figures 0 through 4 down and 5 thru 9 up.
pH (s.u.)	Report pH to the nearest tenth of a pH standard unit.
D.O. mg/L **D.O.** (% saturation)	Report dissolved oxygen to the nearest tenth of a mg/L. Report % saturation to the nearest tenth of a percent
Specific Conductance (micro siemens/cm)	Report specific conductance to only three significant figures if the value exceeds 100. Do not report ORP which is displayed by some multiprobes.
Salinity (ppt)	Report salinity values above 2.0 ppt to the nearest tenth of a part per thousand. In estuarine waters report the actual values displayed by the multiprobe above 2.0 ppt and values less than 2.0 as <2.0 or <1.0 only. Determine if a station is estuarine (i.e., experiences cases where salinity is >2.0 ppt) and always report salinity at this station, regardless of the salinity during periods of high flow.
Secchi Disk (meters)	Report Secchi depth transparency in meters to two significant figures.
Days Since Last Significant Precipitation(days)	Report whole numbers. If it is raining when the sample is collected or has rained within the last 24 h, report a value of <1. If it has been over a week since a rainfall event, report a value of > 7.
Flow (ft^3/s)	Report instantaneous flow values less than 10 ft^3/s to two significant figures. Report flow values greater than 10 ft^3/s to the nearest whole number, but no more than three significant figures. When there is no flow (pools), report as 0.0. When there is no water, don't report a value, but report as "dry" in the observations.
Flow Severity (1-no flow, 2-low, 3-normal, 4-flood, 5-high, 6-dry)	When there is no flow (pools), report the severity as 1, and the instantaneous flow as 0.0 ft^3/s. If the stream is dry, record only flow severity, as a value of 6.

BEAUFORT SCALE: Specifications and equivalent speeds for use at sea

FORCE	EQUIVALEN SPEED 10 m above ground		DESCRIPTION	SPECIFICATIONS FOR USE AT SEA
	Miles/hour	knots		
0	0-1	0-1	Calm	Sea like a mirror
1	1-3	1-3	Light air	Ripples with the appearance of scales are formed, but without foam crests.
2	4-7	4-6	Light Breeze	Small wavelets, still short, but more pronounced. Crests have a glassy appearance and do not break.
3	8-12	7-10	Gentle Breeze	Large wavelets. Crests begin to break. Foam of glassy appearance. Perhaps scattered white horses.
4	13-18	11-16	Moderate Breeze	Small waves, becoming larger; fairly frequent white horses.
5	19-24	17-21	Fresh Breeze	Moderate waves, taking a more pronounced long form; many white horses are formed. Chance of some spray.
6	25-31	22-27	Strong Breeze	Large waves begin to form; the white foam crestsare more extensive everywhere. Probably some spray.
7	32-38	28-33	Near Gale	Sea heaps up and white foam from breaking waves begins to be blown in streaks along the direction of the wind.
8	39-46	34-40	Gale	Moderately high waves of greater length; edges of crests begin to break into spindrift. The foam is blown in well-marked streaks along the direction of the wind.
9	47-54	41-47	Severe Gale	High waves. Dense streaks of foam along the direction of the wind. Crests of waves begin to topple, tumble, and roll over. Spray may affect visibility.
10	55-63	48-55	Storm	Very high waves with long over- hanging crests. he resulting foam, in great patches, is blown in dense white streaks along the direction of the wind. On he whole the surface of the sea takes on a white appearance. The 'tumbling' of the sea becomes heavy and shock-like. Visibility affected.

Last edited on 09 January, 1999 Dave Wheeler weatherman@zetnet.co.uk
Web Space kindly provided by Zetnet Services Ltd, Lerwick, Shetland.
http://www.zetnet.co.uk/sigs/weather/Met_Codes/beaufort.htm

Field Collection Procedures for Water Samples

Scope and Application

This protocol describes the techniques used to collect water samples in the field in a way that neither contaminates, loses, or changes the chemical form of the analytes of interest. The samples are collected in the field into previously cleaned and tested (if necessary) sample bottles of a material appropriate to the analysis to be conducted. Pre-cleaned sampling equipment is used for each site, whenever possible and/or when necessary. Appropriate sampling technique and measuring equipment may vary depending on the location, sample type, sampling objective, and weather. Trade names used in connection with equipment or supplies do not constitute an endorsement of the product.

Summary of Method

Appropriate sample containers and field measurement gear as well as sampling gear are transported to the site where samples are collected according to each sample's protocol. Water velocity, turbidity, temperature, pH, conductivity, dissolved oxygen as well as other field data are measured and recorded using the appropriate equipment. These field data measurement protocols are provided in the SWAMP Field Measurement SOP. Samples are put on ice and appropriately shipped to the processing laboratories. This procedure has been modified from the Texas Natural Resources Conservation Commission's Procedure Manual for Surface Water Quality Monitoring, with major input from the United State's Geological Survey's (USGS's) National Water Quality Assessment (NAWQA) Protocol for Collection of Stream Water Samples, for which due credit is herewith given.

WATER SAMPLE COLLECTION

Water chemistry and bacteriological samples, as requested, are collected at the same location. *Water samples are best collected before any other work is done at the site*. If other work (e.g., sediment sample collection, flow measurement or biological/habitat sample collection or assessment) is done after or downstream of the collection of water samples, it might be difficult to collect representative samples for water chemistry and bacteriology from the disturbed stream. Care must be taken, though, to not disturb sediment collection sites when taking water samples.

The following general information applies to all types of water samples, unless noted otherwise:

Sample Collection Depth

Sub-Surface Grab Sample Samples are collected at 0.1 m below the water surface. Containers should be opened and re-capped under water in most cases.

Depth-integrated Sample If a depth-integrated sample is

taken, the sample is pumped from discrete intervals within the entire water column.

Surface Grab Sample Samples are collected at the surface when water depth is <0.1 m. Since there is a difference in water chemistry on the surface, compared to subsurface, surface water should be noted on the field data sheet as 0 m.

Where to Collect Samples

Water samples are collected from a location in the stream where the stream visually appears to be completely mixed. Ideally this would be at the centroid of the flow (*Centroid* is defined as the midpoint of that portion of the stream width, which contains 50% of the total flow), but depth and flow do not always allow centroid collection. For stream samples, the sampling spot must be accessible for sampling physicochemical parameters, either by bridge, boat or wading. Sampling from the shoreline of any water body (meaning standing on shore and sampling from there) is the least acceptable method, but in some cases is necessary.

In reservoirs, lakes, rivers, and coastal bays, samples are collected from boats at designated locations provided by Regional Water Quality Control Boards (Regional Boards).

Sampling Order if Multiple Media are Requested to be Collected

The order of events at every site has to be carefully planned. For example, if sediment is to be collected, the substrate can not be disturbed by stepping over or on it; water samples can not be taken where disturbed sediment would lead to a higher content of suspended matter in the sample. *For the most part, water samples are best collected before any other work is done at the site*. This information pertains to walk-in sampling.

Sample Container Labels

Label each container with the station ID, sample code, matrix type, analysis type, project ID, and date and time of collection (in most cases, containers will be pre-labeled). After sampling, secure the label by taping around the bottle with clear packaging tape.

Procedural Notes

For inorganic and organic water samples, bottles do not have to be rinsed if they are I-Chem 200 series or higher or ESS PC grade or higher. This means that the sample bottles are analyzed for contamination, and a certification of analysis is included with the bottles. Other sample containers are usually rinsed at least three times if the bottles do not meet these

requirements. See filling instruction for each type of analyses if there is uncertainty. If applicable to the sample and analysis type, the sample container should be opened and re-capped under water.

Sample Short-term Storage and Preservation

Properly store and preserve samples as soon as possible. Usually this is done immediately after returning from the collection by placing the containers on bagged, crushed or cube ice in an ice chest. Sufficient ice will be needed to lower the sample temperature to at least 4 °C within 45 min after time of collection. Sample temperature will be maintained at 4 °C until delivered to the laboratory. Care is taken at all times during sample collection, handling and transport to prevent exposure of the sample to direct sunlight. Samples are preserved in the laboratory, if necessary, according to protocol for specific analysis (acidification in most cases).

Field Safety Issues

Proper gloves must be worn to prevent contamination of the sample and to protect the sampler from environmental hazards (disposable polyethylene, nitrile, or non-talc latex gloves are recommended, **however, metals and mercury sample containers can only be sampled and handled using polyethylene gloves as the outer layer**). Wear at least one layer of gloves, but two layers help protect against leaks. One layer of shoulder high gloves worn as a first (inside) layer is recommended to have the best protection for the sampler. Safety precautions are needed when collecting samples, especially samples that are suspected to contain hazardous substances, bacteria, or viruses.

Sample Handling and Shipping

Due to increased shipping restrictions, samples being sent via a freight carrier require additional packing. Although care is taken in sealing the ice chest, leaks can and do occur. Samples and ice should be bagged placed inside a large trash bag inside the ice chest for shipping. Ice should be double bagged to prevent melted ice water from leaking into the sample. The large trash bag can be sealed by simply twisting the bag closed (while removing excess air) and taping the tail down. Prior to shipping the drain plug of the ice chests have to be taped shut. Leaking ice chests can cause samples to be returned or arrive at the lab beyond the holding time.

Although glass containers are acceptable for sample collection, bubble wrap must be used when shipping glass.

Chain of Custody (COC) Forms

Every shipment must contain a complete Chain of Custody (COC) Form that lists all samples collected and the analyses to be performed on these samples.

Make sure a COC is included for every laboratory, every time you send a shipment of samples. Electronic COCs can also be emailed to the various laboratories but must be sent before the samples arrive at their destinations.
Include region and trip information as well as any special instructions to the laboratory on the COC.

The original COC sheet (not the copies) is included with the shipment (insert into ziplock bag) One copy goes to the sampling coordinator, and the sampling crew keeps one copy.

Samples collected should have the salinity (in ppt), depth of collection, and date/time collected for each station on every COC.

Write a comment on this form, if you want to warn the laboratory personnel about possibly hazardous samples that contain high bacteria, chlorine or organic levels.

Field QC Samples for Water Analyses

Field duplicates are currently submitted at an annual rate of 5%. Field travel blanks are required for volatile organic compounds at a rate of one per cooler shipped. Field blanks are required for trace metals (including mercury and methyl mercury), DOC, and volatile organic compounds in water at a rate of 5%. See Appendix C of the SWAMP QAMP for detailed Field QC requirements.

Field Site Data Sheets

Each visited field site requires a field observation completed SWAMP Field Data Sheet, even if no samples are collected (i.e. at a site which is found to be dry). If water and/or sediment samples are collected, all elements of the SWAMP Field Data Sheet must be completely filled out.

General Pre-Sampling Procedures

Instruments. All instruments must be in proper working condition. Make sure all calibrations are current. Multi-probe sondes should be pre-calibrated every morning prior to sampling and post-calibrated within 24 h of the original calibration. Conductivity should also be calibrated between stations if there is a significant change in salinity. Dissolved oxygen sensors should be re-calibrated if there is a 500 ft

change in elevation.

Calibration Standards. Pack all needed calibration standards.

Sample Storage Preparations. A sufficient amount of cube ice, blue ice and dry ice as well as enough coolers of the appropriate type/size must be brought into the field, or sources for purchasing these supplies identified in advance.

Sample Container Preparation. After arriving at the sample station, pack all needed sample containers for carriage to the actual collection site, and label them with a pre-printed label containing Station ID, Sample Code, Matrix info, Analysis Type info, Project ID and blank fields for date and time (if not already pre-labeled).
Safety Gear. Pack all necessary safety gear like waders, protective gloves and safety vests.

Walk to the site. For longer hikes to reach a sample collection site, large hiking backpacks are recommended for transport of gear, instruments and containers. Tote bins can be used, if the sampling site can be accessed reasonably close to the vehicle.

GPS. At the sampling site, compare/record reconnaissance GPS reading with current site reading and note differences. GPS coordinates should be in Decimal Degrees (e.g. 38.12345 -117.12345).

COLLECTION OF WATER SAMPLES FOR ANALYSIS OF CONVENTIONAL CONSTITUENTS

In most streams, sub-surface (0.1 m below surface) water is representative of the water mass. A water sample for analysis of conventional constituents is collected by the grab method in most cases, immersing the container beneath the water surface to a depth of 0.1 m. Sites accessed by bridge can be sampled with a sample container-suspending device. Extreme care must be taken to avoid contaminating the sample with debris from the rope and bridge. Care must also be taken to rinse the device between stations. If the centroid of the stream cannot be sampled by wading, sampling devices can be attached to an extendable sampling pole.

In some cases, depth-integrated sampling is required, as requested by Regional Boards. This is useful when lakes or rivers are stratified and a sample is wanted that represents the entire water column. Depth-integrated sample collection is explained later in this document.

Conventional Water Constituents, Routinely Requested in SWAMP

Chloride, sulfate, nitrite, nitrate (or nitrate+nitrate), ortho-phosphate, fluoride, total phosphorus, ammonia, TKN, alkalinity, chlorophyll a.

Conventional Water Constituents, Occasionally Requested in SWAMP

Total Suspended Solids (TSS) or Suspended Sediment Concentration (SSC), Total Dissolved Solids (TDS--especially if total metals requested), Total Organic Carbon (TOC), Dissolved Organic Carbon (DOC), hardness (if trace metals analysis is requested).

Conventional Water Constituents Sample Volume

Due to the potential for vastly different arrays of requested analyses for conventional constituents, please refer to table at the end of this document, as well as the Sample Handling Requirements Tables in Appendix C of the QAMP, for information on the proper volume to collect for the various types of analyses.

Conventional Water Constituents Sample Container Type

Due to the potential for vastly different arrays of requested analyses for conventional constituents, please refer to table at the end of this document, as well as the Sample Handling Requirements Tables in Appendix C of the QAMP, for information on the proper type of sample containers.

Chlorophyll a Syringe Sample Method

Chlorophyll a syringe method: Chlorophyll a is sampled by forcing water with a 60-mL syringe through a filter holder containing a 25-mm glass microfiber filter. The 60-mL syringe and an in-line filter holder are rinsed three times with the ambient water before filtration. The syringe is then filled with 60 mL of ambient water. The filter holder is then removed and a 25-mm glass microfiber filter is placed inside. The filter holder is then screwed onto the syringe and the ambient water is then flushed through the filter. The filter holder is removed every time more water needs to be drawn into the syringe. The process is then repeated until the desired amount of Chlorophyll a is present (usually 60 to 360 mL depending on the water clarity). When filtering is complete the filter holder is opened and the filter is removed with tweezers without

touching the Chlorophyll a. The filter is then folded in half, then again, in half with the Chlorophyll a inside the folds. The folded filter is then wrapped in aluminum foil and placed in an envelope labeled with the site information and the volume filtered. The envelope is then immediately placed on dry ice until transferred to the lab.

Collection of Water Samples for Analysis of Trace Metals (Including Mercury)

When deciding to measure total and dissolved metals in water the purpose of the sampling must be considered. Water quality standards for the protection of aquatic life are determined for the dissolved form of heavy metals in most cases, although this, too, can vary within different Basin Plans for different regions. The exception to routinely conducting dissolved metals analyses is usually mercury (and often selenium). Water quality standards usually apply to the total form of mercury (and often selenium), and not the dissolved form of these elements. Several regions are interested in conducting total metals analyses, in order to address specific issues. In order to budget inputs, transport, and accumulation of metals, it is necessary to know the concentration of total metals in the water column, sediments, effluent, etc. Sample collection for trace metals and mercury in water requires "Clean Hands/Dirty Hands" methodology.

Metals-in-water: General Information

Unless otherwise requested to collect for total metals analysis, dissolved metals are collected for all elements with the exception of mercury. Metals-in-water samples should **not** be collected during periods of abnormally high turbidity if at all possible. Samples with high turbidity are unstable in terms of soluble metals, and it is difficult to collect a representative grab sample. Special study sampling, however, may be an exception. For example, wet weather sampling is likely to include some samples with high turbidity.

Metals-in-water: Sample Collection Depth

Collect a metals sample from a depth of 0.1 m using a sub - surface grab method, or at discrete depths using a depth-integrated sampling method with a peristaltic pump (described further down). In most streams, sub-surface water is representative of the water mass. For the purpose of determining compliance with numerical toxic substance standards, a sample taken at the surface is adequate.

Metals-in-water: Sample Volume

Refer to table at end of this document, as well as Sample Handling Requirements Tables in Appendix C of the QAMP, for specific information on the proper volume to collect for trace metals analyses. Generally, for procedures most commonly used for analysis of metals in water (total or dissolved metals); one 60-mL polyethylene container is filled with the salinity recorded on the field data sheet and COC. Generally, for the procedures most commonly used for analysis of mercury in water (whether total or dissolved), one 250-mL glass or teflon container is filled, regardless of the salinity. All containers are pre-cleaned in the lab using HNO_3.

Metals-in-water:

Sampling Equipment

The method of choice for the collection of water samples for trace metals analysis in small, wadeable streams is the grab method, where the sampler submerges the sample bottle or syringe beneath the surface of the water until filled. The procedure for filtration of water samples for trace metals (including mercury) analysis must be performed within the 48-h maximum holding time (as well as acid preservation), and with extreme care to avoid contamination of the water sample. Considering these factors, it is best to use a **field** filtration system, such as a set-up with peristaltic pump with in-line filter, or a set-up with a syringe filter, if filtered water is required. Samples are pumped and/or filtered directly into the sample container. This minimizes contamination by using no intermediate sampling device. Samples can also be filtered in lab if need be Un-powdered (no-talc) polyethylene gloves are always worn during sampling for metals-in-water.

Depth-integrated sampling is useful when lakes or rivers are stratified and a representative sample is wanted which represents the entire water column. The method involves a peristaltic pump system with enough Teflon tubing to pump at the desired depth with an inline filter. Alternatively, mercury and metal samples can be filtered in the laboratory as long as they are filtered within the 48-hr maximum holding time and filter equipment blanks are analyzed for five percent of all cleaned equipment.

Equipment Preparation

It is best if the metals-in-water sampling materials are prepared by a laboratory that can guarantee contamination-free sampling supplies. If a laboratory assembles a Metals-in-Water Sample Collection Kit, it should contain the following items packaged together **for each sample**:

- Tubing with an in-line filter (disposable, 0.45 µm) attached for dissolved metals-in-water sampling. This same tubing is used for total metals-in-water samples without filter. If an in-line pumping system is not used, an acid cleaned syringe and filter are packed.
- Sample containers- polyethylene for total and dissolved samples and blanks; Glass or Teflon for total and dissolved mercury.
- Acid preservation is performed in the laboratory.
- Metals-free DI water (for blanks).

- Powder-free polyethylene gloves

If a laboratory is not assembling collection kits, individuals should take care to keep containers in the original packaging. When removed from the box, sample containers are placed in plastic bags (ziplock bags). Although filters come individually wrapped, they should also be stored in new ziplock bags to avoid possible contamination.

The filtering equipment is pre-cleaned according to laboratory protocol. Clean tubing is put into clean containers, such as large ziplock bags. Metals-free filter cartridges with the capacity to filter several liters are commercially available. Equipment blanks are run at the laboratory on batches of metals-in-water sampling equipment prior to their distribution to field staff. One to two liter containers with metals-free deionized water are taken into the field for travel blanks. Metals-free deionized water is supplied by the laboratory performing metals analysis. The deionized water containers are kept clean and dust-free on the outside by wrapping in two plastic bags.

Dissolved and Total Metals-in-Water: Detailed Collection Techniques

- ***Sub-Surface Grab Method***
- ***Syringe Filtration Method (for sub- surface collection)***
- ***Peristaltic Pumping Method (Using Tubing/In-line Cartridge Filters)for sub- surface collection or for depth-integrated collection***

Metals-in-water Sample Collection:

Sub-Surface Grab Method

Unfiltered Samples (for total metals analysis, if requested, and for mercury almost always, unless otherwise requested): Some samples can be sampled directly from the ambient water either by wading into the stream and dipping bottles under the surface of the water until filled, or by sampling from a boat and dipping the bottle under the surface of the water until it is filled. The bottles are cleaned according to laboratory protocol. It is very critical that all the acid is rinsed out of the bottles before the samples are taken. Personnel involved in field sample collection/processing wear polyethylene gloves. The laboratory pre-cleaned glass or Teflon™ 250 mL (for mercury) or polyethylene 60 mL (for

metals) sample bottles are taken from the double-wrapped plastic bags using "Clean Hands/Dirty Hands" techniques. The dirty hands person opens the first bag, and the clean hands person opens the inner bag around the bottle. The clean hands person then removes the bottle from the inner bag. The clean hands person dips the bottle into the ambient water, with the cap on, to approximately 0.1 m (avoiding disturbing surface scums), placing the cap back on the bottle before being removed from the water, rinses the bottle five times with ambient water, making sure the threads of the bottle get rinsed as well, and fills the bottle to the top. The lid is secured and the bottle is put back into the inner clean bag and sealed by the clean collector. The dirty hands collector then seals the outer bag.

Metals-in-water Sample Collection:

Syringe Filtration Method (for sub-surface collection)

Filtered Samples (for dissolved metals analyses): Sub-surface water samples are filtered for dissolved trace metals analysis (not for mercury, however, in almost all cases) using the following syringe filtration method.

The syringe (60 cc size, pre-cleaned in the laboratory) and in-line filter are pre-packed in two ziplock bags. The syringe and filter are taken out of the bags using "Clean Hands/Dirty Hands" technique, as previously described. The sub-surface water sample is collected by 1) wading out into the centroid portion of the stream, or by leaning over the edge of the boat, and aspirating water into the syringe, filling and rinsing the syringe five times with ambient water; 2) attaching the filter onto the syringe and filling the syringe body; 3) rinsing the filter with a few milliliters of the sample; 4) rinsing the sample bottle five times with the filtered ambient water; and 5) extruding the sample through the syringe filter and completely filling each bottle. The bottles are taken out of and put back into their bags using "Clean Hands/Dirty Hands".

Metals-in-water Sample Collection--

Peristaltic Pump

The basic "Clean Hands/Dirty Hands" technique is also applied in the use of a peristaltic pump with an in-line filter cartridge for metals-in-water sample collection. Dirty Hands removes the plastic cover from the end of the pump tubing and inserts the tubing into the sampling container. Dirty Hands holds the tubing in place. The in-line cartridge filter is attached to the outlet end of the tubing.

Clean Hands takes the plastic cover off the other end of the tubing. Dirty Hands turns on the pump and flushes l L of ambient water through the tubing to purge it for dissolved

metals.

Clean Hands removes the cap from the sample bottle and uses the pump to fill it with ambient water. Clean Hands puts the cap back on the bottle and places it in the plastic bag.

Metals-in-water Sample Collection:

Depth-Integrated Sampling, using In-line Cartridge Filter and Peristaltic Pump

Preparation for Depth-integrated sample collection:
Depth-integrated sampling is useful when lakes or rivers are stratified, and a representative sample is wanted that represents the entire water column to the extent possible. The method utilized to date for SWAMP involves a peristaltic pump system with enough Teflon tubing to pump from the desired depth. Regional Boards must request depth-integrated sampling.

The tubing set consists of a small length of CFLEX tubing that fits in the peristaltic pump, with an appropriate length of Teflon tubing on the suction side of the pump and a 3-ft section of Teflon tubing on the discharge side of the pump.

The tubing set is pre-cleaned in 10% reagent grade HCL at the laboratory, and to date in SWAMP, a new pre-cleaned tubing set is used for each site. However, the same peristaltic tubing set can be used at multiple sites, as long as it has been cleaned in the field between stations, according to protocol as outlined below. If this is to be done, however, and Dissolved or Total Organic Carbon samples are collected, equipment blanks should be collected at each site until it is determined that the blanks are acceptably low.

The field cleaning procedure for tubing that is to be re-used is:

- Pump phosphate free detergent through tubing.
- Pump 10% HCL through tubing.
- Pump methanol through tubing.
- Pump 1 l of blank water (Milli-Q) through.

All reagents must be collected in appropriate hazardous waste containers (separated by chemical), and transport, as well as disposal, must follow appropriate local, state, and federal regulations.

If a field blank is needed, collect it after the 1 L of blank water is pumped through. Pump the amount of ambient water

equivalent to 3 times the volume of the tubing before sampling the next site.

Filtered and Unfiltered Samples, Depth-integrated:
It is recommended to attach the tubing to a line with depth measurement markers (preferably in meters). At the end of this line should be a trace metal-safe weight, which hangs about one meter below the tubing end, avoiding any sediment intake from the bottom of the water column with the pump tubing.

At the site, Dirty Hands sets up the pump, while Clean Hands takes a bottle from the plastic bag and places it in a container holder or on a clean surface. A container holder can be anything trace metal clean that supports the bottle, freeing up the collector's hands. Clean Hands takes the outlet-end of the tubing (with the in-line filter cartridge attached) out of the bag, and places it in the peristaltic pump head. The outlet end is long enough to allow easy bottle filling; the other end is long enough to easily reach beneath the water surface and to the desired depth. Dirty Hands closes the pump head, locking the tubing in place.

Make sure that all bottles are filled with a depth-integrated water sample. This can be accomplished by dividing the total vertical length of the water column into 2 to 10 equal intervals, and sampling each interval equally, filling the bottles at each depth proportional to the number of intervals sampled. For example, if 10 intervals are sampled, every bottle is filled $1/10^{th}$ full at each depth sampled. A very common method of dividing the water column is by first determining the depth of the thermocline. Samples are taken at the midpoint between the surface and the thermocline, at the midpoint between the top of the thermocline and the bottom of thermocline, and at the midpoint between the bottom of the thermocline and just above the bottom of the water column. For these methods, all containers have to be filled at the same time. Note the number of intervals sampled on the data sheet.

When filling bottles, Clean Hands immerses the intake tube directly into the water at the appropriate depth, and Dirty Hands operates the pump to flush the tubing with a minimum of 1L of ambient water through the tubing and filter.

Clean Hands removes the cap from the sample bottle, holds

the tubing outlet with the in-line filter cartridge over the container opening (without touching the container), and allows the container to fill. The container is filled and rinsed five times with ambient water, and is then filled to the top for the actual sample. Clean Hands puts the cap back on the bottle, and places the bottle back it in the plastic bag. Whenever Clean Hands touches the boat or equipment, which may be contaminated, gloves should be changed immediately.

(Note for Unfiltered samples: *If an unfiltered sample is required for total metals, total mercury, conventional constituents, toxicity, or synthetic organics, the same procedure is used as described above, except the filter is detached from the end of the tubing before filling the bottles.)*

When sampling is finished, the tubing is brought to the surface, clean water (Milli-Q or deionized) is pumped through system, and the tubing is stored in a polyethylene bag.

The tubing set can be used at multiple sites, as long as it has been cleaned in the field between stations (see field cleaning procedure above). However, if Dissolved or Total Organic Carbon samples (in water) are collected, equipment blanks should be collected at enough sites until it is determined the blanks are appropriate.

Metals-in-water Sample Collection:

Collecting the Sample:

Composite Bottle

The sample collection methodologies are identical to those described above except the sample is collected first into a composite bottle(s). The sample is collected in an amber glass 4-L bottle for mercury and methyl mercury, and a 4-L polyethylene bottle for other trace metals. The compositing bottle is cleaned according to SWAMP SOP.SC.G.1. It is very critical that all the acid is rinsed out of the bottle and that the bottle is rinsed with sample water (five times) before the sample is taken. The sample is collected by the grab or pumping method after being rinsed five times with ambient water and is brought inside the water quality vehicle or sampling box for processing. Personnel involved in sample processing don polyethylene gloves. During sampling the dirty hands person opens the bag holding the composite bottle and opens the outer plastic bag. The clean hands person opens the inner plastic bag, removes the bottle and holds the bottle while the Dirty Hands sampler controls the flow of water

through the pump into the bottle.

Preparing sample aliquots from a composite bottle into smaller sample bottles using an inline pump and filter:

The dirty hands person opens the first bag, and the clean hands person opens the inner bag around the composite bottle. The clean hands person then removes the bottle from the inner bag and places the bags and the bottle in a designated clean place.

This process is repeated until all sample bottles are lined up on the clean bench with their tops still on.

The top of the bottles are loosened so that they fit very loosely on top of the bottles so the clean hands person can remove the caps and pour or pump water into the bottles easier.

The clean hands person shakes the 4-L sample in a steady and slow up and down motion for two full minutes.

Samples that are not to be filtered (including TSS/SSC) are subsampled out of the bottle by pouring out of the large compositing bottle into the sample bottles. The compositing bottle is shaken for 15 s between these subsamples.

Each sample bottle is rinsed five times with ambient water before filling.

For the clean pumping system setup procedure, see above.

(The equipment or field blank is processed exactly like a sample following the same steps.)

The clean end of the tubing used for suction is placed into 1 L bottle. Approximately 750 mL of Milli-Q are then pumped through the system to purge any residual contamination.

The 250-mL sample bottles are then filled to the neck and capped as soon as possible.

Note: if volatile organics are to be collected they should be pumped directly into the sample containers before the compositing procedure.

Metals-in-water: Short-term Sample Preservation

After collecting the sample, the double-bagged container is placed in another plastic bag for shipping, and placed on ice in the ice chest, cooled to 4 °C. This is to prevent possible contamination from other samples in the ice chest. Metals-in-water samples are acid-preserved in the lab.

Metals-in-water: Sample Container Label

Label each outer sample-bag with the station ID, sample code, matrix type, analysis type, project ID, and date and time of collection.

Metals-in-water: Field Equipment Blank

Pumping Method. If required, field blanks are collected at the last site of a sampling trip, with the same tube and filter used to collect the last dissolved metals-in-water sample of the day (before the ambient sample is collected); and with the tube used for the last total metals-in-water sample of the day. If each sample is taken using a new set of tubing, a separate tubing-set should be used for the blank.

The same Clean Hands/Dirty Hands collection techniques are followed for the field blank as the samples, pumping trace metal-free water from a clean container supplied by the laboratory.

Syringe Method. If required, field blanks are collected in much the same way as in the pumping method. "Clean Hands/ Dirty Hands" techniques are used. The syringe is taken out of the double bags, deionized water is aspirated into the syringe, syringe is rinsed five times with ambient water, the filter is attached, and the blank water is extruded into a sample bottle. A minimum of one blank per trip is taken, if required.

Grab Method. Bottles full of deionized water or Milli-Q are opened at the site for the same length of time the sample bottles are open.

COMPANION SAMPLES FOR METALS-IN-WATER

A hardness analysis should be requested by the Regional Water Control Board whenever metals-in-water are to be analyzed from an inland (freshwater) site. Estuarine/marine sites do not require hardness analysis.

If a total metals sample is collected, it is recommended to submit a sample for total suspended solids/suspended sediment concentration (TSS/SSC) in a companion sample for "conventionals in water".

Hexavalent Chromium

Very rarely, a request may be made for conducting hexavalent chromium analysis in water samples. Acidification alters the hexavalent form of chromium. A separate (un-acidified) sample must be submitted if hexavalent chromium is to be analyzed. Filter and submit a minimum of 500 mL water. The sample is collected in a DI-water-rinsed plastic or glass container, placed on ice, and shipped to the lab in time for analysis to begin within 24 h of collection. The lab must be notified when a hexavalent chromium sample will arrive. Hexavalent chromium is not usually analyzed on unfiltered samples.

FIELD QC SAMPLE COLLECTION REQUIREMENTS FOR METALS-IN-WATER

In order to assess contamination, "blanks" are submitted for analysis. Special projects may have other requirements for blanks. The same group of metals requested for the ambient samples are requested for the blank(s). Run a blank for each type of metal sample collected. Blanks results are evaluated (as soon as available) along with the ambient sample results to determine if there was contamination or not. See Appendix C of the QAMP for MQO's regarding frequency and types of field QC samples.

Field Equipment Blank (Ambient Blank)	Submit an equal volume (equal to the ambient sample) of metals-free deionized water that has been treated exactly as the sample at the same location and during the same time period. Use the same methods as described above (Grab sample, pumping method, syringe method). At least one ambient blank per field trip is required each for trace metal and Mercury samples in water. *If contamination is detected in field equipment blanks, blanks are required for every metals-in-water sample until the problem is resolved*.
Laboratory Equipment Blank	Laboratory Equipment Blanks for pumping and sampling equipment (Metals-in-Water Sample Collection Kits and Syringe Filtration Kits) are run by the laboratory that cleans and distributes the collection materials. It documents that the materials provided by the laboratory are free of contamination. When each batch of tubes, filters, bottles, acid and deionized water are prepared for a sampling trip, about five percent of the Mercury sampling materials are chosen for QC checks. Trace metal equipment needs to be subjected to an initial blank testing series. If these blanks are acceptable only occasional re-testing is required for TM equipment. The QC checks are accomplished by analyzing metals-free water which has been pumped through the filter and tube; collected

in a sample container; and preserved.

Field Duplicates

Five percent Field Duplicates are submitted every year. (If less than 20 samples are collected during an event, submit one set of duplicates per event.)

Collection of Water Samples for Analysis of Synthetic Organic Compounds

Collect organic samples at a depth of 0.1 m by submerging the sample container by hand. If depth-integrated sampling is required, use the in-line peristaltic pump methodology described previously. Since organic compounds tend to concentrate on the surface of the sampling device or container, the sampling device and sample container are ***not*** to be rinsed with ambient water before being filled.

Sample Containers and Collection
Also refer to Appendix C of the SWAMP QAMP for a list of sample volumes and containers.

Pesticides/ Herbicides	The sample container for pesticides and herbicides is a new, clean, unused amber glass jar with a Teflon-liner inside the cap. Collect one liter of water for each of the three sample types (Organophosphorus Pesticides, Organochlorine Pesticides and Chlorinated Herbicides). **EACH ANALYSIS TYPE REQUIRES A SEPARATE JAR**. Minimize the air space in the top of the jar. Preserve immediately after collection by placing on ice out of the sunlight.
Semi-volatile Organics	The sample container for semi-volatile organics must also be new, clean, unused amber glass bottles with a Teflon-liner inside the cap, and pre-rinsed with pesticide-grade hexane, acetone, or methylene chloride. Fill jars to the top and place on ice in the dark. In addition to other sample information, label the jar Semi-volatiles.
Volatile Organics: **Volatile Organic Carbon (VOC), Methyl-Tert Butyl Ether (MTBE) and (BTEX)**	The sample containers for volatiles are VOA vials. Fill the 40-mL VOA vials to the top and cap without trapping any air bubbles. If possible, collect directly from the water, keeping the vial under water during the entire collection process. To keep the vial full while reducing the chance for air bubbles, cap the vials under the water surface. Fill one vial at a time and preserve on ice. The vials are submitted as a set. If the vial has been pre-acidified for preservation, fill the vial quickly, without shaking using a separate clean glass jar. Fill the vial till the surface tension builds a meniscus, which extends over the top end of the vial, then cap tightly and check for bubbles by turning the vial on its head. Ensure that the pH is less than 2. If the water may be alkaline or have a significant buffering capacity, or if there is concern that pre-acidified samples may have the acid wash out, take a

few practice vials to test with pH paper. It may take more than two drops, and it will then be known how to preserve the other samples that are being submitted to the lab. If an alternative method has proven successful, continue with that method.

Note: If vigorous foaming is observed following acidification, discard that sample and collect another set. Do not acidify the second set. Mark the sample clearly "not acidified" and the lab will run them immediately. Holding time is 14 days with acid, 24 h without acid.

Collect three VOA vials, if VOC, MTBE and BTEX are required, two vials, if only VOC is required and two vials, if only MTBE and BTEX are require. The vials may be taped together to keep them together.

Perchlorate

Surface water samples for perchlorate should be collected in a new unused polyethylene or glass container. Perchlorate samples should be placed immediately on ice to maintain temperature at 4 ^{o}C. The sample holding time is 28 days, under refrigeration.

Sample Treatment in Presence of Chlorine

(NOTE: This treatment has not been performed in SWAMP, but may be in the future, or if a known or suspected chlorine residual is suspected and this information is made known by a Regional Board SWAMP contact beforehand.)

If in stream chlorine residual is suspected, measure the chlorine residual using a separate water subsample. Free chlorine will oxidize organic compounds in the water sample even after it is collected. If chlorine residual is above a detectable level, (i.e., the pink color is observed upon adding the reagents) immediately add 100 mg of sodium thiosulfate to the pesticides, herbicides, semivolatiles and VOA samples; invert until sodium thiosulfate is dissolved. Record the chlorine residual concentration in field logbook. If chlorine residual is below detectable levels, no further sample treatment necessary.

VOA Trip Blank

Submit one Trip Blank for VOA samples (2- 40 mL VOA vials) for each sampling event. Trip Blanks are prepared in advance just before the sampling trip and transported to the field. Ask the laboratory for DI water and specify that it is for a VOA trip blank. VOA blanks require special purged water. Trip blanks demonstrate that the containers and sample handling did not introduce contamination. The trip blank vials

are never opened during the trip.

Field QC Samples If required, field Duplicates and field blanks are submitted at a rate subject to the discretion of the project manager. Refer to Appendix C of the SWAMP QAMP for details on required blanks and duplicates.

BACTERIA AND PATHOGENS IN WATER SAMPLES

Summary of Collection Procedure (Based on EPA water quality monitoring procedures)

Make sure the containers are sterilized; either factory-sealed or labeled.

Whirl-pak® bags

- Label the bottle as previously described for SWAMP.
- Tear off the top of the bag along the perforation above the wire tab just prior to sampling. Avoid touching the inside of the bag. If you accidentally touch the inside of the bag, use another one.
- If wading into the stream, try to disturb as little bottom sediment as possible. Be careful not to collect water that has sediment from bottom disturbance. Stand facing upstream. Collect the water sample on your upstream side, in front of you. You may also attach your bottle to an extension pole to sample from deeper water.
- If taking sample from a boat, carefully reach over the side and collect the water sample on the upstream side of the boat.
- Hold the two white pull-tabs in each hand and lower the bag into the water on your upstream side with the opening facing upstream. Open the bag midway between the surface and the bottom by pulling the white pull-tabs. The bag should begin to fill with water. You may need to "scoop" water into the bag by drawing it through the water upstream and away from you. Fill the bag no more than 3/4 full.
- Lift the bag out of the water. Pour out excess water. Pull on the wire tabs to close the bag. Continue holding the wire tabs and flip the bag over at least 4-5 times quickly to seal the bag. Don't try to squeeze the air out of the top of the bag. Fold the ends of the wire tabs together at the top of the bag, being careful not to puncture the bag. Twist them together, forming a loop.
- If the samples are to be analyzed in the lab, place them in a cooler with ice or cold packs for transport to the lab.

Screw cap containers

- Label the bottle as previously described for SWAMP.
- Remove the cap from the bottle just before sampling. Avoid touching the inside of the bottle or cap. If you accidentally touch the inside, use another bottle.
- If wading into the stream, try to disturb as little bottom sediment as possible. Be careful not to collect water that has sediment from bottom disturbance. Stand facing upstream. Collect the water sample on your upstream side, in front of you. You may also attach your bottle to an extension pole to sample from deeper

water.

- If taking sample from a boat, carefully reach over the side and collect the water sample on the upstream side of the boat.
- Hold the bottle near its base and plunge it (opening downward) below the water surface. If you are using an extension pole, remove the cap, turn the bottle upside down, and plunge it into the water, facing upstream. Collect a water sample 2" beneath the surface. You can only use this method if the sample bottles do not contain sodium thiosulfate.
- Turn the bottle underwater into the current and away from you. In slow moving stream reaches, push the bottle underneath the surface and away from you in an upstream direction.
- Alternative sampling method: In case the sample bottle contains preservatives/chlorine removers (i.e. Sodium-Thiosulfate), it cannot be plunged opening down. In this case hold the bottle upright under the surface while it is still capped. Open the lid carefully just a little to let water run in. Fill the bottle to the fill mark and screw the lid tight while the bottle is still underneath the surface.
- Leave a 1-in. air space so that the sample can be shaken just before analysis. Recap the bottle carefully, remembering not to touch the inside.
- If the samples are to be analyzed in the lab, place them in a cooler with ice or cold packs for transport to the lab.

Pouring from another clean bottle

- Due to different sampling conditions (high turbidity, rough water etc.) it is sometimes easy to pour water from another clean bottle into the bacteria bottle. This helps to make sure that the sample water is only being filled to the desired line and no overfilling occurs.

TOXICITY IN WATER

Sample Collection

Using the standard grab sample collection method described previously for water samples, fill (for typical suite of water toxicity tests conducted) the required amount of 2.25-L amber glass bottles with water, put on ice, and cool to 4 °C. Label the containers as described above and notify the laboratory of the impending sample delivery, since there is a 48-hr maximum sample hold time. Sample collection must be coordinated with the laboratory to guarantee appropriate scheduling.

Summary of Sample Container, Volume, Initial Preservation, and Holding Time Recommendations for Water Samples

Parameters for Analysis in WATER Samples	Recommended Containers (all containers pre-cleaned)	Typical Sample Volume (mL)	Initial Field Preservation	Maximum Holding Time (analysis must start by end of max)
Conventional Constituents in Water				
Alkalinity	Polyethylene bottles (see **NOTE(1)** below)	100 mL	Cool to 4°C, dark	14 days at 4°C, dark
Chloride (Cl), Sulfate (SO_4) and Fluoride (F)	Polyethylene bottles (see **NOTE(1)** below)	300 mL	Cool to 4°C, dark	28 days at 4°C, dark
Ortho-phosphate (OPO_4)	Polyethylene bottles (see **NOTE(1)** below)	150 mL	Cool to 4°C, dark	48 h at 4°C, dark
Nitrate + Nitrite (00630) ($NO_3 + NO_2$)	Polyethylene bottles (see **NOTE(1)** below)	150 mL	Cool to 4°C, dark	48 h at 4°C, dark
Total Keldjahl Nitrogen (TKN)	Polyethylene bottles (see **NOTE(1)** below)	600 mL	Cool to 4°C, dark	Recommend: 7 days Maximum: 28 days Either one at 4°C, dark
Total Dissolved Solids (TDS)	Polyethylene bottles (see **NOTE(1)** below)	1000 mL	Cool to 4°C, dark	7 days at 4°C, dark
Ammonia (NH_3)	Polyethylene bottles (see **NOTE(1)** below)	500 mL	Cool to 4°C, dark	48 h at 4C dark; if acidify, 28 days at 4°C, dark
Total Phosphorus (TPO_4)	Polyethylene bottles (see **NOTE(1)** below)	300 mL	Cool to 4°C, dark	28 days at 4°C, dark
(1)NOTE: **The volume of water necessary to collect in order to analyze for the above constituents is typically combined in four 1-L polyethylene bottles, which also allows enough volume for possible re-analysis and for conducting lab spike duplicates. This is possible since the same laboratory is conducting all of the above analyses; otherwise, individual volumes apply.**				
Total Organic Carbon (TOC), Dissolved Organic Carbon (DOC)	125 mL amber glass vial 250 ml amber for TOC/DOC	125 mL for TOC only 250 mL for TOC/DOC	Cool to 4°C, dark Cool to 4°C, dark	28 days at 4°C, dark 28 days at 4°C, dark
Total Suspended Solids (TSS)	250 mL plastic bottle	250 mL	Cool to 4°C, dark	7 days at 4°C, dark
Suspended Sediment Concentration (SSC)	125 mL polyethylene bottle	Up to 125ml depending on turbidity of water	Cool to 4°C, dark	7 days at 4°C, dark
Chlorophyll *a* **Pheophytin *a***	1-L amber polyethylene bottle Aluminum Foil, GFC Filters	1000 mL (one bottle)	Cool to 4°C, dark	Keep at 4°C, dark, but must filter within 48 h.

Parameters for Analysis in WATER Samples	Recommended Containers (all containers pre-cleaned)	Typical Sample Volume (mL)	Initial Field Preservation	Maximum Holding Time (analysis must start by end of max)
Chlorophyll a **Pheophytin *a***		20-420 mL	Freeze, Dry ice	Filters may be stored frozen up to 30 days.
Non-Routine Compounds in Water Samples				
OIL AND GREASE	1-L glass jar with Teflon lid-liner, rinsed with hexane or methylene chloride	1000 mL (one jar)	Add 2 mL conc. H_2SO_4 to pH <2; cool to 4°C, dark.	28 days at 4°C, dark
PHENOLS	1-L glass jar with Teflon lid-liner	1000 mL (one jar)	Add 2 mL conc. H_2SO_4 to pH <2; cool to 4°C, dark.	28 days at 4°C, dark
CYANIDE	1-L cubitainer	1000 mL (one cubitainer)	Add 2 mL 1:1 NaOH to make pH > 12; Add 0.6 g ascorbic acid if residual Cl present. Cool to 4°C, dark.	14 days at 4°C, dark
BIOCHEMICAL OXYGEN DEMAND (BOD)	4-L cubitainer	4000 mL (one cubitainer)	Cool to 4°C, dark. Add 1g FAS crystals per liter, if residual Cl present.	48 h at 4°C, dark
CHEMICAL OXYGEN DEMAND (COD)	1-L cubitainer	110 mL (one cubitainer)	Add 2 mL conc. H_2SO_4 to make pH <2. Cool to 4°C, dark.	28 days at 4°C, dark
Trace Metals in Water Samples				
DISSOLVED METALS (except Dissolved Mercury)	60 mL polyethylene bottle, pre-cleaned in lab using HNO_3	60 mL (one bottle)	Filter at sample site using 0.45 micron in-line filter, or syringe filter. Cool to 4°C, dark. Acidify in lab, within 48 hrs, using pre-acidified container (ultra-pure HNO_3) for pH<2.	Once sample is filtered and acidified, can store up to 6 months at room temperature
DISSOLVED MERCURY	250 mL glass or Teflon bottle, pre-cleaned in lab using HNO_3	250 mL (one bottle)	Cool to 4°C, dark. Filter in lab within 48 h, using bench top Hg filtration apparatus. Acidify in lab within 48 hrs, with pre-tested HCL to	Once sample is filtered and acidified, can store up to 6 months at room temperature

Parameters for Analysis in WATER Samples	Recommended Containers (all containers pre-cleaned)	Typical Sample Volume (mL)	Initial Field Preservation	Maximum Holding Time (analysis must start by end of max)
			0.5%.	
TOTAL METALS (except Total Mercury)	60 mL polyethylene bottle, pre-cleaned in lab using HNO_3	60 mL (one bottle)	Cool to 4°C, dark. Acidify in lab within 48 hrs, with pre-acidified container (ultra-pure HNO_3), for pH<2.	Once sample is acidified, can store up to 6 months at room temperature
TOTAL MERCURY	250 mL glass or Teflon bottle, pre-cleaned in lab using HNO_3	250 mL (one bottle)	Cool to 4°C, dark. Acidify in lab within 48 hrs, with pre-tested HCL to 0.5%.	Once sample is acidified, can store up to 6 months at room temperature.
HEXAVALENT CHROMIUM (filtered)	600 mL plastic or glass bottle	600 mL (one bottle)	Cool to 4°C, dark No acid	Keep at 4°C, dark for up to 24 h; must notify lab in advance.
HARDNESS	200 mL polyethylene or glass bottle	200 mL (one bottle)	Cool to 4°C, dark OR Filter and add 2 mL conc. H_2SO_4 or HNO_3 to pH < 2; Cool to 4°C, dark.	48 h at 4°C, dark 6 months at 4°C, dark
Synthetic Organic Compounds in Water Samples				
VOLATILE ORGANIC ANALYTES (VOA's) including VOC, MTBE and BTEX	40 mL VOA vials	120 mL (three VOA vials)	All vials are pre-acidified (50% HCl or H_2SO_4) at lab before sampling. Cool to 4°C, dark	14 days at 4°C, dark
PESTICIDES & HERBICIDES* □Organophosphate Pesticides □Organochlorine Pesticides □Chlorinated Herbicides **SEMI-VOLATILE ORGANICS*** **POLYCHLORINATED* BIPHEYNYL AND AROCHLOR COMPOUNDS** **TPH, PAH, PCP/TCP***	1-L I-Chem 200-series amber glass bottle, with Teflon lid-liner (per each sample type)	1000 mL (one container) ***Each sample type requires 1000 mL in a separate container**	Cool to 4°C, dark If chlorine is present, add 0.1g sodium thiosulfate	Keep at 4°C, dark, up to 7 days. Extraction must be performed within the 7 days; analysis must be conducted within 40 days.
Toxicity Testing Water Samples				
TOXICITY IN WATER	Four 2.25 L amber glass bottles	9000 mL	Cool to 4°C, dark	48 hrs at 4°C, dark

Parameters for Analysis in WATER Samples	Recommended Containers (all containers pre-cleaned)	Typical Sample Volume (mL)	Initial Field Preservation	Maximum Holding Time (analysis must start by end of max)
Bacteria and Pathogens in Water Samples				
E. Coli	Factory-sealed, pre-sterilized, disposable Whirl-pak® bags or 125 mL sterile plastic (high density polyethylene or polypropylene) container	100 mL volume sufficient for both E. coli and Enterococcus analyses	Sodium thiosulfate is pre-added to the containers in the laboratory (chlorine elimination). Cool to 4°C; dark.	STAT: 6 h at 4°C, dark if data for regulatory purposes; otherwise, 24 hrs at 4C, dark if non-regulatory purpose.
Enterococcus	Factory-sealed, pre-sterilized, disposable Whirl-pak® bags or 125 mL sterile plastic (high density polyethylene or polypropylene) container	100 mL volume sufficient for both E. coli and Enterococcus analyses	Sodium thiosulfate is pre-added to the containers in the laboratory (chlorine elimination). Cool to 4°C; dark.	STAT: 6 h at 4°C, dark if data for regulatory purposes; otherwise, 24 hrs at 4C, dark if non-regulatory purpose.
FECAL COLIFORM	Factory-sealed, pre-sterilized, disposable Whirl-pak® bags or 125 mL sterile plastic (high density polyethylene or polypropylene) container	100 mL volume sufficient for both fecal and total coliform analyses	Sodium thiosulfate is pre-added to the containers in the laboratory (chlorine elimination). Cool to 4°C; dark.	STAT: 6 h at 4°C, dark if data for regulatory purposes; otherwise, 24 hrs at 4C, dark if non-regulatory purpose.
TOTAL COLIFORM	Factory-sealed, pre-sterilized, disposable Whirl-pak® bags or 125 mL sterile plastic (high density polyethylene or polypropylene) container	100 mL volume sufficient for both fecal and total coliform analyses	Sodium thiosulfate is pre-added to the containers in the laboratory (chlorine elimination). Cool to 4°C; dark.	STAT: 6 h at 4°C, dark if data for regulatory purposes; otherwise, 24 hrs at 4C, dark if non-regulatory purpose.

Field Collection Procedures for Bed Sediment Samples

Bed sediment (hereafter termed "sediment") samples are collected after any water samples are collected where water and sediment are taken in the same reach. Care must be taken not to sample sediments that have been walked on or disturbed in any manner by field personnel collecting water samples. Sediment samples are collected into a composite jar, where they are thoroughly homogenized in the field, and then aliquoted into separate jars for chemical or toxicological analysis. Sediment samples for metals and organics are submitted to the respective analytical laboratories in separate glass jars, which have been pre-cleaned according to laboratory protocol.

Sediment chemistry samples give information regarding both trends in contaminant loading and the potential for adverse effects on sediment and aquatic biota. In order to compare samples over time and from site to site, they must be collected in a consistent manner. If a suitable site for collecting sediments cannot be found at a station, sampling personnel should not collect the sediment sample, and should instead attempt to reschedule the sample collection. If this is not possible, make a note so that the missing sample is accounted for in the reconciliation of monitoring events during preparation of sample collection "cruise reports". Sites that are routinely difficult to collect should be considered for elimination or relocation from the sample schedule, if appropriate.

Characteristics of Ideal Sediment Material to be Collected

Many of the chemical constituents of concern are adsorbed onto fine particles. One of the major objectives in selecting a sample site, and in actually collecting the sample while on site, is to obtain recently deposited fine sediment, to the extent possible. Avoid hard clay, bank deposits, gravel, disturbed and/or filled areas. Any sediment that resists being scooped by a dredge is probably not recently deposited fine sediment material. In following this guidance, the collection of sediment is purposefully being biased for fine materials, which must be discussed thoroughly in any subsequent interpretive reporting of the data, in regards to representation of the collected sample to the environment from which it was collected.

Characteristics of an Ideal Site

Quiescent areas are conducive to the settling of finer materials (EPA/USACOE, 1981).

Choose a sampling site with lower hydrologic energy, such as the inner (depositional) side of bends or eddies where the water movement may be slower. Reservoirs and estuaries are generally depositional environments, also.

Selecting the Appropriate Sediment Type for Analysis

Sediment will vary from site to site and can vary between sample events at a particular site.

Streams and Rivers: Sediment collection in flowing streams is often a challenge. In areas of frequent scouring there may not be sufficient sediment for collection during or following periods of high flow. Sediment collection during these times may prove unsuccessful and may have to be rescheduled or cancelled.

When the suspended load in rivers and streams precipitates due to reduction of velocity, most of the resulting sediment will be fine-grained. More often than not, a dredge or mechanical grab device does not function well for collection of sediment in smaller streams. In many cases, sediment will have to be collected using a pre-cleaned polyethylene scoop. Collect the top 2 cm for analysis. Five or more (depending on the volume of sediment needed for conducting analyses) fine-sediment sub-sites within a 100-m reach are sampled into the composite jar.

Reservoirs and Estuaries: Collect the top 2 cm for analysis. Five or more grabs are composited for the sediment sample, depending on the volume of sediment needed for conducting analyses.

GENERAL PROCEDURE FOR COLLECTION OF BED SEDIMENT

After choosing an appropriate site, and identifying appropriate fine-grained sediment areas within the general reach, collect the sample using one or more of the following procedures, depending on the setting:

A. Sediment Scoop Method—Primary Method for Wadeable, Shallow Streams

- The goal is to collect the top 2 cm of recently-deposited fine sediment only.
- Wear gloves and protective gear, in areas of potential exposure hazards, per appropriate protocol (make sure gloves are long enough to prevent water from overflowing gloves while submerging scoop).
- Survey the sampling area for appropriate fine-sediment depositional areas before stepping into the stream, to avoid disturbing possible sediment collection sub-sites.
- Carefully enter the stream and start sampling at the closest appropriate reach, then continue sampling UPSTREAM. Never advance downstream, as this could lead to sampling disturbed sediment.
- Stir, do not shake, collected sediment with a polyethylene scoop for at least 5 min making

sure all sediment is completely homogenized.

- Quickly scoop sediment out of the homogenizing jar into desired sampling jars making sure to stir the sediment in the homogenizing jar in between each aliquot.
- Inspect each individual sediment jar making sure of consistent grain size throughout the entire sample collection.
- Single bag all sediment containers to prevent cross contamination.
- Make sure all containers are capped tightly and stored in a cooler on cube ice at 4 °C.
- Check cooler temperature and record in log book every 8-12 h or whenever sampler suspects that the temperature has not been maintained at 4 °C.

B. Hand Core Method-Alternate method for wadeable shallow streams with fine sediment

- A hand core is used in wadeable streams where there is very fine sediment.
- The hand core sampler consists of a 3-in. diameter polycarbonate core that is 8 inches long. Samplers push the core into the sediment to the desired depth, pull the core out of the sediment, and cap the bottom with a polyethylene core cap or by placing their hand underneath the cap to hold the sediment in place.
- Hand cores are usually measured and marked at 2 cm length so the sampler knows how far to deploy the core into the sediment.
- Sediment is then emptied into a homogenizing jug and aliquoted accordingly.

C. Sediment Grab Method—Primarily for Lake, River, Bridge, and Estuarine Settings (or deeper streams)

Description of sediment grab equipment:

- A mechanical sediment grab is used for the SWAMP bed sediment collection field effort for lake, river, bridge, and estuarine/coastal settings (or deeper, non-wadeable streams).
- The mechanical grab is a stainless steel “Young-modified Van Veen Grab", and is 0.5 m^2 in size.
- The mechanical grab is deployed primarily from a boat, and is used in deeper, non-wadeable waters, such as lakes, rivers, estuaries, and coastal areas.
- It is also deployed by field personnel from land in settings which allow its use: primarily from bridges; from smaller vessels in streams or drainage channels too deep or steep to wade into, but too shallow for a larger boat.

Deploying and retrieving the grab:

- Slowly lower the grab to the bottom with a minimum of substrate disturbance.
- Retrieve the closed dredge at a moderate speed (e.g., less than two feet per second).
- Upon retrieval, open the lids of the sediment grab, examine the sample to ensure that the sediment surface is undisturbed and that the grab sample should not be rejected.

Rejection Criteria—reject the sample if the following are not met:

- Mud surface must not be pressing out of the top of the sampler. If it is, lower the grab more slowly.
- Overlying water must not be leaking out along the sides of the sediment in the grab. This ensures the surficial sediment is not washed out.
- Sediment surface is flat and level in the sampler. If it is not level, the grab has tilted over before closing.

Processing the sediment sample from the grab equipment:

- The water overlying the sediment in the grab is very gently decanted by slightly tipping the grab with the lid closed until the water runs out the top.
- The decanting process should remove all of the overlying water but not remove the surficial sediments. The laboratory reports percent water for the sample, so overlying water is not included in the sample container.
- The sediment is examined for depth of penetration, color and thickness of top aerobic zone, and texture. These observations are recorded in the logbook.
- Collect the top 2 cm from at least five sub samples, and otherwise, exclude the bottom-most layer and composite.
- In streams or other settings with excessive bottom debris (e.g., rocks, sticks, leaves) where the use of a grab is determined to be ineffective (e.g., dredge does not close, causing loss of sediment), samples may be collected by hand using a clean plastic scoop, or by a variety of coring methods, if appropriate for the situation.
- Sediment is handled as described below in the metals and organic sections.

Cleaning the Grab Equipment and Protection from Potential Contaminating Sources:

- The sediment sampler will be cleaned prior to sampling EACH site by: rinsing all surfaces with ambient water, scrubbing all sediment sample contact surfaces with Micro™ or equivalent detergent, rinsing all surfaces with ambient water, rinsing sediment sample contact surfaces with 5% HCl, and rinsing all sediment sample contact surfaces with methanol.
- The sediment grab will be scrubbed with ambient water between successive deployments at ONE site, in order to remove adhering sediments from contact surfaces possibly originating below the sampled layer, thus preventing contamination from areas beyond target sampling area.
- Sampling procedures will attempt to avoid exhaust from any engine aboard any vessel involved in sample collection. An engine will be turned off when possible during portions of the sampling process where contamination from engine exhaust may occur. It is critical that sample contamination be avoided during sample collection. All sampling equipment (e.g., siphon hoses, scoops, containers) will be made of non-contaminating material and will be appropriately cleaned before use. Samples will not be touched with un-gloved fingers. In addition, potential airborne contamination (e.g., from engine exhaust, cigarette smoke) will be avoided.

D. Core Method--alternative for fast-moving, wadeable streams

The core method is used in soft sediments when it is difficult to use the other methodologies. The cores can be used in depths of water from 0 to 10 ft by using a pole deployment device or in deeper water using SCUBA divers. The pole deployment device consists of a pole that attaches to the top of the core. The top of the core is fitted with a one-way valve, which allows the core to be filled with sediment, but when pulled from the sediment catches the sediment within the core. The core is then brought to the surface and the sediments within the core are extruded out the top of the core so that 2 cm of sediment is above the top of the plastic core. The 2 cm of sediment is then sliced off and placed in the homogenizing jar. A new core, homogenizing jar, and device used to slice off the top two cm. are used at each station unless the equipment is cleaned using laboratory protocols.

E. Sediment Grab Method – Primarily used from bridges or for streams with restricted bank access.

Description and sampling procedure for the Eckman sediment grab

- The Eckman grab is 0.2 m^2 in size with a lead "messenger" that triggers the spring loaded doors.
- The primary use is for sampling from bridges or from small vessels in streams or drainage channels too deep or steep to wade into, but too shallow for a larger boat.
- The grab must be cleaned with a Micro™ and tap water rinse before sampling and in-between sample stations.
- To deploy the grab, pull the spring loaded doors open and hook the cables on the actuator plate.
- With a rope, lower the grab to the desired sample reach making sure that the grab has penetrated the sediment. Clip the "messenger" on the rope and release it while maintaining tension on the rope. Pull up the grab once the "messenger" has activated the doors.
- While wearing clean poly gloves, open the top hatch and remove the top 2 cm of sediment with a clean polyethylene scoop. Place the sediment into the homogenizing jug and repeat the sampling process until there is enough desired sediment. See general procedures for processing of bed sediment samples, once they are collected for sediment homogenization and aliquoting into sample jars.

GENERAL PROCEDURE FOR PROCESSING OF BED SEDIMENT SAMPLES, ONCE THEY ARE COLLECTED

Sediment Homogenization, Aliquoting and Transport

For the collection of bed sediment samples, the top 2 cm is removed from the scoop, or the grab, or the core, and placed in the 4-L glass compositing/homogenizing container. The composited sediment in the container is homogenized and aliquoted on-site in the field. The sample is stirred

with a polyethylene scoop for at least 5 min, but longer if necessary, until sediment/mud appears homogeneous. All sample identification information (station numbers, etc.) will be recorded prior to homogenizing and aliquoting.

The sediment sample is then aliquoted, using a clean plastic scoop, into appropriate containers for trace metal chemistry, organic chemistry, and toxicity testing. All sample containers will be packed surrounded by enough ice to keep them cool for 48 h. Each container will be sealed in one large plastic bag to prevent contact with other samples or ice or water.

Metals and Semi-volatile Organics in Sediment
For trace metals and semi-volatile organics, a minimum of three grabs is distributed to the composite bottle and/or sample containers. Mixing is generally done with a polyethylene scoop. Make sure the sample volume is adequate, but the containers do not need to be filled to the top. Seal the jars with the Teflon liner in the lids.

Sediment Conventionals
Sediment conventionals are sometimes requested when sediment organics, sediment metals, and/or sediment toxicity tests are requested for analysis of samples. The collection method is the same as that for metals, semi-volatile organics, and pesticides. Sediment conventionals include: grain size analysis and total organic carbon. These are used in the interpretation of metals and organics in sediment data.

Sample Containers
See "Sediment Sample Handling Requirements" table at end of this document.

Sediment Sample Size
Must collect sufficient volume of sediment to allow for proper analysis, including possible repeats, as well as any requested archiving of samples for possible later analysis. See "Sediment Sample Handling Requirements" Table at end of this document.

Labeling
Label the jars with the station ID, sample code, matrix type, project ID, time, and date of collection, as well as the type of analysis requested (e.g., metals, conventionals, organics, or archives).

Short-term Field Preservation
Immediately place the labeled jar on ice, cool to 4 °C, and keep in the dark at 4 °C until delivery to the laboratory.

Field Notes
Fill out the SWAMP Station Occupation Data Sheet and the Sediment Data Sheet. Make sure to record any field notes that are not listed on the provided data sheets. This information can be reported as comments with the sediment analytical results.

Summary of Sample Container, Volume, Preservation, and Storage Requirements for SWAMP Bed Sediment, Biota, and Tissue Samples (for contaminant analysis)

Parameters for Analysis	Recommended Containers	Typical Sample Volume (mL)	Initial Field Preservation	Maximum Holding Time
Bed Sediment Samples				
Trace Metals, including Hg and As (except for Se--see below)	60-mL I-Chem 300-series clear glass jar with Teflon lid-liner; Pre-cleaned	60 mL (one jar)	Cool to 4 °C, dark, up to 14 days	12 months(1) (-20 °C)
Selenium (separate container required)	60-mL I-Chem 300-series clear glass jar with Teflon lid-liner; Pre-cleaned	60 mL (one jar)	Cool to 4 °C, dark, up to 14 days	12 months(1) (-20 °C)
Synthetic Organic Compounds	250-mL I-Chem 300-series amber glass jar with Teflon lid-liner; Pre-cleaned	500 mL (two jars)	Cool to 4 °C, dark, up to 14 days	12 months(1) (-20 °C)
Sediment TOC	250-mL(3) clear glass jar; Pre-cleaned	125 mL (one jar)	Cool to 4 °C, dark, up to 28 days	12 months(2) (-20 °C)
Sediment Grain Size	250-mL(3) clear glass jar; Pre-cleaned	125 mL (one jar)	Cool to 4 °C, dark, up to 28 days	28 days (4 °C) ***Do not freeze***
Sediment Toxicity Testing	1-L I-Chem wide-mouth polyethylene jar with Teflon lid-liner; Pre-cleaned	2 (two jars filled completely)	Cool to 4 °C, dark, up to 14 days	14 days (4 °C) ***Do not freeze***

(1) Sediment samples for parameters noted with one asterisk (*) may be refrigerated at 4 °C for up to 14 days maximum, but analysis must start within the 14-day period, or the sediment sample must be stored frozen at minus (-) 20 °C for up to 12 months.

(2) Sediment samples for sediment TOC analysis can be held at 4°C for up to 28 days, and should be analyzed within this 28-day period, but can be frozen at any time during the initial 28 days, for up to 12 months at minus (-) 20 °C.

(3) Sediment samples for TOC AND grain size analysis can be combined in one 250 mL clear glass jar, and sub-sampled at the laboratory in order to utilize holding time differences for the two analyses. If this is done, the 250 mL combined sediment sample must be refrigerated only (not frozen) at 4 °C for up to 28 days, during which time the sub-samples must be aliquoted in order to comply with separate storage requirements (as shown above).

APPENDIX F:

SWAMP BIOASSESSMENT SOP

SWAMP
Surface Water Ambient Monitoring Program

SWAMP Bioassessment Procedures 2007

Standard Operating Procedures for Collecting Benthic Macroinvertebrate Samples and Associated Physical and Chemical Data for Ambient Bioassessments in California

February 2007

California Water Boards

www.waterboards.ca.gov/swamp

Standard Operating Procedures for Collecting Benthic Macroinvertebrate Samples and Associated Physical and Chemical Data for Ambient Bioassessments in California

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Preparation Date: January 23rd 2007

Approved by: Beverly van Buuren, SWAMP QA Officer

Approval Date: 01/17/07

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ACKNOWLEDGEMENTS

The protocols described here represent the contributions of a wide range of researchers and field crews. Most of the physical habitat methods are close modifications of those used in the U.S. Environmental Protection Agency's (EPA's) Environmental Monitoring and Assessment Program (EMAP) and developed by EPA's Office of Research and Development (ORD, Peck et al. 2004). The benthic macroinvertebrate collection methods are based on EMAP methods (EPA's targeted riffle methods were derived in turn from methods developed at Utah State University; Hawkins et al. 2003).

The current version of these protocols was established by Peter Ode (Department of Fish and Game's (DFG's) Aquatic Bioassessment Laboratory (ABL)) and David Herbst (UC Santa Barbara's Sierra Nevada Aquatic Research Laboratory) with significant contributions from staff at the ABL (Jim Harrington, Shawn McBride, Doug Post, Andy Rehn, and Jennifer York), the Surface Water Ambient Monitoring Program (SWAMP) Quality Assurance (QA) Team, Thomas Suk and other members of the SWAMP bioassessment committee (Mary Adams, Lilian Busse, Matt Cover, Robert Holmes, Sean Mundell, and Jay Rowan) and three external reviewers: Chuck Hawkins, Dave Peck, and Phil Kaufmann.

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SWAMP GUIDANCE SG

SWAMP GUIDANCE FOR MACROINVERTEBRATE FIELD PROTOCOLS FOR WADEABLE STREAMS

Background: The SWAMP Bioassessment Committee met in December, 2004, and agreed that the SWAMP Quality Assurance Management Plan (QAMP) should be amended to provide greater consistency in bioassessment sampling protocols for wadeable streams. The Committee's recommendations were reviewed and accepted by the full SWAMP Roundtable[1] in February, 2005 (some of the key considerations are contained in Appendix A).

The current guidance for macroinvertebrate sampling under the SWAMP program is as follows:

1. For ambient bioassessment monitoring of wadeable streams in California, two methods are to be used at sites with riffle habitats (i.e., one "multihabitat" sample, and one sample that targets the "richest" habitat):
   - **For sites with sufficient riffle habitat,** the two samples shall be: (1) the reachwide benthos (RWB) method (also known as "multihabitat" sampling.); and (2) the targeted-riffle composite (TRC) method.
   - **For low-gradient sites that do not have sufficient riffle habitat**, the RWB method is the standard method, but we also recommend the option of collecting a sample with (2) the "Margin-Center-Margin" (MCM) method until ongoing methods comparisons are completed (see Appendix A).
   - **Notes:** (1) The protocols for each method are provided in this document; (2) Other appropriate method(s) will be allowed if the specific monitoring objectives require use of alternative method(s). (See Item #2, below.); (3) The protocol recommendations specified above will be reevaluated as results become available from ongoing methods comparison studies. (See Appendix A for more information.)

2. The SWAMP QAMP allows flexibility in sampling methods so that the most appropriate method(s) may be used to address hypothesis tests and project-specific objectives that differ from program objectives. Such situations may include, but are not necessarily limited to, special studies (e.g., evaluation of point source discharges, above/below comparisons where statistical replication is needed), stressor identification investigations, and long-term monitoring projects where consistent data comparability is desired and an alternative method is needed to achieve that comparability. In addition, in some rare cases where funding limitations would make it cost-prohibitive to complete a project in compliance with the protocols listed in #1, above, the project proponent may request to complete laboratory analysis of only one sample, and "archive" one of the macroinvertebrate samples (i.e., the RWB sample in streams with riffles) to reduce lab costs. Deviations from the protocols specified in #1 above may be granted by the SWAMP Bioassessment Coordinator or the full SWAMP Roundtable.

1. The SWAMP Roundtable is the coordinating entity for the program. Participants include staff from the State and Regional Water Boards, USEPA, the Department of Fish and Game, the Marine Pollution Studies Laboratory, Moss Landing Marine Laboratories, contractors, and other interested entities.

SECTION 1 INTRODUCTION

This document describes two standard procedures (TRC and RWB) for sampling benthic macroinvertebrate (BMI) assemblages for ambient bioassessments. This document also contains procedures for measuring instream and riparian habitats and ambient water chemistry associated with BMI samples. These sampling methods replace previous bioassessment protocols referred to as the California Stream Bioassessment Procedure (CSBP, Harrington 1995, 1999, 2002).

These procedures can produce quantitative and repeatable measures of a stream's physical/habitat condition and benthic invertebrate assemblages, but they require field training and implementation of QA measures throughout the field season.

The sampling layout described here provides a framework for systematically collecting a variety of physical, chemical, and biological data. The biological sampling methods are designed to nest within the overall framework for assessing the biotic, physical, and chemical condition of a reach. The layout used in these procedures and most of the physical habitat methods are close modifications of those used in EPA's EMAP and developed by EPA's ORD (Peck et al. 2004). Data collected using this methodology are generally directly comparable to equivalent EMAP data, except for the difference in reach length. Other exceptions are noted in the text.

The following steps are presented in an order suggested for efficient data collection. The specific order of collection for the physical parameters may be modified according to preferences of field crews, with the caveat that care must always be taken to not disturb the substrates within the streambed before BMI samples are collected.

PHYSICAL HABITAT METHODS

The physical habitat scoring methods described here can be used as a stand-alone evaluation or used in conjunction with a bioassessment sampling event. However, measurements of instream and riparian habitat and ambient water chemistry are essential to interpretation of bioassessment data and should always accompany bioassessment samples. This information can be used to classify stream reaches, associate physical and chemical condition with biotic condition, and explain patterns in the biological data.

Because bioassessment samples can be collected to answer a variety of questions, this document describes the component measures of instream and riparian habitat as independent modules. Although individual modules can be added or subtracted from the procedure to reflect specific project objectives, a standard set of modules will normally accompany bioassessment samples. This document describes two standard groupings of modules that represent two different levels of intensity for characterizing the chemical and physical habitat data (Table 1). The BASIC physical habitat characterization represents a minimum amount of physical and chemical data that should be taken along with any ambient BMI sample, the FULL physical habitat characterization represents the suite of data that should be collected with most professional level bioassessment samples (e.g., SWAMP regional monitoring programs). In addition to these data, we also briefly introduce additional data modules (e.g., excess sediment, periphyton) that can be collected as supplements to the full set (OPTIONAL). Table 1 lists the physical and chemical variables that should be measured under the different levels.

Note: *SWAMP intends to develop guidance for selecting appropriate physical habitat modules to the intended uses of data. Until this guidance is available, users of these protocols should consult with representatives of the Regional Water Quality Control Boards (Regional Boards) or the SWAMP Bioassessment Coordinator when selecting modules.*

FIELD CREW SIZE AND TIME ESTIMATES

These methods are designed to be completed by either two or three (or more) person field crews. A very experienced field crew can expect to complete the full suite of physical habitat measurements and the two BMI sampling protocols in approximately two hours. Less experienced crews will probably take closer to three or four hours to complete the work depending on the complexity of the reach. Note that this estimate includes only time at the site, not travel time between sites.

Equipment and Supplies

Recommended equipment and supplies are listed in Table 2.

Table 1. Summary of physical habitat and water chemistry and proposal for basic, full, and optional levels of effort.

Survey Task	Parameter(s)	Basic	Full	Option	Comments
REACH DELINEATION and WATER QUALITY [Conducted before entering stream to sample BMIs or conduct any habitat surveys]	Layout reach and mark transects, record GPS coordinates	X	X		Use 150-m reach length if wetted width ≤10 m; Use 250-m reach length if wetted width > 10 m
	Temperature, pH, specific conductance, DO, alkalinity	X	X		Multi-meter (e.g., YSI, Hydrolab, VWR Symphony)
	Turbidity, Silica			X	Use test kit or meter
	Notable field conditions	X	X		Recent rainfall, fire events, dominant local landuse
CROSS-SECTIONAL TRANSECTS BASIC Measurements at main 11 transects only FULL Measurements at 11 main transects (A, B, C, D, E, F, G, H, I, J, K) or 21 transects (11 main plus 10 inter-transects) for substrate size classes only	Wetted width	X	X		Stadia rod is useful here
	Flow habitat delineation	X	X		Record proportion of habitat classes in each inter-transect zone
	Depth and Pebble Count + CPOM		X		5 -point substrate size, depth and CPOM records at all 21 transects
	Cobble embeddedness		X		All cobble-sized particles in pebble count. Supplement with "random walk" if needed for 25
	Slope (%)	See reach scale	X		Average slope calculated from 10 transect to transect slope measurements. Use autolevel for slopes ≤ 1%; clinometer is OK for steeper gradients
	Sinuosity		X		Record compass readings between transect centers
	Canopy cover	X	X		Four densiometer readings at center of channel (facing L bank R bank, Upstream +Downstream)
	Riparian Vegetation		X		Record % or categories
	Instream Habitat		X		
	Human Influence		X		
	Bank Stability	X	X		Eroding / Vulnerable / Stable
	Bankfull Dimensions		X		
	Excess Sediment Transect Measures (optional)				
	Bankfull width and height, bank angles			X	
	Large woody debris counts			X	Tallies of woody debris in several size classes
	Thalweg profile			X	100 equidistant points along thalweg

Survey Task	Parameter(s)	Basic	Full	Option	Comments
DISCHARGE TRANSECT	Discharge measurements		X		Velocity-Area Method or Neutrally Buoyant Object Method
REACH SCALE MEASUREMENTS:	EPA-RBP visual scoring of habitat features	*		X	*Used for citizen monitoring and comparison with legacy data
	Selected RBP visuals:		X		Channel alteration, sediment deposition, epifaunal substrate (redundant if doing EPA-RBP scoring)
	Slope (%, not degrees)	X	See transect scale		Single measurement for entire reach only for BASIC. Use autolevel for slopes ≤ 1%, clinometer is OK for higher gradients
	Photo documentation	X	X		Upstream (A, F, K) Downstream (F)
OTHER OPTIONAL COMPONENTS					
FOOD RESOURCE QUANTIFICATION	Periphyton (3 replicates)			X	Qualitative characterization of diatom growth and filamentous algal growth, quantification of biomass (AFDM, chl-a)
	CPOM & FPOM (3 replicates)			X	CPOM field measure of wet mass >1 mm particles, FPOM as 0.25 – 1 mm fraction (AFDM in lab)

Table 2. Field equipment and supplies

Physical Habitat	BMI Collection	General/ Ambient Chemistry
• GPS receiver • topographic maps • measuring tape (150-m) • small metric ruler or gravelometer for substrate measurements • digital watch, random number table or ten-sided die • stadia rod • clinometer • autolevel (for slopes < 1%) • handlevel (optional) • current velocity meter • stopwatch for velocity measurements • convex spherical densitometer • flags/ flagging tape • rangefinder	• D-frame kick net (fitted with 500-µ mesh bag) • standard # 35 sieve (500-µ mesh) • wide-mouth 500-mL or 1000 mL plastic jars • white sorting pan (enamel or plastic) • 95% EtOH • fine tipped forceps or soft forceps • waterproof paper and tape for attaching labels • 10-20-L plastic bucket for sample elutriation • preprinted waterproof labels (e.g., Rite-in-the-Rain™) • disposable gloves/ elbow length insulated gloves	• sampling SOP (this document) • hip or chest waders, or wading boots/shoes • field forms printed on waterproof paper (e.g., Rite-in-the-Rain™) • clip board and pencils • digital camera • centigrade thermometer • pH meter • DO meter • conductivity meter • field alkalinity meter • water chemistry containers • calibration standards • spare batteries for meters • first aid kit

SECTION 2
REACH DELINEATION AND WATER QUALITY

REACH LAYOUT AND GENERAL DOCUMENTATION

The systematic positioning of transects is essential to collecting representative samples and to the objective quantification of physical habitat measures. The standard sampling layout consists of a 150-m reach (length measured along the bank) divided into 11 equidistant transects that are arranged perpendicular to the direction of flow (Figure 1, Figure 2). Ten additional transects (designated "inter-transects") located between the main transects give a total of 21 transects per reach. Main transects are designated A through K while inter-transects are designated by their nearest upstream and downstream transects (e.g., AB, BC, etc.). In extreme circumstances, reach length can be shorter than 150 m (e.g., if upstream and downstream barriers preclude a 150-m reach), but this should be avoided whenever possible. If the actual reach length is other than 150 m or 250 m this should be noted and explained on the field forms.

Note 1: *The standard reach length differs from that used in the EMAP design, in which reach length was defined as 40x stream width, with a minimum reach length of 150 m. The EMAP reach length approach is used to ensure that enough habitat is sampled to support accurate fish assemblage estimates and relatively precise characterization of channel characteristics (e.g., residual pool volumes and woody debris estimates, which that are critical for relative bed stability estimates). Programs wishing to sample fish assemblages or produce relative bed stability estimates should strongly consider adopting the EMAP guidance for setting reach length.*

Note 2: *Streams > 10 m wetted width should use a reach length of 250 m. Some very large streams (i.e., > 20-m wetted width) may not be adequately represented even by a 250-m reach. In these cases, field crews should define a reach length that is representative of the larger stream segment being studied (i.e., attempt to include two to three meander cycles, or four to six riffle-pool sequences when possible).*

Note 3: *When the exact reach location is not restricted by the sampling design, attempt to position reaches upstream of bridges to avoid this influence.*

Step 1. Upon arrival at the sampling site, fill out the reach documentation section of the field forms (site and project identification, stream and watershed name, crew members, and date/time). If known at the time of sampling, record the Site Code following SWAMP site code formats. Determine the geographic coordinates of the downstream end of the reach (preferably in decimal degrees to at least four decimal places) with a GPS receiver and record the datum setting of the unit (preferably NAD83/ WGS84).

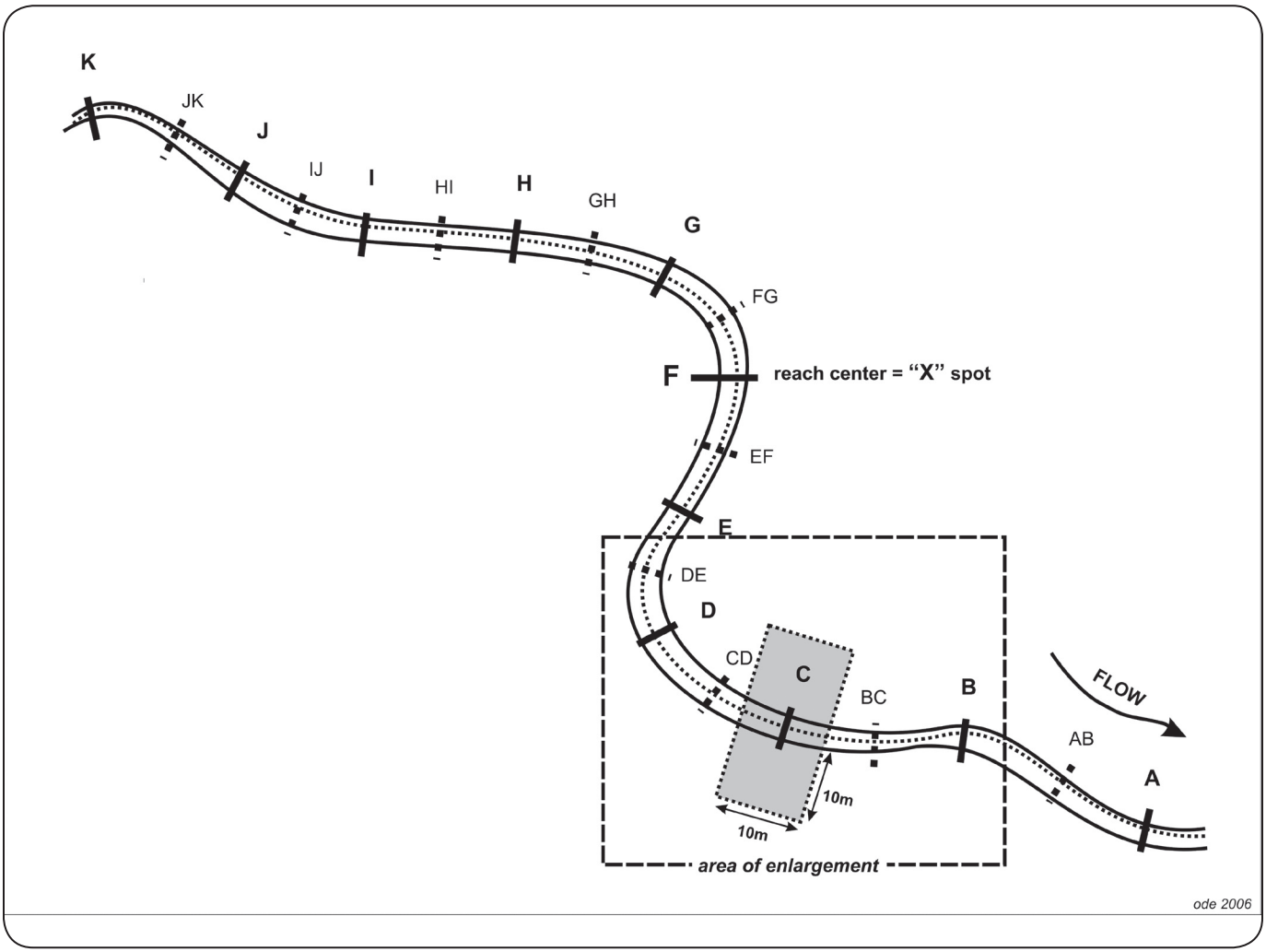

Figure 1. Reach layout geometry for physical habitat and biological sampling showing positions of 11 main transects (A – K) and the 10 supplemental inter-transects (AB- JK). The area highlighted in the figure is expanded in Figure 2. Note: reach length = 150 m for streams ≤ 10-m average wetted width, and reach length = 250 m for streams > 10-m average wetted width.

Step 2. Once a site has been identified, make an initial survey of the reach from the stream banks (being sure to not disturb the instream habitat). If TRC samples will be collected, identify all riffle habitats suitable for sampling (see Section IIIa for suitable habitat types) and note their positions so that a subset can be identified for sampling.

Step 3. Determine if the average wetted width is greater or less than 10 m. If the average wetted width ≤ 10 m, use a 150-m reach length. If the average wetted width > 10 m, use a 250-m reach length.

Step 4. Starting at one end of the reach, establish the position of the 11 main transects (labeled A-K from downstream to upstream) by measuring 15 m (25 m for streams > 10 m wetted width) along the bank from the previous transect. The 10 inter-transects should be established equidistant from the adjacent main transects (i.e., 7.5 m from main transects for 150-m reaches, 12.5 m for 250-m reaches). Since the data collection will start at the downstream end, is often easiest to establish transects starting from the upstream end. For easy setup and breakdown, mark the main transects with easily removable markers (e.g., large washers tied with strips of flagging, surveyor's flags).

Note 1: *While it is usually easiest to establish transect positions from the banks (this also reduces disturbance to the stream channel), this can result in uneven spacing of transects in complex stream reaches. To avoid this, estimate transect positions by projecting from the mid-channel to the banks.*

Note 2: *Flagging of a single bank is recommended to reduce mistakes caused by missed markers.*

Step 5. Measure and record common ambient water chemistry measurements (pH, DO, specific conductance, alkalinity, water temperature) at the downstream end of the reach (near same location as the GPS coordinates were taken). These are typically taken with a handheld water quality meter (e.g., YSI, Hydrolab), but field test kits (e.g., Hach) can provide acceptable information if they are properly calibrated. For appropriate calibration methods and calibration frequency, consult the current SWAMP QAMP (Appendix F), or follow manufacturer's guidelines.

Note 1: *If characteristics of the site prohibit downstream entry, measurements may be taken at other points in the reach. In all cases, ambient chemistry measurements should be taken at the beginning of the reach survey.*

Note 2: *Alkalinity test kits may not perform well in low ionic strength waters. Programs should consider collecting lab samples for these sites (see SWAMP QAMP for guidance on collecting water chemistry samples).*

Step 6. Take a minimum of four (4) photographs of the reach at the following locations: a) Transect A facing upstream, b) Transect F facing upstream, c) Transect F facing downstream, and d) Transect K facing downstream. It may also be desirable to take a photograph at Transect A facing downstream and Transect K facing upstream to document conditions immediately adjacent to the reach. Digital photographs should be used when possible. Record the image numbers on the front page of the field form.

Note 1: *When possible, photograph names should follow SWAMP coding conventions ("StationCode_yyyy_mm_dd_uniquecode"). The unique code should include one of the following codes to indicate direction: RB (right bank), LB (left bank), BB (both banks), US (upstream), DS (downstream). SWAMP suggests using unique codes created by the camera to facilitate file organization. Example: 603WQLB02_2004_03_20_RBDS1253.*

Step 7. Record the dominant land use and land cover in the area surrounding the reach (evaluate land cover within 50 m of either side of the stream reach).

Step 8. At the bottom of the form, record evidence of recent flooding, fire, or other disturbances that might influence bioassessment samples. Especially note if flow conditions have been affected by recent rainfall, which can cause significant under-sampling of BMI diversity (see note in the following section). If you are unaware of recent fire or rainfall events, select the "no" option on the forms.

SECTION 3
COLLECT BENTHIC MACROINVERTEBRATES

MULTIPLE HABITAT AND TARGETED RIFFLE PROTOCOLS

Note 1: *BMI samples intended for ambient bioassessments are generally collected when streams are at or near base flow (i.e., not influenced by surface runoff) as sudden flow increases can dramatically alter local community composition.*

Note 2: *Guidance for choosing among TRC sampling, RWB sampling or both will be provided in a separate document (see Appendix A for current guidance for sampling under SWAMP).*

Once the reach transects have been laid out, the biological samples (BMIs and algae if included) should be collected before any other physical habitat measures so that substrates are not disturbed prior to sampling. Both TRC and RWB methods use 500-µ mesh D-frame nets (see list of BMI sampling equipment in Table 2). The two samples can be collected at the same time by carrying two D-nets and compositing the material from the two samples in their respective nets. If a two person field crew is responsible for both the physical habitat data and benthic invertebrate samples, it is generally best to collect the benthos at each transect, then immediately record the physical habitat data before moving to the next transect. Obviously, this requires especially careful handling of the D-nets during the course of sampling to avoid loss or contamination of the samples. It can be helpful to clearly label the two D nets as RWB and TRC. Larger field crews may choose to split the sampling between biological team and a physical habitat team and have the biological team go through the reach first. The positions of the TRC and RWB subsampling locations are illustrated in Figure 2.

SECTION III A. TARGETED RIFFLE COMPOSITE PROCEDURE

The TRC method is designed for sampling BMIs in wadeable streams that contain fast-water (riffle/run) habitats and is not appropriate for waterbodies without fastwater habitats. The RWB protocol should be used in these situations. Riffles are often used for collecting biological samples (e.g., the old CSBP methods) because they often have the highest BMI diversity in wadeable streams. This method expands the definition to include other fast water habitats, however care should be taken when attempting to apply this method in low gradient streams.

Note: *Since all streams (even low gradient streams) have variation in flow habitats within the channel, this guidance should not be interpreted as including areas within low gradient streams that are only marginally faster than the surrounding habitats. The RWB protocol should be applied in these situations.*

The TRC was developed by the Western Center for Monitoring and Assessment of Freshwater Ecosystems (www.cnr.usu.edu/wmc) in Logan, Utah (Hawkins et al. 2003) and slightly modified by the EPA program (Peck et al. 2004). The TRC has been widely used in California (US Forest Service (USFS), the EMAP Western Pilot, and the California Monitoring and Assessment Program (CMAP)), and in the interest of methodological consistency between state and federal water resource agencies, has been adopted as the standard riffle protocol for bioassessment in California. The version described here is the EMAP modification, which distributes the sampling effort throughout the reach.

Sampling Locations – Acceptable Habitat Types

Riffles are the preferred habitat for TRC sampling, but other fast water habitats are acceptable for sampling if riffles are sparse. Common flow-defined habitat types are listed in Table 3 in decreasing order of energy. Most streams contain some or all of the following fast water habitat types: 1) cascades/falls, 2) rapids, 3) riffles, 4) runs. All of these are acceptable for TRC sampling if riffles are not available.

Note: *Because the common habitat types are arranged on a continuum between high to low energy environments, the categories grade into each other continuously and are not discrete. Thus, determination of habitat types requires somewhat subjective decision-making.*

Table 3. Common habitat types in stream channels, arranged in decreasing order of energy

Flow Habitat Type	Description
Cascades	Short, high gradient drop in stream bed elevation often accompanied by boulders and considerable turbulence
Falls	High gradient drop in elevation of the stream bed associated with an abrupt change in the bedrock
Rapids	Sections of stream with swiftly flowing water and considerable surface turbulence. Rapids tend to have larger substrate sizes than riffles
Riffles	Shallow sections where the water flows over coarse stream bed particles that create mild to moderate surface turbulence; (< 0.5 m deep, > 0.3 m/s)
Step-Runs	A series of runs that are separated by short riffles or flow obstructions that cause discontinuous breaks in slope
Runs	Long, relatively straight, low-gradient sections without flow obstructions. The stream bed is typically even and the water flows faster than it does in a pool; (> 0.5 m deep, > 0.3 m/s)
Glides	A section of stream with little or no turbulence, but faster velocity than pools; (< 0.5 m deep, < 0.3 m/s)
Pools	A reach of stream that is characterized by deep, low-velocity water and a smooth surface ; (> 0.5 m deep, < 0.3 m/s)

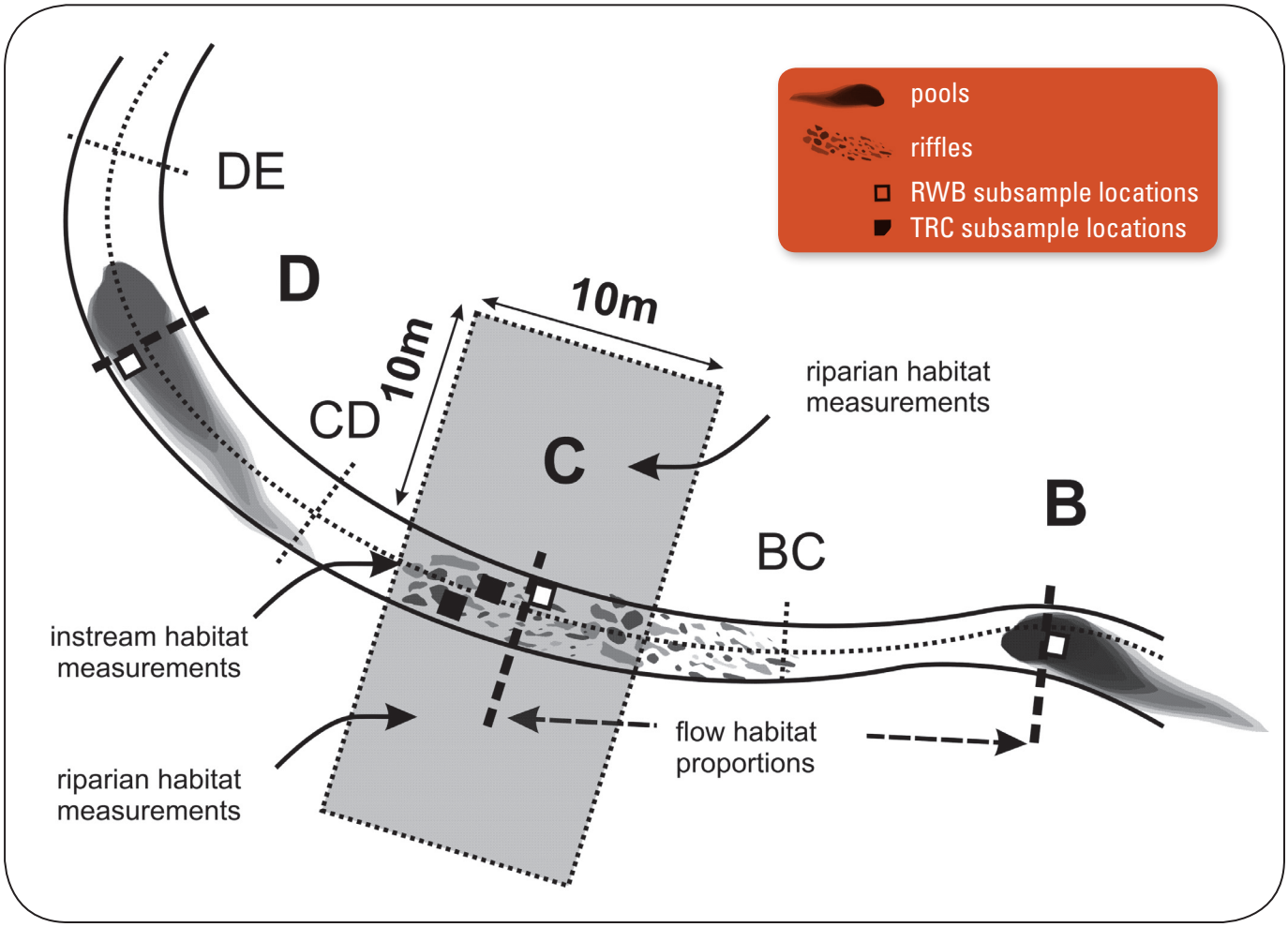

Figure 2. Section of the standard reach expanded from Figure 1 showing the appropriate positions for collecting benthic macroinvertebrate samples, instream and riparian habitat measurements and flow habitat proportion measurements.

Sampling Locations – Selecting Habitat Units

A TRC sample is a composite of eight individual kick samples of 1 ft^2 (0.09 m^2) of substrate each. During your initial layout of the reach, take a mental note of the number and position of the main riffles in a reach (and other fast water habitats if needed). Randomly distribute the eight sub-samples among the fast water habitats in the reach, giving preference to riffles where possible. Unless you are sampling in small streams, try to avoid very small riffle units (i.e., < 5 ft^2). If fewer than eight riffles are present in a reach, more than one sample may be taken from a single riffle, especially if the riffles are large.

Sampling Procedure

Begin sampling at the downstream end of the reach at the first randomly selected riffle and work your way upstream.

TRC-Step 1. Determine net placement within each habitat unit by generating a pair of random numbers between 0 and 9. Examples of convenient random number generators include the hundredths place on the stopwatch feature of a digital watch, a 10 sided die and a random number chart. The first number in each pair (multiplied by 10) represents the percent upstream along the habitat unit's length. The second number in each pair represents the percent of the riffle width from right bank. For example, if the two generated random numbers are 4 and 7, you will walk upstream 40% of the distance of the riffle and then go 70% of the distance across the riffle (see Figure 3). This position is the center of the 1 ft^2 (0.09 m^2) sampling quadrat for that riffle. If you are unable to sample this location because it is too deep or it is occupied by a large boulder, select a new pair of random numbers and pick a new spot.

TRC-Step 2. Position a 500-µ D-net (with the net opening perpendicular to the flow and facing upstream) quickly and securely on the stream bottom to eliminate gaps under the frame. Avoid, and if necessary remove, large rocks that prevent the sampler from seating properly on the stream bottom.

TRC-Step 3. Holding the net in position on the substrate, visually define a square quadrat that is one net width wide and one net width long upstream of the net opening. Since D-nets are 12 inches wide, the area within this quadrat is 1ft^2 (0.09 m^2). Restrict your sampling to within that area. If desired, a wire frame of the correct dimensions can be placed in front of the net to help delineate the quadrat to be sampled, but it is often sufficient to use the net dimensions to keep the sampling area consistent.

TRC-Step 4. Working backward from the upstream edge of the sampling plot, check the quadrat for heavy organisms such as mussels, snails, and stone-cased caddisflies. Remove these organisms from the substrate by hand and place them into the net. Carefully pick up and rub stones directly in front of the net to remove attached animals. Remove and clean all of the rocks larger than a golf ball ($\sim$3 cm) within your sampling quadrat such that all the organisms attached to them are washed downstream into your net. Set these rocks outside your sampling quadrat after you have cleaned them. If the substrate is consolidated or comprised of large, heavy rocks, use your feet to kick and dislodge the substrate to displace BMIs into the net. If you cannot remove a rock from the stream bottom, rub it (concentrating on cracks or indentations) thereby loosening any attached insects. As you are disturbing the plot, let the water current carry all loosened material into the net.

Note 1: *Brushes are sometimes used in other bioassessment protocols to help loosen organisms, but in the interest of standardizing collections, do not use a brush when following this protocol.*

Note 2: *In sandy-bottomed streams, kicking within run habitats can quickly fill the sampling net with sand. In these situations, follow the standard procedures but use care to disturb the substrate gently and avoid kicking.*

TRC-Step 5. Once the coarser substrates have been removed from the quadrat, dig your fingers through the remaining underlying material to a depth of about 10 cm (this material is often comprised of gravels and finer particles). Thoroughly manipulate the substrates in the quadrat.

Note: *The sampler may spend as much time as necessary to inspect and clean larger substrates, but should take a standard time of 30 seconds to perform Step 5.*

TRC-Step 6. Let the water run clear of any insects or organic material before carefully lifting the net. Immerse the net in the stream several times to remove fine sediments and to concentrate organisms at the end of the net, but be careful to avoid having any water or foreign material enter the mouth of the net during this operation.

TRC-Step 7. Move upstream to the next randomly selected habitat unit and repeat steps one through six, taking care to keep the net wet but uncontaminated by foreign material when moving the net from riffle to riffle. Sometimes, the net will become so full of material from the streambed that it is no longer effective at capturing BMIs. In these cases, the net should be emptied into sample jars as frequently as necessary, following guidelines described below in the "Preparation of BMI Sample Jars" section. Continue until you have sampled eight $1ft^2$ ($0.09\ m^2$) of benthos.

TRC-Step 8. PROCEED to Section IIIc. Filling and Labeling BMI Sample Jars.

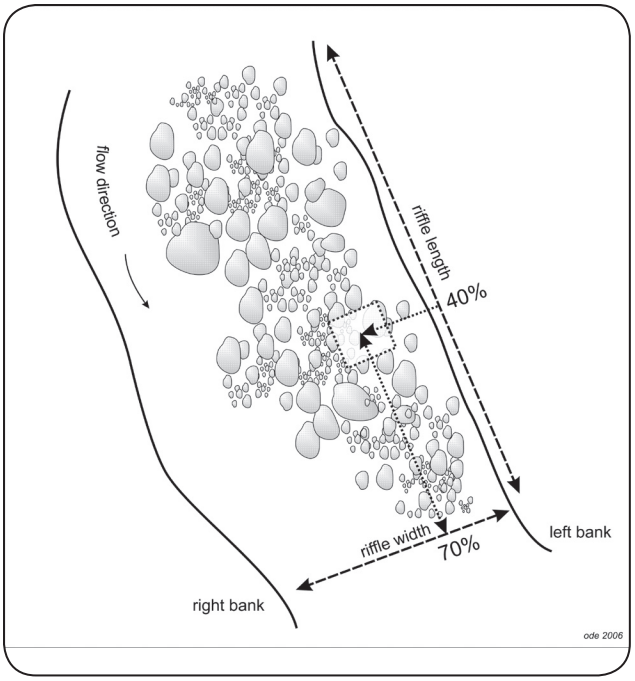

Figure 3. Example showing the method for selecting a subsampling position within a selected riffle under the TRC method. In this example, the random numbers 4 and 7 were selected

SECTION III B. REACHWIDE BENTHOS (MULTIHABITAT) PROCEDURE

The RWB procedure employs an objective method for selecting subsampling locations that is built upon the 11 transects used for physical habitat measurements. The RWB procedure can be used to sample any wadeable stream reach since it does not target specific habitats. Because sampling locations are defined by the transect layout, the position of individual sub-samples may fall in a variety of erosional or depositional habitats.

Note: *Sampling locations should be displaced one meter downstream of the transects to avoid disturbing substrates for subsequent physical habitat assessments.*

RWB -Step 1. The sampling position within each transect is alternated between the left, center and right positions along a transect (25%, 50% and 75% of wetted width, respectively) as you move upstream from transect to transect. Starting with the downstream transect (Transect

A), identify a point that is 25% of the stream width from the right bank (note that the right bank will be on your left as you face upstream). If you cannot collect a sample at the designated point because of deep water obstacles or unsafe conditions, relocate the point as close as possible to the designated position.

Note: *A modification to this procedure is currently being investigated by SWAMP. This "margin-center-margin" (MCM) modification replaces the samples at 25% and 75% of wetted width with samples of the marginal habitats (including emergent and submergent vegetation).*

RWB -Step 2. Place a 500-µ D-net in the water so the mouth of the net is perpendicular to and facing into the flow of the water. If there is sufficient current in the area at the sampling point to fully extend the net, use the normal D-net collection technique to collect the sub-sample (TRC-Step 3 through TRC-Step 6 above). If flow volume and velocity is not sufficient to use the normal collection technique, use the sampling procedure for "slack water" habitats (RWB-Step 3 through RWB-Step 7 below).

RWB -Step 3. Visually define a 1 ft^2 (0.09 m^2) quadrat that is one net-width wide and one net-width long at the sampling point.

RWB -Step 4. Working backward from the upstream edge of the sampling plot, 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. Carefully pick up and rub stones directly in front of the net to remove attached animals. Remove and clean all of the rocks larger than a golf ball within your sampling quadrat such that all the organisms attached to them are washed downstream into your net. Set these rocks outside your sampling quadrat after you have cleaned them. Large rocks that are less than halfway into the sampling area should be pushed aside. If the substrate is consolidated or comprised of large, heavy rocks, use your feet to kick and dislodge the substrate to displace BMIs into the net. If you cannot remove a rock from the stream bottom, rub it (concentrating on cracks or indentations) thereby loosening any attached insects.

RWB -Step 5. Vigorously kick the remaining finer substrate within the quadrat with your feet while dragging the net repeatedly through the disturbed area just above the bottom. Keep moving the net all the time so that the organisms trapped in the net will not escape. Continue kicking the substrate and moving the net for 30 seconds. For vegetation-choked sampling points, sweep the net through the vegetation within a 1ft^2 (0.09 m^2) quadrat for 30 seconds.

Note: *If flow volume is insufficient to use a D- net, spend 30 seconds hand picking a sample from 1ft^2 of substrate at the sampling point, then stir up the substrate with your gloved hands and use a sieve with 500-µ mesh size to collect the organisms from the water in the same way the net is used in larger pools.*

RWB -Step 6. After 30 seconds, remove the net from the water with a quick upstream motion to wash the organisms to the bottom of the net.

RWB -Step 7. PROCEED to Section IIIc: Filling and Labeling BMI Sample Jars

SECTION III C. FILLING AND LABELING BENTHIC MACROINVERTEBRATE SAMPLE JARS

Step 1. Once all sub-samples (eight for TRC, 11 for RWB) have been collected, transfer benthos to a 500-mL or 1000-mL wide-mouth plastic sample jar using one of the following methods.

Note: *Field elutriation should only be used by well-trained field crews who are proficient at removing all benthic organisms from the discarded inorganic material. Training in the recognition of aquatic invertebrates is highly recommended.*

Step 1a. Complete Transfer of all Sampled Material – Invert the contents of the kick net into the sample jar. Perform this operation over a white enameled tray to avoid loss of any sampled material and make recovery of spilled organisms easier. If possible, remove the larger twigs and rocks by hand after carefully inspecting for clinging organisms, but be sure not to lose any organisms. Use forceps to remove any organisms clinging to the net and place these in the sample jar.

Step 1b. Field Elutriation of Samples – Empty the contents of the net into a large plastic bucket (10-20 L is sufficient). Use forceps to remove any organisms clinging to the net and place these in the bucket. Add stream water to the bucket and gently swirl the contents of the bucket in order to suspend the organic material (being certain to not introduce entrained organisms from the source water). Pour the organic matter from the bucket through a 500-µ sieve (or use the 500-µ net). Repeat this process until no additional material can be elutriated (i.e., only inorganic material is left in the bucket). If possible, remove the larger twigs and rocks by hand after carefully inspecting for clinging organisms, but be sure not to lose any organisms. Transfer all of the material in the sieve (invertebrates and organic matter) into the sample jar. Carefully inspect the gravel and debris remaining in the bottom of the bucket for any cased caddisflies, clams, snails, or other dense animals that might remain. Remove any remaining animals by hand and place them in the sample jar.

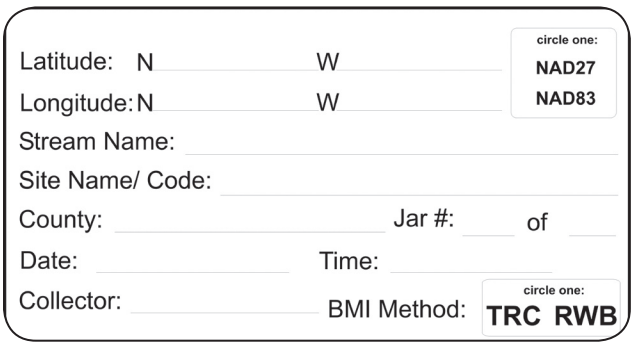

Figure 4. Example date - locality label for all BMI samples.

Step 2. Place a completed date/locality label (see Figure 4) on the inside of the jar (use pencil only as most "permanent" inks dissolve in ethanol) and completely fill with 95% ethanol. Place a second label on the outside of the jar. Note that the target concentration of ethanol is 70%, but 95% ethanol is used in the field to account for dilution from water in the sample. If organic and inorganic material does not accumulate in the net quickly, it may be possible to transfer all the material in the net into one jar. Otherwise, divide the material evenly among several jars

(being careful to clearly label them as part of a set). To ensure proper preservation of benthic macroinvertebrates it is critical that the ethanol is in contact with the BMIs in the sample jar. Never fill a jar more than 2/3 full with sampled material, and gently rotate jars that contain mostly mud or sand to ensure that the ethanol is well distributed. If jars will be stored for longer than a month prior to processing, jars should not contain more than 50% sample material.

SECTION 4
MAIN CROSS-SECTIONAL TRANSECT MEASURES

SECTION IVA. PHYSICAL MEASURES

The majority of physical habitat measurements in this protocol are made relative to the main cross-sectional transects (Figure 5). All the measures taken relative to each transect are recorded on forms specific to that transect. Start with the downstream transect (Transect A) and repeat steps 6-15 for all 11 main transects.

Module A. Transect Dimensions: Wetted Width and Bankfull Dimensions

Wetted Width – The wetted channel is the zone that is inundated with water and the wetted width is the distance between the sides of the channel at the point where substrates are no longer surrounded by surface water. Measure the wetted stream width and record this in the box at the top of the transect form.

Bankfull Width and Depth – The bankfull channel is the zone of maximum water inundation in a normal flow year (one to two year flood events). Since most channel formation processes are believed to act when flows are within this zone (Mount 1995), bankfull dimensions provide a valuable indication of relative size of the waterbody.

Note: *Bankfull dimensions are notoriously difficult to assess, even by experienced field crews (see Heil and Johnson 1995). It is often useful to discuss the interpretation of bankfull locations among the field crew members to reach a consensus. The USFS Stream Team provides a good set of instructional videos for improving consistency in accurate bankfull measurements (http://www.stream.fs.fed.us/publications/videos.html).*

Step 1. Scout along the stream margins to identify the location of the bankfull margins on either bank by looking for evidence of annual or semi-annual flood events. Examples of useful evidence includes topographic, vegetative, or geologic cues (changes in bank slope, changes from annual to perennial vegetation, changes in the size distribution of surface sediments). While the position of drift material caught in vegetation may be a helpful aid, this can lead to very misleading measurements.

Note: *The exact nature of this evidence varies widely across a range of stream types and geomorphic characteristics. It is helpful to investigate the entire reach when attempting to interpret this evidence because the true bankfull margin may be obscured at various points along the reach. Often the bankfull position is easier to interpret from one bank than the other; in these cases, it is easiest to infer the opposite bank position by projecting across the channel. Additionally, height can be verified by measuring the height from both edges of the wetted channel to the bankfull height (these heights should be equal).*

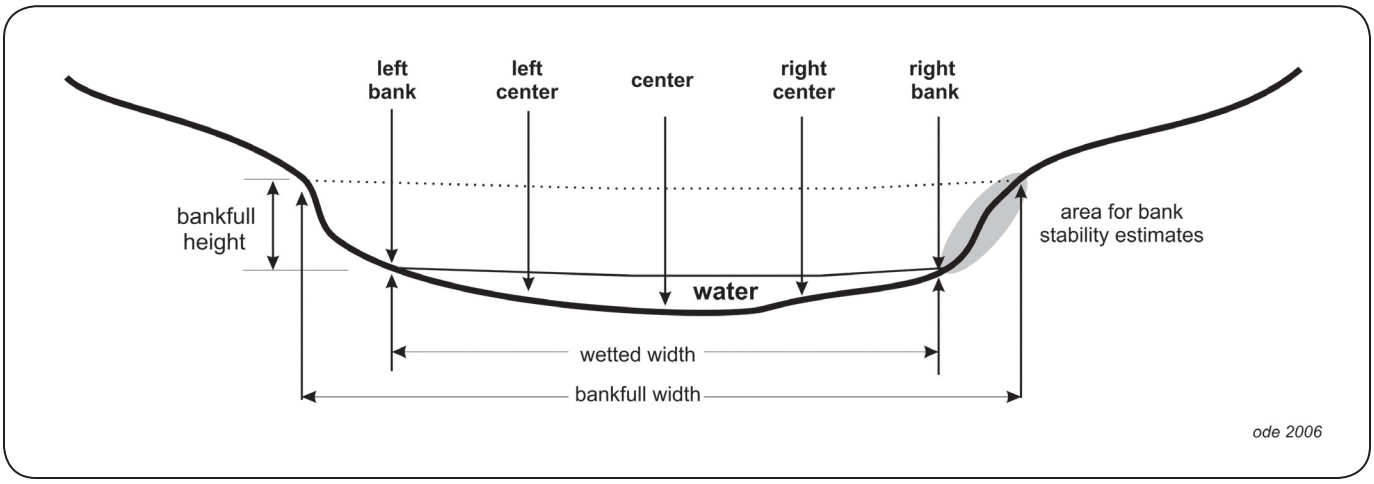

Figure 5. Cross sectional diagram of a typical stream channel showing locations of substrate measurements, wetted and bankfull width measurements, and bank stability visual estimates.

Step 2. Stretch a tape from bank to bank at the bankfull position. Measure the width of the bankfull channel from bank to bank at bankfull height and perpendicular to the direction of stream flow.

Step 3. Measure bankfull height (the vertical distance between the water height of the water and the height of the bank, Figure 5) and record.

Module B. Transect Substrate Measurements

Particle size frequency distributions often provide valuable information about instream habitat conditions that affect BMI distributions. The Wolman pebble count technique (Wolman 1954) is a widely used and cost-effective method for estimating the particle size distribution and produces data that correlates with costly, but more quantitative bulk sediment samples. The method described here follows the EMAP protocol, which records sizes of 105 particles in a reach (five particles from each of 11 main transects and 10 inter-transects).

Note: *The size cutoff for the finest particle sizes in the EMAP protocol (< 0.06 mm) differs from that used by the Sierra Nevada Aquatic Research Laboratory (SNARL) program (0.25 mm), although the narrative description for this cutoff is the same (the point at which fine particles rubbed between one's fingers no longer feel gritty).*

Coarse particulate organic matter (CPOM, particles of decaying organic material such as leaves that are greater than 1.0 mm in diameter) is a general indicator of the amount of allochthonous organic matter available at a site, and its measurement can provide valuable information about the basis of the food web in a stream reach. The presence of CPOM associated with each particle is quantified at the same time that particles are measured for the pebble counts.

Step 1. Transect substrate measurements are taken at five equidistant points along each transect (Figure 5). Divide the wetted stream width by four to get the distance between the five points (Left Bank, Left Center, Center, Right Center and Right Bank) and use a measuring device to locate the positions of these points (a stadia rod is especially helpful here). Once the positions are identified, lower a graduated rod (e.g., a marked ski pole) though the water column perpendicular to both the flow and the transect to objectively select the particle located at the tip of the rod.

Step 2. Measure the depth from the water surface to the top of the particle with the graduated rod and record to the nearest cm.

Step 3. Record the presence or absence of CPOM >1mm within 1 cm of the particle.

Step 4. If the particle is cobble-sized (64-250 mm), record the percent of the cobble that is embedded by fine particles (<2 mm) to the nearest 5% (see cobble embeddedness text below).

Step 5. Remove the particle from the streambed, then measure and record the length of its intermediate axis to the nearest mm (see Figure 6). Alternatively, assign the particle to one of the size classes listed in the bottom of the transect form. Particle sizes classes can be estimated visually or with a quantitative measuring device (e.g., pass/ no-pass template, "gravelometer"). Regardless of the method, all particles less than 0.06 mm should be recorded as fines, all particles between 0.06mm and 2.0 mm recorded as sand. Field crews may want to carry vials containing sediment particles with these size ranges until they are familiar with these particles.

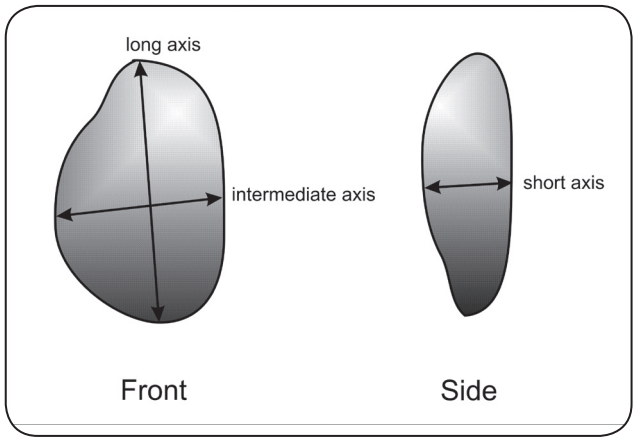

Figure 6. Diagram of three major perpendicular axes of substrate particles. The intermediate axis is recorded for pebble counts.

Module C. Cobble Embeddedness

The quantification of substrate embeddedness has long been a challenge to stream geomorphologists and ecologists (Klamt 1976, Kelley and Dettman 1980). It is generally agreed that the degree to which fine particles fill interstitial spaces has a significant impact on the ecology of benthic organisms and fish, but techniques for measuring this impact vary greatly (this is summarized well by Sylte and Fischenich 2002, http://stream.fs.fed.us/news/streamnt/pdf/StreamOCT4.pdf). Here we define embeddedness as the volume of cobble-sized particles (64-250 mm) that is buried by fine particles (<2.0 mm diameter).

Note: *This method differs from the EMAP method for measuring embeddedness, which measures embeddedness of all particles larger than 2 mm.*

Table 4. Size class codes and definitions for particle size measurements

Size Class Code	Size Class Description	Common Size Reference	Size Class Range
RS	bedrock, smooth	larger than a car	> 4 m
RR	bedrock, rough	larger than a car	> 4 m
XB	boulder, large	meter stick to car	1 - 4 m
SB	boulder, small	basketball to meter stick	25 cm - 1.0 m
CB	cobble	tennis ball to basketball	64 - 250 mm
GC	gravel, coarse	marble to tennis ball	16 - 64 mm
GF	gravel, fine	ladybug to marble	2 – 16 mm
SA	sand	gritty to ladybug	0.06 – 2 mm
FN	fines	not gritty	< 0.06 mm
HP	hardpan (consolidated fines)		< 0.06 mm
WD	wood		
RC	concrete/ asphalt		
OT	other		

Step 1. Every time a cobble-sized particle is encountered during the pebble count, remove the cobble from the stream bed and visually estimate the percentage of the cobble's volume that has been buried by fine particles. Since visual estimates of volume and surface area are subject to large amounts of observer error, field crews should routinely calibrate their estimates with each other and with other field crews.

Step 2. In the spaces to the right of the pebble count data, record the embeddedness of all cobble-sized particles encountered during the pebble count.

Note: *The cobble embeddedness scores do not correspond with the specific particles in the pebble count cells to the left, but are merely a convenient place to record the data.*

Step 3. If 25 cobbles are not encountered during the pebble count, supplement the cobbles by conducting a "random walk" through the reach. Starting at a random point in the reach, follow a transect from one bank to the other at a randomly chosen angle. Once at the other bank reverse the process with a new randomly chosen angle. Record embeddedness of cobble-sized particles in the cobble embeddedness boxes on the transect forms until you reach 25 cobbles. If 25 cobble-sized particles are not present in the entire reach, then record the values for cobbles that are present.

Module D. Canopy Cover

This method uses the Strickler (1959) modification of a convex spherical densiometer to correct for over-estimation of canopy density that occurs with unmodified readings. Read the densiometer by counting the number of line intersections that are obscured by overhanging vegetation (see Figure 7). Taping off the lower left and right portions of the mirror emphasizes overhead vegetation over foreground vegetation (the main source of bias in canopy density measurements). All densiometer readings should be taken with the bubble leveled and 0.3 m (1 ft) above the water surface.

Step 1. Using a modified convex spherical densitometer, take and record four 17-point readings all taken from the center of each transect: a) facing upstream, b) facing downstream, c) facing the left bank, d) facing the right bank.

Note: *This method deviates slightly from that of EMAP (in which two additional readings are taken at the left and right wetted edges to increase representation of bank vegetation).*

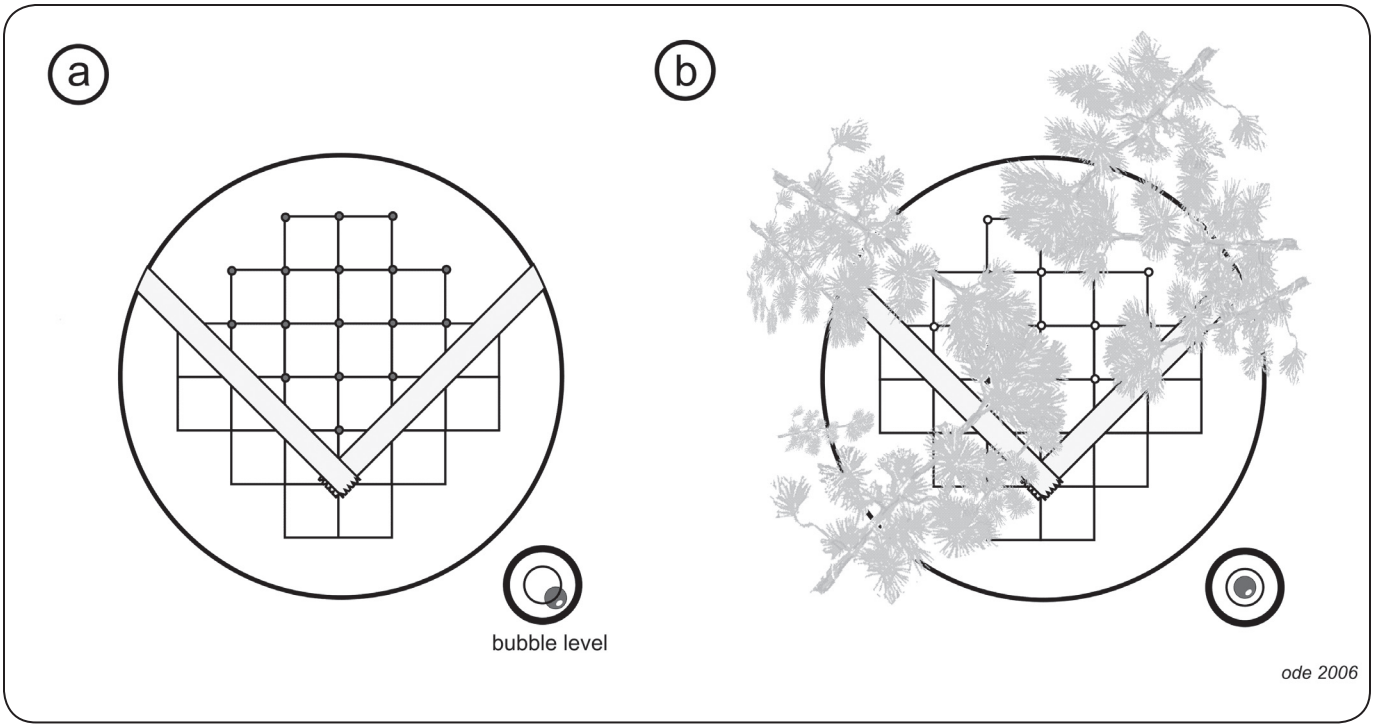

Figure 7. Representation of the mirrored surface of a convex spherical densiometer showing the position for taping the mirror and the intersection points used for the densiometer reading. The score for the hypothetical condition in (b) is 10 covered intersection points out of 17 possible. Note the position of the bubble level in (b) when the densiometer is leveled.

Module E. Gradient and Sinuosity

The gradient of a stream reach 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. The gradient (slope) of a stream reach is often strongly correlated with many BMI metrics and other physical habitat measures and is therefore very useful when interpreting BMI data.

The "full" physical habitat method uses 10 transect to transect measurements to calculate the average slope through a reach. Although this is a little more time intensive than the reach-scale transect measures used in the "basic" protocol, it results in more precise slope determination and the ability to quantify slope variability within a reach. Sinuosity (calculated as the ratio of the length of the flow path between the ends of the reach and the straight line distance between the ends of the reach, Kaufmann et al. 1999) is measured at the same time as slope. These two measurements work best with two people, one taking the readings at the upstream transect ("backsighting") and the other holding a stadia rod at the downstream transect. If you cannot see the mid point of the next transect from the starting point, use the supplemental sections (indicating the proportion of the total length represented by each section). Otherwise, leave these blank.

Note 1: *An auto level should be used for reaches with a percent slope of less than or equal to 1%. All methods (clinometer, hand level, or auto level) may be used for reaches with a percent slope of greater than 1%. The following description is for clinometer-based slope measurements, but the same principles apply to use of an auto or hand level.*

Note 2: *In reaches that are close to 1%, you will not know whether you are above or below the 1% slope cutoff before taking readings. In these cases, default to use of an autolevel.*

Step 1. Beginning with the upper transect (Transect K), one person (the measurer) should stand at the water margin with a clinometer held at eye level. A second person should stand at the margin of the next downstream transect (Transect J) with a stadia rod flagged at the eye level of the person taking the clinometer readings. Be sure you mark your eye level while standing on level ground! Adjust for water depth by measuring from the same height above the water surface at both transects. This is most easily accomplished by holding the base of the pole at water level.

Note: *An alternative technique is to use two stadia rods pre-flagged at the eye-height of the person taking the readings.*

Step 2. Use a clinometer to measure the percent slope of the water surface (not the streambed) between the upstream transect and the downstream transect by sighting to the flagged position on the stadia rod. The clinometer reads both percent slope and degree of the slope. Be careful to read and record percent slope rather than degrees slope (these measurements differ by a factor of ~ 2.2). Percent slope is the scale on the right hand side as you look through most clinometers (e.g., Suunto models).

Note: *If an auto level or hand level is used, record the elevation difference (rise) between transects and the segment length (run) instead of the percent slope.*

Step 3. If the stream reach geometry makes it difficult to sight a line between transects, divide the distance into two or three sections and record the slope and the proportion of the total segment length between transects for each of these sections in the appropriate boxes on the slope form (supplemental segments).

Note: *Never measure slope across dry land (e.g., across a meander bend).*

Step 4. Take a compass reading from the center of each main transect to the center of the next main transect downstream and record this bearing to the nearest degree on the slope and bearing section of the form. Bearing measurements should always be taken from the upstream to downstream transect.

Step 5. Proceed downstream to the next transect pair (I-J) and continue to record slope and bearing between each pair of transects until measurements have been recorded for all transects.

SECTION IVB. VISUAL ESTIMATES OF HUMAN INFLUENCE, INSTREAM HABITAT, AND RIPARIAN VEGETATION

The transect-based approach used here permits semi-quantitative calculations from visual estimates even though most are categorical data (i.e., either presence/ absence or size classes) because we can calculate the percentage of transects that fall into different categories. These modules are adapted directly from EMAP protocols with some modifications as noted.

Module F. Human Influence

The influence of human activities on stream biota is of critical concern in bioassessment analyses. Quantification of human activities for these analyses is often performed with GIS techniques, which are very useful but are not capable of accounting for human activities occurring at the reach scale. Reach scale observations are often critical for explaining results that might seem anomalous on the basis of only remote mapping tools.

Step 1. For the left and right banks, estimate a 10 x 10 m riparian area centered on the edges of the transect (see Figure 2). Record the presence of 11 human influence categories in three spatial zones relative to this 10 x 10 m square (between the wetted edge and bankfull margin, between the bankfull margin and 10 m from the stream, and between 10 m and 50 m beyond the stream margins): 1) walls/rip-rap/dams, 2) buildings, 3) pavement/cleared lots, 4) roads/railroads, 5) pipes (inlets or outlets), 6) landfills or trash, 7) parks or lawns (e.g., golf courses), 8) row crops, 9) pasture/ rangelands, 10) logging/ timber harvest activities, 11) mining activities, 12) vegetative management (herbicides, brush removal, mowing), 13) bridges/ abutments, 14) orchards or vineyards. Circle all combinations of impacts and locations that apply, but be careful to not double-count any human influence observations.

Step 2. Record the presence of any of the 11 human influence categories in the stream channel within a zone 5 m upstream and 5 m downstream of the transect.

Module G. Riparian Vegetation

Riparian vegetation (vegetation in the region beyond the bankfull margins) has a strong influence on the composition of stream communities through its direct and indirect roles in controlling the food base, moderating sediment inputs and acting as a buffer between the stream channel and the surrounding environment. These methods provide a cursory survey of the condition of the riparian corridor. Observations are made in the same 10 x 10 m riparian area used for assessing human influence (see Figure 2).

Note: *Riparian vegetation measurements should only include living or recently dead vegetation.*

The riparian vegetation categories used here were condensed from the EMAP version, which further breaks the canopy classes into different components. However, because we have consolidated EMAP categories into fewer categories rather than creating new categories, existing EMAP data can be easily converted to this format simply by combining the appropriate categories.

Step 1. Divide the riparian zone into three elevation zones: 1) ground cover (< 0.5 m), 2) lower canopy (0.5 m - 5 m), and 3) upper canopy (> 5 m). Record the density of the following riparian classes: 1) Upper Canopy–Trees and Saplings, 2) Lower Canopy–Woody Shrubs and Saplings, 3) Woody Ground Cover–Shrubs, Saplings, 4) Herbaceous Ground Cover–Herbs and Grasses, and 5) Ground Cover–Barren, Bare Soil and Duff. Artificial banks (e.g., rip-rap, concrete, asphalt) should be recorded as barren.

Step 2. Indicate the areal cover (i.e., shading) by each riparian vegetative class as either: 1) absent, 2) sparse (< 10%), 3) moderate (10-40%), 4) heavy (40-75%), or 5) very heavy (> 75%).

Module H. Instream Habitat Complexity

Instream habitat complexity was developed by the EMAP program to quantify fish concealment features in the stream channel, but it also provides good information about the general condition and complexity of the stream channel. Estimates should include features within the banks and outside the wetted margins of the stream.

Step 1. Record the amount of nine different channel features within a zone 5m upstream and 5m downstream of the transect (see Figure 2): 1) filamentous algae (long-stranded algal forms that are large enough to see with the naked eye), 2) aquatic macrophytes (include mosses and vascular plants), 3) boulders (> 25 cm), 4 and 5) woody debris (break into two classes- larger and smaller than 30 cm diameter), 6) undercut banks, 7) overhanging vegetation, 8) live tree roots and 9) artificial structures (includes any anthropogenic objects including large trash objects like tires and shopping carts). Indicate the areal cover of each feature as either: 1) absent, 2) sparse (< 10%), 3) moderate (10-40%), 4) heavy (40-75%), or 5) very heavy (> 75%).

SECTION 5
INTER-TRANSECT MEASURES

While most measures are taken at or relative to the main transects, a few measures are recorded at transects located at the midpoint between main transects. These are called "inter-transects".

Module B (Part 2) Pebble Counts (same as for transects, but no cobble embeddedness measures)

Step 1. Divide the wetted stream width by four to get the distance between the five points (Left Bank, Left Center, Center, Right Center and Right Bank) and use a measuring device to locate the positions of these points (a stadia rod is especially helpful here, see Figure 5). Once the positions are identified, lower a graduated rod through the water column perpendicular to both the flow and the transect to objectively select the particle located at its tip.

Step 2. With the graduated rod, measure the depth from the water surface to the top of the particle and record to the nearest cm.

Step 3. Remove the particle from the streambed, then measure and record the length of its intermediate axis to the nearest mm (see Figure 6). Alternatively, assign the particle to one of the size classes listed in the bottom of the transect form (see Table 3 for a list of size classes). Particle size classes may be estimated visually or with a quantitative measuring device (e.g., pass/ no-pass template, gravelometer). Regardless of the method, all particles less than 0.06 mm should be recorded as fines, while all particles between 0.06 mm and 2.0 mm should be recorded as sand. Field crews may want to carry vials containing sediment particles with these size ranges until they are familiar with these particle size classes.

Step 4. Record the presence (P) or absence (A) of any CPOM within 1 cm of each particle.

Module J. Flow Habitats

Because many benthic macroinvertebrates prefer specific flow and substrate microhabitats, the proportional representation of these habitats in a reach is often of interest in bioassessments. There are many different ways to quantify the proportions of different flow habitats (for example, see text on EMAP's "thalweg profile" below). Like the riparian and instream measures listed above, this procedure produces a semi-quantitative measure consisting of 10 transect-based visual estimates.

Note: *The categories used here are based on those used in the EMAP protocol, with pools combined into one class and cascades and falls combined into another class.*

Step 1. At each inter-transect, identify the proportion of six different habitat types in the region between the upstream transect and downstream transect: 1) cascades/falls, 2) rapids, 3) riffles, 4) runs, 5) glides, 6) pools, 7) dry areas. Record percentages to the nearest 5% — the total percentage of surface area for each section must total 100%.

SECTION 6 DISCHARGE

Stream discharge is the volume of water that moves past a point in a given amount of time and is generally reported as either cubic meters per second (cms) or cubic feet per second (cfs). Because discharge is directly related to water volume, discharge affects the concentration of nutrients, fine sediments and pollutants; and discharge measurements are critical for understanding impacts of disturbances such as impoundments, water withdrawals and water augmentation. Discharge is also closely related to many habitat characteristics including temperature regimes, physical habitat diversity, and habitat connectivity. As a direct result of these relationships, stream discharge is often also a strong predictor of biotic community composition. Since stream volume can vary significantly on many different temporal scales (diurnal, seasonal, inter-annually), it can also be very useful for understanding variation in stream condition.

This procedure (modified from the EMAP protocol) provides for two different methods for calculating discharge. It is preferable to take discharge measurements in sections where flow velocities are greater than 0.15 m/s and most depths are greater than 15 cm, but slower velocities and shallower depths can be used. If flow volume is sufficient for a transect-based "velocity-area" discharge calculation, this is by far the preferred method. If flow volume is too low to permit this procedure or if your flow meter fails, use the "neutrally buoyant object/ timed flow" method.

Note: *Programs that sample fixed sites repeatedly may want to consider installing permanent discharge estimation structures (e.g., stage gauges, wiers).*

Module K. Discharge: Velocity Area Method

The layout for discharge measurements under the velocity-area (VA) method is illustrated in Figure 8. Flow velocity should be measured with either a Swoffer Instruments propeller-type flow meter or a Marsh-McBirney inductive probe flow meter. Refer to the manufacturers' instrument manuals for calibration procedures.

VA-Step 1. Select the best location in the reach for measuring discharge. To maximize the repeatability of the discharge measurement, choose a transect with the most uniform flow (select hydraulically smooth flow whenever possible) and simplest cross-sectional geometry. It is acceptable to move substrates or other obstacles to create a more uniform cross-section before beginning the discharge measurements.

VA-Step 2. Measure the wetted width of the discharge transect and divide this into 10 to 20 equal segments. The use of more segments gives a better discharge calculation, but is impractical in small channels. A minimum of 10 intervals should be used when stream width permits, but interval width should not be less than 15 cm.

VA-Step 3. Record the distance from the bank to the end of the first interval. Using the top-setting rod that comes with the flow velocity meter, measure the median depth of the first interval.

VA-Step 4. Standing downstream of the transect to avoid interfering with the flow, use the top-setting rod to set the probe of the flow meter (either the propeller or the electromagnetic probe) at the midpoint of each interval, at 0.6 of the interval depth (this position generally approximates average velocity in the water column), and at right angles to the transect (facing upstream). See Figure 8 for positioning detail.

VA-Step 5. Allow the flow velocity meter to equilibrate for 10-20 seconds then record velocity to the nearest m/s. If the option is available, use the flow averaging setting on the flow meter.

Note: *Under very low flow conditions, flow velocity meters may register readings of zero even when there is noticeable flow. In these situations, record a velocity of 0.5x the minimum flow detection capabilities of the instrument.*

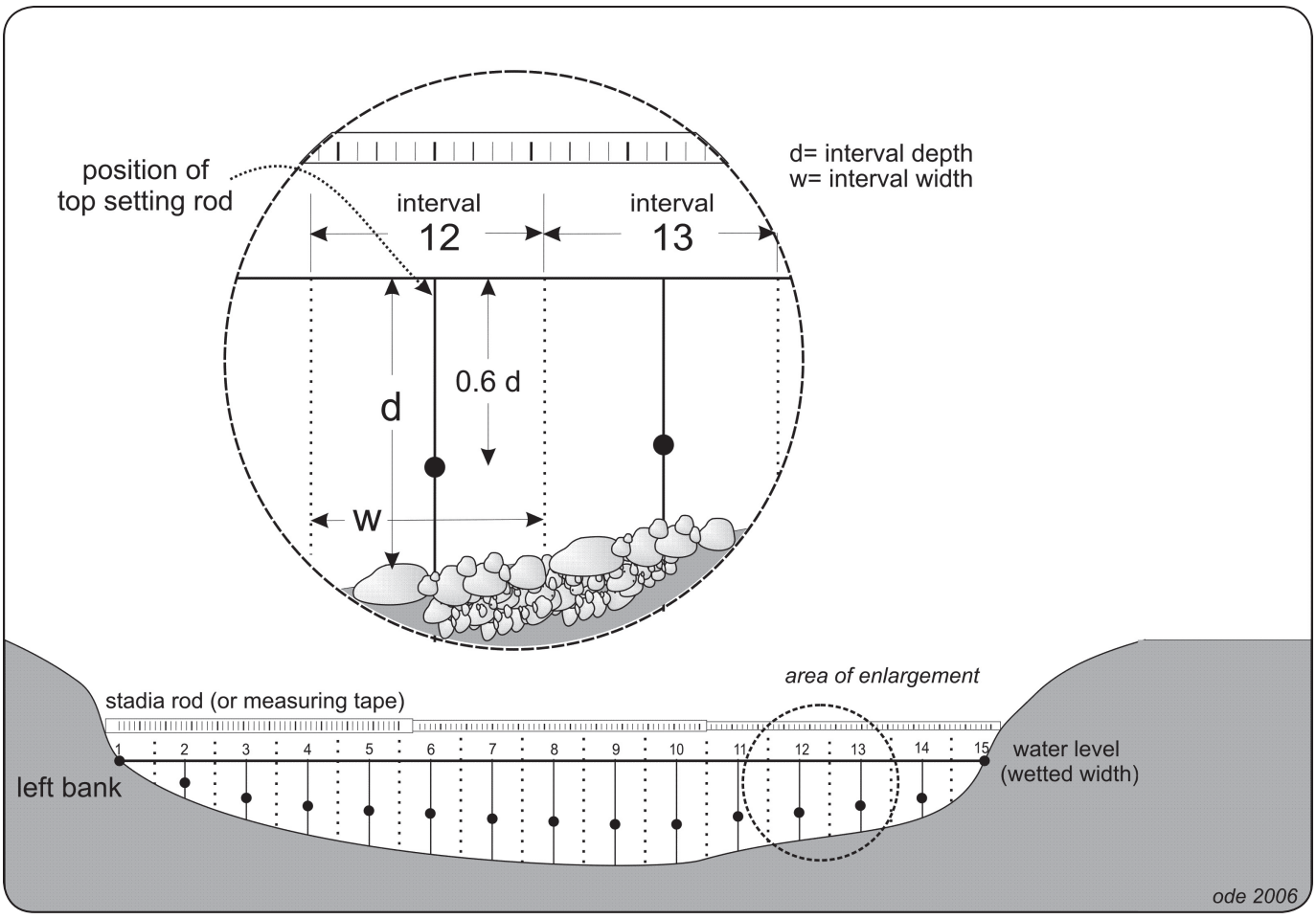

Figure 8. Diagram of layout for discharge measurements under the velocity-area method showing proper positions for velocity probe (black dots).

VA-Step 6. Complete Steps 3 through 5 on the remaining intervals.

Note: *The first and last intervals usually have depths and velocities of zero.*

Module L. Discharge: Neutrally Buoyant Object Method

If streams are too shallow to use a flow velocity meter, the neutrally buoyant object (NBO) method should be used to measure flow velocity. However, since this method is less precise than the flow velocity meter it should only be used if absolutely necessary. A neutrally buoyant object (one whose density allows it to just balance between sinking and floating) will act as if it were nearly weightless, thus it's movement will approximate that of the water it floats in better than a light object. To estimate the flow velocity through a reach, three transects are used to measure the cross-sectional areas within the test section sub-reach and three flow velocity estimates are used to measure average velocity through the test reach. To improve precision in velocity measurements, the reach segment should be long enough for the float time to last at least 10-15 seconds.

NBO-Step 1. The position of the discharge sub-reach is not as critical as it is for the velocity-area method, but the same criteria for selection of a discharge reach apply to the neutrally buoyant object method. Identify a section that has relatively uniform flow and a uniform cross sectional shape.

NBO-Step 2. The cross sectional area is estimated in a manner that is similar but less precise than that used in the velocity area method. Measure the cross sectional area in one to three places in the section designated for the discharge measurement (three evenly-spaced cross sections are preferred, but one may be used if the cross section through the reach is very uniform). Record the width once for each cross section and measure depth at five equally-spaced positions along each transect.

NBO-Step 3. Record the length of the discharge reach.

NBO-Step 4. Place a neutrally buoyant object (e.g., orange, rubber ball, heavy piece of wood, etc.) in the water upstream of the discharge reach and record the length of time in seconds that it takes for the object to pass between the upstream and downstream boundaries of the reach. Repeat this timed float three times.

SECTION 7
POST-SAMPLING OBSERVATIONS

Module M. Rapid Bioassessment Procedures Visual Assessment Scores (for Basic Physical Habitat, or optional supplement)

EPA's Rapid Bioassessment Procedures (RBPs, Barbour et al. 1999) include a set of 10 visual criteria for assessing instream and riparian habitat. The RBP has been used in the CSBP since its first edition (1995) and thus, this information is often valuable for comparison to legacy datasets. The criteria also have a useful didactic role since they help force the user to quantify key features of the physical environment where bioassessment samples are collected.

Module N. Additional Habitat Characterization (Full Physical Habitat only)

The RBP stream habitat visual estimates described in Step 1 are not included in the Full Physical Habitat version because they are generally replaced by more quantitative measurements of similar variables. However, we have found that three of the RBP measures are reasonably repeatable and include them in the reachwide assessment portion of the Full Physical Habitat version.

Note: *This is the only case in which a measurement included in the basic procedure is not included in the full.*

Module O. Reach Slope (for Basic Physical Habitat only)

Reach slope should be recorded as percent slope as opposed to degrees slope to avoid confusion. Slope measurements work best with two people, one taking the readings at the upstream transect and the other holding a stadia rod at the downstream transect. If you cannot see the mid point of the next transect from the starting point, use the supplemental sections (indicating the proportion of the total length represented by each section).

An auto level (with a tripod) should be used for reaches with a percent slope of less than or equal to 1%. All methods (clinometer, hand level, or auto level) may be used for reaches with a percent slope of greater than 1%. In reaches that are close to 1%, you will not know whether you are above or below the 1% slope cutoff. In these cases, default to use of an autolevel.

Step 1. Divide the reach into multiple segments such that stadia rod markings can be easily read with the measuring device to be employed (this is especially a factor for clinometer and hand level readings).

Step 2. Use a clinometer, hand level, or auto level to measure the percent slope of the water surface (not the streambed) between the top and bottom of each segment. Be sure to adjust for water depth by measuring from the same height above the water surface at both transects. Also be sure to record percent slope, not degrees slope. Record the segment length for each of these sections in the appropriate boxes on the BASIC slope form.

SECTION 8
OPTIONAL EXCESS SEDIMENT MEASURES

Future editions of these protocols will include supplemental modules, including a full discussion of the measurements used for calculating the excess sediment index (sometimes referred to as log relative bed stability, LRBS). However, since several of the measurements in EMAP's physical habitat protocols are interwoven into the layout of this protocol, a brief overview of the additional measurements collected for the LRBS calculations is included here for information purposes only. For detailed explanations of these measurements, consult Peck et al. 2004.

Woody Debris Tallies

Large woody debris (logs, snags, branches, etc.) that is capable of obstructing flow when the channel is at bankfull condition (just short of flood stage) contributes to the "roughness" of a channel. The effect of this variable is to reduce water velocity and thereby reduce the stream's competence to move substrate particles. The EMAP protocol tallies all woody debris with a diameter greater than 10 cm (~4") into one of 12 size classes based on the length and width of each object. Tallies are conducted in the zone between the main transects.

Thalweg Measurements

A stream's thalweg is a longitudinal profile that connects the deepest points of successive cross-sections of the stream. The thalweg defines the primary path of water flow through the reach. Thalweg measurements perform many functions in the EMAP protocols, producing measurements for the excess sediment calculations (residual pool volume, stream size, channel complexity) and flow habitat variability.

SECTION 9
OPTIONAL PERIPHYTON QUANTIFICATION

Periphyton Quantification

Characterization of periphyton has a dual role in bioassessments, as periphyton is both a food and habitat resource for benthic macroinvertebrates and fish and an effective bioindicator on its own. Quantification of periphytic resources will be covered under a separate SWAMP bioassessment protocol, but will include procedures for qualitative characterization of diatom assemblages, documentation of filamentous algal growth, and biomass quantification (e.g., ash-free dry mass and chlorophyll a).

SECTION 10
QUALITY ASSURANCE & CONTROL PROCEDURES

The SWAMP bioassessment group is currently developing guidelines for quality assurance and quality control for bioassessment procedures. Future revisions to this document will include guidance covering personnel qualifications, training and field audit procedures, procedures for field calibration, procedures for chain of custody documentation, requirements for measurement precision, health and safety warnings, cautions (actions that would result in instrument damage or compromised samples), and interferences (consequences of not following the standard operating procedure, SOP).

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DEFINITIONS OF TERMS USED IN SOP D

Terms & Definitions	
TERM	**DEFINITION**
ABL	California Department of Fish and Game's Aquatic Bioassessment Laboratory
Allocthonous	Derived from a source external to the stream channel (e.g., riparian vegetation) as opposed to autochthonous, which indicates a source inside the stream channel (e.g., periphyton).
Ambient Bioassessment	Biological monitoring that is intended to describe general biotic condition as opposed to a diagnosis of sources of impairment
Bankfull	The bankfull channel is the zone of maximum water inundation in a normal flow year (one to two year flood events)
BMI	Benthic macroinvertebrates: bottom-dwelling invertebrates large enough to be seen with the unaided eye
Cobble Embeddedness	The volume of cobble-sized particles (64-250 mm) that is buried by fine particles (<2.0 mm diameter)
CPOM	Coarse particulate organic matter (CPOM, particles of decaying organic material such as leaves that are greater than 1.0 mm in diameter)
CSBP	California State Bioassessment Procedures
DFG	California Department of Fish and Game
EMAP	The U.S. Environmental Protection Agency's Environmental Monitoring and Assessment Program
EPA	The U.S. Environmental Protection Agency
Fines	Substrate particles less than 0.06 mm diameter (not gritty to touch)
Inter-transects	Transects established at points equidistant between the main transects
MCM	Margin-Center-Margin alternative procedure for sampling low gradient habitats
ORD	EPA's Office of Research and Development
QAMP	Quality assurance management plan
RBP	EPA's Rapid Bioassessment Procedures
Reach	A segment of the stream channel
Riparian	An area of land and vegetation adjacent to a stream that has a direct effect on the stream.
RWB	Reach-wide benthos composite sampling method for benthic macroinvertebrates, also referred to as multi-habitat method
SCCWRP	Southern Coastal California Water Research Project
SNARL	Sierra Nevada Aquatic Research Laboratory
Substrate	The composition of a streambed, including both inorganic and organic particles
SWAMP	The State Water Resources Control Board's Surface Water Ambient Monitoring Program
Thalweg	A longitudinal profile that connects the deepest points at successive cross-sections of the stream. The thalweg defines the primary path of water flow through the reach

TERM	DEFINITION
Transects	Lines drawn perpendicular to the path of flow used for standardizing sampling locations
TRC	Targeted riffle composite sampling method for benthic macroinvertebrates
USFS	The United States Forest Service
Wadeable Streams	Streams that can be sampled by field crews wearing chest waders (generally less that 0.5 m - 1.0 meters deep)

APPENDIX A

FACTORS TO CONSIDER WHEN RECOMMENDING/ CHANGING BIOASSESSMENT METHODS

Beyond the primary considerations of precision and accuracy, there are at least five other key issues that SWAMP has considered and should consider in the future, when recommending or changing its official methods for bioassessment. These issues include:

1. Costs of Collecting Samples via Multiple Protocols – Collecting, processing, and interpreting samples using more than one method for each indicator (e.g., algae, macroinvertebrates, fish) per site adds significant costs to bioassessment monitoring programs. SWAMP should strive to identify the minimum set of protocols necessary for each indicator. However, this should not come at the expense of sound monitoring. If more than one method is needed to interpret the biological response, then this decision should be based on a cost-benefit assessment.

2. Costs of Maintaining Multiple SWAMP Protocols – While multiple methods for monitoring a given indicator may provide additional accuracy in specific habitats, there are significant costs to maintaining multiple protocols:

a. Need to maintain method-specific infrastructure (e.g., separate reference samples, separate indices of biotic integrity (IBIs), separate O/E models, etc.).
b. May lose or impair ability to compare across sites if different methods are used (see Issue 5 below).
c. Guidance on when to use methods becomes more complex. For example, we need to define very specifically which methods to use at each water body type; and thus, which tools can be used to interpret them.

Recommendation: *SWAMP should maintain as few protocols as necessary. If we elect to add new or modified protocols it should be because we have determined that the added value is worth all of the costs listed above.*

3. Separating Physical Impairment from Water Quality Impairment – One of the original reasons for adding a multihabitat component to SWAMP bioassessment programs was the potential for distinguishing physical and water quality impairment sources (see recommendations in Barbour and Hill 2002). In regards to macroinvertebrate indicators, the conventional wisdom has been that reachwide (RW, sometimes referred to as multihabitat or MH) samples should be relatively more responsive to physical habitat alteration (i.e., fine sediment inputs) than targeted-riffle (TR) samples because it is believed that erosional habitats take longer

to respond to sediment stresses, and because pockets of riffle habitat are thought to act as refugia from habitat loss. To the extent that this is true, RW and TR samples may offer complementary information that allows us to separate these sources of impairment.

While very few studies have addressed this conventional wisdom directly, recent studies suggest that this may not be as much a factor as previously believed. In a recent comparison of TR and RW samples at nearly 200 sites statewide, the ABL found at most weak evidence to support this notion (Rehn et al. 2007). Gerth and Herlihy (2006) came to the same conclusion in their analysis of ~500 sites in the eastern and western United States. However, this issue is far from resolved and SWAMP scientists currently are not in agreement regarding this issue. Since the majority of bioassessment programs in California have emphasized targeted riffle sampling, SWAMP will undoubtedly want to evaluate this question further before making any policy decision to discontinue TR sampling.

Recommendation: *Until this issue can be evaluated further and resolved to SWAMP's satisfaction, ambient macroinvertebrate sampling should include collection of both RW samples and richest targeted habitat (TR or MCM) samples at every site. (The TR method should be used where sufficient riffles are present, and the MCM method should be used at low-gradient sites where sufficient riffle habitat is not available.)*

4. Compatibility with Previous Data – To address this issue, at least three sets of macroinvertebrate sampling method comparisons have been conducted in California.

a. **Targeted Riffle Methods –** Comparisons are complete. Samples collected under the current TR protocols are considered interchangeable with both CSBP and SNARL samples (Ode et al. 2005, Herbst and Silldorff 2006).
b. **Low Gradient Sand-Dominated Streams –** Collaborative studies are currently underway between Water Board Regions 3 and 5, the Southern California Coastal Water Research Project (SCCWRP), and ABL to compare the performance of: (1) the "low-gradient" CSBP; (2) RW samples; and (3) a modification of the RW method designed to emphasize habitats along stream margins (MCM). The results of these low-gradient methods comparisons are not yet available.
c. **Targeted Riffle vs. Reachwide Methods –** A recent comparison of RW and TR samples collected from nearly 200 EMAP/ CMAP sites is in peer review press (Rehn et al. 2007). Results demonstrate remarkably similar performance of the methods across a wide range of habitats. Gerth and Herlihy (2006) recently published a similar analysis with the same conclusions. However, the bioassessment committee has yet to carefully review and discuss these analyses and their implications for SWAMP biomonitoring.

5. Comparability Among Sites – The ability to compare biological condition across sites is a common requirement of most ambient bioassessment programs. This type of analysis is confounded if different methods are used at these sites. One of the big advantages of reachwide (i.e., multihabitat) methods is that they can be applied anywhere because they don't require a specific habitat for sampling. Statewide

bioassessments and most regional programs will require the ability to compare their bioassessment results among multiple sites (e.g., within a watershed, within a region, statewide).

INTERIM RECOMMENDATIONS FOR MACROINVERTEBRATE SAMPLING (UPDATED DECEMBER 2006):

1. Until we can reach consensus on the outstanding issues (i.e., whether a single method for macroinvertebrate sampling will meet our needs, and the outcome of RW vs. MCM comparison studies for low-gradient wadeable streams/rivers), SWAMP recommends collecting both a reachwide (i.e., multihabitat) and a targeted habitat sample at each site. In high gradient streams, this means using both the RW and TR methods. In low-gradient streams, we recommend collecting both RW and MCM samples until the results are available from the low-gradient ("non-riffle") comparison. In rare cases where monitoring objectives cannot be met following these recommendations, the SWAMP Bioassessment Coordinator may authorize deviations. For example, where project-specific objectives differ from ambient monitoring, the SWAMP Bioassessment Coordinator may authorize alternate methods. In rare cases where funding is extremely limited and the cost of following the above recommendations would be prohibitive, the SWAMP Bioassessment Coordinator may authorize cost-saving options such as collecting both samples, but archiving one of the samples for later lab analysis.

2. SWAMP should develop guidance specifying when and where different methods should be used. For example, at "low gradient" sites, what is the slope cut-off (or other channel feature criteria to use) when deciding whether to apply TR or MCM? In addition, while SWAMP may eventually choose to adopt a single method (such as RW) at most sites, some regions may determine that the value of targeted habitat sampling merits continued sampling with supplemental protocols. In the latter case, or if SWAMP determines that distinct methods are needed for different habitat types, the guidance should specify the types of waterbodies or classes of waterbodies that require different methods.

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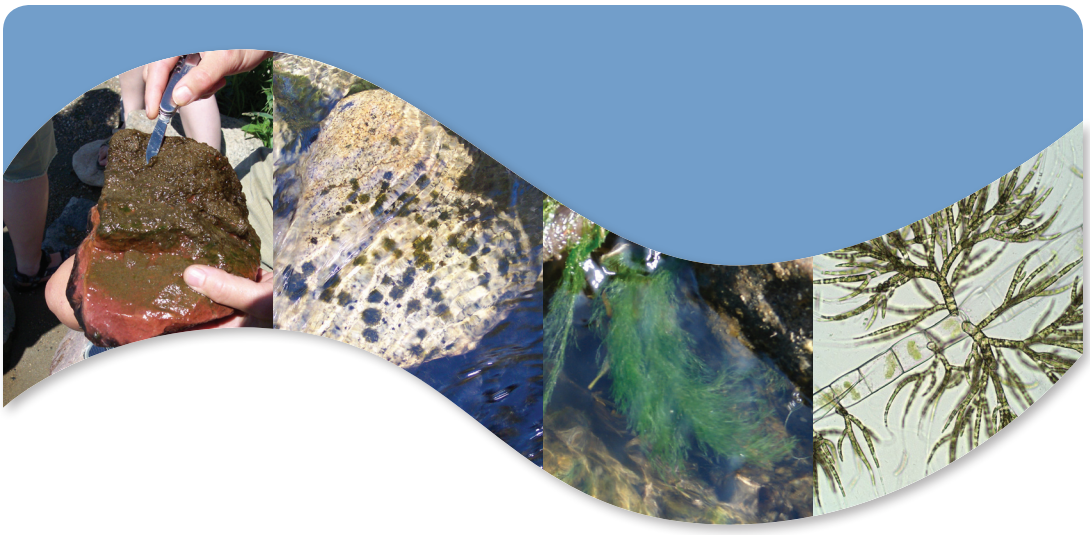
APPENDIX G:

SWAMP ALGAE FIELD SOP



SWAMP
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SWAMP Bioassessment Procedures **2010**

Standard Operating Procedures for Collecting Stream Algae Samples and Associated Physical Habitat and Chemical Data for Ambient Bioassessments in California

June 2009, updated May 2010

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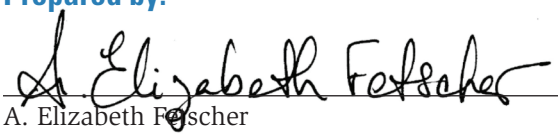

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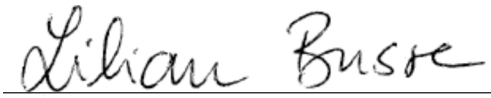

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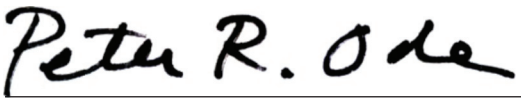

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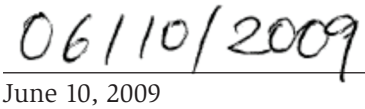

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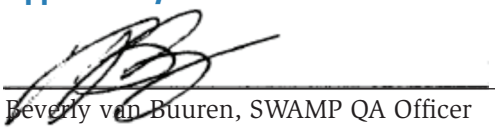

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This Standard Operating Procedures (SOP) manual represents the contributions of a wide range of researchers and field crews. The algal specimen collection methodology presented represents a modification of the U.S. Environmental Protection Agency's (EPA's) Environmental Monitoring and Assessment Program (EMAP) multihabitat sampling protocol (Peck et al. 2006). Point-intercept estimation of macroalgal cover has been adapted from the U.S. Geological Survey's (USGS's) National Water Quality Assessment (NAWQA) pilot procedures (J. Berkman, pers comm.), and assessment of microalgal thickness has been adapted from Stevenson and Rollins (2006). The physical habitat (PHab) methods are identical to those presented in the SWAMP Bioassessment protocol of Ode (2007), with the exception of the point-intercept method for determining algal cover, which is an add-on to the PHab pebble count procedure. The PHab procedures are, in turn, minor modifications of those used in EMAP and developed by EPA's Office of Research and Development (ORD, Peck et al. 2006).

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LIST OF ACRONYMS & ABBREVIATIONS L

Acronyms & Abbreviations	Definitions
AFDM	Ash-Free Dry Mass
BMI	Benthic Macroinvertebrate
chl a	Chlorophyll *a*
CPOM	Coarse Particulate Organic Matter
DO	Dissolved Oxygen
EMAP	Environmental Monitoring and Assessment Program (of the U.S. EPA)
GPS	Global Positioning System
IBI	Index of Biotic Integrity
MCM	Margin-Center-Margin
NAD	North American Datum
NAWQA	National Water Quality Assessment (of the U.S. Geological Survey)
NBO	Neutrally Buoyant Object
NNE	Nutrient Numeric Endpoints
OSPR	Office of Spill Prevention and Response (of the California Department of Fish and Game)
PHab	Physical Habitat
QAPrP	Quality Assurance Program Plan (of SWAMP)
ORD	Office of Research and Development (of the U.S. EPA)
RBP	Rapid Bioassessment Procedures
RWB	Reachwide Benthos
SOP	Standard Operating Procedures
SWAMP	Surface Water Ambient Monitoring Program
TRC	Targeted Riffle Composite

SECTION 1 INTRODUCTION

This document is the Standard Operating Procedure (SOP) for collecting and field-processing stream algae for the California State Water Resources Control Board's Surface Water Ambient Monitoring Program (SWAMP). Instructions are provided for the following:

- collection of samples for taxonomic identification of diatoms and soft-bodied algae
- collection of samples for determination of biomass based on chlorophyll *a* and ash-free dry mass (AFDM)
- estimation of percent algal cover

The document is designed to serve as a stand-alone SOP if algae are the only bioindicators being assessed at a given site. However, it can also serve as an add-on module to the existing SWAMP SOP for bioassessment using benthic macroinvertebrates (BMIs). Much of the procedure for collecting physical habitat (PHab) data is identical for these two assemblages. However, some PHab elements assessed in conjunction with BMI bioassessment are not included for algal bioassessment, because they are more specific to BMI habitat needs than to algae. Conversely, one PHab element for algal bioassessment (i.e., point-intercept estimation of algal cover) is not part of the BMI SOP. It should also be noted that, while the standard PHab protocol associated with BMI sampling includes both a "Full" and a "Basic" (simplified) version, a distinction between basic and full protocols for algae has not been established.

This SOP requires the reachwide benthos (RWB) sampling method to be used whenever algae bioassessment is conducted under the SWAMP program. Other appropriate sampling methods will be allowed if specific monitoring objectives require the use of alternative methods or if consistent data comparability in long-term monitoring projects is desired. For SWAMP funded projects, the project proponent must have the approval of the SWAMP bioassessment coordinator and the SWAMP Quality Assurance Officer before the use of alternative methods. For other projects and/or programs working towards SWAMP comparability, deviations should be approved by their project manager and project Quality Assurance (QA) officer.

For quick reference, Table 1 provides a list of elements common and distinct to the two SWAMP bioassessment assemblages. In general, if both BMIs and algae are being collected at a given site, the PHab procedure as described in Ode (2007) should be followed, with the exception of the pebble count, which should be conducted according to this SOP, because it incorporates instructions for algal cover point-intercept data collection. More specifically, if bioassessment involving the Full BMI protocol plus algae is to be implemented at a given site, practitioners should follow the Full protocol of Ode (2007), and add only Section 3.4 (re: water chemistry), Sections 4 and 5, and Sections 6.9-6.11 from this SOP.

Table 1
Sample and data collection elements included in algal and BMI bioassessment (Ode 2007).
X indicates elements included in algal bioassessment. F indicates elements that are part of the "Full" protocol for conducting BMI bioassessment, B corresponds to elements of the "Basic" BMI protocol, and O indicates elements that are "Optional".

Element	Algae[1]	BMIs
Layout of reach, marking transects, recording GPS coordinates	X	B, F
Notable field conditions	X	B, F
Temperature, pH, specific conductance, DO, alkalinity	X	B, F
Turbidity, Silica	O	O
Water chemistry for lab analysis (see list in Section 3.4)	X	
Algal Sampling for Taxonomic IDs	X	
Algal Sampling for Biomass Assessment	X	O
BMI Sampling for Taxonomic IDs		B, F
Wetted Width	X	B, F
Bankfull Dimensions	X	F
Depth and Pebble Count + CPOM	X	F
Percent Algal Cover (point-intercept with Pebble Count)	X	
Cobble Embeddedness	X	F
Canopy Cover	X	B, F
Gradient	X	B[2], F
Sinuosity		F
Human Influence	X	F
Riparian Vegetation		F
Instream Habitat		F
Bank Stability	X	B, F
Flow Habitat Delineation	X	B, F
Discharge	X	F
Photo documentation	X	B,F
Selected Rapid Bioassessment Procedure (RBP) visuals		F

1. A distinction between Basic and Full protocols for algae has not been established.
2. For BMIs, a single, reachwide measurement of gradient is required for Basic, but gradient is measured between all adjacent transect pairs for Full.

Depending upon the requirements of the monitoring effort, different components of this SOP might be incorporated or omitted. For instance, if stream productivity in terms of algae is the primary concern of the assessment, one may wish to collect only biomass samples and algal cover point-intercept data. Alternatively, one will need to collect algal assemblages (for quantification of diatom and/or soft-bodied algal taxa) in order to make more refined inferences about water quality and stream condition (e.g., by applying an algal Index of Biotic Integrity (IBI)).

This SOP is organized in such a way as to facilitate the inclusion or omission of certain elements based on the goals of the monitoring effort. A list of field supplies is provided in Appendix A. It is organized according to the materials needed for each type of sampling and data collection. In order to facilitate decisions about algal indicators to assess for program-specific needs, the introduction to Section 4 discusses what algal indicators serve which monitoring purposes.

SECTION 2 GETTING STARTED

Several considerations come into play when planning an algae-sampling effort. For instance, time of year can be an important determinant of stream algae abundance as well as the type of community likely to be encountered. Likewise, a minimum amount of surface water is a prerequisite to conducting bioassessment. The following section provides guidelines to help practitioners determine when sampling is appropriate for a given reach and also some pointers to help prepare for field work.

2.1 WHEN TO SAMPLE

It is recommended that sampling for stream algae be carried out during the same period as BMI sampling, generally from May through September, depending on the region. This time frame may eventually be modified (e.g., expanded) based on the results of ongoing index period studies.

It should be noted that high-velocity storm flows can remove macroalgae and biofilms from the stream bottom. Sampling must be done at least a month after any storm event that has generated enough stream power to mobilize cobbles and sand/silt capable of scouring stream substrates, in order to allow ample time for recolonization of scoured surfaces (Round 1991; Kelly et al. 1998; Stevenson and Bahls in Barbour et al. 1999).

2.2 BEFORE SETTING OUT FOR THE FIELD

- Proper precautions should be taken at all times in order to avoid transferring invasive organisms and pathogens between sites. This includes the implementation of effective equipment decontamination procedures. Refer to Appendix B for additional information.
- Use the equipment checklist provided in Appendix A to make sure all necessary supplies are brought along.
- Check with contract lab on sampling containers, and shipping and storage of samples.
- Have in mind at least three sites to visit per day (target two, but plan for at least one additional site as a back up if one of the first two sites is not useable.)
- Prepare, and double check, site dossiers to make sure they are complete with maps/directions to sites and scaled aerial photo(s). Bring along county maps, atlases, and Thomas Guides to further aid location of sites. Also bring along any site access permits, passes, and/or keys, as needed (and be aware that some landowners require notice prior to each site visit).

2.3 BEFORE LEAVING VEHICLE FOR SITE

Make sure the vehicle is parked in a safe spot and there are no "No Parking" signs. Stick a business card with cell phone number in the driver's window. Be sure to display the brown administrative pass placard if you are on National Forest land (or a letter of permission, if applicable).

2.4 DETERMINING WHETHER SITE IS APPROPRIATE FOR SAMPLING

Make an initial survey of the potential monitoring reach from the stream banks (being sure to not disturb the instream habitat). Ensure that there is sufficient water in the stream reach to facilitate collection of algae and water samples. In order for a reach to be in appropriate condition for sampling, at least half of the reach should have a wetted width of at least 1m, and there should be no more than 3 transects that are completely dry. If there is some flexibility in terms of where to place the sampling reach, strive for as few dry transects as possible (and preferably none).

Sites should be safe to sample and legally accessible. The time required to access the sampling sites should also be a consideration in planning which sites to visit, in order to ensure that sample holding times can be met (see Table 2 on page 11 for holding-time information).

3 REACH DELINEATION AND WATER CHEMISTRY SAMPLING

Before sample and data collection can begin, the monitoring reach must be identified and delineated. This requires setting up sampling transects along the stream reach of interest. Once the reach is delineated, information about reach location and condition will need to be documented. Water chemistry parameters must also be recorded, and certain samples collected.

A set of field forms for recording information about monitoring sites, algal samples, and associated water chemistry and PHab data is provided on the SWAMP website (see below). The field forms are also available in electronic version on an portable computer. It is imperative that you confirm throughout the data collection effort at each site that all necessary data have been recorded on the field forms correctly, by double-checking values, and confirming spoken values with your field partner(s). As a general practice, you should conduct a final check across all datasheets to confirm that there are no missing values before you leave the site, and rectify any blanks. *Note: Field forms may be updated periodically. It is imperative that field crews ensure that they are always using the most current field forms. Updated forms can be accessed from the SWAMP website at: http://swamp.mpsl.mlml.calstate.edu/resources-and-downloads*

3.1 DELINEATING AND DOCUMENTING THE MONITORING REACH

To delineate the monitoring reach, you will need to scout it in its entirety in order to make sure that it is of adequate length for sampling algae. During this process, try to stay out of the channel as much as possible, to avoid disturbing the stream bottom, which could compromise the samples and data that will be collected.

SWAMP's standard algae (and BMI) sampling layout consists of a 150 m reach or a 250 m reach, depending upon the average wetted width of the channel. In some circumstances (see below), reach length can be < 150 m, but this should be avoided whenever possible. If the actual reach length is other than 150 m or 250 m, this should be noted and explained on the field forms. Under these circumstances, you will need to determine the useable length of the reach, and how to space your transects so that you can fit them into the reach at equal distances from one to the next.

The wetted channel is the zone that is inundated with water and the wetted width is the distance between the sides of the channel at the point where substrates are no longer surrounded by surface water. Estimate the average wetted width of the reach. If this value is ≤ 10 m, you will end up using 150 m for your monitoring reach length. If the average wetted width is > 10 m, you will use a 250 m long reach.

To set up the monitoring reach, begin a little outside of what you anticipate will be the outer boundary (based on aerials and maps) and count 150 large steps, or 250 large steps (for most adults, a large step is roughly equal to a meter), by walking along the bank. This will give a rough idea about the location of the ends of the study reach. However, keep in mind that once this is determined, the actual distances between transects and intertransects (and consequently, the reach length) will need to be more accurately measured.

As you go, identify where hydrologic inputs that could potentially modify the water chemistry environment occur along the length of the reach. If possible, there should be no tributaries or "end-of-pipe" outfalls feeding into the channel within the monitoring reach. Other features that should not be present within a monitoring reach are: bridge crossings (which shade the stream bottom and can artificially reduce or prevent algal growth), changes between natural and man-made (i.e., concrete) channel bottoms, waterfalls, and impoundments (dams and weirs). If any of such features occur within the reach, and there is not enough room to accommodate a 150-m reach or 250-m reach entirely upstream or downstream of such a feature, then the reach can be somewhat < 150 m. Whatever the reach length turns out to be (150 m, 250 m, or other), record it on the datasheet under "Reach Length".

3.2 MARKING THE TRANSECTS

The monitoring reach will be divided into 11 equidistant main transects that are arranged perpendicularly to the direction of flow. There will also be 10 additional transects (designated "inter-transects"), one between each pair of adjacent main transects, to give a total of 21 transects per monitoring reach. Main transects are designated "A" through "K", while inter-transects are designated by their nearest upstream and downstream main transects ("AB", "BC", etc.).

Once you have identified the upper and lower limits of the monitoring reach, determine the coordinates of the downstream end using a Global Positioning System (GPS) set to the North American Datum 1983 (NAD 83)[3], and record this information in decimal degrees (to five decimal places) on the datasheet under "Reach Documentation". Install a colored flag at water's edge on one of the banks at this location to indicate the first "main transect", or "A". Establish the positions of the remaining transects and inter-transects by heading along the entire length of the monitoring reach (again, staying out of the water/channel as much as possible) and using the transect tape or a segment of rope of appropriate length to measure off successive segments of 7.5 m (for streams of wetted width ≤ 10 m), or 12.5 m (for streams > 10 m wetted width). For monitoring reaches of non-standard length, you will divide the total, targeted length of the reach by 20 to derive the distance between the adjacent main, and inter-, transects. As you measure off the distances, always follow the virtual, mid-channel line, and not the water's edge (which may be irregular, and not reflective of the true stream curvilinear distance).

3. Be aware that some GPS units re-set themselves to factory default settings when the batteries are changed. This can include the datum. Therefore, anytime you remove batteries from your unit, double check that the unit is still set to the NAD83 datum after the batteries have been replaced.

At the end of each measured segment as you head along the stream, mark the transect location on the bank with a flag. We recommend alternating between two different flag colors (e.g., orange could correspond to main transects, and yellow to inter-transects). Determine transect orientations, and where on the banks to place the flags, by visually projecting perpendicularly from the mid-channel to the banks. Refer to Figure 1 for a visual representation of proper transect alignment relative to the stream's direction of flow. When you have finished, the downstream-most flag will correspond to main transect "A", and the upstream-most flag (the 21st in the entire series of main and inter- transects) will correspond to main transect "K".

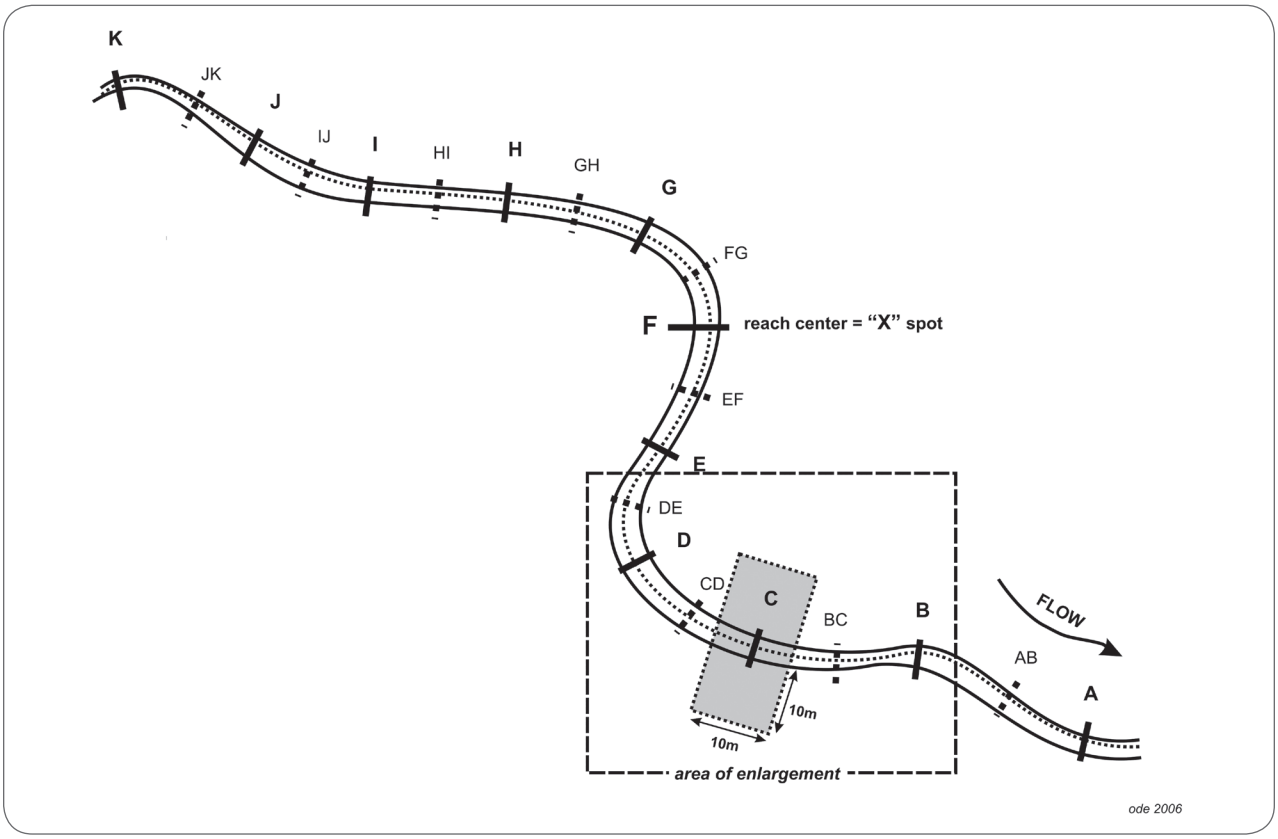

Figure 1. Reach layout geometry for physical habitat (PHab) and biological sampling showing positions of 11 main transects (A-K) and the 10 supplemental inter-transects (AB-JK). The area highlighted in the figure is expanded in Figure 11. Note: reach length = 150 m for streams ≤ 10 m average wetted width, and reach length = 250 m for streams > 10 m average wetted width (reprinted from Ode 2007).

3.3 NOTABLE FIELD CONDITIONS

Record under "Notable Field Conditions" any evidence of recent flooding, fire, or other disturbances that might influence algae samples. Especially note if flow conditions have been affected by recent rainfall, which can cause significant under-sampling of algal biomass and diversity. If you are unaware of recent fire or rainfall events, select the "no" option on the forms. Record the dominant land use and land cover in the area surrounding the reach by evaluating land cover within 50 m of either side of the stream reach. You can

use a scaled aerial photograph of the site and vicinity to guide you. *Note: Before heading out to the field, it is convenient to add a 150 m (or 250 m) line adjacent the stream to be sampled in order to get an idea about the anticipated approximate upstream and downstream boundaries of the monitoring reach.*

3.4 WATER CHEMISTRY

Measure and record common ambient water chemistry measurements (pH, dissolved oxygen (DO), specific conductance, alkalinity, and water temperature) just outside of the reach, at the downstream end, near the same location that the GPS coordinates were taken. This should be done in such a way that it does not interfere with biotic sampling and PHab data collection, but also in such a way that water samples are not compromised by other sampling activities upstream (e.g., by suspension of matter from the stream bottom into the water column, and consequently the introduction of this matter into the water chemistry samples).

Water chemistry measurements are typically taken with a handheld, water-quality meter (e.g., YSI, Hydrolab), but field test kits (e.g., Hach) can provide acceptable information if they are properly calibrated. For appropriate calibration methods, calibration frequency, and accuracy checks, consult the current SWAMP Quality Assurance Program Plan (QAPrP)[4], or follow manufacturer's guidelines. *Note 1: If characteristics of the site prohibit downstream entry, measurements may be taken at other points in the reach. In all cases, ambient chemistry measurements should be taken at the start of the survey (i.e., before algae sampling and PHab data collection). Note 2: A digital titrator (e.g., Hach) using low-concentration acid (such as 0.16N H_2SO_4) as the titrant is recommended for determining alkalinity in low-alkalinity streams (i.e., approximately 100 mg/L $CaCO_3$ or less).*

A suite of analytes must also be evaluated to aid in interpretation of the algal data. These are listed below. Consult the SWAMP QAPrP for specific instructions on the proper techniques for collecting, preserving, and storing these water samples until analysis.

- Nitrate as N (NO_3)
- Nitrite as N (NO_2)
- Ammonia as N (NH_3)
- Nitrogen, Total (TN)[5]
- Orthophosphate as P (dissolved; SRP)
- Phosphorous, Total (TPHOS)
- Dissolved Organic Carbon (DOC)
- Chloride (Cl)

- Silica as SiO_2, dissolved (Note: this analyte is recommended for research purposes, but is not part of the standard algae protocol)

4. This document is available online from the SWAMP website: http://www.swrcb.ca.gov/water_issues/programs/swamp/docs/qapp/swamp_qapp_master090108a.pdf
5. Total Nitrogen can be calculated from Total Kjeldahl Nitrogen (TKN), Nitrate (NO_3) and Nitrite (NO_2)

SECTION 4
REACHWIDE BENTHOS SAMPLING OF ALGAE

The following is a short introduction of several types of algal indicators that can be monitored as part of a bioassessment effort. For a more detailed discussion, see Fetscher and McLaughlin (2008). The most appropriate indicators to include in a given program will ultimately depend upon that program's goals, because the various indicators provide information at varying levels of resolution and applicability to different uses. Likewise, the various indicators require different levels of investment in terms of field work and lab work. Percent algal cover, for instance, is a rapid means of estimating algal primary productivity that can be carried out entirely in the field and is conducted in tandem with the PHab pebble count. Therefore, percent algal cover is an appropriate, fast, and inexpensive parameter for citizen monitoring groups if they are concerned about increased algal biomass. Other estimators of algal biomass include chlorophyll *a* and AFDM, which involve quantitative collection of algae, preservation, and subsequent laboratory analysis. Algal biomass is a key component of the California Nutrient Numeric Endpoints (NNE) framework. Higher resolution information about algal assemblages can be used in algal IBIs, and offers more in-depth insight into water quality. For this type of data, algal specimens must be collected quantitatively (and qualitatively, in the case of soft-bodied algae). The quantitative samples are then fixed/preserved carefully and subjected to taxonomic analysis.

While the percent algal cover data are recorded in conjunction with standard PHab procedures, and do not require the collection of samples, all the other types of data described in this protocol require reachwide benthos (RWB) sampling of algal specimens in a manner analogous to that which is carried out for BMIs.

All four of the algal samples described in this SOP: chlorophyll *a*, AFDM, diatom assemblage, and soft-bodied algal assemblage, can be obtained from a single composite sample generated by the RWB method. Which combination of these samples to prepare and submit for laboratory processing will depend on the needs of the monitoring program. To aid in the selection of algal indicators, Table 2 provides a summary of their attributes.

Table 2
Types of algal indicators and considerations for their assessment.

	Algal indicator for	Collection method	Collection vessel	Preservation/fixation methods and holding times	Qualitative live sample required?
Percent algal cover	Stream productivity measured as algal abundance	Point-intercept add-on to the PHab pebble count	N/A	N/A	N/A
Chlorophyll a[6]	Stream productivity measured as algal biomass; key indicator for the Nutrient Numeric Endpoints (NNE) framework	RWB sample collection	Glass-fiber filter	Wet ice, dark (foil-wrapped); freezing within 4h, and analysis within 28d	N/A
AFDM	Stream productivity measured as biomass of organic matter (including algae); indicator for the NNE framework	RWB sample collection	Glass-fiber filter (pre-combusted[7])	Wet ice, dark (foil-wrapped); freezing within 4h, and analysis within 28d	N/A
Diatoms	Used in IBIs. Indicative of factors such as trophic status; organic enrichment; low DO; siltation; pH; metals	RWB sample collection	50 mL centrifuge tube	Add 10% buffered formalin for a 2% final concentration immediately after collection; keep dark and away from heat	Optional
Soft-bodied algae[8]	Used in IBIs. Indicative of factors such as nitrogen limitation/ trophic status; siltation; pH; temperature, light availability, nuisance/ toxic algal blooms	RWB sample collection	50 mL centrifuge tube	Keep unfixed samples in dark on wet (NOT DRY) ice; add glutaraldehyde (to a 2.5% final concentration) as soon as possible, but no later than 4 days after sampling; after fixing, keep dark and away from heat	Required

6. It is valuable to assess both chlorophyll *a* and phaeophytin *a* (the degradation product of the former) content of algal samples, as this may provide a more robust assessment of algal biomass.
7. Precombustion is recommended in order to remove any possible residual organic matter from the filter.
8. For the purposes of this SOP, the soft-bodied assemblage includes cyanobacteria (an explanation of the rationale for this is provided in Fetscher and McLaughlin 2008)

4.1 GENERAL CONSIDERATIONS FOR SAMPLING ALGAE

This SOP describes the RWB method for collecting stream algae. It employs an objective approach for selecting sub-sampling locations that is built upon the 11 main transects described in the previous section. This approach is analogous to the SWAMP procedure for BMI sampling (Ode 2007), and is ultimately based on EPA's Environmental Monitoring and Assessment Program (EMAP; Peck et al. 2006). After collection, the 11 sub-samples are composited into a single sample per site (sampling reach).

The RWB method can be used to sample any wadeable stream reach since it does not target specific habitats. Because sampling locations are defined by the transect layout, the position of individual sub-sampling spots may fall within a variety of "erosional[9]" or "depositional[10]" habitats, each of which has implications for the type of substrate likely to be encountered and therefore the type of sampling device to use.

For the RWB method, the sub-sampling position alternates between left, center, and right portions of the transects, as one proceeds upstream from one transect to the next. These sampling locations are defined as the points at 25% ("left[11]"), 50% ("center") and 75% ("right") of the wetted width in high-gradient systems, and at "margin-center-margin" (MCM) positions in low-gradient systems. The RWB-MCM method should be only used in low-gradient streams where channel substrates are nearly uniform, resulting in low diversity within the channel. The interim cut-off between "low" and "high" gradient is 1%. Best professional judgment can be used to estimate whether the stream reach should be treated as low- or high-gradient. However, if there is uncertainty about the gradient, it should be measured prior to collecting the biotic assemblage samples. See Section 4.2 for specific instructions about where algae sampling locations should be positioned at the margins of low-gradient sampling reaches.

Algae should be sampled prior to PHab data collection (described in Sections 6-8), so as not to disturb the algae by trampling the transects, as occurs during the PHab process. Furthermore, to avoid disturbing the transects for eventual collection of PHab data, as with BMIs, algae should be collected at a location that is systematically offset from each transect (see Section 4.2).

4.2 COLLECTION OF ALGAE IN CONJUNCTION WITH BENTHIC MACROINVERTEBRATES

If only algae (or only BMIs) are being collected for bioassessment, then the specimens should be collected 1m downstream of the transects. If both assemblages are being sampled, then the algae should be collected upstream of the spot where the BMIs are collected, according to the schematic in Figure 2. BMIs must be collected BEFORE algae at each of the transects, in order to minimize the chances disturbing BMIs during algal collection. After the BMIs are collected at each spot, the algae sample should be taken ¼ m upstream

9. Erosional – habitats in the stream that are dominated by fast-moving water, such as riffles, where stream power is more likely to facilitate erosion (suspension) of loose benthic material than deposition; examples of "erosional" substrates include cobbles and boulders.
10. Depositional – habitats in the stream that are dominated by slow-moving water, such as pools, where deposition of materials from the water column is more likely to occur than erosion (or (re)suspension) of bed materials; examples of "depositional" substrates include silt and sand.
11. For our purposes, "left" is defined as the left bank when facing downstream.

from the center of the upper edge of the "scar" in the stream bottom left from the BMI sampling. It is important to make sure that the surface from which algae will be collected has not been disturbed (by the BMI sampling, or otherwise) prior to sampling the algae.

Note: If only algae (and not BMIs) are being collected in a low-gradient reach, the collection location should be 1 m downstream of the transect and, for each of the "margin" positions, at a distance of 15 cm from the wetted margin of the bank. Fifteen centimeters is chosen because it is approximately ½ the width of a D-frame net.

If duplicates are to be sampled (of either or both assemblages), locations for sampling them should be arranged as depicted in Figure 2 (the duplicates are shown in light grey). *Note: For convenience, only Transects A through C are shown, but the same pattern of placement should be rotated across all 11 transects.*

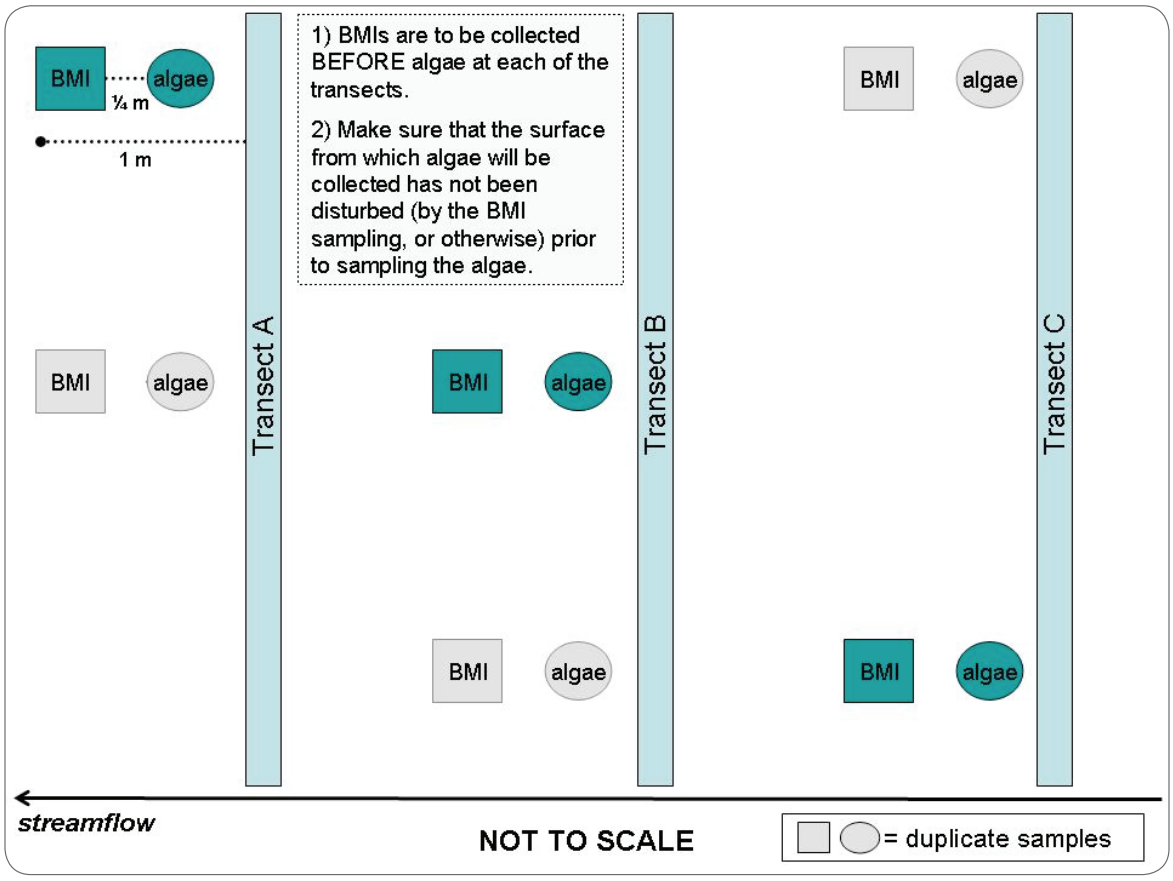

Figure 2. Sampling array for collection of algae, BMIs, and duplicates of each assemblage. For convenience, only Transects A through C are shown, but the same pattern of placement should be rotated across all 11 transects.

4.3 PROCEDURE FOR COLLECTION OF QUANTITATIVE ALGAL SAMPLES

During all phases of algae sampling and processing, in order to preserve specimen integrity, every attempt should be made to keep the sample material out of the sun, and in general, to protect the algae from heat and desiccation, as much as possible. This is necessary in order to reduce the risk of chlorophyll *a* degradation, limit cell division post-collection, and curb senescence/decay of live soft-bodied algae (especially for the qualitative samples; see Section 5.4). The need to maintain the integrity of the algal samples during collection and processing should always be borne in mind when planning the sampling scheme for a given site.

In addition, before sampling at any given site, the dish tub that will contain sample material must be scrubbed with a stiff-bristled brush or scouring pad and thoroughly rinsed with stream water, so that no algal material is carried over from the previous site to contaminate the current sample. The same applies to all other algae sampling apparati (toothbrushes for scrubbing, graduated cylinders, turkey basters, PVC and rubber delimiters, spatulas, syringe scrubbers, etc.).

4.3.1 Identifying the Sampling Locations

As with BMIs, algae sample collection should begin at Transect A and proceed upstream to Transect K. Except in circumstances in which the substrate to be sampled cannot be removed from the stream, a single sample of substrate material that corresponds to the objectively determined sampling point is gathered at each transect and placed in the plastic dish tub. The sample should always be collected from the substrate that is "uppermost" within the stream, and therefore has the highest probability of exposure to sun. For example, if at a given sampling point there is a thick layer of macroalgae above the stream bottom, the substrate collected at that point would be macroalgae itself, not the cobble, sand, or whatever other substrate lies beneath it. Proceeding from transect to transect with the dish tub, the sample collector rotates through the three collection positions in the following order: left at the first transect ("A"), center at the next transect ("B"), right at the next transect ("C"), then back to the left side ("D"), and so on through Transect K.

As substrates are gathered, a tally is taken of the number of samples that correspond to each of the classes of sampling device based on the surface area they sample: 1) 12.6 cm^2 for the PVC or rubber delimiters, and 2) 5.3 cm^2 for the syringe scrubber. The tallies are recorded in the Algae Samples field form under Collection Device. This information will ultimately be used to determine total stream surface area sampled at each site, which in turn will be used to calculate the soft-bodied algal biovolume and the biomass values. It may be helpful to use a tally meter in order to avoid having to carry a datasheet during substrate collection.

4.3.2 Collecting Erosional Substrates

If the substrate type that falls under the sampling spot is in erosional habitat and can be removed from the stream (e.g., a cobble, a piece of wood, or a piece of coarse gravel with an exposed surface area of at least 12.6 cm^2), carefully lift the substrate, moving slowly in an effort to disturb its top surface as minimally as

possible, and remove it from the stream. Then wipe any excess sand, silt, or BMIs, if present, off the bottom of the piece of substrate, and place it in the dish tub. It is helpful to place the substrate in such a way that makes it obvious what surface was facing upward when it was removed from the stream. Eventually, when you isolate a sample of algae from this substrate, you will want to obtain your sample from the portion of the substrate that had been exposed to the surface of the stream (and not buried) during the period leading up to the sampling event. For pieces of substrate with an exposed surface area that is < 12.6 cm^2, the PVC delimiter should be used (Section 4.3.3).

Be sure to place the substrate (e.g., cobble) in the dish tub in such a way that surfaces covered with non-target algae are not rubbing against anything, which could cause non-target algae to slough off into the tub, thus artificially inflating the amount of algae collected. To avoid this problem, and especially if a large number of cobbles are likely to be sampled across a given stream reach, one may choose to isolate the algal specimen from each cobble as it is selected, rather than collecting all the cobbles into the dish tub and then isolating the algal specimens from them after all transects have been sampled. See Section 4.3.9 for further elaboration on this alternative approach.

Figure 3. PVC Delimiter

4.3.3 Collecting Depositional Substrates

If the substrate type that falls under the sampling spot is removable and is in depositional habitat (e.g., silt, sand, fine gravel), and/or has an exposed surface area per particle that is < 12.6 cm^2, you will use a PVC delimiter. This is a plastic coring device with an internal diameter of 4 cm (Figure 3). Instructions for making a PVC delimiter are provided in Appendix C.

Isolate a specific quantity of sand/silt/gravel, centered on the sampling spot, by pressing into the top 1 cm of sediment with a PVC delimiter. Gently slide a masonry or kitchen spatula beneath the delimiter, being careful to keep the collected sediment contained within. Pull the PVC delimiter out of the water (with the spatula still in place) and remove any extra sediment from the spatula around the outside of the delimiter. Transfer the contents held in the delimiter by the spatula to the dish tub. Be sure not to pour the sediment sample on top of any cobbles that may be in the dish tub, as this could result in the sloughing of non-target algae from the cobbles into the dish tub, thus artificially inflating the amount of algae collected.

4.3.4 Collecting Sections of Macroalgae

If the substrate you hit on a given transect is a mass of macroalgae (e.g., a mass of attached filamentous algae underwater, or an unattached, floating mat that is believed to be native to the reach being sampled, and not imported from upstream), position the spatula directly under the macroalgae and press the PVC delimiter into the algae to define a 12.6 cm^2 area. Use a sharp razor blade or knife to cut away and discard any extra material from around the edges of the delimiter (do not simply pull it away, as this will distort the specimen and remove biomass from the targeted material). Add the macroalgal specimen that was isolated by the PVC delimiter to the dish tub.

When collecting a mass/mat of macroalgae, it is important to capture the full thickness of the macroalgae within the delimiter. To do this, from the side of the sampling area, slide your hand under the mat to feel where the bottom is, slide the spatula down to that spot, and then press the PVC delimiter downward slowly to "sandwich" the targeted section of macroalgae between the delimiter and the spatula. It is important to try not to bunch the macroalgae up nor stretch it out unnaturally, as the goal is to collect a representative sample of the algae as it occurs in the stream.

4.3.5 Collecting Sections of Macrophytes

If the substrate to be sampled is part of an immersed macrophyte, or old, dead leaves settled at the bottom of a pool, use the PVC delimiter/spatula combination to isolate a 12.6 cm^2 section of substrate that has been exposed to the surface of the stream. As with the macroalgae (Section 4.3.4), cut away and discard the extra material that falls outside the delimiter using a razor blade.

Figure 4. Syringe Scrubber

4.3.6 Collecting from Concrete, Bedrock, and Boulders

If the substrate falling under a sampling spot cannot be removed from the water (as in the case of bedrock, a boulder, or a concrete channel bottom), use a "syringe scrubber" device (Davies and Gee 1993; Figure 4) to collect an algae sample underwater. Instructions for making a syringe scrubber are provided in Appendix C.

To use this device, affix a fresh, white scrubbing pad circle onto the bottom of the syringe plunger using the Velcro® hooks on the end of the plunger. Press the plunger down so that the bottom of the scrubbing pad is flush with the bottom of the barrel. Then submerge the instrument, press the syringe firmly against the substrate, and rotate the syringe scrubber 3 times in order to collect the biofilm from the substrate surface onto the scrubbing pad. If the surface of the substrate where your sampling point fell is not flat enough to allow for a tight seal with the syringe barrel, objectively choose whatever sufficiently flat area on the exposed face of the substrate is closest to where the original point fell, and sample there.

After sampling, and before removing the syringe scrubber from the substrate, gently retract the plunger just slightly, so it is not up against the substrate anymore, but not so much that it pulls a lot of water into the barrel. Carefully slide the spatula under syringe barrel (which should be pulled just slightly away from the substrate on one side to allow the spatula to slide under), trying not to allow too much water to rush into the barrel. Then pull the instrument back up out of the water with the spatula still firmly sealed against the syringe-barrel bottom.

Hold the syringe scrubber over the dish tub and then remove the spatula, allowing any water to fall into the tub. Carefully detach the pad from the plunger and hold the pad over the tub. Using rinse water sparingly, remove as much algal material from the pad as possible by rinsing it off with the wash bottle, or a turkey baster, filled with stream water (from the current site—never carried over from a previous site), and

wringing it into the dish tub before discarding the used pad. Start this process by rinsing from the backside of the pad (the side that had been affixed to the plunger) to "push" the collected algae forward out of the front surface of the pad.

It is recommended that a fresh (new) pad be used each time a sample is collected, even within the same stream reach. Under no circumstances should the same pad be used at more than one site.

4.3.7 Collecting from Other Substrate Types

If other substrate types are encountered, they can be sampled from as long as there is good reason to believe that they were not recently introduced into the stream (e.g., by flowing from the upstream regions, or by recently falling into the stream), as they would then not be representative of the local instream environment. Use the collection instrument you deem to be most appropriate to sample the substrate and, as with any substrate, be sure to account for the surface area sampled (in this case, using the "Other" box on the Collection Device portion of the field forms).

4.3.8 Removal of Algae from Collected Substrates

After having sub-sampled substrates across the monitoring reach, there should be 11 transects' worth of material in the dish tub. Depending on the types of habitats in the stream and substrates encountered, the tub may contain cobbles, and/or sand, and/or gravel, and/or small pieces of wood, macroalgae, or macrophyte. Now a measured quantity of the algae clinging to these substrates must be removed and suspended in water to form a "composite sample" according to the instructions in the following sections.

For erosional substrate types that were removed from the stream (e.g., cobbles and small pieces of wood), use a rubber delimiter to isolate a 12.6 cm^2 area from which algae will be removed. A rubber delimiter can be made from a mountain bike tube with a hole cut out and reinforced with an appropriately sized rubber washer (Figure 5). Appendix C describes the procedure for making a rubber delimiter.

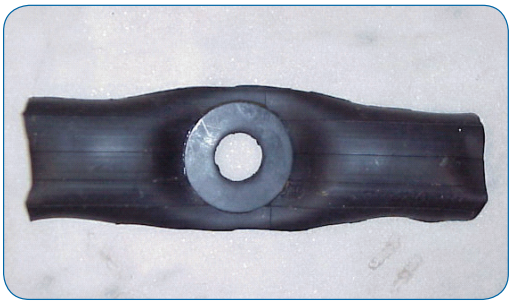

Figure 5. Rubber Delimiter

Wrap the rubber delimiter around the substrate to expose the desired sampling surface through the hole. Take care to ensure that the surface that will be scrubbed is truly the upper (generally at least somewhat "slimy") surface of the substrate as it had been oriented in the stream. Dislodge attached algae from this area by brushing it with a firm-bristled toothbrush (remember that this toothbrush must first have been thoroughly rinsed since the previous site to avoid contamination with algal specimens from other streams). If there is a thick mat of algae, or the algae is firmly encrusted on the surface of the substrate, use forceps or a razor blade first to dislodge the larger matter and put this in the dish tub. Then scrub the area with the brush.

Make sure that the entire surface within the delimiter has been scrubbed well in order to remove all the algae in that area. Fill a wash bottle or turkey baster with stream water from the current site (never carried over from a previous stream). Using as minimal a volume of water as possible, rinse the scrubbed algae from the sample area into the dish tub. Take care to squirt water only on the surface that is showing through the hole in the delimiter, and not anywhere else on the substrate's surface. It is helpful to invert the rock when rinsing so that the target surface is facing down toward the dish tub, and the rinsate drips off the sampling spot directly into the tub rather than flowing along the (non-target) sides of the substrate. Use water sparingly for each piece of substrate, because you should attempt to use no more than 400-500 mL total for the full suite of 11 samples collected along the transects (this includes any water used for rinsing algae off of sampling devices into the dish tub). After scrubbing is complete, rinse the delimiter and the brush into the dish tub, also. The scrubbed part of the substrate should feel relatively rough when you have finished, meaning that essentially all of the algae have been removed. After the sampling area on the piece of substrate has been thoroughly scrubbed and rinsed, the piece of substrate can be returned to the stream.

For depositional samples (e.g., silt, sand, or gravel), there is no need to isolate a specific area of the substrate within the dish tub, because the sample area was pre-isolated by using the PVC delimiter during collection. Simply massage all the sand and/or silt in the dish tub thoroughly between the fingers to dislodge any clinging algae. For pieces of gravel, use a toothbrush to remove algal material from surfaces.

Rinse the sediment thoroughly (but as sparingly as possible) with stream water so as to create a suspension of the dislodged microalgae (i.e., the sample). The final volume of the sample liquid in the dish tub will be measured before the algal taxonomic and biomass samples are prepared (described below). To do this, the liquid in the tub will be separated from the rinsed sediment such that the volume measured does not include sediment. After the liquid sample has been retrieved and measured, the rinsed sediment will be discarded back into the stream.

Other types of substrate, like pieces of macrophyte or dead leaves that had been collected with the PVC delimiter, should also be massaged between the fingers and rinsed into the tub in order to remove the algae coating them.

For macroalgal clumps there is a special step required for processing the samples. This procedure is described in detail in Section 5.3.

4.3.9 Alternative Approach: Processing Samples at Each Transect

It is also acceptable to isolate the algal specimens from each "piece" of substrate collected before moving on to the next transect. This approach has the disadvantage of requiring that all algae sampling/scraping tools be carried along with the collector as s/he proceeds up the stream, and that s/he pause to isolate the algae several times across the stream reach rather than one time at the end of all the transects. However, it limits the amount of substrate material that needs to be carried in the dish tub, thus making it lighter. This could be particularly important if a large number of cobbles are encountered across sampling points, such that it

could be difficult or impossible to carry them all to Transect K, or to carry them in such a way that non-target algae can easily be prevented from sloughing off into the tub via abrasion. For convenience, one may elect to wear a fisherman's vest to facilitate carrying all the algae sampling/scraping tools that will need to be brought along on the substrate sampling trip if employing this alternative approach.

SECTION 5
ALGAL SAMPLE PROCESSING

Four different types of laboratory samples may be prepared from the composite sample:

- Identification/Enumeration Samples
  1. Diatoms
  2. Soft-bodied algae
- Biomass Samples
  3. Chlorophyll *a* ("chl *a*")
  4. Ash-free dry mass ("AFDM")

5.1 GENERAL CONSIDERATIONS FOR PROCESSING ALGAL SAMPLES

The general process for sample preparation is as follows. The Identification/enumeration samples are each aliquoted into 50-mL centrifuge tubes and chemically fixed (preserved). Diatom samples are fixed in the field with formalin immediately following collection, and soft-bodied algae samples are fixed in a laboratory with glutaraldehyde within four days of collection. The chlorophyll *a* and AFDM samples are collected on filters in the field and stored on wet ice, and then frozen as soon as possible after returning from the field (and within four hours of collection). The filters are kept frozen until analysis, which should occur within 28 days of collection. If the field crew is spending the night in a hotel, it is necessary to buy dry ice to freeze the biomass filters upon finishing the day's fieldwork, and to keep them on dry ice until the samples can be transferred to the freezer back at the lab.

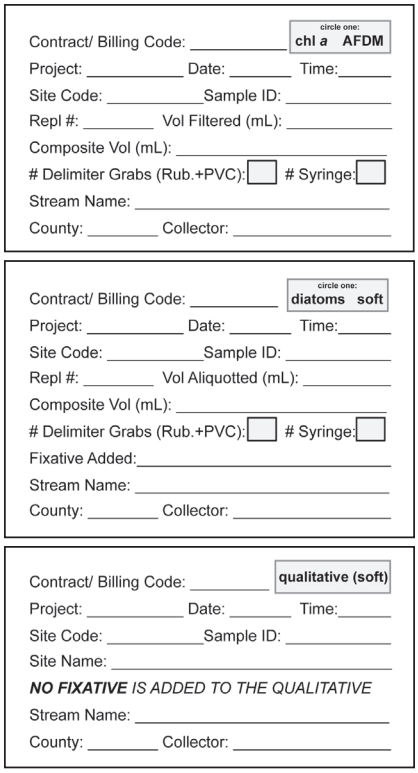

Figure 6. Labels for biomass and taxonomic identification samples.

Algae sample labels are shown in Figure 6. Recorded on each sample label are the volume of the composite sample (described in Sections 5.2.1 and 5.3.2), and the TOTAL area of stream bottom sampled (based on which sampling devices were used; described in Sections 4.3.2 - 4.3.7), as well as the volume of sample aliquoted (for the taxonomic ID samples) or filtered (for the chlorophyll *a* and ADFM samples). All of these values should be recorded on the field forms, as well, under the Algae Samples section. On the sample labels, the sample type: "chl *a*", "AFDM", "diatoms", or "soft" is circled, and all the remaining information on each label, like Site Code, Date, and site coordinates is filled out.

Before preparing the algae samples it is necessary to determine two things:

- Are there any clumps of macroalgae in the composite sample (as opposed to just microalgae suspended in liquid)?

AND

- Is a soft-bodied algal taxonomic sample going to be prepared?

The answers to these questions will determine the course of action for preparing the algae samples for a given site:

- If there is no macroalgal clump, liquid composite sample will simply be added to each taxonomic ID sample tube (40 mL for diatoms and 45 mL for soft-bodied algae). Biomass samples will also be prepared using the liquid composite sample, as is.
- If there is a macroalgal clump present, but no soft-bodied sample will be prepared, the entire clump will be chopped into fine bits (resulting in strands that are eyelash-length or shorter) and incorporated directly into the liquid portion of the composite sample, and the mixture will be shaken to homogenize it before preparing the diatom and/or biomass samples.
- If there is a macroalgal clump AND a soft-bodied algal taxonomic ID sample is to be prepared, then a more complex procedure must be employed in order to properly process the macroalgae before preparing the various samples.

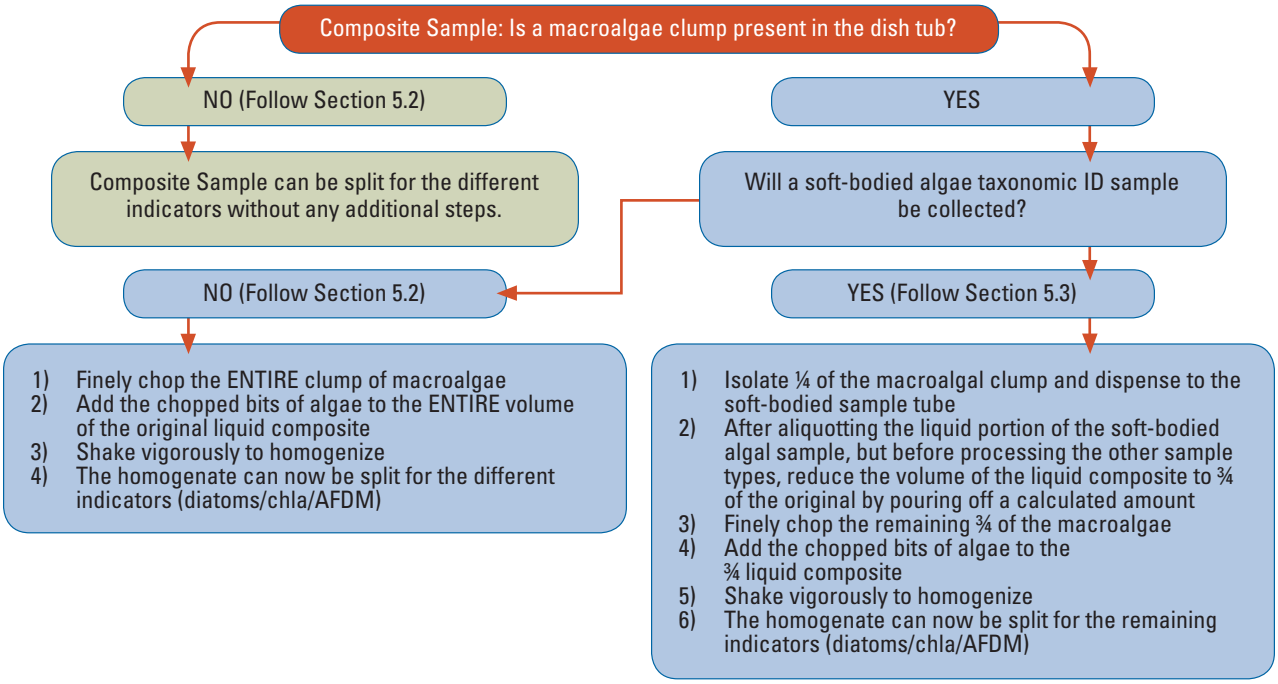

Figure 7. Summary of major sample-processing decision points based on presence of macroalgal clump(s) and need to prepare soft-bodied algal samples.

Figure 7 provides a summary of the various sample-processing steps that are involved, and the following sections describe the procedures in detail. Use Section 5.2. if there is NO macroalgal clump present in the dish tub OR a soft-bodied algal sample will be NOT be prepared. Use Section 5.3 if there IS a macroalgal clump present AND a soft-bodied algal sample will be prepared.

5.2 SAMPLE PROCESSING WHEN THERE IS NO MACROALGAL CLUMP OR WHEN NO SOFT-BODIED SAMPLE IS BEING PREPARED

This section describes the sample-processing procedure for the situation in which there was either 1) no macroalgal clump in the dish tub containing the composite sample material, or 2) no soft-bodied algal sample will be prepared. If there was no macroalgal clump but both soft bodied algae AND other sample types are to be prepared, follow all the instruction in this section with the exception of the final portion of Section 5.2.1 that is italicized and in orange font, and discusses how to process macroalgae when preparing only diatom and/or biomass samples.

If there is a macroalgal clump, but no soft-bodied algae sample is to be collected, follow the instructions in this section, including the final, italicized portion of Section 5.2.1, and skip Section 5.2.2, which deals with soft-bodied algal sample processing.

5.2.1 Measuring the Composite Liquid Volume

Once algal specimens have been removed from all the substrates (e.g., sand, gravel, cobble, wood, leaves) in the dish tub, according to the procedure described in Section 4, thoroughly agitate the liquid to get as much as possible of the microalgae into suspension, and then immediately pour the liquid into a CLEAN graduated cylinder to measure its volume. Try to leave all substrate material (e.g., silt, sand) behind. Transfer the measured liquid into a CLEAN 1L plastic bottle. Using a minimal amount of stream water, rinse the substrate once or twice with stream water until it appears that little to no additional suspended material (microalgae) is coming off. Add this rinsate to the graduated cylinder to measure it also. If necessary, repeat this process (regularly agitating the dish tub) until all the liquid has been measured and transferred to the sample bottle. Use water sparingly, because the total sample volume (plus rinsate) should be no more than about 400-500 mL.

Because you are leaving as much as possible of the silt, sand, and any large substrate material behind, the final volume should reflect only the liquid component of the sample plus rinse water. Record the total volume of all the liquid that had been in the dish tub, including any that was used for rinsing the substrates and sampling devices, on the field sheet under the Algae Samples section. This is the COMPOSITE VOLUME. This value will also be recorded on all algae sample labels (i.e., for the diatom and soft-bodied algae taxonomic ID samples, the chlorophyll *a*, and the AFDM).

Note: If no soft-bodied algae sample is to be prepared, but there is a macroalgal clump in the sample, separate the clump from the liquid portion of the sample, measure and record the composite volume of the liquid (as described above), then cut the macroalgal clump into very fine pieces (resulting in strands that are eyelash-length or shorter) with CLEAN scissors and add these pieces to the composite liquid. The pieces should be chopped small enough so that they practically "blend" into the liquid (i.e., distinct fragments of macroalgae are not easily discernible), because the goal is to "homogenize" the macroalgae into the liquid as much as possible. Shake vigorously to homogenize the macroalgal fragments into the liquid. Then proceed to Section 5.2.3 and beyond to prepare the diatom and/or biomass samples.

5.2.2 Preparing the Soft-Bodied Algae Taxonomic ID Sample

Pour freshly-agitated liquid composite sample into the soft-bodied algae sample tube to the 45 mL mark. Midway through pouring, the composite sample should be swirled some more (first clockwise, then counter-clockwise) to ensure that the microalgae are still fully suspended. Cap the tube tightly. Completely fill out a sample label and affix it to the sample tube. Cover the label completely with clear plastic tape to prevent the writing on the label from smearing. Place the tube in the wet ice chest to keep it in the dark and as cold as possible, but make sure it is never allowed to freeze.

Note: As soon as possible, and no longer than four days after collection of the sample, glutaraldehyde must be added to the tube (to a final concentration of 2.5%, by adding 5 mL of 25% glutaraldehyde to 45 mL of sample) and distributed throughout the sample by agitation and turning the tube upside down repeatedly. Glutaraldehyde is necessary for fixing soft-bodied algal samples in order to preserve fine morphological features and the color of pigments, as both can be crucial characters for taxonomic determination. Glutaraldehyde is a hazardous substance that poses a number of safety risks. As such, it should be handled in a fume hood by trained personnel wearing appropriate gear. Refer to Appendix D for an SOP for the use of glutaraldehyde.

Members of the field crew can either have the glutaraldehyde added to the samples back at their own lab, or arrange for the glutaraldehyde to be added to the samples by the taxonomy lab. In either case, the unfixed samples must be kept in the dark and on wet ice (but not allowed to freeze), and must be fixed within four days of collection (and preferably sooner). If the taxonomy lab will be adding the fixative, it is imperative to plan ahead to arrange for this to be done, and also to clearly mark which tubes will need to have fixative added to them. Once the samples are fixed, it is no longer imperative to store them on wet ice. Following fixation, they can simply be stored in a cool, dark place.

5.2.3 Preparing the Diatom Taxonomic ID Sample

Diatom samples should be fixed as soon as possible after collection to reduce the possibility of cell division post-sampling. A 10% solution of buffered formalin is used to fix diatoms, and instructions for preparing this solution are provided in Appendix C.

To prepare the diatom sample, aliquot 40 mL of freshly-agitated composite liquid into the diatom ID sample tube, swirling the composite sample bottle again midway through pouring to keep the microalgae suspended. Add 10 mL of the 10% buffered formalin to the sample. This can be done using a small syringe or bulb pipette. Alternatively, if preferred, the centrifuge tubes for the diatom samples can be pre-loaded with 10 mL of the 10% buffered formalin and 40 mL of sample can be added carefully to the fixative, to avoid having to dispense the fixative in the field.

Notes: Fixatives such as formalin must be used with great care. Be sure to wear formalin-safe gloves and safety goggles when using the fixative, as it should never be touched with bare hands or allowed to splash onto skin or into eyes. Also make sure it is used only in a very well-ventilated place and avoid breathing in any fumes. Minimize the amount of time that vessels containing formalin are open. Fixative added to the sample must not be allowed to ooze outside the vessel that contains it, including the sample tubes. Refer to Appendix E for an SOP for the use of formalin.

Cap the tube tightly and shake it to mix the formalin into the sample. Fill out a sample label and affix it to the sample tube. Cover the label completely with clear plastic tape to prevent the writing on the label from smearing. Keep the fixed diatom samples in the dark and away from extreme heat.

5.2.4 Preparing the Biomass Samples

The remaining composite sample liquid can be used to prepare the chlorophyll *a* and AFDM filters as described below.

Chlorophyll *a* samples:

The procedure to filter chlorophyll *a* samples should be carried out quickly, and in the shade as much as possible, to minimize exposure of the sample to light, and minimize chlorophyll *a* degradation thereby. For the chlorophyll *a* samples, use CLEAN filter forceps (rinsed with DI water three times) to center a glass fiber filter (47 mm, 0.7 µm pore size) onto the mesh platform of a CLEAN filtering tower apparatus (rinsed with DI water three times), and rinse the filter a little with DI water to seat it well into the mesh before attaching the filter reservoir on top. Never touch the filters with hands or anything other than clean forceps.

Agitate the composite sample to resuspend all the microalgal material. Carefully measure 25 mL using a small, CLEAN graduated cylinder (rinsed with DI water three times). Midway through pouring the 25 mL, swirl the composite sample again to ensure that the material is still fully suspended. Pour the remainder of the 25 mL, and then pour the measured sample into the filter reservoir. Once empty, rinse the graduated cylinder with a few mL of DI water, and add this to the reservoir.

To filter the sample, create a gentle vacuum with the hand pump. Be sure to proceed very slowly, and pump only one stroke at a time until all of the liquid in the sample is passed through the filter. Pressure on the sample should never exceed 7 psi, as this could cause cells to burst and release contents, including chlorophyll *a*, into the filtrate and be lost. If it becomes impossible to filter a whole 25 mL of the sample and

remove the water efficiently, discard the filter and try again with a smaller volume (e.g., 10 mL). It is not necessary to collect on multiple filters to try to achieve a total volume of 25 mL. Simply filter as much as possible on a single filter, up to 25 mL, and then use that filter as the sample. Be sure to record the volume of the composite sample that was actually filtered, both on the datasheet, and on the sample label.

Rinse the sides of the filter reservoir with a few mL of DI water, and continue filtering until the water is drawn down. The filter should not be sucked dry, but rather left slightly moist, in order to avoid applying excessive pressure to the sample, which could cause algal cells to burst. After the liquid has passed through, check the filter to see if there are any bits of non-algal plant matter (like tiny seedlings or bits of leaves). If so, remove them with clean forceps, being careful not to remove any algae in the process. If possible, rinse the removed items with DI water onto the filter before discarding them. Remove the filter from the filtering device. *Note: Always thoroughly rinse the sides of the filter reservoir and the interface between the mesh filter seating and the screw-on part of the reservoir with DI water between samples.*

Being careful not to remove any of the collected material from the filter, fold the filter in half (with the sample material on the inside) using the forceps, and place it inside a clean, snap-top Petri dish[13]. Envelop the Petri dish completely within a small sheet of aluminum foil in order to prevent any light from reaching the filter. Place the covered Petri dish and its corresponding, completely filled-out sample label (face outward) into a 100 mL Whirl-pak bag[14], purge as much of the air out of the bag as possible, "whirl" it shut, and seal it tightly with its wire tabs, so that water in the cooler will not be able to enter the bag. *Note 1: If the Whirl-pak bags contain a lot of air, they will float on top of the ice water in the cooler, and they then run the risk of not being kept cold enough. Shove the sample packet down into the ice in the cooler. Note 2: A clean, clear plastic centrifuge tube is also an acceptable container in which to store the filter. It must also be properly labeled, wrapped in aluminum foil, and kept submerged in wet ice.*

Keep chlorophyll *a* filters as cold as possible and place in the freezer or dry ice as soon as possible (and within four hours of collection); the holding time for the chlorophyll *a* filters is 28 days from collection, when kept frozen.

Ash-free dry mass (AFDM) samples:

For the AFDM samples, you should use glass-fiber filters (47 mm, 0.7 µm pore size) that have been precombusted. Never touch the filters with hands or anything other than a CLEAN forceps (rinsed with DI water three times). The filters to use should be labeled "for AFDM", and stored in aluminum sleeves. Follow the same process as that used for chlorophyll *a* sample filtering. After all the liquid has passed through, check the filter to see if there are any pieces of non-algal plant matter (such bits of leaves or wood). If so, remove them with a clean forceps (rinsed with DI water three times), being careful not to remove any algae in the process. The goal with AFDM, for the purposes of this SOP, is to target the ALGAL portion of the organic matter in the sample, and therefore field crews should do their best to remove non-algal contributors

13. It may be beneficial to write the Site Code or sample ID code on the Petri dish itself, in addition to filling out the full sample label.
14. Other bag types are acceptable only if they are water-tight (note that Ziploc bags often leak when submerged).

of organic matter from the sample. Remove the filter from the filtering device. *Note: Always thoroughly rinse the sides of the filter reservoir and the interface between the mesh filter seating and the screw-on part of the reservoir with DI water between samples.*

Use the forceps to fold the AFDM filter in half (with the sample material on the inside) and wrap it loosely in a small sleeve of clean aluminum foil. Be careful not to squeeze the filter, which could cause the sample to ooze from the filter onto the aluminum sleeve. Store the filter in a sealed 100 mL Whirl-pak bag containing a completely filled-out sample label, including the volume that was filtered (i.e., 25 mL or otherwise). As with the chlorophyll *a*, purge as much of the air out of each bag as possible, "whirl" it shut, and seal tightly with the wire tabs. Shove the sample packet down into the ice in the cooler. *Note: A clean, clear plastic centrifuge tube is also an acceptable container in which to store the filter. It must also be properly labeled and kept submerged in wet ice.*

Keep AFDM filters as cold as possible until the samples can be frozen back at the lab that evening, or place on dry ice until they can be stored in the lab freezer. The holding time for the AFDM samples is 28 days from collection, when kept frozen.

5.3 PROCESSING SOFT-BODIED AND OTHER SAMPLE TYPES WHEN A MACROALGAL CLUMP IS PRESENT

The following is a description of how to proceed when a soft-bodied algal taxonomic ID sample is to be prepared AND macroalgal clump(s) are present in the sample in the dish tub. A flowchart of this procedure is provided in Appendix F. It is recommended that this flowchart be printed out in color, laminated (if possible) or printed out on water-proof paper, and brought along to the field for quick reference on handling macroalgal clumps in the composite sample. The reason for the extra step in the processing of the macroalgae for the purposes of the soft-bodied algae sample is that it maintains larger, more intact macroalgal specimens for examination in the laboratory, rather than chopping up all of the macroalgal specimens before sending them to the lab. This is important, because availability of intact specimens greatly improves the chances that the taxonomist will be able to identify the soft-bodied algae to low taxonomic levels.

5.3.1 Isolating and Dividing the Macroalgal Clump

For this procedure, the macroalgal clump is first removed from the dish tub, wrung out gently, and rolled into a cylinder shape that is relatively even in thickness along its length. If there is more than one type of macroalgae in the sample, the various types should be layered on top of one another lengthwise so that they are represented in roughly constant proportions across the length of the "cylinder". The cylinder is measured with a ruler and a quarter of its length is cut off with scissors and put into the (still empty) soft-bodied algae ID centrifuge tube[15]. The clump is pushed down into the tube, and the top is flattened, so that the volume

15. It is unlikely that the ¼ macroalgal clump will occupy all the space in the sample tube, but if it does, a second tube will be needed in order to accommodate all the sample material plus liquid. If such an action is taken, it should be noted in the Comments section of the field sheets and the tubes should be clearly identified as belonging to the same sample, for record keeping purposes.

of the clump can be estimated using the graduations on the tube. The estimated volume of this clump will be used in a calculation (see Equation 1 and Figure 8). The remaining three-quarters length of cylinder is set aside in the shade/cool. It is recommended that this section be placed in a Ziploc bag, sealed, and put in the wet ice cooler.

5.3.2 Measuring the Composite Liquid Volume

Once algal specimens have been removed from all the substrates (e.g., sand, gravel, cobble, wood, leaves) in the dish tub, according to the procedure described in Section 4, gently agitate the dish tub to suspend the microalgae in the liquid, and then start pouring this suspension into a CLEAN graduated cylinder to measure the volume of the liquid. Try to leave all substrate material (e.g., silt, sand) behind. Transfer the measured liquid into a CLEAN 1L plastic bottle. Using a minimal amount of stream water, rinse the substrate once or twice with stream water until it appears that little to no additional suspended material (microalgae) is coming off. Add this rinsate to the graduated cylinder to measure it also. If necessary, repeat this process (regularly agitating the dish tub) until all the liquid has been measured and transferred to the sample bottle. Use water sparingly, because the total sample volume plus rinsate should be no more than about 400-500 mL.

Because you are leaving as much of the silt, sand, and any large substrate material behind as possible, the final volume should reflect only the liquid component of the sample plus rinse water. Record the total volume of all the liquid that had been in the dish tub, including any that was used for rinsing the substrates and sampling devices, on the field sheet under the Algae Samples section. This is the COMPOSITE VOLUME. This value will also be recorded on all algae sample labels (i.e., for the diatom and soft-bodied algae taxonomic ID samples, the chlorophyll *a*, and the AFDM).

5.3.3 Preparing the Soft-Bodied Algae Taxonomic ID Sample

Pour freshly-agitated liquid composite sample from the 1-L bottle into the soft-bodied algae sample tube (on top of the clump of macroalgae) up to the 45 mL mark. Midway through pouring, the composite sample should be swirled some more (first clockwise, then counter-clockwise) to ensure that the microalgae are still fully suspended. Cap the tube tightly. Completely fill out a sample label and affix it to the sample tube. Cover the label completely with clear plastic tape to prevent the writing on the label from smearing. Place the tube in the wet ice chest to keep it in the dark and as cold as possible, but make sure it is never allowed to freeze.

Note: As soon as possible, and no longer than four days after collection of the sample, glutaraldehyde must be added to the tube (to a final concentration of 2.5%, by adding 5 mL of 25% glutaraldehyde to 45 mL of sample) and distributed throughout the sample by agitation and turning the tube upside down repeatedly. Glutaraldehyde is necessary for soft-bodied algal samples in order to preserve fine morphological features and the color of pigments, as both can be crucial characters for taxonomic determination. Glutaraldehyde is a hazardous substance that poses a number of safety risks. As such, it should be handled in a fume hood by trained personnel wearing appropriate gear. Refer to Appendix D for an SOP for the use of glutaraldehyde.

Members of the field crew can either have the glutaraldehyde added to the samples back at their own lab, or arrange for the glutaraldehyde to be added to the samples by the taxonomy lab. In either case, the unfixed samples must be kept in the dark and on wet ice (but not allowed to freeze), and must be fixed within four days of collection (and preferably sooner). If the taxonomy lab will be adding the fixative, it is imperative to plan ahead to arrange for this to be done, and also to clearly mark which tubes will need to have fixative added to them. Once the samples are fixed, it is no longer imperative to store them on wet ice. Following fixation, they can simply be stored in a cool, dark place.

5.3.4 Preparing the Diatom Taxonomic ID Sample

After the soft-bodied algal sample has been prepared, and before preparing the diatom sample (and biomass samples, which will be discussed in the next section), the volume of the remaining composite liquid must be reduced to equal ¾ of the original volume[16]. This is necessary because ¼ of the macroalgae clump was taken out of the composite sample but a full ¼ was not removed from the water portion. As such, the original ratio between water and macroalgae must be restored before further sample preparation.

The following procedure is used to reduce the volume of liquid composite to ¾ of the original. For convenience, you can use this formula (Figure 8) to calculate how many mL to pour off and discard from the composite:

Equation 1. Adjusting the volume of composite sample

volume (mL) of composite to pour off = (0.25 * C) – 45 + A

where "C" is the original composite volume and "A" is the approximate volume of the clump of macroalgae that was placed in the soft-bodied algae sample tube (tamped down and flattened). You may wish to fill out a copy of the Ratio Restoration worksheet shown in Figure 8 to calculate the amount of composite to pour off.

Liquid Portion of Composite Sample: _____ mL = C

Volume of 1/4 macroalgal chunk: _____ mL = A

Volume of Liquid Composite to Pour Off: (0.25 * _____ C) – 45 + _____ A = _____ mL

Figure 8. Ratio restoration worksheet.

16. For example, if the original composite volume was 480mL, you will be discarding enough composite liquid to get down to 360 mL.

As always, be sure to agitate the composite liquid adequately in order to resuspend any settled microalgae before pouring off the calculated volume.

Once the required amount of composite liquid has been discarded, the remaining ¾ of the macroalgal clump ("cylinder") is cut into very fine pieces with a scissors (resulting in strands that are eyelash-length or shorter), and these are added to the reduced-volume composite liquid. The pieces should be chopped small enough so that they practically "blend" into the liquid (i.e., distinct fragments of macroalgae are not easily discernible), because the goal is to "homogenize" the macroalgae into the liquid as much as possible. Now the ratio of macroalgae to liquid from the original sample in the dish tub is restored. Cap the composite bottle and shake vigorously to homogenize the bits into the liquid as much as possible, while not agitating so hard as to risk busting cells and releasing chlorophyll.

Diatom samples should be fixed as soon as possible after collection to reduce the possibility of cell division post-sampling. A 10% solution of buffered formalin is used to fix diatoms, and instructions for preparing this solution are provided in Appendix C.

To prepare the diatom sample, aliquot 40 mL of freshly-agitated sample homogenate into the diatom ID sample tube, swirling the composite sample bottle again midway through pouring to keep the algal material suspended. Add 10mL of the 10% buffered formalin to the sample. This can be done using a small syringe or bulb pipette. Alternatively, if preferred, the centrifuge tubes for the diatom samples can be pre-loaded with 10 mL of the 10% buffered formalin and 40 mL of sample can be added carefully to the fixative, to avoid having to dispense the fixative in the field.

Note: Fixatives such as formalin must be used with great care. Be sure to wear formalin-safe gloves and safety goggles when using the fixative, as it should never be touched with bare hands or allowed to splash onto skin or into eyes. Also make sure it is used only in a very well-ventilated place and avoid breathing in any fumes. Minimize the amount of time that vessels containing formalin are open. Fixative added to the sample must not be allowed to ooze outside the vessel that contains it, including the sample tubes. Refer to Appendix E for an SOP for the use of formalin.

Cap the tube tightly and shake it to mix the formalin into the sample. Fill out a sample label and affix it to the sample tube. Cover the label completely with clear plastic tape to prevent the writing on the label from smearing. Keep the fixed diatom samples in the dark and away from extreme heat.

5.3.5 Preparing the Biomass Samples

The remaining composite sample homogenate can be used to prepare the chlorophyll *a* and AFDM filters according to the following procedure.

Chlorophyll *a* samples:

The procedure to filter chlorophyll *a* samples should be carried out quickly, and in the shade as much as possible, to minimize exposure of the sample to light, and minimize chlorophyll *a* degradation thereby. For the chlorophyll *a* samples, use CLEAN filter forceps (rinsed with DI water three times) to center a glass fiber filter (47 mm, 0.7 µm pore size) onto the mesh platform of CLEAN filtering tower apparatus (rinsed with DI water three times), and rinse the filter a little with DI water to seat it well into the mesh before attaching the filter reservoir on top. Never touch the filters with hands or anything other than clean forceps.

Agitate the composite sample homogenate to resuspend all the macroalgal fragments and microalgal material. Carefully measure 25 mL using a small, CLEAN graduated cylinder (rinsed with DI water three times). Midway through pouring the 25 mL, swirl the homogenate again to ensure that the material is still fully suspended. Pour the remainder of the 25 mL, and then pour the measured homogenate into the filter reservoir. Once empty, rinse the graduated cylinder with a few mL of DI water, and add this to the reservoir.

To filter the sample, create a gentle vacuum with the hand pump. Be sure to proceed very slowly, and pump only one stroke at a time until all of the liquid in the sample is passed through the filter. Pressure on the sample should never exceed 7 psi, as this could cause cells to burst and release contents, including chlorophyll *a*, into the filtrate and be lost. If it becomes impossible to filter a whole 25 mL of the sample and remove the water efficiently, discard the filter and try again with a smaller volume (e.g., 10 mL). It is not necessary to collect on multiple filters to try to achieve a total volume of 25 mL. Simply filter as much as possible on a single filter, up to 25 mL, and then use that filter as the sample. Be sure to record the volume of the composite sample that was actually filtered, both on the datasheet, and on the sample label.

Rinse the sides of the filter reservoir with a few mL of DI water, and continue filtering until the water is drawn down. The filter should not be sucked dry, but rather left slightly moist, in order to avoid applying excessive pressure to the sample, which could cause algal cells to burst. After all the liquid has passed through, check the filter to see if there are any bits of non-algal plant matter (like tiny seedlings or bits of leaves). If so, remove them with clean forceps, being careful not to remove any algae in the process. Remove the filter from the filtering device. *Note: Always thoroughly rinse the sides of the filter reservoir and the interface between the mesh filter seating and the screw-on part of the reservoir with DI water between samples.*

Fold the filter in half (with the sample material on the inside) using the forceps, and place it inside a clean, snap-top Petri dish. Envelope the Petri dish completely within a small sheet of aluminum foil in order to prevent any light from reaching the filter. Place the covered Petri dish and its corresponding, completely filled-out sample label (face outward) into a 100 mL Whirl-pak bag, purge as much of the air out of the bag as possible, "whirl" it shut, and seal it tightly with its wire tabs, so that water in the cooler will not be able to enter the bag. *Note: If the Whirl-pak bags contain a lot of air, they will float on top of the ice water in the cooler, and they then run the risk of not being kept cold enough. Shove the sample packet down into the ice in the cooler.*

Keep chlorophyll *a* filters as cold as possible and place in the freezer or dry ice as soon as possible (and within four hours of collection); the holding time for the chlorophyll *a* filters is 28 days from collection, when kept frozen.

Ash-free dry mass (AFDM) samples:

For the AFDM samples, you should use glass-fiber filters (47 mm, 0.7 µm pore size) that have been precombusted. Never touch the filters with hands or anything other than a clean forceps (rinsed with DI water three times). The filters to use should be labeled "for AFDM", and stored in aluminum sleeves. Follow the same process as that used for chlorophyll *a* sample filtering. After all the liquid has passed through, check the filter to see if there are any bits of non-algal plant matter (such as bits of leaves or wood). If so, remove them with clean forceps (rinsed with DI water three times), being careful not to remove any algae in the process. Remove the filter from the filtering device. *Note: Always thoroughly rinse the sides of the filter reservoir and the interface between the mesh filter seating and the screw-on part of the reservoir with DI water between samples.*

Use the forceps to fold the AFDM filter in half (with the sample material on the inside) and wrap it loosely in a small sleeve of clean aluminum foil. Be careful not to squeeze the filter, which could cause the sample to ooze from the filter onto the aluminum sleeve. Store the filter in a sealed 100 mL Whirl-pak bag containing a completely filled-out sample label, including the volume that was filtered (i.e., 25 mL or otherwise). As with the chlorophyll *a*, purge as much of the air out of each bag as possible, "whirl" it shut, and seal tightly with the wire tabs. Shove the sample packet down into the ice in the cooler.

Keep AFDM filters as cold as possible until the samples can be frozen back at the lab that evening, or place on dry ice until they can be stored in the lab freezer. The holding time for the AFDM samples is 28 days from collection, when kept frozen.

5.4 PROCEDURE FOR COLLECTING QUALITATIVE ALGAL SAMPLES

If your program calls for the collection of soft-bodied taxonomic ID samples, then you will also need to collect a "qualitative" sample at every monitoring reach. The qualitative samples consist of a composite of all types of soft-bodied algae visible within the reach. This is of value because it can provide a fairly exhaustive list of soft-bodied algal taxa present at the site and can also aid identification of taxa captured in the RWB sampling, since it allows larger, more intact specimens to be collected than those that may end up in the more heavily processed quantitative sample (described above). In addition, if the qualitative sample is kept cool and in the dark, and is delivered to the lab in a timely manner (i.e., as quickly as possible, and within two weeks of collection), the live specimens can be cultured, which can also aid in identification. For example, some taxa in the Zygnematales cannot be identified to species level unless they are in a sexual phase during examination. If asexual at the time of collection (which is the typical situation), live specimens could be induced to a sexual phase in the lab. Collection of a qualitative diatom sample is optional, and is typically not needed for general bioassessment purposes.

For qualitative soft-bodied algal samples, collect specimens of all obviously different types of macroalgal filaments and mats, as well as microalgae (in the forms of scrapings using a razor blade or knife), and depositional samples (suctioned from along the surface of sediments using a clean turkey baster). Note that some algae (e.g., species of *Chara*, *Paralemanea*, and *Vaucheria*) have morphologies similar to submerged macrophytes or mosses. In addition, algae are not always green, and may instead be dark-brownish, golden, reddish, or bluish-green. Some cyanobacteria (which should also be collected for the qualitative sample), such as members of the genus *Nostoc*, look like gelatinous globules, or sacs, ranging in size anywhere from smaller than a pea to larger than a lime. Online image searches of these taxa and others will help the collector develop an eye for the variety of types of algae that may be encountered in streams. If you suspect something may be algae, but are not sure, it is always preferable to collect some of it for the qualitative sample. The laboratory will determine whether it qualifies for inclusion in the species list. Collect from as many distinct locations as possible throughout the reach so as to capture as much of the apparent diversity in the reach as you can. Also, when possible, try to grab part of the holdfast structures that attached the macroalgae to the substrate, as these structures can be useful for taxonomic identification.

Since these samples are merely qualitative, and not quantitative, you need not worry about collecting them in a manner that is representative of their relative abundances within the reach. *Note: If there is only a small amount of macroalgae in the stream, it should be allocated preferentially to the soft-bodied algae laboratory sample, as opposed to the diatoms (if a diatom qualitative sample is being collected), because it is primarily needed for the soft-bodied algal identification work (although diatoms can live as epiphytes on macroalgae, so macroalgal samples are also of values for the diatom work).*

Using a thick, waterproof marker, label a Whirl-pak bag with the Site Code, Date, Sample ID, and "soft" (or "diatom", if also collecting a diatom sample). Fill the bag with a total volume of up to 100 mL of qualitative algae sample + water. Purge any extra air from the bag, seal with the wire tabs by twisting them together (not just folding them, as this can result in leakage), tuck the ends of the wire tabs inward so that they cannot poke other bags, and store in the cooler on wet ice in the field. Be careful not to place the bags right up against ice or frozen blue-ice bags, because this could cause the algae to freeze and thus destroy the sample. Unlike with the quantitative samples, **do not add glutaraldehyde or formalin** (or any other fixative) to these qualitative samples. Keep the qualitative samples on wet ice and refrigerate immediately upon return to the lab. Because they are not preserved, these samples should be examined by a taxonomist as soon as possible (and within two weeks, at most), as they can decompose fairly rapidly. Decomposition is of particular concern for the soft-bodied algae sample.

If it is impossible to get the soft-bodied qualitative samples to a taxonomist within two weeks of sample collection, then split the qualitative samples in half, transfer one half to a 50 mL centrifuge tube and preserve it with glutaraldehyde (to a 2.5% final concentration) and leave the other half un-fixed (but continue to store in the cold/dark until examination by a taxonomist). This should be done in order to preserve part of the sample for morphological identification, but still maintain some possibility of keeping some specimens alive, in case culturing is necessary. *Note: Glutaraldehyde is a hazardous substance that can*

pose health and safety risks. Add glutaraldehyde in a fume hood, wearing safety goggles and glutaraldehyde-safe gloves. Refer to Appendix D for more detailed instructions on the safe handling of glutaraldehyde.

5.5 ALGAL SAMPLING QUALITY ASSURANCE / QUALITY CONTROL

The SWAMP bioassessment group is currently developing guidelines for quality assurance and quality control for bioassessment procedures. Future revisions to this document will include more specific information covering personnel qualifications, training and field audit procedures, procedures for field calibration, procedures for chain of custody documentation, requirements for measurement precision, health and safety warnings, cautions (actions that would result in instrument damage or compromised samples), and interferences (consequences of not following the SOP).

It is recommended that duplicate sampling of algae occur at 10% of study sites. The recommended method for collecting duplicates is at adjacent positions along the sampling transect according to the scheme depicted in Figure 2. Both samples should be collected at each transect before moving on to the next transect. When duplicate samples are collected at a site, the full suite of information about the algae samples (composite volume, numbers of each sampling device used, amount filtered, etc.) will need to be recorded for each replicate. This information can be recorded on a duplicate copy of the "Algae Samples" field sheet. Alternatively, the data cells on this sheet can be divided in half to accommodate information for each replicate. If the latter, it is important to keep track of which values go with which replicate.

In addition to including composite volume, area sampled (total, for all sampling devices used), and amount filtered (for the biomass samples) on the sample labels and field sheets, this information should also be included on the chain-of-custody sheets that are submitted to the algae analytical and taxonomy laboratories. This will facilitate efficient calculation of several types of data output, because this information is needed both for the biomass results and for the soft-bodied algae biovolume results.

6 PHYSICAL HABITAT TRANSECT-BASED MEASUREMENTS TO ACCOMPANY ALGAL BIOASSESSMENT

Once all algae samples have been collected at a given transect, PHab data collection can begin there. PHab data are designed to assess the physical habitat conditions of the stream reach being sampled. Knowledge about the PHab parameters can aid interpretation of the biotic assemblage data collected. Data for the following PHab parameters will be entered on transect-specific datasheets (corresponding to each of the 11 main transects along the monitoring reach). These datasheets are provided on the SWAMP website.

It should be noted that the data collection procedures for the parameters below reflect those that are described in the SWAMP BMI Bioassessment SOP (Ode 2007). With respect to PHab assessment, the only deviation between this SOP and that of Ode (2007) is in terms of omission of certain parameters. However, where there is overlap in parameters between the two SOPs, they are assessed in exactly the same manner. The one exception to this is the addition, in this SOP, of percent algal cover determination to the pebble count as described in Ode (2007). Also, note that because the datasheets are multi-purpose datasheets, developed for both BMIs and algae, they include some PHab parameters that are not a part of this SOP. Specifically, the following PHab data that appear on the datasheets are not collected when only algae are being sampled: 1) Riparian vegetation, and 2) Instream habitat complexity. As such, these sections are not filled out on the datasheets when only algae samples are being collected.

6.1 WETTED WIDTH

The wetted channel is the zone that is inundated with water and the wetted width is the distance between the sides of the channel at the point where substrates are no longer surrounded by surface water. Measure the wetted stream width and record this in the box at the top of the Transect data form.

6.2 BANKFULL WIDTH

The bankfull channel is the zone of maximum water inundation in a normal flow year (one-to-two year flood events). Since most channel formation processes are believed to act when flows are within this zone, bankfull dimensions provide a valuable indication of relative size of the waterbody.

Scout along the stream margins to identify the location of the bankfull margins on either bank by looking for evidence of annual or semi-annual flood events. Examples of useful evidence include topographic, vegetative, or geologic cues (changes in bank slope, changes from annual to perennial vegetation, changes in the size distribution of surface sediments). While the position of drift material caught in vegetation may be

a helpful aid, this can lead to very misleading measurements. *Note: The exact nature of this evidence varies widely across a range of stream types and geomorphic characteristics. It is helpful to investigate the entire reach when attempting to interpret this evidence because the true bankfull margin may be obscured at various points along the reach. Often the bankfull position is easier to interpret from one bank than the other; in these cases, it is easiest to infer the opposite bank position by projecting across the channel. Additionally, height can be verified by measuring the height from both edges of the wetted channel to the bankfull height (these heights should be equal).*

Stretch a tape from bank to bank at the bankfull position. Measure the width of the bankfull channel from bank to bank at bankfull height and perpendicular to the direction of stream flow.

6.3 BANKFULL HEIGHT

Measure bankfull height (the vertical distance between the **water surface** and the height of the bank, Figure 9) and record in the boxes at the top of the Transect data form under "Bankfull Width" and "Bankfull Height".

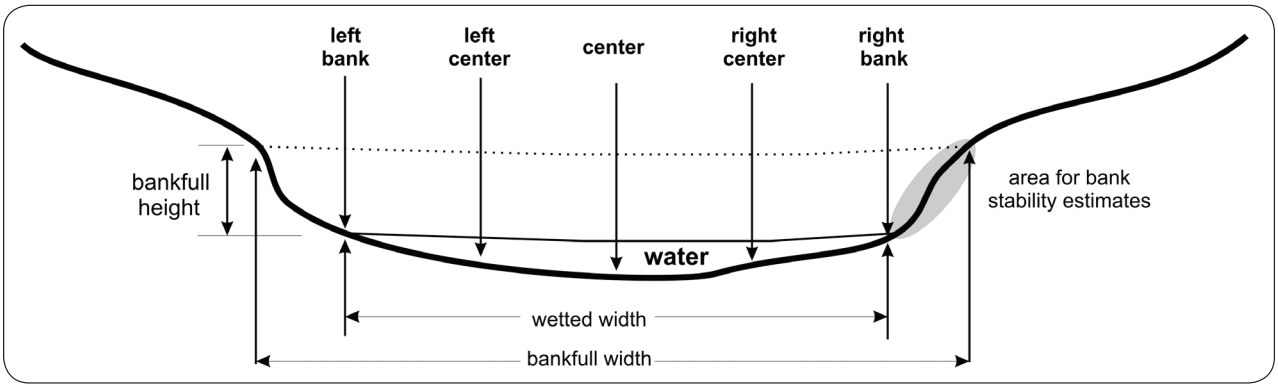

Figure 9. Cross sectional diagram of a typical stream channel showing locations of substrate measurements, wetted and bankfull width measurements, and bank stability visual estimates (reprinted from Ode 2007).

6.4 "PEBBLE COUNT": TRANSECT SUBSTRATES

Particle size frequency distributions often provide valuable information about instream habitat conditions that affect benthic communities. The Wolman pebble count technique is a widely used and cost-effective method for estimating the particle size distribution and produces data that correlate with costly, but more quantitative bulk sediment samples. Coarse particulate organic matter (CPOM, particles of organic material such as leaves that are greater than 1.0 mm in diameter) is a general indicator of the amount of

allochthonous organic matter available at a site, and its measurement can provide valuable information about the basis of the food web in a stream reach. The presence of CPOM associated with each particle is quantified at the same time that particles are measured for the pebble counts.

Transect substrate measurements are taken at five equidistant points along each transect (Figure 9). Divide the wetted stream width by four to get the distance between the five points (Left Bank, Left Center, Center, Right Center and Right Bank) and use a measuring device to locate the positions of these points (e.g., a stadia rod or measuring tape). Once the positions are identified, lower a folding meter stick through the water column perpendicular to both the flow and the transect to identify the particle located at the tip of the meter stick. *Note: It is important that you are not subjective about selecting a particle, as this will result in failing to generate an accurate assessment of the size class distribution of particles present in that stream reach.*

6.5 DEPTH

With the folding meter stick, measure the depth from the water surface to the top of the particle to the nearest cm and record on the datasheet.

Table 3
Particle size class codes, descriptions, and measurements (adapted from Ode 2007)

Size Class	Code Size Class Description	Common Size Reference	Size Class Range
RS	bedrock, smooth	larger than a car	> 4 m
RR	bedrock, rough	larger than a car	> 4 m
XB	boulder, large	meter stick to car	1 - 4 m
SB	boulder, small	basketball to meter stick	25 cm - 1.0 m
CB	cobble	tennis ball to basketball	64 - 250 mm
GC	gravel, coarse	marble to tennis ball	16 - 64 mm
GF	gravel, fine	ladybug to marble	2 – 16 mm
SA	sand	gritty to ladybug	0.06 – 2 mm
FN	fines	not gritty	< 0.06 mm
HP	hardpan (consolidated fines)		< 0.06 mm
WD	wood		
RC	concrete/ asphalt		
OT	other		

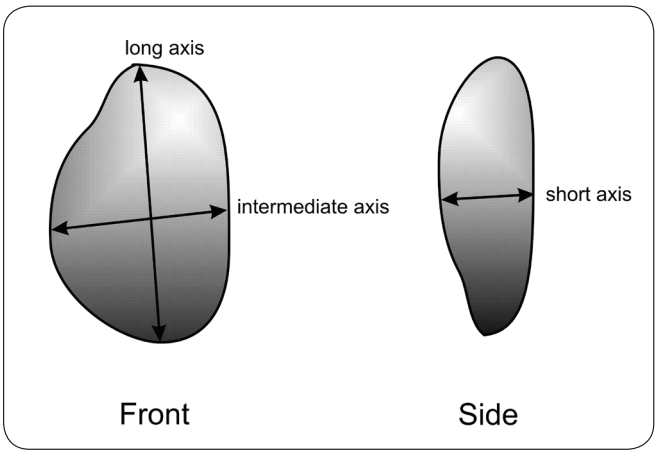

Figure 10. Diagram of three major perpendicular axes of substrate particles. The intermediate axis is recorded for pebble counts (reprinted from Ode 2007).

6.6 PARTICLE SIZE CLASS

Remove the particle from the streambed. Assign the particle to one of the size classes listed in Table 3 (these are also provided in a box on the transect form), based on its intermediate axis length (Figure 10). Record this information under Substrate size class.

Be sure to use measurements or the established codes for particle size class. If the latter, confirm the 2-letter codes for the particles as you call them out to your partner recording the data to ensure you are using the correct codes.

6.7 COBBLE EMBEDDEDNESS

It is generally agreed that the degree to which fine particles fill interstitial spaces has a significant impact on the ecology of benthic organisms and fish, but techniques for measuring this impact vary greatly. Here we define embeddedness as the volume of cobble-sized particles (64-250 mm) that is buried by fine and sand particles (< 2.0 mm diameter).

When a cobble-sized particle is encountered during the pebble count, visually estimate the percentage of the cobble's volume that has been buried by fine/sand particles (this will likely require removing the cobble from the streambed). Record, to the nearest 5%, the embeddedness of up to 25 cobble-sized particles within the sampling reach in the corresponding "% Cobble Embed" field for each cobble.

If 25 cobbles are not encountered during the pebble count, supplement the cobbles by conducting a "random walk" through the reach. Starting at a random point in the reach, follow a line from one bank to the other at a randomly chosen angle. Once at the other bank reverse the process with a new randomly chosen angle. Enter any of these additional embeddedness values at the bottom of the first page of the set of field forms, under "ADDITIONAL COBBLE EMBEDDEDNESS MEASURES".

If 25 cobble sized particles are not present in the entire reach, then record the values for however many cobbles are present.

6.8 CPOM

Record the presence or absence of Coarse Particulate Organic Matter (CPOM) that is > 1 mm diameter, and within 1 cm of the particle.

6.9 ALGAL COVER

Algal cover refers to the amount of algae in the stream reach, both in terms of 1) microalgal coatings ("slimy-ness") on stream substrates and 2) macroalgae (e.g., filaments, mats, globules). Algal cover is estimated by a point-intercept approach that entails collecting information about the presence/absence of both types of algae (as well as thickness, for the microalgae) at each of the points along the transects associated with the pebble count. If the imaginary point corresponding to each pebble in the pebble count intercepts algae, then algae is recorded as "present" at that point. The percentage of the points across the sampling reach that have algae present yields an estimate of the percent algal cover.

For each point along the pebble count, record information about algae as follows. For any film-like coating of algae (referred to as "Microalgae" on the datasheet) present on the surface of the substrate at that point, estimate the presence/thickness category according to the scheme in Table 4. For thicker microalgal layers, a small metal or plastic rod with demarcations at 1, 5, and 20 mm can be used for measurement. For layers too thin to measure, use the diagnostic criteria listed in the last column of Table 4. Note that these thickness codes refer only to microalgal coatings/films on substrate. They do not refer to thickness of macroalgal filaments/mats; macroalgal thickness is not assessed in this protocol. Be sure to collect microalgal thickness data from whatever substrate is topmost within the stream and therefore is most likely to be exposed to sunlight. Sometimes this substrate is not the actual pebble used in the pebble count, but rather a substrate type that occurs above the pebble, such as a thick mat of macroalgae that is above (and covering) the stream bottom. Microalgal species (which can include diatoms and unicellular soft-bodied algae) can grow as "epiphytes" upon macroalgal filaments and mats, coating them with a slimy, tinted film.

Table 4
Microalgal thickness codes and descriptions (adapted from Stevenson and Rollins 2006).

Code	Thickness	Diagnostics
0	No microalgae present	The surface of the substrate feels rough, not slimy.
1	Present, but not visible	The surface of the substrate feels slimy, but the microalgal layer is too thin to be visible.
2	<1mm	Rubbing fingers on the substrate surface produces a brownish tint on them, and scraping the substrate leaves a visible trail, but the microalgal layer is too thin to measure.
3	1-5mm	
4	5-20mm	
5	> 20mm	
UD	Cannot determine if a microalgal layer is present	
D	Dry point	

Note: Sometimes, due to the nature of the substrate, it can be difficult to discern whether a microalgal layer is present (particularly if it is very thin). For example, in the case of very fine sediments, the dark color of the silt can obscure the diagnostic color of a microalgal layer, and the inherent "sliminess" of very fine silt may make tactile determination of microalgae impossible. Therefore, when silt is the substrate, only relatively thick layers of microalgae might be easily discernible. If presence/absence of a microalgal layer cannot be determined with certainty, score microalgal thickness as "UD".

In addition to recording the presence and thickness of microalgae on the surfaces of substrates, record the presence/absence of attached macroalgae in the water column, as well as unattached, floating macroalgal mats on the water's surface, corresponding to each pebble count sampling point. Do this by envisioning an imaginary "line" extending from the water's surface down to the stream bottom where the target "pebble" lies (particularly in turbulent water, it may be helpful to use a viewing bucket (Appendix C) in order to see below the water's surface; the use of the viewing bucket is optional). If this line intercepts macroalgae, either floating on the water's surface, or somewhere within the water column, the appropriate algal class(es) should be recorded as "present". Attached macroalgal filaments have an obvious physical connection to something (like a cobble, boulder, or a gravel bed) lying on the bottom of the stream, whereas for unattached macroalgae, there is no obvious physical connection with the streambed, and the algae is just freely floating at or near the water's surface. For each class of macroalgae (Attached and Unattached), mark "P" (for "present") if intercepted by the sampling point and "A" (for "absent") if not intercepted.

Bear in mind that, because pebble counts span the "wetted width" of each transect, the expectation is that even the pebbles at the bank positions will generally be at least moist, and sometimes even submerged. As such, it is important to realize that algal cover can occur at the bank positions of the pebble count as well as intermediate positions across the stream. An exception to this is when the pebble surface is completely dry. Section 6.11 provides instructions for data collection in this situation.

6.10 MACROPHYTES

If a vascular plant (i.e., a macrophyte) is intercepted by the imaginary line associated with the pebble count point, mark "P" for "present" under Macrophytes. Otherwise, mark "A" for absent. Include only herbaceous plants that are rooted underwater. Examples of macrophytes include cattails, tules, rushes, sedges, monkeyflowers, speedwells, knotweeds, and watercress.

6.11 DRY SUBSTRATES

To determine how to collect data at dry sampling points, it is necessary to first establish whether the dry area in question lies within the stream's active channel (i.e., therefore regularly inundated during storms), or whether the point is on a stable island (i.e., therefore rarely, if ever, inundated). Stable islands are typically

vegetated, often with woody shrubs or trees, and have heights near or exceeding bankfull height. Pebble counts should not be conducted on stable islands. If the transect spans a portion of the study reach in which the channel is bifurcated such that there are two channels with an intervening island, the entire transect should be placed across the dominant channel, and all five pebble count points should be located on that side.

If the point falls on a dry surface that is within the usual active channel (i.e., subject to regular disturbance by flows), then pebble count/algal cover data from the dry point should be recorded as follows:

- score Depth as 0
- score particle Size Class and Embeddedness as described above for wet particles
- score all the algae variables (Microalgae, Macroalgae Attached, and Macroalgae Unattached) as "D" for "dry"
- leave CPOM and Macrophytes "blank" (i.e., do not circle anything). These parameters will register as NR (Not Recorded) in the database.

Ordinarily, the sampling transect would span the wetted width of the channel, but when no water is present at a given transect, evidence of the typical wetted extent of the active channel will need to be used to infer appropriate transect boundaries. Such indicators can include the transition from vegetated to unvegetated area (i.e., moving from banks to active channel), as well as the presence of dried algae, water stains, microtopographic transitions, changes in substrate composition, and others.

6.12 BANK STABILITY

The vulnerability of stream banks to erosion is often of interest in bioassessments because of its direct relationship with sedimentation.

For each transect, record a visual assessment of bank vulnerability in the region between the wetted width and bankfull width of the stream margins and between the upstream and downstream inter-transects. Choose one of three vulnerability states: eroded (evidence of mass wasting), vulnerable (obvious signs of bank erosion or unprotected banks), or stable.

6.13 HUMAN INFLUENCE

For the left and right banks, estimate a 10 x 10 m riparian area centered on the edges of the transect (see Figure 11). In the "Human Influence" section of the Transect data sheet, record the presence of 14 human influence categories in three spatial zones relative to this 10 x 10 m square (between the wetted edge and bankfull margin, between the bankfull margin and 10 m from the stream, and between 10 m and 50 m beyond the stream margins): 1) walls/rip-rap/dams, 2) buildings, 3) pavement/cleared lots, 4) roads/

railroads, 5) pipes (inlets or outlets), 6) landfills or trash, 7) parks or lawns (e.g., golf courses), 8) row crops, 9) pasture/ rangelands, 10) logging/ timber harvest activities, 11) mining activities, 12) vegetative management (herbicides, brush removal, mowing), 13) bridges/ abutments, 14) orchards or vineyards. Circle all combinations of impacts and locations that apply, but be careful to not double-count any human influence observations.

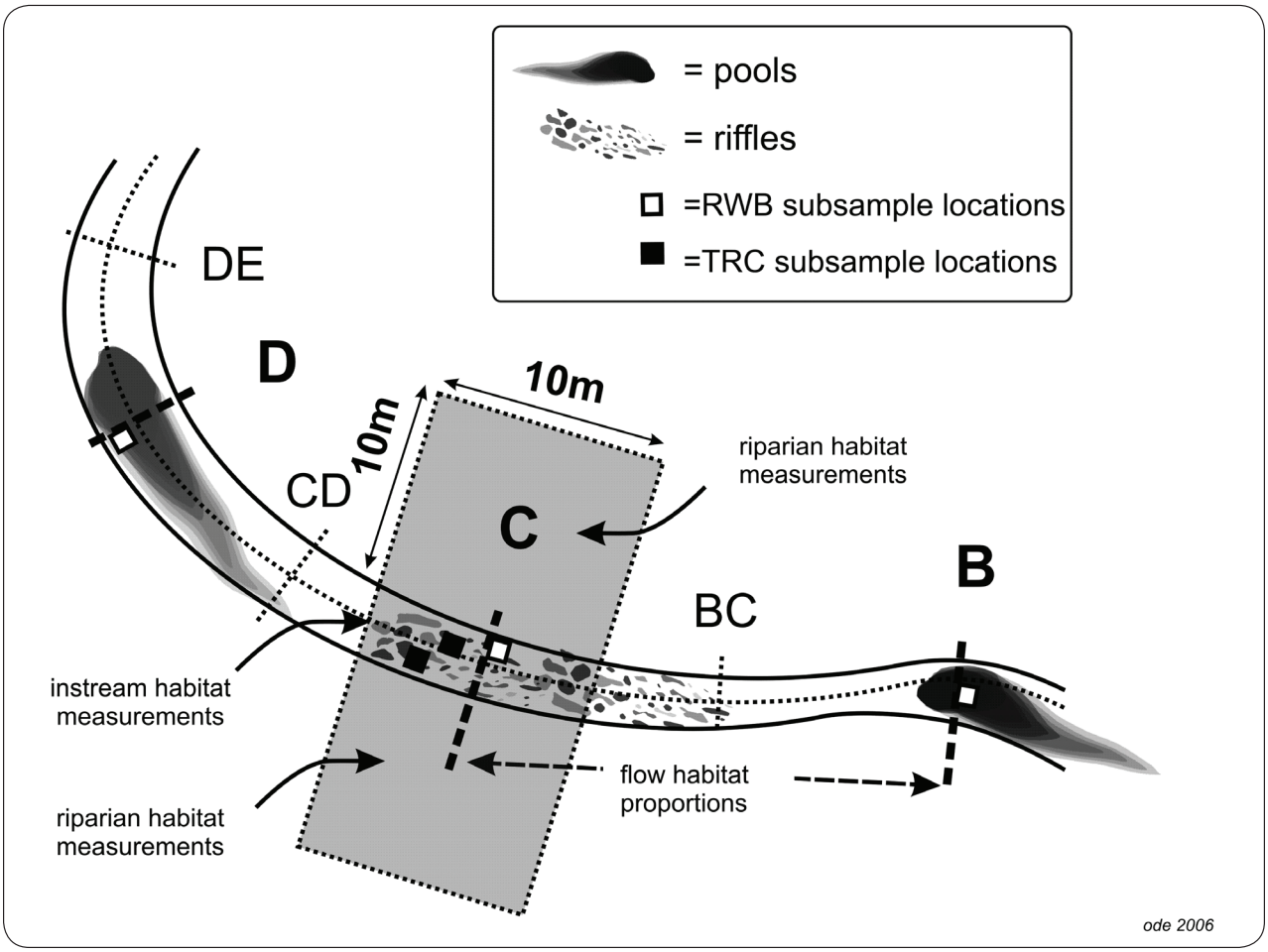

Figure 11. Section of the standard reach expanded from Figure 1 showing the appropriate positions for collecting algae samples (the white square, labeled "RWB" in the legend box) and flow habitat proportion measurements (reprinted from Ode 2007).

Record the presence of any of the 14 human influence categories in the stream channel within a zone 5 m upstream and 5 m downstream of the transect.

6.14 DENSIOMETER READINGS (CANOPY COVER)

The densiometer is read by counting the number of line intersections that are obscured by overhanging vegetation. Before using, the densiometer should be modified by taping off the lower left and right portions of the mirror in order to emphasize overhead vegetation over foreground vegetation (the main source of bias in canopy density measurements; see Figure 12.)

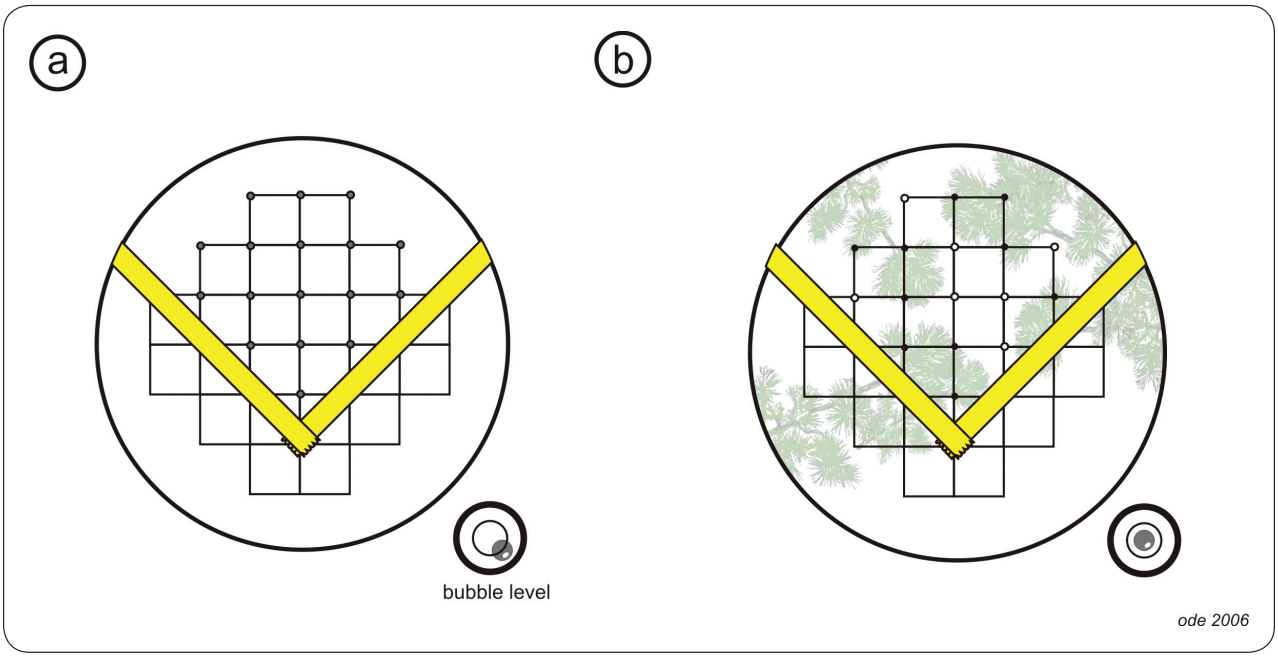

Figure 12. Representation of the mirrored surface of a convex spherical densiometer showing the position for taping the mirror and the intersection points used for the densiometer reading. The score for the hypothetical condition in (b) is 10 covered intersection points out of 17 possible. Note the position of the bubble level in (b) when the densiometer is leveled (reprinted from Ode 2007).

All densiometer readings should be taken with the bubble leveled, and 0.3 m (1 ft) above the water surface. The densiometer should be held just far enough from the squatting observer's body so that his/her forehead is just barely obscured by the intersection of the two pieces of tape.

Take and record four 17-point readings from the center of each transect: a) facing upstream, b) facing downstream, c) facing the left bank, d) facing the right bank. Optional readings can also be taken at the left and right banks (facing away from the stream, for these positions).

7 PHYSICAL HABITAT INTER-TRANSECT-BASED MEASUREMENTS

While most measures are taken at or relative to the main transects, a few measures are recorded at transects located at the midpoint between main transects. These are called "Inter-transects". The following measurements are taken relative to the Inter-transects: 1) Wetted Width, 2) Flow Habitats, and 3) "Pebble Count": Transect Substrates (including algal cover, as for the main transects).

7.1 INTER-TRANSECT WETTED WIDTH

Measure the same way that Transect wetted width was measured.

7.2 INTER-TRANSECT SUBSTRATES AND PERCENT ALGAL COVER

Collect these data the same way that Transect substrates and percent algal cover data were collected.

7.3 FLOW HABITATS

Because many benthic organisms prefer specific flow and substrate microhabitats, the proportional representation of these habitats in a reach is often of interest in bioassessments. There are many different ways to quantify the proportions of different flow habitats. This procedure produces a semi-quantitative measure consisting of 10 transect-based visual estimates.

At each Inter-transect, identify the percentage of six different habitat types in the region between the upstream Transect and downstream Transect: 1) cascades, 2) falls, 3) rapids, 4) riffles, 5) runs, 6) glides, 7) pools, and 8) dry areas. Record percentages to the nearest 5% — the total percentage of surface area for each section must equal 100%.

A description of each of these flow habitat types is provided below:

- cascades: short, high-gradient drops in stream bed elevation often accompanied by boulders and considerable turbulence
- falls: high-gradient drops in elevation of the stream bed associated with an abrupt change in the bedrock
- rapids: sections of stream with swiftly flowing water and considerable surface turbulence (rapids tend to have larger substrate sizes than riffles)
- riffles: "shallow/fast"; riffles are shallow sections where the water flows over coarse stream bed particles that create mild to moderate surface turbulence (< 0.5 m deep, > 0.3 m/s)
- runs: "deep/fast"; long, relatively straight, low-gradient sections without flow obstructions. The stream bed is typically even and the water flows faster than it does in a pool (> 0.5 m deep, > 0.3 m/s)
- glides: "shallow/slow"; sections of stream with little or no turbulence, but faster velocity than pools (< 0.5 m deep, < 0.3 m/s)
- pools: "deep/slow"; a reach of stream that is characterized by deep, low-velocity water and a smooth surface (> 0.5 m deep, < 0.3 m/s)
- dry: any surface area within the channel's wetted width that is above water

After you have collected all the above Transect-, and Inter-transect-, based measurements, collect data on Gradient. Also, if you have not already done so, take photographs at specific Transects, as indicated below. After you have collected Gradient data at each Transect, and have taken photographs where indicated, remove the corresponding flag from the stream bank.

7.4 PHOTOGRAPHS

Take a minimum of four (4) photographs of the reach at the following locations: a) Transect A facing upstream, b) Transect F facing upstream, c) Transect F facing downstream, and d) Transect K facing downstream. It is also desirable to take a photograph at Transect A facing downstream and Transect K facing upstream to document conditions immediately adjacent to the reach. Digital photographs should be used. Record the image numbers on the front page of the field form under "Photographs". *Note: An easy way to keep track of which site each series of photographs belongs to is to take a close-up of the front data sheet (containing legible site code and date) for that site prior to taking the series of photos.*

SECTION 8
REACHWIDE MEASUREMENTS

This last section describes PHab measurements of attributes specific to the stream reach as a whole. These include gradient of the reach and stream discharge.

8.1 GRADIENT

The gradient of a stream reach 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. The data collected for gradient are recorded on the "Slope and Bearing" form.

Note: An autolevel should be used for reaches with a percent slope of less than or equal to 1%. Either a clinometer or an autolevel may be used for reaches with a percent slope of greater than 1%, and sometimes a clinometer is preferable in really steep areas that are also heavily vegetated. The following description is for clinometer-based slope measurements. In reaches that are close to 1%, you will not know whether you are above or below the 1% slope cutoff. In these cases, default to use of an autolevel, which is described further below.

Clinometer method:

Transect to transect measurements taken with a clinometer are used to calculate the average slope through a reach. This measurement works best with two people, one taking the readings at the upstream transect ("backsighting") and the other holding a stadia rod at the downstream transect. If you cannot see the mid point of the next transect from the starting point, use the supplemental sections (indicating the proportion of the total length represented by each section). Otherwise, leave these blank.

Beginning with the upper transect (Transect K), one person (the measurer) should stand at the water margin with a clinometer held at eye level. A second person should stand at the margin of the next downstream transect (Transect J) with a stadia rod flagged at the eye level of the person taking the clinometer readings. Be sure you mark your eye level while standing on level ground! Adjust for water depth by measuring from the same height above the water surface at both transects. This is most easily accomplished by holding the base of the stadia rod at water level. *Note: An alternative technique is to use two stadia rods pre-flagged at the eye-height of the person taking the readings.*

Use a clinometer to measure the percent slope of the water surface (not the streambed) between the upstream transect and the downstream transect by sighting to the flagged position on the stadia rod. The clinometer reads both percent slope and degree of the slope. Be careful to read and record percent slope

rather than degrees slope (the measurements differ by a factor of ~2.2). Percent slope is the scale on the right hand side as you look through most clinometers. *Note: If an autolevel or hand level is used, record the elevation difference (rise) between transects and the segment length (run) instead of the percent slope (autolevel instructions are provided in the following section).*

If the stream reach geometry makes it difficult to sight a line between transects, divide the distance into two sections and record the slope and the proportion of the total segment length between transects for each of these sections in the appropriate boxes on the slope form (supplemental segments). Do not measure slope across dry land (e.g., across a meander bend).

Proceed downstream to the next transect pair (I-J) and continue to record slope between each pair of transects until measurements have been recorded for all transects. If you have finished all the other transect and inter-transect based measurements for PHab, you may remove the transect flags as you go.

Autolevel method (preferred):

To measure gradient using an autolevel, identify a good spot to set up the autolevel, preferably somewhere around the center of the reach (if there is good visibility from this location to both the upstream and downstream ends of the reach.) Set up the autolevel on very stable, and preferably fairly flat, ground. Set the height of the autolevel to comfortable eye level for the operator. Level the plane of view of the autolevel by balancing it using the bubble. Start by adjusting the legs, and then fine-tune the adjustment using the knobs. Once balanced, begin "shooting" the change in the height of the water level of the stream from transect to transect. Try to start with one of the outer transects (like A). Have a field partner at Transect A hold the Stadia rod at water's edge and perpendicular to the ground. Viewing through the autolevel (and focusing as necessary), look at the Stadia rod and note to the smallest demarcation on the stadia rod the height at which the autolevel line of view (i.e., the middle line in the viewfinder) hits. Record this information, and then have the Stadia rod holder proceed to the next transect (e.g., Transect B), again holding the base of the Stadia rod at water's edge. Very carefully, rotate the head of the autolevel so that it points to the new Stadia rod location. *Note: Take care not to bump the autolevel out of its position, because if this happens, you will not be able to take a height measurement of Transect B's water surface relative to that of Transect A, to determine the slope between the two transects.*

If the autolevel is bumped out of position before all the measurements are done, or if there is a point along the reach at which there is no longer a clear line of sight from the autolevel to the Stadia rod positioned at the transect, at water's edge, a new location must be set up for the autolevel. In order to maintain a relationship between water heights of the various transects already measured, it will be necessary to "re-shoot" the height of the water at the last transect for which a valid measurement was attained. From there, assuming there is no more disturbance to the position of the autolevel, you can continue cycling through the remaining transects from the new position. On the Slope and Bearing Form corresponding to autolevel use, indicate when the autolevel's position has been changed. If it is necessary to move the autolevel at some point, the transect that was measured from the original and the new position will be listed twice on

the datasheet: once for the original position, and once for the new. Also indicate the distance between main transects (i.e., 15 m, 25 m or other). These pieces of information will later be used to determine the slopes between transects and for the reach as a whole.

8.2 STREAM DISCHARGE

Stream discharge is the volume of water that moves past a point in a given amount of time and is generally reported as cubic feet per second (cfs) or cubic meters per second (cms). Because discharge is directly related to water volume, discharge affects the concentration of nutrients, fine sediments and pollutants; and discharge measurements are critical for understanding impacts of disturbances such as impoundments, water withdrawals and water augmentation. Discharge is also closely related to many habitat characteristics including temperature regimes, physical habitat diversity, and habitat connectivity. As a direct result of these relationships, stream discharge is often also a strong predictor of biotic community composition. Since stream volume can vary significantly on many different temporal scales (diurnal, seasonal, inter-annually), it can also be very useful for understanding variation in stream condition.

It is preferable to take discharge measurements in sections where flow velocities are greater than 0.5 ft/s and most depths are greater than 15 cm, but slower velocities and shallower depths can be used. If flow volume is sufficient for a transect-based "velocity-area" discharge calculation, this is by far the preferred method. If flow volume is too low to permit this procedure or if your flow meter fails, use the "neutrally buoyant object/ timed flow" method.

8.2.1 Discharge: Velocity Area Method

The layout for discharge measurements under the velocity-area (VA) method is illustrated in Figure 13. Flow velocity should be measured with either a Swoffer Instruments propeller-type flow meter or a Marsh-McBirney inductive probe flow meter.

Select the best location in the reach for measuring discharge. To maximize the repeatability of the discharge measurement, choose a transect with the most uniform flow (select hydraulically smooth flow whenever possible) and simplest cross-sectional geometry. It is acceptable to move substrates or other obstacles to create a more uniform cross-section before beginning the discharge measurements.

Data for this parameter will be entered in the "Discharge Measurements" section of the datasheet with the basic site information at the top ("Reach Documentation"). Measure the wetted width of the discharge transect and divide this into 10 to 20 equal segments. The use of more segments gives a better discharge calculation, but is impractical in small channels. A minimum of 10 intervals should be used when stream width permits, but interval width should not be less than 15 cm.

At each interval, record the distance from the bank to the end of the interval. Using the top-setting rod that comes with the flow velocity meter, measure the median depth of the interval. Standing downstream of the

transect to avoid interfering with the flow, use the top-setting rod to set the probe of the flow meter at the midpoint of the interval, at 0.6 of the interval depth (this position generally approximates average velocity in the water column), and at right angles to the transect (facing upstream). See Figure 13 for positioning detail.

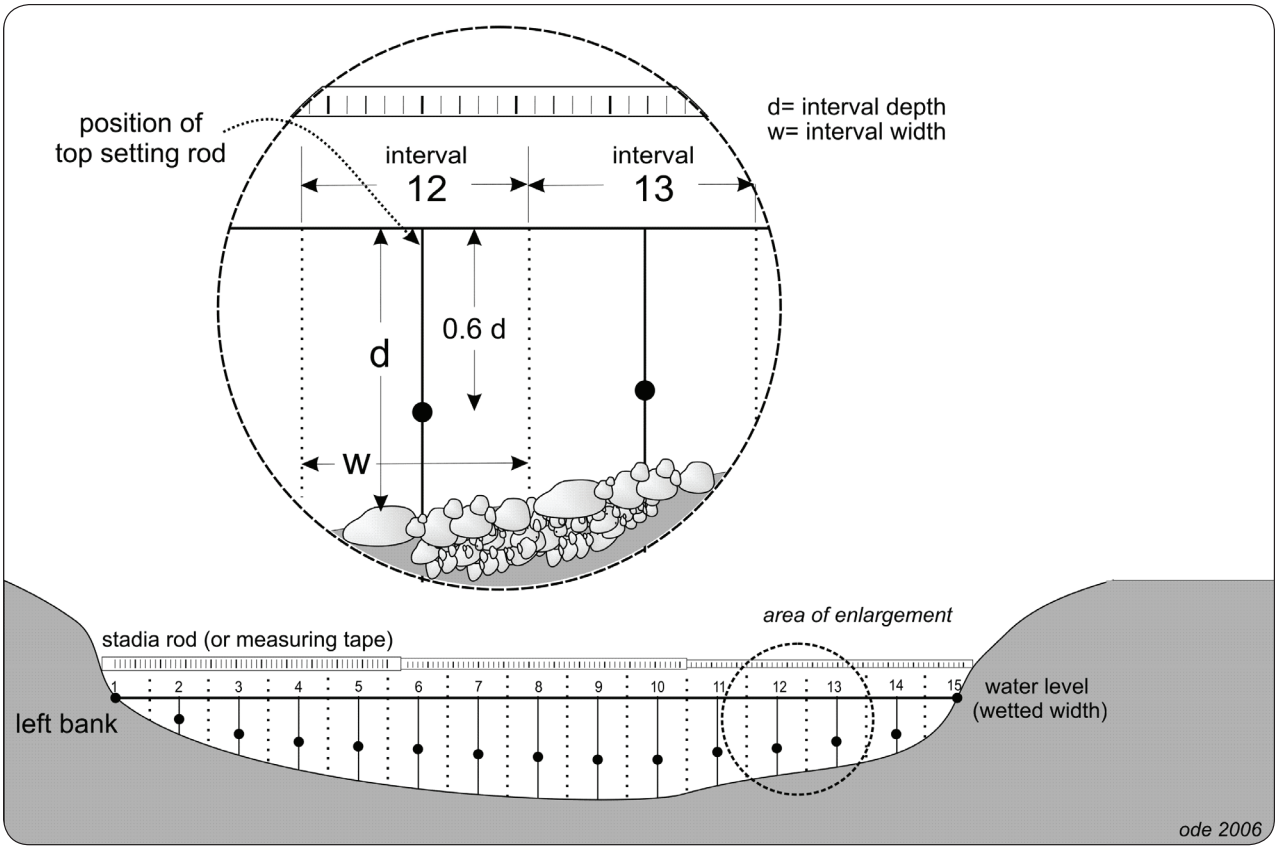

Figure 13. Diagram of layout for discharge measurements under the velocity-area method showing proper positions for velocity probe (black dots; reprinted from Ode 2007).

Allow the flow velocity meter to equilibrate for 10-20 seconds, then record velocity to the nearest ft/s. If the option is available, use the flow averaging setting on the flow meter. *Note 1: Under very low flow conditions, flow velocity meters may register readings of zero even when there is noticeable flow. In these situations, record a velocity of 0.5x the minimum flow detection capabilities of the instrument. Complete these steps on each of the intervals across the stream. Note 2: The first and last intervals usually have depths and velocities of zero.*

8.2.2 Discharge: Neutrally Buoyant Object Method

If streams are too shallow to use a flow velocity meter, the neutrally buoyant object (NBO) method should be used to measure flow velocity. However, since this method is less precise than the flow velocity meter

it should only be used if absolutely necessary. A neutrally buoyant object (one whose density allows it to just balance between sinking and floating) will act as if it were nearly weightless, thus its movement will approximate that of the water it floats in better than a light object. A piece of orange peel works well. To estimate the flow velocity through a reach, three transects are used to measure the cross-sectional areas within the test section sub-reach and three flow velocity estimates are used to measure average velocity through the test reach. To improve precision in velocity measurements, the reach segment should be long enough for the float time to last at least 10-15 seconds.

The position of the discharge sub-reach is not as critical as it is for the velocity-area method, but the same criteria for selection of a discharge reach apply to the neutrally buoyant object method. Identify a section that has relatively uniform flow and a uniform cross sectional shape.

The cross sectional area is estimated in a manner that is similar to, but less precise than, that used in the velocity area method. Measure the cross sectional area in one to three places in the section designated for the discharge measurement (three evenly-spaced cross sections are preferred, but one may be used if the cross section through the reach is very uniform). Record the width once for each cross section and measure depth at five equally-spaced positions along each transect.

Record the length of the discharge reach.

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GLOSSARY OF TERMS USED IN SOP G

Terms & Definitions	
TERM	**DEFINITION**
Aliquot	a measured portion of a sample, or subsample, or to measure a portion of a sample or subsample
Ash-free dry mass (AFDM)	the portion, by mass, of a dried sample that is represented by organic matter; the concentration of AFDM per stream surface area sampled can be used as a surrogate for algal biomass
Benthic algae	algae that are anchored to, or have at one point been anchored to, the stream bottom, in contrast to planktonic algae which are free-floating in the water column
Biofilm	a matrix/film adhering to stream substrata and consisting of microorganisms (e.g., algae, fungi, bacteria, protozoans) and detritus
Chlorophyll *a*	primary light receptor/photosynthetic pigment in algae and higher plants; the concentration of this pigment per stream surface area sampled provides an estimate of algal biomass
Composite sample	volume of all the liquid material amassed during sampling, including water used for rinsing substrate and sampling devices. Final composite volume should not exceed 400-500 ml.
Cyanobacteria	historically referred to as "blue-green" algae, but actually bacteria that are capable of photosynthesis and co-occur with true benthic algae in streams; useful as a bioindicator, and field-sampled and laboratory-processed concurrently with soft-bodied algae
Depositional	habitats in the stream that are dominated by slow-moving water, such as pools, where deposition of materials from the water column is more likely to occur than erosion (or (re)suspension) of loose bed materials; examples of "depositional" substrates include silt and sand
Diatom	a unicellular alga that possesses a rigid, silicified (silica-based) cell wall in the form of a "pill box"
Erosional	habitats in the stream that are dominated by fast-moving water, such as riffles, where stream power is more likely to facilitate erosion (suspension) of loose benthic material than deposition; examples of "erosional" substrates include cobbles and boulders
Homogenate	mixture of liquid composite sample and finely chopped fragments of macroalgae
Index of Biotic Integrity (IBI)	a quantitative assessment tool that uses information about the composition of one or more assemblages of organisms to make inferences about condition the environment they occupy (e.g., the assemblage of interest could be diatoms or benthic macroinvertebrates living in a stream)
Macroalgae	soft bodied algae that form macroscopically discernible filaments, mats, or globose structures
Microalgae	diatoms and microscopic soft-bodied algae, including unicellular forms; can co-occur with other microorganisms in a biofilm
Reachwide benthos (RWB)	method for biotic assemblage sample collection that does not target a specific substrate type, but rather objectively selects sampling locations across the reach, allowing for any of a number of substrate types to be represented in the resulting composite sample
Soft-bodied algae	non-diatom algal taxa; for the purposes of this SOP, cyanobacteria are subsumed under this assemblage
Wetted width	the distance between the sides of the channel at the point where substrates are no longer surrounded by surface water

List of Supplies for Stream Algae Sampling and Associated Data Collection A

Table 5.
General Supplies and Ambient Water Chemistry Collection

Item	Quantity / Site	Specifications
Sampling SOP (this document)	1/person	
Equipment decontamination supplies		See Appendix B
Hip or chest waders, or wading boots/shoes (not felt-soled)	at least 1 pair/ person	
Digital camera	1	
Full set of datasheets printed on waterproof paper (e.g., Rite-in-the-Rain™)	1 full set (and spare set recommended)	
Fine-tipped and thick-tipped waterproof/alcohol-proof pens and markers; pencils	2-3 each	
Clipboard	2-3	
Site dossier containing site maps, aerials, etc.	1	Add a 150 m scale line to aerials adjacent to stream
Thomas Guide and regional maps	as needed	
Centigrade thermometer	1	
pH meter	1	
DO meter and spare membrane	1	
Conductivity meter	1	
Turbidimeter and vial(s) (optional)	1	
Field alkalinity meter or test kit (e.g., Hach)	1	
Water chemistry containers	as needed	
Calibration standards	1	
Spare batteries for meters	as needed	
First aid kit	1	

Table 6.
Algal Taxonomic and Biomass Sample Collection

Needed for[1]:	Item	Quantity / Site	Specifications
D, S, C, A	White dish tub, rectangular, plastic, 11.5 qt	1	Use white, not colored
D, S, C, A	Scrubbing brush or scouring pad to clean dish tub, etc.	1	
D, S, C, A	Composite sample receiving bottle with cap, 1 L, plastic	1	Fisher 02-912-038
D, S, C, A	Graduated cylinder, 500 mL and 25 mL, plastic	1 each	Fisher 03-007-42 & 03-007-39
D, S, C, A	Bottle brush to clean graduated cylinders, etc.	1 sm, 1 lg	
D, S, C, A	PVC delimiter, 12.6 cm^2 area	1	See Appendix C
D, S, C, A	Spatula (> 12.6 cm^2 surface area)	1	
D, S, C, A	Rubber delimiter, 12.6 cm^2 area	1	See Appendix C
D, S, C, A	Toothbrush, firm-bristled	1	
D, S, C, A	Syringe scrubber, 60 mL syringe, 5.3 cm^2 area	1	See Appendix C
D, S, C, A	White (non-pigmented) scrubbing-pad circles	11 per replicate	See Appendix C
D, S, C, A	Tally meter (optional)	1	Ben Meadows 9JB-102385
D, S, C, A	Scissors	1	
D, S, C, A	Wash bottles	2	Label bottles with "stream water", and "DI water"
D, S, C, A	Razor blades or Swiss army knife	1	
D, S, C, A	Sample labels (printed on waterproof paper)	4 per replicate	See Figure 6
D, S, C, A	Clear plastic tape, 5 cm wide	Length of ~20cm per replicate	
D, S, C, A	Ice chest with wet ice	1 (2 preferred if multiple sites to be sampled)	
D, S, C, A	Fisherman's vest (optional)	1	
D, S	Centrifuge tubes, 50 mL, plastic	2 per replicate	Cole Parmer 06344-27
D, S	Rack for 50 mL centrifuge tubes	1	
D	10% formalin solution buffered with borax	10 mL per replicate	See Appendix C
D	Formalin-resistant gloves	1 pair	
D	Safety goggles or face shield	1	
D	Small syringe or bulb pipette	1	

1. "D" = diatom sample, "S" = soft-bodied algal sample, "C" = chlorophyll a sample, "A" = ash-free dry mass sample

Needed for[1]:	Item	Quantity / Site	Specifications
D	Vermiculite packing material	as needed	
S	Turkey baster	1	
S (see note)	25% glutaraldehyde solution (to be dispensed in a laboratory fume hood, wearing appropriate safety gear)	5 mL per replicate	*Note: could be added by taxonomy lab, with prior notification*
S	Calculator	1	
S	Small metric ruler (waterproof)	1	
S	Small Ziploc bag	1	
S, C, A	Whirl-pak bag, 100 mL	3 per replicate	Cole Parmer 06498-00
C, A	Filter forceps	1	Fisher 0975350
C, A	Filtering chamber/tower, 47 mm, plastic	1	Hach 2254400
C, A	Hand vacuum pump	1	Fisher 13-874-612B
C, A	Aluminum foil	~100 cm^2 per replicate	
C, A	Deionized water	500 mL	
C, A	Dry ice (if not returning to lab immediately following the day's fieldwork)	10 lbs	
C	Glass fiber filter, 47 mm, 0.7 µm pore size	1 per replicate	Fisher 09804142H
C	Snapping Petri dish, 47 µm	1 per replicate	Fisher 08-757-105
A	Glass fiber filter, 47 mm, 0.7 µm pore size; foil-wrapped and pre-combusted for ash-free dry mass (AFDM)	1 per replicate	

1. "D" = diatom sample, "S" = soft-bodied algal sample, "C" = chlorophyll a sample, "A" = ash-free dry mass sample

Table 7.
Physical Habitat Data Collection

Item	Quantity / Site	Specifications
GPS receiver	1	
Transect tape; 150 m	1	
Lengths of rope (7.5 m and 12.5 m)	1 each	
Small metric folding ruler (waterproof)	1	
Digital watch	1	
Stadia rod	1	
Clinometer	1	
Autolevel and tripod	1	
Current velocity meter and top-setting rod	1	
Convex spherical densiometer	1	Taped to expose only 17 intersections of the grid (see Figure 12)
Transect flags	21 total	Two colors; label with main transect (11 ct.) and inter-transect (10 ct.) names
Algae viewing bucket (optional)	1	See Appendix C
Small/slender rod with 1, 5, and 20 mm marks	1	For measuring microalgal thickness
Rangefinder (optional)	1	
Fresh orange peel	1	

B Information Resources for Avoiding Introduction of Invasive Species and Pathogens into Streams

The following is an adaptation of an excerpt taken from an EMAP-based Quality Assurance Project Plan developed by the California Department of Fish and Game Aquatic Bioassessment Laboratory (2008).

Organisms of concern in the U.S. include, but may not be limited to, Eurasian watermilfoil (*Myriophyllum spicatum*), New Zealand mud snail (*Potamopyrgus antipodarum*), zebra mussel (*Dreissena polymorpha*), *Myxobolus cerebralis* (the sporozoan parasite that causes salmonid whirling disease), and *Batrachochytrium dendrobatidis* (a chytrid fungus that threatens amphibian populations).

Field crews must be aware of regional species of concern, and take appropriate precautions to avoid transfer of these species. Crews should make every attempt to be apprised of the most up-to-date information regarding the emergence of new species of concern, as well as new advances in approaches to hygiene and decontamination to prevent the spread of all such organisms (e.g., Hosea and Finlayson, 2005; Schisler et al., 2008).

There are several online sources of information regarding invasive species, including information on cleaning and disinfecting gear:

Whirling Disease Foundation
www.whirling-disease.org

USDA Forest Service - Preventing Accidental Introductions of Freshwater Invasive Species
www.fs.fed.us/invasivespecies/documents/Aquatic_is_prevention.pdf

California Department of Fish and Game
www.dfg.ca.gov

U.S. Geological Survey Nonindigenous Aquatic Species: general information about freshwater invasive species
http://nas.er.usgs.gov

Protect Your Waters - Co-sponsored by the U.S. Fish and Wildlife Service
www.protectyourwaters.net/hitchhikers

The California State Water Resources Control Board Aquatic Invasive Species website

www.swrcb.ca.gov/water_issues/programs/swamp/ais

REFERENCES

Hosea, R.C. and B. Finlayson. 2005. Controlling the spread of New Zealand mudsnails of wading gear. California Department of Fish and Game, Office of Spill Prevention and Response, Administrative Report 2005-02, Sacramento.

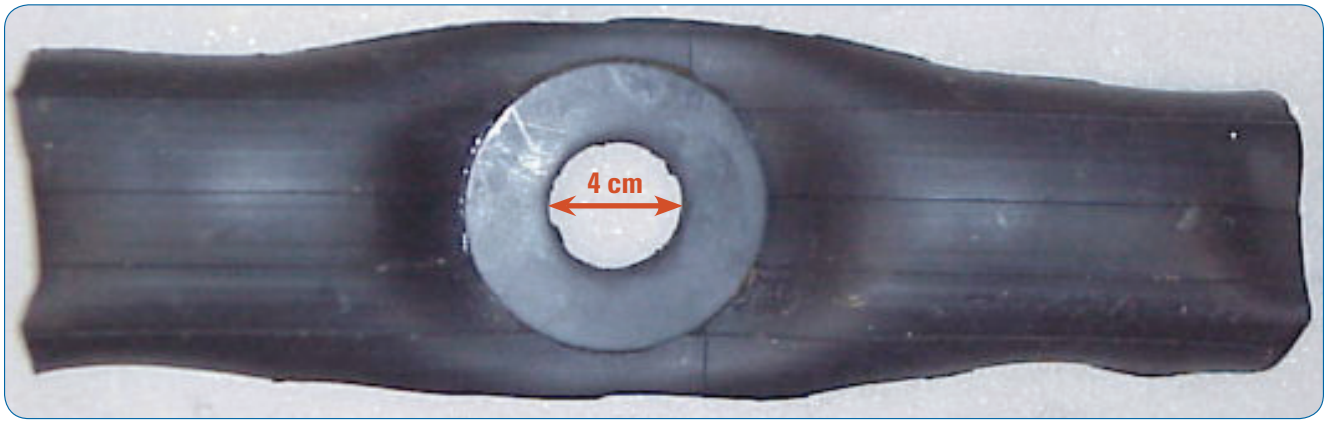
Schisler, G.J., N.K.M. Vieira, and P.G. Walker. 2008. Application of Household Disinfectants to Control New Zealand Mudsnails. North American Journal of Fisheries Management 28:1171-1176.

CONSTRUCTION OF ALGAE SAMPLING TOOLS C

This appendix provides step-by-step instructions for constructing the devices used for sampling algae. It also provides a recipe for the formalin fixative for diatoms.

1. RUBBER DELIMITER

The rubber delimiter for use on "erosional"/hard substrates like cobbles and wood is made from a sliced-open mountain bike inner tube that has a 4-cm diameter hole cut in the middle. The hole should be reinforced with a rubber gasket affixed to the tube with rubber cement.

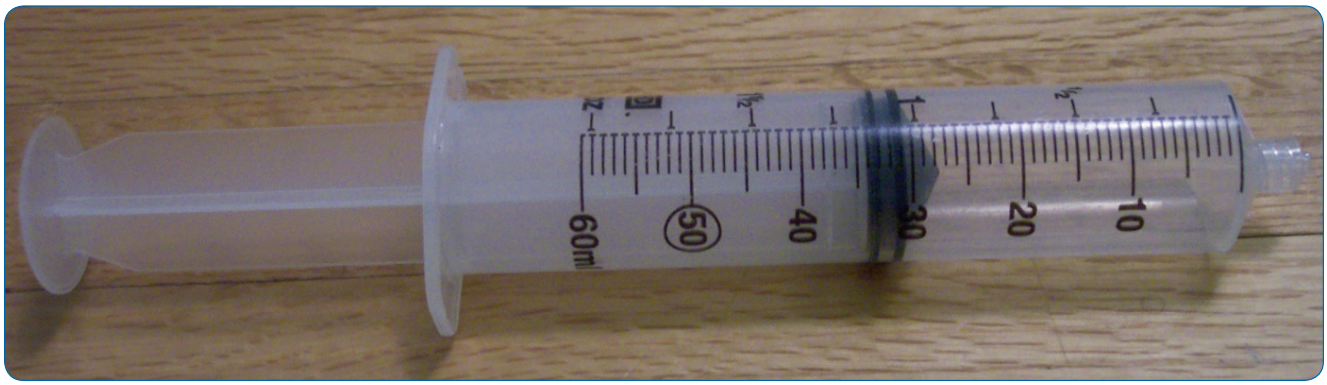


2. PVC DELIMITER

The PVC delimiter for "depositional"/soft substrates like sand, small gravel, and silt is made from a 1½" sewer cleanout, which can be found at a home-improvement or plumbing supply store. The hole in the bottom of the cleanout is 4 cm in diameter. The bottom edge of the cleanout is filed to make it sharp, to ease insertion into silt/sand. To facilitate consistent sampling, it is useful to paint a bright line indicating a depth of 1 cm around the outer surface of the bottom of the sampling device. This indicates the depth to which to insert the delimiter when sampling.

3. SYRINGE SCRUBBER

The syringe scrubber is for use on hard substrates that cannot be picked up out of the stream, like submerged bedrock and concrete channel bottoms. It is made from a 60 mL syringe barrel with the end cut off and its plunger fitted with Velcro-type material. Disposable, white (non-pigmented) scrubbing pads circles are then affixed to the end of the plunger and used to scrub the algae from the substrate.



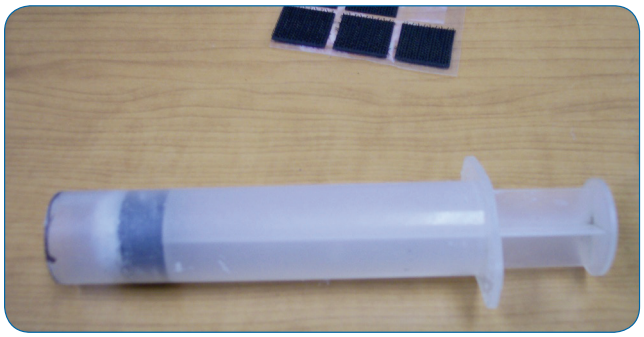
You will need a 60-mL plastic syringe for each sampler you want to make. Remove the plunger and saw the conical end off the plastic syringe, then sand the bottom so it is flat all the way around and fits tightly against a flat surface.

Firmly affix the rubber end to the plastic plunger by removing the rubber tip, applying glue to the "naked" end of the plunger, and replacing the rubber cap. Allow glue to cure. Then cut the conical part off the plunger tip so that only a flat surface of rubber remains.

Cut a circle of Velcro®-style hook material to fit the size of the plunger. Use a waterproof adhesive to affix the "Velcro®" circle to the end of the plunger.

Obtain some white scrubbing pad material (make sure it is not pigmented so it will not end up interfering with eventual chlorophyll *a* analysis of the samples collected.) Cut a supply of circles to fit the size of the plunger.

Before each sampling event, attach a fresh circular scrubbing pad to the end of the plunger. This is a head-on view of the plunger, with the scrubbing pad circle attached.



This is what the syringe sampler looks like when it is ready to be used.

4. VIEWING BUCKET (OPTIONAL)

A viewing bucket can be useful for visualizing submerged algae, particularly in instances of a turbulent stream surface that obscures the stream bottom. A viewing bucket can be constructed from a narrow cylinder of clear Plexiglas (approximately 8 inches in diameter) whose bottom is fitted with a circle of thick glass, and secured in place with a silicone seal. If desired, one or two handles can also be fashioned out of Plexiglas and attached to the side(s) of the cylinder. The use of the viewing bucket is optional.

5. PREPARING A 1-L SOLUTION OF 10-PERCENT BUFFERED FORMALIN (MOULTON ET AL. 2002)

1. Add 100 mL of formaldehyde (37-40%) to 900 mL of water in a chemically resistant, non-breakable bottle.
2. Add about 3 g of borax to 10 mL of water and mix.
3. Add dissolved borax solute, to buffer formalin solution.
4. Tightly seal the bottle and mix by carefully inverting the bottle several times.
5. Label the outside of the bottle with "10-percent buffered formalin," the date of preparation, and related hazardous chemical stickers.

REFERENCE

Moulton II, S.R., J.G. Kennen, R.M. Goldstein, and J.A. Hambrook. 2002. Revised protocols for sampling algal, invertebrate, and fish communities as part of the National Water-Quality Assessment Program, Open-File Report 02-150.

STANDARD OPERATING PROCEDURES (SOP) FOR USING GLUTARALDEHYDE FOR THE PRESERVATION OF SOFT ALGAE

D

(adapted from the Aquatic Bioassessment Laboratory, California Department of Fish and Game)

Note: Glutaraldehyde must only be handled by trained individuals who understand the safe handling and use of this chemical

1. SCOPE AND APPLICATION

Glutaraldehyde is a colorless liquid with a pungent odor used as a preservative and sterilant. This SOP covers the use of Glutaraldehyde by Department of Fish and Game OSPR laboratories as a preservative for soft bodied algae.

2. PHYSICAL HAZARDS

The physical hazards associated with the use of Glutaraldehyde include:

- Incompatibility with strong oxidizing substances and bases
- Corrosive to metals
- Production of Carbon Monoxide and Carbon Dioxide during decomposition
- Discolors on exposure to air

3. HEALTH HAZARDS

The health hazards associated with the use of Glutaraldehyde include;

Inhalation

- Regulatory limit of 0.05 ppm as a ceiling level
- Chemical burns to the respiratory tract
- Asthma and shortness of breath
- Headache, dizziness, and nausea

Skin

- Sensitization or allergic reactions, hives
- Irritations and burns
- Staining of the hands (brownish or tan)

Eyes

- Irritation and burns. Eye contact causes moderate to severe irritation, experienced as discomfort or pain, excessive blinking and tear production
- May cause permanent visual impairment
- Conjunctivitis and corneal damage

Ingestion

- Gastrointestinal tract burns; Central nervous system depression, excitement
- Nausea, vomiting
- Unconsciousness, coma, respiratory failure, death

*Note: Oral toxicity of Glutaraldehyde **increases** with dilution*

4. ENGINEERING CONTROLS

Strict engineering controls will be followed when using Glutaraldehyde. This chemical and processes using this chemical will only be used under a laboratory fume hood meeting the requirements of Title 8, CCR Section 5154.1. At no time will containers of Glutaraldehyde be opened outside of an operating fume hood.
Personnel using Glutaraldehyde will designate an area of the lab for its use. The area where it is used will be noticed with a sign reading:

CAUTION GLUTARALDEHYDE IN USE

Only trained personnel will be allowed to enter the designated area when using Glutaraldehyde.

5. PERSONAL PROTECTIVE EQUIPMENT

Personal Protective Equipment (PPE) is required to be worn at all times when working with Glutaraldehyde. This includes:

Eye Protection

- Chemical splash goggles; or
- Safety glasses with face shield

Hand Protection

- Nitrile or Polyvinyl Chloride (vinyl) gloves

Body Protection

- Lab coat with polypropylene splash apron that cover the arms

Any PPE with noticeable contamination will be immediately removed and the affected area washed with water. Gloves and apron will be removed before leaving the designated area. Disposable PPE (gloves and aprons) will not be re-worn. Disposable PPE will be disposed of in a sealed waste receptacle approved for hazardous waste. Any non-disposable PPE (lab coats, chemical goggles) with noticeable contamination will be rinsed or cleaned as soon as practical, and secured in a manner that does not allow contamination of laboratory personnel. Respiratory protection will not be required as long as strict engineering controls are followed.

6. SAFETY SHOWER AND EYEWASH

All employees using Glutaraldehyde must be aware of the location and use of the laboratory safety shower and eyewash, and must be able to reach it within 10 seconds from the time of contamination. At no time will processes using Glutaraldehyde be allowed that do not provide access to a safety shower and eyewash. Employees who have skin or eye contact with Glutaraldehyde will immediately stop all processes and proceed to the safety shower and eyewash station. The employee will rinse the affected area for a minimum of 15 minutes. If eye contact has occurred, the upper and lower eyelids must be lifted to allow adequate flushing of the eyes.

7. SPECIAL HANDLING PROCEDURES AND STORAGE REQUIREMENTS

Procedures will be followed that reduce exposure to Glutaraldehyde vapor to the lowest reasonable level. This includes:

- Ensure Glutaraldehyde is only used under a fume hood
- Use only enough Glutaraldehyde to perform the required procedure
- Every effort must be made to minimize splashing, spilling, and personnel exposure
- Once specimens are preserved, they will be capped or secured in a way that does not allow Glutaraldehyde vapor to escape into the lab
- At no time will open containers be removed from the fume hood
- All containers of Glutaraldehyde or solutions containing Glutaraldehyde will be appropriately marked with the chemical name, and hazard warning label at the end of the work day or whenever there is a personnel change
- Glutaraldehyde will be stored in tightly closed containers in a cool, secure, and properly marked location

8. WASTE DISPOSAL

Excess Glutaraldehyde and all waste material containing Glutaraldehyde must be placed in an unbreakable secondary container labeled with the following “HAZARDOUS WASTE GLUTARALDEHYDE.” Wastes will be disposed of through the laboratory hazardous waste contract.

9. SPILL AND ACCIDENT PROCEDURES

Drips and splashes will be wiped up immediately with a sponge, towel, or mop. Any material used to clean spills will be disposed of as hazardous waste. Large spills (Greater than 300 CC) require response by a local Hazmat team. The Hazmat team will be called by the laboratory supervisor. In the event of a large spill personnel will immediately leave the laboratory, and not re-enter until cleared by the laboratory supervisor.

10. TRAINING

All personnel engaged in the use of Glutaraldehyde will be trained on the hazards associated with this chemical, before use. The training will include;

- OSPR's Hazard Communication Program and information contained in the chemical's Material Safety Data Sheet (MSDS)
- Health hazards and routes of exposure
- Specific procedures and techniques for use and handling
- Use of PPE and engineering controls
- The contents and requirements of this Standard Operating Procedure.

STANDARD OPERATING PROCEDURES (SOP) FOR USING FORMALIN FOR THE PRESERVATION OF DIATOMS

E

(adapted from the US EPA EMAP program; Peck et al. 2006)

Note: Formalin must only be handled by trained individuals who understand the safe handling and use of this chemical. All personnel engaged in the use of formalin will be trained on the hazards associated with this chemical before use. The training will include the information contained in the chemical's Material Safety Data Sheet (MSDS).

Formaldehyde (or formalin) is highly allergenic, toxic, and dangerous to human health (potentially carcinogenic) if utilized improperly. Formalin vapors and solution are extremely caustic and may cause severe irritation on contact with skin, eyes, or mucous 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 immediate 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.

During the course of field activities, a team 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. You should always err on the side of personal safety for yourself and your fellow field crew members.
- 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 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.

Follow Department of Transportation (DOT) and the Occupational Safety and Health Administration (OSHA) regulations for handling, transporting, and shipping hazardous material 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. Dispose of all wastes in accordance with approved procedures (e.g., National Institute for Occupational Safety and Health 1981, US EPA 1986).

To dispense formalin in the field, wear formalin-safe gloves and safety goggles. Use a small syringe or bulb pipette to add 10 mL of 10% buffered formalin solution to 40 mL of the diatom sample in a 50 mL centrifuge tube. Alternatively, in order to avoid dispensing formalin solution in the field, clean 50 mL centrifuge tubes that will hold the diatom samples can also be pre-loaded with 10 mL of 10% buffered formalin in a laboratory fume hood prior to going into the field.

The preparation of the 10% buffered formalin stock solution should always be done by trained personnel under a laboratory fume hood while wearing protective gloves, clothing, and goggles.

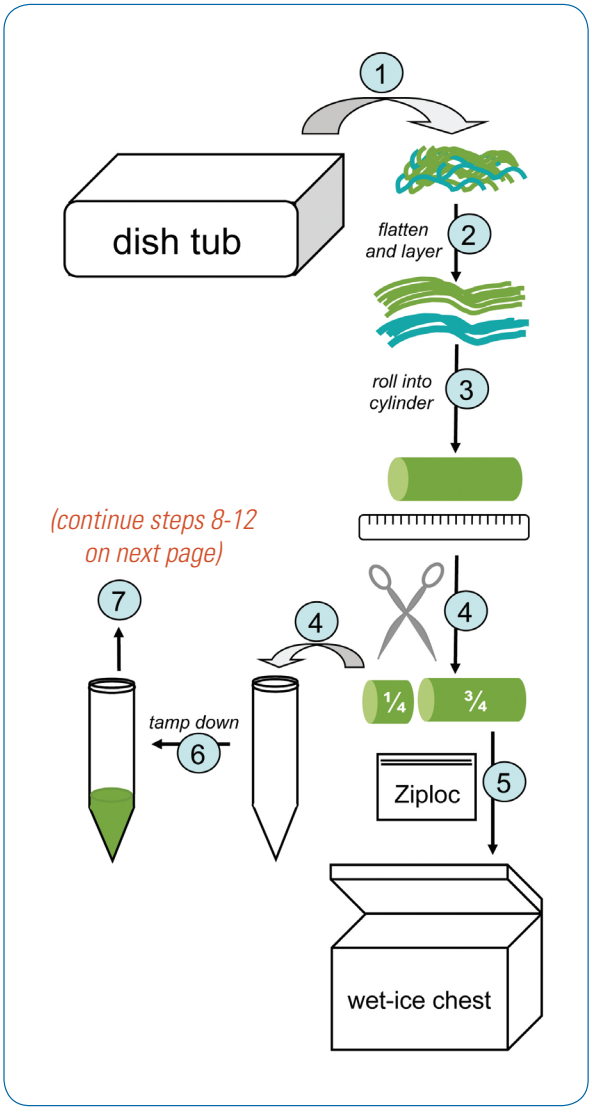
REFERENCE

Peck, D. V., A. T. Herlihy, B. H. Hill, R. M. Hughes, P. R. Kaufmann, D. Klemm, J. M. Lazorchak, F. H. McCormick, S. A. Peterson, P. L. Ringold, T. Magee, and M. Cappaert. 2006. Environmental Monitoring and Assessment Program-Surface Waters Western Pilot Study: Field operations manual for wadeable streams. U.S. Environmental Protection Agency, Washington, D.C. EPA/620/R-06/003.

PROCESSING SOFT-BODIED ALGAL AND DIATOM SAMPLES WHEN MACROALGAL CLUMPS ARE IN THE SAMPLE

F

The first step involves delivering a known quantity of macroalgae to the soft-bodied algae sample tube.



1) Gently wring excess water out of macroalgae and remove from dish tub.

2) Flatten each distinct taxon of macroalgae into even "sheets" and lay atop one another to distribute the volume of each as equally as possible.

3) Once the various layers of macroalgae are evenly spread upon one another, gently roll the stack into a cylinder shape that is roughly straight and even in thickness along its length.

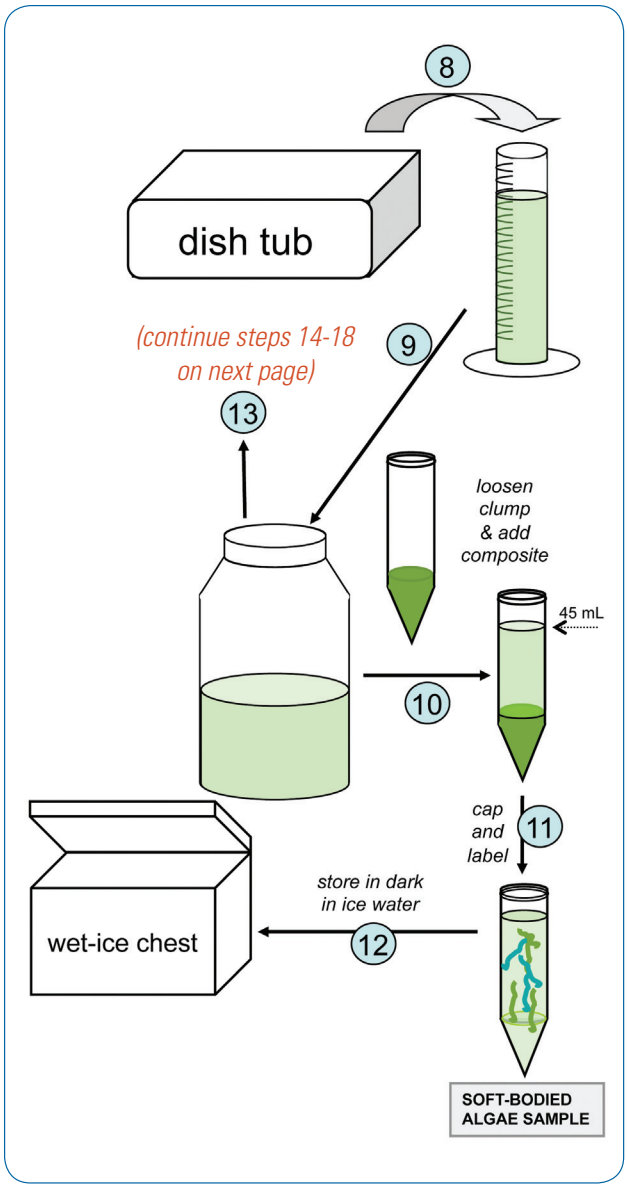
4) Measure and cut off ¼ of the cylinder and place that piece into the empty soft-bodied algae taxonomic ID sample tube.

5) Seal the remaining ¾ of the macroalgae in a clean plastic bag and place inside a cool, dark place such as the wet-ice chest.

6) Using a clean, blunt-ended object, tamp down the ¼ clump of macroalgae in the tube to make it dense and flatten the top surface. Estimate the volume of the macroalgal clump using the graduations on the tube and record this value on the Ratio Restoration worksheet (Figure 8).

7) Add composite sample solution to the tube according to directions on next page.

Some of the liquid composite sample is now added to the tube containing the macroalgae, but first the volume of the entire liquid composite collected must be measured.



8) Agitate the composite sample in the dish tub in order to suspend and mix the microalgae. Wait a few seconds to let the sand/silt settle. Quickly pour only the liquid (leaving silt/sand behind) into a graduated cylinder to measure its volume. Rinse substrate as necessary. Record TOTAL volume of the composite liquid (+ rinsate) on the datasheet, sample labels, and Ratio Restoration worksheet.

9) Pour the liquid composite sample into a clean, 1 L sample bottle

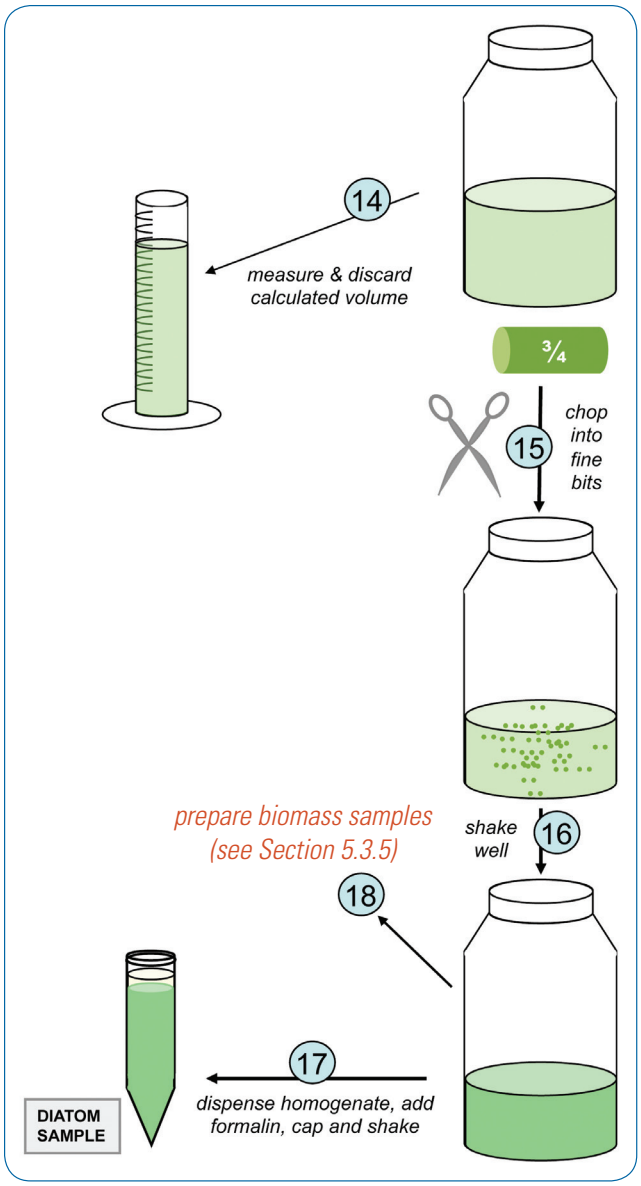
10) Loosen the macroalgae in the sample tube a little so it is no longer a dense clump lodged in the bottom and then pour freshly-agitated composite liquid into sample tube up to the 45 mL mark.

11) Cap the sample tube tightly. Affix a filled-out label to sample tube and cover with clear tape.

12) Place the tube in the dark in a wet-ice chest (not dry ice). Do not allow the algae to freeze. **Glutaraldehyde will need to be added to the tube within 4 days of sample collection, and preferably as soon as possible.**

13) According to directions on the next page, restore the original ratio of macroalgae to liquid composite in order to prepare the remainder of the samples.

The remainder of the macroalgae is now cut into tiny bits, which are added back to the liquid composite. **But the original ratio of macroalgae:liquid must first be restored.** The diatom and biomass samples are then prepared.



14) Use your Ratio Restoration worksheet to determine how much of the liquid composite to pour off. First shake the bottle vigorously, then measure and discard the appropriate volume.

15) Remove the macroalgal clump from the wet-ice chest. Chop the algae into **very fine** (eyelash length or smaller) pieces and add these to the liquid composite.

16) Cap and shake the bottle vigorously in order to homogenize the chopped algae into the liquid as thoroughly as possible.

17) Pour 40 mL of the freshly-agitated homogenate into the diatom sample tube. Add 10 mL 10% buffered formalin solution, observing all formalin safety precautions. Cap the tube, shake, and affix a sample label.

18) After both taxonomic ID samples have been prepared, the remainder of the homogenate is used for the biomass samples (chlorophyll *a* and ash-free dry mass). 25 mL of freshly shaken homogenate is filtered for each biomass sample. See Section 5.3.5.

SWAMP
Surface Water
Ambient Monitoring
Program

For more information, please contact:

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Southern California Coastal Water Research Project
3535 Harbor Blvd., Suite 110
Costa Mesa, California 92626
bettyf@sccwrp.org

CALIFORNIA
Water Boards

www.waterboards.ca.gov/water_issues/programs/swamp/

APPENDIX H:

INSTRUMENT CALIBRATION AND MEASUREMENTS

This Appendix represents current equipment used by the District which may change as programmatic updates are implemented.

ISCO 3700 Portable Sampler

For operational instructions with graphics, along with important notes, please refer to the **Operation Manual which can be found online at the following website** **http://www.isco.com/pcfiles/PartPDF/SL000004/UP0019H4.pdf** .

Hydrolab Quanta Multi-Parameter Water Quality Monitor

For detailed instructions with graphics, along with important notes, please refer to the **Operation Manual**.

A. Calibration

1. Ensure that the sensor probe is connected.

2. Wash the sensor in distilled water a few times and put some of the pH 4 standard solution into the calibration beaker to the marked line.

3. Immerse the sensor in the calibration beaker.

4. Press the **POWER** key.
   a. Use arrow keys to select 'CALIB' and press Enter
   b. Select parameter you wish to calibrate and press Enter
   c. Use arrow keys to adjust reading to standard solution press Enter

5. Fully rinse probe and calibration cup with DI water then dry before filling with next standard solution.
   a. When calibrating pH calibrate 7.0 first then 10.0 or 4.0. (NOTE Quanta only does 2 point pH calibration)
   b. When calibrating DO sensor fill cup with DI water to O-ring of membrane on DO probe. Use accurate mmHg to calibrate %DO this will calibrate mg/L DO at the same time.

B. Measurement

1. Always attach the Guard to protect to the sensors before taking measurements.

2. Slowly immerse the sensor in the sample.

3. Turn the power on and allow measurements to stabilize.

4. Record the measurements on the *Field Data Sheet* while out in the field. Select the SCREEN icon and use the **ENTER** key to switch measurement items.

5. After taking the measurement:

   5.1 Turn the power off.

   5.2 Use DI water to completely wash off the sample on the sensor and then wipe water drops.

   5.3 Pour about 20 mL (about 2 cm from the bottom) of distilled water in the storage cup and install it on the sensor probe and store the instrument in the carrying case.

C. Specifications

Performance Specifications

	Range	Accuracy	Resolution
Temperature	-5°C to 50°C	±0.2°C	0.01°C
Dissolved Oxygen	0 to 50 mg/L	±0.2 mg/L ≤ 20 mg/L ±0.6 mg/L > 20 mg/L	0.01 mg/L
Specific Conductance	0 to 100 mS/cm	±1% of reading ±1 count	4 digits
pH	2 to 12 units	±0.2 units	0.01 units
ORP	-999 to 999 mV	±25 mV	1 mV
Vented Depth (10m)	0 to 10 m	±0.003 m (±0.01 ft)	0.001 m
Depth (25m)	0 to 25 m	±0.1 m	0.1 m
Depth (100m)	0 to 100 m	±0.3 m	0.1 m
Turbidity	0 to 1000 NTU	±5% of reading ±1 NTU	0.1 NTU < 100 NTU 1 NTU ≥ 100 NTU
Salinity	0 to 70 PSS	±1% of reading ±1 count	0.01 PSS

YSI 6920 V2 Multi-Parameter Water Quality Sonde

For detailed instructions with graphics, along with important notes, please refer to the **User Manual**.

A. Calibration

1. Ensure that the sensor probe is connected and sensor probe is fully immersed in standard solution.

2. Press the **POWER** key.
   - d. Select Sonde Menu and press Enter
   - e. Select Calibrate and press Enter
   - f. Select parameter you wish to calibrate and press Enter
   - g. Select 1-3 point calibration dependent on parameter and press Enter
   - h. Type in standard solution value and press Enter
   - i. Wait for readings to stabilize then press Enter to calibrate sensor

3. Fully rinse probe and calibration cup with DI water then dry before filling with next standard solution.
   - a. When calibrating pH calibrate 7.0 first then 10.0 or 4.0.
   - b. When calibrating optical sensors (DO or Turbidity clean optics before calibrating)
   - c. When calibrating DO sensor allow atmosphere in calibration cup to stabilize for 15 mins with DI water before calibration.

B. Measurement

1. Always attach the flow-thru Guard to protect to the sensors before taking measurements.

2. Slowly immerse the sensor in the sample.

3. Turn the power on and allow measurements to stabilize.

4. Record the measurements on the *Field Data Sheet* while out in the field.

5. After taking the measurement:

   5.1 Turn the power off.

   5.2 Use DI water to completely wash off the sample on the sensor and then wipe water drops.

   5.3 Pour about 20 mL (about 2 cm from the bottom) of distilled water in the storage cup and install it on the sensor probe and store the instrument in the carrying case.

C. Specifications

6920V2-2 SONDE

Available Sensors	Temperature, Conductivity, pH, ORP, one ion selective electrode (ammonium, nitrate, or chloride), and Depth (shallow, medium, shallow vented). Two total optical sensors (ROX Optical DO, Turbidity, Chlorophyll, Rhodamine WT, BGA-PC, or BGA-PE).
Operating Environment	Medium: fresh, sea, or polluted water Temperature: -5 to +50 °C for most sensors Depth: 0 to 200 feet (61 meters)
Storage Temperature:	-40 to +60 °C for sonde and all sensors except pH , pH/ORP, ISE and optical sensors -10 to +60 °C for pH, pH/ORP, ISE, and optical sensors
Material:	Polyurethane, PVC, Stainless Steel
Diameter:	2.9 inches (7.4 cm)
Length:	Approximately 18.25 inches (46.4 cm) with no depth, 19.63 inches (49.9 cm) with depth
Weight:	Approximately 3.74 pounds (1.7 kg)
Computer Interface:	RS-232C, SDI-12
Internal logging memory size:	384 kilobytes (150,000 individual parameter readings)
Power:	8 AA-size Alkaline Batteries or External 12 VDC
Battery Life:	Approximately 32 days at 20 C at a 15 minute logging interval with ROX Optical DO, another optical sensor (turbidity, chlorophyll, Rhodamine WT, BGA-BC, or BGA-PE), temperature, conductivity, and pH active. Battery life is heavily dependent on sensor configuration and is given above for a typical sensor ensemble. If you have a different sensor configuration, set up your sonde for a deployment in the Run|Unattended menu and check the projected approximate battery life.

Temperature

Sensor Type.....Thermistor
Range.....-5 to 50 °C
Accuracy.....+/- 0.15 °C
Resolution.....0.01 °C
Depth.....200 meters

ROX Optical Dissolved Oxygen, mg/L (Calculated from % air saturation, temperature and salinity)
Sensor Type..... Optical, Luminescence Lifetime
Range.....0 to 50 mg/L
Accuracy.....0 to 20 mg/L, +/- 1 % of the reading or 0.1 mg/L, whichever is greater
20 to 50 mg/L, +/- 15 % of the reading; Relative to Calibration Gases.
Resolution.....0.01 mg/L
Temperature Range -5 to 50 C

pH
Sensor Type.....Glass combination electrode
Range.....0 to 14 units
Accuracy.....+/- 0.2 units
Resolution.....0.01 units
Temperature Range -5 to 50 C
Depth.....200 meters

Turbidity
Sensor type..... Optical, 90 ° scatter, with mechanical cleaning
Range..... 0 to 1000 NTU
Accuracy.....+/- 2% of the reading or 0.3 NTU (whichever is greater), in YSI AMCO-AEPA standards
Resolution.....0.1 NTU
Temperature Range -5 to 50 C
Depth..... 61 meters

Conductivity*
Sensor Type.....4 electrode cell with autoranging
Range.....0 to 100 mS/cm
Accuracy.....+/- 0.5% of reading + 0.001 mS/cm
Resolution.....0.001 mS/cm to 0.1 mS/cm (range dependent)
Temperature Range -5 to 60 C
Depth.....200 meters

Horiba U-22XD Multi-Parameter Water Quality Monitoring System

For detailed instructions with graphics, along with important notes, please refer to the **Operation Manual**.

A. Calibration
   1. Ensure that the sensor probe is connected.

   2. Press the **POWER** key.

   3. Wash the sensor in distilled water a few times and put some of the pH 4 standard solution into the calibration beaker to the marked line.

   4. Immerse the sensor in the calibration beaker.

   5. Press the CAL auto calibration mode key in one of the Measurement modes **pH**, **COND**, **TURB**, **DO**. **AUTO** and **CAL** appear and the instrument enters the **AUTO** calibration mode.

   6. Press the **ENT** key to start **AUTO** calibration. Upon completion of the **pH**, **COND**, **TURB**, **DO**, and **DEP** modes, "End" will be displayed on the screen.

   7. Press the **MEAS** key to return to the Measurement mode.

B. Measurement
   1. Slowly immerse the sensor in the sample.

   2. Select the measurement item. Use the **MEAS** key to switch measurement items in the following order: **pH**, **COND**, **TURB**, **DO**, **TEMP**, **DEP**, **SAL**, **TDS**, **σ**, **ORP**, **TIME**, then back to **pH**.

   3. After taking the measurement:

      3.1 Turn the power off.

      3.2 Use tap water to completely wash off the sample on the sensor and then wipe water drops.

      3.3 Pour about 20 mL (about 2 cm from the bottom) of distilled water in the probe cap and install it on the sensor probe. Place the rubber cap on the connector and store the instrument in the carrying case.

   4. Water quality data should always be recorded to the *Field Data Sheet* while out in the field. To additionally manually store the measurement data (up to 2880 sets) in the instrument for later download:
      4.1 Make sure that **MAN** is displayed as the Measurement mode.

      4.2 Press the **ENT** key. Data storage starts, **DATA IN** and the data set number is displayed on the screen, and the measured value to be stored and the measurement item are displayed in order at approximately 0.5 second intervals. After the data are stored to memory, the screen returns to the original Measurement mode. Note the data set number on the *Field Data Sheet*.

5. Viewing stored data on the Horiba (note that the data cannot be transferred to a PC, only to a Horiba printer):
   5.1 Press the **DATA** key in the Measurement mode. The instrument goes into **DATA** mode.

   5.2 Press the **DATA** key. The measurement data are displayed. Data you want to call can be displayed by selecting a measurement item and data number.

   5.3 Press the **UP/DOWN** keys to switch the measurement item or number that has been selected with the **DATA** key.

   5.4 Press the **DATA** key. Use the **UP/DOWN** keys to switch between "Year, Month, Day' and "Hour, Minute, Second".

C. Specifications

Measurement range

Measurement item		Measurement range		Measurement units
		Expanded	Standard	
pH		0.00 to 14.00	0.0 to 14.0	pH
		—	−1999 to 1999	mV in pH measurement
Conductivity (COND)	Range 1	0.90 to 9.99	0.9 to 9.9	S/m
		9.0 to 99.9	9 to 99	mS/cm
	Range 2	0.090 to 0.999	0.09 to 0.99	S/m
		0.90 to 9.99	0.9 to 9.9	mS/cm
	Range 3	0.0 to 99.9	0 to 99	mS/m
		0.000 to 0.999	0.00 to 0.99	mS/cm
Turbidity (TURB) *1		0.0 to 800.0	0 to 800	NTU (nephelometric turbidity units) or mg/L
Dissolved-oxygen (DO)		0.00 to 19.99	0.0 to 19.9	mg/L
		0.0 to 199.9	0 to 199	%
Temperature (TEMP)		0.00 to 55.00	0.0 to 55.0	°C
Water depth (DEP)		0.0 to 100.0	0 to 100	m
		0.0 to 330.0	0 to 330	ft
Salinity (SAL)		0.00 to 4.00	0.0 to 4.0	%
Total dissolved solids	Range 1	5.5 to 65.0	5 to 65	g/L
(TDS) *2	Range 2	0.55 to 6.50	0.5 to 6.5	g/L
	Range 3	0.000 to 0.650	0.00 to 0.65	g/L
Seawater specific gravity (σ_t)		0.0 to 50.0	0 to 50	–
Oxygen-reduction potential (ORP)		—	−1999 to 1999	mV

*1: Depending on the concentration range, the minimum turbidity is displayed as follows:
0 to 100 NTU ... 1 NTU for standard readout, 0.1 NTU for expanded readout.
100 to 800 NTU ... 10 NTU for standard readout, 1 NTU for expanded readout.
*2: The TDS range depends on the TDS factor settings. (Above ranges are given for a TDS coefficient of 0.65.)

Oakton Waterproof pHTestr 2

For detailed instructions with graphics, along with important notes, please refer to the **Operation Manual**.

A. Calibration

1. Before you begin: Remove electrode cap. To condition electrode, immerse electrode in electrode storage solution, buffer, or tap water for at least 30 minutes. **DO NOT USE DE-IONIZED WATER.**

2. Calibration: Calibration should be done regularly, typically every day that the *Testr* is used. Calibrate the *pHTestr 2* at three points (pH 4, 7, 10).

   2.1 Press **ON/OFF** button to switch unit on.

   2.2 Dip electrode 1/2" to 1" into chosen buffer (pH 4, 7, or 10).

   2.3 Press **CAL** button to enter Calibrate (CA) mode. 'CA' flashes on the display. Then, a pH value close to the pH buffer value will flash repeatedly.

   2.4 After at least 30 seconds (about 30 flashes) press the **HOLD/CON** button to confirm calibration. The display will show '**CO**' and then switch to the buffer value reading.

   2.5 Repeat with other buffers if necessary (*pHTestr 2* only). Rinse electrode in tap water before dipping into next buffer.

B. Measurement

1. Remove cap from the electrode and press the **ON/OFF** button to switch *Testr* on.

2. Dip the electrode 1/2" to 1" into the test solution. Stir once and let the reading stabilize.

3. Note the pH or press **HOLD/CON** button to freeze the reading. Press **HOLD/CON** again to release the reading.

4. Press **ON/OFF** to turn off *Testr*. If you do not press a button for 8.5 minutes, the *Testr* will automatically shut off to conserve batteries.

5. If possible, keep a small piece of paper or sponge in the electrode cap – moistened with clean water or electrode storage solution (**NOT DE-IONIZED WATER**) – and close the cap over the electrode.

C. Specifications

Specifications

	WP pHTestr 1	WP pHTestr 2
Range	–1.0 to 15.0 pH	
Resolution	0.1 pH	
Accuracy	±0.2 pH	±0.1 pH
Calibration	1 point (pH 4.0; 7.0; or 10.0)	3 points (pH 4.0; 7.0 and 10.0)
ATC	No	Yes
Operating Temperature	0 to 50°C (32 to 122°F)	
Functions	ON/OFF; HOLD; CA (Calibrate); CO (Confirm display); auto buffer recognition; auto-shutoff after 8.5 min. of nonuse	
Power	Three 1.5 V batteries (included). 24 hours continuous use (approx. 720 tests per battery set)	
Dimensions	6.5"L x 1.5" dia. (165 x 38 mm dia.)	
Weight	3.25 oz (90 gms)	

Oakton TDSTestr 20

For detailed instructions with graphics, along with important notes, please refer to the **Operation Manual**.

A. Calibration

1. Before you Begin: Remove electrode cap. Switch unit on for 15 minutes to stabilize the batteries. Soak electrodes for a few minutes in alcohol to remove oils. **Caution: Never immerse the electrode above color band as it will damage the instrument electronics.**

2. Calibration: Your tester features push- button calibration at two points (one per range). Select a calibration standard appropriate for each range: low range, between 150 to 1999 µS; high range, between 2.0 to 19.99 mS. It is best to select a standard close to the test solution value, and one that has a similar chemical make-up to the test solution.

   2.1 Rinse the electrode in tap or deionized water, then in a known calibration standard.

   2.2 Switch unit on (ON/OFF button).

   2.3 Dip electrode into calibration standard. **DO NOT immerse above color band!**

   2.4 Press CAL/CON button to enter Calibrate mode. 'CA' flashes on the display. An uncalibrated conductivity value close to the calibration standard value will flash.

   2.5 Wait at least 30 seconds (about 30 flashes) for the reading to stabilize. Press the HOLD/INC button repeatedly to adjust reading to match value of the known calibration standard.

   2.6 Press CAL/CON button to confirm calibration. The display will show 'CO' and then switch to a calibrated conductivity reading.

   2.7 For second range, repeat steps 1-6.

B. Measurement

1. Remove cap from electrode. Switch unit on (ON/OFF button).

2. Rinse the electrode in tap or deionized water, then in a small portion of test solution.

3. Dip the electrode into the test solution. Stir once and let the reading stabilize. **DO NOT immerse electrode above color band!**

4. Allow time for the Automatic Temperature Compensation to correct the readings for solution temperature changes.

5. Note the reading once the display is stable. Press HOLD/CON button to freeze the reading. Press HOLD/CON again to release the reading.

6. Press ON/OFF to turn off Testr. If you do not press a button for 8.5 minutes, the Testr will automatically shut off to conserve batteries.

C. Specifications

Specifications

TDSTestr	10	20
Range	0 to 999 ppm/ 1.00 to 9.99 ppt	0 to 1999 µS/ 2.0 to 19.99 mS
Resolution	1 ppm/ 0.01 ppt	1 µS/ 0.01 mS
Accuracy	±2% full scale	
Calibration	two-point push button slope adjustments (one in each range)	
Calibration Standard	100 to 999 ppm 2.00 to 9.99 ppt	150 to 1999 µS/ 2.0 to 19.99 mS
Operating Temperature	32 to 122°F / 0 to 50°C	
ATC	32 to 122°F / 0 to 50°C (1.11% per °F / 2% per °C)	
Power	Four 1.5 V alkaline batteries (Eveready A76BP; supplied) 20 hrs. continuous use Alternate replacement Model Eveready 303 silver oxide, 70 hrs. continuous use.	
Dimensions	5.9" x 1.7" x 1" (151 x 42 x 24 mm)	
Weight	3.25 oz (90 gms)	

Hanna Instruments HI 98129 Waterproof pH, EC/TDS & Temperature Multimeter

For detailed instructions with graphics, along with important notes, please refer to the **Operation Manual**.

A. Calibration
   1. pH calibration:
      1.1 From measurement mode, press and hold the MODE button until CAL is displayed on the lower LCD. Release the button. The LCD will display pH 7.01 USE or pH 6.86 USE (if you have selected the NIST buffer set). The CAL tag blinks on the LCD.

      1.2 For a single-point pH calibration, place the electrode in any buffer from the selected buffer set (e.g. pH 7.01 or pH 4.01 or pH 10.01). The meter will recognize the buffer value automatically.
         1.2.1 If using pH 4.01 or pH 10.01, the meter will display OK for 1 second and then return to measurement mode.

         1.2.2 If using pH 7.01, after recognition of the buffer the meter will ask for pH 4.01 as second calibration point. Press the MODE button to return to measurement mode or, if desired, proceed with the 2-point calibration as explained below.

      1.3 A two-point pH calibration give better accuracy. For a two-point pH calibration, place the electrode in pH 7.01 (or 6.86 if you have selected the NIST buffer set). The meter will recognize the buffer value and then display pH 4.01 USE.
         1.3.1 Rinse the electrode thoroughly to eliminate cross-contamination.

         1.3.2 Place the electrode in the second buffer value (pH 4.01 or 10.01, or, if using NIST, pH 4.01 or 9.18). When the second buffer is recognized, the LCD will display OK for 1 second and the meter will return to normal measurement mode.

      1.4 The CAL symbol on the LCD means that the meter is calibrated.

   2. EC/TDS calibration:

      2.1 From measurement mode, press and hold the MODE button until CAL is displayed on the lower LCD.

      2.2 Release the button and immerse the probe in the proper calibration solution: HI7031 (1413 µS/cm) for HI98129 and HI7030 (12.88 mS/cm) for HI98130.

      2.3 Once the calibration has been automatically performed, the LCD will display OK for 1 second and the meter will return to normal measurement mode.

      2.4 Since there is a known relationship between EC and TDS readings, it is not necessary to calibrate the meter in TDS

      2.5 The CAL symbol on the LCD means that the meter is calibrated.

B. Measurement

1. pH measurement:

1.1 Select the pH mode with the SET/HOLD button.

1.2 Submerge the electrode in the solution to be tested.

1.3 The measurements should be taken when the stability symbol on the top left of the LCD disappears.

1.4 The pH value automatically compensated for temperature is shown on the primary LCD while the secondary LCD shows the temperature of the sample.

2. EC/TDS measurement:

2.1 Select either EC or TDS mode with the SET/HOLD button.

2.2 Submerge the probe in the solution to be tested. Use plastic beakers to minimize any electromagnetic interferences.

2.3 The measurements should be taken when the stability symbol on the top left of the LCD disappears.

2.4 The EC (or TDS) value automatically compensated for temperature is shown on the primary LCD while the secondary LCD shows the temperature of the sample.

C. Specifications

Range Temperature:	0.0 to 60.0°C or 32.0 to 140.0°F	
HI 98129	pH: 0.00 to 14.00	
	EC: 0 to 3999 µS/cm	
	TDS: 0 to 2000 ppm	
HI 98130	pH: 0.00 to 14.00	
	EC: 0.00 to 20.00 mS/cm	
	TDS: 0.00 to 10.00 ppt	
Resolution	0.1°C or 0.1°F	
HI 98129	0.01 pH; 1 µS/cm ; 1 ppm	
HI 98130	0.01 pH, 0.01 mS/cm ; 0.01 ppt	
Accuracy (@20°C/68°F)	Temperature	±0.5°C or ±1°F
	EC/TDS	±2% f.s.
	pH	±0.01
Typical EMC Deviation	Temperature	±0.5°C or ±1°F
	pH	±0.02 pH
	EC/TDS	±2% f.s.
Temp.Compensation	pH: Automatic	
	EC/TDS: with β=0.0 to 2.4%/°C	
Environment	0 to 50°C (32 to 122°F); RH 100%	
EC/TDS Conversion Factor	0.45 to 1.00 (CONV)	
Calibration	pH: at 1 or 2 points with 2 sets of memorized buffers (pH 4.01/7.01/10.01 or pH 4.01/6.86/9.18)	
	EC/TDS: automatic, at 1 point	
EC/TDS Calibration Solutions		
HI 98129	HI7031 (1413 µS/cm)	
	HI7032 (1382 ppm; CONV=0.5)	
	HI70442 (1500 ppm; CONV=0.7)	
HI 98130	HI7030 (12.88 mS/cm)	
	HI70038 (6.44 ppt; CONV=0.5 or 9.02 ppt; CONV=0.7)	
Electrode	HI 73127 pH electrode (**included**)	
Battery Type/Life	4 x 1.5V with BEPS / typical 100 hours	
Auto-off	After 8 min.	
Dimensions	163 x 40 x 26 mm (6.4 x 1.6 x 1.0")	
Weight	85 g (3.0 oz)	

APPENDIX I:

SWAMP DATA GUIDANCE MANUALS

SWAMP
Surface Water Ambient Monitoring Program

SWAMP DATA MANAGEMENT PLAN

Chemistry Template

Surface Water Ambient Monitoring Program

April 13, 2009

CALIFORNIA Water Boards

www.waterboards.ca.gov/swamp

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A. Analytical Data

The typical method for submitting Laboratory Analytical Data to the SWAMP Database is electronically through the online data checker. This section will focus on the tools and steps involved in submitting these data.

The following information should be used in conjunction with the SWAMP QAPrP for complete compliance with the SWAMP program including measurement quality objectives and required quality assurance (QA) samples.

1. Analysis Authorization Forms

2. Water Quality (Chemistry and Bacteria) Data

The data for sediment chemistry, water column chemistry, and bacteria analyses are treated in a similar way in the SWAMP Database and for this reason will be treated together in this portion of the document.

a. Chemistry Data Template

The Chemistry Data Template is available online at http://swamp.mpsl.mlml.calstate.edu/resources-and-downloads/database-management-systems/swamp-25-database/templates-25/#Chem.

b. Formatting the Data

A Microsoft Excel template exists for laboratories to use to format the laboratory data in a manner that can be easily loaded into the SWAMP database. As previously discussed, for many labs working under the SWAMP master contract, the station collection information can be obtained from the Analysis Authorization form. This section will discuss each of the fields in the template and how to populate them.

i. Chemistry Results Worksheet

There are two worksheets that must travel with the data for the data package to be considered complete. The first holds all chemistry or bacteria results, including QA data, and should be named **Results** in a worksheet tab. Each record in this sheet represents a result from a specific analysis for a particular parameter at a single station or for a single QA sample. This worksheet will also contain all supporting QA sample results. If beginning with an Analysis Authorization form, the following fields will likely be pre-populated in the **Data** worksheet: *EventCode*, *ProtocolCode*, *StationCode*, *LocationCode*, *CollectionMethodCode*, *SampleTypeCode*, *Replicate*, *CollectionDepth*, *UnitCollectionDepth*, *ProjectCode*, and *AgencyCode*. Please note that all fields are required to have data except when otherwise noted below. Examples of special types of samples are listed in the Special Circumstances section.

All valid LookUp list values are available at http://ftp.mpsl.mlml.calstate.edu/LookUpLists.php. At any time, if the necessary field does not appear in the LookUp list, the SWAMP Data Management Team (DMT) must be contacted to have it added.

Template Field Name	LookUp List	Description & Business Rules
LabSampleID		The LabSampleID is a recommended field intended to provide lab specific identification for an analyzed sample. **BR:** The format and content is determined by the lab. It is preferable to add -Dup, -MS, -MSD to the end of the ID to help confirm the SampleType and the LabSampleID of the native sample.
StationCode	StationLookUp	StationCode represents a unique sampling site in a sampling design. A single waterbody may have multiple stations. **BR:** The format for the unique alphanumeric description of the station is ###ABC123, where ### is the Hydrologic Unit number and ABC123 is an alphanumeric description of the Station. An example is 111EELBRN which is Hydrologic Unit 111 and an abbreviated code to indicate “Eel River - South Fork near Branscomb”.

Template Field Name	LookUp List	Description & Business Rules
EventCode	EventLookUp	EventCode represents the initial intent of the sampling event at a particular station. **BR:** The EventCode will be in a hierarchical order as follows: BA – If the initial intent of sampling is for Bioassessment (Tissue and/or WaterQuality samples may or may not also be collected) TI – If the initial intent of sampling is for Tissue (WaterQuality samples may or may not also be collected; no associated Bioassessment samples collected) WQ – If the initial intent of sampling is for WaterQuality (no associated Bioassessment or Tissue samples collected) For example, if the initial intent of sampling on Day 1 was for Tissue and WaterQuality, the EventCode would be TI. If for some reason the WaterQuality had to be re-sampled the next day, on Day 2, the event for the re-sampling would still be TI because Tissue was the initial intent of sampling on Day 1 even though the WaterQuality was sampled on Day 2.
ProtocolCode	ProtocolLookUp	ProtocolCode represents the sampling protocol used, which includes the set of methods, methodology and/or specifications, such as MPSL-DFG_Field_v1.0. Established protocols may be used or Regions may document their own sampling protocols. **BR:** It is preferable to combine protocols per StationCode and date so that all WaterQuality, Bioassessment and Tissue data are combined under the same EventCode. For example, if Tissue and WaterQuality are sampled at a station, the EventCode would be TI. If the protocols are different for Tissue and WaterQuality, the Tissue protocol would be used and the WaterQuality protocol would be listed in the SampleComments. If that is not preferable, separate EventCodes may be used with each individual protocol. Default value is Not Recorded.

Template Field Name	LookUp List	Description & Business Rules
LocationCode (previously SampleLocation)	LocationLookUp	LocationCode describes the physical location in the waterbody where the sample was collected. One sampling event may have a single or multiple locations. **BR:** For a single point of sampling, the physical location in the waterbody can be used such as Bank, Thalweg, Midchannel, OpenWater. The LocationCode for field results should be the same as the location for the WaterQuality collection method. For TI EventType sampling, the physical location plus the CollectionMethod is used such as BankNet1, BankShock1, OpenWaterTrawl1, OpenWaterNet1. For resident mussel or clam collections, the LocationCode would be the physical location in the water body plus the generic CollectionMethod, e.g. BankTissue_Grab. OpenWater sampling with multiple sub-locations within a single water body or station could have locations of OpenWaterTrawl1, OpenWaterTrawl2 describing one large location with two smaller areas of sampling within the OpenWater Location. Multiple physical locations within a single station could consist of a LocationCode such as BankShock1, BankNet1, OpenWaterHook1. If recording specific locations within a station are necessary for the project, a LocationCode such as Location1Net1, Location1Net2, Location2Shock1 may be used.
SampleDate		SampleDate refers to the date the sample was collected in the field. **BR:** The format for date in the templates is dd/mmm/yyyy, such as 10/Nov/2007. For samples with collection times that last longer than one day, like autosamplers, the sample date is the date sampling began. When entering data using the forms, the format is mm/dd/yy. For transplanted bivalves, the SampleDate is the date when the bivalves were deployed in the field.

Template Field Name	LookUp List	Description & Business Rules
CollectionTime (previously SampleTime)		CollectionTime refers to the time when the first sample of a sampling event at a specific station was collected in the field. **BR:** If the sampling crew collects 18 bottles at a single station, the CollectionTime for each would be the time of the first bottle collected. By doing so, the samples can easily be linked and any holding time issues will be consistent, and conservative, for the laboratory work. The CollectionTime format should be expressed as xx:xx in 24 hour time, such as 13:30 for 1:30 pm.
CollectionMethodCode	CollectionMethodLookUp	CollectionMethodCode refers to the general method of collection such as Sed_Grab, Sed_Core, Water_Grab, Autosampler24h, Autosampler7d. **BR:** The SWAMP water default is Water_Grab and the sediment default is Sed_Grab.
SampleTypeCode	SampleTypeLookUp	SampleTypeCode refers to the type of sample collected or analyzed. **BR:** Some commonly used SampleTypeCode choices include Grab, Integrated, MS1, CRM, LCS, LabBlank, CNEG, Composite.
Replicate (previously SampleReplicate)		The Replicate number is used to distinguish between replicates created at a single collection in the field. **BR:** The default is 1. Field Duplicates will be identified by a Replicate of 2. Field Blind Duplicates will be identified with a different SampleTypeCode of FieldBLDup, not a Collection Replicate, because they are collected blind. Laboratory replicates will be identified by a replicate of 2 in the LabReplicate field, not a Collection Replicate. When field duplicates are collected, the samples are given a StationCode of FIELDQA and a SampleTypeCode of Grab or Integrated with a Replicate of 2. When the chemistry data are loaded into the database, the StationCode FIELDQA will be given the correct StationCode of the native sample and the Replicate will be used to distinguish the duplicate sample from the native sample. EventCode, ProtocolCode, LocationCode, SampleDate, CollectionTime, CollectionMethodCode, CollectionDepth, UnitCollectionDepth, ProjectCode and AgencyCode should be identical for both samples taken.

Template Field Name	LookUp List	Description & Business Rules
CollectionDepth (previously DepthSampleCollection)		CollectionDepth records the level, from the surface in the water or sediment column, at which the sample was collected. **BR:** This information should be listed on the Chain of Custody (COC) document that accompanies the samples from the field. CollectionDepth for water samples would be measured from the water surface and recorded in meters (m) while depth collected for sediment would be measured from the sediment surface and recorded in centimeters (cm). Since depths for ambient monitoring Grab samples are generally "subsurface", defaults have been established to indicate this. For water samples the default value is 0.1 m and for sediment samples the default value is 2 cm. For Integrated samples collected from the same depth at different points across a waterbody or for samples collected at multiple times, i.e. an autosampler, the actual sample depth should be recorded. This applies to both water and sediment samples. Integrated samples collected at multiple depths, i.e. samples integrated from the water column or sediment cores, should receive a depth of -88 and the actual depths of collection should be recorded in the CollectionComments field.
UnitCollectionDepth (previously DepthUnit)	VariableCodesLookUp	UnitCollectionDepth refers to the units used in the CollectionDepth including cm (centimeters) and m (meters). This information should be listed on the Chain of Custody (COC) document that accompanies the samples from the field.
ProjectCode (previously ParentProjectID)	ProjectLookUp	ProjectCode references the project that is associated with the sample. **BR:** The formatting for a ProjectCode is as follows Program_Project. For example, the format for a SWAMP Regional Board project is SWAMP_RBX where SWAMP is the program, RB indicates it is a Regional Board sample and X is the Regional Board number. The format for a SWAMP State Board project is SWAMP_SB_XXX where SWAMP is the program, SB indicates it is a State Board sample and XXX represents the State Board project. For example, the ProjectCode for the Bioaccumulation Oversight Group would be SWAMP_SB_BOG. For a project other than SWAMP such as the Friends of the Los Angeles River, the project is FLAR_XXX where FLAR indicates it is a Friends of the Los Angeles River program and XXX represents the Friends of the Los Angeles River project. In some cases, there may not be a specific project but only a program. In this case the ProjectCode would be just the program; e.g. UMD.

Template Field Name	LookUp List	Description & Business Rules
AgencyCode	AgencyLookUp	AgencyCode refers to the organization or agency that collected the sample. This should be listed on the Chain of Custody (COC) document that accompanies the samples from the field. **BR:** If an environmental sample is used to perform laboratory QC, i.e. a matrix spike or lab duplicate, the AgencyCode still refers to the agency that collected the native sample, not the agency that created the QC sample. See 'Special Circumstances' for LABQA business rules.
CollectionComments (Not Required)		CollectionComments records any comments relating to the collection of the field sample for laboratory analysis. **BR:** This field can also be used for laboratory QC samples, e.g. Matrix Spike was performed on a FieldBlank.
SampleID (Not Required)		SampleID is a unique identifier supplied by the organization directing the sampling or sampling agency and is used to track the sample throughout the sampling and analysis processes. This field can be used to tie a result to the sample. **BR:** This SampleID, which is different from the StationCode, will likely be on the sample container the laboratory receives from the field crew or on the COC. If there is no number, leave this field blank.
PreparationPreservation (previously Preparation)	PrepPreservationLookUp	PreparationPreservation references the preparation or preservation method performed on the samples prior to analysis. **BR:** If no preparation or preservation method was performed the default value is None.
PreparationPreservationDate		PreparationPreservationDate is the date and time the preparation or preservation was started. **BR:** The format is dd/mmm/yyyy hh:mm. If there is no preparation or preservation method performed (None) then the PreparationPreservationDate should be listed as 01/Jan/1950 00:00 (the SWAMP default date/time value for none).

Template Field Name	LookUp List	Description & Business Rules
DigestExtractMethod	DigestExtractLookUp	DigestExtractMethod references the digestion or extraction method performed on the sample prior to analysis. **BR:** If no digestion or extraction method was performed the default value is None.
DigestExtractDate		DigestExtractDate is the date and time the digestion or extraction was started. **BR:** The format is dd/mmm/yyyy hh:mm. If there is no digestion or extraction performed on the sample (None) then the DigestExtractDate should be listed as 01/Jan/1950 00:00 (the default value for none).
LabBatch		The LabBatch is a unique code, provided by the laboratory, that represents a group of samples processed together. It groups all environmental samples with their supporting QC samples and will be used to verify completeness based on the SWAMP QAPrP. **BR:** The LabBatch is assigned to and identifies all samples digested or extracted together in one batch. When a digestion or extraction is not performed as part of the method, the LabBatch represents all samples within a unique analysis run. Please see the File and Batch Naming Convention to correctly format the LabBatch name. All lab batches listed in the Results worksheet need to be listed one time only in the LabBatch worksheet.
AnalysisDate		AnalysisDate is the date and time the sample was processed on the analytical instrument. **BR:** This date/time should be expressed as dd/mmm/yyyy hh:mm.
LabReplicate		The LabReplicate number is used to distinguish between replicates created in the laboratory. It differentiates the original field sample that was analyzed from all subsequent laboratory duplicates. **BR:** The default is 1 for the first sample and increases by one for each successive replicate analyzed in the laboratory.

Template Field Name	LookUp List	Description & Business Rules
MatrixName	MatrixLookUp	MatrixName refers to the sample matrix. **BR:** Water - For field-generated water samples, the MatrixName is samplewater. For lab-generated QC samples, the matrix should be the type of water that was used for the analysis of the sample, either labwater or blankwater. Labwater is water coming either directly from the tap in the laboratory or purchased spring water. Blankwater is laboratory Type I or Type II water, purchased reagent water or water that is run through a filtration process in a laboratory, such as Deionized (DI) or Milli-Q (MQ) water. Sediment - For field-generated sediment samples, the MatrixName is sediment. For lab-generated QC samples, blankmatrix could be used as the MatrixName which is a matrix used to identify a commercial- or lab-produced medium in tissue or sediment QC samples. If this is not the case then the MatrixName for lab-generated QC samples would be sediment which would include samples where water, solvent or nothing was used as a matrix. Tissue - For field-generated tissue samples, the MatrixName is tissue. For lab-generated QC samples, blankmatrix could be used as the MatrixName which is a matrix used to identify a commercial- or lab-produced medium in tissue or sediment QC samples. If this is not the case then the MatrixName for lab-generated QC samples would be tissue which would include samples where water, solvent or nothing was used as a matrix.
MethodName	MethodLookUp	MethodName refers to the analysis method used by the laboratory to analyze the sample. **BR:** Methods are expressed with a MethodName such as SM 4500-NH3 C or EPA 600/R-99-064 and must be fully described in the Method Lookup list and in the laboratory records. If a laboratory has modified an EPA or Standard Method, the laboratory agency needs to add "M" to end of the MethodName. In such situations, the modification should be documented and communicated to the SWAMP DMT for notation in the database. For instance, a lab would report a modified EPA 600/R-99-064 as EPA 600/R-99-064M accompanied by a description of the modification made. Any method for the SWAMP Project which is not in the current SWAMP database lookup list must be approved by the SWAMP QA Team prior to being added to the database.

Template Field Name	LookUp List	Description & Business Rules
AnalyteName	AnalyteLookUp	The AnalyteName is the name of the analyte or parameter for which the analysis is conducted and result is reported. The lookup list includes the acceptable abbreviation or name of the variable used by the database, enabling consistency across reporting.
FractionName	FractionLookUp	FractionName is a specific descriptor of the Analyte. **BR:** For instance, metals are often expressed as Total or Dissolved, each of which would be expressed as the Fraction, distinguishing the appropriate Analyte. If there is no need for further description of the analyte, type 'None' in this field.
Unit	UnitLookUp	Unit refers to how the chemistry result is measured or expressed. **BR:** Each combination of Analyte and Matrix requires that a specific Unit be used in the SWAMP database to ensure comparability across data. This listing can be found in the SWAMP QAPrP, online at http://www.swrcb.ca.gov/swamp/qapp.html. ??? Water units are indicated by weight/volume, i.e. ng/L. Sediment and tissue units are indicated by weight/weight and includes whether the sample result is reported as wet weight (ww) or dry weight (dw). For example, ng/g ww for ng/g wet weight. Surrogates recovery results will use a unit of %.

Template Field Name	LookUp List	Description & Business Rules
DilFactor		DilutionFactor is the factor by which a sample was diluted and is reported as a whole number. It is equal to the final volume divided by the initial volume of solution, or DF = Vf ÷ Vi. ??? **BR:** Default value is 1. A dilution other than 1 is recorded when a sample requires an additional dilution to fit into the standard curve of the instrument. It does not apply to dilutions that are standard in the method being used for analysis. Final reported results must be corrected for dilution that was carried out during the process of analysis. 1 part sample plus 9 parts blank is a DilutionFactor of 10. A 50% dilution is equivalent to a DilutionFactor of 2. When a sample requires a dilution, the MDL and RL must also be raised by that DilutionFactor. A QACode of D is required when a dilution is performed on the sample.
Result		Final numeric result of a given analyte, stored as text to retain trailing zeros **BR:** The chemistry Result is expressed as a real number rather than a calculation. The result should be reported with appropriate number of significant figures. A result of 3.7266945 with 3 significant figures should be reported as 3.73. A result of 1.350 with 4 significant figures must display 1.350 in the Excel file. If you only see 1.35, that is the result that will be loaded to the database and the 4th significant figure will be dropped.

Template Field Name	LookUp List	Description & Business Rules
ResultQualCode (previously ResQualCode) (Not Required)	ResQualLookUp	The Result Qualifier Code or ResultQualCode qualifies the analytical result of the sample. **BR:** This field may be left blank for results that are considered detected. The database will be populated with an equal sign (=) when the data are loaded. When a result is Not Detected (ND) or Detected Not Quantified (DNQ) a ResultQualCode is required and the appropriate code would be applied. When the result is -88, a ResultQualCode is required. If the ResultQualCode value is NR for Not Recorded, then a reason for this code must be written into the LabResultComments field and/or an appropriate QACode would be applied.
MDL		Method Detection Limit (MDL) is the minimum concentration of an analyte that undergoes the entire measurement process and can be reported with a stated level of confidence that the analyte concentration is greater than zero. It is the detection limit associated with the method used to analyze the analyte, or parameter, in the sample. **BR:** If no MDL is used, enter -88. A value other than -88 must be used for either the MDL or the RL.
RL		Reporting Limit (RL) is the minimum value below which data are documented as nonquantifiable. It is the reporting limit for the sample analyzed, as determined by the laboratory. **BR:** If no RL is used, enter -88. A value other than -88 must be used for either the RL or the MDL.
QACode	QALookUp	QACode is applied to the result to describe any special conditions, situations or outliers that occurred during or prior to the analysis to achieve the result. **BR:** The default code, indicating no special conditions, is None. If more than one code should be applied to a record, the convention is to list them in alphabetical order separated by commas and no spaces; i.e. GB,SC

Template Field Name	LookUp List	Description & Business Rules
ExpectedValue (Not Required)		The ExpectedValue is the concentration of the analyte in a reference standard, laboratory control sample or matrix spike sample or the value expected to obtain from analysis of the QC Sample. This consists of the native sample result concentration plus the spike amount. **BR:** For surrogate samples, the expected value should be 100, representing 100%. This field is required for SampleTypeCodes of MS1, MS2, CRM and LCS.
LabResultComments (Not Required)		The LabResultComments field holds any comments related to the lab result or analysis of the sample. **BR:** These could be comments needed to clarify any portion of the analysis or a comment that is not accommodated by any other field, e.g. Percent Recovery or Relative Percent Difference. For reference standards, laboratory control samples and matrix spikes, it is recommended to include the Percent Recovery as PR xx and the Relative Percent Difference as RPD xx. When used in combination, as in Matrix Spike Duplicate samples, the convention would be PR xx, RPD xx.

ii. Chemistry LabBatch Worksheet

The second worksheet to travel with the data holds information specific to the laboratory batch in which data is analyzed. This worksheet should be named **LabBatch** (with no spaces) in a worksheet tab. The fields in this sheet are and should be completed as follows:

Template Field Name	LookUp List	Description & Business Rules
LabBatch		The LabBatch is a unique code, provided by the laboratory, that represents a group of samples processed together. It groups all environmental samples with their supporting QC samples and will be used to verify completeness based on the SWAMP QAPrP. **BR:** The LabBatch is assigned to and identifies all samples digested or extracted together in one batch. When a digestion or extraction is not performed as part of the method, the LabBatch represents all samples within a unique analysis run. Please see the File and Batch Naming Convention to correctly format the LabBatch name. All lab batches listed in the Results worksheet need to be listed one time only in the LabBatch worksheet.
LabAgencyCode	AgencyLookUp	LabAgencyCode refers to the organization, agency or laboratory that performed the analysis on the sample.
LabSubmissionCode (previously BatchQualifierCode)	LabSubmissionLookUp	The LabSubmissionCode is a unique batch qualifier code assigned to the LabBatch as a whole by the analyzing laboratory which references the quality of the data in the LabBatch. **BR:** If the LabSubmissionCode of A is used, meaning Acceptable, the laboratory is ensuring that all SWAMP QAQC protocols were met for the lab batch. If anything other than A is used, a LabBatchComment is required.
SubmittingAgencyCode	AgencyLookUp	SubmittingAgencyCode is the organization or agency that is responsible for submission of the data to the database. This agency may be different from LabAgencyCode if the analytical data were subcontracted to another agency.
LabBatchComm (Not Required)		LabBatchComments records any comments relating to the LabBatch as a whole. **BR:** If the LabSubmissionCode is anything other than "A", a LabBatchComment is required.

c. Special Circumstances

There are several types of special circumstances discussed in this section. One type includes samples that are generated or created by the laboratory (LABQA). Another type includes environmental samples that are modified by the laboratory. A third type includes samples that are created by the sampling agency that are not environmental samples and may or may not have been created in the field (FIELDQA).

For a list of QA sample types required for each type of chemical analysis, please see the SWAMP QAPrP.

i. Laboratory-generated QA samples (LABQA)

All samples generated from within the laboratory, such as LabBlank, LCS, CRM, etc. have specific business rules, which are as follows:

LabSampleID	Recommended - provide lab specific identification for an analyzed sample
StationCode	LABQA
EventCode	WQ for water and sediment chemistry
ProtocolCode	Not Applicable
LocationCode	Not Applicable
SampleDate	Date sample was digested/extracted, expressed as dd/mmm/yyyy. When no digestion/extraction was performed, *SampleDate* is equal to the analysis date.
CollectionTime	0:00 There are situations within a batch when two identical sample types are used for QA reasons and the only way to differentiate between them is to give them each a different *CollectionTime*. For example, when more than one LabBlank, CRM, or LCS is digested, extracted, or analyzed in the same batch on the same day but are not replicates of each other, one *CollectionTime* should be 0:00 and the other 0:15, increasing the time by 15 minutes for each additional sample.
CollectionMethodCode	Not Applicable
SampleTypeCode	Select from SampleTypeLookUp List
Replicate	1
CollectionDepth	-88
UnitCollectionDepth	m for water or cm for sediment
ProjectCode	Not Applicable
AgencyCode	Organization or agency that analyzed the sample
Matrix	Water samples - labwater or blankwater Sediment samples - blankmatrix (commercially generated product) or sediment (if laboratory is using solvent, water, or nothing)

ii. Laboratory-modified QA samples

There are several types of samples discussed in this section that are generated or modified within the laboratory. The first is a Matrix Spike, which is a modified or analyte-spiked field sample. The second is a laboratory-generated duplicate of a field sample. At times, laboratories use samples not generated through the SWAMP program to satisfy SWAMP batch QA requirements. This third type is a Non-Project sample.

(a) Matrix Spike and Laboratory Duplicate Samples

For these samples, all fields describing the sample (*StationCode*, *EventCode*, *ProtocolCode*, *LocationCode*, *SampleDate*, *CollectionTime*, *CollectionMethodCode*, *CollectionDepth*, *UnitCollectionDepth*, *ProjectCode*, *AgencyCode*) remain the same as the native sample. For Matrix Spike samples, the only fields that are different than the native field sample is the *SampleTypeCode*, which should be MS1, MS2, or MSBLDup, and potentially the *Replicate*. For laboratory-generated Duplicate samples, the only field that is different than the native field sample is the *LabReplicate*.

LabSampleID	Recommended - provide lab specific identification for an analyzed sample. It is preferable to add -Dup, -MS, -MSD to the end of the Lab ID to help confirm the *SampleTypeCode* and the *LabSampleID* of the native sample.
StationCode	Same as native field sample
EventCode	Same as native field sample
ProtocolCode	Same as native field sample
LocationCode	Same as native field sample
SampleDate	Same as native field sample
CollectionTime	Same as native field sample
CollectionMethodCode	Same as native field sample
SampleTypeCode	Same as native field sample - laboratory-generated Duplicates MS1 - Matrix Spike performed on a Grab or Integrated sample MS2 - Matrix Spike performed on a FieldDup (Grab or Integrated with a *Replicate* of 2) MSBLDup - Matrix Spike performed on a FieldBLDup There are situations when a Matrix Spike was unintentionally performed on a blank sample such as a FieldBlank, TravelBlank, EquipBlank, DIBlank or FilterBlank. A batch may include two or more of these types of native samples where the only difference between them is the *SampleTypeCode*. The only way to differentiate them is to give each a different *CollectionTime*. For example, when a batch contains both a DIBlank and an EquipBlank (both with an original time of 0:00) and a Matrix Spike was performed on the EquipBlank, one *CollectionTime* should be 0:00 and the other 0:15. Then the MS1 *CollectionTime* should correspond to the correct native sample time.
Replicate	1
CollectionDepth	Same as native field sample
UnitCollectionDepth	Same as native field sample

ProjectCode	Same as native field sample
AgencyCode	Same as native field sample
LabReplicate	2 - lab-generated Duplicates 1 - Matrix Spike 2 - Matrix Spike Duplicate
Matrix	Same as native field sample

(i) Matrix Spike Samples performed on Field Duplicates

Following is a table that describes the way to format matrix spike samples performed on field duplicates, field blind duplicates, and composite blind duplicates in both the v2.2 and v2.5 SWAMP databases. The *SampleTypeCode* MS represents an MS/MSD pair.

v2.2 SWAMP Database			**v2.5 SWAMP Database**	
SampleTypeCode	*Sample Replicate*		*SampleTypeCode*	*Replicate*
One sample - sampled or split in triplicate - blind				
Grab	1	=	Grab	1
FieldBLDup	1	=	FieldBLDup or CompBLDup	1
FieldBLTrip	1	=	FieldBLDup or CompBLDup	2
One sample - sampled or split in triplicate				
Grab	1	=	Grab	1
FieldDup	1	=	Grab	2
FieldTrip	1	=	Grab	3
One pair of MS/MSD associated to one grab				
Grab	1	=	Grab	1
MS	1	=	MS1	1
One pair of MS/MSD associated to one grab - FieldDup present				
Grab	1	=	Grab	1
FieldDup	1	=	Grab	2
MS	1	=	MS1	1
One pair of MS/MSD associated to one FieldDup				
Grab	1	=	Grab	1
FieldDup	1	=	Grab	2
MSFDup	1	=	MS2	1
One pair of MS/MSD associated to one FieldBLDup				
Grab	1	=	Grab	1
FieldBLDup	1	=	FieldBLDup or CompBLDup	1
MSFBLDup	1	=	MSBLDup	1
Two pairs of MS/MSD, one associated to the Grab and one associated to the FieldDup				
Grab	1	=	Grab	1
FieldDup	1	=	Grab	2
MS	1	=	MS1	1
MSFDup	1	=	MS2	1

(ii) Calculating Matrix Spike Percent Recovery

The reported lab *Result* is the number gathered from the instrument and is the net amount recovered from the sample including the spike concentration. For spiked samples, the *ExpectedValue* is the total concentration of the analyte in the native sample plus the spiked concentration. Matrix Spike Percent Recovery is calculated by subtracting the native result from both the MS lab *Result* and the MS *ExpectedValue*, then dividing the two by each other and multiplying by 100. To illustrate:

$$\frac{\text{MS Lab Result} - \text{Native}}{\text{MS Expected} - \text{Native}} \times 100 \quad \text{or} \quad \frac{5-1}{10-1} = \frac{4}{9} \times 100 = 44\%$$

If the sample being used for the matrix spike requires a dilution, the reported values for the MS and the native sample are the dilution corrected values, not the actual values from the instrument.

(b) Non-Project Matrix Spike and Duplicate Samples (000NONPJ)

At times, laboratories use samples not generated through the SWAMP program to satisfy SWAMP batch QA requirements. These samples have different formatting rules, as follows:

LabSampleID	Recommended - provide lab specific identification for an analyzed sample. It is preferable to add -Dup, -MS, -MSD to the end of the Lab ID to help confirm the *SampleTypeCode* and the *LabSampleID* of the native sample.
StationCode	000NONPJ
EventCode	WQ for water and sediment chemistry
ProtocolCode	Not Applicable
LocationCode	Not Recorded
SampleDate	Date sample was digested/extracted, expressed as dd/mmm/yyyy. When no digestion/extraction was performed, *SampleDate* is equal to the analysis date.
CollectionTime	0:00 There are situations within a batch when two identical sample types are used for QA reasons and the only way to differentiate between them is to give them each a different *CollectionTime*. For example, when more than one 000NONPJ sample with the same *SampleTypeCode* is digested, extracted or analyzed in the same batch on the same day but are not replicates of each other, one *CollectionTime* should be 0:00 and the other 0:15, increasing the time by 15 minutes for each additional sample.
CollectionMethodCode	Not Recorded
SampleTypeCode	Grab - lab-generated Duplicates MS1 - Matrix Spikes
Replicate	1
CollectionDepth	-88
UnitCollectionDepth	m for water or cm for sediment

ProjectCode	Not Applicable
AgencyCode	Organization or agency that analyzed the sample
SampleID	The *LabSampleID* or *Source ID* can be used here as the *SampleID* as an indicator to identify the native sample
PreparationPreservation	Actual preparation or preservation performed
PreparationPreservationDate	Actual preparation or preservation date and time expressed as dd/mmm/yyyy xx:xx
LabReplicate	2 - lab-generated Duplicates 1 - Matrix Spike 2 - Matrix Spike Duplicate
Matrix	samplewater or sediment
QACode	QAX, when the native sample is not included in the batch reported

iii. Field-generated QA samples (FIELDQA)

There are two types of samples discussed in this section that are generated in the field. The first is when a field-generated QA sample is created at a specific station and that station information is important to record. The second is when a field-generated QA sample is created for a sampling trip or if the station is not recorded.

(c) Station Specific

For analyses that require an EquipBlank, FieldBlank, or FilterBlank to accompany a sampling event and it is important to record the station information, the data is entered into the SWAMP database in the same manner as the samples in the same group. The specifics are as follows:

LabSampleID	Recommended - provide lab specific identification for an analyzed sample
EventCode	WQ for water and sediment chemistry
ProtocolCode	Protocol associated with the sample or Not Recorded
StationCode	Station where sample was created
LocationCode	Location where sample was created
SampleDate	Date sample was created
CollectionTime	Time sample was created or 00:00 There are situations within a batch when two identical sample types are used for QA reasons and the only way to differentiate between them is to give them each a different *CollectionTime*. For example, when more than one EquipBlank, FieldBlank, or FilterBlank is created on the same day but are not replicates of each other, one *CollectionTime* should be 00:00 and the other 00:15, increasing the time by 15 minutes for each additional sample.
CollectionMethodCode	Not Applicable
SampleTypeCode	EquipBlank, FieldBlank, or FilterBlank

Replicate	1
CollectionDepth	-88
UnitCollectionDepth	m for water or cm for sediment
ProjectCode	Project associated with the sample
AgencyCode	Organization or agency that created the sample
CollectionComment	EquipBlank - comment includes type of equipment cleaned and location (lab or field)
Matrix	labwater or blankwater

(d) Not Station-Specific (FIELDQA)

For analyses that require an EquipBlank, FieldBlank, FilterBlank, TravelBlank or DIBlank to accompany a sampling event and it is not important to record the station information, the data is entered into the SWAMP database in the same manner as the samples in the same group. The specifics are as follows:

LabSampleID	Recommended - provide lab specific identification for an analyzed sample
EventCode	WQ for water and sediment chemistry
ProtocolCode	Protocol used or Not Recorded
StationCode	FIELDQA
LocationCode	Not Applicable
SampleDate	Date sample was created TravelBlank should be entered as the date the TravelBlank becomes part of the sample group (i.e., leaves the lab for the sampling event).
CollectionTime	Time sample was created or 0:00 There are situations within a batch when two identical sample types are used for QA reasons and the only way to differentiate between them is to give them each a different *CollectionTime*. For example, when more than one EquipBlank, FieldBlank, FilterBlank, TravelBlank, or DIBlank is created on the same day but are not replicates of each other, one *CollectionTime* should be 00:00 and the other 00:15, increasing the time by 15 minutes for each additional sample.
CollectionMethodCode	Not Applicable
SampleTypeCode	EquipBlank, FieldBlank, FilterBlank, TravelBlank, or DIBlank
Replicate	1
CollectionDepth	-88
UnitCollectionDepth	m for water or cm for sediment

ProjectCode	Project associated with the sample
AgencyCode	Organization or agency that created the sample
CollectionComment	EquipBlank - comment includes type of equipment cleaned and location (lab or field)
Matrix	labwater or blankwater

iv. Other Types of Samples

There are two types of samples discussed in this section that have special circumstances. The first is a pore water matrix sample and the second is a sample where bacteria is analyzed.

(e) Interstitial Water (Pore Water)Analysis

Certain sampling events create a special set of rules that apply for a few of the entry fields in the SWAMP database. One of these would be the collection of sediment from which interstitial water is extracted and then analyzed. Below are the fields that differentiate these samples from the norm and how they should be completed.

CollectionMethodCode	Sed_Grab
SampleTypeCode	Integrated
UnitCollectionDepth	cm
PreparationPreservation	Centrifuged plus any additional preparation done at the lab (Centrifuged, X)
PreparationPreservationDate	If no preparation was performed at the analyzing lab, enter the centrifuge date for *PreparationPreservationDate*. If a preparation or preservation was performed at the analyzing laboratory, enter the preparation date and time and include the date of centrifuge in the *CollectionComments*.
Matrix	sediment, interstitialwater
Unit	water units

(f) Bacteria Samples

Bacteria/Pathogen samples are generally recorded in the same way as chemistry samples, except as follows:

AnalysisDate	Include the analysis time in the *AnalysisDate* field as dd/mmm/yyyy xx:xx
DilFactor	Dilution factor should be recorded
ResultQualCode	This field records the <, <= and >, >= as related to the results. Please note that if the result is less than the reporting limit and the lab is confident in reporting "Non Detect", this should be entered as ND in the *ResultQualCode* with a value of -MDL in *Result* field.

SWAMP
Surface Water Ambient Monitoring Program

SWAMP DATA MANAGEMENT PLAN

Toxicity Template

Surface Water Ambient Monitoring Program

April 15, 2009

California Water Boards

www.waterboards.ca.gov/swamp

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A. Analytical Data

The typical method for submitting Laboratory Analytical Data to the SWAMP Database is electronically through the online data checker. This section will focus on the tools and steps involved in submitting these data.

The following information should be used in conjunction with the SWAMP QAPrP for complete compliance with the SWAMP program including measurement quality objectives and required quality assurance (QA) samples.

1. Analysis Authorization Forms

2. Water Quality (Chemistry and Bacteria) Data

The Chemistry Data Template Documentation is available online at http://swamp.mpsl.mlml.calstate.edu/resources-and-downloads/database-management-systems/swamp-25-database/documentation-25/#Chemistry.

3. Toxicity Data

Toxicity Data consists of Toxicity Test, Point, and Results data along with Toxicity Summary data for each effort, which is based on the replicate data. The toxicity data is submitted to the SWAMP database in three worksheets; Results, Summary, and ToxBatch.

a. Toxicity Data Template

The Toxicity Data Template is available online at http://swamp.mpsl.mlml.calstate.edu/resources-and-downloads/database-management-systems/swamp-25-database/templates-25/#Tox.

b. Formatting the Data

A Microsoft Excel template exists for laboratories to use to format the toxicity laboratory data in a manner that can be easily loaded into the SWAMP database. As previously discussed, for many labs working under the SWAMP master contract, the station collection information can be obtained from the Analysis Authorization form. This section will discuss each of the fields in the template and how to populate them.

i. Tox Results Worksheet

There are three worksheets that must travel with the data for the toxicity data package to be considered complete. The first holds all toxicity results, including quality control (QC) data, and should be named **Results** in a Microsoft Excel worksheet tab. Each record in this sheet represents a result from a specific analysis for a particular parameter at a single station or for a single QC sample. This worksheet will also contain all supporting QC sample results. If beginning with an Analysis Authorization, the following fields will likely be pre-populated in the **Data** worksheet: *EventCode*, *ProtocolCode*, *StationCode*, *LocationCode*, *CollectionMethodCode*, *SampleTypeCode*, *Replicate*, *CollectionDepth*, *UnitCollectionDepth*, *ProjectCode*, and *Agency Code*. Please note that all fields are required to be populated except when otherwise noted below. Examples of special types of samples are listed in the Special Circumstances section.

All valid LookUp list values are available at http://ftp.mpsl.mlml.calstate.edu/LookUpLists.php. At any time, if the necessary field does not appear in the LookUp list, the SWAMP Data Management Team (DMT) must be contacted to have it added.

(*Note that the fields through Dilution are identical to those in the summary worksheet*)

Template Field Name	LookUp List	Description & Business Rules
LabSampleID		The LabSampleID is a recommended field intended to provide lab specific identification for an analyzed sample. BR: The format and content is determined by the lab. It is preferable to add -Dup, -MS, -MSD to the end of the ID to help confirm the SampleType and the LabSampleID of the native sample.
StationCode	StationLookUp	StationCode represents a unique sampling site in a sampling design. A single waterbody may have multiple stations. BR: The format for the unique alphanumeric description of the station is ###ABC123, where ### is the Hydrologic Unit number and ABC123 is an alphanumeric description of the Station. An example is 111EELBRN which is Hydrologic Unit 111 and an abbreviated code to indicate "Eel River - South Fork near Branscomb".

Template Field Name	LookUp List	Description & Business Rules
EventCode	EventLookUp	EventCode represents the initial intent of the sampling event at a particular station. BR: The EventCode will be in a hierarchical order as follows: BA – If the initial intent of sampling is for Bioassessment (Tissue and/or WaterQuality samples may or may not also be collected) TI – If the initial intent of sampling is for Tissue (WaterQuality samples may or may not also be collected; no associated Bioassessment samples collected) WQ – If the initial intent of sampling is for WaterQuality (no associated Bioassessment or Tissue samples collected) For example, if the initial intent of sampling on Day 1 was for Tissue and WaterQuality, the EventCode would be TI. If for some reason the WaterQuality had to be re-sampled the next day, on Day 2, the event for the re-sampling would still be TI because Tissue was the initial intent of sampling on Day 1 even though the WaterQuality was sampled on Day 2.
ProtocolCode	ProtocolLookUp	ProtocolCode represents the sampling protocol used, which includes the set of methods, methodology and/or specifications, such as MPSL-DFG_Field_v1.0. Established protocols may be used or Regions may document their own sampling protocols. BR: It is preferable to combine protocols per StationCode and date so that all WaterQuality, Bioassessment and Tissue data are combined under the same EventCode. For example, if Tissue and WaterQuality are sampled at a station, the EventCode would be TI. If the protocols are different for Tissue and WaterQuality, the Tissue protocol would be used and the WaterQuality protocol would be listed in the SampleComments. If that is not preferable, separate EventCodes may be used with each individual protocol. Default value is Not Recorded.

Template Field Name	LookUp List	Description & Business Rules
LocationCode (previously SampleLocation)	LocationLookUp	LocationCode describes the physical location in the waterbody where the sample was collected. One sampling event may have a single or multiple locations. BR: For a single point of sampling, the physical location in the waterbody can be used such as Bank, Thalweg, Midchannel, OpenWater. The LocationCode for field results should be the same as the location for the WaterQuality collection method. For TI EventType sampling, the physical location plus the CollectionMethod is used such as BankNet1, BankShock1, OpenWaterTrawl1, OpenWaterNet1. For resident mussel or clam collections, the LocationCode would be the physical location in the water body plus the generic CollectionMethod, e.g. BankTissue_Grab. OpenWater sampling with multiple sub-locations within a single water body or station could have locations of OpenWaterTrawl1, OpenWaterTrawl2 describing one large location with two smaller areas of sampling within the OpenWater Location. Multiple physical locations within a single station could consist of a LocationCode such as BankShock1, BankNet1, OpenWaterHook1. If recording specific locations within a station are necessary for the project, a LocationCode such as Location1Net1, Location1Net2, Location2Shock1 may be used.
SampleDate		SampleDate refers to the date the sample was collected in the field. BR: The format for date in the templates is dd/mmm/yyyy, such as 10/Nov/2007. For samples with collection times that last longer than one day, like autosamplers, the sample date is the date sampling began. When entering data using the forms, the format is mm/dd/yy. For transplanted bivalves, the SampleDate is the date when the bivalves were deployed in the field.

Template Field Name	LookUp List	Description & Business Rules
CollectionTime (previously SampleTime)		CollectionTime refers to the time when the first sample of a sampling event at a specific station was collected in the field. BR: If the sampling crew collects 18 bottles at a single station, the CollectionTime for each would be the time of the first bottle collected. By doing so, the samples can easily be linked and any holding time issues will be consistent, and conservative, for the laboratory work. The CollectionTime format should be expressed as xx:xx in 24 hour time, such as 13:30 for 1:30 pm.
CollectionMethodCode	CollectionMethodLookUp	CollectionMethodCode refers to the general method of collection such as Sed_Grab, Sed_Core, Water_Grab, Autosampler24h, Autosampler7d. BR: The SWAMP water default is Water_Grab and the sediment default is Sed_Grab.
SampleTypeCode	SampleTypeLookUp	SampleTypeCode refers to the type of sample collected or analyzed. BR: Some commonly used SampleTypeCode choices include Grab, Integrated, MS1, CRM, LCS, LabBlank, CNEG, Composite.
Replicate (previously SampleReplicate)		The Replicate number is used to distinguish between replicates created at a single collection in the field. BR: The default is 1. Field Duplicates will be identified by a Replicate of 2. Field Blind Duplicates will be identified with a different SampleTypeCode of FieldBLDup, not a collection Replicate, because they are collected blind. Laboratory replicates will be identified by a replicate of 2 in the LabReplicate field, not a collection Replicate. When field duplicates are collected, the samples are given a StationCode of FIELDQA and a SampleTypeCode of Grab or Integrated with a Replicate of 2. When the chemistry data are loaded into the database, the StationCode FIELDQA will be given the correct StationCode of the native sample and the Replicate will be used to distinguish the duplicate sample from the native sample. EventCode, ProtocolCode, LocationCode, SampleDate, CollectionTime, CollectionMethodCode, CollectionDepth, UnitCollectionDepth, ProjectCode and AgencyCode should be identical for both samples taken.

Template Field Name	LookUp List	Description & Business Rules
CollectionDepth (previously DepthSampleCollection)		CollectionDepth records the level, from the surface in the water or sediment column, at which the sample was collected. BR: This information should be listed on the Chain of Custody (COC) document that accompanies the samples from the field. CollectionDepth for water samples would be measured from the water surface and recorded in meters (m) while depth collected for sediment would be measured from the sediment surface and recorded in centimeters (cm). Since depths for ambient monitoring Grab samples are generally "subsurface", defaults have been established to indicate this. For water samples the default value is 0.1 m and for sediment samples the default value is 2 cm. For Integrated samples collected from the same depth at different points across a waterbody or for samples collected at multiple times, i.e. an autosampler, the actual sample depth should be recorded. This applies to both water and sediment samples. Integrated samples collected at multiple depths, i.e. samples integrated from the water column or sediment cores, should receive a depth of -88 and the actual depths of collection should be recorded in the CollectionComments field.
UnitCollectionDepth (previously DepthUnit)	VariableCodesLookUp	UnitCollectionDepth refers to the units used in the CollectionDepth including cm (centimeters) and m (meters). This information should be listed on the Chain of Custody (COC) document that accompanies the samples from the field.

Template Field Name	LookUp List	Description & Business Rules
ProjectCode (previously ParentProjectID)	ProjectLookUp	ProjectCode references the project that is associated with the sample. BR: The formatting for a ProjectCode is as follows Program_Project. For example, the format for a SWAMP Regional Board project is SWAMP_RBX where SWAMP is the program, RB indicates it is a Regional Board sample and X is the Regional Board number. The format for a SWAMP State Board project is SWAMP_SB_XXX where SWAMP is the program, SB indicates it is a State Board sample and XXX represents the State Board project. For example, the ProjectCode for the Bioaccumulation Oversight Group would be SWAMP_SB_BOG. For a project other than SWAMP such as the Friends of the Los Angeles River, the project is FLAR_XXX where FLAR indicates it is a Friends of the Los Angeles River program and XXX represents the Friends of the Los Angeles River project. In some cases, there may not be a specific project but only a program. In this case the ProjectCode would be just the program; e.g. UMD.
AgencyCode	AgencyLookUp	AgencyCode refers to the organization or agency that collected the sample. This should be listed on the Chain of Custody (COC) document that accompanies the samples from the field. BR: If an environmental sample is used to perform laboratory QC, i.e. a matrix spike or lab duplicate, the AgencyCode still refers to the agency that collected the native sample, not the agency that created the QC sample. See 'Special Circumstances' for LABQA business rules.
CollectionComments (previously SampleComments) (Not Required)		CollectionComments records any comments relating to the collection of the field sample for laboratory analysis. BR: This field can also be used for laboratory QC samples, e.g. Matrix Spike was performed on a FieldBlank.

Template Field Name	LookUp List	Description & Business Rules
SampleID (Not Required)		SampleID is a unique identifier supplied by the organization directing the sampling or sampling agency and is used to track the sample throughout the sampling and analysis processes. This field can be used to tie a result to the sample. BR: This SampleID, which is different from the StationCode, will likely be on the sample container the laboratory receives from the field crew or on the COC. If there is no number, leave this field blank.
MatrixName	MatrixLookUp	MatrixName refers to the sample matrix. BR: Water - For field-generated water samples, the MatrixName is samplewater. For lab-generated QC samples, the matrix should be the type of water that was used for the analysis of the sample, either labwater or blankwater. Labwater is water coming either directly from the tap in the laboratory or purchased spring water. Blankwater is laboratory Type I or Type II water, purchased reagent water or water that is run through a filtration process in a laboratory, such as Deionized (DI) or Milli-Q (MQ) water. Sediment - For field-generated sediment samples, the MatrixName is sediment. For lab-generated QC samples, blankmatrix could be used as the MatrixName which is a matrix used to identify a commercial- or lab-produced medium in tissue or sediment QC samples. If this is not the case then the MatrixName for lab-generated QC samples would be sediment which would include samples where water, solvent or nothing was used as a matrix. Tissue - For field-generated tissue samples, the MatrixName is tissue. For lab-generated QC samples, blankmatrix could be used as the MatrixName which is a matrix used to identify a commercial- or lab-produced medium in tissue or sediment QC samples. If this is not the case then the MatrixName for lab-generated QC samples would be tissue which would include samples where water, solvent or nothing was used as a matrix.

Template Field Name	LookUp List	Description & Business Rules
MethodName	MethodLookUp	MethodName refers to the analysis method used by the laboratory to analyze the sample. BR: Methods are expressed with a MethodName such as SM 4500-NH3 C or EPA 600/R-99-064 and must be fully described in the Method Lookup list and in the laboratory records. If a laboratory has modified an EPA or Standard Method, the laboratory agency needs to add "M" to end of the MethodName. In such situations, the modification should be documented and communicated to the SWAMP DMT for notation in the database. For instance, a lab would report a modified EPA 600/R-99-064 as EPA 600/R-99-064M accompanied by a description of the modification made. Any method for the SWAMP Project which is not in the current SWAMP database lookup list must be approved by the SWAMP QA Team prior to being added to the database.
TestDuration	ToxTestDurLookUp	ToxTestDurCode indicates the duration of the toxicity test as a number and includes the associated units. BR: Some methods allow for a test to be completed early if all the necessary data has been obtained. If this is the case, the ToxTestDurCode is recorded as the duration of the test initially indicated by the method. For example, a method indicates a 7 day test is to be performed but the laboratory ends the test one day early. The ToxTestDurCode would be 7 days, not 6 days.
OrganismName (previously ToxSpeciesName)	OrganismLookup	OrganismName refers to the scientific name of the species used in the toxicity test.
ToxBatch		The ToxBatch is assigned by the laboratory and groups all environmental samples and supporting QA samples within a unique analysis batch. It is used to compare field samples with their associated NegativeControls for statistical analysis and will be used to verify completeness based on the SWAMP QAPrP. BR: The ToxBatch should be listed only one time in the ToxBatch worksheet for each unique ToxBatch found in the Results and Summary worksheets. Follow the File and Batch Name Convention to correctly identify the batch. It is recommended to include the start date and an abbreviation of the OrganismName in the lab-specific portion of the ToxBatch.

Template Field Name	LookUp List	Description & Business Rules
ToxTestComments (Not Required)		Use the ToxTestComments field to note any comments necessary to describe special circumstances for the toxicity test for the specific record.
Treatment	AnalyteLookUp	Treatment refers to any treatment performed on the sample, such as a pH adjustment. BR: The default value is None.
Concentration		Concentration refers to the adjusted final concentration or value of the analyte applied to the toxicity sample, expressed as a number. BR: The default value is 0. ??? Or the adjusted concentration level of the analyte i.e. pH or temp adjustment to 7 or 15 degrees
UnitTreatment	UnitLookUp	UnitTreatment refers to the units used in the treatment. BR: If a Treatment did not occur, the default value is none.
Dilution		Dilution is recorded as a proportion of the original sample. A sample with 80% sample and 20% blankwater has a Dilution Value of 80. BR: If no dilution is performed, the default value of 100 is used.
LabReplicate (previously ToxReplicate)		The LabReplicate identifies the individual splits of the toxicity sample and is used to identify from which replicate a result originated. BR: For toxicity replicates, the default is 1 and increases by one for each successive replicate. If a water quality measurement record is associated with a single replicate, it should have the same value as the replicate it measures. If the water quality measurements are taken at the sample level, the LabReplicate should be recorded as replicate 0.
ToxPointMethod	MethodLookUp	ToxPointMethod refers to the general method used in obtaining or calculating the result. BR: Toxicity replicate and summary data have a default value of None unless a method other than the test MethodName is used for the calculations. Water quality measurement results have a default value of ToxWQMeasurement.

Template Field Name	LookUp List	Description & Business Rules
AnalyteName (previously EndPointCode)	AnalyteLookUp	AnalyteName refers to the parameter being measured. BR: Toxicity examples include Survival, Young/female, Biomass (weight/orig indiv). Water quality measurment examples include pH, Ammonia as NH3, Salinity. The recommendation is "Biomass" be calculated for all fish species (total weight of surviving individuals divided by the original number of organisms at the start of the test) and "Growth" be calculated for Hyalella growth weight (total weight of surviving individuals divided by the number of survivors at the end of the test). Toxicity endpoints in the database will change to represent this correction; the endpoints for all fish species will be expressed as biomass instead of growth weight. It is the recommendation of the panel that we do not go back and recalculate results in the database where growth was calculated instead of biomass. All previous measurements would be listed as growth or biomass according to analysis performed by the analyzing laboratory.
FractionName	FractionLookUp	FractionName is a specific descriptor of the Analyte. BR: Ammonia as NH3 is expressed as Total or Unionized, each of which would be expressed as the Fraction, distinguishing the appropriate Analyte. If there is no need for further description of the analyte the default value is None.
WQSource	MatrixLookUp	WQSource differentiates between water quality measurements taken in the overlying water as well as in the sediment or interstitialwater. BR: Overlyingwater is used for the overlying water measurements which is the default for all water quality measurements. Interstitialwater is used for the interstitial water measurements. NA is used for for all toxicity results.

Template Field Name	LookUp List	Description & Business Rules
TimePoint	TimePointLookUp	TimePoint refers to the point in time during the test at which the measurement was recorded for water quality measurements. BR: For water quality measurements, timepoints such as initial or final are used to indicate the measurments recorded at the beginning and end of the test. High and low are used to indicate the highest and lowest values recorded for a particular sample during the entire length of the test. Day 2 is used indicate a measurement taken on the 2nd day of the test. For toxicity measurements, the TimePoint should match the TestDuration value. So if a 7 day test is performed, the TimePoint for the toxicity results would be 7 Days.
UnitAnalyte (previously ToxResultsUnit)	UnitLookUp	UnitAnalyte indicates the units used in the measurment of the AnalyteName.
Result		Numeric result of test, stored as text to retain trailing zeros BR: The toxicity Result is expressed as a real number rather than a calculation. The result should be reported with the appropriate number of significant figures. A result of 3.7266945 with 3 significant figures should be reported as 3.73. A result of 1.350 with 4 significant figures must display 1.350 in the Excel file. If you only see 1.35, that is the result that will be loaded to the database and the 4th significant figure will be dropped.
ResultQualCode (Not Required)	ResQualLookUp	The Result Qualifier Code or ResultQualCode qualifies the analytical result of the sample. BR: This field may be left blank for results that are considered detected. The database will be populated with an equal sign (=) when the data are loaded. When a result is Not Detected a ResultQualCode of ND is required. When the result is -88, a ResultQualCode is required. If the ResultQualCode value is NR for Not Recorded or NS for No Survival, then a reason for this code must be written into the ToxResultComments field.

Template Field Name	LookUp List	Description & Business Rules
ToxResultQACode (Not Required)	ToxResultQALookUp	A ToxResultQACode is used to further qualify the analytical result of the sample. BR: When a test has a secondary toxpoint where young are measured and the first toxpoint is a Male instead of a female, the ToxResultQACode for the second toxpoint is MAL for male.
ToxResultComments (Not Required)		In the ToxResultsComments field note any comments necessary to describe special circumstances for the toxicity results data for the specific record. These could be comments needed to clarify any portion of the analysis which is not described in any other field. BR: When the ResultQualCode value is NR for Not Recorded or NS for No Survival, then a reason for this code must be written into the ToxResultsComments field.

ii. Toxicity Summary Worksheet

The second worksheet to travel with the data holds information specific to toxicity summary data. This worksheet should be named **Summary** in its worksheet tab. The fields in this sheet are and should be completed as follows. Examples of special types of samples are listed in the Special Circumstances section.

(*Note that the fields through Dilution are identical to those in the Results*)

Template Field Name	LookUp List	Description & Business Rules
LabSampleID		The LabSampleID is a recommended field intended to provide lab specific identification for an analyzed sample. BR: The format and content is determined by the lab. It is preferable to add -Dup, -MS, -MSD to the end of the ID to help confirm the SampleType and the LabSampleID of the native sample.
StationCode	StationLookUp	StationCode represents a unique sampling site in a sampling design. A single waterbody may have multiple stations. BR: The format for the unique alphanumeric description of the station is ###ABC123, where ### is the Hydrologic Unit number and ABC123 is an alphanumeric description of the Station. An example is 111EELBRN which is Hydrologic Unit 111 and an abbreviated code to indicate "Eel River - South Fork near Branscomb".

Template Field Name	LookUp List	Description & Business Rules
EventCode	EventLookUp	EventCode represents the initial intent of the sampling event at a particular station. BR: The EventCode will be in a hierarchical order as follows: BA – If the initial intent of sampling is for Bioassessment (Tissue and/or WaterQuality samples may or may not also be collected) TI – If the initial intent of sampling is for Tissue (WaterQuality samples may or may not also be collected; no associated Bioassessment samples collected) WQ – If the initial intent of sampling is for WaterQuality (no associated Bioassessment or Tissue samples collected) For example, if the initial intent of sampling on Day 1 was for Tissue and WaterQuality, the EventCode would be TI. If for some reason the WaterQuality had to be re-sampled the next day, on Day 2, the event for the re-sampling would still be TI because Tissue was the initial intent of sampling on Day 1 even though the WaterQuality was sampled on Day 2.
ProtocolCode	ProtocolLookUp	ProtocolCode represents the sampling protocol used, which includes the set of methods, methodology and/or specifications, such as MPSL-DFG_Field_v1.0. Established protocols may be used or Regions may document their own sampling protocols. BR: It is preferable to combine protocols per StationCode and date so that all WaterQuality, Bioassessment and Tissue data are combined under the same EventCode. For example, if Tissue and WaterQuality are sampled at a station, the EventCode would be TI. If the protocols are different for Tissue and WaterQuality, the Tissue protocol would be used and the WaterQuality protocol would be listed in the SampleComments. If that is not preferable, separate EventCodes may be used with each individual protocol. Default value is Not Recorded.

Template Field Name	LookUp List	Description & Business Rules
LocationCode (previously SampleLocation)	LocationLookUp	LocationCode describes the physical location in the waterbody where the sample was collected. One sampling event may have a single or multiple locations. BR: For a single point of sampling, the physical location in the waterbody can be used such as Bank, Thalweg, Midchannel, OpenWater. The LocationCode for field results should be the same as the location for the WaterQuality collection method. For TI EventType sampling, the physical location plus the CollectionMethod is used such as BankNet1, BankShock1, OpenWaterTrawl1, OpenWaterNet1. For resident mussel or clam collections, the LocationCode would be the physical location in the water body plus the generic CollectionMethod, e.g. BankTissue_Grab. OpenWater sampling with multiple sub-locations within a single water body or station could have locations of OpenWaterTrawl1, OpenWaterTrawl2 describing one large location with two smaller areas of sampling within the OpenWater Location. Multiple physical locations within a single station could consist of a LocationCode such as BankShock1, BankNet1, OpenWaterHook1. If recording specific locations within a station are necessary for the project, a LocationCode such as Location1Net1, Location1Net2, Location2Shock1 may be used.
SampleDate		SampleDate refers to the date the sample was collected in the field. BR: The format for date in the templates is dd/mmm/yyyy, such as 10/Nov/2007. For samples with collection times that last longer than one day, like autosamplers, the sample date is the date sampling began. When entering data using the forms, the format is mm/dd/yy. For transplanted bivalves, the SampleDate is the date when the bivalves were deployed in the field.

Template Field Name	LookUp List	Description & Business Rules
CollectionTime (previously SampleTime)		CollectionTime refers to the time when the first sample of a sampling event at a specific station was collected in the field. BR: If the sampling crew collects 18 bottles at a single station, the CollectionTime for each would be the time of the first bottle collected. By doing so, the samples can easily be linked and any holding time issues will be consistent, and conservative, for the laboratory work. The CollectionTime format should be expressed as xx:xx in 24 hour time, such as 13:30 for 1:30 pm.
CollectionMethodCode	CollectionMethodLookUp	CollectionMethodCode refers to the general method of collection such as Sed_Grab, Sed_Core, Water_Grab, Autosampler24h, Autosampler7d. BR: The SWAMP water default is Water_Grab and the sediment default is Sed_Grab.
SampleTypeCode	SampleTypeLookUp	SampleTypeCode refers to the type of sample collected or analyzed. BR: Some commonly used SampleTypeCode choices include Grab, Integrated, MS1, CRM, LCS, LabBlank, CNEG, Composite.
Replicate (previously SampleReplicate)		The Replicate number is used to distinguish between replicates created at a single collection in the field. BR: The default is 1. Field Duplicates will be identified by a Replicate of 2. Field Blind Duplicates will be identified with a different SampleTypeCode of FieldBLDup, not a collection Replicate, because they are collected blind. Laboratory replicates will be identified by a replicate of 2 in the LabReplicate field, not a collection Replicate. When field duplicates are collected, the samples are given a StationCode of FIELDQA and a SampleTypeCode of Grab or Integrated with a Replicate of 2. When the chemistry data are loaded into the database, the StationCode FIELDQA will be given the correct StationCode of the native sample and the Replicate will be used to distinguish the duplicate sample from the native sample. EventCode, ProtocolCode, LocationCode, SampleDate, CollectionTime, CollectionMethodCode, CollectionDepth, UnitCollectionDepth, ProjectCode and AgencyCode should be identical for both samples taken.

Template Field Name	LookUp List	Description & Business Rules
CollectionDepth (previously DepthSampleCollection)		CollectionDepth records the level, from the surface in the water or sediment column, at which the sample was collected. BR: This information should be listed on the Chain of Custody (COC) document that accompanies the samples from the field. CollectionDepth for water samples would be measured from the water surface and recorded in meters (m) while depth collected for sediment would be measured from the sediment surface and recorded in centimeters (cm). Since depths for ambient monitoring Grab samples are generally "subsurface", defaults have been established to indicate this. For water samples the default value is 0.1 m and for sediment samples the default value is 2 cm. For Integrated samples collected from the same depth at different points across a waterbody or for samples collected at multiple times, i.e. an autosampler, the actual sample depth should be recorded. This applies to both water and sediment samples. Integrated samples collected at multiple depths, i.e. samples integrated from the water column or sediment cores, should receive a depth of -88 and the actual depths of collection should be recorded in the CollectionComments field.
UnitCollectionDepth (previously DepthUnit)	VariableCodesLookUp	UnitCollectionDepth refers to the units used in the CollectionDepth including cm (centimeters) and m (meters). This information should be listed on the Chain of Custody (COC) document that accompanies the samples from the field.

Template Field Name	LookUp List	Description & Business Rules
ProjectCode (previously ParentProjectID)	ProjectLookUp	ProjectCode references the project that is associated with the sample. BR: The formatting for a ProjectCode is as follows Program_Project. For example, the format for a SWAMP Regional Board project is SWAMP_RBX where SWAMP is the program, RB indicates it is a Regional Board sample and X is the Regional Board number. The format for a SWAMP State Board project is SWAMP_SB_XXX where SWAMP is the program, SB indicates it is a State Board sample and XXX represents the State Board project. For example, the ProjectCode for the Bioaccumulation Oversight Group would be SWAMP_SB_BOG. For a project other than SWAMP such as the Friends of the Los Angeles River, the project is FLAR_XXX where FLAR indicates it is a Friends of the Los Angeles River program and XXX represents the Friends of the Los Angeles River project. In some cases, there may not be a specific project but only a program. In this case the ProjectCode would be just the program; e.g. UMD.
AgencyCode	AgencyLookUp	AgencyCode refers to the organization or agency that collected the sample. This should be listed on the Chain of Custody (COC) document that accompanies the samples from the field. BR: If an environmental sample is used to perform laboratory QC, i.e. a matrix spike or lab duplicate, the AgencyCode still refers to the agency that collected the native sample, not the agency that created the QC sample. See 'Special Circumstances' for LABQA business rules.
CollectionComments (previously SampleComments) (Not Required)		CollectionComments records any comments relating to the collection of the field sample for laboratory analysis. BR: This field can also be used for laboratory QC samples, e.g. Matrix Spike was performed on a FieldBlank.

Template Field Name	LookUp List	Description & Business Rules
SampleID (Not Required)		SampleID is a unique identifier supplied by the organization directing the sampling or sampling agency and is used to track the sample throughout the sampling and analysis processes. This field can be used to tie a result to the sample. BR: This SampleID, which is different from the StationCode, will likely be on the sample container the laboratory receives from the field crew or on the COC. If there is no number, leave this field blank.
MatrixName	MatrixLookUp	MatrixName refers to the sample matrix. BR: Water - For field-generated water samples, the MatrixName is samplewater. For lab-generated QC samples, the matrix should be the type of water that was used for the analysis of the sample, either labwater or blankwater. Labwater is water coming either directly from the tap in the laboratory or purchased spring water. Blankwater is laboratory Type I or Type II water, purchased reagent water or water that is run through a filtration process in a laboratory, such as Deionized (DI) or Milli-Q (MQ) water. Sediment - For field-generated sediment samples, the MatrixName is sediment. For lab-generated QC samples, blankmatrix could be used as the MatrixName which is a matrix used to identify a commercial- or lab-produced medium in tissue or sediment QC samples. If this is not the case then the MatrixName for lab-generated QC samples would be sediment which would include samples where water, solvent or nothing was used as a matrix. Tissue - For field-generated tissue samples, the MatrixName is tissue. For lab-generated QC samples, blankmatrix could be used as the MatrixName which is a matrix used to identify a commercial- or lab-produced medium in tissue or sediment QC samples. If this is not the case then the MatrixName for lab-generated QC samples would be tissue which would include samples where water, solvent or nothing was used as a matrix.

Template Field Name	LookUp List	Description & Business Rules
MethodName	MethodLookUp	MethodName refers to the analysis method used by the laboratory to analyze the sample. BR: Methods are expressed with a MethodName such as SM 4500-NH3 C or EPA 600/R-99-064 and must be fully described in the Method Lookup list and in the laboratory records. If a laboratory has modified an EPA or Standard Method, the laboratory agency needs to add "M" to end of the MethodName. In such situations, the modification should be documented and communicated to the SWAMP DMT for notation in the database. For instance, a lab would report a modified EPA 600/R-99-064 as EPA 600/R-99-064M accompanied by a description of the modification made. Any method for the SWAMP Project which is not in the current SWAMP database lookup list must be approved by the SWAMP QA Team prior to being added to the database.
TestDuration	ToxTestDurLookUp	ToxTestDurCode indicates the duration of the toxicity test as a number and includes the associated units. BR: Some methods allow for a test to be completed early if all the necessary data has been obtained. If this is the case, the ToxTestDurCode is recorded as the duration of the test initially indicated by the method. For example, a method indicates a 7 day test is to be performed but the laboratory ends the test one day early. The ToxTestDurCode would be 7 days, not 6 days.
OrganismName (previously ToxSpeciesName)	OrganismLookup	OrganismName refers to the scientific name of the species used in the toxicity test.
ToxBatch		The ToxBatch is assigned by the laboratory and groups all environmental samples and supporting QA samples within a unique analysis batch. It is used to compare field samples with their associated NegativeControls for statistical analysis and will be used to verify completeness based on the SWAMP QAPrP. BR: The ToxBatch should be listed only one time in the ToxBatch worksheet for each unique ToxBatch found in the Results and Summary worksheets. Follow the File and Batch Name Convention to correctly identify the batch. It is recommended to include the start date and an abbreviation of the OrganismName in the lab-specific portion of the ToxBatch.

Template Field Name	LookUp List	Description & Business Rules
ToxTestComments (Not Required)		Use the ToxTestComments field to note any comments necessary to describe special circumstances for the toxicity test for the specific record.
Treatment	AnalyteLookUp	Treatment refers to any treatment performed on the sample, such as a pH adjustment. BR: The default value is None.
Concentration		Concentration refers to the adjusted final concentration or value of the analyte applied to the toxicity sample, expressed as a number. BR: The default value is 0. ??? Or the adjusted concentration level of the analyte i.e. pH or temp adjustment to 7 or 15 degrees
UnitTreatment	UnitLookUp	UnitTreatment refers to the units used in the treatment. BR: If a Treatment did not occur, the default value is none.
Dilution		Dilution is recorded as a proportion of the original sample. A sample with 80% sample and 20% blankwater has a Dilution Value of 80. BR: If no dilution is performed, the default value of 100 is used.
ToxPointMethod	MethodLookUp	ToxPointMethod refers to the general method used in obtaining or calculating the result. BR: Toxicity replicate and summary data have a default value of None unless a method other than the test MethodName is used for the calculations. Water quality measurement results have a default value of ToxWQMeasurement.

Template Field Name	LookUp List	Description & Business Rules
AnalyteName (previously EndPointCode)	AnalyteLookUp	AnalyteName refers to the parameter being measured. BR: Toxicity examples include Survival, Young/female, Biomass (weight/orig indiv). Water quality measurment examples include pH, Ammonia as NH3, Salinity. The recommendation is "Biomass" be calculated for all fish species (total weight of surviving individuals divided by the original number of organisms at the start of the test) and "Growth" be calculated for Hyalella growth weight (total weight of surviving individuals divided by the number of survivors at the end of the test). Toxicity endpoints in the database will change to represent this correction; the endpoints for all fish species will be expressed as biomass instead of growth weight. It is the recommendation of the panel that we do not go back and recalculate results in the database where growth was calculated instead of biomass. All previous measurements would be listed as growth or biomass according to analysis performed by the analyzing laboratory.
FractionName	FractionLookUp	FractionName is a specific descriptor of the Analyte. BR: Ammonia as NH3 is expressed as Total or Unionized, each of which would be expressed as the Fraction, distinguishing the appropriate Analyte. If there is no need for further description of the analyte the default value is None.
WQSource	MatrixLookUp	WQSource differentiates between water quality measurements taken in the overlying water as well as in the sediment or interstitialwater. BR: Overlyingwater is used for the overlying water measurements which is the default for all water quality measurements. Interstitialwater is used for the interstitial water measurements. NA is used for for all toxicity results.

Template Field Name	LookUp List	Description & Business Rules
TimePoint	TimePointLookUp	TimePoint refers to the point in time during the test at which the measurement was recorded for water quality measurements. BR: For water quality measurements, timepoints such as initial or final are used to indicate the measurments recorded at the beginning and end of the test. High and low are used to indicate the highest and lowest values recorded for a particular sample during the entire length of the test. Day 2 is used indicate a measurement taken on the 2nd day of the test. For toxicity measurements, the TimePoint should match the TestDuration value. So if a 7 day test is performed, the TimePoint for the toxicity results would be 7 Days.
UnitAnalyte (previously ToxResultsUnit)	UnitLookUp	UnitAnalyte indicates the units used in the measurment of the AnalyteName.
RepCount		RepCount is the total number of sample replicates analyzed for the associated toxpoint in the toxicity test. BR: There are a few circumstances where a replicate should not be counted or used in the calculations. For all tests, if a replicate was spilled before a result could be recorded, the RepCount would decrease by one replicate. For all tests but primarily Ceriodaphnia dubia, if the first toxpoint had a single individual and it was a male, the second toxpoint of Young/female would not be possible so the RepCount for the second toxpoint would decrease by one. For Hyalella only, if the first toxpoint had a Survival of 0, the second toxpoint of Growth (weight/surv indiv) would not be included in the calculations so the RepCount would decrease by one replicate.
Mean		Mean is the average result calculated from all replicates of a single sample.
StdDev		StdDev or standard deviation is a statistic that indicates how tightly all the replicates are clustered around the mean in a set of data. This calculation includes all the applicable replicates from a single sample.
StatisticalMethod (previously Statistical Test Code)	VariableCodesLookUp	StatisticalMethod is the statistical test or method used to calculate the probability of whether a test is significant or not. Esentially, whether the sample replicates are significantly different from the control.

Template Field Name	LookUp List	Description & Business Rules
AlphaValue		AlphaValue is the predetermined statistical acceptance level that is not calculated, but is chosen by the laboratory. BR: The default value for SWAMP is 0.05.
Probability		Probability is the calculated probability using a standard statistical analysis between the replicates of the field samples and the control or reference sample within a ToxBatch. BR: Negative control samples (CNEG) probability is 0.5.
MSD		The minimum significant difference (MSD) is a measurement that can be produced for each statistical comparison performed between sample and control, or among multiple concentrations of a sample and control. It represents the smallest significant difference from the control and is unique for each statistical comparison. BR: A threshold is generated for each batch of samples associated with a single control. The sample response will be evaluated by a statistical comparison to the control response, and then comparing it to the generated threshold. This provides a two-tier system for designating a sample as toxic.
EvalThreshold		Evaluation threshold or EvalThreshold is the associated level that is used to identify that an environmental sample is significantly different from its associated control sample and is recorded as a percentage, e.g. 80%. EvalThreshold = Control % - MSD % BR: Control sample should be reported as 80 for the corresponding endpoint. Unless supplied, the default EvalThreshold will be 80.

Template Field Name	LookUp List	Description & Business Rules
SigEffect	SigEffectLookUp	The toxicity significant effect code or SigEffect indicates whether the sample result is significantly different from the control. BR: The code is based on two criteria used to determine significance. The first or first two letters of the code refers to the significance compared to the negative control, which is determined whether it is above or below the alpha value of 0.05. The last letter of the code refers to the whether the %control is above or below the evaluation threshold. For example NSG, is not significant compared to negative control based on statistical test, it was below the alpha of 5%, and above the evaluation threshold. For labs submitting only one criterion to determine significance, the SWAMP DMT will apply the last letter of the code based on an EvalThreshold of 80 unless the lab provides an alternate EvalThreshold value. The SigEffectCode will receive an NA code in this instance. Sediment and water samples must be compared to the negative control sample. Reference sediment or salinity controls may be reported in the database for informational purposes. While the ToxSigEffectCode will always reflect a combination of the probability and evaluation threshold, best professional judgment of the lab can be reflected in the sample comment.
TestQACode (previously TestQualCode)	QALookUp	TestQACode is applied to each sample's toxpoint and water quality measurements to describe any special conditions, situations or outliers that occurred during or prior to the analysis to achieve the result. BR: The default code, indicating no special conditions, is None. If more than one code should be applied to a record, the convention is to list them in alphabetical order separated by commas and no spaces; i.e. BY,TW
SummaryComments (Not Required)		In the SummaryComments field note any comments necessary to describe special circumstances for the toxicity summary data for the specific record.

iii. Tox Batch Worksheet

The third worksheet to travel with the data holds information specific to the laboratory batch in which data is analyzed. This worksheet should be named **ToxBatch** (with no spaces) in its worksheet tab. The fields in this sheet are and should be completed as follows:

Template Field Name	LookUp List	Description & Business Rules
ToxBatch		The ToxBatch is assigned by the laboratory and groups all environmental samples and supporting QA samples within a unique analysis batch. It is used to compare field samples with their associated NegativeControls for statistical analysis and will be used to verify completeness based on the SWAMP QAPrP. BR: The ToxBatch should be listed only one time in the ToxBatch worksheet for each unique ToxBatch found in the Results and Summary worksheets. Follow the File and Batch Name Convention to correctly identify the batch. It is recommended to include the start date and an abbreviation of the OrganismName in the lab-specific portion of the ToxBatch.
LabAgencyCode	AgencyLookUp	AgencyCode refers to the organization, agency or laboratory that performed the analysis of the sample.
StartDate		StartDate refers to the date the test began.
RefToxBatch		RefToxBatch lists the Reference Tox Batch ID run with this batch of samples.
OrganismSupplier (Not Required)		OrganismSupplier refers to the agency that supplied the test organisms.
OrganismAgeAtTestStart (Not Required)		OrganismAgeAtTestStart indicates the age or age range (e.g. 7 days or 7-10 days) of the test organisms at the beginning of the test. The age or range is usually recommended by the method.
LabSubmissionCode (previously BatchQualCode)	LabSubmissionLookUp	The LabSubmissionCode is a unique batch qualifier code assigned to the ToxBatch as a whole by the analyzing laboratory which references the quality of the data in the ToxBatch. BR: If the LabSubmissionCode of A is used, meaning Acceptable, the laboratory is ensuring that all SWAMP QAQC protocols were met for the toxicity batch. If anything other than A is used, a ToxBatchComment is required.

Template Field Name	LookUp List	Description & Business Rules
SubmittingAgencyCode	AgencyLookUp	SubmittingAgencyCode is the organization or agency that is responsible for submission of the data to the database. This agency may be different from LabAgencyCode if the analytical data were subcontracted to another agency.
ToxBatchComments (Not Required)		The ToxBatchComments field is intended to record any comments relating to the ToxBatch as a whole. BR: If the LabSubmissionCode is anything other than A, a ToxBatchComment is required

c. Special Circumstances

i. Laboratory-generated QA samples (LABQA)

All samples generated from within the laboratory, such as CNEG, CNSL, etc. have specific alternative rules, which are as follows:

LabSampleID	Recommended - provide lab specific identification for an analyzed sample
StationCode	LABQA
EventCode	WQ for water and sediment chemistry and toxicity results
ProtocolCode	Not Applicable
LocationCode	Not Applicable
SampleDate	Date test started, expressed as dd/mmm/yyyy
CollectionTime	0:00
CollectionMethodCode	Not Applicable
SampleTypeCode	Select from SampleTypeLookup List
Replicate	1
CollectionDepth	-88
UnitCollectionDepth	m for water or cm for sediment
ProjectCode	Not Applicable
AgencyCode	Organization or agency that analyzed the sample
Matrix	Water samples - labwater or blankwater Sediment samples - blankmatrix (commercially generated product) or sediment (if laboratories is using solvent, water or nothing)

ii. Interstitial Water (Pore Water) Analysis

Certain sampling events create a special set of rules that apply for a few of the entry fields in the SWAMP database. One of these would be the collection of sediment from which interstitial water is extracted and then analyzed. Below are the fields that differentiate these samples from the norm and how they should be completed.

CollectionMethodCode	Sed_Grab
SampleTypeCode	Integrated
UnitCollectionDepth	cm
PreparationPreservation	Centrifuged plus any additional preparation done at the lab (Centrifuged, X)
PreparationPreservationDate	If no preparation was performed at the analyzing lab, enter the centrifuge date for *PreparationPreservationDate*. If a preparation or preservation was

	performed at the analyzing laboratory, enter the preparation date and include the date of centrifuge in the *CollectionComments*.
Matrix	sediment, interstitialwater
Unit	Water units

SWAMP
Surface Water Ambient Monitoring Program

SWAMP DATA MANAGEMENT PLAN

Taxonomy Results Template

Surface Water Ambient Monitoring Program

October 20, 2010

CALIFORNIA Water Boards

http://www.waterboards.ca.gov/water_issues/programs/swamp

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E. ANALYTICAL DATA

The typical method for submitting Laboratory Analytical Data to the SWAMP Database is electronically through the online data checker. This section will focus on the tools and steps involved in submitting these data.

The following information should be used in conjunction with the SWAMP QAPrP for complete compliance with the SWAMP program including measurement quality objectives and required QA samples.

2. TAXONOMY ANALYSIS AUTHORIZATION FORMS

Prior to a sampling event, it is helpful to organize and identify what taxonomic samples are to be analyzed. For those organizations contracting with the Department of Fish and Game (DFG) for sampling activity, the tool used for this expectation of work is called a Taxonomy Analysis Authorization Form. This AA form is a copy of the Taxonomy Results Template (also available online) with specific samples listed for analysis.

The Analysis Authorization (AA) form is an Excel workbook provided to the labs in an electronic format prior to sampling. It is used as a detailed supplement to the Chain of Custody (COC) documentation that travels with the samples from the field to the lab. Should any discrepancy between the two types of documentation occur, contact the provider of the Analysis Authorization for further clarification.

	A	B	C	D	E	F	G	H	I	J	K	L	M	N	O
1	EventCode	ProtocolCode	StationCode	SampleDate	AgencyCode	ProjectCode	UserName	SampleComments	LocationCode	GeometryShape	CollectionTime	CollectionMethodCode	SampleTypeCode	Replicate	CollectionDeviceName
2	EXAMPLE MARINE INVERTEBRATES														
11	WQ	MPSL-DFG_Field_v1.0	306MSLG22	04/Jun/2004	MPSL-MLML	SWAMP_RB3	msigala	Station in deep water	OpenWater	Point	10:00	Sed_Grab	Grab	1	MPSL-MLML Mod.VanV
12	WQ	MPSL-DFG_Field_v1.0	306MSLG22	04/Jun/2004	MPSL-MLML	SWAMP_RB3	msigala	Station in deep water	OpenWater	Point	10:00	Sed_Grab	Grab	1	MPSL-MLML Mod.VanV
13	WQ	MPSL-DFG_Field_v1.0	306MSLG22	04/Jun/2004	MPSL-MLML	SWAMP_RB3	msigala	Station in deep water	OpenWater	Point	10:00	Sed_Grab	Grab	2	MPSL-MLML Mod.VanV
14	WQ	MPSL-DFG_Field_v1.0	306MSLG22	04/Jun/2004	MPSL-MLML	SWAMP_RB3	msigala	Station in deep water	OpenWater	Point	10:00	Sed_Grab	Grab	2	MPSL-MLML Mod.VanV
15	WQ	MPSL-DFG_Field_v1.0	306MSLG22	04/Jun/2004	MPSL-MLML	SWAMP_RB3	msigala	Station in deep water	OpenWater	Point	10:00	Sed_Grab	Grab	2	MPSL-MLML Mod.VanV
16	WQ	MPSL-DFG_Field_v1.0	306MSLG22	04/Jun/2004	MPSL-MLML	SWAMP_RB3	msigala	Station in deep water	OpenWater	Point	10:00	Sed_Grab	Grab	1	MPSL-MLML Mod.VanV
17	WQ	MPSL-DFG_Field_v1.0	306MSLG22	04/Jun/2004	MPSL-MLML	SWAMP_RB3	msigala	Station in deep water	OpenWater	Point	10:00	Sed_Core	Grab	1	Core_0.0071m2
18	EXAMPLE BMI INVERTEBRATES														
19	BA	SWAMP_WS	401VCJVCL	05/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	BMI_TRC	Integrated	1	D-Frame Kick Net
20	BA	SWAMP_WS	401VCJVCL	05/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	BMI_RWB	Integrated	1	D-Frame Kick Net
21	BA	SWAMP_WS	401VCJVCL	05/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	BMI_RWB	Integrated	1	D-Frame Kick Net
22	BA	SWAMP_WS	401VCJVCL	05/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	BMI_RWB	Integrated	1	D-Frame Kick Net
23	BA	SWAMP_WS	401VCJVCL	05/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	BMI_RWB	Integrated	1	D-Frame Kick Net
24	BA	SWAMP_WS	401VCJVCL	05/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	BMI_RWB	Integrated	1	D-Frame Kick Net
25	BA	SWAMP_WS	401VCJVCL	05/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	BMI_RWB	Integrated	1	D-Frame Kick Net
26	BA	SWAMP_WS	401VCJVCL	05/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	BMI_RWB	Integrated	1	D-Frame Kick Net
27	BA	SWAMP_WS	401VCMADL	07/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	BMI_RWB	Integrated	1	D-Frame Kick Net
28	BA	SWAMP_WS	401VCMADL	07/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	BMI_RWB	Integrated	2	D-Frame Kick Net
29	EXAMPLE DIATOMS														
30	BA	SWAMP_WS	401VCMADL	07/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	Algae_SWAMP	Integrated	1	RubDelim & PVCDelim
31	BA	SWAMP_WS	401VCMADL	07/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	Algae_SWAMP	Integrated	1	RubDelim & PVCDelim
32	BA	SWAMP_WS	401VCMADL	07/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	Algae_SWAMP	Integrated	1	RubDelim & PVCDelim
33	BA	SWAMP_WS	401VCMADL	07/Jun/2006	MPSL-DFG	SWAMP_RB4	admin		X	Point	11:00	Algae_SWAMP	Integrated	1	RubDelim & PVCDelim

Notes_Information | BenthicCollectionResults | BenthicCollectionResultsEXAMPLE | AgencyLookUp | CollectionDevice_FieldResLookUp

- The **BenthicCollectionResults** worksheet provides the sample information in the correct template format for the laboratory to use in order to enter the analysis results.
- A **BenthicCollectionResults Example** worksheet is also listed which gives an example of the final formatting of the taxonomy template.

Regardless of whether an organization chooses to use these Analysis Authorization Forms or others like them that serve the same purpose, they are useful tools to help communicate and maintain consistency regarding samples collected and analyzed. If interested, contact the SWAMP DMT to receive an electronic version of an AA form template.

7. TAXONOMIC DATA

Data for marine benthic infauna, freshwater benthic macroinvertebrate (BMI), algae, and diatom taxonomic analyses are treated in a similar way in the SWAMP database and for this reason will be treated together in this portion of the document.

a. Taxonomy Results Template

The Taxonomy Results Template is available online at http://swamp.mpsl.mlml.calstate.edu/resources-and-downloads/database-management-systems/swamp-25-database/templates-25#Taxonomic.

b. Formatting the Data

A Microsoft Excel template exists for laboratories to use to format and submit taxonomy results in a manner that can be easily loaded into the SWAMP database. As previously discussed, for many labs working under the SWAMP master contract, the station collection information can be obtained from the Taxonomy Analysis Authorization form. This section will discuss each of the fields in the template and how to populate them.

All valid LookUp list values are available at http://ftp.mpsl.mlml.calstate.edu/LookUpLists.php. If the necessary field and/or values do not appear in the LookUp list, a request to have it added can be submitted to the SWAMP Data Management Team (DMT).

i. Benthic Collection Results Worksheet

There is one worksheet that must travel with the data for the data package to be considered complete. This worksheet holds all marine benthic infauna, freshwater benthic macroinvertebrate (BMI), algae, and diatom collection and result data, and should be named **BenthicCollectionResults** in a worksheet tab. Each record in this sheet represents a unique taxonomic identification from each sample and collection event. If beginning with a Taxonomy Analysis Authorization form, the following fields will likely be pre-populated in the **BenthicCollectionResults** worksheet: *EventCode*, *ProtocolCode*, *StationCode*, *SampleDate*, *AgencyCode*, *ProjectCode*, *LocationCode*, *CollectionTime*, *CollectionMethodCode*, *SampleTypeCode*, *Replicate*, *CollectionDeviceName*, *SieveSize*, *CollectionDepth*, and *UnitCollectionDepth*. Please note that all fields are required to have data except when otherwise noted below. Examples of special types of samples are listed in the Special Circumstances section.

Template Field Name	LookUp List	Description & Business Rules
StationCode	StationLookUp	StationCode represents a unique sampling site in a sampling design. A single waterbody may have multiple stations. BR: StationCode should represent a geographic location not a geographic location combined with a replicate reference. There cannot be multiple StationCodes in the StationLookUp with the same target latitute/longitude coordinates. The format for the unique alphanumeric description of the station is ###ABC123, where ### is the Hydrologic Unit number and ABC123 is an alphanumeric description of the Station. An example is 111EELBRN which is Hydrologic Unit 111 and an abbreviated code to indicate “Eel River - South Fork near Branscomb”. If the exact Hydrologic Unit is not known, populate the first number with the regional board number; i.e. the station is known to be in California Regional Water Board 3, the StationCode would be 300######. If the station is not in California, the Hydrologic Unit is 000.

Template Field Name	LookUp List	Description & Business Rules
SampleDate		SampleDate refers to the date the sample was collected in the field. BR: The format for date in the templates is dd/mmm/yyyy, such as 10/Nov/2007. For samples with collection times that last longer than one day, like autosamplers, the sample date is the date sampling began. When entering data using the forms, the format is mm/dd/yy. For transplanted bivalves, the SampleDate is the date the transplanted organisms were collected, removed, or retrieved from the field.
ProjectCode	ProjectLookUp	ProjectCode is a unique code referencing a project which includes data of a single study design; e.g. RWB5_StS_FY0708, RWB2_Status_YR1, RWB7_Trend_2003, SWB_RCMP_2008. BR: ProjectCodes with FY in the name indicate samples were collected in a fiscal year calendar cycle, e.g. FY0708 would indicate fiscal year 2007-2008 which is July 1, 2007 through June 30, 2008. ProjectCodes without FY indicate samples were collected in a calendar year, e.g. 2003 would indicate calendar year 2003 which is January 1, 2003 through December 31, 2003. Each Project must have an associated QAPrP or Project QAPP listed. SWAMP moves data to the permanent side of the database by ProjectCode.

Template Field Name	LookUp List	Description & Business Rules
EventCode	EventLookUp	EventCode represents the initial intent of the sampling event at a particular station. BR: The EventCode will be in a hierarchical order as follows: BA – If the initial intent of sampling is for Bioassessment (PHab, BMI [freshwater], and/or Algae) (Tissue and/or WaterQuality samples may or may not also be collected) TI – If the initial intent of sampling is for Tissue (WaterQuality samples may or may not also be collected; no associated Bioassessment samples collected) WQ – If the initial intent of sampling is for WaterQuality (Water, Sediment, Toxicity, and/or Marine Benthic Abundance) (no associated Bioassessment or Tissue samples collected) For example, if the initial intent of sampling on Day 1 was for Tissue and WaterQuality, the EventCode would be TI. If for some reason the WaterQuality had to be re-sampled the next day, on Day 2, the event for the re-sampling would still be TI because Tissue was the initial intent of sampling on Day 1 even though WaterQuality was sampled on Day 2.

Template Field Name	LookUp List	Description & Business Rules
ProtocolCode	ProtocolLookUp	ProtocolCode represents the sampling protocol used, which includes the set of methods, methodology, and/or specifications; e.g. MPSL-DFG_Field_v1.0 or SWAMP_WS_2007. Established protocols may be used or Regions may document their own sampling protocols. BR: It is preferable to combine protocols per StationCode and date so that all WaterQuality, Bioassessment, and Tissue data are combined under the same EventCode and Protocol. For example, if Tissue and WaterQuality are sampled at a station, the EventCode would be TI. If the protocols are different for Tissue and WaterQuality, the Tissue protocol would be used and the WaterQuality protocol would be listed in the SampleComments. If that is not preferable, separate EventCodes may be used with each individual protocol. Not Recorded is only used for historic data. All non-historic SWAMP projects require a true protocol reference. Not Applicable is used for LABQA and 000NONPJ samples only.
AgencyCode	AgencyLookUp	AgencyCode refers to the organization or agency that collected the sample. This should be listed on the Chain of Custody (COC) document that accompanies the samples from the field. BR: If an environmental sample is used to perform laboratory QC, i.e. a matrix spike or lab duplicate, the AgencyCode still refers to the agency that collected the native sample, not the agency that created the QC sample. See 'Special Circumstances' for LABQA business rules.
SampleComments (Not Required)		SampleComments records any comments related to the sampling event at a particular station. BR: When a PurposeFailureCode of Other is used, a SampleComment is required.

Template Field Name	LookUp List	Description & Business Rules
LocationCode	LocationLookUp	LocationCode describes the physical location in the waterbody where the sample was collected. One sampling event may have a single or multiple locations. BR: For a single point of sampling, the physical location in the waterbody can be used such as Bank, Thalweg, Midchannel, OpenWater, Transect 1, or X (general location for bioassessments). For field results, the LocationCode should be the same as the location for the WaterQuality collection method. For TI EventType sampling, the physical location plus the CollectionMethod is used such as BankNet1, BankShock1, OpenWaterTrawl1, OpenWaterNet1. For resident mussel or clam collections, the LocationCode would be the physical location in the water body plus the generic CollectionMethod, e.g. BankTissue_Grab1. OpenWater sampling with multiple sub-locations within a single water body or station could have locations of OpenWaterTrawl1, OpenWaterTrawl2 describing one large location with two smaller areas of sampling within the OpenWater Location. Multiple physical locations within a single station could consist of a LocationCode such as BankShock1, BankNet1, OpenWaterHook1. If recording specific locations within a station are necessary for the project, a LocationCode such as Location1Net1, Location1Net2, Location2Shock1 may be used.
GeometryShape (Not Required)	VariableCodesLookUp	GeometryShape is the physical shape of the location; e.g. Line, Point, Line Centroid, Polygon, Polygon Centroid.

Template Field Name	LookUp List	Description & Business Rules
CollectionTime		CollectionTime refers to the time when the first field measure of a sampling event at a specific station was recorded in the field. BR: If the sampling crew collects 18 field measurements at a single station, the CollectionTime for each would be the time of the first measurement recorded. By doing so, the samples can easily be linked and any holding time issues will be consistent, and conservative, for laboratory work. The CollectionTime format should be expressed as xx:xx in 24 hour time, such as 13:30 for 1:30 pm.
CollectionMethodCode	CollectionMethodLookUp	CollectionMethodCode refers to the general method of collection. BR: For BA EventCodes, use the appropriate CollectionMethodCode for BMIs (e.g., BMI_RWB, BMI_CSBP) and for Algae (e.g., Algae_SWAMP, Algae_EMAP) that follows the associated protocol. The SWAMP marine invertebrates default is Sed_Grab.
SampleTypeCode	SampleTypeLookUp	SampleTypeCode refers to the type of sample collected or analyzed. BR: Some commonly used SampleTypeCode choices include Grab, Integrated, Macroalgae, Microalgae, Epiphyte, Qualitative, etc. When laboratories report soft algae taxonomy data with multiple sample types (Epiphyte, Macroalgae, Microalgae, Integrated, Qualitative), a comment should be added to the BenthicCollectionComment identifying if no taxonomic results were found for any or all of the SampleTypeCodes. It is the data submitter's responsibility to update the BenthicCollectionComment for this particular SampleTypeCode where no results were found and to notify the data loader of the change. The submitting laboratory should not delete the SampleType row from the MS Excel file.

Template Field Name	LookUp List	Description & Business Rules
Replicate		Replicate is the collection replicate number used to distinguish between replicates created at collection. BR: If a replicate was performed in the lab, indicate replicate 2 and make a comment in the BenthicCollectionComments that it was a lab replicate.
CollectionDeviceName	CollectionDeviceLookUp	CollectionDeviceName refers to the specific device used in the collection of the sample such as MPSL-MLML Mod.VanVeen Grab-Dbl_0.05m2, 0.05m2 or Core_0.0071m2 for marine invertebrate collections, D-Frame Kick Net for BMI invertebrate collections, and RubDelim & PVCDelim or Individual Collection by Hand for algae and diatom collections.
SieveSize	VariableCodesLookUp	SieveSize is the size of the sieve the sample was passed through; e.g. 0.5mm, none
CollectionDepth		CollectionDepth records the level, from the surface in the water or sediment column, at which the field measurements were recorded. BR: This information should be listed on the field data sheet from the field crew. CollectionDepth for water samples would be measured from the water surface and recorded in meters (m). Since depths for ambient monitoring field measurements are generally "subsurface", defaults have been established to indicate this. For water samples, the default value is 0.1 m. For freshwater bioassessments, algae, and diatom samples where samples are composited, the default is -88 m. If samples were composited from multiple locations at different depths, CollectionDepth is -88.
UnitCollectionDepth	VariableCodesLookUp	UnitCollectionDepth refers to the units used in the CollectionDepth including cm (centimeters) and m (meters). This information should be listed on the Chain of Custody (COC) document that accompanies samples from the field.

Template Field Name	LookUp List	Description & Business Rules
SampleID (Not Required)		SampleID is a unique identifier supplied by the organization directing the sampling or sampling agency and is used to track the sample throughout the sampling and analysis processes. This field can be used to tie a result to the sample. BR: This SampleID, which is different from the StationCode, will likely be on the sample container the laboratory receives from the field crew or on the COC. If there is no number, leave this field blank.
BenthicCollectionComments (Not Required)		BenthicCollectionComments records any comments relating to the collection of the field sample for laboratory analysis. BR: If no results were found, enter a comment stating that no organisms were identified. If a sample collection is missing an expected bottle, or the bottle broke or was lost enroute to or at the laboratory, enter a comment here describing why results are not reported. The submitting laboratory should not delete the sample row from the MS Excel file in either situation.
GrabSize (Not Required)		GrabSize is the total area of substrate collected for the sample, regardless of CollectionDevice area size. This is determined by the sampling device area and, if applicable, the number of transects or grabs sampled.
UnitGrabSize (Not Required)	VariableCodesLookUp	UnitGrabSize refers to the units used for GrabSize; from UnitGrabSizeList in VariableCodesLookUp; e.g. m2 or cm2.
ReplicateName (Not Required)		ReplicateName is the name of the Replicate Number if applicable; e.g. Transect 1 or T-1. This field was used previously in the CalEDAS database to identify if replicate samples were collected.
ReplicateCollectionDate (Not Required)		ReplicateCollectionDate is the date of the ReplicateCollection; format as dd/mmm/yyyy
NumberJars (Not Required)		NumberJars is the number of jars into which the sample fit for transport to the analytical lab
BenthicCollectionDetailComments (Not Required)		BenthicCollectionDetailComments are comments related to the BenthicCollectionDetail

Template Field Name	LookUp List	Description & Business Rules
AgencyCode_LabEffort	AgencyLookUp	AgencyCode_LabEffort is the agency that sorted or processed the taxonomic sample
PersonnelCode_LabEffort	PersonnelLookUp	PersonnelCode_LabEffort refers to the Lab personnel (sorter or taxonomist) who processed the sample, performed the initial sorting of the sample, and was responsible for this entry.
PercentSampleCounted (Not Required)		PercentSampleCounted refers to the percent of the sample that was counted.
TotalGrids (Not Required)		TotalGrids is the total number of grids onto which the sample was spread for subsampling.
GridsAnalyzed (Not Required)		GridsAnalyzed is the number of grids (in integers) of material pulled from to achieve the TargetOrganismCount.
GridsVolumeAnalyzed (Not Required)		Volume of grids included in the analysis required to achieve the TargetOrganismCount; i.e. if 0.25 from each of 3 grids were analyzed, GridsVolumeAnalyzed would be 0.75. Units are in grids.
TargetOrganismCount (Not Required)		TargetOrganismCount is the number of organisms at which subsampling will cease.
ActualOrganismCount (Not Required)		ActualOrganismCount is the total number of organisms recovered by lab sorter in all grids analyzed, including the count above and beyond the target total for the subsample.
ExtraOrganismCount (Not Required)		ExtraOrganismCount is the number of organisms subsampled beyond the target count.
QCOrganismCount (Not Required)		QCOrganismCount refers to the number of organisms in the subsample counted during the quality control (QC) check. It refers to cases where a sample is re-sorted and more organisms are found. If the original sort found 500 organisms and then it was re-sorted by a different person who found 3 organisms, the QCOrganismCount would be 3 rather than 503.
DiscardedOrganismCount (Not Required)		DiscardedOrganismCount is the number of organisms in the subsample determined to be unsuitable for identification.

Template Field Name	LookUp List	Description & Business Rules
EffortQACode	EffortQALookUp	EffortQACode is a unique code applied to the result which describes any special conditions, situations or outliers occurring during or prior to lab sorting.
BenthicLabEffortComments (Not Required)		BenthicLabEffortComments are the comments related to lab sorting or sample processing.
FinalID	OrganismLookup	FinalID refers to the lowest taxon level identified for the organism.
LifeStageCode	LifeStageLookUp	LifeStageCode is a unique code referencing the stage of life of the organism; e.g. adult, juvenile, larvae.
Distinct		Distinct is an indicator of whether this is a distinct (>=1) or non-distinct (0) taxon.
BAResult		BAResult is the number of individuals of a given FinalID and stage that were identified within a sample replicate. This is for unadjusted (raw) counts and is to be used for cases where a TargetOrganismCount is used. Either BAResult or Result should be populated (unless QACodes and ResQualCodes other than the defaults are used), but NOT both fields.
Result		Result is the final numeric result of a given FinalID and stage scaled up to grab size. Result is for counts adjusted to the area sampled and for biovolumes and it may represent raw counts if the full sample is sorted. BR: Either BAResult or Result should be populated (unless QACodes and ResQualCodes other than the defaults are used), but NOT both fields.
UnitName	UnitLookUp	UnitName refers to how the taxonomic result is measured or expressed. BR: Taxonomic units are indicated by count or volume/area, e.g. um3/cm2

Template Field Name	LookUp List	Description & Business Rules
ResQualCode	ResQualLookUp	ResQualCode or Result Qualifier Code qualifies the analytical result of the sample. BR: This field should have an equal sign (=) for results that are recorded. When a result is Greater Than (>) or Less Than (<), a ResQualCode is required and the appropriate code would be applied. When both the BAResult and Result are blank, a ResQualCode is required. If the ResQualCode value is NR for Not Recorded, then a reason for this code must be written in the BenthicResultComments field and/or an appropriate QACode would be applied.
QACode	QALookUp	QACode is applied to the result to describe any special conditions, situations or outliers that occurred during or prior to the identification to achieve the result. BR: The default code, indicating no special conditions, is None. If more than one code should be applied to a record, the convention is to list them in alphabetical order separated by commas and no spaces; e.g. GB,SC.
ComplianceCode	ComplianceLookUp	ComplianceCode is a unique code referencing the Compliance with the associated QAPP.
BatchVerificationCode	BatchVerificationLookUp	BatchVerificationCode is a unique code referencing the Verification of a Batch.
TaxonomicQualifier (Not Required)	VariableCodesLookUp	TaxonomicQualifier is a code representing the taxonomist's justification for not identifying a specimen to standard effort; from TaxonomicQualifierList in VariableCodesLookUp.
ExcludedTaxa (Not Required)	VariableCodesLookUp	ExcludedTaxa is a code representing the taxonomist's justification for excluding a specimen from analysis; from ExcludedQualifierList in VariableCodesLookUp.
PersonnelCode_Result	PersonnelLookUp	PersonnelCode_Result is a unique code referencing the personnel or person who performed the taxonomy and produced the FinalID. In some cases, the PersonnelCode_LabEffort and PersonnelCode_Result are the same person. In other cases, the lab pays a student to do the initial sorting of pulling organisms out of the sample (PersonnelCode_Lab Effort) and then has a taxonomist do the actual identification (PersonnelCode_Results).

Template Field Name	LookUp List	Description & Business Rules
LabSampleID (Not Required)		LabSampleID is a recommended field intended to provide lab specific identification for an analyzed sample. The format and content is determined by the lab.
EnterDate (Not Required)		EnterDate is the date the data were entered into the template; formatted as dd/mmm/yyyy.
BenthicResultComments (Not Required)		BenthicResultComments are comments related to the BenthicResult or individual taxa count.

c. *Special Circumstances*

There are two types of special circumstances discussed in this section. SWAMP is currently developing QA/QC guidance for freshwater bioassessment taxonomy.

(a) Qualitative Organism Identifications

Qualitative organism identifications refer to samples where the field personnel note the presence of algae in the stream without noting the abundance of the taxon. The special circumstances for recording these types of results are as follows:

SampleTypeCode	Qualitative
Result	Leave null
ResQualCode	P (present)
Unit	count

(b) Colonial Organism Counts

Colonial organism counts refer to samples where the objective is to obtain a count for a taxon but a colonial organism is found. Laboratories should not provide a count in these cases due to the difficulty in obtaining a count for certain organism types (e.g. a sponge or hydroid). If a biovolume or biomass is the objective, then record the result and appropriate unit. The special circumstances for recording colonial organism counts are as follows:

SampleTypeCode	Epiphyte, Macroalgae, etc. (not Qualitative)
Result	Leave null
ResQualCode	COL (Colonial)
Unit	count

d. *Procedure for Adding New Organisms to the Database*

There are instances when new organisms will need to be added to the SWAMP database. Following is the procedure to add those organisms.

i. Adding a New Organism

The first step is to check the OrganismLookUp list online http://ftp.mpsl.mlml.calstate.edu/LookUpLists.php to determine if the organism already exists in the list. If the organism does not exist in SWAMP, the name and associated information must be submitted to the SWAMP DMT for consideration to be added to the database. Additional information may be found through SCAMIT for marine taxonomic references and SAFIT for freshwater taxonomic references.

ii. Organism_DetailLookUp Worksheet

Once it is determined that an organism is not in the current lookup list, complete the Organism_DetailLookUp worksheet in the SWAMP Taxonomy Results Template. Please note that all fields are required to have data except when otherwise noted below, but it is preferred to populate all fields if the information is known. In cases where an organism has multiple LifeStageCodes, a record consisting of each FinalID and LifeStageCode combination would be expected.

Template Field Name	LookUp List	Description & Business Rules
CommonName (Not Required)		CommonName refers to the common name of the organism collected.
FinalID		FinalID is the lowest taxon level identified for the organism. BR: Do not use sp. or spp. after the genus name as part of the FinalID if the organism can only be identified down to the genus and not the species. It is allowable if the organism is known as Genus sp A, where Species A is a known and documented species.
ParentFinalID (Not Required)		ParentFinalID is the FinalID of the parent Taxon ID.
Phylum		Phylum is the phylum of the organism; a taxonomic rank used in the classification of organisms.
Subphylum (Not Required)		Subphylum is the subphylum of the organism; a taxonomic rank used in the classification of organisms.
Superclass (Not Required)		Superclass is the superclass of the organism; a taxonomic rank used in the classification of organisms.
Class		Class is the class of the organism; a taxonomic rank used in the classification of organisms.
Subclass (Not Required)		Subclass is the subclass of the organism; a taxonomic rank used in the classification of organisms.
Superorder (Not Required)		Superorder is the superorder of the organism; a taxonomic rank used in the classification of organisms.
Order		Order is the order of the organism; a taxonomic rank used in the classification of organisms.

Template Field Name	LookUp List	Description & Business Rules
Suborder (Not Required)		Suborder is the suborder of the organism; a taxonomic rank used in the classification of organisms.
Superfamily (Not Required)		Superfamily is the superfamily of the organism; a taxonomic rank used in the classification of organisms.
Family		Family is the family of the organism; a taxonomic rank used in the classification of organisms.
Subfamily (Not Required)		Subfamily is the subfamily of the organism; a taxonomic rank used in the classification of organisms.
Tribe (Not Required)		Tribe is the tribe of the organism; a taxonomic rank used in the classification of organisms.
GenusComplex (Not Required)		GenusComplex is the genus complex of the organism; a taxonomic rank used in the classification of organisms.
Genus		Genus is the genus of the organism; a taxonomic rank used in the classification of organisms.
SpeciesComplex (Not Required)		SpeciesComplex is the species complex of the organism; a taxonomic rank used in the classification of organisms.
Species		Species is the species of the organism; a taxonomic rank used in the classification of organisms.
Subspecies		Subspecies is the subspecies of the organism; a taxonomic rank used in the classification of organisms.
Variety (Not Required)		Variety is the variety of the organism; a taxonomic rank used in the classification of organisms.
TaxonomicLevelCode	TaxonomicLevelLookup	TaxonomicLevelCode is a unique code referencing the level of the FinalID.
LifeStageDefault	LifeStageLookUp	LifeStageDefault is the default LifeStageCode of each organism or FinalID one expects to find and to be used in analyses. BR: Default value is NR.

Template Field Name	LookUp List	Description & Business Rules
TaxonomicAuthority (Not Required)		TaxonomicAuthority is the Taxonomist or reference (including year) used to identify the organism; e.g. Barnum 1905. BR: If the FinalID is defined to genus and species, a TaxonomicAuthority is required. If it is defined to the family level or higher, this field may be left blank if not applicable.
CaliforniaTaxon (Not Required)		CaliforniaTaxon is a yes/no field used to record if the organism is a CaliforniaTaxon, i.e. that it exists in California regardless if it exists in other states or not. BR: Limit values to TRUE or FALSE where TRUE identifies the FinalID as a California taxon.
DateFinalIDRevised (Not Required)		DateFinalIDRevised is the date the FinalID was revised. BR: Format is dd/mmm/yyyy.
FinalIDAuthority	AgencyLookUp	FinalIDAuthority represents the Agency that has the authority to change values for the FinalID; e.g. SAFIT, SCAMIT.
Source (Not Required)	AgencyLookUp	Source is the agency that introduced or added the organism to the database; e.g. MPSL-MLML, DFG-ABL, SFEI.
OrganismComments (Not Required)		OrganismComments are comments related to the organism.
LifeStageCode	LifeStageLookUp	LifeStageCode is a unique code referencing the stage of life of the organism; e.g. adult, larvae. Each FinalID can have multiple yet different LifeStageCodes to create unique records; i.e. one FinalID can have both an adult and larvae LifeStageCode but not two adult LifeStageCodes.

iii. Submit Data to SWAMP

Submit taxonomic results and new LookUp values to SWAMP by following the Submitting Data To SWAMP Data Management Team procedures and using the Online Data Checker.

iv. Authority Check

Proposed new organisms will be forwarded to SCCWRP who will then forward it to the appropriate authority (e.g. SAFIT or SCAMIT) for review and consideration to be added to the SWAMP database. Follow-up questions or comments may be needed before final resolution is completed.

v. Clarification of New Organisms

If the authority has questions about the new organism, SCCWRP will be contacted. SCCWRP will then contact the SWAMP DMT who will contact the submitting laboratory to obtain the answers. This information will be relayed back to the appropriate authority for final approval of the new organism.

vi. Addition of New Organisms

Once the authority approves the new organism for addition to the SWAMP database, SCCWRP will be notified. SCCWRP will then notify the SWAMP DMT. The SWAMP DMT will then add the new organism to the SWAMP database.

APPENDIX J:

HEALTH AND SAFETY PROCEDURES

4. CONSOLIDATED MONITORING PROGRAM

A. Health and Safety

The information provided in this section are general safety guidelines. Additional, site-specific safety instructions are provided for most CMP stations. This guidance is available in David Ortega's (951-955-4390) office.

Safety of the sampling team is paramount. The field vehicle should start out with a full tank of gas and be in good repair. Extra care must be taken when driving at night or in the rain. If the sampling location is unsafe, make note of the unsafe situation and either do not collect a sample or come back after the hazardous situation has ceased.

1. Before You Leave

1.1 Always let someone else at the office know where you are, what your intended route or set of activities is, when you intend to return, and what to do if you don't come back at the appointed time. If in rough terrain, communicate with base when leaving and arriving at sampling sites.

1.2 Inspect the vehicle prior to leaving base. Make sure communication equipment and vehicle are in proper working condition. All safety equipment should be in the vehicle. Gas up the vehicle before you leave.

1.3 Know your equipment, sampling instructions, and procedures before going out into the field. Prepare labels, calibrate field meters, and clean equipment before you get started.

1.4 Know how to use and store chemicals. Do not expose chemicals or equipment to temperature extremes or long term direct sunshine.

2. Arriving at the Sampling Location

2.1 Prior to sampling, always assess the situation at the site. If it is unsafe, move on and return later or find a safer alternate location that will give comparable water quality results. If the situation remains unsafe, record the hazardous conditions in the *Field Data Sheet*, including photos, and do not collect a sample. **Your safety is more important than the data.**

2.2 If visibility is poor, use the vehicle lights to sweep the sampling site for safe entry and to illuminate the path. Use flashlights for additional lighting.

2.3 Do not cross low water crossings as the depth of the water and the integrity of the underlying roadway is uncertain. Floating debris may damage the vehicle or even push it from the roadway. Water as shallow as two feet deep can float a car.

2.4 Never cross private property without the permission of the landowner. Carry your employee identification card with you.

2.5 Watch for irate dogs, farm animals, wildlife, and insects such as ticks, hornets, wasps, and bees. Know what to do if you get bitten or stung.

3. Sampling Safety

3.1 When working with potentially hazardous materials, follow EPA, OSHA, and specific health and safety procedures. HAZWOPER certification is required if you will be sampling potentially hazardous materials.

3.2 Confined space entry is not allowed.

3.3 Avoid contact between chemical reagents and skin, eyes, nose, and mouth.

3.4 Know chemical cleanup and disposal procedures. Wipe up all spills when they occur.

3.5 Monitoring should be a team activity. Two person teams are required for night and wet weather sampling.

3.6 If you have health considerations such as epilepsy, severe allergies, or diabetes, let your partner know, as well as instructions on what to do if you need emergency assistance.

3.7 If in an active traffic area, cones should be placed around the parked vehicle, and personnel should be wearing reflective clothing. Make sure you are visible to passing traffic. Do not stand in front of the vehicle while sampling; better yet, park the vehicle a safe distance away. If visibility is poor, use flashlights when walking to and from the sampling site.

3.8 If sampling from a bridge, be wary of passing traffic, and personnel should be wearing reflective clothing. Never lean over the bridge rails unless you are firmly anchored to the ground or the bridge with good hand/foot holds. Never stand in front of the vehicle while sampling if it is parked on the bridge. Place flashing lights uproad when sampling off a roadway bridge. If visibility is poor, use flashlights when walking to and from the sampling site.

3.9 The person performing the sampling should be properly secured, such as on a lifeline, and be wearing adequate protective equipment.

3.10 Stay out of the channel if at all possible. Dip sample rather than enter the channel.

3.1 If you must enter the channel, use two hands to descend and ascend the iron ladder attached to the channel wall. Have your partner lower the sample bottles in an ice chest attached to a rope.

3.12 Test the traction before walking down a slope.

3.13 Do not walk on unstable stream banks. Disturbing these banks can accelerate erosion, contaminate your sample, and might prove dangerous if a bank collapses.

3.14 Be very careful when walking in the stream itself. Rocky-bottom streams can be very slippery and can contain deep pools; muddy-bottom streams might also prove treacherous in areas where mud, silt, or sand have accumulated in sink holes. If you must cross the stream, use a walking stick to steady yourself and to probe for deep water or muck. Your partner(s) should wait on dry land ready to assist you if you fall. Never wade in swift or high water. A rule of thumb used by USGS is to not attempt wading in a stream where the depth multiplied by the velocity is equal to or greater

than 10 sq ft/s. Watch the stream stage, especially if there is a chance it could rise rapidly.

3.15 Wear hip boots or chest waders. Boots and waders protect against contaminants, cold, and underwater objects. Be aware that traction may be impaired while wearing them. Do not allow water to get into the boots or waders.

3.16 If sampling at night, carry a bright flashlight or use a head-mounted lantern. Avoid shining the light in others' eyes.

3.17 Do not go sampling if severe weather is predicted. If there is active lightning, find cover to avoid being struck.

3.18 Watch for poison ivy, oak or sumac, and other types of vegetation in your area, such as nettles, that can cause rashes and irritation.

3.19 Never drink the water in a stream, no matter how pristine the environment appears. Assume it is unsafe to drink, and bring your own water or sports drink from home. After monitoring, wash your hands with antibacterial soap.

3.20 The ideal comfort range for humans is between 60 and 90 degrees Fahrenheit. Hypothermia (cold) and hyperthermia (heat) may occur outside this range. Be aware of the symptoms of either and leave the sampling area if symptoms begin to appear.

3.21 If you become injured in the field, contact your supervisor or another supervisor for instructions. Contact the nearest worker's compensation medical clinic (Appendix D.4) for an appointment. If the injury creates an emergency situation, call 911 and contact the District after the situation is stabilized.

Medical Referral for Treatment of Work-related Injuries or Illnesses

The treatment of work-related injuries or illnesses is handled through the County of Riverside Human Resources Department, Workers' Compensation Division.

The following website will locate the closest medical provider. The Medical Provider Network (MPN) includes the Kaiser Occupational Clinics in Riverside and Fontana. Upon clicking the link below, the MPN search page will load. Simply type in the name of the provider or choose a specialty option and click "Find Providers". An option to adjust the range of the search is also provided by utilizing the "within miles" tool.

http://www.compcaremed.com/riverside/

More information on the MPN is accessible on the County's workforceExchange Website:

http://www.workforceexchange.net/HRToolbox/WorkersCompensation/MedicalProviderNetwork/tabid/1632/Default.aspx

Questions regarding the MPN program may be answered by contacting the following:

CorVel Corporation, MPN Department	800-966-5307
Riverside County Workers' Compensation Division (HR Dept)	951-955-3530

APPENDIX K:

NPDES CONTACTS

NPDES Watershed Coordinators

Santa Ana Region
Julianna Gonzalez
951-955-8064
juliannagonzalez@rcflood.org

Santa Margarita Region
Eric Lomeli
951-955-8589
erlomeli@rcflood.org

Whitewater River Region
Scott Bruckner
951-955-0843
sebruckner@rcflood.org

NPDES Program Manager
Jason Uhley
951-955-1273
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County and other District Contacts

Riverside County Executive Office

Mr. Steven Horn
4080 Lemon Street, 4th Floor
Riverside, CA 92501
Tel:951.955.1110
Fax: 951.955.1105
shorn@rceo.org

Riverside County Department of Environmental Health

Dottie Ellis-Merki or Jeff Johnson
3880 Lemon Street, 2nd Floor
Riverside, CA 92501
Tel: 951. 358.5172 or 951.358.5055
DEllisme@rivcocha.org or JeffJohnson@rivcocha.org

Riverside County Facilities Management

Mr. John Alfred or Ms. Vikki Kuntz
3403 10th Street, Suite 500
Riverside, CA 92501
Phone: 951.955.4884 or 951.955.0166
Jalfred@rivcoeda.org or VKuntz@rivcoeda.org

Riverside County Transportation Land and Management Agency (TMLA)

Ms. Claudia Steiding or Ms. Adriana Soares
4080 Lemon Street, 8th Floor
Riverside, CA 92501
Tel: 951.955.1694 or 951.955.8823
Fax: 951.955.6721
csteiding@rctlma.org or ASoares@rctlma.org

Monitoring Program Manager

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Riverside County Flood Control and Water Conservation District
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Riverside, CA 92501
Tel: 951.955.2901
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Code Enforcement Contacts

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Cell: 951-830-8782
gflanner@rctlma.org

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lelandc@RanchoMirageCA.gov

Fire Department Contacts

The County of Riverside contracts with the following cities for emergency response services. For hazardous materials incidents within these cities, or within unincorporated Riverside County, contact County Environmental Health at 951-358-5055

Banning	Beaumont	Calimesa	Canyon Lake
Cathedral City	Coachella	Desert Hot Springs	Indian Wells
Indio	La Quinta	Lake Elsinore	Moreno Valley
Murrieta	Norco	Palm Desert	Palm Springs
Perris	Rancho Mirage	Rubidoux	San Jacinto
Temecula			

The following cities have their own hazardous material management program:

City of Riverside
Fire Station No. 2
9449 Andrew St.
Riverside, CA 92503
951-351-6102

City of Corona
Call 9-1-1

City of Hemet
Hemet Fire Department
510 E. Florida Ave.
Hemet, CA 92543
951-765-2450 during business hours
951-765-2400 outside of business hours

APPENDIX L:

CHARACTERISTICS OF CONTAMINANTS COMMONLY ASSOCIATED WITH VARIOUS FACILITIES AND ACTIVITIES

Table 4.[1] Typical Chemical and Physical properties of Industrial Non-Stormwater Discharges.

Industrial Categories Major Classifications SIC Group Numbers	Odor	Color	Turbidity	Floatables	Debris and Stains	Structural Damage	Vegetation	pH	TDS
Primary Industries									
20 Food and Kindred Products									
201 Meat Products	Spoiled Meats, Rotten Eggs and Flesh	Brown to Reddish-Brown	High	Animal Fats, Byproducts, Pieces of Processed Meats	Brown to Black	High	Flourish	Normal	High
202 Dairy Products	Spoiled Milk, Rancid Butter	Grey to White	High	Animal Fats, Spoiled Milk Products	Grey to Light Brown	High	Flourish	Acidic	High
203 Canned and Preserved Fruits and Vegetables	Decaying Products Compost Pile	Various	High	Seeds, Skins, Cores, Leaves	Brown	Low	Normal	Wide Range	High
204 Grain Mill Products	Slightly Sweet & Musty, Grainy	Brown to Reddish Brown	High	Grain Hulls and Skins, Straw & Plant Fragments	Light Brown	Low	Normal	Normal	High
205 Bakery Products	Sweet and/or Spoiled	Brown to Black	High	Cooking Oils, Lard, Flour, Sugar	Grey to Light Brown	Low	Normal	Normal	High
206 Sugar and Confectionary Products	NA	NA	Low	Low Potential	White Crystals	Low	Normal	Normal	High
207 Fats and Oils	Spoiled Meats, Lard or Grease	Brown to Black	High	Animal Fats, Lard	Grey to Light Brown	Low	Normal	Normal	High
208 Beverages	Flat Soda, Beer or Wine, Alcohol, Yeast	Various	Moderate	Grains & Hops, Broken Glass, Discarded Canning Items	Light Brown	High	Inhibited	Wide Range	High
21 Tobacco Manufactures	Dried Tobacco, Cigars, Cigarettes	Brown to Black	Low	Leaves, Papers and Fillers	Brown	Low	Normal	Normal	Low
22 Textile Mill Products	Wet Burlap, Bleach, Soap, Detergents	Various	High	Fibers, Oils, Grease	Grey to Black	Low	Inhibited	Basic	High
23 Apparel and Other Finished Products	NA	Various	Low	Some Fabric Particles	NA	Low	Normal	Normal	Low

[1] Dr. Robert Pitt of the University of Alabama developed a table of chemical and physical properties of Industrial Non-Stormwater Discharges. This provides an aid to identifying potential sources of illicit discharges when no other evidence is available.

Table 4.[1] Typical Chemical and Physical properties of Industrial Non-Stormwater Discharges.

Industrial Categories Major Classifications SIC Group Numbers	Odor	Color	Turbidity	Floatables	Debris and Stains	Structural Damage	Vegetation	pH	TDS
Material Manufacture									
24 Lumber & Wood Products	NA	NA	Low	Some Sawdust	Light Brown	Low	Normal	Normal	Low
25 Furniture & Fixtures	Various	Various	Low	Some Sawdust,Solvents	Light Brown	Low	Normal	Normal	Low
26 Paper & Allied Products	Bleach, Various Chemicals	Various	Moderate	Sawdust, Pulp Paper, Waxes, Oils	Light Brown	Low	Normal	Wide Range	Low
27 Printing, Publishing, and Allied Industries	Ink, Solvents	Brown to Black	Moderate	Paper Dust, Solvents	Grey to Light Brown	Low	Inhibited	Normal	High
31 Leather & Leather Products	Leather, Bleach, Rotten Eggs or Flesh	Various	High	Animal Flesh & Hair, Oils, Grease	Grey to Black, Salt Crystals	High	Highly Inhibited	Wide Range	High
33 Primary Metal Industries	Various	Brown to Black	Moderate	Ore, Coke, Oils, Limestone, Millscale	Grey to Black	High	Inhibited	Acidic	High
34 Fabricated Metal Products	Detergents, Rotten Eggs	Brown to Black	High	Dirt, Grease, Oils, Sand, Clay Dust	Grey to Black	Low	Inhibited	Wide Range	High
32 Stone, Clay, Glass, and Concrete Products	Wet Clay, Mud, Detergents	Brown to Reddish-Brown	Moderate	Glass Particles Dust from Clay or Stone	Grey to Light Brown	Low	Normal	Basic	Low
Chemical Manufacture									
28 Chemicals & Allied Products									
2812 Alkalies and Chlorine	Strong Halogen or Chlorine, Pungent, Burning	Alkalies - NA Chlorine - Yellow to Green	Low	NA	Alkalies – White Carbonate Scale Chlorine - NA	High	Highly Inhibited	Basic	High
2816 Inorganic Pigments	NA	Various	High	Low Potential	Various	Low	Highly Inhibited	Wide Range	High
282 Plastic Materials and Synthetics	Pungent, Fishy	Various	High	Plastic Fragments, Pieces of Synthetic Products	Various	Low	Inhibited	Wide Range	High

Table 4.[1] Typical Chemical and Physical properties of Industrial Non-Stormwater Discharges.

Industrial Categories Major Classifications SIC Group Numbers	Odor	Color	Turbidity	Floatables	Debris and Stains	Structural Damage	Vegetation	pH	TDS
283 Drugs	NA	Various	High	Gelatin Byproducts for Capsulating Drugs	Various	Low	Highly Inhibited	Normal	High
284 Soap, Detergents & Cleaning Preparations	Sweet or Flowery	Various	High	Oils, Grease	Grey to Black	Low	Inhibited	Basic	High
285 Paints, Varnishes, Lacquers, Enamels and Allied Products (SB - Solvent Base)	Latex - Ammonia SB – Dependent Upon Solvent (Paint Thinner, Mineral Spirits)	Various	High	Latex - NA SB - All Solvents	Grey to Black	Low	Inhibited	Latex-Basic SB-Normal	High
286 Indust. Organic Chemicals									
2861 Gum and Wood Chemicals	Pine Spirits	Brown to Black	High	Rosins and Pine Tars	Grey to Black	Low	Inhibited	Acidic	High
2865 Cyclic Crudes, & Cyclic Intermediates Dyes, & Organic Pigments	Sweet Organic Smell	NA	Low	Translucent Sheen	NA	Low	Highly Inhibited	Normal	Low
287 Agricultural Chemicals									
2873 Nitrogenous Fertilizers	NA	NA	Low	NA	White Crystalline Powder	High	Inhibited	Acidic	High
2874 Phosphatic Fertilizers	Pungent Sweet	Milky White	High	NA	White Amorphous Powder	High	Inhibited	Acidic	High
2875 Fertilizers, Mixing Only	Various	Brown to Black	High	Pelletized Fertilizers	Brown Amorphous Powder	Low	Normal	Normal	High
29 Petroleum Refining and Related Industries									
291 Petroleum Refining	Rotten Eggs, Kerosene, Gasoline	Brown to Black	High	Any Crude or Processed Fuel	Black Salt Crystals	Low	Inhibited	Wide Range	High

Table 4.[1] Typical Chemical and Physical properties of Industrial Non-Stormwater Discharges.

Industrial Categories Major Classifications SIC Group Numbers	Odor	Color	Turbidity	Floatables	Debris and Stains	Structural Damage	Vegetation	pH	TDS
30 Rubber & Miscellaneous Plastic Products	Rotten Eggs, Chlorine, Peroxide	Brown to Black	Moderate	Shredded Rubber Pieces of Fabric or Metal	Grey to Black	Low	Inhibited	Wide Range	High
Transportation & Construction									
15 Building Construction	Various	Brown to Black	High	Oils, Grease, Fuels	Grey to Black	Low	Normal	Normal	High
16 Heavy Construction	Various	Brown to Black	High	Oils, Grease, Fuels, Diluted Asphalt or Cement	Grey to Black	Low	Normal	Normal	High
Retail									
52 Building Materials, Hardware, Garden Supply, and Mobil Home Dealers	NA	Brown to Black	Low	Some Seeds, Plant Parts, Dirt, Sawdust, or Oil	Light Brown	Low	Normal	Normal	Low
53 Gen. Merchandise Stores	NA	NA	NA	NA	NA	Low	Normal	Normal	Low
54 Food Stores	Spoiled Produce, Rancid, Sour	Various	Low	Fragments of Food, Decaying Produce	Light Brown	Low	Flourish	Normal	Low
55 Automotive Dealers & Gasoline Service Stations	Oil or Gasoline	Brown to Black	Moderate	Oil or Gasoline	Brown	Low	Inhibited	Normal	Low
56 Apparel & Accessory Stores	NA	NA	Low	NA	NA	Low	Normal	Normal	Low
57 Home Furniture, Furnishings, & Equip. Stores	NA	NA	Low	NA	NA	Low	Normal	Normal	Low
58 Eating & Drinking Places	Spoiled Foods Oil & Grease	Brown to Black	Low	Spoiled or Leftover Foods	Brown	Low	Normal	Normal	Low
Miscellaneous									
Coal Steam Electric Power	NA	Brown to Black	High	Coal Dust	Black Amorphous Powder	Low	Normal	Slightly Acidic	Low
Nuclear Steam Electric Power	NA	Light Brown	Low	Oils, Lubricants	Light Brown	Low	Normal	Normal	Low

Table 5.[2] Contaminants Commonly Associated With Selected Facilities and Activities.

POTENTIAL SOURCE	CONTAMINANT
Commercial / Industrial	
Above-ground storage tanks	Arsenic, Barium, Benzene, Cadmium, 1,4-Dichlorobenzene or P-Dichlorobenzene, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Lead, Trichloroethylene (TCE), Tetrachloroethylene or Perchlorethylene (Perc)
Automobile, Body Shops/Repair Shops	Arsenic, Barium, Benzene, Cadmium, Chlorobenzene, Copper, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, 1,4-Dichlorobenzene or P-Dichlorobenzene, Lead, Fluoride, 1,1,1-Trichloroethane or Methyl Chloroform, Dichloromethane or Methylene Chloride, Tetrachloroethylene or Perchlorethylene (Perc), Trichloroethylene (TCE), Xylene (Mixed Isomers)
Boat Repair/Refinishing/Marinas	Benzene, Cadmium, cis 1,2-Dichloroethylene, Coliform, Cryptosporidium, Dichloromethane or Methylene Chloride, Giardia Lambia, Lead, Mercury, Nitrate, Nitrite, trans 1,2-Dichloroethylene, Tetrachloroethylene or Perchlorethylene (Perc), Trichloroethylene (TCE), Vinyl Chloride, Viruses
Cement/Concrete Plants	Barium, Benzene, Dichloromethane or Methylene Chloride, Ethylbenzene, Lead, Styrene, Tetrachloroethylene or Perchlorethylene (Perc), Toluene, Xylene (Mixed Isomers)
Chemical/Petroleum Processing	Acrylamide, Arsenic, Atrazine, Alachlor, Aluminum (Fume or Dust), Barium, Benzene, Cadmium, Carbofuran, Carbon Tetrachloride, Chlorobenzene, Copper, Cyanide, 2,4-D, 1,2-Dibromoethane or Ethylene Dibromide (EDB), 1,2-Dichlorobenzene or O-Dichlorobenzene, 1,4-Dichlorobenzene or P-Dichlorobenzene, 1,1-Dichloroethylene or Vinylidene Chloride, cis 1,2 Dichloroethylene, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl) adipate, Di(2-ethylhexyl) phthlate, 1,2-Dichloroethane or Ethylene Dichloride, Dioxin, Endrin, Epichlorohydrin, Ethylbenzene, Hexachlorobenzene, Hexachlorocyclopentadiene, Lead, Mercury, Methoxychlor,Polychlorinated Biphenyls, Selenium, Styrene, Sulfate, Tetrachloroethylene or Perchlorethylene (Perc), Toluene, 1,2,4-Trichlorobenzene, 1,1,1-Trichloroethane or Methyl Chloroform, Trichloroethylene (TCE), Vinyl Chloride, Xylene (Mixed Isomers), Zinc (Fume or Dust)
Construction/Demolition	Arsenic, Asbestos, Benzene, Cadmium, Chloride, Copper,Cyanide, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Fluorides, Lead, Selenium, Tetrachloroethylene or Perchlorethylene (Perc), 1,1,1-Trichloroethane or Methyl Chloroform, Trichloroethylene (TCE), Turbidity, Xylene (Mixed Isomers), Zinc (Fume or Dust)
Dry Cleaners/Dry Cleaning	Tetrachloroethylene or Perchlorethylene (Perc), 1,1,1-Trichloroethane or Methyl Chloroform, 1,1,2-Trichloroethane

[2] EPA maintains a chart that lists some potential facilities and activities where one might find the contaminants referred to as primary and secondary drinking water standards. The listing of a contaminant does not mean that it will always occur at the associated source, nor does it encompass all contaminants that may be present. This list is intended as a resource guide for creating an inventory list. A state or local community may have different sources of concern from the list below, based on local variability such as existing industrial activity, and known contaminant occurrence information.

Table 5.[2] Contaminants Commonly Associated With Selected Facilities and Activities.

POTENTIAL SOURCE	CONTAMINANT
Dry Goods Manufacturing	Barium, Benzene, Cadmium, Copper, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl) phthlate, Lead, 1,1,1-Trichloroethane or Methyl Chloroform, Polychlorinated Biphenyls, Tetrachloroethylene or Perchlorethylene (Perc), Toluene, Trichloroethylene (TCE), Xylene (Mixed Isomers)
Electrical/Electronic Manufacturing	Aluminum (Fume or Dust), Antimony, Arsenic, Barium, Benzene, Cadmium, Chlorobenzene, Copper, Cyanide, Carbon Tetrachloride, 1,2-Dichlorobenzene or O-Dichlorobenzene, 1,2-Dichloroethane or Ethylene Dichloride, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl) phthlate, Ethylbenzene, Lead, Mercury, Polychlorinated Biphenyls, Selenium, Styrene, Sulfate, Tetrachloroethylene or Perchlorethylene (Perc), 1,1,1-Trichloroethane or Methyl Chloroform, 1,1,2-Trichloroethane, Trichloroethylene (TCE), Thallium, Toluene, Vinyl Chloride, Xylene (Mixed Isomers), Zinc (Fume or Dust)
Fleet/Trucking/ Bus Terminals	Arsenic, Acrylamide, Barium, Benzene, Benzo(a)pyrene, Cadmium, Chlorobenzene, Cyanide, Carbon Tetrachloride, 2,4-D, 1,2-Dichlorobenzene or O-Dichlorobenzene, 1,4-Dichlorobenzene or P-Dichlorobenzene, 1,2-Dichloroethane or Ethylene Dichloride, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl) phthlate, Epichlorohydrin, Heptachlor (and Epoxide), Lead, Mercury, Methoxychlor, Pentachlorophenol, Propylene Dichloride or 1,2-Dichloropropane, Selenium, Styrene, Toxaphene, Tetrachloroethylene or Perchlorethylene (Perc), Toluene, 1,1,1-Trichloroethane or Methyl Chloroform, Trichloroethylene (TCE), Vinyl Chloride, Xylene (Mixed Isomers)
Food Processing	Arsenic, Benzene, Cadmium, Copper, Carbon Tetrachloride, Dichloromethane or Methylene Chloride, Lead, Mercury, Picloram, Tetrachloroethylene or Perchlorethylene (Perc), Toluene, 1,1,1-Trichloroethane or Methyl Chloroform, Trichloroethylene (TCE), Xylene (Mixed Isomers)
Funeral Services/Taxidermy	Glyphosate, Dichloromethane or Methylene Chloride, Nitrate, Nitrite, Total Coliforms, Viruses
Furniture Repair/Manufacturing	Barium, 1,2-Dichloroethane or Ethylene Dichloride, Dichloromethane or Methylene Chloride, Ethylbenzene, Lead, Mercury, Selenium, Trichloroethylene (TCE)
Gas Stations (see also above ground/underground storage tanks, motor-vehicle drainage wells)	cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Tetrachloroethylene or Perchlorethylene (Perc), Trichloroethylene (TCE)
Graveyards/Cemetaries	Dalapon, Lindane, Nitrate, Nitrite, Total Coliforms, Viruses.
Hardware/Lumber/Parts Stores	Aluminum (Fume or Dust), Barium, Benzene, Cadmium, Chlorobenzene, Copper, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl)adipate, Di(2-ethylhexyl) phthlate, 1,4-Dichlorobenzene or P-Dichlorobenzene, Ethylbenzene, Lead, Mercury, Tetrachloroethylene or Perchlorethylene (Perc), 1,1,1-Trichloroethane or Methyl Chloroform, Trichloroethylene (TCE), Toluene, Xylene (Mixed Isomers)
Historic Waste Dumps/Landfills	Atrazine, Alachlor, Carbofuran, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Diquat, Dalapon, Glyphosate, Dichloromethane or Methylene Chloride, Nitrate, Nitrite, Oxamyl (Vydate), Sulfate, Simazine, Tetrachloroethylene or Perchlorethylene (Perc), Trichloroethylene(TCE)

Table 5.[2] Contaminants Commonly Associated With Selected Facilities and Activities.

POTENTIAL SOURCE	CONTAMINANT
Home Manufacturing	Arsenic, Barium, Benzene, Cadmium, Chlorobenzene, Copper, Carbon Tetrachloride, 1,2-Dichlorobenzene or O-Dichlorobenzene, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl) phthlate, Ethylbenzene, Lead, Mercury, Selenium, Styrene, Tetrachloroethylene or Perchlorethylene (Perc), 1,1,1-Trichloroethane or Methyl Chloroform, Trichloroethylene (TCE), Toluene, Turbidity, Xylene (Mixed Isomers)
Industrial Waste Disposal Wells (see UIC for more information on concerns, and locations)	Acrylamide, Arsenic, Atrazine, Alachlor, Aluminum (Fume or Dust), Ammonia, Barium, Benzene, Cadmium, Carbofuran, Carbon Tetrachloride, Chlorobenzene, Copper, Cyanide, 2,4-D, 1,2-Dibromoethane or Ethylene Dibromide (EDB), 1,2-Dichlorobenzene or O-Dichlorobenzene, 1,4-Dichlorobenzene or p-Dichlorobenzene, 1,1-Dichloroethylene or Vinylidene Chloride, cis 1,2 Dichloroethylene, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl) adipate, Di(2-ethylhexyl) phthlate, 1,2-Dichloroethane or Ethylene Dichloride, Dioxin, Endrin, Epichlorohydrin, Hexachlorobenzene, Hexachlorocyclopentadiene, Lead, Mercury, Methoxychlor, Oxamyl (Vydate), Polychlorinated Biphenyls, Selenium, Styrene, Sulfate, Tetrachloroethylene or Perchlorethylene (Perc), Toluene, 1,2,4-Trichlorobenzene, 1,1,1-Trichloroethane or Methyl Chloroform, Trichloroethylene (TCE), Vinyl Chloride, Xylene (Mixed Isomers), Zinc (Fume or Dust)
Junk/Scrap/Salvage Yards	Barium, Benzene, Copper, Dalapon, cis 1,2-Dichloroethylene, Diquat, Glyphosate, Lead, Polychlorinated Biphenyls, Sulfate, Simazine, Trichloroethylene (TCE), Tetrachloroethylene or Perchlorethylene (Perc)
Machine Shops	Arsenic, Aluminum (Fume or Dust), Barium, Benzene, Boric Acid, Cadmium, Chlorobenzene, Copper, Cyanide, Carbon Tetrachloride 2,4-D, 1,4-Dichlorobenzene or P-Dichlorobenzene, 1,2-Dichloroethane or Ethylene Dichloride, 1,1-Dichloroethylene or Vinylidene Chloride, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl) phthlate, Ethylbenzene, Fluoride, Hexachlorobenzene, Lead, Mercury, Polychlorinated Biphenyls, Pentachlorophenol, Selenium, Styrene, Tetrachloroethylene or Perchlorethylene (Perc), Toluene, 1,1,1-Trichloroethane or Methyl Chloroform, 1,1,2-Trichloroethane, Trichloroethylene (TCE), Xylene (Mixed Isomers), Zinc (Fume or Dust)
Medical/Vet Offices	Arsenic, Acrylamide, Barium, Benzene, Cadmium, Copper, Cyanide, Carbon Tetrachloride, Dichloromethane or Methylene Chloride, 1,2-Dichloroethane or Ethylene Dichloride, Lead, Mercury, Methoxychlor, 1,1,1-Trichloroethane or Methyl Chloroform, Radionuclides, Selenium, Silver, Tetrachloroethylene or Perchlorethylene (Perc), 2,4,5-TP (Silvex), Thallium, Xylene (Mixed Isomers)
Metal Plating/Finishing/Fabricating	Antimony, Aluminum (Fume or Dust), Arsenic, Barium, Benzene, Cadmium, Carbon Tetrachloride, Chlorobenzene, Chromium, Copper, Cyanide, 1,4-Dichlorobenzene or P-Dichlorobenzene, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl) adipate, Ethylbenzene,Lead, Mercury, Polychlorinated Biphenyls, Pentachlorophenol, Selenium, Styrene, Sulfate, Tetrachloroethylene or Perchlorethylene (Perc), , Thallium, Toluene, 1,1,1-Trichloroethane or Methyl Chloroform, 1,1,2-Trichloroethane, Trichloroethylene(TCE), Vinyl Chloride, Xylene (Mixed Isomers), Zinc (Fume or Dust)
Military Installations	Arsenic, Barium, Benzene, Cadmium, Chlorobenzene, 1,2-Dichlorobenzene or O-Dichlorobenzene, 1,2-Dichloroethane or Ethylene Dichloride, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Hexachlorobenzene, Lead, Mercury, Methoxychlor, 1,1,1-Trichloroethane or Methyl Chloroform, Radionuclides, Selenium, Tetrachloroethylene or Perchlorethylene (Perc), , Toluene, Trichloroethylene (TCE)

Table 5.[2] Contaminants Commonly Associated With Selected Facilities and Activities.

POTENTIAL SOURCE	CONTAMINANT
Mines/Gravel Pits	Lead, Selenium, Sulfate, Tetrachloroethylene or Perchlorethylene (Perc), 1,1,1-Trichloroethane or Methyl Chloroform, Turbidity
Motor Pools	cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride,
Motor Vehicle Waste Disposal Wells (gas stations, repair shops) See UIC for more on concerns for these sources http://www.epa.gov/safewater/uic/cv-fs.html	Arsenic, Barium, Benzene, Cadmium, Chlorobenzene, Copper, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, 1,4-Dichlorobenzene or P-Dichlorobenzene, Lead, Fluoride, 1,1,1-Trichloroethane or Methyl Chloroform, Dichloromethane or Methylene Chloride, Tetrachloroethylene or Perchlorethylene (Perc), Trichloroethylene (TCE), Xylene (Mixed Isomers)
Office Building/Complex	Barium, Benzene, Cadmium, Copper, 2,4-D, Diazinon, 1,2-Dichlorobenzene or O-Dichlorobenzene, Dichloromethane or Methylene Chloride, Diquat, 1,2-Dichloroethane or Ethylene Dichloride, Ethylbenzene, Glyphosate, Lead, Mercury, Selenium, Simazine, Tetrachloroethylene or Perchlorethylene (Perc), 1,1,1-Trichloroethane or Methyl Chloroform, Trichloroethylene (TCE), Vinyl Chloride, Xylene (Mixed Isomers)
Photo Processing/Printing	Acrylamide, Aluminum (Fume or Dust), Arsenic, Barium, Benzene, Cadmium, Carbon Tetrachloride, Chlorobenzene, Copper, Cyanide, 1,1-Dichloroethylene or Vinylidene Chloride, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl) phthlate, 1,2-Dichlorobenzene or O-Dichlorobenzene, 1,4-Dichlorobenzene or P-Dichlorobenzene, 1,2-Dichloroethane or Ethylene Dichloride, 1,2-Dibromoethane or Ethylene Dibromide (EDB), Heptachlor epoxide, Hexachlorobenzene, Lead, Lindane, Mercury, Methoxychlor, Propylene Dichloride or 1,2-Dichloropropane, Selenium, Styrene, Tetrachloroethylene or Perchlorethylene (Perc), 1,1,1-Trichloroethane or Methyl Chloroform, Toluene, 1,1,2-Trichloroethane, Trichloroethylene(TCE), Vinyl Chloride, Xylene (Mixed Isomers), Zinc (Fume or Dust)
Synthetic / Plastics Production	Antimony, Arsenic, Barium, Benzene, Cadmium, Carbon Tetrachloride, Chlorobenzene, Copper, Cyanide, 1,2-Dichlorobenzene or O-Dichlorobenzene, 1,4-Dichlorobenzene or P-Dichlorobenzene, 1,2-Dichloroethane or Ethylene Dichloride, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl) adipate, Di(2-ethylhexyl) phthlate, Ethylbenzene, Hexachlorobenzene, Lead, Mercury, Methyl Chloroform or 1,1,1-Trichloroethane, Pentachlorophenol, Selenium, Styrene, Tetrachloroethylene or Perchlorethylene (Perk), Toluene, Trichloroethylene (TCE), Vinyl Chloride, Xylene (Mixed Isomers), Zinc (Fume or Dust)
RV/Mini Storage	Arsenic, Barium, Cyanide, 2,4-D, Endrin, Lead, Methoxychlor
Railroad Yards/Maintenance/Fueling Areas	Atrazine, Barium, Benzene, Cadmium, Dalapon, 1,4-Dichlorobenzene or P-Dichlorobenzene, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Lead, Mercury, Tetrachloroethylene or Perchlorethylene (Perc), Trichloroethylene (TCE).
Research Laboratories	Arsenic, Barium, Benzene, Beryllium Powder, Cadmium, Carbon Tetrachloride, Chlorobenzene, Cyanide, 1,2-Dichloroethane or Ethylene Dichloride, 1,1-Dichloroethylene or Vinylidene Chloride, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Endrin, Lead, Mercury, Polychlorinated Biphenyls, Selenium, Tetrachloroethylene or Perchlorethylene (Perc), Thallium, Thiosulfates, Toluene, 1,1,1-Trichloroethane or Methyl Chloroform, Trichloroethylene (TCE), Vinyl Chloride, Xylene (Mixed Isomers)
Retail Operations	Arsenic, Barium, Benzene, Cadmium, 2,4-D, 1,2-Dichloroethane or Ethylene Dichloride, Lead, Mercury, Styrene, Tetrachloroethylene or Perchlorethylene (Perc), Toluene, 1,1,1-Trichloroethane, Vinyl Chloride

Table 5.[2] Contaminants Commonly Associated With Selected Facilities and Activities.

POTENTIAL SOURCE	CONTAMINANT
Underground Storage Tanks	Arsenic, Barium, Benzene, Cadmium, 1,4-Dichlorobenzene or P-Dichlorobenzene, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Lead, Tetrachloroethylene or Perchlorethylene (Perc), Trichloroethylene (TCE).
Wood Preserving/Treating	cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Lead, Sulfate
Wood/Pulp/Paper Processing	Arsenic, Barium, Benzene, Cadmium, Carbon Tetrachloride, Copper, Dichloromethane or Methylene Chloride, Dioxin, 1,2-Dichloroethane or Ethylene Dichloride, Ethylbenzene, Lead, Mercury, Polychlorinated Biphenyls, Selenium, Styrene, Tetrachloroethylene or Perchlorethylene (Perc), Trichloroethylene (TCE), Toluene, 1,1,1-Trichloroethane or Methyl Chloroform, Xylene (Mixed Isomers)
Residential / Municipal	
Airports (Maintenance/Fueling Areas)	Arsenic, Barium, Benzene, Cadmium, Carbon Tetrachloride, cis 1,2- Dichloroethylene, Dichloromethane or Methylene Chloride, Ethylbenzene, Lead, Mercury, Selenium, Tetrachloroethylene or Perchlorethylene (Perc), 1,1,1-Trichloroethane or Methyl Chloroform, Trichloroethylene (TCE), Xylene (Mixed Isomers)
Apartments and Condominiums	Atrazine, Alachlor, Coliform, Cryptosporidium, Dalapon, Diquat, Giardia Lambia, Glyphosate, Nitrate, Nitrite, Picloram,Sulfate,Simazine, Vinyl Chloride, Viruses
Camp Grounds/RV Parks	Benomyl, Coliform, Cryptosporidium, Diquat, Dalapon, Giardia Lambia, Glyphosate, Isopropanol, Nitrate, Nitrite, Picloram,Sulfate,Simazine, Turbidity, Vinyl Chloride, Viruses
Cesspools - Large Capacity (see UIC for more information)	Atrazine, Alachlor, Carbofuran, Coliform, Cryptosporidium, Diquat, Dalapon, Giardia Lambia, Glyphosate, Nitrate, Nitrite, Oxamyl (Vydate), Picloram,Sulfate,Simazine, Vinyl Chloride, Viruses
Drinking Water Treatment Facilities	Atrazine, Benzene, Cadmium, Cyanide, Fluoride, Lead, Polychlorinated Biphenyls, Toluene, Total Trihalomethanes, 1,1,1-Trichloroethane or Methyl Chloroform
Gas Pipelines	cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Tetrachloroethylene or Perchlorethylene (Perc), Trichloroethylene or TCE
Golf Courses and Urban Parks	Arsenic, Atrazine, Benzene, Chlorobenzene, Carbofuran, 2,4-D, Diquat, Dalapon, Glyphosate, Lead, Methoxychlor, Nitrate, Nitrite, Picloram, Simazine, Turbidity
Housing developments	Atrazine, Alachlor, Coliform, Cryptosporidium, Carbofuran, Diquat, Dalapon, Giardia Lambia, Glyphosate, Dichloromethane or Methylene Chloride, Nitrate, Nitrite, Picloram, Simazine, Trichloroethylene (TCE), Turbidity, Vinyl Chloride, Viruses
Landfills/Dumps	Arsenic, Atrazine, Alachlor, Barium, Benzene, Cadmium, Carbofuran, cis 1,2 Dichloroethylene, Diquat, Glyphosate, Lead, Lindane, Mercury, 1,1,1-Trichloroethane or Methyl Chloroform, Dichloromethane or Methylene Chloride, Nitrate, Nitrite, Picloram, Selenium, Simazine, Trichloroethylene (TCE)
Public Buildings (e.g., schools, town halls, fire stations, police stations) and Civic Organizations	Arsenic, Acrylamide, Barium, Benzene, Beryllium Powder, Cadmium, Carbon Tetrachloride, Chlorobenzene, Cyanide, 2,4-D, 1,2-Dichlorobenzene or O-Dichlorobenzene, 1,4-Dichlorobenzene or P-Dichlorobenzene, Dichloromethane or Methylene Chloride, Di(2-ethylhexyl) phthlate, 1,2-Dichloroethane or Ethylene Dichloride, Endothall, Endrin, 1,2-Dibromoethane or Ethylene Dibromide (EDB), Lead, Lindane, Mercury, Methoxychlor, Selenium, Toluene, 1,1,1-Trichloroethane or Methyl Chloroform, Trichloroethylene (TCE), Vinyl Chloride, Xylene (Mixed Isomers)

Table 5.[2] Contaminants Commonly Associated With Selected Facilities and Activities.

POTENTIAL SOURCE	CONTAMINANT
Septic Systems	Atrazine, Alachlor, Carbofuran, Coliform, Cryptosporidium, Diquat, Dalapon, Giardia Lambia, Glyphosate, Nitrate, Nitrite, Oxamyl (Vydate), Picloram, Sulfate, Simazine, Vinyl Chloride, Viruses
Sewer Lines	Coliform, Cryptosporidium, Diquat, Dalapon, Giardia Lambia, Glyphosate, Nitrate, Nitrite, Oxamyl (Vydate), Picloram,Sulfate,Simazine, Vinyl Chloride, Viruses
Stormwater infiltration basins/injection into wells (UIC Class V), runoff zones	Atrazine, Alachlor, Coliform, Cryptosporidium, Carbofuran, Chlorine, Diquat, Dalapon, Giardia Lambia, Glyphosate, Dichloromethane or Methylene Chloride, Nitrate, Nitrite, Nitrosamine, Oxamyl (Vydate), Phosphates, Picloram, Simazine, Trichloroethylene(TCE), Turbidity, Vinyl Chloride, Viruses
Transportation Corridors (e.g., Roads, railroads)	Dalapon, Picloram, Simazine, Sodium, Sodium Chloride, Turbidity
Utility Stations	Arsenic, Barium, Benzene, Cadmium, Chlorobenzene, Cyanide, 2,4-D, 1,4-Dichlorobenzene or P-Dichlorobenzene, 1,2-Dichloroethane or Ethylene Dichloride, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Lead, Mercury, Picloram, Toluene, 1,1,2,2- Tetrachloroethane, Tetrachloroethylene or Perchlorethylene (Perc), Trichloroethylene (TCE), Xylene (Mixed Isomers)
Waste Transfer /Recycling	Coliform, Cryptosporidium, Giardia Lambia, Nitrate, Nitrite, Vinyl Chloride, Viruses
Wastewater Treatment Facilities/Discharge locations (incl. land disposal and underground injection of sludge)	Cadmium, Coliform, Cryptosporidium, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Dichloromethane or Methylene Chloride, Fluoride, Giardia Lambia, Lead, Mercury, Nitrate, Nitrite, Tetrachloroethylene or Perchlorethylene (Perc) Selenium, sulfate,Trichloroethylene (TCE), Vinyl Chloride, Viruses
Agricultural / Rural	
Auction Lots/Boarding Stables	Coliform, Cryptosporidium, Giardia Lambia, Nitrate, Nitrite,Sulfate,Viruses
Animal Feeding Operations/ Confined Animal Feeding Operations	Coliform, Cryptosporidium, Giardia Lambia, Nitrate, Nitrite, Sulfate, Turbidity, Viruses
Bird Rookeries/Wildlife feeding /migration zones	Coliform, Cryptosporidium, Giardia Lambia, Nitrate , Nitrite , Sulfate, Turbidity, Viruses
Crops - Irrigated + Non-irrigated	Benzene, 2,4-D, Dalapon, Dinoseb, Diquat, Glyphosate, Lindane, Lead, Nitrate, Nitrite , Picloram, Simazine, Turbidity
Dairy operations	Coliform, Cryptosporidium, Giardia Lambia, Nitrate , Nitrite,Sulfate,Turbidity, Viruses
Drainage Wells, Lagoons and Liquid Waste Disposal - Agricultural	Atrazine, Alachlor, Coliform, Cryptosporidium, Carbofuran, Diquat, Dalapon, Giardia Lambia, Glyphosate, Nitrate, Nitrite, Oxamyl (Vydate), Picloram,Sulfate,Simazine, Vinyl Chloride, Viruses
Managed Forests/Grass Lands	Atrazine, Diquat, Glyphosate, Picloram, Simazine, Turbidity
Pesticide/Fertilizer Storage Facilities	Atrazine, Alachlor, Carbofuran, Chlordane, 2,4-D, Diquat, Dalapon, 1,2-Dibromo-3-Chloropropane or DBCP, Glyphosate, Nitrate, Nitrite, Oxamyl (Vydate), Picloram, Simazine, 2,4,5-TP (Silvex)
Rangeland/Grazing lands	Coliform, Cryptosporidium, Giardia Lambia, Nitrate, Nitrite, Sulfate, Turbidity, Viruses
Residential Wastewater lagoons	Atrazine, Alachlor, Carbofuran, Coliform, Cryptosporidium, Diquat, Dalapon, Giardia Lambia, Glyphosate, Nitrate, Nitrite, Oxamyl (Vydate), Picloram,Sulfate,Simazine, Vinyl Chloride, Viruses

Table 5.[2] Contaminants Commonly Associated With Selected Facilities and Activities.

POTENTIAL SOURCE	CONTAMINANT
Rural Homesteads	Atrazine, Alachlor, Carbofuran, Coliform, Cryptosporidium, cis 1,2-Dichloroethylene, trans 1,2-Dichloroethylene, Diquat, Dalapon, Giardia Lambia, Glyphosate, Nitrate, Nitrite,Oxamyl (Vydate), Picloram, Sulfate, Simazine, Vinyl Chloride, Viruses

MISCELLANEOUS SOURCES	
Abandoned drinking water wells (conduits for contamination)	Atrazine, Alachlor, Coliform, Cryptosporidium, Carbofuran, Diquat, Dalapon, Giardia Lambia, Glyphosate, Dichloromethane or Methylene Chloride, Nitrate, Nitrite, Oxamyl (Vydate), Picloram, Simazine, Trichloroethylene (TCE), Turbidity, Vinyl Chloride, Viruses
Naturally Occurring	Arsenic, Asbestos, Barium, Cadmium, Chromium, Coliform, Copper, Cryptosporidium, Fluoride, Giardia Lambia, Iron, Lead, Manganese, Mercury, Nitrate, Nitrite, Radionuclides, Selenium, Silver, Sulfate, Viruses, Zinc (Fume or Dust)
Underground Injection Control (UIC) Wells CLASS I - deep injection of hazardous and non-hazardous wastes into aquifers separated from underground sources of drinking water	see UIC
UIC Wells CLASS II deep injection wells of fluids associated with oil/gas production (for more detailed list of sites click here)	see UIC
UIC Wells CLASS III re-injection of water/steam into mineral formations for mineral extraction	see UIC
UIC Wells CLASS IV - officially banned. Inject hazardous or radioactive waste into or above underground sources of drinking water	see UIC

Table 6.[3] Possible Contaminating Activities (PCAs).

Risk	Activities			
	Commercial/Industrial	**Residential/Municipal**	**Agricultural/Rural**	**Other**
Very High (VH)	-Gas stations -Chemical/petroleum processing/storage -Dry cleaners -Metal plating/ finishing/fabricating -Plastics/synthetics producers	-Airports – maintenance/fueling areas -Landfills/dumps -*Septic systems - High density (>1/acre) (for groundwater sources otherwise M) -*Wastewater Treatment Plants	-* Animal Feeding Operations -* Concentrated Aquatic Animal Production Facilities -* Managed Forests (VH for surface water otherwise H)	-Underground injection of commercial/ industrial discharges -Historic gas stations -Historic waste dumps/landfills -Injection wells/dry wells/ sumps -Known contaminant plumes -Military installations -Historic Mining operations -Active Mining operations -Confirmed leaking underground storage tanks
High (H)	-Auto Body shops -Auto Repair shops -Boat services/repair/ refinishing -Chemical/petroleum pipelines -Electrical/electronic manufacturing -Fleet/trucking/bus terminals -Furniture repair/ manufacturing -Home manufacturing -Junk/scrap/salvage yards -Machine shops -Photo processing/ printing -Research laboratories -Wood preserving/ treating -Lumber processing and manufacturing -Wood/pulp/paper processing and mills -*Sewer collection systems	-Railroad yards/ maintenance/fueling areas -*Sewer collection systems -Utility stations - maintenance areas -*Wastewater Treatment Plants	-* Grazing (> 5 animals/ acre) -* Animal Feeding Operations -* Other animal operations -Concentrated Aquatic Animal Production Facilities -Other aquatic animal operations -Farm chemical distributor/ application service -Farm machinery repair -*Septic systems- low density (<1/acre) -*Lagoons/liquid wastes -Machine shops -Pesticide/fertilizer/ petroleum storage and transfer areas -Managed Forests (VH for surface water otherwise H) -Agricultural Drainage and Irrigation Wells	-Industrial discharges -Illegal activities/unauthorized dumping -Mining - Sand/Gravel -Wells- Oil, Gas, Geothermal -Salt water intrusion -*Recreational area - surface water source -Non-regulated Underground storage tanks (tanks smaller than regulatory limit) -Not yet upgraded or registered Underground storage tanks -Snow Ski Areas -Recent (< 10 years) Burn Areas -Dredging

[3] The Navy Environmental Health Center (NEHC) has prepared tables based on EPA's State Source Water Assessment And Protection Programs Guidance, identifying Possible Contaminating Activities (PCAs) associated with various potential risks for drinking water contamination. The concept is largely applicable to stormwater, as activities with the runoff potential to contaminate drinking water are also likely to impact stormwater. In addition, rising contaminated groundwater could enter an MS4. This table is taken from the NEHC's efforts. It identifies PCAs divided by land use type and risk to drinking/receiving water. The risk rankings are based on the general nature of activities and the contaminants associated with them. An asterisk (*) indicates PCAs that may be associated with microbiological contamination.

Table 6.[3] Possible Contaminating Activities (PCAs).

Risk	Activities			
	Commercial/Industrial	**Residential/Municipal**	**Agricultural/Rural**	**Other**
Moderate (M)	-Car washes -Parking lots/malls (>50 spaces) -Cement/concrete plants -*Food processing -Funeral services/ graveyards -Hardware/lumber/parts stores	-*Septic systems - High density (>1/acre) -Drinking water treatment plants -Golf courses -Housing – High density (>1 house/0.5 acres) -Motor pools -Parks -Waste transfer/ recycling stations	-* Other animal operations -Other aquatic animal operations (H in Zones for surface water, otherwise M) -Crops, irrigated (berries, hops, mint, orchards, sod, greenhouses, vineyards, nurseries, vegetables) NOTE: Drip-irrigated crops are considered Low risks. -*Sewage sludge (biosolids) land application -Fertilizer, pesticide/ herbicide application -Managed Forests (M for ground water) -Agricultural Drainage	-Above ground storage tanks -Wells - water supply -Construction/demolition staging areas -Contractor or government agency equipment storage yards -Managed forests -Freeways/state highways -Railroads -Historic railroad right-of-ways -Road right-of-ways (herbicide use areas) -Hospitals -Storm drain discharge points -Storm water detention facilities -Artificial recharge projects – nonpotable water (includes recycled, storm, and untreated imported water) -Injection wells -Spreading basins -Snow Ski Areas (H in Zones for surface water, otherwise M) -Recent (< 10 years) Burn Areas (H in Zones for surface water, otherwise M) -Dredging (H in Zones for surface water, otherwise M)
Low (L)	-*Sewer collection systems -Appliance/Electronic repair -Office buildings/ complexes -Rental yards -RV/mini storage	-*Sewer collection systems -Apartments and condominiums -Campgrounds/ Recreational areas -Fire stations -RV parks -Schools -Hotels, Motels	-Crops, non-irrigated (e.g. Christmas trees, grains, grass seeds, hay) (or drip-irrigated crops) -* Septic systems - low density (<1/acre)	-Decommissioned underground storage tanks – inactive -Upgraded and/or registered underground storage tanks – active -Roads/Streets -Artificial recharge projects – potable water-Injection wells -Artificial recharge projects – potable water -Spreading basins -Medical/dental offices/clinics -Veterinary offices/clinics -*Surface water - streams/lakes/ rivers -Wells - Monitoring, test holes, borings

APPENDIX M:

HYDSTRA DATA ENTRY PROCEDURES

Hydstra Data Entry Procedures

1. Receive Analytical Data from Laboratory

2. Import Laboratory and Field Data Into Hydstra
   E.S. Babcock Lab will send "edd"s – electronic data text files, periodically via email. The text files will be attachments to the email. A sample data file name is given for this example and will be different for each file sent by the lab.

   2.1 Match the paper copy of the lab results with the text file from the email. The paper copy of the lab results will accompany the invoices from the lab.
   2.2 Open up the text file and review its format. It should read similar to:

   "A5D1827-01 813 Bactee Sampling 04/22/2005 14:30 David Ortega 04/22/2005 16:35 Fecal Streptococcus 40 MPN/100 mL 20 SM 9230B 04/22/2005 SAMP"

      2.2.1 The first item (**A5D1827-01**) is the lab number.
      2.2.2 The Hydstra station number (**813**) is next. This number should match the Sample ID on the *Chain of Custody* form.
      2.2.3 The project name (**BACTEE Sampling**) should match the Project Name on the *Chain of Custody* form**.**
      2.2.4 The date (**4/22/05**) should match the sample date on the *Chain of Custody* form.
      2.2.5 The time (**14:30**) should match the sample time on the *Chain of Custody* form.
      2.2.6 The name (**David Ortega**) should match the name under Sampler Information on the *Chain of Custody* form.
      2.2.7 The next date (**4/22/05**) is the date that the samples were received by the lab, and should match the *Chain of Custody* form.
      2.2.8 The next time (**16:35**) is the time that the lab received the samples, and should match the *Chain of Custody* form.
      2.2.9 **Fecal Streptococcus** refers to the type of lab analysis performed by the lab.
      2.2.10 **MPN/100 mL** refers to the reporting units of the lab results.
      2.2.11 **SM 9230B** refers to the analytical method use by the lab.

   2.3 If there is a discrepancy or difference between the paper copy of the lab report and the electronic lab data, contact Rebekah Guill ~~Arlene Chun~~ (951-955-2901 ~~951-955-1330~~) at the District and Taylor Cariaga ~~Hsin-Yi Li~~ (951-653-3351) at the lab.
   2.4 Open **Q:\NPDES\LABORATORY REPORTS\Babcock Data 04-05** (the reporting year at the end of the filename may be different).
   2.5 If there are no discrepancies between the paper copy of the lab report and the electronic lab data, then copy the text file and paste to the file **Babcock Data 04-05** in **Q:\NPDES\LABORATORY REPORTS.** Make a note of the entire text file name, *e.g.*: **A5D1827 FINAL SIMPLE 27 Apr 05 10481.txt**
   2.6 Log on to the Hydstra database, double click on file: **"Programs by Function"**, double click on file: **"Hydstra/WQ – Water Quality"**, double click on **"RFCWQIN – Import WQ Samples from lab"**. Wait for Window labeled **"RFCWQIN – Import WQ Samples from lab"** appears. Click **"Program"** tab, click **"Previous run F8"**. Insert Data File Name: **Q:\NPDES\LABORATORY REPORTS\Babcock Data 04-05\.: A5D1827 FINAL SIMPLE 27 Apr 05 10481.txt**, Insert Work file name: **A5D1827**, Insert Date Format: **B** (for Babcock), click **RUN**.
   2.7 Successful import will produce a page on the HYEXPLORE window. The page will read: **Perl – opening input file [Q:\NPDES\LABORATORY REPORTS\Babcock**

Data 04-05\.: A5D1827]. The next line will read: **Perl – Preprocessor read** a number **lines, wrote** same number **lines**. The following line will read: **Summary for Samples**.

2.8 Double click on **WQWRK – WQ Samples and Results Management**.

2.9 Click on work file: **A5D1827** to highlight it, then click on **OK**.

2.10 Click on the **Toggle browse/form view** tab for each site number listed in the work file. Check the site number, sample number, collection date and time. Match this information to the *Chain of Custody* form, *Field Data Sheet* and the lab results. Perform a check off procedure as you match the information.

2.11 The Lab Sample number should match the Sample number on this page for Dry Weather or Wet Weather sampling (WWR, SMR and SAR). For TMDL samples, the sample number will begin with an S (example: S010205O5A).

2.12 To manually enter in the Field Data, click on **RESULTS – Water Quality Results (work area)** while in WQWRK Manage.

2.13 Click on the green plus symbol button at the top left to **"Add a New Record"**.

2.14 Enter in a three or four digit numerical variable from *Field Data Sheet*:

- 2.14.1 **262: CFS, Discharge (Flow) Q**
- 2.14.2 **1690: Turbidity (NTU)**
- 2.14.3 **1200: Conductivity (µS/cm or mS/cm)**
- 2.14.4 **1435: Dissolved Oxygen, Field Concentration (mg/L)**
- 2.14.5 **1515: ORP (Redox Potential) Field (mV)**
- 2.14.6 **1655: Temperature Field (^{o}C)**
- 2.14.7 **1660: Temperature Field (^{o}F)**
- 2.14.8 **1705: pH, Field**
- 2.14.9 **8998: IC/ID sample; Value: 1 = Yes; 0 = No**
- 2.14.10 **8999: Visited not Sampled; Value: 1 = Yes; 0 = No**
- 2.14.11 **9000: Visited (Sampled); Value: 1 = Wet weather; 0 = Dry weather**

2.15 Enter in value and enter in a comment (if needed).

2.16 Double click on **WQ Manage – WQ Archive Management**

2.17 Highlight a site number that matches with the work file, then click **SAMPLES – Water Quality Samples**.

2.18 Click the **Options** button at the top.

2.19 Click on **Import a WQ workfile**.

2.20 Click on the Workfile number to import into **Archive**.

2.21 Successful Archiving will allow you to go to the site number(s) and check the sample number and results as a double check against the *Chain of Custody* form, *Field Data Sheets* and the paper copy of the lab results.

APPENDIX N:

IC/ID AND IDDE FORMS

IC/ID Incident Reporting Form

Riverside County Flood Control & Water Conservation District

Illicit Connection / Illegal Discharge Incident Reporting Form

Received by: _____
Date: _____ Time Received: _____
Complaint Routed to: _____

Reporting Party

Name: _____ ☐Anon. Agency: _____
Address: _____ City: _____ Zip Code: _____
Phone: _____ Ext.: _____ Pager/Cell: _____ e-mail: _____

Incident Location

Incident Address: _____ City: _____ Zip Code: _____
Incident Location or Business Name: _____ Thos. Bros. Page _____ Zone: ____
Incident Date: _____ Time (24-hr clock): _____ Discharge Currently Occurring: ☐Yes ☐No
Incident Description (attach add'l sheets as needed): _____
_____
_____
_____
_____ Photos Available: ☐Yes ☐No

Substance Involved

Substance Description/Chemical Name: _____
Quantity: ☐Less than ☐Greater than Amount: _____ Units: _____
Color: _____ Odor: _____ Duration of Discharge: _____
Other Details: _____
_____
Special Precautions Needed: ☐No ☐Yes _____
Other parties contacted: ☐HazMat Team ☐County Env. Health ☐County Exec. ☐City of _____
by Reporting Party ☐RWQCB ____ ☐OES – Control #_____ ☐Other _____

Containment

_____% Contained Containment Measure Used: _____
Waterbody or MS4 Involved: _____
Cleaned Up: ☐No ☐Yes, by whom _____ on Date _____ Time (24-hr) _____

Alleged Responsibility Party/Parties (If Known)

Name: _____ Business: _____
Address: _____ City: _____ Zip Code: _____
Phone: _____ Vehicle License No.: _____ Make: _____ Model: _____
Precautions Needed: ☐No ☐Yes _____

Action Needed

Investigation Required: ☐No ☐Yes Details: _____
_____
Investigation Team: Name: _____ Agency: _____ Phone No. _____
Name: _____ Agency: _____ Phone No. _____
Name: _____ Agency: _____ Phone No. _____
Name: _____ Agency: _____ Phone No. _____
Copy sent to: ☐City of _____ via ☐Mail ☐Fax ☐e-mail ☐_____

IC/ID Incident Reporting Form Instructions

A. Reporting Party
   1. *Self-explanatory.*
   2. *If the person does not want to give out their name, try to get at least their last name. If the caller hesitates in giving out a phone number, ask for a callback number. Be sure to also fill in the "Received by" information in the box above.*

B. Incident Location
   1. *Mostly self-explanatory.*
   2. *Get detailed directions to the incident location, including cross streets and Thomas Bros. map page, especially if it is in an out-of-the-way location.*
   3. *Ask the complainant if evidence of the incident is still present. If the incident occurred more than two days prior to the complaint, inform the caller that if there is no evidence of the incident nothing may be able to be done this time and request that the caller call sooner next time.*
   4. *Get as detailed a description of the incident as possible, including whether the complainant has taken photos or video. This information may be needed if formal enforcement actions will be taken.*
   5. *Let the caller know that if a District facility is not involved, you will be referring their complaint to the local jurisdiction. Thank the person for the call and tell him/her that you appreciate their helping to prevent pollution.*

C. Substance Involved
   1. *Mostly self-explanatory.*
   2. *Find out if the substance discharged has any known risks, such as toxic, respiratory irritant, etc.*
   3. *Get the names and agencies of other people the complainant contacted, if available.*
      a. *The complainant may have already notified several other people, and may feel they are getting the runaround or that their complaint is not being taken seriously. If you have to refer the complainant to another person, let the complainant know and try to get them a specific person to call.*
      b. *If another County or City employee referred the complainant to you and the referral was not appropriate, help the complainant and notify the municipal coordinator.*
      c. *The complainant may have contacted someone from a state or federal agency.*
      d. *If someone on the County Board of Supervisors is involved, the priority of response may need to be elevated, even if it is a minor issue.*

D. Containment – *Self-explanatory.*

E. Alleged Responsible Party
   1. *Mostly self-explanatory.*
   2. *Note if any precautions will be needed in approaching the alleged responsible party. If anyone at the incident site behaves in a threatening manner, leave the site and contact your supervisor for further instruction.*

F. Action Needed – *Self-explanatory.*

IC/ID Incident Investigation Report

Riverside County Flood Control & Water Conservation District

Illicit Connection / Illegal Discharge

Incident Investigation Report

Response Time: 1-6 hrs 12 hrs 24 hrs 48 hrs Other: _____

Responsible Party

Name: _____ Business: _____
Address: _____ City: _____ Zip Code: _____
Phone: _____ Ext.: _____ Pager/Cell: _____ e-mail: _____
Responsible Party Notified: ☐No ☐Yes, via ☐Mail ☐Fax _____ ☐e-mail ☐_____
Repeat Violation: ☐Yes ☐No Discharge Stopped: ☐Yes ☐No ☐Industrial ☐Commercial ☐Residential
Corrective Action Required: ☐No ☐Yes, describe _____

Outreach

Outreach Material Distributed:
☐None ☐Door Hanger ☐Business Card ☐Supplement "A" ☐Brochure _____
Other: _____

Follow-up Visit

Date: _____ Time (24-hr): _____ Investigator's Name: _____
Discharge Stopped: ☐Yes ☐No Proper Cleanup Action Taken: ☐Yes ☐No
Explain "No" answer: _____
Further Action Required: ☐No ☐Yes _____
Additional Follow-up Visit(s) Required: ☐Yes ☐No
Details: _____

Investigation

Description of Discharge and Analyses Made: ☐Attach Field Data Sheet for Additional Details
Date/Time Discharge Started: _____ Date/Time Discharge Ceased: _____ Total Amount: _____
Incident Occurred: ☐On Land ☐In Water ☐In Air Waterbody/MS4 Involved: ☐No ☐Yes _____
Substance(s) Involved: ☐ Oil & Grease ☐Soil/Sediment ☐Sewage ☐Reclaimed Water ☐Petroleum (Gas/Diesel/Jet Fuel)
☐ Chemicals _____ ☐Other _____
Photos Taken: ☐Yes (attach) ☐No Field Testing: ☐Yes ☐No Samples Collected: ☐Yes ☐No
Attach pages as needed for investigation details, photos, analyses, phone logs, meeting notes, etc.
Other parties contacted: ☐HazMat Team ☐County Env. Health ☐County Exec. ☐City of _____
☐RWQCB ____ ☐OES – Control #_____ ☐Other _____
Reason for Investigation: ☐Discharge/Spill Response ☐Citizen Complaint ☐Sewage Spill
☐Visual Monitoring ☐Construction Concern ☐Industrial Concern

Enforcement

Enforcement: ☐None ☐Verbal Warning ☐Door Hanger ☐Written Warning (attach copy)
☐Cease and Desist Order: ☐Verbal ☐Written ☐Stop Work Order
Other Enforcement Actions: _____
Investigator's Name: _____ Agency: _____ Phone No. _____
Signature: _____ Date: _____

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IC/ID Incident Investigation Report Instructions

Fill out this form as completely as possible. All of the information is required for the Annual Reports.

A. Responsible Party
   1. *Self-explanatory.*
   2. *Ensure that the "Received by" information in the box above is filled out.*

B. Outreach
   1. *Self-explanatory.*
   2. *If no outreach materials are distributed, explain why.*

C. Follow-up Visit – *Self-explanatory.*

D. Investigation – *Self-explanatory.*

E. Enforcement
   1. *Self-explanatory.*
   2. *If no enforcement action is taken, explain why.*

APPENDIX O:

MONITORING CONSULTANT APPROVAL AND RESPONSIBILITIES FORMS

Monitoring Contractor Approval and Responsibilities Form

Riverside County Quality Assurance Project Plan for Water Quality Monitoring

Contractor Name

Address	
County Monitoring Project Name and Elements	
District Agreement Number and Approval Date	
Project Manager	
Signature	
Phone Number	
E-mail	
Quality Assurance Officer	
Signature	
Phone Number	
E-mail	

The Project Manager is responsible implementing monitoring activities and data management in accordance with the requirements of this QAPP and the corresponding elements of a Project's Monitoring Plan. The Quality Assurance (QA) Officer and is responsible for quality assurance and quality control procedures for sampling and data management procedures in this QAPP. The QA officer will review and assess procedures during the project against QAPP requirements. The Project Manager will report all findings to the QA Officer and District's Project Manager, including all requests for corrective action. The QA Officer and/or District's Project Manager may stop monitoring activities, if there are significant deviations from required practices or if there is evidence of a systematic failure.

The Contractor will be responsible for ensuring QAPP/Monitoring Plan compliance by subcontractors hired on single or various elements of the Monitoring Project named above.