

The sampling point for the site runoff should be located so as to capture as much of the runoff from the test or control location as possible. (Multiple sampling points may be required if more than 10% of the site's area drains separately). This point should be situated up gradient at least above the 5-year floodway of the receiving stream.

When to Sample:

Instream sampling around the test and reference sites can be broken into three distinct stages: Pre-Construction (baseline); During Construction (active phase); and Post-Construction. In general, monitoring conducted during construction will be most useful in analyzing erosion prevention & sediment control (EPSC) methods at the test and control building sites, while the post-construction monitoring will be most useful in analyzing how the finished site designs affects site runoff quality long-term.

For the benthic macroinvertebrate monitoring, the sample collection frequency and timing should remain the same throughout all three phases of the project (Pre, During, Post construction). Benthic samples should be collected twice per year, once in the spring / high-flow season (April, May), and once in the late summer / low-flow season (late July – early October).

Instream Chemical Monitoring can be divided into two basic categories : baseflow and storm-event sampling. Baseflow samples should be collected 3 times per year, during the months of January, and May, and September, throughout all three phases of the BOB project. ****The “baseflow” of a stream is considered to be from groundwater input only, not related to direct precipitation runoff, and samples should be collected only when the stream's hydrograph has remained at a low level for at least three days. OR : Baseflow samples should be collected at least 7 days since a previous rainfall event? ****

Instream Storm-Event sampling frequency will vary over the three phases of the project. During Pre-Construction monitoring, at least 3 qualifying storm events should be collected, preferably spread apart temporally as much as is feasible. During the active construction phase, every qualifying storm event should be sampled if possible. After the permanent stabilization of a site, at least 3 post-construction qualifying events should be sampled, also spaced throughout a calendar year if possible. A “qualifying” rain event will be defined as : At least 1/2 inch of precipitation over a period of 4 hours or less, occurring at least 24 hours after the previous qualifying rain event. Storm event chemical samples should be collected within the first 30 minutes of discharge initiation from the BOB or control site into the receiving stream.

What to sample :

The following **chemical parameters** should be collected at all instream sampling points, and site runoff samples :

Total Suspended Solids	Nitrate+Nitrite	pH
Total Settleable Solids	TKN	Temperature
Turbidity	Ammonia	
	Total Phosphate	Fecal Coliform
	Dissolved Phosphate	Fecal Streptococcus

In addition, the metals Copper, Lead, and Zinc should be analyzed from selected composited storm event samples collected from the site runoff at the paired control and test construction sites.

For each chemical sample, a standardized Monitoring Form should be completed, recording various metadata such as sampler, time/date, previous and current weather, streamflow and stage (see next paragraph), stream appearance, etc.

In addition, a discharge or stage measurement of the stream or runoff should be taken concurrently with any chemical sample. For instream sampling, the preferred method would be an actual gauging of the flow. If this is not possible, an estimated flow measurement using basic cross-section x velocity methods may be used. Staff gauges should also be established at each sample point as well, and the stage recorded during each sample. During periods of high flows, this may be the only estimate of discharge volume possible. The volume of site runoff should be measured using either a calibrated weir, or other instrumentation. See Appendix B for more information.

Biological samples will consist of the collection of benthic macroinvertebrates from 2m² of productive riffle habitat (wetted long enough to establish colonization). If available, one square meter should be sampled from two separate riffles from both the upstream and downstream sample reaches (i.e. two kicks of 1m² each upstream, and two kicks of 1m² downstream). If the stream is small, a total of 2m² of riffle habitat may be spread out over multiple riffles along each sampling reach (see Biological Sampling SOP for more info). If not practical, pick samples from the bank may be used as an alternative, with careful recording of the sites where samples are collected.

In conjunction with each biological sample, a Monitoring Form should be completed, as well as a Habitat Assessment Form.

How to Sample :

1) *Sample Containers*

Biological samples may be collected in any container that will not leak the 80% ethanol preservative. A wide-mouth jar is preferable, especially with an opening large enough to stick your hand into. Mason Jars also work well.

For chemical samples, differing sizes and preservatives of sample containers will be required for the various parameters to be analyzed. Collection containers for bacteriological samples must be sterile. Nutrient samples should be preserved with sulphuric acid. Metals samples should be preserved with nitric acid. Other parameters such as solids, BOD, and should be collected in unpreserved gallon jugs. All samples bottles should be put on ice for transportation, and refrigerated until analyzed. A complete list of bottle types, preservatives, and holding times from the TDEC SOP follows. Please note that all holding times should be confirmed with the particular lab doing the analysis.

Table 2: Chemical Sample Bottles, Preservatives, and Holding Times

Sample Type	Bottle Type	Preservative	Holding Time
Fecal Coliform, Fecal Strep	Two 250mL bottles	Sodium thiosulfate (Na ₂ S ₂ O ₃)	6 hours
PH, Temp	1 liter, or directly measured	none	Test immediately
TSS, Tot Sett Solids, Turbid	1 liter or 1 gallon	none	ASAP, no longer than 48 hours
NO ₂ +NO ₃ , TKN, NH ₄ , Tot P, Diss P	500 mL plastic	1 mL sulfuric acid (H ₂ SO ₄)*	28 days
Cu, Pb, Zn	1 liter plastic	5 mL 70% nitric acid (HNO ₃)*	6 months
Oil & Grease	1 liter glass, wide mouth	2 mL sulfuric acid (H ₂ SO ₄)*	28 days
TOC	125 mL plastic	None	None specified

2) *Chemical Sample Collection Procedures*

Instream samples should be collected at mid-depth in the center of the thalweg, except for oil & grease or petroleum, which should be collected at the surface. **Always sample the most downstream point first, and work progressively upstream to avoid contamination.** Minimize use of any perfumes, lotions, bug sprays, or any other chemical compounds on the samplers hands and arms. If possible, collect samples directly into the appropriate containers, being careful not to wash out the preservative. If the stream is too deep or too swift, it may be necessary to use a swing-sampler device, or collect off a bridge with a pre-cleaned bucket. If an automatic sampler is used, the samples should be transferred to the appropriate preserved container for transport to a lab.

In streams and rivers shallow enough to safely wade, submerge the sample container directly in the water column (grab sample) to collect the sample. Wading is appropriate if the stream has a noticeable current and the samples are collected upstream of all movement. If multiple sample containers are going to be filled at the same station, fill the unpreserved sample (routine) first. Collect subsequent samples upstream of the previous sample to avoid possible contamination from the substrate or previous preservatives.

To collect a surface water sample using the sample container, wade to the middle of the channel, face upstream and collect the sample without disturbing the sediment. If sediment disturbance is unavoidable collect the sample upstream of the sediment plume or wait until the disturbed sediment moves downstream. Remove the lid without contaminating the lid or the inside of the sample container. Grasp the bottle near the base and dip it midway in the water column with a forward upstream motion. If the sample bottle contains a preservative, do not overfill it and displace the preservative. Tightly replace the lid and shake preserved bottles to assure adequate mixing of the preservative.

After collection, uniquely label each sample container in some manner so as to record the site, date, sampler, and sample type, at a minimum. All samples should then be placed in a cooler with wet ice for transport, and stored at 4 deg C until analyzed.

3) *Chemical Sample Analysis*

Chemical samples should be analyzed using methodologies and equipment consistent with the currently approved editions of *Standard Methods for Examination of Water and Wastewater*, American Public Health Association, Washington, D.C.

4) *Biological Sampling Procedures*

A. *Semi-quantitative Riffle Kick (SQKICK)*

1. Use a (two-person) one square meter kick net with a 500-micron mesh to sample the riffle. If necessary, use rocks to weight the bottom edge to prevent the flow of water beneath the net. At each site, collect two kicks: one from an area of fast current velocity and one from an area of slower current velocity. Always collect the downstream sample first to avoid organism drift. Avoid areas with large leaf packs caught on the rocks if possible. If the stream is too small to use the one meter kick net sample multiple riffles using the modified SQKICK for small streams (see Method B. below).

2. One biologist holds the net at an angle that allows the current to flow into it. making sure the bottom is in contact with the substrate and the top of the net is above the surface of the water. The second biologist disturbs the substrate for approximately one-meter distance and the width of the net (one meter) upstream of the net by kicking and shuffling the substrate. This causes organisms and debris to flow into the net. Larger rocks may be lifted and rubbed with the hands to remove clinging organisms.

3. Once the kick is completed, allow time for the lighter debris to finish floating into the net. The biologist who performed the kick then grabs the two pole ends at the bottom of the net and carefully lifts the net out of the water while the other biologist continues to hold the upper end making sure the top of the net does not dip below the water surface, thereby allowing organisms to escape. If the top of the net dips under the water and debris flows out, discard the sample and collect another kick. Carry the net horizontally to the bank for processing.

Composite the debris from both kicks. Using forceps, remove all organisms clinging to the net and add them to the sample. Thoroughly rinse the sample using sieved water to remove fine sediment. Large rocks or organic material such as whole leaves or twigs, are discarded after rinsing and removing clinging organisms. If it does not appear that a minimum of 200 organisms have been collected after 2 kicks, perform additional kicks in the same reach until at least 200 organisms are assured. Document the number and location of kicks on the field survey form and write the number of kicks on the sample tag.

Place the composited debris in a wide mouth plastic container, or other suitable container, and preserve with 80% ethanol. Include an internal tag (written in pencil on water-proof paper) with the station number, date, sampler's initials and sample type inside the container with the debris. Attach an external sample tag to the outside of the container. Instead of an external tag, the site information can be printed in indelible ink (i.e. Sharpie) on the sample lid or bottle. The external tag information must include the Station ID, Stream name, location, sampler's initials, date sampled time sampled and sample type.

B. Modified SQKICK (small streams)

In extremely small streams, where riffles are less than one meter wide, collect a one person stationary kick using a single handle dip net with a 500-micron mesh.

Sample 4-8 separate riffle locations. Starting with a downstream riffle, hold the net perpendicular to the flow making certain the bottom of the net is in contact with the substrate at all time. Disturb the substrate upstream of the net for an area approximately 18 inches long and the width of the net. Do not allow the net to move during the kick as it might cause organisms to drift under the net. Once the kick is complete, allow time for all debris to finish flowing into the net. Repeat this procedure until a total riffle area of approximately 2m² has been sampled.

Composite the debris from all kicks. Use forceps, to remove all organisms clinging to the net and add them to the debris. Thoroughly rinse the sample using sieved water to remove fine sediment. Large rocks or organic material such as whole leaves or twigs, are discarded after rinsing and removing clinging organisms. If it does not appear that 200 organisms are in the composited sample, collect additional kicks and add them to the composite. Document the total number and area of kicks on the sample tag and on the field survey form.

Place the composited debris in a wide mouth plastic container and preserve with 80% ethanol. Include an internal tag with the station number, date, sampler's initials and sample type inside the container with the debris. Attach an external sample tag to the outside of the container. Instead of an external tag, the site information can be written in indelible ink (i.e. Sharpie) on the sample lid. The external tag information must include the Station ID, Stream name, location, sampler's initials, date sampled time sampled and sample type.

5) *Biological Sample Processing*

All semi-quantitative samples are to be reduced to a 200+/- 20% (160-240) organism subsample using the following technique. This method comes directly from section 7.3 (pages 7-9) of the 1999 guidance, Rapid Bioassessment Protocols for Use in Wadeable Streams and Rivers (EPA 841-B-99-002).

Thoroughly rinse the sample in a 500-micron mesh sieve to remove preservative and fine sediment. Large organic material (whole leaves, twigs, etc.) not removed in the field should be rinsed, visually inspected and discarded. It may be necessary to soak the sample contents in water for about 15 minutes to hydrate the benthic organisms, which will prevent them from floating on the water surface during sorting. If the sample was stored in more than one container, the contents of all containers for a given sample should be combined at this time. Gently mix the sample by hand while rinsing to make it homogenous.

Transfer the cleaned sample to a gridded pick subsampler (or similar apparatus). The subsampler is a white plastic cutting tray that measures approximately 18" x 12½" x 2¼." The tray is divided into 28, 2"x2" grids and marked with indelible ink. Note: it is preferable that a sieve insert or raised grid divider be used to separate the grids. Remove the animals and debris using a combination of scoop and transfer pipette.

If the debris will not fit in one tray, use two or more trays. Thoroughly mix the debris and divide equally between the trays. Sort the same grids for both trays. For example, if grid # 5 is randomly selected, both # 5 grids are picked. This will count as one grid out of 28.

Add enough water to evenly distribute the debris. Gently shake and swirl the tray until the organisms are evenly distributed within the tray. Remove the excess water with a suction device (i.e. turkey baster with a 500 micron or smaller screen over the aperture), to the point where the sample is settled onto the bottom of the tray. If a raised grid insert is not being used, care should be taken not to pull organisms towards the area of suction.

Randomly select four numbers corresponding to squares (grids) within the gridded subsampling pan. Remove all material (organisms and debris) from the four grids and place the material into a dish or jar with a small amount of water. Use a magnifying light to make sure all organisms and debris were removed from the grids. Any organism that

is lying over a line separating two grids is considered to be on the grid containing its head. If it is not possible to determine the location of the head (i.e. oligochaetes), the organism is considered to be in the grid containing most of the body.

If there appears to be 160-240 organisms (cumulative of the four grids) then subsampling is completed. If there appears to be fewer than 160 organisms, continue selecting grids one at a time until between 160 and 240 organisms are selected. If more than 240 organisms are contained in the first four grids, transfer the contents of the four grids to a second gridded pan. Randomly select grids for this second subsampling as was done for the first, sorting grids four (and then one) at a time until the second subsample contains 160-240 organisms. If it is estimated that the first four grids of the second subsample contain more than 240 organisms, transfer the four grids to another pan and conduct a third subsample. Continue creating subsamples until there are 160-240 organisms.

Transfer the subsample, a small amount at a time to a petris dish for sorting (removing organisms). Complete all sorting under a dissecting scope, removing and preserving all organisms in 80% ethanol. If the number of organisms from the four-grid subsample does not equal the specified number of 160-240, randomly choose a fifth grid and pick out all organisms in that grid. If the addition of the fifth grid fulfills the quota, than the subsampling is complete. If not, choose additional grids (one at a time) until the quota is reached or surpassed. All the organisms from the final grid that is randomly selected are removed even if the quota is reached midway through the picking of the grid.

If, after microscopic sorting, more than 240 organisms are found, transfer all organisms to a small gridded dish (36 grids). Subsample by groups of first four and then one random grid until the target of 160-240 organisms is achieved.

8. Place the sorted debris in a separate container and preserve in 80% ethanol. Include both external and internal tags (Figures 2 and 3). Add the words “sorted debris” to the standard information on the tag. Save the remaining unsorted sample debris residue in a separate container labeled “sample residue”. This container should include the original sample label and internal tag.

9. Place the sorted 160-240 organism subsample into a glass vial and preserve in 80% ethanol. Place an internal tag written in pencil on waterproof paper citing the log number, station number, date collected and taxonomist inside each vial (Figure 4). Position the label so it can be read through the vial.

Chironomids and oligochaetes are mounted on slides in a permanent mounting media (i.e. CMC-10). Slides are labeled with the station id, date collected and initials of the taxonomist and slide box number.

10. After sorting is completed, record the appropriate information (log number, station ID, sorters initials, date sorted and the number of organisms found) in the QC logbook (Figure 8, Section II-C).

11. Identify all organisms to genus except Acari, Nematoda, Hydra, Brachiobdellida, immature Tubificidae, Lumbriculidae and Nematomorpha.

6) *Biological Data Analysis*

Using the raw benthic data from the semi-quantitative subsample, calculate a numerical value for at least each of the seven following biometrics. Calculate all biometrics using taxa identified to the genus level except for specified taxa (Acari, Branchiobdellida, Nematomorpha, Nematoda, Hydra, immature Tubificidae, Lumbriculidae) or those too young or too damaged to identify to this level. Species identification is not to be used.

a. **EPT** (Ephemeroptera Plecoptera Trichoptera Richness)

Total the number of genera within the orders Ephemeroptera, Plecoptera and Trichoptera. Taxa that could only be identified to family are included only if they are the only taxon found in that family or it is probable that they are distinct from other taxa identified to genus within the family.

b. **TR** (Taxa Richness)

Total the number of distinct genera found in the subsample. Taxa that could only be identified to family are included only if it is probable that they are distinct from other taxa identified to genus within the family.

c. **%OC** (Percent oligochaetes and chironomids)

$$\%OC = \frac{\text{Total number of Oligochaeta} + \text{Chironomidae}}{\text{Total number of individuals in the subsample}} \times 100$$

d. **% EPT** (EPT Abundance)

$$\% \text{ EPT} = \frac{\text{Number of Ephemeroptera} + \text{Plecoptera} + \text{Trichoptera}}{\text{Total number of individuals in the subsample}} \times 100$$

e. **NCBI** (North Carolina Biotic Index)

$$NCBI = \sum \frac{x_i t_i}{n}$$

where: x_i = number of individuals within a taxon
 t_i = tolerance value of a taxon (Appendix C)
 n = total number of individuals in the subsample

f. **% Dominant** (Percent contribution of the single most dominant taxon)

$$\% \text{ Dominant} = \frac{\text{Total individuals in the single most dominant taxon}}{\text{Total individuals in the sample}} \times 100$$

- g. **% Clingers** (Percent contribution of organisms that build fixed retreats or have adaptations to attach to surfaces in flowing water)

A list of taxa designated as clingers is located in Appendix C.

$$\% \text{ Clingers} = \frac{\text{Total number of clinger individuals}}{\text{Total individuals in the sample}} \times 100$$

APPENDIX A - Habitat Assessment Guidelines for using Standard EPA Habitat Form

Evaluate all ten habitat parameters. Base score on a scale of 0 to 20 for each parameter, with 20 being the highest attainable score. Scores are divided into four categories (optimal, suboptimal, marginal and poor) with a range of five scores possible in each category. Specific guidance for scoring is located on the habitat sheets. The parameters that are evaluated in each sample reach are:

1. Epifaunal Substrate/Available Cover (high and low gradient streams)

Estimate the relative quantity and variety of natural structures in the stream such as cobble riffles, large rocks, fallen trees, logs and branches, and undercut banks that are available as refugia, feeding, spawning or nursery functions for macroinvertebrates and fish. Do not count “newly fallen trees and unstable habitats.

- 2a. Embeddedness (high gradient streams)

Estimate the percent that rocks (gravel, cobble, and boulders) and snags are covered or sunken into the silt, sand, or mud of the stream bottom. To avoid confusion with sediment deposition (another habitat parameter), observations of embeddedness should be taken in the upstream and central portions of riffles and cobble substrate areas.

- 2b. Pool Substrate Characterization (Low gradient)

Evaluate the type and condition of the bottom substrate in the pools. Firmer sediment such as gravel and sand, and rooted aquatic plants support a wider variety of organisms and should be scored higher than a pool substrate dominated by mud or bedrock with no plants. In addition, a stream that has a uniform substrate will support fewer types of organisms and should score lower than a stream that has a variety of substrate types.

- 3a. Velocity/Depth Combinations (high gradient)

Determine the patterns of velocity and depth. The four basic patterns are slow-deep, slow-shallow, fast-deep, and fast-shallow. The best streams will have all four patterns present. The general guidelines are 0.5 meter depth to separate shallow from deep and 0.3 m/sec to separate fast from slow.

3b Pool Variability (low gradient)

Rate the overall mixture of pool types found in the stream, according to size and depth. The four basic types of pools are large-shallow, large-deep, small-shallow, and small-deep. A stream having many different pool types will support a wider variety of aquatic species and should score higher. General guidelines are any pool dimension (length, width, oblique) greater than half the cross-section of the stream for separating large from small and 1 meter depth separating shallow and deep.

4. Sediment Deposition (high and low gradient)

Estimate the amount of sediment deposition. This is observable through the formation of islands, point bars (areas of increased deposition at the beginning of a meander that increase in size as the channel is diverted toward the outer bank) or shoals. Determine whether pools and runs are filling in. Usually deposition is evident in areas that are obstructed by natural or manmade debris and areas where the stream flow decreases, such as bends or pools.

5. Channel Flow Status (high and low gradient)

Estimate the degree to which the channel is filled with water. When water does not cover much of the streambed, the amount of suitable substrate for aquatic organisms is limited and the stream should score lower.

6. Channel Alteration (high and low gradient)

Determine how much, if at all, the stream has been altered. Channel alteration is present when artificial embankments, riprap, and other forms of artificial bank stabilization or structures are present; when the stream is very straight for significant distances; when dams and bridges are present; when dredging or gravel removal is evident and when other such artificial changes have occurred. Scouring is often associated with channel alteration.

7a. Frequency of Riffles or Bends (high gradient)

Determine the pattern of stream morphology by estimating the sequencing of riffles. In high gradient streams where distinct riffles are uncommon, a run/bend ratio can be used as a measure of meandering or sinuosity. In headwaters, riffles are usually continuous and the presence of cascades or boulders provides a form of sinuosity. To

determine this parameter, a longer segment or reach than that designated for sampling should be incorporated into the evaluation.

7b. Channel Sinuosity (low gradient)

Evaluate the meandering or sinuosity of the stream. A high degree of sinuosity provides diverse habitat for macroinvertebrates and the stream is better able to handle surges when the flow fluctuates due to rain events. To estimate this parameter, a longer segment or reach than that designated for sampling should be incorporated into the evaluation. (This will vary by site, but should include at least two bends).

8. Bank Stability (high and low gradient)

Determine whether the stream banks are eroded or have the potential for erosion. Steep banks are more likely to collapse and suffer from erosion than are gently sloping banks, and are therefore considered less stable. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil. Each bank is evaluated separately on a scale of 0 to 10 and the cumulative score of both banks is used for this parameter. (This parameter should be evaluated within the 100-meter sample reach.)

9. Bank Vegetative Protection (high and low gradient)

Determine the amount of vegetative protection afforded to the stream bank and near-stream portion of the riparian zone. The object is to determine the ability of the bank to resist erosion as well as the ability of the plants to uptake nutrients, control instream scouring, supply food to shredders and provide stream shading. Streams that have various types (shrubs, trees etc.) of native vegetation providing full natural plant growth will score highest. In some regions, the introduction of exotics, such as kudzu, has virtually replaced all native vegetation. The value of exotic vegetation to the quality of the habitat structure and contribution to the stream ecosystem should be evaluated, generally resulting in a lower score. Each bank is evaluated separately on a scale of 0 to 10 and the cumulative score of both banks is used for this parameter. (This parameter should be evaluated within the 100-meter sample reach.)

10. Riparian Vegetative Zone Width (high and low gradient)

Estimate the width of natural vegetation from the edge of the stream bank out through the riparian zone (approximately 18 meters). Disturbance to the riparian zone occurs when roads, parking lots, fields, lawns, bare soil, or buildings are near the stream bank. Residential developments, urban centers, golf courses, pastures and row crops are common causes of degradation of the riparian zone. However, the presence of old fields (previously grazed fields, not currently in use), paths, and walkways in an otherwise undisturbed riparian zone may be judged to be inconsequential to altering the riparian zone and may be given relatively high scores. Each bank is evaluated

separately on a scale of 0 to 10 and the cumulative score of both banks is used for this parameter. (This parameter should be evaluated within the 100-meter sample reach.)

APPENDIX B - Methods and Theory of Flow Measurement and Estimate

(from : (to be added later)

APPENDIX C - Quality Control and Quality Assurance

(From : TN Division of Water Pollution Control QS-SOP for Chemical & Bacteriological Sampling of Surface Water)

Quality Assurance/Quality Control samples must be collected at least 10 percent of the surface water sampling events. The goal is to demonstrate the accuracy and precision of the chemical and bacteriological collection process, the reproducibility of the methodology, and to ensure unbiased treatment of all samples. Choose QC sites that will be representative of the all samples analyses in the 10-site QC set.

II.A. General QC Practices

- 1. Quality Team Leader (QC Coordinator)** - A centralized chemical and bacteriological QC coordinator is designated with the responsibility of ensuring that all QC protocols are met. This person will be an experienced water quality professional. Major responsibilities include monitoring QC activities to determine conformance, distributing quality related information, training personnel on QC requirements and procedures, reviewing QA/QC plans for completeness, noting inconsistencies, and signing off on the QA plan and reports.
- 2. Quality Team Member (In-house QC officer)** - One WPC staff member in each EAC will be designated as the Quality Team Member (in-house QC officer.) This person will be responsible for performing and/or ensuring that quality control is maintained and for coordinating activities with the central Quality Team Leader (QC coordinator).
- 3. Training** – There is no substitute for field experience. All samplers must have at least 6 months of field experience before selecting sampling sites. For on the job training, new employees should accompany experienced staff for as many different studies and sampling situations as possible. During this training period, the new employee needs to perform all tasks involved in sample collection under the supervision of experienced staff

II.B. Quality Control Samples

It is mandatory that Trip Blanks, Field Blanks, and Duplicate samples be collected at a minimum of 10 percent of sampling events. The QC set must be representative of all types of samples collected in a series of ten sites. Collect Equipment Blanks at 10

percent of sample collections when a discrete depth-sampling device (Kemmerer) or any reusable sampling equipment that has direct contact with sample water has been used. A Temperature Blank to measure cooler temperature must accompany every cooler delivered to the laboratory if more than two hours from collection. If any QC sample is to be analyzed for metals or mercury, the modified clean technique (Protocol C) must be employed.

1. **Trip Blanks** – The Trip Blank is used to determine if samples were contaminated during storage or transportation to the laboratory. In the EAC lab, immediately before departing for a sampling trip, fill the appropriate QC sample containers with organic-free reagent grade water. Label the tag and Sample Request Form with the associated station ID, county code, date, military time, sampler, preservative, and sample type. Write Trip Blank on the description line of the Sample Request Form and in the remarks box on the sample tag. Attach a completed sample tag to each sample container and place the trip blank samples in a zip-type colorless plastic bag. Store the Trip Blank QC sample on ice in a clean cooler. The sample is to remain sealed the remainder of the trip.
2. **Field Blanks** – The Field Blank is used to determine if contamination originated from sources not associated with the surface water conditions. At the vehicle parked near the sampling location, before collecting surface water samples, pour organic-free reagent grade water from the storage container to the sample container(s). New gallon size pre-cleaned, single use plastic bottles or glass bottles that have been properly cleaned (Section I.H) work well for the storage of organic-free reagent grade water. Label the tags and Sample Request Form as Field Blank and complete the associated station ID, county code, date, military time, sampler, preservative, and sample type. Attach a completed tag identified as Field Blank in the remarks box. Place the sample container in a zip-type colorless plastic bag and store the sample on ice in a clean cooler.
3. **Duplicate Sample** – The purpose of the duplicate sample is to determine variability of contaminants in surface water samples. Immediately after collecting a sample, fill a second sample container using the same technique. Label the tag and Sample Request Form as Duplicate and complete the station ID, county code, date, military time, sampler, preservative, and sample type. Attach the tag to the sample and place it in a zip-type colorless plastic bag and store on ice in a clean cooler until delivery to the laboratory.
4. **Temperature Blank** – A temperature blank, small bottle filled with water, is placed inside each cooler at the time the samples are stored in the cooler. When the samples are delivered to the laboratory the temperature of the sample cooler is measured in the temperature blank to ensure it is 4°C or less. Samples maintained at higher temperatures are flagged. (Note: If samples are delivered to the laboratory within 2 hours of collection, then temperatures greater than 4°C are acceptable.)

- 5. Equipment Field Blank** – After reusable equipment such as a discrete depth sampler or automatic sampler is cleaned it is necessary to demonstrate that it is contaminate free. Collect the equipment blank in the field prior to collecting the first sample. Pour a sample of organic-free reagent grade water over and through the equipment and collected the sample into the appropriate sample container. Label the tag and Sample Request Form as Equipment Field Blank and complete with the associated station ID, county code, date, military time, sampler, preservative, and sample type. Attach the tag to the sample, place the sample in a colorless zip-type plastic bag, and store on ice in a clean cooler until delivery to the laboratory.
- 6. Instantaneous Field Water Parameter QC** – Calibrate all probes each day before use. (If overnight travel is involved, the probes may be calibrated at the beginning of the trip.) Take duplicate water parameter readings at each site. If time is a constraint, duplicate readings may be reduced to the first and last site each day. To take a duplicate reading, lift the probe completely out of the water, then place it upstream of the original reading and allow the meter to equilibrate before recording results. If the readings are off by more than 0.2 units (or 10% for conductivity), repeat the procedure until reproducible results are obtained.

Upon return to the EAC Lab, perform a QC drift check on each meter at the end of the day (or at the end of the trip on multiple night trips). If the meter calibration is off by more than 0.2 for pH, DO or temperature or by more than 10% for conductivity, all readings between the initial calibration and the drift check must be marked as questionable. On the stream survey sheet and Chemical Request Form, preceded questionable readings with an N (uncertainty in result). If Chemical Request Forms have already been submitted to the laboratory, notify the central office in writing (e-mail or fax) of questionable readings. See Protocol J for additional information.

- 7. Continuous Water Parameter QC** – At 10 percent of the diurnal monitoring stations anchor a second continuous monitoring probe beside the first. After the data from both probes has been down loaded review the reading to ensure that they are within 10 percent of each other. If there is more than 10 percent difference between the two probe readings notify the supervisor and note on all associated paper work (N) that there was a calibration error. See Protocol K for additional information.
- 8. Flow Measurement QC** – Take a second flow measurement at 10 percent of the sites. The readings must be taken on the same day and in the same transect. If the original and the QC flow measurements differ by more than 10 percent, make a notation of N (uncertainty of results) on the associated paperwork. See Protocol L for additional information.