



Water Resource Evaluation Standard Operating Procedures Manual

**City of Austin
Watershed Protection & Development Review
Environmental Resource Management Division**

August, 2004

Water Resource Evaluation Section Organizational Chart

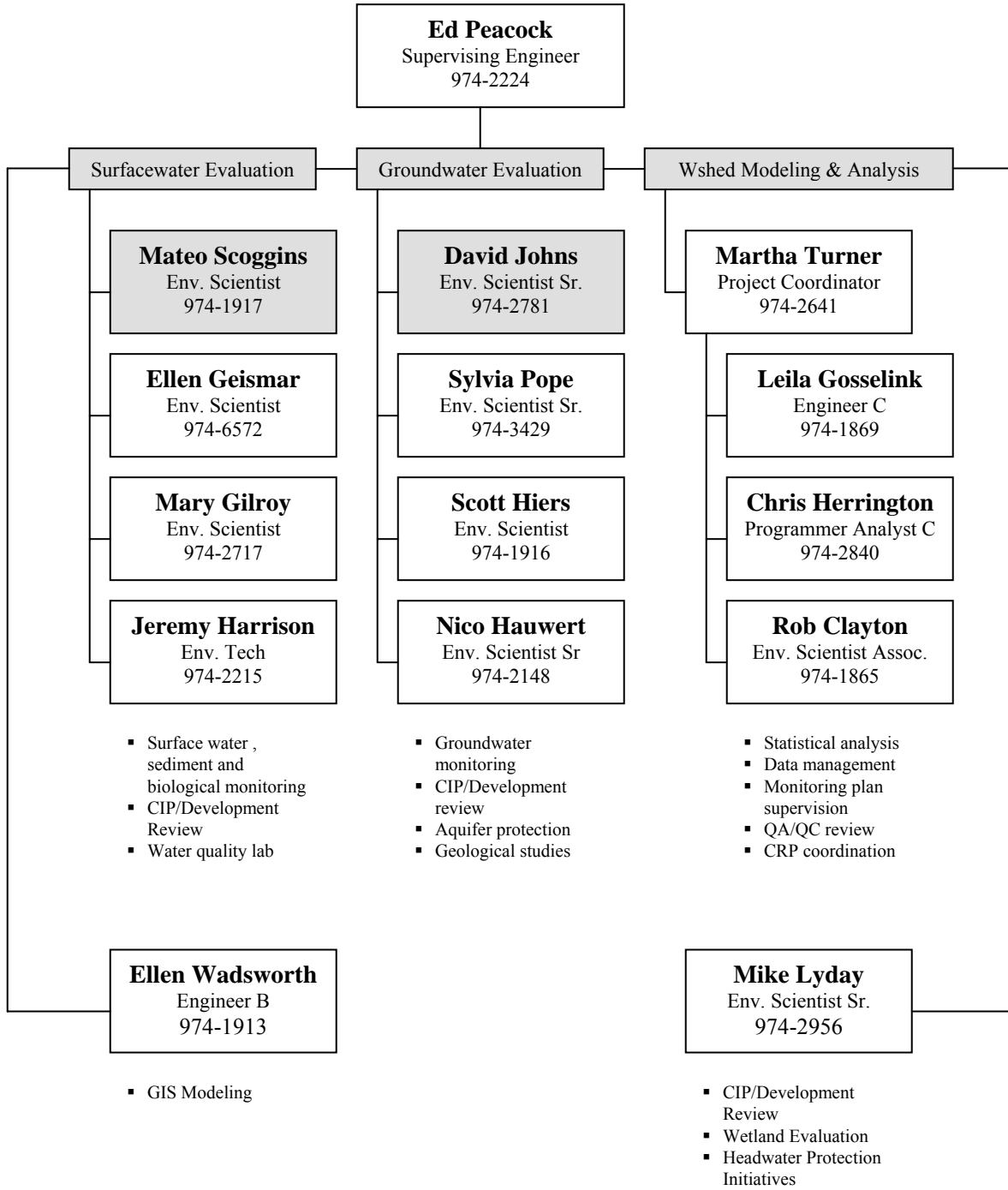


Table of Contents

Water Resource Evaluation Section Organization Chart.....	2
1.0 Purpose.....	5
2.0 Field Instrumentation.....	6
2.1 Hydrolab/Quanta Multi-probe	
2.2 Cole-Parmer pHCON10 meter	
2.3 Hach 2100 Turbidimeter	
2.4 Secchi Disk Transparency	
2.5 Marsh-McBirney Flow Meter	
2.6 Diurnal (24-hour) Field Measurements	
3.0 Water Sample Collection.....	14
3.1 Site Selection & Access	
3.2 Surface Water Sample Collection	
3.3 Groundwater Sample Collection	
3.4 QA/QC Sample Requirements	
3.5 Antecedent Flow Requirements	
3.6 Core Parameters	
3.7 Field Sheets	
3.8 Field Book Notation Guidelines	
3.9 Chain of Custody	
4.0 Sediment Sample Collection.....	21
4.1 Equipment	
4.2 Sample Collection	
4.3 Chain of Custody	
4.4 Preservation	
4.5 Routine Sediment Parameters	
5.0 Biological Sample Collection.....	23
5.1 Algae Transects	
5.2 Flow requirements & Timing of Biological Sample Collection	
5.3 Benthic Macroinvertebrates	
5.4 Diatoms	
6.0 Habitat Data Collection.....	28
6.1 Non-Contact Recreation Assessment	
6.2 EPA Habitat Quality Protocol	
6.3 Pfankuch Channel Assessment Evaluation	
6.4 Densimeter	
7.0 In-house analytical techniques.....	36
7.1 Ohmicron Nitrate	
7.2 PAH Soil Extraction & Analysis	
7.3 Hach NH ₃	
7.4 Hach OP	
7.5 TSS	

7.6 Fecal Coliform	
7.7 Scale Operation	
7.8 Glassware washing procedure	
7.9 Shelf-life tracking of laboratory supplies	
8.0 References.....	43

1.0 Purpose

This Standard Operating Procedures Manual describes the routine activities of the Water Resource Evaluation (WRE) staff of the City of Austin Watershed Protection and Development Review Department (WPDRD).

WRE staff collect data from multiple matrices throughout the greater Austin area in fulfillment of our stated mission:

..to protect and improve water quality in Austin's creeks, lakes and aquifers for our community and aquatic life by preventing, detecting, evaluating and reducing water pollution.

Program specific data may be found in the current version of the WRE Quality Management Plan or sampling plans for any individual project.

All data collected by WRE staff should be stored in the Field Sampling Database (FSDB). Procedures for correct data input and output from the FSDB may be found in the most current version of the FSDB Reference Manual.

The standard operating procedures presented in this manual are generally derived from reference materials and professional experience. Omissions or confusion about specific monitoring procedures should be resolved by consulting the most current version of the Texas Commission on Environmental Quality (TCEQ) Surface Water Quality Procedures Manual (TCEQ 2003).

2.0 Field measurements

Collection of field data at the time of sample collection is crucial to a more complete understanding of laboratory analysis results. This section details the calibration and use of multi-probe instrumentation as well as other field instruments such as the Secchi disk and Marsh-McBirney flow meters commonly used by WRE staff. Post-calibration procedures, a check for instrument drift after field data collection, are also detailed.

All instruments should be properly calibrated prior to field deployment. For any parameter, if the instrument does not read within accuracy range after calibration, perform maintenance on the instrument and re-calibrate. If it is still not within range, take the instrument in for repair. Do not record or enter into the FSDB measurements for parameters which do not properly calibrate prior to instrument deployment. Calibration results should be recorded in the Calibration Log, located in the WRE laboratory.

Instruments should be post-calibrated where specified, and post-calibration results must be entered into the FSDB to properly qualify field data accuracy.

2.1 Hydrolab/Quanta Multi-probes

The Hydrolab and Quanta multi-probes are used not only for instantaneous field measurements of routine physical parameters for a variety of programs, but also deployed for long periods of time collecting semi-continuous (data at 5 to 30 minute intervals) data. These datasondes are also used to collect the 24-hour average dissolved oxygen measurements submitted to TCEQ as part of the Clean Rivers Program.

Calibration

Record the results of all calibration procedures in the calibration logbook located in the WRE laboratory. Consult database staff for assistance in entering calibration data into the appropriate form of the FSDB.

To begin, place the calibration cup (open at both ends) on the instrument; invert and place instrument in a clamp on the ring stand. If the instrument is going to be inverted overnight, fill the cup above the DO sensor with de-ionized water and cover with the hard white plastic cap.

To calibrate the pH probe:

1. Pour a small amount of DI water into the calibration cup, then place the soft rubber cap on top and shake well. Repeat.
2. Following the same procedure, rinse the probe with the 7.0 pH standard.
3. Fill the cup with the pH 7.0 standard, and allow time to stabilize.
4. Record the reading in the **pH initial** column in the logbook.
5. Press CALIBRATE, select pH, and press ENTER.
6. Type in the correct pH, and press ENTER, and select Y (for Yes, Save the Calibration).
7. Record the final reading in the **pH final** column in the logbook.
8. Rinse the probe well with DI water, and then repeat from step 2 above using the 10.0 pH standard.

To calibrate the conductivity probe:

1. Rinse probe twice with DI water, then place the probe into ambient air with no liquid (dry) in the calibration cup.
2. Record the stable reading in the **COND 0 initial** column in the logbook.

3. Press CALIBRATE, then select C and press ENTER.
4. Type in the correct value (0 in open air), press ENTER, and select Y (for Yes, save the calibration.).
5. Record the final reading in the **COND 0 final** column of the calibration logbook.
6. Rinse and then fill the calibration cup with the 500 $\mu\text{S}/\text{cm}$ standard. Allow time for reading to stabilize.
7. Record the stable reading in the **COND 500 initial** column of the logbook.
8. Press CALIBRATE, then select C and press ENTER.
9. Type in the correct value (500), press ENTER, and select Y (for Yes, save the calibration.).
10. Record the final reading in the **COND 500 final** column of the calibration logbook.

To calibrate the DO probe:

1. Check the DO membrane for wrinkles, tears or air bubbles and replace if necessary. (See Maintenance Section in Hydrolab manual for replacement instructions)
2. Fill the cup with water to just below the O-ring securing the DO membrane.
3. Remove any water droplets by wiping the membrane with a Kimwipe.
4. Place the plastic storage lid on the cup and allow about five minutes to stabilize. Record the water temperature (from the display) and the barometric pressure (760) in the logbook.
5. Use temperature and barometric pressure to determine the theoretical DO value from the DO nomograph (printed in the Hydrolab manual). Record this in the **DO std** (theoretical) column in the logbook.
6. Once the instrument has stabilized, record the actual DO reading in the **DO initial** column in the logbook.
7. Press CALIBRATE, select the O (for DO), and press ENTER.
8. Type in the theoretical value in mg/L, press ENTER, and then select Y (for Yes, Save the Calibration).
9. Return to the main screen and verify that the instrument is reading the calibrated value. If the logbook is set up for it, record this value in the **DO final** column.

To calibrate the depth probe:

1. Pour out any solution and hold the Hydrolab downward (probes pointing down), resting the calibration cup on the floor.
2. Record the reading in the **Depth** column.
3. Press CALIBRATE, select D (for Depth), and press ENTER.
4. Type in 0.00 (change negative to positive if necessary), press Enter, and select Y (for Yes, Save the calibration).
5. Check the depth reading to make sure it is within range.

To calibrate the turbidity probe:

1. Press the SCREEN ESCAPE key to allow the instrument display to show turbidity.
2. Rinse the probe with DIUF water several times and shake off as much residual water as possible.
3. Fill the calibration cup with DIUF, being careful not to trap bubbles.
4. Allow to stabilize, then record the turbidity reading in the **Zero T initial** column of the calibration logbook.
5. Press CALIBRATE, select T (for Turbidity) and press ENTER.
6. Type in 0.00, press ENTER, and select Y (for Yes, Save the Calibration)
7. Record the final reading in the **Zero T final** column.

Measurement

Remove the plastic screw cap from the datasonde, and attach the weight/stirrer. Place the instrument into the water so that all instruments are submerged. Turn on the datasonde, and record measurements once readings have equilibrated to ambient conditions at that site (usually once temperature and conductivity stabilize). Prior to recording field measurements, be certain that a stirrer is attached and functioning or there is a sufficient amount of velocity at the site to insure properly water circulation around sensors.

For specifics on programming and downloading datasondes in any situation, refer to Hydrolab manual “Datasonde 4 and MiniSonde User’s Manual” or “Datasonde 3 Operating Manual”.

Turn off the instrument, and replace plastic screw cap. Be sure that enough liquid remains in screw cap to keep sensors wet between readings. Use ambient site water in screw cap if tap water spills, being certain to partially fill screw with tap water (chlorine in tap water restricts algal growth on sensors) once post-calibration has been performed in the laboratory.

Post-Calibration

Post-calibration values are compared to expected ranges to determine if field measurements may be reported with confidence. If post-calibration values fall outside error limits, all data for that parameter since the last calibration is unreliable. If the instrument fails to calibrate and properly post-calibrate again, maintenance is necessary.

Post-calibration must be performed after each use of the instrument. Calibration and post-calibration should be no more than 24-hours apart.

To post-calibrate, follow the initial calibration procedures but DO NOT adjust the instrument, recording the instrument readings for each standard in the calibration log (with notations indicating post-calibration). Record the instrument reading for each standard in the **initial** column, and the standard or theoretical value in the **final** column. If field sheets allot space to record post-calibration, populate those fields with the appropriate information as well.

If the instrument is to be used on the next consecutive day, the next morning’s calibration may be used as post-calibration for the prior day’s records.

If measurements do not fall within error limits (Table 2.1), appropriate routine maintenance (changing DO membrane, cleaning sensors, etc.) should be performed. If post cal values are consistently out of limits, the instrument should be taken in for repair.

Table 2.1. Post Calibration Guidelines for Field Instruments

Field Parameter	TNRCC error limits for multi-probes	Accuracy Value		
		Hydrolab H20 (DS3,DS4)	Horriba	^a Corning M90 ^b Hach pH tester
Dissolved Oxygen	± 0.5 mg/L	± 0.2 mg/L	± 0.1 mg/L	----
Temperature	± 1 C	± 0.15 (±0.10) C	± 0.3 C	----
pH	± 0.5 su	± 0.2 su	± 0.05 su	0.02 (at room T) ^b
Sp. Conductance	± 5 %	± 1 % of range	± 1 % range	± 1.5 % range ^a
TDS	--	----	----	± 1.5 % range ^a
Depth	± 0.2 at 1 m	± 0.45 m	---	---
Turbidity	---	± 5 % of range	± 3 % range	----

2.2 Cole-Parmer pHCON10 (19825-00)

The Cole-Parmer pHCON10 meter is a useful tool for rapid assessment of pH, conductivity and water temperature.

Calibration

Calibrate the Cole-Parmer pHCON10 for pH using a two-point calibration curve.

1. Press MODE to select pH mode. It will appear in the upper right corner of the display.
2. Rinse the probe thoroughly with DI water, then with the 7.0 pH solution.
3. Immerse the probe in a small amount of calibration buffer 7.0 in a clean beaker.
4. Check the upper left corner of the display for MEAS (measuring indicator); If it is not in this mode (it would show CAL), then press CAL/MEAS to switch the display.
5. Wait until the reading is stabilized (READY in upper left of display); write this value in the pH initial column in the logbook.
6. Press CAL/MEAS to begin calibration. The primary (first line) display will show the measured reading while the smaller secondary display will show the pH standard value. If the pH standard value showing is different from the standard you are using, press the up or down arrow key to scroll to the correct value.
7. When the READY indicator shows, the reading is stable and you should confirm the calibration by pressing ENTER.
8. Press CAL/MEAS key and allow the reading to stabilize, then record the value in the pH final column of the logbook.
9. The secondary display automatically scrolls to the next buffer calibration option. If not, press the up or down arrow keys to select the next buffer.
10. Follow steps 2-9 for the second buffer 10.0 solution.

To calibrate for conductivity measurement:

1. Press MODE to select Conductivity. Make sure 'MEAS' is displaying.
2. Rinse the probe with DI water, then with the calibration standard solution.
3. Immerse the probe in the standard, then tap the probe to remove air bubbles.
4. When the READY indicator lights, record the value in the COND initial column in the logbook. Record the temperature reading (second line in display) also.
5. Press the CAL/MEAS key. The CAL indicator will show in the upper left of the display, and the first line of display will show the factory default value.
6. Press the up or down arrow keys to scroll to the value of the conductivity standard being used.
7. Press ENTER to confirm the calibration.

8. Upon calibration, the CON indicator appears and the meter switches back into measurement mode. The display shows the calibrated value. Record this in COND final column of the logbook.

Measurement

1. Turn the probe on and make sure 'MEAS' (not 'CAL') is displayed. Press MODE button until display reads MEAS.
2. Immerse the probe into the sample, stirring gently. Tap the probe to remove air bubbles.
3. When READY shows on the display, the reading is stable and may be recorded.
4. To switch from pH to Conductivity (display will read "µS"), press the MODE key

2.3 Hach 2100p Turbidimeter

The Hach 2100p turbidimeter may be used to measure turbidity (in NTU) in both the field or laboratory.

Calibration

Calibration of the Hach 2100p turbidimeter should be performed only by appropriate lab personnel.

1. Check calibration by reading the three turbidity standard cell marked:
7.25 ; Acceptable range (7.61 – 6.89)
54.5 ; (57.23 - 51.78)
490 ; (514.50 – 465.50)
The values read with each of these standards must fall within the ranges above. If they do not, notify laboratory personnel.

Measurement

1. Obtain a clean, dry cylindrical cuvet.
2. Agitate the sample whirlpak in order to fully suspend any particles that may have settled out after the sample was gathered.
3. Rinse the cylindrical cuvet with a small amount of sample. Repeat twice.
4. Fill the cuvet to its marked white line with sample water. Replace and tighten cuvet top.
5. Before placing cuvet in instrument, place one small drop of silicone oil on outside of the glassware and wipe thoroughly with black cloth. This action compensates for any distortions in the glass.
6. Place the cuvet in the meter's receptacle lining up the white arrow on sample cell to the tab on meter (i.e. arrow should be towards you).
7. Cover the cuvet with the meter cover.
8. Ensure that the meter is on and then press "READ" button.
9. If "NTU" flashes on the display, push "RANGE" button once.
10. Read and record results.
11. Wash cuvet immediately in soapy water, rinse with tap water three times, then with deionized water three times according to approved bottle washing procedures.

2.4 Secchi Disk Transparency

A weighted, 20 cm black-and-white Secchi Disk is used to assess transparency of standing water (TCEQ 2003). If possible, do not use in direct sunlight, or while wearing sunglasses as the glare will inhibit correct measurements. If the current prevents the Secchi Disk from dropping straight

down, additional weight should be added. The disk is lowered into the water slowly, until it disappears from view, and then raised until it reappears. The secchi disk transparency is the mean of these two values.

2.5 Marsh-McBirney Flow Meter

Instream flow measurement with the Marsh-McBirney Flow Meter is ideally performed in a straight, flat laminar channel free of large rocks, weeds and protruding obstructions. When these qualities are not present, it may be necessary to modify the channel cross-section to create more acceptable conditions. Building dikes to reduce dead water and shallow flows or removing rocks and debris from two meters upstream of the measurement transect is acceptable.

To begin, stretch a measuring tape across the stream at a right angle to the direction of flow. Measure and record the wetted stream width, and determine the number of flow measurements to be made. The preferable number of flow measurements is 20-30. If the stream is greater than 5 feet (1.5 meters) wide, at least 10 measurements must be taken. If the stream is less than 5 feet, use flow sections of 0.5 feet (0.15 meters). Although the flow measurements do not have to be equal width, no flow measurement section should have greater than 10% of the total flow. Where flow is deeper or faster, it is good to measure smaller and more numerous sections

Record all pertinent information on the appropriate field sheet (Figure 2.1), including:

- Date and time
- Site name
- Staff measuring flow
- Current and antecedent weather conditions, including an estimation of flow severity (flow type)
- Total transect width
- All specifics (width, depth, velocity) for each measurement

To measure flow, record the width of the first measurement section and then use the wading rod to measure the depth at the mid-point of that section to the nearest 0.01 feet. Adjust the height of the sensor to the appropriate depth. If the total depth in that section is greater than 2.5 feet, measure velocity at two different depths within that section (at depths of 0.2 times the total depth and 0.8 times the total depth), then average these two velocities. If the depth is less than 2.5 feet, measure velocity twice at the same depth and average these two velocities.

When measuring velocity, keep the rod vertical and perpendicular to both the tape and the flow. Measure velocity for a minimum of 20 seconds, and always stand in a position that least affects the velocity of the water passing the meter (at least 1.5 feet downstream).

Repeat this procedure for as many flow measurement sections as if necessary at the site. Multiply the depth, width and average velocity to calculate the flow within each section. Sum the flows for all sections to yield the total discharge.

Discharge values less than 10 ft³/s should be reported to two significant figures. Discharge values greater than 10 ft³/s should be reported to the nearest whole number, but not more than three significant figures.

Figure 2.1. Field sheet for flow data collection



WPDRD FLOW DATA FIELD SHEET

Date: _____ Time: _____ Team: _____
 Meter: _____ Instrument No. _____ Site: _____

Substrate: smooth/irregular/rough - bedrock, sand/silt, gravel, cobble, boulder
 Total Transect (ft): _____ Number of Intervals: _____ Section Intervals (ft): _____

Sec.	Width (ft)	Depth (ft)	V ₁ (fps)* 0.2 or 0.6	V ₂ (fps)* 0.8	V _{avg} ** (fps)	Discharge (cfs)	Notes
1							
2							
3							
4							
5							
6							
7							
8							
9							
10							
11							
12							
13							
14							
15							
16							
17							
18							
19							
20							
21							
22							
23							
24							
25							
Total:							

* Depth > 2.5', velocity measured at 0.2 and 0.8.
 Depth < 2.5', velocity measured at 0.6.
 ** V_{avg} = 1/2(V₁ + V₂)

2.6 Diurnal (24-hour) Field Measurements

Average measurements of dissolved oxygen are collected and submitted to TCEQ for inclusion in the 303(d)/305(b) review process by WRE staff in conjunction with the Clean Rivers Program. All procedures for 24-hour average field measurements should comply with TCEQ (2003) requirements.

All diurnal measurements for the Clean Rivers Program must occur within the index period (March 15-October 15), with at least $\frac{1}{2}$ to $\frac{2}{3}$ of measurements occurring within the critical period (July 1-September 30). Only the first 24-hours of measurements from the time of deployment may be used to calculate average values.

Flow measurements must be taken at or near the time of diurnal measurements. Flow must be greater than the published 7Q2 value (TSWQS 1997).

For shallow streams, diurnal measurements should be collected from the surface, with the main goal to best represent the conditions of that water body. Diurnal sampling within reservoirs should be conducted within the mixed surface layer.

The minimum, maximum and average parameter concentration should be entered into the FSDB along with the number of measurements used to generate those summary statistics.

Datasondes must properly calibrate prior to deployment, and all parameters must meet post-calibration requirements (see table 2.1).

3.0 Water Sample Collection

Water sample collection should be performed before any other activity is done at the site, unless holding time restrictions require field parameter collection first.

3.1 Site Selection & Access

Sampling locations should be chosen to best characterize water quality for a given stream reach or segment. Program specific sampling locations are listed in each QAPP.

When choosing sites, consider historical water quality data availability, location of flow gage stations, coordination with the EII and access. Sampling should be conducted at sites which can be accessed safely under most expected flow conditions. Sampling should be conducted on public property unless specific permission is received from private landowners to access sites on private property.

3.2 Surface Water Sample Collection

Prior to sample water collection, sample bottles should be labeled with (at a minimum) date, time, site name and database number, collecting staff and preservation method. Use waterproof pens (such as Sharpies) for sample bottle labeling.

For surface water samples except bacteria, sample containers should be pre-rinsed three times with sample water by putting a small amount of water in the sample bottle, loosely capping, shaking, pouring the water out, then repeating twice.

After pre-rinsing, water should be collected from the top one-third of the water column for shallow streams (no greater than one foot deep for deeper streams), being careful not to get surface debris or bottom sediments into the container. Avoid contaminating chlorophyll-a samples with pollen floating on the surface of the water. Chlorophyll-a sample collection bottles should be held upright with the mouth of the bottle facing the water surface, the bottle mouth should be submerged below the surface of the water, then turn the bottle upright to avoid direct sampling of the water surface.

Bacteria bottles do not require pre-rinsing. Leave a head space of at least ½ inch in bacteria bottles (often referred to as Bac-T bottles) to allow for agitation prior to filtering.

Where required for preservation, acid should be added to samples in the field prior to placing on ice for transport to the lab. The appropriate amount of acid should be added with a dropper, then samples should be capped and agitated to ensure adequate mixing.

All samples should be well iced to at least 4°C for transport to the lab and kept out of sunlight, making sure to meet holding times for each requested analysis.

Sample Collection with Kemmerer

Depth sampling of Town Lake is conducted using a Kemmerer device, designed to conduct discrete water sampling at depth. Additionally, the Kemmerer may be used to generate split samples for quality control purposes.

The instrument will collect three liters of standing water. Before deployment, the sampler must be opened. Pushing the metal rod flush with the top seal will open the bottom seal of the

container. The top seal can then be pulled up. Ensure that the seals stay open by holding the sampler by the rope and moving it gently. Lower the sampler to the desired depth, being sure to keep the metal messenger on your (above water) end of the rope.

When at the desired sampling depth, drop the messenger down the rope with some force to trigger the sampler to close. Sometimes a tug on the rope will also help to seal the sampler. Retrieve the sampler from depth. Use the spout on the bottom of the sampler to rinse the sample bottles at least three times each, and then fill the sample bottles. Once all sample bottles are filled, the remaining water can be released by opening the sampler.

3.3 Groundwater Sample Collection

In most springs, the near surface discharge is representative of the local groundwater. As a result, groundwater grab samples are collected at springs by directly filling the sample container as close to the spring's discharge point as possible. If the spring's discharge is very low, a sterile whirlpak (or sterile intermediate container) is used to grab and transfer aliquots of sample water into the sample container. An alternative method to using a whirlpak is to use a portable peristaltic pump with sterile tubing to fill the sample container. In both cases, extreme care is used to avoid contaminating the sample with debris. In addition, the spring's discharge is noted on the field sheet by either direct measurement or field observation estimation.

To collect a dissolved metals sample, it must be filtered in the field with peristaltic pump and in-line filter set. To minimize contamination unpowdered latex gloves are always worn during sample collection and when possible an intermediate collection bottle is not used. When the peristaltic pump is used, field equipment blanks are collected before a ambient sample is collected for 10% of sample set at a site(s) with the same tube and filter that will be used to collect the sample. This involves pumping De-Ionized Ultra Filtered (DIUF) water through the in-line filter set into sample container for analysis.

All quality control and sampling labeling protocols follow those in the TCEQ guidance (TCEQ 2003).

Immediately after collection the appropriate preservation method is followed for each sample container. Samples are stored on ice in an ice chest and if required acidified with the appropriate acid for transport to the lab. Sufficient ice is used to lower the sample temperature to less than 4°C. Samples should be kept out of sunlight.

3.4 QA/QC Sample Requirements

Each sampling program may have more stringent quality assurance/quality control measures as specified in the program-specific QAPP. However, each surface and groundwater sampling program must collect at least one split sample per program per sampling event and one field blank per program per year. Field split and blank samples should be labeled according to pre-assigned "dummy" names, listed in each QAPP. "Dup", "Blank" or "Split" should not appear on the sample label or chain of custody form for any qa/qc sample submitted to a contract laboratory. Field notebooks and data collection sheets, however, should indicate where, when and how the qa/qc sample was generated, and that data should be entered into the FSDB in appropriate comments and qc type fields.

3.5 Antecedent Flow Requirements

Baseflow conditions, determined using antecedent rainfall, must exist at all sites at the time of water quality sample collection unless specified in special studies fully documented in the program QAPP.

Average rainfall totals will be calculated using current FEWS data, available at:

<http://wpdclaytonr/surber/resources/tools/fewstools/>

Average rainfall totals will be calculated from the 24-hour, 48-hour and 72-hour periods preceding 08:00am (or estimated time of first sample collection) on the day of the sampling event and compared to the criteria (Table 3.1) to determine if baseflow conditions exist and sampling may occur. Gages used to calculate average may vary by program, and should be listed in the program-specific QAPP.

Non-baseflow (storm-influenced) flow conditions exist if the average rainfall exceeds the range during the antecedent periods specified. Field personnel will note the flow condition on field sheets, and input the flow type into the FSDB along with field and lab data.

Table 3.1. Rainfall totals within antecedent time periods used to determine storm-influenced flow conditions.

Average Rainfall in 24-Hour Period* Prior to Sampling (Inches)	Time to Wait until Baseflow Conditions Return (Hours)
≥0.10	24
≥0.25	48
>1.00	72

*24-hour period ends at 08:00am on the day of sampling.

3.6 Core Parameters

Lists of routine laboratory (Table 3.2) and field (Table 3.3) parameters were developed by WRE committee, and should be collected for all intensive study programs. Additional parameters may be collected as specified in the QAPP in conjunction with documented special studies or at USGS gages.

Additional parameters will be collected at USGS gage sites for intensive study creeks in conjunction with the rotating 3-year EII schedule. Consult the EII sampling plan for the most up-to-date list of intensive study sites where these additional parameters will be collected.

The additional parameters collected at USGS gage locations are:

TKN, Total phosphorus, COD, total As, total Cu, total Fe, total Pb, and total Zn.

Table 3.2. Routine core laboratory parameters for water samples.

Lab Parameter	Bottle/Preservative	Reason for Monitoring
Ammonia-Nitrogen, mg/L	250mL Plastic H ₂ SO ₄	NH ₃ is used by TCEQ for identifying secondary concerns; high levels of NH ₃ may be toxic to aquatic organisms; NH ₃ has multiple anthropogenic sources
Nitrate+Nitrite-Nitrogen, mg/L		NO ₃ +NO ₂ is also used by TCEQ for identifying secondary concerns in Texas waters; high levels of NO ₃ may be toxic to humans and aquatic life; NO ₃ has multiple anthropogenic sources
Total Orthophosphorus, mg/L	250mL Plastic Ice	OP is the bioavailable portion of phosphorus in water; the state standard is for dissolved OP, and additional sampling techniques (field filtering of samples) would have to be employed for this data to be considered valid for inclusion in 303(d)/305(b) assessments. Total OP can still be used as a screening tool in non-official comparisons to dissolved OP standards.
Total Suspended Solids, mg/L	500mL Plastic Ice	TSS is useful in quantifying suspended sediment loads and is required by the masterplan. BMP design criteria in TCEQ Edwards Rules (and others) are based on TSS removal
<i>Escherichia coli</i> , cfu/100mL	Bac-T Glass Ice	State standards for bacteria are shifting to E Coli.


Table 3.3. Routine core field parameters for water samples.

Field Parameter	Reason for Monitoring
Dissolved Oxygen, mg/L	Water quality standards exist for DO; despite high variability, DO is a useful screening tool in assessing water quality impairments
Water Temperature, °C	Water quality standards exist for temperature; temperature is useful in normalizing the concentrations of DO
Conductivity, µS/cm	Water quality standards exist for conductivity; conductivity does change with increasing urbanization; extreme concentrations of conductivity may be detrimental to humans and livestock
pH, standard units	Water quality standards exist for pH; extreme pH values may be harmful to aquatic life
Instream Flow, ft ³ /s	Flow must be collected with field samples not only to enable pollutant loading calculations but also to document changing patterns in flow regimes over time

3.7 Field Data Collection Forms

Field data will be collected on a standardized form (Figure 3.1). Because of the depth profiles conducted in Town Lake, an additional form (Figure 3.2) will be utilized for reservoir sampling.

Figure 3.1. Standard field data collection form.



WPRD FIELD DATA SHEET

Database Entry

Locked: P F

Date: _____

Entered by: _____

Ref. No: _____

Database No.: _____

Site Name: _____

Date: _____

Time: _____

Personnel: _____

Current Weather: _____

Project: _____

Geologic Formation: _____

Watershed: _____

QA/QC Name: _____

Blank, Split, Duplicate

Antecedent Weather: _____

Day Since Sign. Rain: (> 0.1") _____

FEWS Gauge/Other: _____

Flow Type: Baseflow (B) - Stormflow (S) - Special Events (E) - No Flow or Dry (N)
Flow Severity: Dry - No Flow (pools only) - Low Flow - Normal Flow - High Flow - Flood
Sample Type: Grab, Grab Composite **Medium:** Surface Water, Groundwater, Sediment, Soil, Other: _____

Field Parameters:

		Passed Post-Calibration				Passed Post-Calibration	
		Yes	No			Yes	No
Dissolved Oxygen	_____ mg/l	<input type="checkbox"/>	<input type="checkbox"/>	Total Dissolved Solids	_____ mg/l	<input type="checkbox"/>	<input type="checkbox"/>
Water Temp.	_____ °C or °F *			Flow	_____ gpm or cfs *		
pH	_____ Std.Units	<input type="checkbox"/>	<input type="checkbox"/>	Conductivity	_____ μS/cm	<input type="checkbox"/>	<input type="checkbox"/>
Turbidity	_____ NTUs	<input type="checkbox"/>	<input type="checkbox"/>	Dissolved Oxygen %	_____ % Sat.	<input type="checkbox"/>	<input type="checkbox"/>
Depth	_____ ft or m *			* Annual Calibration Check			

Method: Hydrolab-MiniSonde, Hydrolab-DataSonde, Hydrolab-Quanta, Cole Parmer, Other SN# or Instrument # _____
Flow : Estimate, Marsh McBirney, Measured w/ bottle, USGS gauge _____

Lab Analysis: **Laboratory Analyzing Samples:** COA-WWW, LCRA, COA-ERM, Other: _____

Nutrients	Ions	Semivolatiles	Chlorophyll-α
TSS	Pesticides	PCBs	Isotopes
Bacteria	Herbicides	TOC	Others: _____
Metals	VOAs	VSS	Others: _____

Notes: (Sampling Location, Site Description - Algae Type, Algae % Cover, Substrate Description, Flow, Water Color and Clarity, General Vegetation, Benthics, Fish, Trash, Sample Collection and Field Measurement Location, Number of Grab Samples and etc.)

Calibration Notes: Instrument Passed Post Calibration (Yes or No) : _____

(Dissolved Oxygen ±0.5 mg/l; pH ±0.5 Std.Units; Conductivity and TDS ±5%; Temp. ±1°C, annual check; Depth ±0.2 m, annual check; Turbidity ±0.5 NTUs)

Form : ERM10-2003

3.8 Field Book Notation Guidelines

The field book is not meant to be a duplicate of field sheets, which contain much more information. But, because the field sheets are eventually archived with F-locked data, the field notebooks can provide an accessible, quick overview of sampling activity by each individual.

Field notebooks should contain at a minimum the date, site name and number, initials of staff and time of visit. Additional notes can include unusual circumstances, particularly those related to data collection (or lack of it). For example, if two sites in a sampling 'run' are dry or have no flow, this information should be recorded in the field notebook. Problems with sample collection or field equipment should be noted, especially if equipment fails and data is not collected.

In addition, if the individual does field work that does not involve sampling, such as CIP reviews, notes from these site visits should also be kept in field notebooks to provide a permanent record of the visit.

3.9 Chain of Custody

Chain of custody forms provided by the contract laboratory will be completed for all samples submitted for analysis.

4.0 Sediment Sample Collection

Sediment collected in creeks and lakes should be representative of the sampling reach. Samples should be obtained from recently deposited fine bed sediments. Every effort should be made to collect sediment in depositional areas of lower hydrologic energy to insure a maximum amount of new, fine materials. Avoid hard-packed bank sediments and disturbed (scoured) areas, as these are most-likely not freshly deposited materials.

In shallow creeks, visual assessment of the entire area is possible before collecting the sample. In deeper areas such as reservoirs, visual inspection is impossible and several attempts to collect appropriate sediment may be necessary.

4.1 Equipment

Sampling equipment consists of a Teflon coated scoop, disposable pre-sterilized plastic scoops or the stainless steel petite ponar dredge for deeper areas. The selection of sampling equipment is determined by the substrate (rocky areas and small pockets of deposited materials preclude the use of the dredge), the depth of the water and the parameters for analysis (to avoid contamination).

Clean, washed sampling equipment is used. The sampling equipment is pre-rinsed at each site with native water. The equipment is rinsed between samples, removing all sediment from previous samples.

4.2 Sample Collection

A minimum of three (preferably more, depending on the amount of sediment needed) sub-samples are collected and deposited into a clean glass bowl, which is pre-rinsed at each site with native water.

For lake sampling, only the top layer, or “recent deposits” are obtained from the dredge sample as described in the most recent version of the SWQM manual (TCEQ 2003). Reservoir samples should be collected from aerobic zones, generally the uppermost 5cm of sediment.

Samples are then composited directly into properly labeled and certified clean glass containers or unused sterilized whirlpaks. Care should be taken to minimize water content of sediment samples to avoid inflation of detection limits. If necessary, slowly decant overlying water from glass bowl after fine sediments have generally settled prior to emptying sample into final container.

If sediment and water samples are to be collected during the same sampling event, water samples should be collected prior to sediment samples.

Field notation should include at a minimum:

- Type of composite (both time and space-composited?)
- Number of grabs
- Duration of sampling
- General description of the sample including color, texture and odor.

4.3 Chain of Custody

Chain of custody for sediment samples must be maintained and documented on the appropriate chain of custody forms provided by each contract laboratory.

4.4 Preservation

Place samples on ice and cool to 4°C for transport to the appropriate laboratory. Avoid exposure of sample containers to sunlight.

4.5 Routine Sediment Parameters

Routine sediment investigation parameters have been determined by committee, and may be found in the WRE core parameter documentation (WRE 2003). For convenience, the list of core sediment parameters is presented (Table 4.1). Except for specific investigations with pre-determined objectives, this list may be considered the standard analytes for all sediment samples.

A need for periodic screening of chlorophenoxy herbicides and organophosphorus pesticides has been recognized, and should occur on a project-by-project basis as suggested by data analysis.

Table 4.1 List of routine WRE sediment parameters

Category	Parameter
Nutrients	Ammonia
General	Dry Weight Percent
	Texture
	TOC
	TPH (by method TX 1005)
Metals	Arsenic
	Cadmium
	Chromium
	Copper
	Iron
	Lead
	Mercury
	Nickel
	Silver
	Zinc
Organics	PAHs
Pesticides	Organochlorine Pesticides
Other	Polychlorinated Biphenyls

5.0 Biological Sample Collection

Routine biological sampling by WRE staff includes assessment of benthic macroinvertebrates, diatoms and algae.

5.1 Algae Transects

Monitoring of the relative coverage and composition of aquatic macrophytes and algae is used as another indicator of change in water quality over time. Coverage is estimated along linear transects by standard plant ecology techniques (Barbour 1980, Blum 1957, Hynes 1970). The distance that all plants or unvegetated substrates project through the plane of the line is tallied and expressed as a percentage of the total length of the line.

Surveys are typically conducted in pools, with each pool divided by three to six equally-spaced transects depending on the length of the pool. Measurement is typically performed with two staff members. As one person records the data the other attaches the tape measure across the stream, observes the benthic cover and calls out the number of feet observed in the various categories. If waters are deep, a diving mask may be used to observe the cover.

All aquatic vascular macrophytes are identified to the genus level using Correll and Correll (1972) as a reference. Non-filamentous algae with macrophyte morphology such as *Nitella* and *Chara* species are identified by genus. Commonly encountered spongy composites of blue-green algae, diatoms, and sediment are identified as "carpet algae" if the cover has enough integrity to bind together when disturbed. Substrate will be described as silt if it breaks into fine suspended solids when disturbed.

Filamentous algae are categorized as *Cladophora* species or *Spirogyra* species types depending on their texture and branching habit, as described below. All blue-green algae are lumped together, and un-vegetated substrates are identified as one of seven categories. Five categories of un-vegetated substrate are characterized by particle size (Compton 1985), and the other two un-vegetated substrates were characterized as bedrock and leaf litter (included any dead or decomposing organic matter). Altogether, 26 commonly encountered categories of cover are listed on the field data sheet, with several blank columns available for the addition of rarely encountered plants or substrates. These categories may be lumped into four super categories for analysis purposes: un-vegetated substrate, filamentous algae, non-filamentous algae, and aquatic macrophytes.

Cladophora species algae is coarse in texture and is multi-branched in morphology, making field identification of this genus rather easy. Other non-branching, slimy textured algae are identified as a *Spirogyra* type on the data sheet.

5.2. Flow Requirements

Biological sampling must occur during baseflow conditions. For biological sampling, there must not only be a minimum flow, but also an absence of scouring flow events, prior to sampling. Antecedent flow conditions are determined by analysis of daily mean flows at USGS gages, available from the USGS website at: <http://tx.waterdata.usgs.gov/nwis/current?type=flow>.

The specific gages to be used may be different for each project, and should be clearly documented in the sampling plan for that project.

The 25th percentile of daily mean flow over the past 20 years is used as an arbitrary indication of minimum baseflow conditions. Gages used for the EII project along with the 25th percentile of flow are presented in table 5.1. The specific gages to be used for a given project must yield mean daily flow values equal to or greater than the 25th percentile value *for at least 10 consecutive days prior to biological sampling*. In addition, all gages must have had measurable (non-zero) flow for 90 consecutive days prior to biological sampling events, based on observed recolonization rates.

Table 5.1. Minimum mean daily flow required for biological sampling.

USGS Gage	Minimum Daily Mean Flow (cfs) 10 days prior to sampling
Bear Creek @ FM 1826 (08158810)	0.01
Bull Creek @ Loop 360 (08154700)	1.0
Walnut Creek @ Webberville Rd (08158600)	3.0

In addition to the minimum flow requirements, biological sampling must occur no sooner than 2 weeks after high flow scouring events. Scouring flow events are arbitrarily defined as the 90th percentile of daily mean flow over the past 20 years. If flow magnitudes exceed the 90th percentile in the previous 14 days, biological sampling must be postponed. The 90th percentile mean daily flow values for the EII gages are presented in table 5.2.

Table 5.2. Mean daily high flow values resulting in 2-week postponement of sampling.

USGS Gage	High Daily Mean Flow (cfs)
Bear Creek @ FM 1826 (08158810)	10
Bull Creek @ Loop 360 (08154700)	50
Walnut Creek @ Webberville Rd (08158600)	75

5.3. Timing of sampling events

Screening level sampling and EII sampling should be conducted in or about June, but may be adjusted as flow patterns dictate. If an especially dry spring occurs with flows dropping below the 25th percentile in March, screening level sampling events may be moved forward on the calendar, assuming that the sampling program is not Clean Rivers Program-compliant.

Intensive surveys and all Clean Rivers Program-compliant programs should have biological sample collection two times per site per year in accordance with TCEQ procedures (SWQM 2004). Both samples will be collected within the index period (March 15 to October 15), and one sample collected within the critical period (July 1 to September 30).

5.3. Benthic Macroinvertebrates

Flow, collected by standard WRE field procedures as defined in the Standard Operating Procedures Manual, will be collected at each site when biological samples are collected and documented on field sheets. An example field data sheet that could be used for benthic macroinvertebrate sample collection is presented (Figure 5.1).

Figure 5.1. Example biological sample field data sheet.

EII Benthic/Diatom Survey Field Sheet

DUID#: _____	Project: _____
Site Name: _____	Geologic Formation: _____
Date: _____	Watershed: _____
Time: _____	QA/QC Name: _____
Personnel: _____	
Current Weather: _____	Antecedent Weather: _____
	Days Since Sig. Rain (0.1"): _____
	FEWS Gauge/Other: _____
Flow Type (circle one): Baseflow (B) Stormflow (S)	Special Event (E)
Surface Flow Conditions: Flowing (F) Pooled (N)	Dry (D)

Bugs:	Composited?	YES	NO	Subsampled?	YES	NO
Grids #'s	# of organisms	Sorter	Detritus description	Notes(QA?)		
Total # of Orqs:				Total # of Surbers:		

Diatoms:			
# of Scrapings	Periphyton layer (thickness, color, etc)	Scraper	Notes(QA?)

For all surveys, count, note and remove all Corbicula from samples. Permanent paper labels describing the location and time of sample collection will be placed in all field vials.

Field teams will need the following equipment: one surber sampler (1 ft², 600µm mesh-size), one Caton sub-sampler, sample vials and a picking pan for each person who will be sorting. Three surber samples, including all detritus, will be collected from the bottom, middle and top parts of the riffle at each site.

Screening level programs and the EII are treated differently from intensive study programs like the Bull, Barton or Walnut creek studies. For screening level programs and the EII, the three surber samples will be composited in the sub-sampler, distributing material evenly through the grid. For intensive study programs such as the Bull, Barton or Walnut Creek studies, the three surbers will be preserved as three discrete samples and labeled appropriately so that they can be spatially located (a, b, c, etc., with notations on field sheets). Intensive study surbers should be picked in their entirety except as noted below for high abundance (>1000 organisms) surber samples.

Once the sample(s) have been deposited in the pan or sub-sampler, abundance is noted visually and used to determine if sub-sampling is necessary:

- For high abundance samples (>1000 organisms), one grid (out of 30) will be randomly selected. That grid will be removed and transferred to the picking pan to be completely picked. For screening level investigations and the EII program, sequential grids will be picked until the target number of 200 organisms (+/- 20%) is reached. For intensive study programs, each surber may be sub-sampled until 100 organisms per surber (for a total of 300 organisms) is reached. Additionally, intensive study surbers which are sub-

sampled should be reviewed for large and rare organisms, which are stored in a fourth vial and used in calculation of richness/diversity metrics.

- (SCREENING LEVEL ONLY) Lower abundances will require more than one grid per person picking (2-9). As a guide, if each grid has < 7 organisms, you will have to pick the whole pan to get your target. Think rapid and pull enough grids to give each picker approximately 25–50 organisms.
- (SCREENING LEVEL ONLY) Extremely low abundance samples can be picked in their entirety and supplemented with sequential surbers until the target number of organisms is reached (200 +/- 20%). However, the original 3 surber composited sample will be maintained as a discrete sample, and each subsequent surber after that initial discrete sample will be maintained separately. Thus, the 3-surber composite will be one sample in the FSDB and each subsequent surber will be one sample in the FSDB.
- (FOR INTENSIVE STUDIES ONLY) In case of an excessively depauperate site, after the first three surbers have been sorted (3 samples in DB), successive surbers (one sample each) may be collected and sorted if the field staff desires to increase total number of organisms to 100 (+/-20%) in order to calculate. However, this is not required and the three-surber sample with uniform area corresponding to other sites should always be the base data source for analysis purposes.

For all cases, the number of surbers, number of grids picked if sub-sampled, and the estimated total number of organisms should be recorded in the lower portion of the field sheet. The number of grids/surbers sub-sampled is noted along with the estimated number of organism in each grid/surber to document the level of effort (area) to reach the target number of organisms.

If Intensive and EII surveys have crossover and the data will need to be used for both purposes, the Intensive survey method should be used.

Each sorted pan of benthic macroinvertebrate samples will be checked for missed organisms by a different member of the field staff. The detritus from 1 out of every 10 samples will be preserved and returned to the lab for verification. Percent missed will be noted to monitor effectiveness of each field team.

5.4. Diatoms

Routine diatom sample collections are to be made from hard substrates (cobble) in the same riffle that is sampled for benthic macroinvertebrates. A field sheet similar to that presented for benthic macroinvertebrates (Figure 5.1) should be used.

Prior to collecting material from the stream, examine the collection equipment for debris or leftover material. Rinse everything thoroughly and remove any remaining material before beginning sampling.

At each monitoring site, three samples are taken from rocks randomly selected from the study riffle. Rocks are sampled individually and labeled **a**, **b** and **c** (from upstream to downstream) and composited into a single sample. The objective is to collect a single composite sample that is representative of the periphyton found at the site. A small petri dish (47 cm²) and sharp object are used to mark the area on the rock to be sampled. This area is then scraped with a wire brush, and the particulate matter deposited into a shallow collecting pan. A sufficient quantity of ambient creek water is used to flush the finer plant material from the substrate and the scraping tools. The contents of the pan are poured into a plastic sample bottle (approximately 125 ml) and

more water is used to flush all remaining particulate matter from the pan. Extreme care is taken to ensure that the total volume of water required to flush the sample rock, tools, and pan does not exceed the volume of the bottle. Samples are then preserved with formalin in the field, labeled appropriately with at a minimum the name of the site, the date, the collectors name and the database sample site number. The samples should be refrigerated for transport to the ERM lab as soon as possible.

NOTE: in cases where samples will not be processed within one week, they should be preserved using 2.5 mL of M-3. Alternatively, if the preservative M-3 is not available, the samples should be allowed to settle overnight, then approximately 50% of the sample water decanted and sample bottle refilled with 70 % isopropyl alcohol.

Duplicate samples will be collected from 10% of the total number of sites.

Additionally, densiometer readings will be made to estimate percent canopy cover at each riffle where diatom samples are collected. Descriptions of densiometer use appear in the habitat section of this manual (Section 6.5).

6.0 Physical Habitat

Protecting the physical integrity of waterways is a vital component of an urban watershed protection and management program. Physical habitat data is collected in conjunction with the EII as well as intensive study programs. Clean Rivers Program-compliant studies conduct habitat assessments in accordance with TCEQ protocols (SWQM 2004, RWA 1999).

6.1 Non-Contact Recreation

The aesthetics of Austin waterways are assessed for the EII program using the Non-contact Recreation Use field assessment form (Figure 6.1). This visual/sensory assessment of flow volume and water appearance is conducted through field observation of the creek and adjacent stream banks.


The area surveyed encompasses approximately 100 meters upstream of each site, 100 meters downstream and the visible area of each stream bank. Field assessment forms include a description of the conditions for six parameters in four scoring categories: Excellent, Good, Poor and Bad. Parameters are ranked on a 20-point scale that represents the overall conditions of stream bank at that site. Notes and photographs are used to document the conditions of the creek and the overall appearance of the creek at the time of the survey.

The visual/sensory assessment parameters are:

- **Clarity:** a visual assessment of turbidity.
- **Litter:** a visual survey of the litter conditions at each site.
- **Flow Volume:** description of the amount of flow occurring at each site.
- **Odor:** a sensory assessment of objectionable odors existing at each site.
- **Percent Algae Cover:** a visual assessment of algae occurring on or below the water's surface at each site.
- **Surface Appearance:** a visual assessment of the percent of organic floatables, plants and woody debris excluding algae at each site.

Figure 6.1. Non-Contact Recreation assessment field sheet.

Figure 3: Non-Contact Recreation Field Assessment Form

DATE _____ TIME _____		OBSERVER _____			
SITE _____					
Indicator	Excellent (20-16)	Good (15-11)	Fair (10-6)	Poor (5-1)	Score
Clarity	Clear to slightly cloudy, visibility mostly good	Slightly cloudy to cloudy, visibility somewhat impaired	Cloudy to very cloudy, impaired visibility	Very cloudy/murky, can't see below surface	Score: _____
Litter	Very little litter present in the creek and on the banks; no glass	Some litter present in the creeks and on the banks; mostly small items (paper, wrappers); no glass	Litter present in the creeks and on the banks; larger items (cups, containers, cans); a few pieces of glass	Large volume of litter in the creeks and on the banks; dumping; glass and/or sharp metal objects readily apparent	Score: _____
Flow volume	Steady, running flow; water reaches both channel banks	Moderately running; evidence of diminished flow; some areas of the creek channel are dry.	Slight, trickling flow, most of the creek bed is dry	Dry creek bed, isolated stagnant pools	Score: _____
Odor	No odor to very faint offensive odor (e.g. fishy, sulphurous, petroleum, dead animal)	Slight offensive odor (e.g. fishy, sulphurous, petroleum, dead animal)	Noticeable offensive odor (e.g. fishy, sulphurous, petroleum, dead animal)	Strong offensive odor (e.g. fishy, sulphurous, petroleum, dead animal)	Score: _____
Percent Algae Cover	Surface less than 10% covered by algae	Surface 10-20% covered with algae	Surface 20-30% covered by algae	Surface more than 30% covered with algae	Score: _____
Surface appearance	Surface less than 10% covered by organic floatables, plants, woody debris, oil or foam, etc...	Surface 10-20% covered by organic floatables, algae, plants, woody debris, oil or foam, etc...	Surface 20-30% covered by organic floatables, algae, plants, woody debris, oil or foam, etc...	Surface more than 30% covered with organic floatables, algae, plant woody debris, oil or foam, etc...	Score: _____
Fish Presence / Absence (yes/no) <input type="checkbox"/>					
Greenbelt/ Buffer Rating: _____ Trail Access Rating: _____					
Notes: _____ _____ _____					Total: <input type="text"/> <input type="text"/>
					

6.2 EPA Habitat Quality Protocol

The EPA Physical Habitat Quality evaluation is a visual assessment of existing erosion-related in-stream and riparian conditions conducted through field observation. The field assessment sheet is derived from Barbour and Striblings (1998), and describes four categories of conditions (optimal, sub-optimal, marginal and poor) for 10 parameters.

- **Epifaunal substrate/available cover:** the complexity and quality of available cover for benthic macroinvertebrates and fish.
- **Embeddedness:** the degree that the stream bed substrate is covered by fine sediment, based on the USGS method (USGS 1993) in which five larger-sized particles are randomly selected along a stream cross-section and rated according to the amount of their surface area covered by fine sediment.
- **Velocity/Depth regimes:** the evaluation of a diverse stream morphology characterized by the presence of different velocity regimes.
- **Sediment deposition:** the changes in a stream's morphology resulting from the erosive forces of water and the accumulation of sediments to form new surfaces. The amount and pattern of sediment deposition is indicative of the severity of stream bank erosion.
- **Channel flow status:** the volume of water present in the stream, visually estimated by comparing the width of water within the channel with the amount of exposed channel substrate.
- **Channel alteration:** the amount of human-induced modifications in the natural geometry of the stream channel including removal of meanders and reinforcing embankments of hard engineered materials.
- **Frequency of riffles:** a measure of the sinuosity of the stream based on the frequency of occurrence of riffles relative to the width of the stream
- **Bank Stability:** the current stability of the stream banks including consideration of the presence and magnitude of recent erosion or bank failure.
- **Vegetative Protection:** the amount and composition of vegetative cover present along stream banks.
- **Riparian vegetative zone width:** the width of the natural vegetative zone and amount of human influence within close proximity to the stream channel.

Figure 6.2a Habitat assessment field data sheet (front)

HABITAT ASSESSMENT FIELD DATA SHEET—HIGH GRADIENT STREAMS (FRONT)				
STREAM NAME _____		SITE _____		
DUID # _____ RIVERMILE _____		DAYS SINCE RAIN _____		
STAFF/YOU _____		DATE _____	PROJECT _____	
		TIME _____ AM PM		

Habitat Parameter	Condition Category			
	Optimal	Suboptimal	Marginal	Poor
1. Epifaunal Substrate/ Available Cover	Greater than 70% of substrate favorable for epifaunal colonization and fish cover; mix of snags, submerged logs, undercut banks, cobble or other stable habitat and at stage to allow full colonization potential (i.e., logs/snags that are not new fall and not transient).	40-70% mix of stable habitat; well-suited for full colonization potential; adequate habitat for maintenance of populations; presence of additional substrate in the form of newfall, but not yet prepared for colonization (may rate at high end of scale).	20-40% mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed.	Less than 20% stable habitat; lack of habitat is obvious; substrate unstable or lacking.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
2. Embeddedness	Gravel, cobble, and boulder particles are 0-25% surrounded by fine sediment. Layering of cobble provides diversity of niche space.	Gravel, cobble, and boulder particles are 25-50% surrounded by fine sediment.	Gravel, cobble, and boulder particles are 50-75% surrounded by fine sediment.	Gravel, cobble, and boulder particles are more than 75% surrounded by fine sediment.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
3. Velocity/Depth Regime	All four velocity/depth regimes present (slow-deep, slow-shallow, fast-deep, fast-shallow). (Sow is < 0.3 m/s, deep is > 0.5 m.)	Only 3 of the 4 regimes present (if fast-shallow is missing, score lower than if missing other regimes).	Only 2 of the 4 habitat regimes present (if fast-shallow or slow-shallow are missing, score low).	Dominated by 1 velocity/depth regime (usually slow-deep).
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
4. Sediment Deposition	Little or no enlargement of islands or point bars and less than 5% (<20% for low-gradient streams) of the bottom affected by sediment deposition.	Some new increase in bar formation, mostly from gravel, sand or fine sediment; 5-30% (20-50% for low-gradient) of the bottom affected; slight deposition in pools.	Moderate deposition of new gravel, sand or fine sediment on old and new bars; 30-50% (50-80% for low-gradient) of the bottom affected; sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools prevalent.	Heavy deposits of fine material, increased bar development; more than 50% (80% for low-gradient) of the bottom changing frequently; pools almost absent due to substantial sediment deposition.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
5. Channel Flow Status	Water reaches base of both lower banks, and minimal amount of channel substrate is exposed.	Water fills >75% of the available channel; or <25% of channel substrate is exposed.	Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed.	Very little water in channel and mostly present as standing pools.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

Figure 6.2b Habitat assessment field data sheet (back)

Habitat Parameter	Condition Category			
	Optimal	Suboptimal	Marginal	Poor
6. Channel Alteration	Channelization or dredging absent or minimal; stream with normal pattern.	Some channelization present, usually in areas of bridge abutments; evidence of past channelization, i.e., dredging, (greater than past 20 yr) may be present, but recent channelization is not present.	Channelization may be extensive; embankments or shoring structures present on both banks; and 40 to 80% of stream reach channelized and disrupted.	Banks shored with gabion or cement; over 80% of the stream reach channelized and disrupted. Instream habitat greatly altered or removed entirely.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
7. Frequency of Riffles (or bends)	Occurrence of riffles relatively frequent; ratio of distance between riffles divided by width of the stream <7:1 (generally 5 to 7); variety of habitat is key. In streams where riffles are continuous, placement of boulders or other large, natural obstruction is important.	Occurrence of riffles infrequent; distance between riffles divided by the width of the stream is between 7 to 15.	Occasional riffle or bend; bottom contours provide some habitat; distance between riffles divided by the width of the stream is between 15 to 25.	Generally all flat water or shallow riffles; poor habitat; distance between riffles divided by the width of the stream is a ratio of >25.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
8. Bank Stability (score each bank) Note: determine left or right side by facing downstream.	Banks stable; evidence of erosion or bank failure absent or minimal; little potential for future problems. <5% of bank affected.	Moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion.	Moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; obvious bank sloughing; 60-100% of bank has erosional scars.
SCORE (LB)	Left Bank 10 9	8 7 6	5 4 3	2 1 0
SCORE (RB)	Right Bank 10 9	8 7 6	5 4 3	2 1 0
9. Vegetative Protection (score each bank)	More than 90% of the streambank surfaces and immediate riparian zone covered by native vegetation, including trees, understory shrubs, or nonwoody macrophytes; vegetative disruption through grazing or mowing minimal or not evident; almost all plants allowed to grow naturally.	70-90% of the streambank surfaces covered by native vegetation, but one class of plants is not well-represented; disruption evident but not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	50-70% of the streambank surfaces covered by vegetation; disruption obvious; patches of bare soil or closely cropped vegetation common; less than one-half of the potential plant stubble height remaining.	Less than 50% of the streambank surfaces covered by vegetation; disruption of streambank vegetation is very high; vegetation has been removed to 5 centimeters or less in average stubble height.
SCORE (LB)	Left Bank 10 9	8 7 6	5 4 3	2 1 0
SCORE (RB)	Right Bank 10 9	8 7 6	5 4 3	2 1 0
10. Riparian Vegetative Zone Width (score each bank riparian zone)	Width of riparian zone >18 meters; human activities (i.e., parking lots, roadbeds, clear-cuts, lawns, or crops) have not impacted zone.	Width of riparian zone 12-18 meters; human activities have impacted zone only minimally.	Width of riparian zone 6-12 meters; human activities have impacted zone a great deal.	Width of riparian zone <6 meters; little or no riparian vegetation due to human activities.
SCORE (LB)	Left Bank 10 9	8 7 6	5 4 3	2 1 0
SCORE (RB)	Right Bank 10 9	8 7 6	5 4 3	2 1 0

Total Score _____

6.3 Pfankuch Channel Stability Evaluation

A comprehensive evaluation of the stability and erosion occurring within stream reaches is conducted using the method developed for the U.S. Department of Agriculture by Pfankuch (1975) to answer three basic questions: (1) what are the magnitudes of the hydraulic forces at work, (2) how resistant are the various organic and inorganic bank and channel components to the recent stream flow exerted on them, and (3) what is the capacity to adjust and recover from potential changes in flow volume and increases in sediment production.

Although the Pfankuch channel stability evaluation is considered non-scoring, supplemental information for the EII, it does provide useful information about the capacity of streams to recover from change by systematic measurements of the resistive capacity of stream channels to the detachment of bed and bank materials.

The parameters evaluated are:

Landform Slope: the lateral extent and ease to which banks can be eroded and the potential slough which can enter the water. Bank slopes may be measured with a clinometer.

Mass Wasting: mass movement of banks by slumping or sliding introducing large volumes of soil and debris into the channel causing constriction and increasing sedimentation rates.

Debris Jam Potential: the potential for increasing woody impediments to the natural direction and force of flow of the channel.

Bank Protection from Vegetation: density, coverage and root mat continuity as a measure of protection of the bank from overland flows.

Degree of Entrenchment: ratio of bankfull width to valley width as a relative index of channel incision.

Bank Rock Content: the relative resistance of bank rocks to detachment by flow forces.

Obstructions, Deflectors and Sediment Traps: objects within the stream that change the direction and velocity of flow as indicators of increasing channel damage.

Cutting: the loss of aquatic vegetation by scouring or uprooting, sometimes characterized by vertical channel walls.

Deposition: a measure of the amount and size of material that has recently been moved in or around a channel.

Rock Angularity: resistance of rock movement is evaluated by visually estimation of the sharpness of rock angles and shapes.

Brightness: the brightness of rock surfaces is an indicator of rock movement.

Consolidation: a measure of the unstable substrates that form in loose attachments, as opposed to more stable materials that pack together with voids filled over time.

Bottom Size Distribution and Percent Stable Materials: the change from the natural variation of component size classes and the percentage of all components judged to be stable.

Scouring and Deposition: a rating of the percentage of channel bottom affected by scouring and deposition.

Clinging Aquatic Vegetation: an evaluation of the community and coverage of macro-aquatic biomass as an indicator of stream stability.

Figure 6.3 Pfankuch Channel Stability Evaluation field sheet.

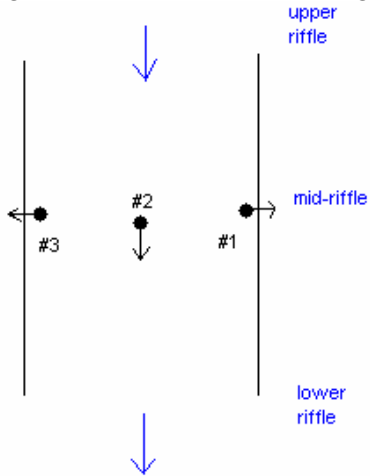
EII Reach Inventory and Channel Stability Evaluation					
EII Site Number:	Stream:	Start:	Date:	Time:	
Days since last rain (<+0.25"):	Avg. stream depth:	Avg. stream width:	Est. stream discharge (cfs):	Pool 1 =	Pool 2 =
Note flow condition (Flooding, high flow, moderate flow, low flow or dry):	Bankfull width: Riffle 1 =		Riffle 2 =		
Parameter	Excellent	Good	Poor	Score	
Upper Banks (From normal high water mark to the next bank in general slope; Relatively perennial with terracing and armoring)					
Landform Slope	Bank slope gradient < 30%	Bank slope gradient 30-40%	Bank slope gradient 40-60%	(6)	Bank slope gradient > 60%
Mass Wasting (existing or potential)	No evidence of past or potential for future mass wasting into channel.	Infreq. and/or very small, mostly healed over, low future potential.	Moderate freq. & size, with some raw spots eroded by high flows	(9)	Freq. or large, causing sediment loss nearly yearlong.
Debris Jam Potential (floatable objects)	Essentially absent from immed. channel	Present, but mostly small twigs and limbs	Present, volume and size are both increasing	(6)	Moderate to heavy amounts, larger sizes (8)
Bank Protection From Vegetation	90% + plant density, vigor and variety suggest deep, dense root mass	70-90% density, fewer species or lower vigor suggest less dense or deep root mass	50-70% density, fewer species, less vigor indicate poor, discont. or shallow root mass	(9)	>50% density, fewer species, less vigor indicate poor, discont. or shallow root mass
Lower Banks (Immediately submerged degree of entrenchment (entrenchment ratio))					
Bank Rock Content	Little or no entrenchment, ratio > 2.5	Minimal entrenchment, ratio of 2.0-2.5	Moderate entrenchment, ratio of 1.2-2.0	(3)	Highly entrenched, ratio < 1.2
Obstructions, Deflectors, Sediment Traps	+65% w/ large, angular boulders (12" dia.) abund.	40-65% small boulders to cobble (6-12" dia.)	20-40% with most in the 3-6" dia. class.	(6)	<20% rock fragments of gravel size (2-4" dia.)
Cutting	Rocks, old logs firmly embedd., flow pattern or pool/riffle stable w/out cutting or deposition	Some present, causing erosive cross currents and minor pool filling, obstructions and deflectors newer and less firm	Moderately frequent, mod. unstable obstruct. & deficit; move with high water causing bank cuttin and filling of pools	(6)	Freq. obstruct. and deficit, cause bank erosion yearlong, sed. traps full, channel migration occurring
Deposition	Little or none evident, infrequent raw banks less than 6" high	Some, intermittent at outcaves & constrictions, raw banks be up to 12" high	Significant, cuts 12-24" high, root mat overhangs and sloughing evident	(12)	Almost continuous cuts some over 24" high, failure of overhangs frequent
Bottom (Generally submerged portion of channel, totally aquatic)					
Rock Angularity	Little or no enlargement of channel or point bars	Some new increase in bar formation, most from coarse gravels	Moderate deposition of new gravel & coarse sand on old/new bars	(12)	Extensive dep. of predom. fine particles, accelerated bar development
Brightness (Clean rocks)	Sharp edges and corners, plane surfaces roughened	Rounded corners & edges, surfaces smooth & flat	Comers & edges well rounded in two dimensions	(3)	Well rounded in all dimensions, surf. smooth
Consolidation or Particle Packing	Surfaces dull, darkened or stained, gen. not bright	Mostly dull but may have up to 35% bright surfaces	Mixture, 50-50% dull and bright, (+, - 15%) mostly a loose assortment with no apparent overlap	(3)	Predom. bright, 65% exposed or scored surf.
Bottom Size Dist. & % Stable Materials	Assorted sizes tightly packed and/or overlapping	Moderately packed with some overlapping	Moderate change in sizes, stable material 20-50%	(6)	No packing evident, loose assortment, easily moved.
Scouring and Deposition	No change in sizes evident, stable materials 80-100%	Distribution shift slight, stable materials 50-80%	Moderate change in sizes, stable material 20-50%	(12)	Marked dist. change, stable materials 0-20%
Clinging Aquatic Vegetation (moss, algae)	Less than 5% of bottom affected by scouring and deposition	5-30% affected, scour at constrict. and where grades steepen, some dep. in pools	30-50% affected, dep. & scour at obstruct. constrict. and bends, pools filling	(18)	More than 50% of bottom in a stat of flux or change nearly yearlong
Attachment ratio = Width of the flood-prone area (width at 2x bankfull height) divided by width at bankfull	Abundant, growth largely moss-like, dark green, perennial, in swift flow also.	Common, algal forms in low velocity & pools, moss in pools and riffles	Present but spotty, mostly in backwater areas, seasonal blooms make rocks slick	(3)	Perennial veg. scarce or absent, yell-gr., short term blooms may be present.
Notes: (Large erosion sites, niche-points, mass ve deposition, excessive debris, exceptional integrity, etc...)					Total:

Figure 3.2. EII Reach Inventory and Channel Stability Evaluation Form

6.4 Densiometer

Densiometer readings will be made to estimate percent canopy cover at each riffle where diatom samples are collected. Three readings will be collected at each site, holding the densiometer out from the hip and level (Figure 6.4). The three readings will be averaged to yield average percent canopy cover.

Figure 6.4. Densiometer reading locations.



7.0 In-House Analytical Techniques

Some quantitative environmental analyses may be performed in the WRE water quality laboratory (Table 7.1). However, data analyzed at the WRE laboratory at the present time may not be submitted to TCEQ for inclusion in the 303(d)/305(b) assessment process under the Clean Rivers Program.

Table 7.1 In-house analytical methods by parameter.

Parameter	Method	Method	Equivalencies	Preservative	Holding Time	Detection Limits
Orthophosphate	Hach 8048		USEPA 365.1 SM 4500-P-E	ice and/or refrigeration to 4°C.	24 hrs. with refrigeration	0.01 ppm
Ammonia	Hach 8155 (Salicylate) OR Orion NH ₃ Electrode		USEPA 350.3 SM 4500-NH3-F-G	ice and/or refrigeration to 4°C.	run analysis as soon as possible or w/n 28 days if preserved w/ H ₂ SO ₄ to pH < 2	0.01 ppm
Nitrate	Hach 8192 (LR)/ 8171(MR) Cadmium reduction method	InQuest nitrate screen zinc reduction method		ice and/or refrigeration to 4°C.	Store at 4degrees c. or lower, run sample within 48 hrs	0.06 ppm
Turbidity	Hach 8237 (FTU)	Hach 2100P		ice and/or refrigeration to 4°C.	Store at 4°C or lower, run sample within 48 hrs	0.01 NTU
	Hach 16800 (NTU)	SM 2130 B		ice and/or refrigeration to 4°C.	Store at 4degrees c. or lower, run sample within 48 hrs	0.01 NTU
Fecal coliform	SM 9222D	n/a		ice and/or refrigeration to 4°C.	6 hours	1 CFU/100ml
Total Suspended Solids	SM 2540D	n/a		ice and/or refrigeration to 4°C.	24 hrs.- 7 days	0.5 mg/L

7.1 Ohmicron Nitrate Analysis

The Ohmicron spectrophotometer may be used to measure nitrate-nitrogen (NO₃ as N) and polycyclic aromatic hydrocarbons (PAH) in soils.

When analyzing nitrate on the Ohmicron spectrophotometer, always wear gloves and goggles. Check for the 550 filter block in spectrophotometer prior to analysis.

1. Set out and label Ohmicron tubes with the name of each sample and each QA/QC check (blank, low standard, high standard and the designated split). Place the tubes in the Ohmicron rack. Note that although the meter will print out the results, the printout will not contain site names. Thus, it is CRITICAL to keep the tubes in order throughout the testing process.
2. Use the micropipettor to measure 1000 µl (1 mL) of DIUF water into the “blank” tube. Stopper and shake the tube to prevent clumping of reagent powder.
3. Use the micropipettor to measure 900 µl (0.9 mL) of DIUF water into the low standard tube, then stopper and shake.

4. Use the micropipettor to measure 500 µl (0.5 mL) of DIUF into the high standard tube, then stopper and shake.
5. Use the micropipettor to measure 1000 µl (1 mL) of each sample into its respective tube. Stopper and shake each tube after the sample is added to prevent reagent clumping.
MAKE SURE TO GET A FRESH TIP ON THE PIPETTOR BETWEEN EACH SAMPLE TO PREVENT CROSS CONTAMINATION.
6. Pipette 100 µl (0.1 mL) of standard into the low standard tube, 500 µl (0.5 mL) of standard into the high standard tube, then stopper and shake both.
7. Remove all stoppers, and place each stopper in the row directly behind each corresponding tube (again, to prevent cross contamination).
8. Add 100 µl of developing solution to each tube, stopper and shake again.
9. Press START on the timer (a two-minute reaction countdown begins).

NOTE: The readings must all be taken within 2 minutes after the timer beeps, otherwise the color will overdevelop and generate false high readings!!!

10. When the timer beeps, take each tube in turn, wipe it with a chemwipe, place it in the spectrophotometer, and remove it when the spectrophotometer readout directs you to do so.
11. After the meter prints out its results, write on the printout the names of the standards and sites that correspond to the printed results.
12. Records the readings on the appropriate lab bench sheet.
13. Dispose of samples in sink.

7.2 Ohmicron PAH analysis

PAH may be analyzed in soil and sediment samples, although an extraction must be performed on the samples prior to analysis with the Ohmicron spectrophotometer.

Rapid Prep PAH Soil Extraction

Always wear gloves and goggles when performing soil extraction protocol.

1. Collect soil by weight (10grams)
2. Weigh 10-25 grams separately on a large filter paper and put in oven to dry.
3. Add 20 ml of PAH Extraction solution to collection device containing 10 grams of sample soil.
4. Replace cap and shake for one minute. Let stand for at least five minutes, until liquid appears clear and soil has settled.
5. Use filter apparatus to remove small amount of liquid:
 - a. Disassemble filtration plunger from filtration barrel.
 - b. Insert bulb pipet into top (liquid) layer in extraction jar and draw up sample. Transfer at least ½ bulb capacity into the filtration barrel. *Do NOT use more than one full bulb.*
 - c. Press plunger firmly into barrel until adequate filtered sample is available or unit snaps together.
 - d. Repeat for each sample to be tested.

Dilute the filtered extract into the appropriate extract diluent as described by analyte directions. For PAH analyses, use 250 µL filtered extracted to 12.24 mL diluent volume.

PAH Analysis

Always wear gloves and goggles when using the Ohmicron spectrophotometer for PAH analyses. Be sure that the 450 nm filter block is in the spectrophotometer when analyzing PAH.

1. Turn On Rapid Assay Spectrophotometer to allow for warm up.
2. Prepare PAH Enzyme conjugate for use- reconstitute as directed.
3. Remove upper rack from magnetic base. Label test tubes for use (Table 7.2).

Table 7.2. Test tube lab instructions for PAH analysis.

Tube #	Content/Label
0	Diluent/Zero
1,2	Standard 0, 0 ppb
3,4	Standard 1, 2.0 ppb
5,6	Standard 2, 10.0 ppb
7,8	Standard 3, 50.0 ppb
9	Control
10 thru # of samples	Sample 10 ... # of samples

4. Add 250 μ L of either standard, control or diluted soil extract to the bottom of each test tube by inserting the pipet tip all the way into the tube without touching the sides or the bottom of the tube.
5. Add 250 μ L of enzyme conjugate down the inside wall of each tube by aiming the pipet tip $\frac{1}{4}$ " to $\frac{1}{2}$ " below the tube rim without touching the rim or tube wall with the pipet tip; deliver liquid gently.
6. Add 500 μ L of thoroughly mixed PAH Antibody Coupled Magnetic Particles down the inside wall of each tube by using the technique described in #4. Vortex for 1 to 2 seconds at low speed to minimize foaming.
7. Incubate for 30 minutes at room temperature.
8. Combine the upper rack with the magnetic base; press all tubes into base; allow 2 minutes for the particles to separate.
9. Do not separate upper rack from lower base; using a smooth motion, invert the combined rack assembly over a sink and pour out the tube contents; keep inverted and gently blot the test tube rims on several layers of paper towels.
10. Add 1 mL of PAH washing solution down the inside wall of each tube and vortex each tube. Wait 2 minutes. Repeat step #8.
11. Lift the upper rack (with the tubes) off the magnetic base; add 500 μ L of color reagent down the inside wall of each tube by using the technique previously described. Vortex for 1 to 2 seconds at low speed to minimize foaming.
12. Incubate for 20 minutes at room temperature (15-30°C.) During this period add 1 mL washing solution to a clean test tube for use as an instrument blank in Step 12.
13. Add 500 μ L of stopping solution down the inside wall of each tube using the technique previously described. Read results at 450 nm within 15 minutes after adding the stopping solution. Multiply test results by appropriate dilution factor then divide by dry weight calculation for final concentrations.

7.3 NH₃ by Hach Spectrophotometer

This method for ammonia as nitrogen analysis by Hach spectrophotometer is adapted from FWPCA (1969) Methods for Chemical Analysis of Water and Wastes. Always wear gloves, goggles and dust mask when performing this analysis.

1. Set out and label small post-its with the name of each QA/QC check (blank and standards), each site and the designated split sample(s).
2. Rinse each cuvet 3 times with the sample or standard solution it will contain during testing.
3. Select HACH method 385, then press enter.
4. Rotate wavelength dial to 655 nm.
5. Press read/enter (display will show *mg/L N NH₃ Salic*).
6. Add one *Ammonia Salicylate* powder pillow to each bottle, INCLUDING the blank.
7. Stopper and shake each bottle until the reagent has dissolved.
8. Press Shift/Timer (a 3-minute countdown will begin).
9. When the timer beeps, remove the stoppers from each bottle, setting them behind the bottle they were removed from with the wide end down. This prevents cross contamination between samples.
10. Add one *Ammonia Cyanurate* powder pillow to each bottle.
11. Stopper and shake until dissolved.
12. Press Shift/Timer (a 15-minute countdown will begin).
13. When timer beeps, clean the outside of the blank with a chemwipe or lint-free cloth and place into the meter. Close the cover.
14. Press ZERO to zero the machine.
15. Clean the outside of all sample and standard cuvettes with a chemwipe or lint-free cloth and place them into the meter one a time. Press READ/ENTER.
16. Record the results on the lab bench sheet.
17. Dispose of bottle contents down the sink with plenty off tap water.

7.4 Orthophosphorus by Hach Spectrophotometer.

This method for orthophosphorus (reactive) as phosphorus analysis by Hach spectrophotometer is adapted from FWPCA (1969) Methods for Chemical Analysis of Water and Wastes. Always wear gloves and goggles when performing this analysis.

1. Set out and label small post-its with the name of each QA/AC check (blank, high standard and low standard), each site and the designated split sample(s).
2. Rinse each bottle 3 times with the sample or standard it will hold during testing. Keep the fourth pouring of each liquid in its bottle.
3. Press 491 READ/ENTER (for spec #1) or 496 READ/ENTER (for spec # 2 and #3).
4. Turn the dial of the meter to 890 nm. Press READ/ENTER.
5. Add one reagent pillow to each bottle, EXCEPT the blank.
6. Stopper bottles and shake each to mix reagent with sample.
7. Press SHIFT TIMER (a 2-minute reaction countdown will begin).
8. Clean the glass of the blank with a lint-free towel, place it in the meter, and close the cover.
9. Press ZERO (the display will show WAIT, then 0.00 mg/mL P).
10. Clean the glass of each bottle with a lint-free towel.
11. Place each into the meter, close the cover, and press READ/ENTER.
12. Record the readings for each bottle on the bench sheet.

13. Dispose of bottle contents down the sink with plenty of water, and clean up the table for the next crew.

7.5 Total Suspended Solids

Total suspended solids (TSS), also known as total non-filterable residue, is analyzed by Standard Methods 2540-D. Always wear latex gloves to ensure that human oil and dirt from skin does not impact this test.

1. With DI water, rinse the filtering apparatus three times.
2. Remove a dried, pre-weighed metal pan and filter from the dessicator, noting the number written on the bottom of the pan.
3. On the TSS bench sheet, write in the name of your sample in the space that corresponds with the number of the pan you just chose.
4. With forceps, place the pre-weighed glass-fiber filter on the filtering apparatus.
5. Attach the funnel portion of the apparatus carefully, so the portion that tightens down is threaded properly. This ensures a good seal for the vacuum.
6. Swirl or otherwise agitate the sample container to re-suspend the solids in the water column.
7. Measure out 500 mL of the water sample in a graduated cylinder.
8. Slowly pour the measured sample into the “funnel” at the top of the filtering apparatus; avoid having a great amount of water sitting in the funnel, or the suspended solids may be lost on the sides of the apparatus.
9. Create a vacuum to pull the water through the filter either by a hand pump or by water bypass.
10. When the water has dissipated from the “funnel” portion of the apparatus, fill the graduated cylinder you just used with the rest of your sample.
11. Record the total amount of sample used (in liters) on the TSS bench sheet, next to the sample name (e.g., “.986 L” [500 mL + 486 mL]).
12. Activate the vacuum again.
13. When the sample has dissipated from the funnel, pour about 100 mL of DI water into the cylinder and rinse both the cylinder and the sides of the apparatus funnel, allowing this rinse water to be vacuum filtered as well.
14. Disassemble the apparatus and use forceps to remove the filter.
15. Place the filter on its metal pan, and place both in the oven to dry for 24 hours.
16. Rinse the filter and funnel of the apparatus 3 times, disposing of the sample in the sink with plenty of tap water then clean up the table for the next crew.

7.6 Fecal Coliform

Fecal coliform are part of the total coliform group that ferment in lactose in 24 hours (⁺/ 2 hours) at 44.5°C. These gram-negative non-spore forming rod bacteria will produce acidity appearing as a blue colony in the membrane filter procedure. The major species is *Escheria coli* and is indicative of fecal pollution and possible presence of enteric pathogens.

Samples must be delivered to the laboratory within six hours of sample collection. An additional 2 hours of refrigerated processing/testing time is allowed at the laboratory, yielding a total holding time of 8 hours from the time of sample collection to the time of filter insertion in the incubation over.

Analysis of fecal coliform is adapted from FWPCA (1969) Methods for Chemical Analysis of Water and Wastes, and is equivalent to Standard Methods 9221-E. Always wear latex gloves to discourage contamination of the test.

1. Put on latex gloves to discourage contamination of the test field
2. Label the disposable petri dishes with the appropriate date and site name on the side of the dish that has raised writing on it ; set the labeled dishes aside
3. Use DI water to cleanse the suction apparatus three times, then assemble
4. Measure 90 mL of DI water into a graduated cylinder
5. Sterilize a pair of forceps by dipping the tips in alcohol, then holding their tips briefly in the blue cone of a flame
6. Use the sterilized forceps to remove the filter's protective plastic coating and place it on the apparatus with the grid side visible to the technician
7. Pour approximately 45 mL of the DI water from the graduated cylinder into the assembled apparatus
8. Pull a 10 mL amount of the water sample from the whirlpak with a pipette
9. Release the 10 mL of the water sample from the pipette into the assembled apparatus
10. Pour the remaining DI water from the graduated cylinder into the apparatus
11. Attach the suction gun and pump it to evacuate the rest of the water from the top of the apparatus to the collection cup below
12. Add one tube of blue nutrient broth to the dish
13. Disassemble the apparatus
14. Resterilize the forceps, using the method outlined above
15. Gently remove the filter from the apparatus with the forceps and, using the "roll" method, place the filter, grid side up, in the bottom of the petri dish
16. Place the cap on the petri dish and twist to close it tightly
17. Cut a 1"- wide strip of Parafilm and wrap it tightly around the circumference of the dish to seal the contents inside.
18. DI all parts of the apparatus three times to cleanse

7.7 Scale Operation

The scale is frequently used to weigh filters for use in TSS analysis. Always wear latex gloves when calibrating or using the scale to prevent oil and dirt from human skin from affecting measurements.

Calibration

Calibration should only be performed by trained personnel.

1. Place the 30 g weight on the scale, close the door and record the reading once the "U" has disappeared.
2. Replace the 30 g weight with the 50 g weight and repeat the procedure.
3. Remove the weight from the scale and replace it in the appropriate container.
4. Press "TARE" and hold it down until "CAL" appears in the readout.
5. Place the 30 g weight on the scale, close the door and wait until the readout settles to "30.000"
6. Remove the weight and replace it in the appropriate container. press "TARE" and hold it down until "CAL" appears in the readout.

7. Place the 50 g weight on the scale, close the door and wait until the readout settles to "50.000".
8. Replace the weight in the appropriate container.

Measurement

1. Put on gloves to prevent oils, etc from affecting measurements
2. Place container (vial, paper, etc) on the scale and close the door.
3. Wait until the "U" on the left hand side of the readout disappears.
4. Press "TARE" to zero the scale.
5. Place the material to be weighed on the scale, and record the readout once the "U" disappears.

7.8 Glassware washing procedure

Wash all glassware in soapy water, using the (brown) liquinox low-phosphorus soap. After scrubbing all debris from the glassware, rinse thoroughly with tap water at least six times until all evidence of soap is gone.

Rinse three times with DI water, then hang glassware on rack or place in dish to air dry.

7.9 Shelf Life Tracking of Laboratory Supplies and Standards

1. When unpacking shipment, inspect the shipping invoice, compare to the PRF to ensure all supplies ordered have been received. Give the invoice to appropriate Admin staff after verification.
2. Apply HazMat label , with Health, Fire, Reactivity and Hazard numbers obtained from the MSDS book. Include the date received on the edge of the Label.
3. When ordering a new chemical or standard, request a MSDS sheet, insert in the MSDS book and mail or fax a copy to the safety office.

8.0 References

- American Public Health Association. 1995. Standard methods for the examination of water and wastewater. 19th Edition.
- Barbour, M.G., J.H. Burk, and W.D. Pitts. 1980. Terrestrial Plant Ecology. Benjamin/Cummings, California.
- Barbour, M.T. and J.B. Striblings. 1991. Use of habitat assessment in evaluating the biological integrity of stream communities. In Biological Criteria: Research and Regulation, 1991, pp 25-38. EPA-440/5-91-005. U.S. EPA, Office of Water, Washington, DC.
- Blum, J.L. 1957. An Ecological Study of the Algae of the Saline River, Michigan. *Hydrobiologia* 9:361-408.
- Hynes, H.B.N. 1972. The Ecology of Running Waters. University of Toronto Press, Canada.
- Correll, D.S. and H.B. Correll. 1972. Aquatic and Wetland Plants of Southwestern United States. U.S. Government Printing Office, Washington, DC.
- Compton, R.R. 1985. Geology in the Field. John Wiley and Son, Toronto.
- Pfankuch, D.J. 1975. Stream reach inventory and channel stability evaluation. USDA Forest Service, R1-75-002. Government Printing Office #696-260/200.
- Texas Commission on Environmental Quality (TCEQ). 2003. Surface Water Quality Monitoring Procedures Manual, Volume 1: Physical and Chemical Monitoring Methods for Water, Sediment, and Tissue. RG-415 Texas Commission on Environmental Quality, Monitoring Operations Division.
- Texas Surface Water Quality Standards (TSWQS). 1997. Texas Administrative Code Title 30, Chapter 307.
- Water Resource Evaluation Section (WRE). 2003. WRE Section Core Routine Monitoring Parameter List. SR-03-09. City of Austin Water Resource Evaluation Section.
- City of Austin DUD ERM 1997. Environmental Integrity Index Water Quality Technical Assessment Methodology. COA-ERM/WRE 1997-03.
- USGS (M.R. Meador et al). 1993. Methods for characterizing stream habitat as part of the National Water Quality Assessment Program. Open File Report 93-408.