STILLWATER CREEK WATERSHED

WATER QUALITY MONITORING PROGRAM

By

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WATER QUALITY MONITORING PROGRAM

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Abstract: The city of Stillwater contracted the Environmental Science Graduate Program (ESGP) to create an ambient water quality monitoring program for streams that are within city limits. The ESGP was also contracted to collect sample data following the designed water quality monitoring program (WQMP) for the duration of this project. This WQMP is part of a larger stormwater project that the city contracted with Meshek Engineering and Jacobs Engineering. The WQMP is designed following the guidelines set forth by the Clean Water Act (CWA). The project area is within the Stillwater Creek Watershed and includes streams that flow through the Stillwater city limits. This project includes a selection of sample site locations, parameter sampling based on designated beneficial uses for each waterbody, a cost analysis of the WQMP, a stand-alone sampling manual document, data analysis, future considerations for the city, and a conclusion. This WQMP is for the city of Stillwater's use to collect water quality data for submission to the Oklahoma Integrated Report.

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CHAPTER I

DOCUMENT OVERVIEW

This document is a report created for the city of Stillwater for ambient water quality monitoring of surface waters in the Stillwater area. The "Introduction" section covers the importance of surface water quality, the city contract with the Environmental Science Graduate Program (ESGP), the Clean Water Act (CWA) and sections within, the overview of the Stillwater Creek Watershed, and an explanation of the organizations in Oklahoma dealing with water quality. The "Water Quality Monitoring Program" section is an overview of the program that has been designed for the city to monitor surface waters. This section covers the design of the program, the cost analysis, and a sample manual for application in the field. The "Data Summary" section includes water quality data from 2020-2021 sampling. This data is analyzed for attainment of water quality standards for the assigned beneficial uses for that waterbody. The "Future Considerations" section addresses suggestions for the city in moving forward with the water quality monitoring. The "Conclusion" section summarizes the findings from this project and the importance of water quality monitoring.

CHAPTER II

INTRODUCTION

Surface water is a valuable resource that is increasingly more important as water scarcity becomes a global issue (UN, 2021). The quality of that surface water is also valuable for the aquatic, riparian, and terrestrial ecosystems they serve. Surface water is often used for municipal drinking supply and the quality of that water can directly impact human health (USGS, 2021).

Freshwater only accounts for 2.8% of the world's total water and only 0.001% of that percentage is stored in rivers (Allan, 1995). Rivers play a vital role in terrestrial ecosystems and in anthropogenic uses even though they account for such a small portion of the world's water. Streams also play a major role in nutrient distribution throughout watersheds big and small. The global importance of rivers for the earth's survival gives reason for strategic stream management.

The city of Stillwater contracted with Oklahoma State University's ESGP to create and implement an ambient water quality monitoring program. The purpose will be to regularly monitor the water quality (WQ) of the streams in the city of Stillwater. The water quality monitoring program (WQMP) includes a sampling manual and a cost analysis for a comprehensive program and a minimally effective program. The ESGP was also tasked with taking WQ samples during the duration of this project from 2020-2021. Meshek Engineering and Jacobs Engineering are the lead contractors for the stormwater project. They have worked

closely with the ESGP department on this water quality monitoring program design and implementation.

Clean Water Act

The first Clean Water Act (CWA) was implemented in 1948 and was initially titled the Federal Water Pollution Control Act (FWPCA). The goal of this Act was to establish a system for regulating pollutants that are discharged into United States waters. The Act also addresses the regulation of surface water quality standards. The FWPCA was significantly revised and renamed the CWA in 1972. The CWA has given the Environmental Protection Agency (EPA) the authority to create and establish water quality monitoring programs and regulations that each state is required to uphold for their corresponding surface waters (EPA, 2021). The scope of this project and water quality monitoring program is defined by the CWA and the regulations that the state of Oklahoma must maintain for the state's surface water quality.

Integrated Report

The CWA requires that states create and submit a report every two years that summarizes the current conditions of their surface waters. Section 305(b) of the CWA requires that states report a full inventory of the waterbodies in the state. This allows the EPA to assess the overall quality of waterbodies in the country. This portion of the Integrated Report generated by each state also serves as a progress report for improving impaired waterbodies and assessing their "fishable/swimmable" attainment (ODEQ, 2021).

Per section 303(d) of the CWA, states must also maintain a list of waterbodies that do not meet water quality standards. This list is measured against the beneficial use parameters

assigned to each waterbody as defined by the Code of Federal Regulations (CFR) and the Oklahoma Water Resources Board (OWRB). The CWA requires that a Total Maximum Daily Load (TMDL) be created for waterbodies on the 303(d) list. The purpose is to improve the quality of that waterbody, so it is no longer impaired. A TMDL measures the maximum amount of pollutant that can be discharged in a particular waterbody while still meeting water quality standards (ODEQ, 2021).

The Integrated Report is a combined report of both the 305(b) and 303(d) report requirements of the CWA (ODEQ, 2020). The Integrated Report lists all the water quality standards required to meet each beneficial use for waterbodies. These standards have been used to determine attainability of beneficial uses for each waterbody in this project based on the data that has been collected so far.

NPDES Permitting

The CWA established a National Pollutant Discharge Elimination System that serves to regulate point source pollutant discharges allowed in surface waters across the United States. The EPA has given 47 of the 50 states authorization to assign NPDES permits under the authority of the EPA (EPA, 2021). The city of Stillwater has an NDPES permit for stormwater and is therefore required to monitor the number of pollutants that enter the surface waters in the Stillwater city limits. The stormwater permit is called a Municipal Separate Storm Sewer System (MS4) permit. The MS4 permit requires that a Stormwater Management Plan be created by the permit holder. A part of the Stormwater Management Plan includes monitoring water quality of the surface waters where stormwater is discharged. This WQMP will meet the requirements for

water quality monitoring for the MS4 permit. Water quality monitoring has not been performed by the city up to this point.

Oklahoma Water Organizations

The Oklahoma Department of Environmental Quality (ODEQ), the Oklahoma Water Resources Board (OWRB), and the Oklahoma Conservation Commission (OCC) are the predominant governing bodies that are involved with surface water quality in Oklahoma. The ODEQ authors the Integrated Report and manages point source discharges. The EPA has given the ODEQ authority to issue NPDES permits to municipalities in the state. The OWRB has authority to set water quality standards for surface waters as well as assigning their beneficial uses. The OWRB has an ambient water quality monitoring program for surface water and groundwater in the state. The OCC focuses on non-point source pollution and has an ambient water quality monitoring program for surface water in the state.

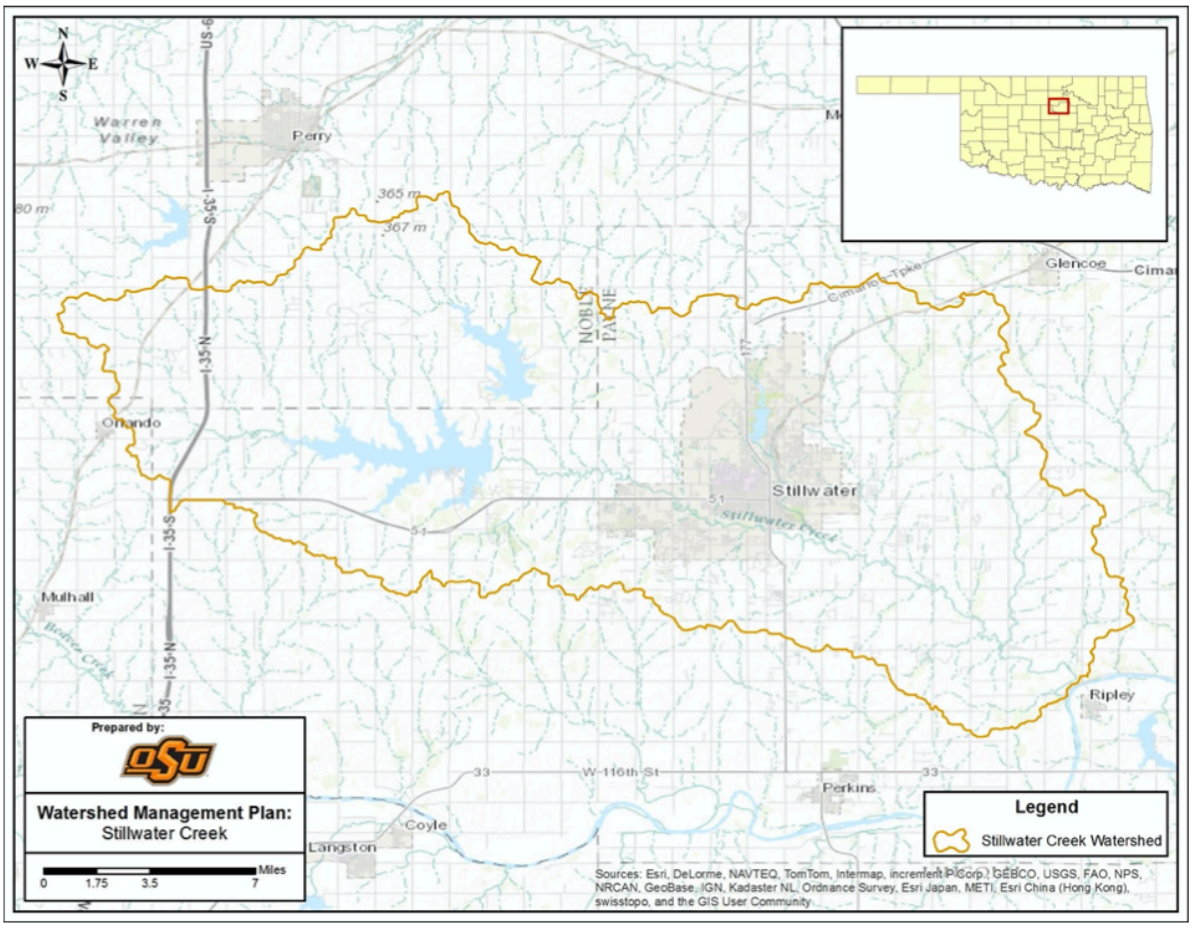

Figure 1. Map of the Stillwater Creek Watershed. This figure was obtained from the Watershed Based Plan for the Stillwater Creek Watershed (Barnes et al, 2015).

Stillwater Creek Watershed

The Stillwater Creek Watershed (SWC) falls within Noble, Logan, and Payne counties. The area of the SWC is 276 square miles and is within the Hydrologic Unit Codes (HUC) 11050003030 and 11050003040. It is in the Lower Cimarron sub-basin. Stillwater Creek is the primary stream that drains the watershed towards the southeast and eventually converges with the Cimarron River. Stillwater Creek enters the watershed from the west. It forms Lake Carl Blackwell and then flows through the city of Stillwater before reaching the Cimarron River confluence. Stillwater Creek is fed by Stillwater Creek North, Cow Creek, Duck Creek, Sanborn Hazen Lake Creek, Boomer Creek, Brush Creek, and East Brush Creek (Barnes et al, 2015).

For the scope of this water quality monitoring program the section of Stillwater Creek that flows through Stillwater has been chosen for sampling. Boomer Creek, Sanborn-Hazen Creek, Duck Creek, Cow Creek, and Stillwater Creek, North have also been chosen for sampling as they drain Stillwater and converge with Stillwater Creek.

Table 1 illustrates the current water quality status as reported in the 2020 Integrated Report for each stream and the associated impairment reason for streams that did not meet water quality standards for their designated beneficial uses (ODEQ, 2020).

305(b) Assessment from 2020 Oklahoma Integrated Report

Waterbody ID	Waterbody Name	Size (miles)	Category	Impairment
OK620900040070_10	Stillwater Creek	16.43	5a	5c Benthic Macroinvertebrates, 5a Dissolved Oxygen, 5a Turbidity
OK620900040140_00	Boomer Creek	2.28	5c	Benthic Macroinvertebrates
OK620900040180_00	Boomer Creek	6.49	3	
OK620900040150_00	Sanborn-Hazen Lake Creek	3.59	5c	Benthic Macroinvertebrates
OK620900040195_00	Duck Creek	3.06	3	
OK620900040200_00	Cow Creek	8.26	5c	Benthic Macroinvertebrates
OK620900040230_00	Stillwater Creek, North	6.80	3	

Category	
1	Attaining the water quality standard and no use is threatened
2	Attaining some of the designated uses; no use is threatened; and insufficient or no data and information is available to determine if the remaining uses are attained or threatened
3	Insufficient or no data and information to determine if any designated use is attained
4	Impaired or threatened for one or more designated uses but does not require the development of a TMDL
4A	TMDL has been completed
4B	Other pollution control requirements are reasonably expected to result in the attainment of the water quality standard in the near future
4C	Impairment is not caused by a pollutant
5	The water quality standard is not attained. The waterbody is impaired or threatened for one or more designated uses by a pollutant(s), and requires a TMDL
5A	TMDL is underway or will be scheduled
5B	A review of the Water Quality Standards will be conducted before a TMDL is scheduled
5C	Additional data and information will be collected before a TMDL or review of the Water Quality Standards is scheduled

Table 1. Current Water Quality Status. Information in this table was obtained from the Oklahoma 2020 Integrated Report (ODEQ, 2020).

CHAPTER III

WATER QUALITY MONITORING PROGRAM

An effective water quality monitoring program (WQMP) requires carefully chosen sample sites that accurately represent the character of the sampled waterbody. The beneficial uses of the sampled waterbodies are necessary to know what parameters should be tested. The frequency of sampling and temporal range of sample dates is important so that the waterbody can be accurately represented over different seasons. Samples should be taken under antecedent base flow conditions to ensure that data is not skewed by high flow rainfall runoff events. The more sample data that can be collected will always result in the most accurate determination of water quality for any given waterbody (Dressing, 2005).

The design of this monitoring program was directly modeled after the Oklahoma Conservation Commission's (OCC) sample collection methodology and sample frequency. The OCC Standard Operating Procedures (SOP) document contains instructions for each sample parameter that the OCC collects samples for (OCC, 2014). The sample manual in this document follows the template of the OCC SOP document for sampling instructions. Sampling frequencies for this WQMP are taken directly from the OCC Small Watershed Rotating Basin Monitoring Program Quality Assurance Project Plan (QAPP), (OCC, 2017) . This is the monitoring program that the OCC follows for their ambient water quality monitoring in the state. The EPA requires that stringent data collection methodology be followed for the submission of data for the Integrated Report. The OCC is a leading contributor for data in the Oklahoma Integrated Report and thus meets the requirements for data collection. The OCC methodology is also appropriate

for the city's use because they monitor for non-point source pollution in the state of Oklahoma. Stormwater drainage into surface waters is largely affected by non-point source pollution. This means that the source for pollution from stormwater is often untraceable to a single point of discharge.

Design

The streams chosen for monitoring flow through the city of Stillwater and receive discharge from the city stormwater runoff as well as rural rainfall runoff. The monitoring program is intended to test the parameters of each stream according to the beneficial uses and water quality standards that have been assigned by the OWRB. Sample sites were chosen based on accessibility and distance from the nearest confluence of a converging stream. The sample location is far enough upstream from the confluence to determine the water quality of the sampled stream without influence from the converging stream backflow. Sample sites were first chosen by map observation and were then analyzed in the field by OSU and Jacobs Engineering personnel to evaluate the feasibility and quality of the site location.

Site Locations

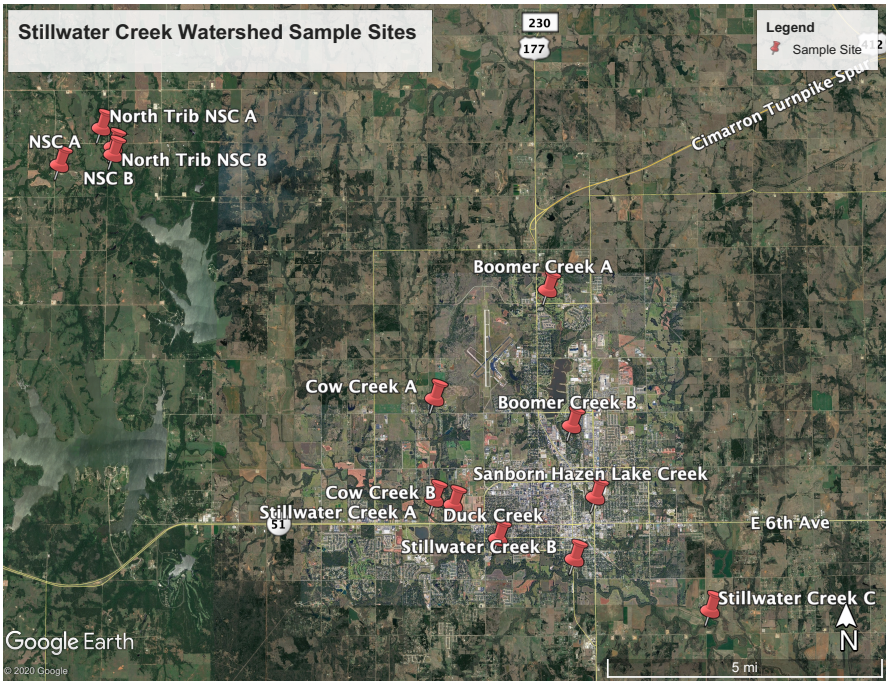

Figure 2. Stillwater Creek Watershed Sample Sites. This map was created using Google Earth.

Figure 2 shows the sample site locations chosen for this WQMP. The Cow Creek sample locations were chosen to capture the water quality north of the Oklahoma State University agricultural fields and facilities. The second location is located south of these facilities to determine their impact on water quality (Figure 2).

Stillwater Creek sample sites were chosen to obtain water quality before the confluence of contributing streams on the west side of Stillwater, toward the middle east-west of Stillwater, and the third site was chosen to capture the water quality of the creek as it flows from the city limits on the eastern most edge. It should be noted that for the samples taken in 2019-2021, the Stillwater Creek C sample site was not utilized due to lack of landowner cooperation. A new sample site with public access or with landowner permission should be chosen along this portion of Stillwater Creek for future sampling.

Boomer Creek sample sites were chosen north and south of Boomer Lake to evaluate the water quality of the creek at the inflow to the reservoir as well as the outflow. Single sample sites for both Duck Creek and Sanborn Hazen Lake Creek were chosen just north of their confluence with Stillwater Creek to evaluate the impact of the city stormwater on these two creeks.

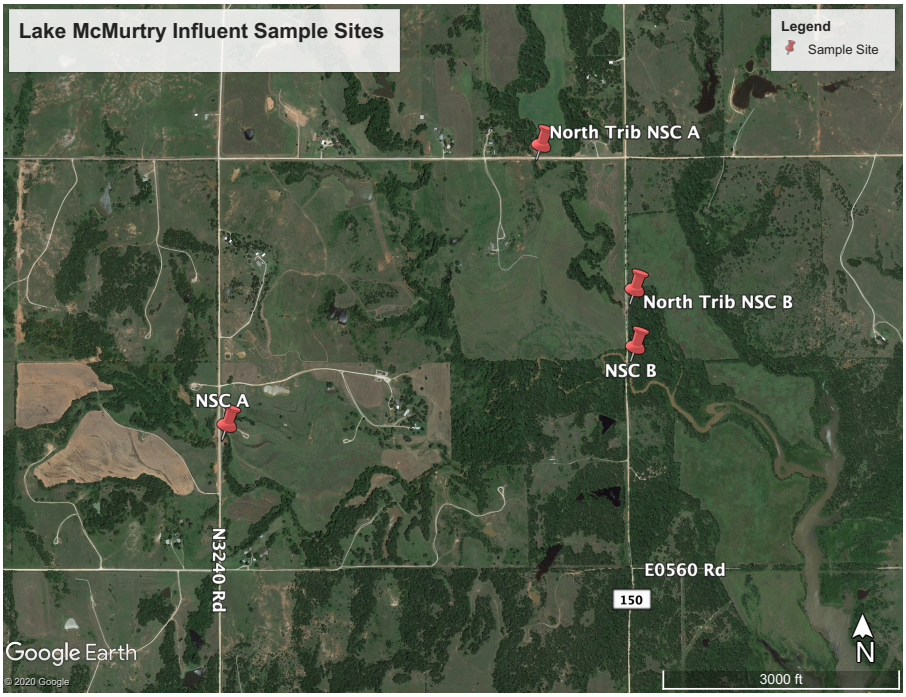

Figure 3. Northwest Stillwater Creek Watershed Sampling Sites. This map was created using Google Maps.

Four additional sampling sites were added to the project for evaluation northwest of Lake McMurtry Figure 3). The purpose of these sites is to examine the potential non-point source pollution (NPS) responsible for harmful algal blooms and increasing sediment concentrations that are occurring at Lake McMurtry. These locations were also chosen for accessibility and for their proximity to Lake McMurtry inflow. North Stillwater Creek (NSC) is the main water source for the Lake McMurtry reservoir and North Trib NSC is a tributary that feeds North Stillwater Creek.

Sample Parameters

Beneficial Uses per OAC 785:45-5 & 785: Appendix A

Waterbody ID	Waterbody Name	Size (miles)	Category	Aes	Ag	HLAC	WWAC	FC	PBCR	SBCR	PPWS	EWS	SWS
OK620900040070_10	Stillwater Creek	16.43	5a	•	•	•		•		•	•	•	
OK620900040140_00	Boomer Creek	2.28	5c	•	•		•	•	•		•		
OK620900040180_00	Boomer Creek	6.49	3	•	•		•	•	•		•		*
OK620900040150_00	Sanborn-Hazen Lake Creek	3.59	5c	•	•		•	•	•				
OK620900040195_00	Duck Creek	3.06	3	•	•		•	•	•				
OK620900040200_00	Cow Creek	8.26	5c	•	•		•	•	•				
OK620900040230_00	Stillwater Creek, North	6.8	3	•	•		•	•	•		•		*

Beneficial Use	Parameters Tested
Aesthetic	Nutrients, Oil & Grease
Agriculture	Total Dissolved Solids, Chlorides, Sulfates,
Habitat Limited Aquatic Community	Dissolved Oxygen, Toxicants, pH, Fish Collection, Macroinvertebrates, Turbidity, Oil & Grease, Sediment
Warm Water Aquatic Community	Dissolved Oxygen, Toxicants, pH, Fish Collection, Macroinvertebrates, Turbidity, Oil & Grease, Sediment
Fish Consumption	Using long-term average numerical parameters
Primary Body Contact Recreation	E. coli, Enterococci
Secondary Body Contact Recreation	E. coli, Enterococci
Public and Private Water Supply	Toxicants, Total Coliform, Oil & Grease, Chlorophyll-α/Total Phosphorus
Emergency Water Supply	**All waterbodies designated Emergency Water Supply beneficial use shall be deemed to be attaining the beneficial use for all water quality related issues.
Sensitive Water Supply	See *OAC 785:45-5-25-a-4*

Aes = Aesthetic
Ag = Agriculture
HLAC = Habitat Limited Aquatic Community
WWAC = Warm Water Aquatic Community
FC = Fish Consumption
PBCR = Primary Body Contact Recreation
SBCR = Secondary Body Contact Recreation
PPWS = Public and Private Water Supply
EWS = Emergency Water Supply
SWS = Sensitive Water Supply

Table 2. Beneficial Uses of Waterbodies. Information in this table was obtained from the Oklahoma 2020 Integrated Report (OCC, 2020).

Sample parameters for each sample site were chosen based on the beneficial uses that have been assigned to each waterbody by the OWRB. Beneficial uses are defined in the Oklahoma Administrative Code Chapter 45.

Sample Frequency

Table 1: Sampling Frequency

Parameter	Collection Frequency
Physical and chemical field parameters	With each collection (fish and water quality collections)
Chemical "lab samples"	10x / year fixed interval sampling, every 35 days
Benthic Macroinvertebrates	2 in summer / 2 in winter
Fish	Once (summer collections)
Flow	With each sample collection and habitat assessment.
Habitat	Once (with fish) / two years
Bacteria	Monthly (May - September)

Table 3. Sampling Frequency. This table was obtained from the Oklahoma Conservation Commission Small Watershed Rotating Basin Monitoring Program Quality Assurance Project Plan (OCC, 2017).

Table 3 shows the sample frequency used by the OCC for their rotating basin monitoring program (OCC, 2017). The rotating basin monitoring program is designed by the OCC to monitor non-point source pollution in small feeder streams. This program monitors a total of 250 streams spanning the five watershed basins in Oklahoma. Monitoring occurs every two years per basin (OCC, 2021). This sample frequency would be considered the most comprehensive approach to monitoring so that the water quality in each waterbody is analyzed with a large set of data over time. This sampling frequency is recommended for this WQMP. The most accurate determination of water quality can be made with the most collected data over every season in the year (Dressing, 2005).

YSI probe samples were taken every month for the sampling duration of this project. Parameters measured by the YSI probe would be considered chemical field/"lab samples" according to Table 3 and should be measured every 35 days according to OCC standards. This is the sample frequency that was followed for this project during the samples collected from February 2020 to May 2021. Some sampling events did not take place due to COVID-19 restrictions and low flow conditions at some sample sites.

The EPA has minimum sampling frequency requirements for data collected for the Integrated Report, which are less frequent than the OCC sampling frequency. The minimum required samples for determining beneficial use attainment in streams is a minimum of 10 samples every 5 years for data submission. This applies to the parameters: pH, temperature, Dissolved Oxygen (DO), coliform bacteria, salts, and dissolved solids. A minimum of 5 samples in 5 years is required for toxicants. A minimum of 4 macroinvertebrate samples collected over at least a two-year period is required for biological assessment. A WQMP following this sample frequency would be considered minimally effective. Taking the minimum required samples for submission to the Integrated Report does not result in the most accurate water quality determination for a given waterbody. The minimum required sampling frequency is not recommended for this WQMP.

Cost Analysis

This section includes both a "comprehensive" cost analysis and a "minimal" cost analysis and equipment costs. The comprehensive cost analysis is for the recommended sampling frequency for this WQMP based on OCC guidelines. The minimal cost analysis is calculated based on the minimal requirements for sampling frequencies required by the EPA.

The budget estimations are subject to change based on the sample processing costs of the third-party laboratory services and the current cost of equipment setup and maintenance. The sampling costs for laboratory services are estimates given by Accurate Labs in Stillwater. The samples estimated for Accurate Labs are phosphorus and nitrogen (PAN) nutrients, total phosphorus, *Escherichia Coli*, *Enterococcus*, and total coliform. Macroinvertebrate sorting and identification costs are estimated from laboratory services provided by Oklahoma State University in the Integrative Biology department. The cost of each test is multiplied by the

number of sample sites required for that parameter to be tested. This results in the total cost for each parameter per sampling event.

Comprehensive Cost Analysis

Water Quality Sampling Cost Analysis

	Test	Number of samples	Price per sample	Total Price
January				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
OSU	Macroinvertebrates	10	$200.00	$2,000.00
February				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
March				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
April				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
May				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
Accurate Labs	E Coli	10	$75.00	$750.00
Accurate Labs	Enterococcus	10	$75.00	$750.00
Accurate Labs	Total Coliform	6	$75.00	$450.00
June				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
Accurate Labs	E Coli	10	$75.00	$750.00
Accurate Labs	Enterococcus	10	$75.00	$750.00
Accurate Labs	Total Coliform	6	$75.00	$450.00
OSU	Macroinvertebrates	10	$200.00	$2000.00
July				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
Accurate Labs	E Coli	10	$75.00	$750.00
Accurate Labs	Enterococcus	10	$75.00	$750.00
Accurate Labs	Total Coliform	6	$75.00	$450.00
August				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
Accurate Labs	E Coli	10	$75.00	$750.00
Accurate Labs	Enterococcus	10	$75.00	$750.00
Accurate Labs	Total Coliform	6	$75.00	$450.00
September				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
Accurate Labs	E Coli	10	$75.00	$750.00
Accurate Labs	Enterococcus	10	$75.00	$750.00
Accurate Labs	Total Coliform	6	$75.00	$450.00
October				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
November				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
December				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
Annual Total				$40150.00

PAN Nutrients include: Ammonia, K, Nitrate, Nitrite, pH, Total Phosphorus, and TKN
Total Phosphorus includes: Total Phosphorus and Dissolved Phosphorus

Table 4. Comprehensive Water Quality Sampling Cost Analysis. These costs are estimated from Accurate Labs and the Oklahoma State University Integrative Biology Department. This cost analysis follows the sampling frequency of the OCC QAPPs (OCC, 2017).

A comprehensive cost analysis was performed to estimate the cost of the most ideal water quality monitoring program. The most ideal water quality monitoring program would exhibit sampling frequencies as demonstrated by the OCC QAPPs. This program would be the most comprehensive in assessing the surface water quality of the streams in the Stillwater Creek Watershed. The total sample processing cost of the recommended WQMP is $40,150 per year. This cost does not include sample collection.

Minimal Cost Analysis

Water Quality Sampling Cost Analysis

	Test	Number of samples	Price per sample	Total Price
May				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
Accurate Labs	E Coli	10	$75.00	$750.00
Accurate Labs	Enterococcus	10	$75.00	$750.00
Accurate Labs	Total Coliform	6	$75.00	$450.00
OSU	Macroinvertebrates	10	$200.00	$2000.00
September				
Accurate Labs	PAN Nutrients	10	$185.00	$1850.00
Accurate Labs	Total Phosphorus	10	$35.00	$350.00
Accurate Labs	E Coli	10	$75.00	$750.00
Accurate Labs	Enterococcus	10	$75.00	$750.00
Accurate Labs	Total Coliform	6	$75.00	$450.00
Annual Total				$10300.00

Table 5. Minimal Water Quality Sampling Cost Analysis. These costs are using estimates from Accurate Labs and the Oklahoma State University Department of Integrative Biology. This analysis follows the EPA's minimum sampling frequency requirements for the Integrated Report (ODEQ, 2020).

Table 4 shows the cost of a WQMP that follows the minimum requirements for data submission to the Integrated Report. This cost is for sample processing and does not include

sample collection. The minimum requirement for chemical samples is 10 samples every five years. This frequency would only require that two samples be taken per year for five-year intervals to be submitted to the Integrated Report. The minimal cost analysis is $10,300 per year.

Equipment Costs

Equipment Cost Analysis

Sampling Device	Parameters Included	Cost
YSI ProDSS Multiparameter Water Quality Meter w/ GPS	Conductivity, Specific Conductance, Salinity, Total Dissolved Solids (TDS), Resistivity, Seawater Density, Depth, GPS Coordinates, Temperature, Barometric Pressure	$2,160.00
ProDSS Turbidity Sensor	Turbidity	$1,100.00
ProDSS ODO Optical Dissolved Oxygen Sensor	Dissolved Oxygen	$1,000.00
ProDSS Total Algae Sensor, PC Freshwater	Total Algae	$3,200.00
Global Water Flow Probe FP311	Flow Rate	$1,090.00
Total Cost		$8,550.00

Table 6. Equipment Cost Analysis. This cost analysis includes prices of equipment needed for the WQMP.

The Equipment costs for the WQMP include the initial purchase of the YSI probe and sensors, as well as a flow meter. These costs were taken from the YSI website and reflect prices at the time of this project. The total cost of equipment needed for this WQMP is $8,550.

Sampling Manual

The draft sampling manual found in Appendix A, is a stand-alone document written specifically for the purposes of water quality monitoring by the city of Stillwater. The template and content of the manual is written closely following the OCC SOPs (OCC, 2014). Sections of this manual are directly quoted from the OCC SOPs and are noted as such. This manual serves as

a guideline for sampling every parameter for the beneficial uses for the streams in this project. The manual contains instructions for using a chain of custody and labeling sample bottles, measuring flow with a flow meter, taking inorganic and bacteria bottle samples, taking quality assurance samples, how to sample with the YSI probe, and macroinvertebrate and fish collection instructions. All samples should be taken under antecedent base flow conditions to ensure sample accuracy. Rainfall runoff events and other unique conditions can skew sample data. Flow is a necessary measurement for some attainment calculations for meeting water quality standards. Attainment analysis requirements can be found in the Integrated Report, (ODEQ, 2020).

The sections of this manual are necessary for taking samples following this WQMP. The EPA has requirements for data collection methodology for data to be submitted in the Integrated Report. Following the procedures in this manual ensures that data collected by the city is acceptable for submission to the Integrated Report. This sampling manual applies to both frequencies of sampling outlined by this project. The manual can be used for both the recommended comprehensive OCC sampling frequency or the minimal sampling frequency required by the EPA for data submission to the Integrated Report.

CHAPTER IV

DATA ANALYSIS

Samples were taken using the YSI probe for the duration of this project. Budget restrictions due to the COVID-19 pandemic did not allow for samples to be analyzed by third party contractors. The only parameters measured were those that the ESGP department YSI probe measured. These parameters include Conductivity (uS/cm), Specific Conductance (uS/cm), Salinity (psu), Total Dissolved Solids (mg/L), Temperature (°F), Resistivity (ohms-cm), Turbidity (FNU), Dissolved Oxygen (% Sat, mg/L), Chlorophyll (RFU, ug/L), Phycocyanin (RFU, ug/L), Pressure (psi a), Depth (m), and Vertical Position (m). The following table shows the parameters that are necessary to analyze for attainment purposes for the waterbodies that were sampled.

Beneficial Uses per OAC 785:45-5 & 785: Appendix A

Waterbody ID	Waterbody Name	Size (miles)	Category	Aes	Ag	HLAC	WWAC	FC	PBCR	SBCR	PPWS	EWS	SWS
OK620900040070_10	Stillwater Creek	16.43	5a	•	•	•		•		•	•	•	
OK620900040140_00	Boomer Creek	2.28	5c	•	•		•	•	•		•		
OK620900040180_00	Boomer Creek	6.49	3	•	•		•	•	•		•		*
OK620900040150_00	Sanborn-Hazen Lake Creek	3.59	5c	•	•		•	•	•				
OK620900040195_00	Duck Creek	3.06	3	•	•		•	•	•				
OK620900040200_00	Cow Creek	8.26	5c	•	•		•	•	•				
OK620900040230_00	Stillwater Creek, North	6.8	3	•	•		•	•	•		•		*

Beneficial Use	Parameters Tested
Aesthetic	Nutrients, Oil & Grease
Agriculture	Total Dissolved Solids, Chlorides, Sulfates,
Habitat Limited Aquatic Community	Dissolved Oxygen, Toxicants, pH, Fish Collection, Macroinvertebrates, Turbidity, Oil & Grease, Sediment
Warm Water Aquatic Community	Dissolved Oxygen, Toxicants, pH, Fish Collection, Macroinvertebrates, Turbidity, Oil & Grease, Sediment
Fish Consumption	Using long-term average numerical parameters
Primary Body Contact Recreation	E. coli, Enterococci
Secondary Body Contact Recreation	E. coli, Enterococci
Public and Private Water Supply	Toxicants, Total Coliform, Oil & Grease, Chlorophyll-α/Total Phosphorus
Emergency Water Supply	**All waterbodies designated Emergency Water Supply beneficial use shall be deemed to be attaining the beneficial use for all water quality related issues.
Sensitive Water Supply	See *OAC 785:45-5-25-a-4*

Aes = Aesthetic
Ag = Agriculture
HLAC = Habitat Limited Aquatic Community
WWAC = Warm Water Aquatic Community
FC = Fish Consumption
PBCR = Primary Body Contact Recreation
SBCR = Secondary Body Contact Recreation
PPWS = Public and Private Water Supply
EWS = Emergency Water Supply
SWS = Sensitive Water Supply

*The red text indicates parameters measured during this project

Table 7. Beneficial Uses of Waterbodies and YSI Probe Sampling Parameters. Information in this table was obtained from the Oklahoma 2020 Integrated Report (ODEQ, 2020).

The following section includes the data collected for TDS, DO, Turbidity, and Chlorophyll- *a* for each of the waterbodies in this project. The data is analyzed using water quality standards for each parameter. The data determines if each waterbody meets attainment for the given parameter. Data points that do not meet water quality standards are bolded in the tables. Water quality standards used for attainment determination can be found in the Integrated Report (ODEQ, 2020).

Stillwater Creek OK620900040070_10

Agriculture

Total Dissolved Solids (mg/L): Attained

Sample Date	Stillwater Creek A	Stillwater Creek B	Stillwater Creek C

2/29/20	349	445	494
3/30/20	263	290	312
4/30/20	336	374	(not sampled)
5/31/20	427	504	542
7/31/20	265	261	239
9/2/20	693	503	339
9/29/20	693	**761**	551
10/30/20	194	185	179
1/28/21	513	494	478
2/24/21	558	593	586
3/25/21	302	314	323
5/1/21	327	340	(thrown out)

Only one value exceeds 700 mg/L. This requires that attainment be measured by the mean of all TDS samples not exceeding the yearly mean standard (YMS) as found in the OAC 785:45 Appendix F. The TDS samples must also not exceed the sample standard (SS) for TDS as found in OAC 785:45 Appendix F by 10% of the samples taken.

Habitat Limited Aquatic Community

Dissolved Oxygen (mg/L): Not Attained

Sample Date	Stillwater Creek A	Stillwater Creek B	Stillwater Creek C
2/29/20	11.3	11.5	11.8
3/30/20	9.5	9.4	9.4
4/30/20	8.5	8.7	(not sampled)
5/31/20	5.9	6.5	7.9
7/31/20	3.1	4.1	6.4
9/2/20	**0.7**	**2.3**	5.4
9/29/20	**1.9**	**2.9**	5.9
10/30/20	9.0	10.3	10.9
1/28/21	11.9	12.6	11.9
2/24/21	14.6	12.3	12.5
3/25/21	10.4	10.2	10.1
5/1/21	8.3	8.3	9

HLAC is considered not attained if more than 10% of the samples have DO concentrations less than 4.0 mg/L from April 1 – June 15 (3.0 mg/L from June 16 – March 31)

Turbidity (NTU): Not Attained

Sample Date	Stillwater Creek A	Stillwater Creek B	Stillwater Creek C
2/29/20	20	15.3	10.5
3/30/20	27.1	33.0	43.2
4/30/20	30.4	33.6	(not sampled)

5/31/20	44.2	41.8	33.5
7/31/20	**94.8**	**73.6**	**53.6**
9/2/20	24.7	25.9	22.4
9/29/20	27.9	6.5	13.0
10/30/20	**69.7**	**55.8**	**64.1**
1/28/21	26.2	27.7	39.1
2/24/21	10.9	12.7	13.3
3/25/21	33.5	41.7	**57.7**
5/1/21	31.4	32.5	12.5

Turbidity is not attained because more than 10% of the samples measured greater than 50 NTUs.

Public and Private Water Supply

Chlorophyll - *a*: N/A

Stillwater Creek is not listed in OAC 785:45-5-10(7) as a waterbody that utilizes chlorophyll – *a* as a parameter to determine PPWS use attainment.

Boomer Creek OK620900040180_00

Agriculture

Total Dissolved Solids (mg/L): Attained

Sample Date	Boomer A	Boomer B
2/29/20	492	235
3/30/20	368	264
4/30/20	469	287
5/31/20	468	310
7/31/20	256	256
9/2/20	297	268
9/29/20	333	439
10/30/20	272	283
1/28/21	363	286
2/24/21	432	320
3/25/21	432	309
5/1/21	423	356

No sample value exceeds 700 mg/L. Therefore, the TDS parameter is attained.

Warm Water Aquatic Community

Dissolved Oxygen (mg/L): Attained

Sample Date	Boomer A	Boomer B
2/29/20	11.5	10.9
3/30/20	8.3	8.8
4/30/20	6.4	7.5
5/31/20	**5.7**	**4.6**
7/31/20	6.2	7.2
9/2/20	**4.6**	**4.9**
9/29/20	5.7	5.2
10/30/20	10.7	11.4
1/28/21	12.2	13.1
2/24/21	13.2	12.8
3/25/21	9.4	11.4
5/1/21	6.2	8.0

WWAC is not attained if more than 10% of the samples have DO concentrations less than 6.0 mg/L from April 1 – June 15 (5.0 mg/L from June 16 – March 31)

Turbidity (NTU): Attained

Sample Date	Boomer A	Boomer B
2/29/20	12.4	6.2
3/30/20	18.0	10.9
4/30/20	28.4	17.1
5/31/20	13.6	15.9
7/31/20	13.3	18.7
9/2/20	5.7	12.4
9/29/20	3.8	5.6
10/30/20	26.4	6.4
1/28/21	21.6	7.0
2/24/21	8.1	4.0
3/25/21	24.9	9.1
5/1/21	14.9	16.4

Turbidity is attained because 10% or fewer of the samples measured greater than 50 NTUs.
Public and Private Water Supply

Chlorophyll – *a* (mg/L): Not Attained

Sample Date	Boomer A	Boomer B
2/29/20	**0.014**	**0.012**
3/30/20	0.010	**0.016**
4/30/20	0.007	**0.012**
5/31/20	0.006	0.007
7/31/20	**0.030**	**0.014**
9/2/20	**0.023**	0.007

9/29/20	0.005	0.005
10/30/20	0.006	**0.015**
1/28/21	0.007	0.009
2/24/21	0.004	0.006
3/25/21	0.008	**0.016**
5/1/21	0.007	**0.013**

Waterbodies that are given the beneficial use of Sensitive Water Supply (SWS) must not exceed 0.010 mg/L at a depth of 0.5 meters below the surface for a long-term average. The average value of the samples collected so far is 0.01083. This average exceeds the maximum value for chlorophyll- α and therefore the parameter is not attained.

Sanborn-Hazen Lake Creek OK620900040150_00

Agriculture

Total Dissolved Solids (mg/L): Not Attained

Sample Date	Sanborn-Hazen Lake Creek
2/29/20	**722**
3/30/20	607
4/30/20	510
5/31/20	576
7/31/20	302
9/2/20	219
9/29/20	**760**
10/30/20	204
1/28/21	459
2/24/21	584
3/25/21	403
5/1/21	640

Two sample values exceed 700 mg/L. This requires that attainment be measured by the mean of all TDS samples not exceeding the yearly mean standard (YMS) as found in the OAC 785:45 Appendix F. The TDS samples must also not exceed the sample standard (SS) for TDS as found in OAC 785:45 Appendix F by 10% of the samples taken. Two of the twelve samples taken exceed 700 mg/L indicating that more than 10% of the samples exceed the maximum standard. This parameter is not attained.

Warm Water Aquatic Community

Dissolved Oxygen (mg/L): Not Attained

Sample Date	Sanborn-Hazen Lake Creek
2/29/20	10.9

3/30/20	7.7
4/30/20	**5.9**
5/31/20	**4.7**
7/31/20	6.6
9/2/20	**3.3**
9/29/20	6.9
10/30/20	11.5
1/28/21	12.6
2/24/21	10.5
3/25/21	10.5
5/1/21	**4.2**

WWAC is considered not attained if more than 10% of the samples have DO concentrations less than 6.0 mg/L from April 1 – June 15 (5.0 mg/L from June 16 – March 31)

Turbidity (NTU): Attained

Sample Date	**Sanborn-Hazen Lake Creek**
2/29/20	8.0
3/30/20	12.1
4/30/20	10.0
5/31/20	12.6
7/31/20	34.2
9/2/20	10.3
9/29/20	5.2
10/30/20	19.0
1/28/21	12.9
2/24/21	4.5
3/25/21	21.0
5/1/21	15.4

Turbidity is attained because 10% or fewer of the samples measured greater than 50 NTUs.

Duck Creek OK620900040195_00

Agriculture

Total Dissolved Solids (mg/L): Not Attained

Sample Date	**Duck Creek**
2/29/20	**1006**
3/30/20	**996**
4/30/20	623
5/31/20	661

7/31/20	Not sampled
9/2/20	352
9/29/20	598
10/30/20	223
1/28/21	**855**
2/24/21	**907**
3/25/21	**874**
5/1/21	**1077**

Six sample values exceed 700 mg/L. This requires that attainment be measured by the mean of all TDS samples not exceeding the yearly mean standard (YMS) as found in the OAC 785:45 Appendix F. The TDS samples must also not exceed the sample standard (SS) for TDS as found in OAC 785:45 Appendix F by 10% of the samples taken. Six of the eleven samples taken exceed 700 mg/L indicating that more than 10% of the samples exceed the maximum standard. This parameter is not attained.

Warm Water Aquatic Community

Dissolved Oxygen (mg/L): Attained

Sample Date	**Duck Creek**
2/29/20	14.4
3/30/20	9.0
4/30/20	11.6
5/31/20	14.3
7/31/20	Not sampled
9/2/20	6.4
9/29/20	8.5
10/30/20	11.3
1/28/21	13.8
2/24/21	12.8
3/25/21	14.1
5/1/21	**4.9**

WWAC is considered attained if 10% or fewer of the samples have DO concentrations less than 6.0 mg/L from April 1 – June 15 (5.0 mg/L from June 16 – March 31)

Turbidity (NTU): Attained

Sample Date	**Duck Creek**
2/29/20	0.8
3/30/20	1.7
4/30/20	1.3
5/31/20	2.4
7/31/20	Not sampled

9/2/20	7.3
9/29/20	5.9
10/30/20	12.6
1/28/21	4.4
2/24/21	1.9
3/25/21	2.9
5/1/21	1.6

Turbidity is attained because 10% or fewer of the samples measured greater than 50 NTUs.

Cow Creek OK620900040200_00

Agriculture

Total Dissolved Solids (mg/L): Not Attained

Sample Date	Cow A	Cow B
2/29/20	684	668
3/30/20	655	640
4/30/20	**744**	**708**
5/31/20	614	529
7/31/20	Not sampled	258
9/2/20	438	**703**
9/29/20	Not sampled	637
10/30/20	106	135
1/28/21	304	299
2/24/21	364	438
3/25/21	413	366
5/1/21	677	663

Three sample values exceed 700 mg/L. This requires that attainment be measured by the mean of all TDS samples not exceeding the yearly mean standard (YMS) as found in the OAC 785:45 Appendix F. The TDS samples must also not exceed the sample standard (SS) for TDS as found in OAC 785:45 Appendix F by 10% of the samples taken. Three of the 22 samples taken exceed 700 mg/L indicating that more than 10% of the samples exceed the maximum standard. This parameter is not attained.

Warm Water Aquatic Community

Dissolved Oxygen (mg/L): Not Attained

Sample Date	Cow A	Cow B
2/29/20	11.4	11.4
3/30/20	8.1	8.5
4/30/20	7.0	6.2

5/31/20	**4.3**	**5.9**
7/31/20	Not sampled	**4.1**
9/2/20	**3.6**	**1.9**
9/29/20	Not sampled	**3.0**
10/30/20	9.8	10.2
1/28/21	10.6	11.1
2/24/21	11.4	12.4
3/25/21	10.0	9.8
5/1/21	7.6	6.7

WWAC is considered not attained if more than 10% of the samples have DO concentrations less than 6.0 mg/L from April 1 – June 15 (5.0 mg/L from June 16 – March 31)

Turbidity (NTU): Not Attained

Sample Date	**Cow A**	**Cow B**
2/29/20	5.6	11.7
3/30/20	10.8	14.2
4/30/20	5.7	14.2
5/31/20	16.2	15
7/31/20	Not sampled	20.4
9/2/20	3.7	8.4
9/29/20	Not sampled	8.6
10/30/20	47.0	**68.0**
1/28/21	42.4	**65.4**
2/24/21	16.6	17.7
3/25/21	16.7	30.7
5/1/21	12.1	12.0

Turbidity is not attained because more than 10% of the samples measured greater than 50 NTUs.

North Stillwater Creek OK620900040230_00

Agriculture

Total Dissolved Solids (mg/L): Attained

Sample Date	**NSC A**	**NSC B**
4/30/20	610	593
5/31/20	535	325
7/31/20	Not sampled	281
9/2/20	Not sampled	264
9/29/20	Not sampled	276
10/30/20	157	127

1/28/21	236	249
2/24/21	339	323
3/25/21	367	375
5/1/21	501	491

No sample value exceeds 700 mg/L. Therefore, the TDS parameter is attained.

Warm Water Aquatic Community

Dissolved Oxygen (mg/L): Attained

Sample Date	**NSC A**	**NSC B**
4/30/20	9.2	6.8
5/31/20	6.8	10.1
7/31/20	Not sampled	**4.8**
9/2/20	Not sampled	7.4
9/29/20	Not sampled	5.1
10/30/20	9.6	10.4
1/28/21	13.1	12.8
2/24/21	11.6	10.0
3/25/21	10.1	9.5
5/1/21	8.0	8.3

WWAC is considered attained if 10% or fewer of the samples have DO concentrations less than 6.0 mg/L from April 1 – June 15 (5.0 mg/L from June 16 – March 31)

Turbidity (NTU): Not Attained

Sample Date	**NSC A**	**NSC B**
4/30/20	43.6	22.6
5/31/20	**64.1**	29.5
7/31/20	Not sampled	**72.1**
9/2/20	Not sampled	37.5
9/29/20	Not sampled	**57.4**
10/30/20	**169.0**	**200.2**
1/28/21	**133.2**	**106.0**
2/24/21	48.2	46.0
3/25/21	**67.7**	**86.1**
5/1/21	**86.7**	32.5

Turbidity is not attained because more than 10% of the samples measured greater than 50 NTUs.

Beneficial Uses per WQS 785:45-5 & 785: Appendix A

Waterbody ID	Waterbody Name	Size (miles)	Ag	HLAC	WWAC	PPWS
OK620900040070_10	Stillwater Creek	16.43	TDS: A	DO: NA Turbidity: NA		
OK620900040180_00	Boomer Creek	6.49	TDS: A		DO: A Turbidity: A	Chlorophyll- a: NA
OK620900040150_00	Sanborn-Hazen Lake Creek	3.59	TDS: NA		DO: NA Turbidity: A	
OK620900040195_00	Duck Creek	3.06	TDS: NA		DO: A Turbidity: A	
OK620900040200_00	Cow Creek	8.26	TDS: NA		DO: NA Turbidity: NA	

Table 8. Attainment of beneficial uses for 2020-2021 samples collected from streams in Stillwater, OK.

The samples collected during this project suggest that every tested waterbody is impaired for at least one parameter. Stillwater Creek is impaired for DO and turbidity. This matches the findings reported in the 2020 Integrated Report (ODEQ, 2020). Boomer Creek is impaired for chlorophyll – *a*. Sandborn-Hazen Lake Creek is impaired for TDS and for DO. Duck Creek is impaired for TDS. Cow Creek is impaired for all three parameters tested with the YSI probe: TDS, DO, and turbidity.

CHAPTER V

FUTURE CONSIDERATIONS

Beneficial Use Changes

Some of the beneficial uses listed for the streams in this project were in question as the water quality monitoring program was designed. The ESGP and Zack Henson determined that some of the beneficial uses with stringent monitoring requirements don't meet the city's use for that waterbody. The Emergency Water Supply (EWS) beneficial use given to Stillwater Creek may be determined as an unsupported beneficial use by the city of Stillwater. The attainment parameters for EWS states that "all waterbodies designated EWS beneficial use shall be deemed to be attaining the beneficial use for all water quality related issues" (OAC 785:45). This description requires that the EWS meet water quality standards in every way possible.

The OWRB is the entity that assigns beneficial uses to waterbodies. Jade Jones of the Water Quality Standards division of OWRB provided information regarding the process to change beneficial uses for waterbodies. The process is "both a technically and regulatory rigorous process and in the majority of cases this is not a viable option; however, it has and can be done under the right circumstances" (Jones, 2021). A Use Attainability Analysis would be required to change a beneficial use. More information about Use Attainability Analysis can be found on the EPA website. The city of Stillwater may pursue the process of changing the beneficial use of a waterbody by providing more information and specific details to the OWRB.

Data Submission

Water quality data can be submitted to the Oklahoma Department of Environmental Quality (ODEQ) every two years for the Integrated Report. Data submission requirements and email address or mailing address for submission can be found on the ODEQ website under the "Public Solicitation of Water Quality Data for the *year* Integrated Report" (ODEQ, 2021). Only data collected before April 30, 2019 was used in the 2020 Integrated Report submission. The data collected for this project will need to be submitted for the 2022 Integrated Report.

Macroinvertebrate Sampling

Current macroinvertebrate sampling events are performed by Blue Thumb crews on the streams in this project. Blue Thumb is a citizen science organization under the water quality division of the OCC. They train volunteers to take water samples on streams. The macroinvertebrate sampling that Blue Thumb performs is the only parameter that they submit for the Integrated Report (OCC, 2015). Partnering with Blue Thumb for future macroinvertebrate sampling events could potentially benefit the city. Sample sites for this project have been strategically chosen and are recommended for future macroinvertebrate sampling. Joint macroinvertebrate sampling with Blue Thumb at these chosen sample sites would unify water quality monitoring efforts in the city and improve accuracy. This could reduce the potential cost of macroinvertebrate sampling for the city as well as ensure accuracy in sampling. Blue Thumb is operated by the OCC and therefore follows the OCC macroinvertebrate sampling procedures.

CHAPTER VI

CONCLUSION

The ESGP recommends that the city of Stillwater follow this WQMP for data collection to be submitted for the Integrated Report. The comprehensive sampling frequency following OCC QAPPs is recommended for the most accurate data collection and analysis. This WQMP will cost the city a total of $40,150 annually for sample processing with an initial equipment cost of $8,550. The minimal sampling frequency can be used for the WQMP if budget requirements are not met for the recommended frequency. The minimally effective frequency required by the EPA for the Integrated Report would cost the city $10,300 for sample processing annually with an initial equipment cost of $8,550. These cost estimates do not include the cost of labor for sample collection. It should be noted that the more data collected for analysis of beneficial use attainment, results in the most accurate water quality determination of a waterbody (Dressing, 2005). Following the minimal sampling frequency means that only two samples are taken per year at each sample site. This could result in an inaccurate water quality analysis if one or both of the chosen sampling dates are not representative of the average water quality in that waterbody. If a sampling event takes place in unusual weather conditions such as drought or following a rainfall runoff event, the data collected would not be representative of the typical conditions for the sampled waterbodies and could result in a skewed attainment analysis. More sampling events per year allow for more accurate averages and can compensate for unusual conditions during sampling events.

Water quality monitoring is a vital part of the stormwater management program because it serves as a measure by which stormwater drainage can be evaluated. All six streams in this WQMP have been determined to be impaired for at least one parameter according to the data collected during this project. The data provided by this project serves as a foundation for measuring the impact of the current stormwater infrastructure on surface water quality. The WQMP created for the city satisfies the MS4 permit monitoring requirements. Data collected from this WQMP can be submitted to the Integrated Report and can be a tool for improving water quality in the future.

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APPENDICES

APPENDIX A: Sampling Manual

SAMPLING MANUAL

Standard Operating Procedures
AND
Quality Assurance/Quality Control Measures
FOR
Water Quality Monitoring and Measurement Activities

Adapted from the Oklahoma Conservation Commission SOPs

CHAIN OF CUSTODY AND SAMPLE LABELING

1.0 Procedural Section

1.1 Scope and Application
A Chain of Custody form is necessary to ensure that all parties involved in the handling of a sample understand what the sample is for, when the sample was taken, and where the sample was taken from. The associated laboratory that facilitates the testing of the sample should provide a Chain of Custody form that will then be filled out by the individual collecting the sample. All samples should be labeled clearly so that the sample can be accurately analyzed (OCC, 2014).

1.2 Summary of Method
The Chain of Custody form provides record of who has possession of each sample. This ensures the ability to track where the sample has been and at what time. This process allows the integrity of the sample to remain intact and ensures that only the appointed personnel have had access to the sample at a given time. This document serves as a reference for quality assurance purposes and data accuracy (OCC, 2014).

1.2.1 Definitions
- Possession: A person has "possession" of a sample if it is in that person's physical custody, has been stored in a tamper-proofed manner by that person, or is stored in a location restricted to that individual (OCC, 2014)

1.3 Health and Safety Warnings
None

1.4 Cautions
None

1.5 Interference
- Illegible writing
- Where applicable: insufficient pen pressure causing carbon copy to be illegible

1.6 Personnel Qualification
Field sample collectors are required to be trained in the filling out of a Chain of Custody form. The training must be approved by the entity providing the Chain of Custody form as well as the supervising authority with the city of Stillwater.

1.7 Apparatus & Materials
- Chain of Custody form
- Permanent marker for sample labels
- Ball point pen for COC form

1.8 Procedure

1.8.1 Sample labeling:

Each sample should contain the following information at a minimum. The entity providing the COC may require more information. Label the sample bottle at the time of collection.

- Site ID
- Sample date
- Sample time
- Preservatives used (if any)
- Field collector's name

2.0 QA/QC Section

2.1 Training

All sampling personnel are required to be trained in all operating procedures and will be monitored in practicing such procedures before solo field operation.

2.2 Maintenance

N/A

2.3 QC Procedures

N/A

FLOW MEASUREMENT
(FP311 Global Water Flow Probe)

1.0 Procedural Section

1.1 Scope and Application
Flow is the measure of water volume discharge per unit time. This is calculated by determining the water velocity which is the change in distance per unit time (OCC, 2014).

1.2 Summary of Method
"The Global Water Flow Probe is a rugged and highly accurate water velocity instrument for measuring flows in open channels and partially filled pipes. The water velocity probe consists of a protected propeller and water bearing for measuring water velocity, coupled to a telescoping probe handle ending in with a LCD display flow computer. The Flow Probe is ideal for storm water runoff studies, sewer flow measurements, measuring flows in rivers and streams, and monitoring water velocity in ditches and canals" (Global Water, 2021).

1.2.1 Definitions
Flow = volume/time

1.3 Health and Safety Warnings
The flow measurement should not be made by wading into the stream when the velocity of the stream is high and/or the stream depth is too deep. Wearing waders can be dangerous in deep or high flowing waters if they fill with water. Flow measurements should always be made with at least two field agents present (OCC, 2014).

1.4 Cautions
Take care when using the probe and prevent the digital monitor from submerging in the water. Keep the base of the probe (with the propeller) from hitting rough substrate or getting tangled in debris.

1.5 Interference
Debris can interfere with probe readings and result in inaccurate data. Low flow that cannot be detected by the probe can also interfere with results.

1.6 Personnel Qualification
Field operators are required to be trained in the use of the flow meter from an experienced supervising field agent. Practice sessions will be monitored before the agent takes flow measurements without supervision. All agents must familiarize themselves with the flow meter user manual before training.

1.7 Apparatus & Materials
- FP311 Global Water Flow Probe
- Spare batteries

1.8 Instrument/Method Calibration
Reference the flow meter User Manual

1.9 Equipment Operation & Preparation
Reference the flow meter User Manual for operation.

1.9.1 Wading Rod
(This section is taken directly from the OCC SOP Manual)

Two accepted methods for determining velocities are as follows:

- Measure the velocity at 60% of the depth (from the top) and use this as the mean

- Measure the velocity at 20% and 80% of the depth (from the top). Use the average of these velocities as the mean.

The purpose of the top setting wading rod is to conveniently set the sensor at 20%, 60%, or 80% of the total depth. The total depth can be measured using the gauge rod. The rod is divided into feet and tenths of feet (not inches). Each single mark represents 0.10 ft, each double mark represents 0.50 ft, and each triple mark represents 1.00 ft.

The wading rod is designed to facilitate the determination of the correct sensor depth as shown in the following examples (Refer to Figure2):

To calculate 60% of depth from the top:

1. Determine depth of segment to be measured using the gradations on the wading rod (e.g. 2.7 ft).
2. Slide smaller rod up until the "2" on the rod lines up with the “7” on the rod handle.

To calculate 80% of depth from the top:

3. Determine depth of segment to be measured (e.g. 2.7 feet).
4. Divide depth by 2 = 1.35.
5. Slide small rod until "1" on the rod lines up with the 3.5 on the rod handle.

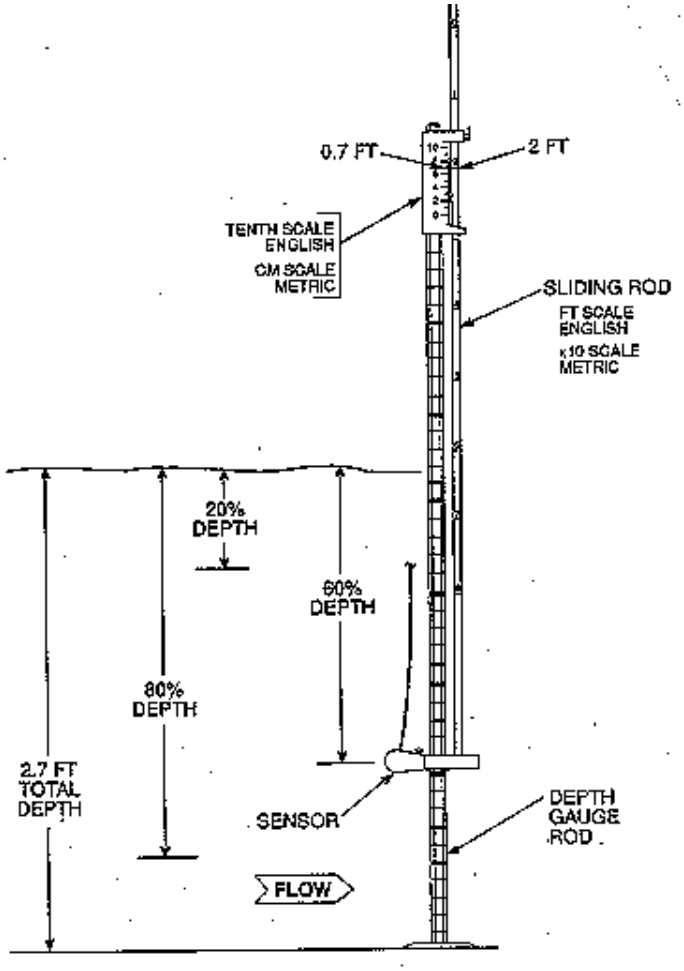
To calculate 20% of the depth from the top:

6. Determine depth of segment to be measured (e.g. 2.7 feet).
7. Multiply depth by 2 = 5.4 feet.
8. Slide small rod until "5" on the rod lines up with the 4 on the rod handle.

Using the 60% method by itself to determine velocity is probably the least accurate option because it assumes that there is consistent flow throughout the depth profile. However, in shallow waters, this is an acceptable method because it may not be practical

to measure velocity at other depths. Therefore, for OCC purposes, the selection of the method depends on the depth of the water column.

1. If the depth is less than 1.5 feet, the velocity should be measured at 60% of the profile from the surface.
2. If the depth is greater than or equal to 1.5 feet, the velocity should be measured at 20% and 80% of the profile from the surface and averaged.



1.10 Sample Collection

1.10.1 Meter Setting

Reference the flow meter User Manual for proper meter setting for samples.

1.10.2 Site Selection and Preparation

Sampling sites should be in an area of the stream where flow appears to be uniform across the stream. The section of the stream should not have a drastic change in the stream floor or be where there is significant riffles or uneven substrate. The narrowest section of smaller streams is ideal to decrease the number of measurements (OCC, 2014).

Samples should be taken across the stream's width in equally divided segments. This will give a more accurate average flow measurement than one single measurement. Segments of about 1 foot should be divided across the stream unless the stream is less than 20 feet across and then segments should be no shorter than 0.5 feet.

In most scenarios the stream depth will not be adequate at 1 foot from the bank on either side, so the first measurement should be taken in stream as far as an adequate depth is reached.

1.10.3 Measurement Procedure

1. Turn meter on.
2. Stretch a graduated string or tape from bank to bank on the stream cross section to be sampled.
3. Divide the distance into equal segments so that each segment account for 5% of the stream.
4. Place the wading rod at the first interval with the sensor pointed upstream directly into the current. Stand behind and to the side of the wading rod.
5. Measure depth.
6. Adjust wading height based on water depth. (<1.5 ft take reading at 60% of depth; >1.5 ft take readings at 20% and 80% of depth).
7. Hold the rod steady while the reading is being measured. Once the reading has stabilized or averaged, record the reading on the probe (OCC, 2014).

1.11 Sample Handling & Preservation

N/A

1.12 Sample Preparation and Analysis

N/A

1.13 Troubleshooting

Reference User Manual

1.14 Data Acquisition, Calculation & Data Reduction

N/A

1.15 Computer Hardware & Software

N/A

1.16 Chain of Custody Procedure

N/A

2.0 QA/QC

2.1 Training

All sampling personnel are required to be trained in all operating procedures and will be monitored in practicing such procedures before solo field operation.

2.2 Maintenance

- Attempt to keep digital monitor from being submerged in water
- Clear debris from propeller and rinse with water if necessary
- Keep flow meter in protective canister when not in use
- Reference the User Manual for further maintenance

2.3 QC Procedures

The meter should have a reading of “0” before every sampling

INORGANIC AND BACTERIA SAMPLE COLLECTION

1.0 Procedural Section

1.1 Scope and Application
The proper sampling technique is pertinent to gathering accurate water quality data. Water samples that are collected and then taken to a lab should be as small as possible to be transported easily, while still maintaining an adequate sample amount for analysis (OCC, 2014)

1.2 Summary of Method
This collection method outlines the correct way to obtain a water sample in a bottle for bacteria and inorganic analysis. The bacteria analysis is for total coliform analysis and *Escherichia coli*. The bacteria analysis is only performed from May 1 – September 30 during the recreational period. The inorganic analysis consists of total phosphorus, ortho-phosphorus, chloride, sulfate, nitrate, nitrite, ammonia, total suspended solids, acidity, total dissolved solids, and most cations (metals: Mn, Mg, Fe, Al, Ca). (OCC, 2014)

1.2.1 Definitions
- Grab Samples: A sample collected at a specific place and time
- Composite Samples: A combination of various grab samples taken at different time intervals or locations
- Integrated Samples: A combination of various grab samples taken at different points at the same time (OCC, 2014)

1.3 Health and Safety Warnings
- Some sample bottles may contain an acidic reagent or preservative. Always wear proper gloves, eye protection, and clothing that covers the limbs to prevent bodily harm.
- Sample collections should be made with caution in swift or deep waters (OCC, 2006).

1.4 Cautions
- Only use new sample bottles or certified clean bottles when applicable
- Keep bottles sealed and sterile until time of sampling to prevent contamination
- Do NOT rinse bottles prior to sampling for bacteria collection
- Avoid taking samples near man-made structures that could alter the sample integrity
- Always collect enough sample requested by the lab facility for the proper analysis (OCC, 2014)

1.5 Interference
- Contaminated bottles or pipettes
- Improper preservation conditions for sample bottles

1.6 Personnel Qualification
Field operators are required to be trained in the proper method of sample collection from an experienced supervising field agent. Practice sessions will be monitored before the agent takes

samples without supervision. All agents must familiarize themselves with proper sampling techniques prior to training.

1.7 Apparatus & Materials

- Bottles supplied by laboratory facility
- Ice if necessary
- Coolers (may be provided by lab facility)
- Permanent marker for labeling

1.8 Instrument/Method Calibration

N/A

1.9 Preparation

Preparation of sample bottles prior to sampling will be specified by the laboratory facility

1.10 Sample Collection

1.10.1 Inorganic Grab Sample

1. The sample should be taken where flow is the most uniform and is upstream of riffles. The collection should also take place upstream from man-made structures to avoid contamination. The sample should also be taken in the same section of stream as other samples in the sample site.
2. Make sure to label the bottle with permanent marker prior to sampling. Mark all minimum required labels and those specified by the lab.
3. Make sure to face upstream as not to allow interference from sediment that may have been disturbed while wading. Rinse the bottle and cap 3 times with the same area that the sample will be taken.
4. Place the sample bottle underwater so that the mouth of the bottle is 6 inches below the surface of the water. Position the bottle so that it is upright, and the neck is pointing toward the flow of water. Be sure to not collect any surface scum
5. Use a separate smaller container for collecting the sample into a larger vessel if the stream depth is too shallow to submerge the sample container.
6. Be sure to leave as much air space as requested by the lab for each sample bottle. If no air is required, fully submerge the bottle and screw the cap on while the bottle is submerged.

1.10.2 Bacteria Sample Collection (only performed during the recreational period May 1 – September 30)

1. Label the sample bottle with necessary labels using a permanent marker
2. Denote on the label if there is increased cattle activity or high flow during sampling to notify the lab that the sample may need to be diluted prior to analysis.
3. Do not rinse the bottle. Unscrew the cap and submerge the bottle with the opening facing down and fill the bottle slowly with the opening facing slightly upstream. Make sure your arm and hands are behind the flow, not in front of the opening.
4. Remove the bottle with the opening facing upward and cap the bottle.
5. Place the sample on ice immediately and ensure that there is no water for an ice water bath in the cooler.

6. Do not allow direct contact with sunlight or ice (OCC, 2014)

1.11 Sample Handling & Preservation
Follow protocol of the testing laboratory for proper handling of sample bottles and preservation.

1.12 Sample Preparation and Analysis
Follow protocol of the testing laboratory for proper sample preparation.

1.13 Troubleshooting
Contact testing laboratory or supervisor.

1.14 Data Acquisition, Calculation & Data Reduction
N/A

1.15 Computer Hardware & Software
N/A

1.16 Data Management & Records Management
All samples should be recorded on the Chain of Custody supplied by the testing laboratory. Keep a copy of the COC for record.

2.0 QA/QC SECTION

2.1 Training
All sampling personnel are required to be trained in all operating procedures and will be monitored in practicing such procedures before solo field operation.

2.2 Maintenance
N/A

2.3 Quality Assurance Measures
Blanks, spikes, duplicates, and replicate samples should be taken when indicated (See "Spike, Duplicate, Replicate, and Blank Samples/Measurements for Routine QA" section)

SPIKE, DUPLICATE, REPLICATE, AND BLANK SAMPLES/MEASUREMENTS FOR ROUTINE QA

1.0 Procedural Section

1.1 Scope and Application
Quality assurance samples must be taken to evaluate potential bias and accuracy of sampling by the field agent. Spikes, duplicates, replicates, and blanks should be collected at each sampling event to monitor these measures (OCC, 2014).

1.2 Summary of Method

1.2.1 Definitions
QA/AC SAMPLES COLLECTED

- Blank: De-ionized water is obtained from the lab facility and brought to the field. The field agent will rinse a sample bottle 3 times with the de-ionized water and then fill the bottle with the de-ionized water. The sample bottle will then be handled and preserved in the same manner as the other sample bottles. The blank sample will then be analyzed to determine if any contamination takes place during the sampling process.

- Split/DUP: This process requires that two separate samples for the same sample site and parameter be obtained to ensure that the results of analysis are the same. This would reveal any potential field error.

- Replicate: These samples are taken in different locations or at different times. A spatial replicate is preferred to evaluate a stretch of the same stream at least 50 meters from the sampling site of an area with similar habitat. This analysis can account for potential variation in sample results depending on stream location.

- Spike: This sample requires an added known reagent to a grab sample to evaluate the accuracy of analysis. This sample is only taken for specific project QAPPs (OCC, 2014).

FIELD QA/QC READINGS

- Conductivity, pH, DO, and Turbidity: Perform replicate measurement recordings for these parameters in-situ

1.3 Health and Safety Warnings
N/A

1.4 Cautions
N/A

1.5 Interferences
N/A

1.6 Personnel Qualification
Field operators are required to be trained in the proper method of sample collection from an experienced supervising field agent. Practice sessions will be monitored before the agent takes samples without supervision. All agents must familiarize themselves with proper sampling techniques prior to training.

1.7 Apparatus & Materials
N/A

1.8 Procedure
These QA measures should be taken at each sampling episode at one sample location. Be sure to choose a site for the QA samples that is not experiencing unusual or extreme conditions to reduce variability (OCC, 2014).

1.8.1 Preparation or Measurement of Blank Samples
De-ionized water should be obtained from the lab facility prior to the sampling event, but shortly before the sampling event. The water should be stored in a polyethylene bottle that has been rinsed 3 times with the same deionized water.

The bottle should be labeled as other samples in the field, but with the label of "field blank" where the stream name would normally be written. The blank bottle should be rinsed 3 times with the deionized water and then filled with the deionized water. The blank bottle with acid reagent in it should also be filled with deionized water. The blank bottle samples should then be treated the same as the other samples from that sampling event (OCC, 2014).

1.8.2 Preparation or Measurement of Split/Duplicate Samples
Label one sample bottle for acid preservation and another for ice preservation. Take grab samples following the "Inorganic and Bacteria Sample Collection" section of the sampling manual (OCC, 2014).

1.8.3 Preparation for Measurement of Replicate Samples
Find a location near the sample site at least 50 meters away that is of the same habitat as the sample site. Label sample bottles for both preservation on ice and with acid. Take grab samples at this replicate location following the "Inorganic and Bacteria Sample Collection" instructions. Take replicate samples for parameters using the YSI probe in-situ (OCC, 2014).

1.8.4 Preparation of Spiked Samples
Follow the specific instructions from the laboratory facility for spiked samples.

2.0 QA/QC SECTION

2.1 Training
All sampling personnel are required to be trained in all operating procedures and will be monitored in practicing such procedures before solo field operation.

2.2 Maintenance
N/A

2.3 QC Procedures
Ensure that all equipment used for sampling is properly calibrated prior to sampling.

YSI ProDSS MULTI-PARAMETER METER for DO, CONDUCTIVITY, SPECIFIC CONDUCTANCE, TEMPERATURE MEASUREMENT, TURBIDITY, and CHLOROPHYLL-A

Probe Protocols can be found in Appendix C of this document.

1.0 Procedural Section

1.1 Scope and Application
The YSI ProDSS Multi-Parameter Meter has four ports for water quality sensors. The sensors for this WQMP are conductivity, DO, turbidity, and chlorophyll-a. The meter measures the following parameters: Conductivity (uS/cm), Specific Conductance (uS/cm), Salinity (psu), Total Dissolved Solids (mg/L), Temperature (°F), Resistivity (ohms-cm), Turbidity (FNU), Dissolved Oxygen (% Sat, mg/L), Chlorophyll (RFU, ug/L), Phycocyanin (RFU, ug/L), Pressure (psi a), Depth (m), and Vertical Position (m).

1.2 Summary of Method
Probe samples should be taken in a section of the stream that is representative of the entire stream. Samples should not be taken below a riffle or in a hydrologically modified section to provide an accurate reading. The probe should be submerged fully for sampling and should be submerged as close to the middle of the water column as possible in the middle of the stream from bank to bank. When the probe is submerged, wait for the sample values to steady before taking a sample. Some parameters take time to reach their final measurement, such as DO. Make sure these values are not still changing and reach their final measurement before saving the data. Please see Appendix B of this project document for probe protocols.

1.3 Cautions
Ensure that the probe does not drag along the bottom of the creek or that the cord does not get tangled. These precautionary measures will prevent damage to the probe.

1.4 Interference
Make sure that sampling individual stands downstream of the probe when taking sample collections. This ensures that sediment stirred from wading does not interfere with the sample collection.

1.5 Personnel Qualification
Field operators are required to be trained in the proper method of sample collection from an experienced supervising field agent. Practice sessions will be monitored before the agent takes samples without supervision. All agents must familiarize themselves with proper sampling techniques prior to training.

1.6 Apparatus & Materials
YSI Probe

1.7 Instrument/Method Calibration
Follow the User Manual for calibration instructions.

1.8 Troubleshooting
Contact YSI for any troubleshooting questions or refer to the User Manual.

1.9 Computer Hardware & Software
Download the YSI KorDSS Software to a computer for downloading data following a collection event.

1.10 Data Management & Records Management
All sample data should be stored on a reliable computer and back-up hard drive or flash drive.

2.0 QA/QC SECTION

2.1 Training
All sampling personnel are required to be trained in all operating procedures and will be monitored in practicing such procedures before solo field operation.

2.2 Maintenance
Maintenance of the YSI Probe should be performed following the YSI ProDSS User Manual.

2.3 Quality Assurance Measures
Taking routine duplicate samples with the probe will ensure that contamination or user interference does not influence data.

MACROINVERTEBRATE COLLECTION, SUBSAMPLING, AND PICKING

AND FISH COLLECTION

The OCC collection procedures for both Macroinvertebrates and Fish can be found in Appendix B of this project document. These sampling procedures require that a member of the OCC train all city personnel in sampling technique. In addition, an OCC trained field operator will accompany city of Stillwater personnel for at least the first sampling event.

APPENDIX B: Macroinvertebrate and Fish Collection taken directly from the OCC SOPs

MACROINVERTEBRATE COLLECTION, SUBSAMPLING, AND PICKING8

1.0 PROCEDURAL SECTION

1.1 Scope and Application[19,20]

Most free flowing water bodies with acceptable water quality and habitat conditions support diverse macroinvertebrate communities in which there is a reasonably balanced distribution of species among the total number of individuals present. Macroinvertebrate community responses to environmental perturbations are useful in assessing water quality and habitat impacts. The composition and density of macroinvertebrate communities in flowing water are reasonably stable from year to year. However, seasonal fluctuation associated with life-cycle dynamics of individual species may result in extreme variation at specific sites within any calendar year. Assessing the impact of pollution generally involves comparison of macroinvertebrate communities and their habitats at sites influenced by pollution with those collected from adjacent unaffected sites.

Macroinvertebrate collections, for purposes of stream assessment, are made from the community that requires or prefers flowing (lotic) water. Reasons why this community type is sampled rather than various lentic communities include:

1. The flowing water community is routinely exposed to the average water quality of the stream;
2. The metrics used to analyze the macroinvertebrate community of streams were designed for the flowing water community;
3. The database of pollution tolerance of macroinvertebrates found in Oklahoma is much larger for lotic communities; and
4. The organisms most sensitive to water quality degradation tend to live in flowing water.

Due to these factors, looking at the flowing water community is more suitable for assessing the condition of a stream than looking at the pool community where more tolerant organisms are found, regardless of the stream's water quality.

Lotic communities require a substrate of some type to attach to. The most common substrates of this type include rocky riffles, streamside vegetation/root masses, and woody debris. Where possible, a rocky riffle should be sampled. If a rocky riffle is not present, if the riffle is of dubious quality, or if rocky riffles cannot be found at all streams of a given ecoregion, both of the other two alternate habitats (root masses and woody debris) should be sampled. At present, it appears that the streamside vegetation is superior to woody debris for macroinvertebrates, but until that is definitely established, both should be sampled. The sampling methodology for the three habitat types is included in this SOP.

Macroinvertebrate communities are constantly changing throughout the year as species emerge and new species hatch. Consequently, it is not possible to infer water quality from the invertebrate community of a stream by comparing it to a reference stream community that was collected at a different time of year. The springtime communities are especially unstable, as many of the insects that over-winter as larvae begin to emerge. By summertime, however, the insects that only have one generation per year have mostly emerged, and the insects left are ones that hatch repeatedly throughout the summer. This period of the summer when collections from different streams can be compared to each other is termed the Summer Index Period.

Fall is also a poor time to collect to be used for comparing the water quality of different streams. Many insects lay eggs in the summer, and these do not hatch until the water temperature cools down. As these insects hatch and grow large enough to see, they start appearing in collections. Since they hatch at different times and grow at different rates, collections can be very different if they are sampled at different times in the fall. Wintertime communities, on the other hand, tend to be stable. Very few insects emerge in the wintertime, and Oklahoma streams stay warm enough that the invertebrates in them remain actively growing. The wintertime period in which macroinvertebrate collections from different streams can be compared to each other is called the Winter Index Period.

1.2 Summary of Method

A modified version of EPA Rapid Bioassessment Protocol (RBPs) was adopted for macroinvertebrate collections. As stated above, the collection methods are geared toward assessing communities that require or prefer flowing water. Lotic communities require a substrate of some type to attach to. The most common substrates encountered are rocky riffles, streamside vegetation, and woody debris. All three substrates can be sampled (when available) to provide an accurate representation of the various communities in the stream. A combination of collection techniques is used for each habitat. Organisms collected from these habitats are subsampled and sent to a professional macroinvertebrate taxonomist and enumerated to genus level, when possible.

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[19] Text taken directly or in part from Standard Methods (APHA, AWWA, WPCF, 1995).

[20] Text taken directly or in part from Dan Butler, Senior Biologist, Oklahoma Conservation Commission (2000)

1.2.1 Definitions

- Riffle: Any sudden downward change in the level of the streambed such that the surface of the water becomes disrupted by small waves. A riffle substrate must be composed of gravel, or cobble from 1" to 12" in the longest dimension; substrates of bedrock or tight clay are not considered suitable. If composed of gravel and sand, it must be >50% gravel.
- Streamside Vegetation: Any streamside vegetation which offers fine structure for invertebrates to dwell within or upon that receives suitable flow. Most habitat is located along undercut banks where fine roots of riparian vegetation are hanging in the water.
- Woody Debris: Any dead wood with or without bark located in the stream with suitable current flowing over it.
- Summer Index Period: **June 1** to **September 15.**
- Winter Index Period: **January 1** to **March 15**

1.3 Health and Safety Warnings

- Proper precautions should be taken when handling 100% ethanol.
  - Flammable
  - Intoxicant
  - Eye irritant

1.4 Cautions

- Stream stage must not be greater than 3 cm (~1 inch) above base flow during the collection.
- Collections must be done in flowing water.
- In no case should the Mason jar be filled more than 3/4 full of loose sample.
- There should always be enough room in the jar to have at least 5 cm (~2 inch) of free ethanol over the sample.

1.5 Interference

None

1.6 Personnel Qualification

Field personal must be trained and evaluated on sample collection technique. Sample collection is subject to approval by the QA Officer and/or the Environmental Monitoring Coordinator. Training will be done through dry run exercises in the laboratory and field to familiarize field personnel with procedures and techniques.

1.7 Apparatus & Materials

- Absolute ethanol (200 proof; ~100%)
- Clean quart size Mason jars
- New mason jar lids
- Pencil & indelible marker
- 1 m^2 kick net composed of # 30 nylon mesh
- Handheld dip net composed of #30 size nylon mesh

1.8 Instrument/Method Calibration

Not applicable

1.9 Preparation

Determine if flow conditions are suitable for collection. Samples must be collected in flowing water no greater than 3 cm (~1 inch) above the seasonal base flow. After a high flow event, 5 – 7 days should lapse before a collection is made to allow the benthic organisms to return to the preferred substrate. Furthermore, collection should be delayed for two weeks after a stream has gone from no flow (interrupted, or dry conditions) to base flow conditions.

1.10 Sample Collection

There are three possible habitat types for collection. The methods for each are described below.

1.10.1 Collection of Benthic Macroinvertebrates from Rocky Riffles

- **Suitable Substrate** - A riffle is defined as any sudden downward change in the level of the streambed such that the surface of the water becomes disrupted by small waves. For this collection method the substrate of the riffle must be

composed of gravel, or cobble from 1" to 12" in the longest dimension. Riffles with substrates of bedrock or tight clay are not suitable. If the riffle substrate is composed of only gravel and sand it must contain at least 50% gravel.

- **Where to Sample the Riffle** - Three 1 m^2 areas of the riffle must be sampled. They can be square, rectangular or trapezoidal so long as each area equals 1 m^2 in area. One should be in the fastest part of the riffle where the largest rocks and the smallest amount of interstitial sediment will generally be found. The second should be in the slowest part of the riffle, often near the edge of the stream where the smallest rocks and the greatest amount of interstitial sediment will be found. The third sample should be in an area intermediate between the first two

- **Method of Collecting the Sample** - Support a 1 m^2 kick net composed of a double layer of fiberglass window screen or a net of number 30 mesh in such a way that any organisms dislodged from the substrate will be carried into it by the current. The bottom of the net should be tight against the bottom of the stream and the current must be sufficient to insure that dense organisms such as small mollusks will be carried into the net from the sampling area. There is no definite cutoff for stream velocity in the sampling area, but if possible, riffles with average velocities of 1 foot/second or greater are preferred and should be chosen if possible.

  By kicking the substrate, vigorously agitate the substrate of a 1 m^2 area of the bed of the riffle immediately upstream of the net until all rocks and sediment to a depth of at least five inches have been thoroughly disturbed. Organisms living between and upon the rocks will have been dislodged and carried into the net by the current. Any rocks too large to kick should be brushed by hand on all surfaces. This can be done using your hands or with the aid of a brush. If a brush is used, you must be very careful to clean it after each site to prevent contamination of the next sample with invertebrates from the previous site. Continue agitation and brushing until it can be seen that the area being sampled is producing no new detritus, organisms, or fine sediment.

  At this point, rinse leaves, sticks and other large debris caught in the net in the current in a manner such that organisms on them are carried into the net. When the volume of the sample is reduced so that three 1 m^2 samples will loosely fill a 1-quart mason jar three fourths (3/4) full or less, remove all of the material from the net and place it in the Mason jar. In no case should the Mason jar be filled more than 3/4 full of loose sample. Add 100% ethanol to the jar until the sample is covered and there is free ethanol on top of the sample. There should always be enough room in the jar to have at least 5 cm (2 inches) of free ethanol over the sample.

  Label the sample appropriately following the instructions presented in section 1.11 Sample Handling & Preservation.

1.10.2 Collection of Macroinvertebrates from Streamside Vegetation

- **Suitable Substrate** - Any streamside vegetation in current that offers fine structure for invertebrates to dwell within or upon is suitable. The vegetation being sampled must be in the current so that it offers suitable habitat for organisms which collect drifting particles or which need flowing water for other reasons. This habitat will often be found along the undercut banks of runs and bends where the fine roots of grasses, sedges, and trees, such as willow and sycamore, hang in the water.

- **Method of Collecting the Sample** - This type of sample should be collected with a dip net made of #30 size mesh material. The net should be placed around or immediately downstream of the vegetation being sampled. The organisms can be dislodged from the roots either by vigorously shaking the net around the roots or by shaking the roots by hand while the roots are inside the net.

- **Where and How Long to Sample** - Sampling should continue for **3 minutes** of actual root shaking. Do not count the time that elapses between sampling areas. Be careful to only sample roots in current. Usually, only one or two sides of a given root mass are in current. Be careful not to sample the backside of a root mass that is in still water.

At this point, rinse leaves, sticks and other large debris caught in the net so that organisms are not lost. When the volume of the sample is reduced so that it will loosely fill a 1-quart mason jar three fourths (3/4) full or less, remove all of the material from the net and place it in the Mason jar. In no case should the Mason jar be filled more than 3/4 full of loose sample. Add 100% ethanol to the jar until the sample is covered and there is free ethanol on top of the sample. There should always be enough room in the jar to have at least 5 cm (2 inches) of free ethanol over the sample. Label the sample appropriately following the instructions presented in section 1.11 Sample Handling & Preservation.

1.10.3 Collection of Macroinvertebrates from Woody Debris

- **Suitable Substrate** - Any dead wood with or without bark in the stream is suitable as long as it is in current fast enough to offer suitable habitat for organisms which collect drifting particles or which need flowing water for other reasons. The final sample should consist of organisms collected from an even mixture of wood of all sizes and in all stages of decay.

- **Method of Collecting the Sample** - This type of sample should be collected with a dip net made of #30 size mesh material. The net should be placed around or immediately downstream of the debris being sampled. The organisms can be dislodged from the debris either by vigorously shaking the net around the woody debris or by shaking the debris by hand while the debris is inside the net. Large logs that are too big to shake should be brushed or rubbed vigorously by hand while the net is held immediately downstream.

- **Where and How Long to Sample** - Sample for total of **5 minutes** counting only the time that debris is actually being agitated. Include as many types of debris in the sample as possible. These types often include wood that is very rotten and spongy with or without bark, wood that is fairly solid which has loose and rotten bark, wood that is solid with firmly attached bark and any combination of these states. They should range in size from 1/4" to about 8" in diameter.

After sampling, rinse leaves, sticks and other large debris caught in the net so that organisms are not lost. When the volume of the sample is reduced so that it will loosely fill a 1-quart mason jar three fourths (3/4) full or less, remove all of the material from the net and place it in the Mason jar. In no case should the Mason jar be filled more than 3/4 full of loose sample. Add 100% ethanol to the jar until the sample is covered and there is free ethanol on top of the sample. There should always be enough room in the jar to have at least 5 cm (2 inches) of free ethanol over the sample.

Label the sample appropriately following the instructions presented in section 1.11 Sample Handling & Preservation.

1.11 Sample Handling & Preservation

1. **Pack the Mason Jar Properly.** In no case should the Mason jar be filled more than 3/4 full of loose sample. Add 100% ethanol to the jar until the sample is covered and there is free ethanol on top of the sample. There should always be enough room in the jar to have at least 5 cm (2 inches) of free ethanol over the sample.
2. **Label the Sample.** The Mason jar should be labeled on the lid using a fine tip permanent ink marker (Sharpie) as described below. In addition, a small sheet of paper (approximately 2" x 2") should be filled out with the same information written in pencil and placed in the jar.

Jar Lid & Sample Insert

Site Date
Stream Name
Waterbody ID #
Site Time
Legal Description
County
Type of sample (riffle, woody, vegetation)
Sampler's Initials

3. **Complete the Chain of Custody Form (COC).** Follow the instructions in the **Chain of Custody and Sample Labeling SOP**. A new COC should be completed for each collection episode. Each substrate collection (riffle, woody, vegetation) should occupy a separate line on the COC. There should be only one box of samples per COC, i.e., one box, one COC.
4. **Transfer samples to the Macroinvertebrate Sample Custodian.** Correctly labeled macroinvertebrate samples, along with a Chain of Custody form (COC), should be transferred to the Macroinvertebrate Sample Custodian for subsampling. The box should be conspicuously labeled with COC number. Once the samples have been received and the COC signed, the field sampler should make a photocopy of the COC for their records.

1.12 Sample Preparation and Analysis

In some instances it may be necessary to drain the liquid from the sample and add fresh 100% ethanol. This is necessary when the sample contains a large amount of algae or other material with high water content or material that will rapidly become rancid. This will help preserve the morphological integrity of the invertebrates and greatly aid in taxonomic identification.

1.12.1 Subsampling and Picking of Macroinvertebrates from field Collected Samples

The waterbody assessment procedure utilized by OCC requires that a random sample of macroinvertebrates be collected, identified and enumerated, from the portion of the waterbody being assessed. In order to make this test cost effective it is not possible to identify more than about 150 organisms from each site. This procedure describes the procedure used to subsample a field-collected sample, which may contain 200-10,000 organisms.

1. **Obtain the Field Sample to be Subsampled.** The field collected macroinvertebrate samples and the original COC will be transferred to the individual(s) designated to complete the macroinvertebrate subsampling and picking. At this time a copy of the COC (signed by the subsampling designee) will be sent to the Data Manager.

   The sample will come from the field in 1-quart mason jars preserved with 100% ethanol. Mason jar lids have a sealing compound that is not particularly resilient. Care must be taken so that the lids are not damaged when they are opened or resealed. If a lid is damaged it must be replaced with a new one. Keep a fresh supply of lids handy in case this happens. If you use a new lid, label it exactly the same as the one that was originally used.

   Samples will be subsampled/picked in the order assigned on the Chain of Custody form.

2. **Decant Ethanol.** Without shaking or disturbing the contents, pour the liquid from the sample through a sieve made of #30 or finer screen. Save the ethanol to preserve the unused portion of the sample.

3. **Rinse Sample.** At this point, any silt, clay or fine sand in the sample should be GENTLY rinsed out of the sample. Be careful not to break off any of the delicate appendages that are used for identification of the animals. The sample will be easier to process if any large pieces of leaf, bark, stones, etc., are discarded. **Any material to be discarded must first be carefully rinsed within the sieve.**

4. **Prepare Sample for Picking.** Spread the sample out in a rectangular tray/pan that is divided into 28 sections of equal area. The size and shape of the divisions are not important so long as they are all equal in size. A pan with a white background may facilitate the collection since there will be a contrast between the organisms and the pan.

   A clear glass pan or baking dish can be effectively used by creating a grid system on the bottom of the pan using a permanent marker. Each square should be numbered (1-28), and a sheet of white paper can be glued or taped over the outside bottom of the pan. If very many samples will be subsampled it will be worth your time to construct a divider for the tray similar in construction to an ice cube tray divider. This will not only demarcate the subsampling squares, but it will also prevent animals from drifting from one square to another during subsampling.

5. **Remove Large Pieces of Detritus and Sediment.** Large leaves and big pieces of wood and bark should be removed. Be VERY CAREFUL to pick all macroinvertebrates off of them before discarding. At this stage, any debris removed must be viewed through a magnifying lens to ensure the removal of all small invertebrates. At this point, the material remaining in the dish should consist of a mixture of sand, fine gravel, small organic detritus, pieces of leaves < 1-2 cm wide, fine roots, algae and macroinvertebrates.

6. **Spread the Sample Out.** All detritus and sediment should be as uniformly distributed over the bottom of the dish as possible.

7. **Visually Estimate Invertebrate Density of the Sample.** Determine if the sample must be subdivided. The decision will be based on three requirements: (1) individual squares MUST have AT LEAST 3 animals in them (providing the entire sample has at least 100 animals total), (2) you MUST pick AT LEAST 5 squares, and (3) each square can have absolutely NO MORE THAN 25 animals. Simulid (blackfly) larvae are not to be counted as individuals for this purpose. That is, the density estimate should be independent of any blackfly larvae present. The goal is to have roughly 10 TO 20 ANIMALS PER SQUARE. This is a compromise between the statistical ideal of very few organisms per square and ease of subsampling where the entire sample is picked from one square. If you estimate that there are less than 200 animals in the entire sample, you should process the entire sample.

The purpose of this estimate is to make the sample statistically valid. Fewer than 80 animals does not provide a good representation of the population to draw conclusions from, and more than 130 animals biases the sample making it appear that that stream has more taxa than it really does. A total of 100 invertebrates is the absolute minimum number of individual invertebrates to pick from a sample, except when a sample contains fewer than 100. Although 80 is the minimum number of individuals for statistical analysis, it is imperative that a cushion is incorporated to allow for the potential difference in the sub-samplers count and the final count by the taxonomist. Often invertebrates are tossed out by the taxonomist due to an inability to identify individuals caused by missing or damaged body parts or other various reasons.

8. **Subdivide the Sample.** If each square is estimated to have more than 50 animals, it is important to grossly subdivide the sample prior to picking. For instance, divide the sample in half or quarters depending on the animal density. Ideally, 10 to 20 animals per square is desirable.

   For dividing in fourths, cut the sample into top and bottom halves and then into right and left halves. After dividing the sample, the four portions should appear as equal as possible in terms of the amount of detritus and sediment present. Choose one quarter by random means. For instance, flip a coin to select either the top half or the bottom half, and then flipping the coin again to select either the right or left side of the first half selected. If the sample is not too dense, that is the selected quarter has a density of 10 to 20 animals per square, then no additional subdivision is necessary. If the number of invertebrates is still too dense, divide the remaining portion in half and select one half by flipping the coin again. Continue dividing the sample until the density appears to fall in the correct range.

9. **Return the Unselected Portion(s) to the Mason Jar.** Return the unselected portion(s) of the sample to the original Mason jar and add the reserved alcohol. Be very careful to remove the entire portion of unselected subsample. A proportionately high density of small macroinvertebrates can remain hidden in the sediment and detritus.

10. **Fill the Tray About 1 to 2 cm Deep With Water.** Add enough tap water to fill the tray to a depth of 1–2 cm (~0.5 to 0.75 inches) or the depth necessary to cover all sample material (debris and invertebrates). The water aids in the subsampling process. The organisms and individual pieces of detritus do not clump together as when they are dry. If the water is run into the tray very slowly, the remaining leaves and large pieces of detritus can be rinsed and discarded.

11. **Distribute the Sample Evenly.** Make sure that all materials in the tray are evenly distributed, especially the gravel and leaves. This is most easily accomplished by gently homogenizing the sample mixture by hand in the tray and distributing the mixture evenly over the entire pan. If a divider is to be used, place it in the tray now. Once the sample has been distributed, do not move the pan. Jostling can cause the organisms to move outside of their designated square. This could lead to a sampling bias.

12. **Fill Out Macroinvertebrate Picking Data Sheet.**
    Complete the Macroinvertebrate Picking Data Sheet (see SOP Appendix: Data Sheets) as described below:

    SITE / SUBSAMPLING INFORMATION
    - **Picking Date.** The date of subsampling and picking should be recorded in MM/DD/YY format.
    - **Site Name.** The name of the site as it is written on the sample jar.
    - **WBID #.** The waterbody identification number on the sample jar
    - **Picker.** The name of the person subsampling / picking.
    - **Site Date.** The date the sample was collected.
    - **COC #.**
    - **Lab Log #.**
    - **Site Time.** Record the site time in military format. The "site time" is when initial activities began at the site.
    - **Sample Type.** Type of sample "Woody", "Vegetation", or "Riffle"
    - **Sample Description.** Exclusive of invertebrates, estimate the composition of the sample according to the following list: silt and clay, sand, fine gravel (<2mm), coarse gravel (>2mm), woody debris (twigs, bark, roots, etc.), whole leaves, rotted pieces of leaves, filamentous algae, and unidentifiable organic material. Record the percentage of each type.
    - **Proportion Picked.** Record the fraction of the sample that was placed into the tray for sampling –e.g. 1/2, 1/4, or 1/8 of the original Mason jar sample. This is important because the density calculations depend on this number.

- **Square # / # Organisms.** List the number of the square that was picked on the lab notebook along with the number of organisms that were picked from that square.
- **Total Number of Organisms.** Record the sum total of organisms from all squares picked. This number is not used in the calculation of the IBI scores, but provides an estimate of the total number of organisms.

INFORMATION TO INCLUDE	SAMPLE NOTEBOOK PAGE
Subsampling Date	11/18/99
Site Date	07/16/99
Stream Name	Griever Creek
Site Time	13:30
Legal Description & County	E 9 T22N R15W, Major County
WBID #	OK620920-01-0130g
COC #	COC# 1772
Sampling Type	Riffle kick
Sample Description	40%fine gravel; 10% film. algae; 30% well-rotted leaves; 10% whole leaves; 5% woody debris; 5% coarse gravel
Amount of Sample Picked	¼ of the original sample was prepared for picking
Squares Picked	Square # 1, 12, 3, 23, 28, 10
# of animals found in each square	# picked 15, 27, 10, 18, 8, 24
Subsampler's name	John Hassell

13. **Randomly Select Squares.** Using some method to generate a series of random numbers (random number generator or number table), select at least 6 squares. The random number generators found on most pocket calculators or the Excel spreadsheet function will give a series of three digit numbers—usually >0 but <1. For OCC purpose, use only the last two numbers. Starting with the first random number generated, record all numbers between 01 and 28 until there are 6. These numbers represent the numbered squares to pick. Picking must follow the order in which the number were generated.

14. **Confine the Organisms to the Selected Square.** Place rectangles or squares constructed of clear plastic or other material that are the same or greater height as the water in each square. This will keep the organisms from drifting out of the squares during the picking process. If a large piece of detritus (leaves, roots, algae masses) crosses the boundary of two or more squares, it may be sliced along the edge of the square so it is contained with the boundary of the selected square.

    If there are any organisms that cross square boundaries, do not cut them. Place them in the square in which their head is already lying.

15. **Pick <u>All</u> the Invertebrates Out of the First Square Selected.** Locate and collect all the organisms in the selected square. Keep track of the number of non-blackfly organisms picked. Place the organisms picked in a scintillation vial that is filled up to the neck with 70-100% ethanol. If any large organisms (that are too big to fit in the vial with the other organisms) are picked such as crayfish or hellgrammites, place them in a separate vial. If there is some question if something is an organism, place it in the vial but <u>DO NOT COUNT</u> it as part of the total. Place five (5) blackflies and five (5) scuds in the vial but <u>DO NOT COUNT</u> them as part of the total. Pickers should be trained to identify blackfly larval forms—if the picker cannot identify blackflies, they should not be subsampling without further training. Note the general abundance of blackflies and scuds in the comments.

    When all of the organisms are picked out of the square, record the number of non-blackflies/scuds that were picked from that square under the number of that square.

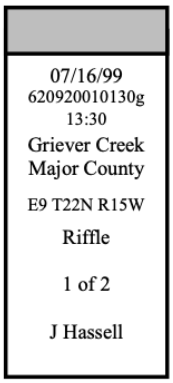
16. **Continue to Pick Squares Until 100 Organisms Have Been Collected.** Using the random number list, continue to select squares until 100 (non-blackfly) animals have been collected. Once a square has been started, all of the animals must be collected from that square. A subsample will typically have 100-130 organisms / vial. If there are more than

130, the sample was not properly subdivided. If the sample has more than 150 organisms in it, it must be mixed back in with the rest of the "unpicked sample" and re-picked. If the finished invertebrate sample has less than 100 organisms in it and there is unpicked sample available, add all of the picked sample (debris and invertebrates) back to the unpicked sample portion and begin the process again with the full sample. The ONLY time that it is acceptable to have a sample with less than 100 organisms is when the entire mason jar of material has been picked. All other samples containing less than 100 invertebrates will be rejected as data with bad QA/QC.

17. **Label the Vial(s).** Using a pencil and a fine point permanent ink marker, label the vial. The top and side of the vial should both be labeled.

    The vial should be labeled IN PENCIL on waterproof paper taped to the vial after labeling with the following information:
    - Stream name
    - WBID #
    - Site date
    - Site time
    - Legal location and County
    - Type of sample (riffle, woody, vegetation)
    - Number of vials for this sample (e.g. 1 of x, where x = total number vials for one site (Mason jar))

    The cap should be labeled IN PERMANENT INK with the following information:
    - Stream name
    - WBID #
    - Site date
    - Time
    - Type of collection
    - Number of vials for this sample



Scintillation Vial

Scintillation Vial Cap

Figure 1: Example label for bug picking sample.

18. **Place Clear Tape Over the Label.** To protect the pencil-written label from wear, place clear tape (scotch tape) over the writing.

19. **Transfer Picked Samples to the Taxonomist**. Once macroinvertebrate samples have been picked and placed in properly labeled scintillation vials, the vials and the original COC will be transferred to the laboratory for taxonomic identification. At this point, a copy of the COC should be forwarded to the Data Manager with the signature of the taxonomist. The original COC will be returned to the Data Manager by the taxonomist or laboratory custodian.

20. **Archive Remaining Sample.** The remainder of the field collected macroinvertebrate samples (un-picked) should be delivered to Nathan Carter or designee for archive purposes. The COC number should be conspicuously labeled on the end of the box.

1.13 Troubleshooting
Consult with the Environmental Monitoring Coordinator

1.14 Data Acquisition, Calculation & Data Reduction
Not applicable

1.15 Computer Hardware & Software
Not applicable

1.16 Data Management & Records Management

1.16.1 Field Notation
A duplicate sample should be collected for every 10 sampling sites and noted on the **Sampling Episode Sheet** (see **SOP Appendix: Data Sheets**). All measurements and observations made at each site should be recorded on the **Site Collection Sheet** (see **SOP Appendix: Data Sheets**). Data should be recorded following procedures outlined in the **Procedure for Completing Field Data Sheets SOP**.

1.16.2 Habitat Form
Regardless of the habitat sampled, a **Macroinvertebrate Habitat Assessment Sheet** (see **SOP Appendix: Data Sheets**) must be filled out at each collection site.

The following bullets describe how to fill out the Macroinvertebrate Habitat Sheet:

DATA SHEET HEADER INFORMATION:

- SITE NAME: Record the stream name from the USGS 7-1/2' map name. If a county map, soil map, or other map has a different name, the USGS 7-1/2' map takes precedence. If a stream is unnamed on the USGS map, but named on another map, use that name, but write the name of the map in parentheses beside the stream name.
- WBID #: Record the Water Body Identification number.
- LEAD INVESTIGATOR: Record the name of the person responsible for data custody and reporting
- DATE: Record the site date in MM/DD/YR format
- TIME: Record the site time in military format. The "site time" is when initial activities began at the site. The site time should be the same on all forms associated with this site.

The form is broken into three columns, one for each habitat type (riffle, streamside vegetation and woody debris). Fill out the appropriate information for each habitat type collected. If one or two of the three sample types are not collected, write "not collected" above the habitat type.

RIFFLE

- % of SAMPLE COLLECTED Usually all of the sample collected will fit in a one quart mason jar (3/4 full). If the sample will not fit, even after removing leaves, rocks, and sticks, without overfilling the jar, mix the sample until all the components (algae, leaves, twigs, rocks, sand, etc.) appear to be uniformly mixed and discard enough of it so that the remainder will fit without over packing the jar. Write down the % of sample you estimate that you have placed in the jar

- UNIT of EFFORT: Refers to the area of riffle sampled. Three 1 m^2 samples should be collected. Record the area sampled.

- EMBEDDEDNESS: This quantifies the amount of silt, clay and sand that has been **DEPOSITED IN RIFFLES.** If there is no fine material surrounding the cobble and gravel of riffles, and there is at least some free space under the rocks, that is 0 percent embedded. If the free space under the rocks is filled but the sides are untouched, count that as 5 percent embedded. As the level of fines rises up the cobble sides, estimate the percentage of the total height of the cobbles that is covered. This is the embeddedness estimate. You can often see this line quite distinctly if you lift the rocks out of the water.

- CPOM in SAMPLE: "Coarse Particulate Organic Matter" Refers to the % of sample composed of partially or well-rotted plant material not counting the substrate being sampled. This should mostly be composed of leaf material. Do not count freshly fallen leaves that have not started to rot. Circle the appropriate number.
  1. Absent 0%
  2. Sparse > 0% but < 5%
  3. Moderate 5% to 25%
  4. Abundant > 25%

- SUBSTRATE TYPE & %: This is an approximate classification of the riffle substrate <u>where the collection is being made</u>. Estimate the proportion each type comprises of the entire substrate. The total of all substrate components should add up to 100%.
  1. Silt & Clay Refers to loose particles < 0.05 mm.
  2. Sand Refers to particles 0.1 to 2 mm is size.
  3. Gravel Refers to particles 2 to 50 mm is size.
  4. Cobble Refers to particles 50 to 250 mm is size.
  5. Boulder Refers to particles >250 mm is size.
  6. Bedrock Refers to rock that is attached to the earth's crust. If a rock can be moved by any means, it is not bedrock.
  7. Hard Pan Clay Refers to a smooth (relatively) surface of clayey material, firm to hard that is moderately resistant to erosion, and provides stable habitat.

- SUBSTRATE ROUGHNESS: Refers to the roughness of the rocks in the riffle. If you can easily assign the riffle to one of these categories by a visual estimate of the roughness no scraping is necessary. If you are not sure, pick up a typical rock and scrape it with a pocketknife. Circle the appropriate number.
  1. Low — >75% of the visible periphyton is removed when scraped with a pocketknife or spatula.
  2. Moderate — 25 to 75% of the visible periphyton is removed when scraped with a pocketknife or spatula.
  3. High — <25% of the visible periphyton is removed when scraped with a pocketknife or spatula.

- VELOCITY TYPICAL MAX: This is an estimate of the average velocity of the habitat sampled in the fastest part of the stream. In a riffle this would be the thalweg. This velocity can be estimated using a floating object and a watch. Circle the appropriate number.
  1. Low — (0.2-0.5 FPS; 0.061-0.15 MPS) FTS = feet/second; MPS = meters/second
  2. Moderate — (0.5-1 FPS; 0.152-0.305 MPS)
  3. High — (>1 FPS; 0.305 MPS)

- NON-FILAMENTOUS ALGAE: Refers to the areal percent of the substrate sampled which is covered with algae that lacks a stringy appearance. Circle the appropriate number.
  1. Sparse — When rocks, bedrock, limbs, trash, etc., are free of attached algae or have only a thin film of greenish or brownish algae that cannot be measured by holding a ruler perpendicular to the surface of the submerged object.
  2. Moderate — When the submerged surfaces have a slight fuzzy or blanketed appearance. The thickness of the attached algae does not exceed 5mm.
  3. Abundant — Submerged surfaces have a definite fuzzy or blanketed (covered with gelatinous mat) appearance. The thickness of attached growth exceeds 5mm.

- FILAMENTOUS ALGAE: Refers to the areal percent of the substrate sampled which is covered with algae that is hair-like or stringy in appearance. Circle the appropriate number.
  1. Absent — 0%
  2. Sparse — > 0% but < 5%
  3. Moderate — 5% to 25%
  4. Abundant — > 25%

- AQUATIC MOSS: Refers to the areal percent of the substrate sampled which is covered with aquatic moss. Circle the appropriate number.
  1. Absent — 0%
  2. Sparse — > 0% but < 5%
  3. Moderate — 5% to 25%
  4. Abundant — > 25%

STREAMSIDE VEGETATION

- % of SAMPLE COLLECTED Usually all of the sample collected will fit in a one quart mason jar. If the sample will not fit, even after removing leaves and sticks, without overfilling the jar, mix the sample until all the components (algae, leaves, twigs, rocks, sand, etc.) appear to be uniformly mixed and discard enough of it so that the remainder will fit without over packing the jar. Write down the % of the total sample that is placed in the jar

- UNIT of EFFORT: Refers to the amount of time the vegetation was agitated. The collection should proceed for **3** minutes. Record the actual time of collection in minutes.

- CPOM in SAMPLE: "Coarse Particulate Organic Matter" Refers to the % of sample composed of partially or well-rotted plant material not counting the substrate being sampled. This should mostly be composed of leaf material. Do not count freshly fallen leaves that have not started to rot. Circle the appropriate number.
  1. Absent — 0%

2. Sparse > 0% but < 5%
3. Moderate 5% to 25%
4. Abundant > 25%

- PRESENCE: Refers to the amount of suitable streamside vegetation habitat present in the stream. Circle the appropriate number.
  1. Occasional Indicates that you must walk more than 50 meters to get a good 3-minute sample.
  2. Common Indicates that you must walk 10 to 50 meters to get your sample.
  3. Abundant Indicates that a good sample can be collected in less than 10 meters of stream.

- TYPE: Refers to the type of streamside vegetation sampled. Circle <u>all</u> that makes up at least ¼ of the total habitat sampled.
  1. Grass-like Leaves Leaves of aquatic or semi aquatic grasses & sedges which have been hanging in the water long enough to develop a periphyton and/or slime coat.
  2. Fine Roots Root masses where most of the roots are <2 mm in diameter.
  3. Coarse Roots Root masses where most of the roots are >2 mm but <6 mm in diameter.
  4. *Ludwigia* Stems Stream macrophyte—not suitable habitat.

- VELOCITY TYPICAL MAX: This is an estimate of the average velocity of the habitat sampled in the fastest part of the stream. For streamside vegetation it would be on the outside (streamside) edge of the root mass. This velocity can be estimated using a floating object and a watch.
  1. Low 0.2 to 0.5 ft/sec
  2. Medium 0.5 to 1.0 ft/sec
  3. High >1.0 ft/sec.

- NON-FILAMENTOUS ALGAE: Refers to the areal percent of the substrate sampled which is covered with algae that are not stringy in appearance. Circle the appropriate number.
  1. Sparse When rocks, bedrock, limbs, trash, etc., are free of attached algae or have only a thin film of greenish or brownish algae that cannot be measured by holding a ruler perpendicular to the surface of the submerged object.
  2. Moderate When the submerged surfaces have a slight fuzzy or blanketed appearance. The thickness of the attached algae does not exceed 5mm.
  3. Abundant Submerged surfaces have a definite fuzzy or blanketed (covered with gelatinous mat) appearance. The thickness of attached growth exceeds 5mm.

- FILAMENTOUS ALGAE: Refers to the areal, percent of the substrate sampled which is covered with algae that is hair-like or stringy in appearance. Circle the appropriate number.
  1. Absent 0%
  2. Sparse > 0% but < 5%
  3. Moderate 5% to 25%
  4. Abundant > 25%

WOODY DEBRIS

- % of SAMPLE COLLECTED Usually all of the sample collected will fit in a one quart mason jar. If the sample will not fit, even after removing leaves and sticks, without overfilling the jar, mix the sample until all the components (algae, leaves, twigs, rocks, sand, etc.) appear to be uniformly mixed and discard enough of it so that the remainder will fit without over packing the jar. Write down the % of sample you estimate that you have placed in the jar

- UNIT of EFFORT: Refers to the amount of time the vegetation was agitated. The collection should proceed for **5** minutes. Record the actual time of collection in minutes.

- CPOM in SAMPLE: "Coarse Particulate Organic Matter" Refers to the % of sample composed of partially or well-rotted plant material not counting the substrate being sampled.

This should mostly be composed of leaf material. Do not count freshly fallen leaves that have not started to rot. Circle the appropriate number.

1. Absent 0%
2. Sparse > 0% but < 5%
3. Moderate 5% to 25%
4. Abundant > 25%

- PRESENCE: Refers to the amount of suitable woody debris habitat present in the stream. Circle the appropriate number.
  1. Occasional Indicates that you must walk more than 50 meters to get a good 3-minute sample.
  2. Common Indicates that you must walk 10 to 50 meters to get your sample.
  3. Abundant Indicates that a good sample can be collected in less than 10 meters of stream.

- SIZE: Refers to the average diameter of the woody debris sampled. Check all lines where that size class makes up at least 1/4 of the habitat sampled.
  1. Small 0.6 to 2.0 cm
  2. Medium 2.0 to 7.5 cm
  3. Large >7.5 cm

- STATE OF DECAY: Refers to the state of decay of the woody debris sampled. Circle all that apply where debris of this type makes up at least ¼ of the habitat sampled. All of these categories may or may not have bark on them. These categories are determined by firmly pressing your thumbnail into the wood (not bark) of the debris sampled perpendicular to the grain. The depth of the indentation, if any that remains when your thumbnail is removed is measured to determine the state of decay.
  1. Low Indentation is 0 to 0.5 mm deep
  2. Moderate Indentation is 0.5 to 2 mm deep
  3. High Indentation is > 2 mm deep

- VELOCITY TYPICAL MAX: This is an estimate of the average velocity of the habitat sampled in the fastest part of the stream. For woody debris, it would be the average velocity of the water passing over the sides of the wood. This velocity can be estimated using a floating object and a watch.
  1. Low 0.2 to 0.5 ft/sec
  2. Medium 0.5 to 1.0 ft/sec
  3. High >1.0 ft/sec.

- NON-FILAMENTOUS ALGAE: Refers to the areal percent of the substrate sampled which is covered with algae that are not stringy in appearance. Circle the appropriate number.
  1. Sparse When rocks, bedrock, limbs, trash, etc., are free of attached algae or have only a thin film of greenish or brownish algae that cannot be measured by holding a ruler perpendicular to the surface of the submerged object.
  2. Moderate When the submerged surfaces have a slight fuzzy or blanketed appearance. The thickness of the attached algae does not exceed 5mm.
  3. Abundant Submerged surfaces have a definite fuzzy or blanketed (covered with gelatinous mat) appearance. The thickness of attached growth exceeds 5mm.

- FILAMENTOUS ALGAE: Refers to the areal percent of the substrate sampled which is covered with algae that is hair-like or stringy in appearance. Circle the appropriate number.
  1. Absent 0%
  2. Sparse > 0% but < 5%
  3. Moderate 5% to 25%
  4. Abundant > 25%

COMMENTS Record any useful information that provides insight to the sample collection process, conditions, or miscellaneous information.

1.16.3 Chain of Custody Procedure

Collection of inorganic sample requires the use of a Chain of Custody form (COC). The handling of COC should follow the procedures described in the **Chain of Custody and Sample Labeling SOP.** The manifest is routed as follows:

1. Macroinvertebrate samples are collected in the field and the COC is completed and signed by the field personnel involved with collection.
2. Samples are submitted to the Macroinvertebrate Sample Custodian. That person signs the COC and forwards a copy to Data Manager or logs the information on the web page.
3. Samples are assigned to subsampling/picking personnel for processing. They must sign the COC.
4. Processed samples are sent to the taxonomist for identification. The taxonomist must sign the COC. The person who sends the samples to the taxonomist, forwards a copy of the COC to the Data Manager.
5. After identification, the taxonomic identification sheets will be forwarded with the signed COC to the Data Manager. The laboratory will include the laboratory tracking or log numbers used to reference the identification sheet.

2.0 QA/QC SECTION

2.1 Training

Training of field personnel will be done through dry run exercises in the laboratory to familiarize them with instrument operation, calibration and maintenance. All operators are required to become familiar with the SOP documents. Prior to solo sample collection, subsampling or picking, personnel are evaluated in for proper use of equipment and sample collection protocol. Annual audits are performed on sample collectors following procedures outlined in the **Quality Management Plan**.

2.2 Maintenance

Not applicable

2.3 QC Procedures

A set of field QA samples will be collected for every sampling episode or one set per 10 sampling sites (10%). The QA samples will include at a minimum a Field Replicate. Spatial replicates should be obtained by implementing the aforementioned sampling procedures upstream of the sampling site being careful to sample with equal effort a similar composition of habitat to the original sampling site. If required by the QAPP, Field Splits will be collected. Subsampling and picking QA/QC is the responsibility of the contracted facility. The OCC will evaluate QA/QC procedures through blind checks and spot inspections.

3.0 REFERENCES

APHA, AWWA, and WPCF (1995) Standard Methods for the Examination of Water and Wastewater, 19th edition, eds. L.S. Clesceri, A.E. Greenberg, and R.R. Trussell, American Public Health Association, Washington, D.C.

Butler, D., (1999) Personal Communication, Senior Biologist, Oklahoma Conservation Commission, Oklahoma City, OK.

FISH COLLECTION

1.0 PROCEDURAL SECTION

1.1 Scope and Application[7],[8]

Fish assemblage monitoring is an integral component of the Oklahoma Conservation Commission's Water Quality program. Assessment of the fish assemblage measures the structure and function of the ichthyofaunal community to evaluate the integrity of a stream. Sampling occurs during the summer period, as defined below, with care to avoid collection in waters with sensitive species or species of concern earlier than June 1.

1.2 Summary of Method

The collection of fish follows a modified version of the EPA Rapid Bioassessment Protocol V (EPA, 1989) supplemented by other documents. Specific techniques for, and relative advantages of seining and electrofishing vary considerably according to stream type and conductivity. The specifics are discussed in detail in Fisheries Techniques (edited by L.A. Nielsen and D.L. Johnson and published by the American Fisheries Society 1983).

The collection of fish involves the use of two collection methods, seining and electroshocking. The combination of methods was selected in order to produce a representative fish collection. Variations of habitat, type of fish, and water chemistry dictate the use of different collection techniques. In general, each stream is sampled for a distance of 400 m. Both techniques are used at each site when practical, in attempt to reduce gear bias or selectivity to the extent possible. Occasionally site conditions will limit the effectiveness of one form of sampling or the other, to the point of rendering them impractical or unsafe. These judgments will be determined by the crew chief or the crew member most experienced with the site.

Seining can be broadly defined as the use of a net, manually pulled through the water, in attempt to encircle and capture fish. Seine height is dictated by water depth, and length is determined by width of the water being sampled. If possible, the seine should be 15-25% longer than the width of the waterbody being sampled and about 25% higher than the depth of the water. The seine is hauled with the current because fish tend to orient towards the current.

Electrofishing can be defined as the use of electrical current passed through the water, in a controlled manner, in an attempt to momentarily stun fish, rendering them easy to capture. Electrofishing can be accomplished using three different methods and equipment types: (1) backpack shocker, (2) tote barge, or (3) boat shocker. The backpack shocker is the primary means of electrofishing and consists of a trailing stainless steel cable electrode and ring electrode mounted on the end of a fiberglass pole. The ring should be substituted with a diamond shaped array when sampling in waters of higher conductivity (>1000 µS/cm). The shocking team consists of at least two people. One carries and operates the shocker while the other(s) net stunned fish. The shocker is most useful where a seine cannot be used effectively in areas such as brush piles, root wads, and cobble substrates. The forward electrode is gradually passed back and forth as the team walks downstream. As fish are stunned, they usually roll over and become more visible, allowing the netters to see and capture them.

In waters of high conductivity (> 1000 µS/cm) the effectiveness of electroshocking declines. Under these conditions, electrofishing may be limited to targeted shocking in shallow habitat that is not possible to seine. At high conductivity levels it is up to the discretion of the crew leader, whether electrofishing is effective. The backpack shocker is rated for use up to 2150 µS/cm, and should not be used at higher conductivity levels. The backpack system's level of power and size of field is designed for and works well in narrow (<15 meter wide) streams with short wadeable pools and numerous riffles. Stream channels that are wider, with longer pools and fewer and shorter riffles that still meet wadeable criteria, require a larger electrical field and more power than can be supplied by a backpack shocker. Additionally, if 30-40% percent of the reach to be sampled is greater than the depth of the shortest crew member's elbow, an alternate shocking technique should be used.

In deep or wide streams where backpack shocking is impractical, the OCC employs the use of a tote barge. The barge system is powered by a 5500 watt (10horsepower) generator and is housed in a 4x5'x12"plastic tub. The barge allows for reasonable mobility of the large (>100lb) generator. The system includes a handheld anode pole, identical in scope and design to that described for the backpack system, but connected by a 50' cable. The cathode consists of an electrically connected aluminum grid system mounted to the bottom of the barge (below waterline). A pull rope is attached to the front of the barge to allow the team to pull the barge across short riffles and other obstacles when necessary. The increased power of the larger generator not only enables the crew to improve its efficiency in open water but is also helpful in mitigating the effects of increased conductivity

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[7] Text taken directly or in part from "Rapid Bioassessment Protocols for Use in Wadeable Stream and Rivers, 2nd Edition", US EPA 841-B-99-002 July 1999

[8] Text taken directly or in part from Dan Butler, Senior Biologist, Oklahoma Conservation Commission (2000)

when encountered. Due to safety concerns the barge team should include an additional crew member responsible for overseeing the safety of the crew and monitoring the control box (and the emergency shut-off switch)

Occasionally, fish surveys must be completed on stream reaches that offer very limited wading potential. When presented with this obstacle, we employ the use of a boat mounted electrofishing system. The system includes a 3250 watt generator, a single bow mounted anode pole/array, and a control. The aluminum boat hull, which houses the electrofishing system, acts as the cathode. The boat is equipped with a raised deck and safety rail. The primary netter(s) position themselves on the deck. The current is activated when one of the netters stands on a pressure activated switch, completing the circuit. The boat is propelled by a small outboard motor. The navigator maneuvers the boat in a way that positions the anode in or near habitat. When riffles or other areas of shallow water are encountered, the system comes equipped with a junction box, which allows the user to quickly convert from a bow mounted anode to a handheld anode attached by a 50' cable. This enables a crew, wearing chest waders, to use the boat in a very similar manner as the tote barge. Electroshocking from a boat requires at least three crew members.

In general, all fish are placed in 10% formalin immediately after capture. However, if larger fish (> 100 g) can be positively identified in the field, they are returned to the water in a location where recapture is unlikely. All large fish released are photographed. A representative photograph is taken when large numbers of one fish species is collected and released. Collected organisms are identified to species by an experienced taxonomist.

1.2.1 Definitions

- Summer Collection Period: **May 15*** to **October 31**

  **waters containing sensitive species or species of concern will not be sampled before June 1 to avoid disruption of spawning*

1.3 Health and Safety Warnings

- Primary responsibility for safety while electroshocking rests with the team leader.
- All crew members should receive training in First Aid and CPR. Electro-fishing units have a high voltage output and may deliver dangerous electrical shock. Electric shock can cause heart fibrillations and/or death.
- While electrofishing, avoid contact with water unless sufficiently insulated against electric shock. Use chest waders with non-slip soles and water-tight rubber gloves that cover to the elbow. If they become wet inside, stop fishing until thoroughly dry.
- Avoid contact with anode at all times. <u>At no time while electrofishing should a crewmember reach into the water for any reason.</u>
- The electrofishing equipment provided is equipped with a 45 degree tilt switch which interrupts the current. Do not make any modifications to the electrofishing unit, which would make it impossible to turn off the electricity.
- General safety guidelines should be observed. If waders or gloves develop leaks, leave the water immediately. Avoid operating electrofishing equipment near people, pets or livestock. Discontinue any activity in streams during thunderstorms or heavy rain. Rest if crew becomes fatigued.
- Decision to use electrofishing equipment will depend on size of site, flow, conductivity and turbidity. If the specific conductivity is below 10 µS or > 1000µs; if the flow is too high; if the site is too deep; if the water is too turbid to assure safe footing or locate stunned fish, the crew may consider using the seine only or determine that site cannot be sampled. <u>This is a safety decision.</u>
- Formalin is a carcinogen and can also cause permanent damage to mucous membranes and eyes. Care must be taken when placing fish in formalin so that the fish does not flop around and splash formalin onto people near the jar. Proper precautions should be taken when handling formalin.
  - Protective gloves and eyewear should be worn
  - Avoid inhalation of vapors
- <u>FAILURE TO OBSERVE SAFETY PROCEDURES WILL RESULT IN DISCIPLINARY ACTIONS INCLUDING PROBATION AND DISMISSAL.</u>

1.4 Cautions

- Do not collect fish without the permission of the Monitoring Coordinator, who will have obtained the appropriate permits.
- Do not sample in waters containing sensitive species or species of concern before June 1 to avoid disruption of spawning.

1.5 Interference

- Seine effectiveness is limited by physical obstructions including rocks, sticks, logs, thick vegetation, or anything that would impede the progress of the net. And can also include extreme (>1.5 m) depth and water velocity (>3fps).
- Backpack shocker effectiveness declines above conductivity levels greater than 1,000 µS/cm, and should not be used above 2,150 µS/cm

- Tote barge and boat shockers allow for sampling at higher conductivity levels, but should not be used in waters with higher conductivity than the manufacturer's specifications..

1.6 Personnel Qualification

Field personal must be trained and evaluated on sample collection techniques. Sample collection is subject to approval by the QA Officer and/or Monitoring Coordinator. Training will be done through dry run exercises in the field to familiarize field personnel with procedures and techniques.

1.7 Apparatus & Materials

Clothing

Rubber Gloves	as many pairs as the shocking crew consists of
Waders	as many pairs as the shocking crew consists of, although everyone is responsible for their own waders
Goggles	for use in mixing formalin

Documentation

Field data sheets	**Sampling Episode Sheet, Site Collection Sheet, Flow Meter Sheet** and **Fish Collection**
Waterproof paper	for labels inside jar
Pencils	labeling
Sharpie pen	for labeling jar
Extra white paper	used for a background for fish pictures
Clipboard	
Camera	
Tape measure	to record lengths of released fish if desired

Chemicals

10% buffered formalin

Shocker

Smith Root LR24 backpack shocker system OR
Smith and Root VVP tote barge system mounted on 4'X5'X12' plastic tub housing OR
Midwest Lake Electrofishing system mounted on a 4'X12' aluminum boat.

Nets

4 x 10, 6 x 10, 4 x 20, and 6 x 20 seines and any other seines that are preferred by the crew leader. All seines should be ¼ inch mesh.
Dip nets to collect shocked fish

Containers

Wide mouth 1-gallon jars, at least 4 per site
1 or 2 liter graduated cylinder for mixing 10% formalin (37% formaldehyde)
Whirl-Paks for putting special fish in

Instruments

DO meter
pH meter
Conductivity meter
Turbidity meter
Alkalinity test kit
Flow meter

1.8 Instrument/Method Calibration

Refer to the appropriate SOP and/or owner's manual.

1.9 Preparation

- A representative stream reach is selected and measured such that primary physical features are included in the reach (riffles, runs, and pools)
- The reach should be located away from the influences of major tributaries and bridge/road crossings.
- In general, each stream is sampled for a distance of 400 m.

Seining

- Seine height is dictated by water depth, and length is determined by width of the water being sampled. If possible, the seine should be 15-25% longer than the width of the waterbody being sampled and about 25% higher than the depth of the water. The amount of obstructions in the stream will often preclude the use of longer seines however. When this situation occurs, the crew leader will decide on the most effective combination of seines. OCC utilizes 4 and 6 foot seines in 10, 20, and 30-foot lengths. This will allow the center of the net to form a bag behind the operators where the fish are more likely to stay in the net. The seine is hauled with the current because fish tend to orient towards the current.

Electrofishing

- The shocker is most useful where a seine cannot be used effectively in areas such as brush piles, rootwads, and cobble substrates.
- The choice of electrofishing method and equipment will depend on the stream to be sampled.
    1. For narrow (<15 meter wide) streams with short wadeable pools and numerous riffles, the backpack shocker is most appropriate. The shocker consists of a trailing stainless steel cable electrode and either a ring or diamond electrode mounted on the end of a fiberglass pole. In waters of extremely low conductivity (<40 uS) the ring should be used. In waters of high conductivity (>1000 uS) only the diamond should be used. In very deep water where the ring seems to be ineffective the diamond electrode may offer better results. The shocking team consists of at least two people. One carries and operates the shocker while the other(s) net stunned fish.
    2. In deep or wide streams where backpack shocking is impractical, the OCC employs the use of a tote barge. The system includes a handheld anode pole, identical in scope and design to that described for the backpack system, but connected by a 50' cable. The cathode consists of an electrically connected aluminum grid system mounted to the bottom of the barge (below waterline). A pull rope is attached to the front of the barge to allow the team to pull the barge across short riffles and other obstacles when necessary. Tote barge electrofishing requires at least three crew members.
- In stream reaches that offer very limited wading potential, the boat mounted electrofishing system will be used. The system includes a single bow mounted anode pole/array. The aluminum boat hull, which houses the electrofishing system, acts as the cathode. When riffles or other areas of shallow water are encountered, the system comes equipped with a junction box, which allows the user to quickly convert from a bow mounted anode to a handheld anode attached by a 50' cable. This enables a crew, wearing chest waders, to use the boat in a very similar manner as the tote barge. Boat electrofishing requires at least three crew members.In waters of high conductivity (>1000 µS/cm) electroshocking effectiveness declines, due to the highly conductive nature of the water. Under these conditions, it is up to the discretion of the crew leader if electrofishing is suitable. Electrofishing will not be completed at conductivity levels greater than the manufacturer's recommendation for the equipment .

1.10 Sample Collection

Seining

1. The seine should be manually pulled through the water. Since fish tend to orient towards the current, the direction of the seine haul should generally be with (in the same direction of) the current.
2. The lead line should be kept on the bottom, and in front of the float line.
3. If there are many obstructions on the bottom, the lead line will become caught or bounce and most fish will escape underneath the bottom of the net. If this happens use a smaller net that allows you to avoid obstructions or go to electroshocking.
4. The brailes of the net should be used to disturb the area under any undercut banks or beds of macrophytes near the edge, in order to scare fish hiding under cover out towards the middle of the net.
5. Under ideal conditions the net should be pulled through the water in the manner described above for about 10 meters and dragged out of the water on a gradually sloping pre-selected beach. The person pulling the seine on the side of the stream opposite the beach should swing ahead of the other person so that the seine is pulled out on the beach stretched over the same distance it was stretched in the stream.
6. If the stream does not have gradually sloping banks, the dip method should be used. This method consists of sweeping around and through the area to be sampled, keeping a wide bag and moving the lead line as much under the undercut bank

as possible. Use the brailes to probe repeatedly as far as possible into the undercut area working towards each other until the brailes overlap. The seine should then be swiftly stretched and lifted vertically from the water. An alternative method of retrieving fish under these conditions is to slowly turn the brailes to wind the net up once they have overlapped to form an enclosure. This may entangle the fish with the net and allow them to be lifted out of the water with the rolled up net.

Shocking

1. Before operating or assisting with the shocker, READ AND UNDERSTAND THE MANUALS for the generator and the shocker. Starting procedures, safety procedures and troubleshooting are well documented in these manuals and are not spelled out in this text. The manuals can be obtained from the equipment file in the main office.
2. Collection begins at a shallow riffle or other physical barrier at the downstream limit of the reach, and terminates at a similar barrier at the upstream end of the reach.
3. In general, fish collection procedures commence at the downstream barrier and proceeds in an upstream direction; however, this is up to the discretion of the Crew Leader.
4. A minimum of two people is required for electrofishing.
5. The forward electrode should be gradually passed back and forth over the stream width, including brush piles and root wads. As fish are stunned, they will usually roll over and become more visible, allowing the netter(s) to see and capture them.
6. In very dense brush or root cover, fish often sense the presence of the team before they are close enough to be stunned and then retreat so deeply into cover that it is impossible to net them when they are stunned. It is often better in situations such as these to insert the electrode into the brush before it is turned on, give the fish a minute or so to get used to the new situation and then turn the current on. Many fish will be much closer to the edge of brush pile when they are stunned in this manner.

1.11 Sample Handling & Preservation

1. Fish collected by seining and electroshocking should be kept in separate jars and labeled as to what method was used to capture them. This will make the methods independent if desired for analysis.
2. Label each jug. Using a permanent marker, write the date, WBID #, collection time, stream name, number of jars composing one sample, county, legal location, and crew leader's name on the lid and side of the jug. In general all fish should be placed in 10% formalin immediately after capture. There are a few exceptions made for larger fish (>100 gms or 0.25 lbs), which can be positively identified in the field.
    a. If all team members agree on the identification of such a fish, it can be returned to the water far enough away that recapture is unlikely.
    b. All large fish released must be documented on the **Fish Collection Sheet**. This includes fish such as gars, all types of carpsuckers, black bass, any white bass in water where yellow bass or striped/white hybrids may be found, all buffalo, all redhorse, and any other unusual fish. Please note**,** the golden and black redhorse cannot be told apart without counting lateral line scales and pelvic rays. Unless this information is recorded on the **Fish Collection Sheet**, the fish must be brought in for identification, or recorded as *Moxostoma* sp. Similar notes must be taken when releasing other fish that can be difficult to tell apart in the field such as the river and shorthead redhorses or any of the buffalos.
    c. All large fish released must be photographed. It is important to take photos and label them so that they will be identifiable 5 to 7 years from now. Be sure to follow the Photodocumentation SOP. The photos are data, and should be labeled as to the ID of the fish in the picture, the date, WBID #, site time, stream name, county, and legal location of the site. One copy should be kept in the Crew Leader's files, and one should be forwarded to the Data Manager. In addition, note the photos on the **Fish Collection Sheet**.
3. When preserving fish much larger than 0.3 to 5 kg (0.5 to 10 lbs), the fish should be sliced open along the lower rib in order to allow the formalin to penetrate the body cavity fast enough to prevent decay. A slit through the ribs is preferred to a belly slit to facilitate counting belly scales in the lab.
4. Formalin is a carcinogen and can also cause permanent damage to mucous membranes and eyes. Care must be taken when placing fish in formalin so that the fish does not flop around and splash formalin onto people near the jar. The fish should be put into the jar with the lid tilted open away from the operator so that the lid shields the face and body of the operator. Flood any skin exposed to formalin with plenty of water as soon as possible. If it gets in your eyes, flood the eyes with water immediately and go to the doctor immediately after that.
5. Fill out a **Chain of Custody Form**.
6. The Crew Leader is responsible for transferring the samples to the Fish Sample Custodian.

1.12 Sample Preparation and Analysis

Not applicable

1.13 Troubleshooting

Consult owners' manuals and/or the Environmental Monitoring Coordinator

1.14 Data Acquisition, Calculation & Data Reduction

Not applicable

1.15 Computer Hardware & Software

Not applicable

1.16 Data Management & Records Management

1.16.1 Field Notation

All measurements and observations made at each site should be recorded on the **Site Collection Sheet** (see **SOP Appendix: Data Sheets**); include all physical and chemical information including DO for runs, riffles, pool top, and pool bottom—when available. Data should be recorded following procedures outlined in the **Procedure for Completing Field Data Sheets SOP**. A **Flow Meter Data Sheet** (see **SOP Appendix: Data Sheets**) should also be filled out; see the Flow Measurement for Wadeable Streams SOP. It is mandatory to follow the procedures outlined in the Photodocumentation SOP. Please note photos on the appropriate field sheets.

1.16.2 Fish Collection Sheet:

All observations should be recorded on the **Fish Collection Sheet** (see **SOP Appendix: Data Sheets**).

The following bullets will describe how the **Fish Collection Sheet** should be completed.

DATA SHEET HEADER INFORMATION:

- SITE NAME: Record the stream name from the USGS 7-1/2' map name. If a county map, soil map, or other map has a different name, the USGS 7-1/2' map takes precedence. If a stream is unnamed on the USGS map, but named on another map, use that name, but write the name of the map in parentheses beside the stream name.
- WBID #: Record the Water Body Identification number.
- LEAD INVESTIGATOR: Record the name of the person responsible for data custody and reporting
- DATE: Record the date in MM/DD/YY format.
- TIME: Record the site time in military format. The "site time" is when initial activities began at the site and should be the same on all forms associated with the site.

COLLECTION INFORMATION:

For each collection method used, fill in the appropriate specifications. For the backpack, tote barge and boat-mounted shockers, indicate:

- SHOCKING TIME Record the amount of time spent shocking in seconds
- VOLT/AMPS Record the voltage and amperage on the shocker
- PULSES/SECOND Record the pulses per second setting on the shocker (measure of wave frequency)
- %DUTY CYCLE % of on time; product of pulse width and frequency (the actual time the current is being delivered)
- REACH LENGTH Length of stream used in the fish collection

For the boat-mounted shocker only, also indicate:

- LOW RANGE or HIGH RANGE
- HANDHELD or UMBRELLA ARRAY PROBE.

If a seine is used, indicate:

- SEINING TIME Record the amount of time spent seining in minutes
- SEINE TYPE/SIZE Record the size and type of seines used

FISH IDENTIFIED & RELEASED:

- SPECIES Record the genus and species of the fish released or the common name if the species can be definitely identified later based on that common name
- COUNT Record the number of individual organisms released
  - SHOCK Number released during the shocking effort

○ SEINE	Number released during the seining effort
• COMMENTS	Record any information that was used to help in the identification process
• PHOTO ID #	Record the identification number that corresponds to OCCWQ photo tracking system. It is mandatory to follow the procedure outlined in the Photo-documentation SOP.

1.16.2 Habitat Form

At all sites where fish are collected, a stream habitat evaluation must be performed. It does not have to be done on the same day as the fish are collected, but should be done before major floods change the habitat. Refer to the Habitat Assessment SOP.

1.16.3 Chain of Custody Procedure

Collection of fish requires the use of a Chain of Custody form (COC). . The handling of COC should follow the procedures described in the **Chain of Custody and Sample Labeling SOP**. The manifest is routed as follows:

1. Fish samples are collected in the field and the COC is completed and signed by the field personnel involved with collection.
2. Samples are submitted to the Fish Data Custodian and the person receiving the samples signs the COC.
3. Processed samples are sent to the taxonomist for identification. The taxonomist must sign the COC.
4. After identification, taxonomic identification sheets will be forwarded with a copy of the signed COC to the Data Manager. The COC form returned from the laboratory will include the laboratory tracking or log number(s) used to reference the identification sheet.

2.0 QA/QC SECTION

2.1 Training

Training of field personnel will be done through dry run exercises in the laboratory and field to familiarize them with instrument operation, use, calibration and maintenance. All samplers should read Fisheries Techniques (edited by L.A. Nielsen and D.L. Johnson and published by the American Fisheries Society 1983) prior to collecting fish. All operators are required to become familiar with the SOP documents. Prior to solo sample collection, field personnel are evaluated in a field setting for proper use of equipment and sample collection protocol. Annual field audits are performed on sample collectors following procedures outlined in the **Quality Management Plan**.

2.2 Maintenance

- Maintain the shocking equipment per the owner's manual instructions
- Seines should be stored dry and tangle-free

2.3 QC Procedures

At least one temporal replicate sample should be collected per fish crew leader.. Replicate samples should be completed at the same reach as the original collection. When practicable, replicate samples should be taken during the same season and hydrologic period (i.e., not across major seasonal change such that rainfall and temperature are significantly different between the samples). Replicate sampling within four weeks is preferred.

APPENDIX C: YSI ProDSS Probe Protocols

City of Stillwater
YSI ProDSS Probe Protocols

The city of Stillwater uses a YSI ProDSS Multi-parameter Water Quality Meter. This probe has 4 ports for additional sensors. We have 4 additional sensors:

- Conductivity
- Turbidity
- ODO
- Total Algae

***Be familiar with ALL the ProDSS parameters and their units.**

Before using the probe, you **must** familiarize yourself with the following documents:

- Instrument Overview
- Calibration Guide
- Calibration Solutions

For further training or assistance, you can visit YSI ProDSS University for tutorial videos.
For more detailed operation assistance, read the User Manual.

Using the Probe

1. Calibration

- As researchers, we are expected to collect **quality** data. The probe must be calibrated properly to ensure this. You are responsible for making sure that your data is accurate, therefore you must calibrate the probe before **every sampling event**.

- We calibrate two parameters in the lab before each sampling event:
    1. Conductivity (NOT Specific Conductance)
        - make sure that the calibration value is entered as 1000 µS-cm. This should match the value on the calibration solution bottle.

    2. Turbidity
        - use deionized water (DI water) as the first calibration solution. The calibration value should be 0. (save calibration)
        - use the turbidity solution for the second calibration point (124 FNU). The calibration value should be 124. (save calibration)
        - finish calibration after the second calibration point.

- We calibrate one parameter in the field:
    1. % DO
        - unscrew the calibration cup so that it is loose
        - Perform calibration

- **ALWAYS** keep the protective sleeve on the probe while calibrating.

***** see the Calibration Guide for more detailed instructions on how to calibrate these parameters.

You are responsible for making sure that calibration solution is stocked and in date. When a solution is running low, be sure to order more.

*****Label the bottle with the date you open it and the date it will expire*****
- Conductivity solution expires **1 month** after opening
- Turbidity solution expires **6 months** after opening

2. Operation

- Make sure to log data under your unique user ID and your unique data ID.

- **Do not let the probe drag** on the bottom of streams/lakes. This could damage the probe. As soon as turbidity values increase significantly, pull up the probe to prevent damage.

- Make sure the microUSB port is closed securely, **water can damage the handheld**.

- **ALWAYS** keep the protective sleeve screwed on over the sensors when sampling. (this sleeve should only be unscrewed to add/remove sensors)

- To ensure that you don't lose data, **download data after each sampling event.** You will need the KorDSS Software to download data from the handheld.

3. Storage

- ALWAYS coil the probe cable in a tidy manner.

- Remove any knots or twists in the cable before spooling. **Knots can damage the cable.**

- Make sure that the handheld has a sufficient charge before storing. If the device is below 90% battery, plug-in the handheld to a power source for the next user.

- **Make sure that there is 1cm of water in the calibration cup**. The DO sensor requires a moist environment for storage. Do not overfill— the sensors should not be submerged in water for storage.

4. Troubleshooting

- You are responsible for making sure that equipment is working accurately. If the probe data doesn't look right, or the probe is malfunctioning in any way, stop collecting data. **DO NOT USE THE PROBE FOR SAMPLING UNLESS IT IS WORKING ACCURATELY.**

****You are responsible for finding a solution to any issues that may arise****

Here are some resources for troubleshooting:

1. Read the User Manual
2. Google the problem
3. Use the "chat" feature on the YSI website
4. Call the YSI tech support line

We have a contact for troubleshooting:

Sam: (877)-726-0975 ext. 775

VITA

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Completed the requirements for the Master of Sciences in Environmental Science at Oklahoma State University, Stillwater, Oklahoma in December, 2021.

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