Utah Lake Nutrient Cycling Studies:
Phase I: Annual Report, 2021

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Executive Summary

This report presents the quantitative and statistical data results from Phase I of the Utah Nutrient Cycling Studies project. The research goals for Phase I were to:

1. better understand nutrient cycling in Utah Lake and
2. evaluate the performance of treatments that have the potential to offer holistic and long-term solutions to reduce the intensity, duration, and frequency of harmful algal blooms (HABs) toxins in Utah Lake.

Utah Lake (UL) is a major physical feature in the Utah Valley and a valuable natural resource. UL is a shallow, turbid, slightly saline, eutrophic lake in a semi-arid area. It has a high capacity for degradation and stabilization of pollutants because of its shallow, well-oxygenated, high pH waters. It supports and harbors abundant wildlife as part of a productive ecosystem. The lake offers a wide range of beneficial uses: ecological habitats, water storage, and recreation (e.g., boating, sailing, fishing, and hunting). Over the years, usage of UL water has increased with urbanization in the Salt Lake Valley.

UL is unique in that most nutrient loads or inflows remain in the Lake; current estimates are that over 95% of nutrient inflows remain in the Lake. It is likely that even if anthropogenic inflows were significantly decreased, the lake would remain eutrophic from natural inflows [1].

Current discourse surrounding the Lake ecosystem concerns whether available nutrient concentrations in the water column are directly related to nutrient inflows, are governed by in-lake processes, or are governed by other factors. If nutrient concentrations in the water column are not directly related to nutrient inflows, then controls on nutrient inflows will have little impact on UL water quality. The data we collected in 2021, presented in our final report, will better characterize these processes, and help evaluate such proposed mitigation measures.

The Utah Lake Nutrient Cycling Studies (ULNCS), both Phase I and subsequent phases, supplements the ongoing Utah Lake Water Quality Studies (ULWQS) implemented by the Utah Lake Commission (Commission) and the Utah Department of Environmental Quality, Division of Water Quality (DWQ). The initial Phase I work, completed in close coordination with the Timpanogos Special Service District (TSSD) and reported in this paper, is a scoping study designed to generate data and information to inform the Phase II and Phase III work.

Phase I included the following objectives:

- develop sampling and analysis methods to characterize geochemistry in Utah Lake,
- determine how to place and operate limnocorrals, and
- start to develop baseline data to characterize geochemical processes in Utah Lake.

We used limnocorrals to create large mesocosms that we used to semi-isolate various lake processes and experiments from the surrounding lake. The limnocorrals are 10 m

(30 ft) in diameter with skirts that extend to the lake bottom, where they are sealed to the lakebed sediments by a heavy chain.

In Section 2, our paper outlines the details of corral construction and subsequent redesign following damage that the corrals sustained due to inclement weather on the lake. Together with TSSD, we developed a reinforced limnocorral design that was deployed with success in a preliminary stage during Phase I and will be deployed for use in Phase II during the summer of 2022. Because the corrals sustained damage or lost functionality in other ways during the course of the summer, at times we had to adjust our sampling schedule. Full details concerning our sampling schedule and relevant adjustments are outlined in this report.

In addition to making improvements on the limnocorrals, Phase I work enabled us to learn how best to take samples from the corrals and from the surrounding lake. We performed statistical analyses to understand how well the functional corrals mirrored lake conditions, how many samples should be taken from each corral to make an accurate assessment of each mesocosm, how frequently we needed to calibrate our probes, and other relevant sampling details. Also provided in the report is a full description of our standard operating procedures surrounding each type of sampling trip in our Phase I efforts: Corral Water Sampling, Probe-Only Sampling, and Probe Data Tracks Sampling.

In Section 1.6, we discussed Dr. David Richards' ecological work, performed during Phase I of this study. He will continue to lead these efforts in Phase II. Because of the nature of the data and experiments, those data and results are not reported in our final report but are provided as separate reports.

These include:

- Richards, David C. and Blake Wellard (2022) *Macrophyte Restoration Utah Lake: A Success Story: Technical Memoranda"*, prepared by OreoHelix, LTD, prepared for the Timpanogas Special Service District, April 16, 2022.
- Richards, David C. (2022) *Plankton Biomass, Diets, Production biomass Ratios, And Ecotrophic Efficiency Estimates for Utah Lake Foodweb Model Development: Is It Raining Algae On The Benthos In The Summer?,* prepared by OreoHelix, LTD, prepared for the Wasatch Front Water Quality Council, Salt Lake City, UT, January 2, 2022.
- Richards, David C. (2021) Chlorophyll A Trends In Utah Lake From 1989 To 2019: technical memo, prepared by OreoHelix, LTD, prepared for the Wasatch Front Water Quality Council, Salt Lake City, UT, January 2, 2022.

For all sample analysis activities, we followed the draft Data Quality Objectives (DQOs) for the ULNCS as presented in the request for proposal [2] and TSSD's 2022 Framework for the UL Solutions Program. These draft DQOs follow the United States Environmental Protection Agency's (EPA's) seven-step DQO process. The 2022 framework provides the most recent summary of objectives for this work. We view these DQOs and the 2022 framework as an essential component of the project as they provide the background, purpose, problem statement, conceptual model, and define the framework and requirements for the project. We followed DQOs and procedures from

both the BYU Environmental Analytical Laboratory (EAL) and the TSSD Analytical Laboratory for the sample analysis performed in these laboratories.

We measured pH, dissolved oxygen, temperature, specific conductance, turbidity, phycocyanin, chlorophyll, and depth *in situ* using YSI DSS Pro water quality sondes. This included point samples in the water sample locations, and vertical profiles in each deep corral. Before labor costs, each probe sample cost $67. The number and type of probe measurements we took are detailed in the final report.

We performed our sample analysis in three separate laboratories, as described in our final report. At the TSSD laboratory, we analyzed NH_3, NO_3, NO_2, TP, filtered and unfiltered PO_4^{3-}, TOC, COD, TDS, TSS, and VSS. In the BYU EAL, certified students analyzed aluminum (Al), arsenic (As), boron (B), barium (Ba), calcium (Ca), cadmium (Cd), cobalt (Co), chromium (Cr), copper (Cu), iron (Fe), potassium (K), magnesium (Mg), manganese (Mn), molybdenum (Mo), sodium (Na), nickel (Ni), phosphorus (P), lead (Pb), sulfur (S), selenium (Se), silicon (Si), strontium (Sr), titanium (Ti), vanadium (V), and zinc (Zn) using inductively coupled plasma (ICP) spectroscopy and soluble reactive phosphorus (SRP). Certified students used the BYU Geological Sciences Laboratory (GSL) Dionex ICS-90 for Ion Chromatography (IC) to measure the major anions: fluoride (F^-), chloride (Cl^-), phosphate (PO_4^{3-}), and sulfate (SO_4^{2-}) in the water column. All of our work plans and methods included a Quality Assurance Plan and was reviewed by TSSD and with the ULWQS Science Panel. These sample analyses are detailed in Section 3.2 of the final report. Details concerning the number of samples run for analysis of each constituent are listed in Section 4.3.

We performed several experiments during Phase I to gain a better understanding of conditions in the lake. These experiments are discussed briefly here, but full notes are found in Section 5.

Our serial filtration experiment, detailed in Section 5.2, examined the effectiveness of different filtration methods. We tested three different filter sizes (0.45µm, 0.7µm, and 1.5µm) during our analysis of water samples. Our results led us to using a 0.45µm membrane filter on all filtered samples.

Our ICP analysis of heavy elements is detailed in Section 5.3. We obtained data for 25 elements. Based on our preliminary results, we plan to integrate more rigorous QA/QC methods into our studies during Phase II, potentially including matrix spikes to evaluate filtering activities.

In Section 5.4, we discuss an initial bench-scale experiment that we conducted to evaluate the ability of aluminum to co-precipitate phosphorus and remove it from the water column. We found from our preliminary jar test data that the lake was able to compensate over time for added Al, resulting in no discernable increase in Al concentration after later doses. During Phase II, we will expand upon these jar tests to determine if aluminum salts could effectively sequester phosphorus in UL.

To better understand the amount of isolation produced by the limnocorral mesocosm and the flushing rate of the corrals, we conducted an experiment in which we used rhodamine water tracer dye to coat the surface of the limnocorral-contained water

column. Our results indicated that the actual flushing time in the corrals is about 3-6 days; however, further dilution experiments in later phases will be necessary to get a more accurate estimate of the flushing rate. For now, we can only be sure that complete turnover of water in the corrals occurs in a matter of days, which is an excellent time frame for our purposes. This experiment is fully detailed in Section 5.5.

We have not yet attempted to perform an in-depth analysis of the turbidity or TSS data that we collected, but we have completed a preliminary examination to verify that the data are reasonable. This examination is outlined in Section 5.6. Our preliminary data and analysis indicated that phytoplankton growth, and growth of cyanobacteria in particular, is light limited in UL, and that reduced turbidity results in higher growth. These data are not conclusive; we expect that Phase II data will be more conducive to analysis.

We also examined phosphorus distribution due to its status as a primary nutrient influencing algae growth. Full details on our analysis of total, filtered, and unfiltered reactive phosphorus distributions are found in Section 5.7 of our final report.

In Section 6, we discuss Dr. Carling's work in three Phase I categories: sediment chemistry, pore water chemistry, and geochemical modeling. His methods and results are included in our final report.

As previously stated, the research goals for Phase I were to develop sampling and analysis methods to characterize geochemistry in UL, to determine how to place and operate limnocorrals, and to, if possible, start to develop baseline data. We have accomplished our objectives by developing a sampling schedule and identifying problems with our limnocorrals. Through close coordination with the TSSD, we were also able to obtain preliminary data regarding lake geochemistry that has given us context for our studies and data usage moving forward. Through our partnership with TSSD, we were also able to develop more sophisticated limnocorrals to produce more durable mesocosms that will allow us to gain more consistent data over the course of future project phases. Our Phase I goals met; we are confident in our ability to progress to Phase II.

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Utah Lake Nutrient Cycling Studies: Phase I Annual Report 2021

1. Introduction

1.1. Introduction

This report represents the results from Phase I of the ULNCS project. The research goals for Phase I were to:

1. better understand nutrient cycling in Utah Lake and
2. evaluate the performance of treatments that have the potential to offer holistic and long-term solutions to reduce the intensity, duration, and frequency of harmful algal blooms (HABs) toxins in Utah Lake (UL).

We accomplished this by…

- developing sampling and analysis methods to characterize geochemistry in Utah Lake,
- determining how to place and operate limnocorrals, also referred to as corrals in this report, and
- starting to develop baseline data to characterize geochemical processes in Utah Lake.

The goal of this sampling will be to document nutrient concentrations in the sediment and water column and how they change in response to various parameters within the limnocorrals. The overarching purpose for this investigation is to better understand the geochemical interactions between lake processes and water column.

This report is a description of the data collected during Phase I and describes these data both qualitatively and statistically. It includes a section on issues with and potential improvements to our limnocorral design, as well as some preliminary analysis as examples of the types of information that may be gained from the data collected during Phase I. It is not meant as a detailed report on Phase I data and results, but rather a description of the types of data that were taken and some insight into the data themselves.

Utah Lake (UL) is a major physical feature in the Utah Valley and a valuable natural resource. UL is a shallow, turbid, slightly saline, eutrophic lake in a semi-arid area. It has a high capacity for degradation and stabilization of pollutants because of its shallow, well-oxygenated, high pH waters. It supports and harbors abundant wildlife as part of a productive ecosystem. The lake offers a wide range of beneficial uses: ecological habitats, water storage, and recreation (e.g., boating, sailing, fishing, and hunting). Abundant wildlife and ecological richness are some of its more significant assets [3].

Because of the uniqueness of the UL ecosystem and its history as a valuable resource for Utah County, it is important that the health of the Lake ecosystem is maintained and understood. Our study aims to 1) better understand geochemical interactions between lake processes and the water column (referred to as nutrient cycling) in UL and 2) evaluate the performance of treatments that have the potential to offer holistic and long-

term solutions to reduce the intensity, duration, and frequency of future harmful algal blooms (HABs) in UL.

As noted above, this report does not address these study aims, but instead describes the results of the Phase I exploratory study, which investigated the use of limnocorrals on UL and potential sampling methods and experiment designs. The results of Phase I will inform the study design and methods used during Phase II, and the data from that phase will be used to fulfill the study aims.

1.2. Water Quality and Nutrients

UL is unique in that most nutrient inputs remain in the lake; current estimates are that over 95% of nutrient inflows remain in the Lake. Total nutrient inflows to UL include sources such as streams, overland flow, sediment sources, biological sources (i.e., carp), dust, wastewater treatment plants, and geochemical processes. Taken together, they provide nutrient loadings that are tens of times larger than those that would designate the lake as eutrophic. Current estimates are that even if anthropogenic inflows were significantly decreased, the lake would remain eutrophic from natural inflows [1]. In response to these unique circumstances, our experiments produce necessary background information about the behavior of the lake as well as about the efficacy of potential conservation efforts.

The water quality impacts of nutrient inflows from various sources, including publicly owned treatment facilities, are currently of wide interest [2]. There is considerable debate surrounding the impacts of nutrient loadings on lake water quality. In many freshwater lakes, direct nutrient loads are a primary driver of algal growth, but this may not be the case for UL due to its unique characteristics, including extremely high turbidity levels and high rates of sediment resuspension.

Current discourse surrounding the lake ecosystem concerns whether available nutrient concentrations in the water column are directly related to nutrient inflows, are governed by in-lake processes, or are governed by other factors. If nutrient concentrations in the water column are not directly related to nutrient inflows, then controls on nutrient inflows, such as new restrictions concerning nutrient levels in water treatment plant outflows, will have little impact on UL water quality. The data we collected in 2021 will better characterize these processes and help evaluate such proposed mitigation measures.

1.3. Study Background

Figure 1 Students on the BYU research boat prepare to take samples at the limnocorrals on Utah Lake

In 2021, we started Phase I of the ULNCS, which is designed to conduct a series of experiments to better understand how to characterize UL geochemistry and nutrient cycling processes. The initial Phase I work, reported here, is a scoping study designed to generate data, experimental procedures, and information to inform the Phase II and Phase III work. The ULNCS, both Phase I and subsequent phases, supplements the ongoing Utah Lake Water Quality Studies (ULWQS) implemented by the Utah Lake Commission (Commission) and Utah Department of Environmental Quality, Division of Water Quality (DWQ).

We conducted the Phase I study in close coordination with the Timpanogos Special Service District (TSSD). The study is independent of the ULWQS, but with a goal to complete these studies in parallel to and in collaboration with the ULWQS as a means of investigating and developing solutions to water quality issues in UL.

1.4. Field Sampling

This study is unique in that we used limnocorrals to create large mesocosms that semi-isolated various lake processes and experiments (Figure 1). The limnocorrals are 10 meters (30 feet) in diameter with skirts that extend to the lake bottom, where they are sealed by a heavy chain that connects the skirts to the lake sediments. This creates a water column that is semi-isolated from the surrounding lake. It allows us to perform experiments, such as chemical augmentation to reduce phosphorous, and isolates the corral water column from some of the lake processes such as sediment suspension from wave and wind action.

The Phase I field sampling efforts within and outside of the corrals were completed by students at BYU who were trained by the study Principal Investigators (PIs) (Figure 2). Dr. Williams and Dr. Miller led the BYU team, and each have substantial experience with field sampling and have worked extensively in UL. Dr. David Richards led the ecological studies in this project who is one of the foremost ecologists working on UL issues. Dr. Richards trained and provided guidance to BYU students for biological sampling. Dr. Richards also used several subcontractors and

Figure 2 Lab team members take samples on our skiff near the floating dock.

other groups to assist in biological sampling and analysis. Dr. Richards' research is further detailed in Work Plan and final report [4, 5].

1.5. ULNCS Sampling Plan

We performed all Phase I summer sampling in accordance with field and laboratory checklists that provided detailed procedures including, for example, specific steps on how to correctly collect and store samples, acquire drone images, perform other sampling tasks, and how to calibrate and maintain field equipment and sensors. Using checklists helped ensure quality control of the samples by aiding students in performing tasks consistently and followed SOPs based on DWQ standard operating protocols [6].

A goal of Phase I was to develop a feasible sampling schedule that would meet the need to characterize the lake processes within the constraints of our resources: including students, laboratory capacity, and costs. At the beginning of the summer, we tested several potential sampling schedules. Over time, we developed and refined this schedule to create a consistent sampling and analysis routine. This is discussed in Section 3.1.

Over the course of the summer, many corrals failed due to inclement weather and other issues (details on corral function are provided in Section 0). When all corrals were functioning, we performed sampling trips and collected data as scheduled, weather and equipment permitting. When fewer corrals were functioning, we performed fewer sampling trips, but collected more samples and ran more data tests on the collected samples in the remaining corrals.

Figure 3 presents the sampling timeline for the summer of 2021. Later sections detail the exact procedures marked on this schedule. Periods when the corrals were in place and functional are identified with green bars and periods when the corrals were non-functional due to structural damage are identified with grey bars. Collection dates for each type of sample and measurement are shown as color-coded points along the timeline, and purple bars indicate the dates of extreme weather events. Figure 12 in Section 0.2 presents drone photographs of the condition of the corrals over time. This series of images in combination with the sampling timeline gives good insight into the impacts that inclement weather had on corral integrity.

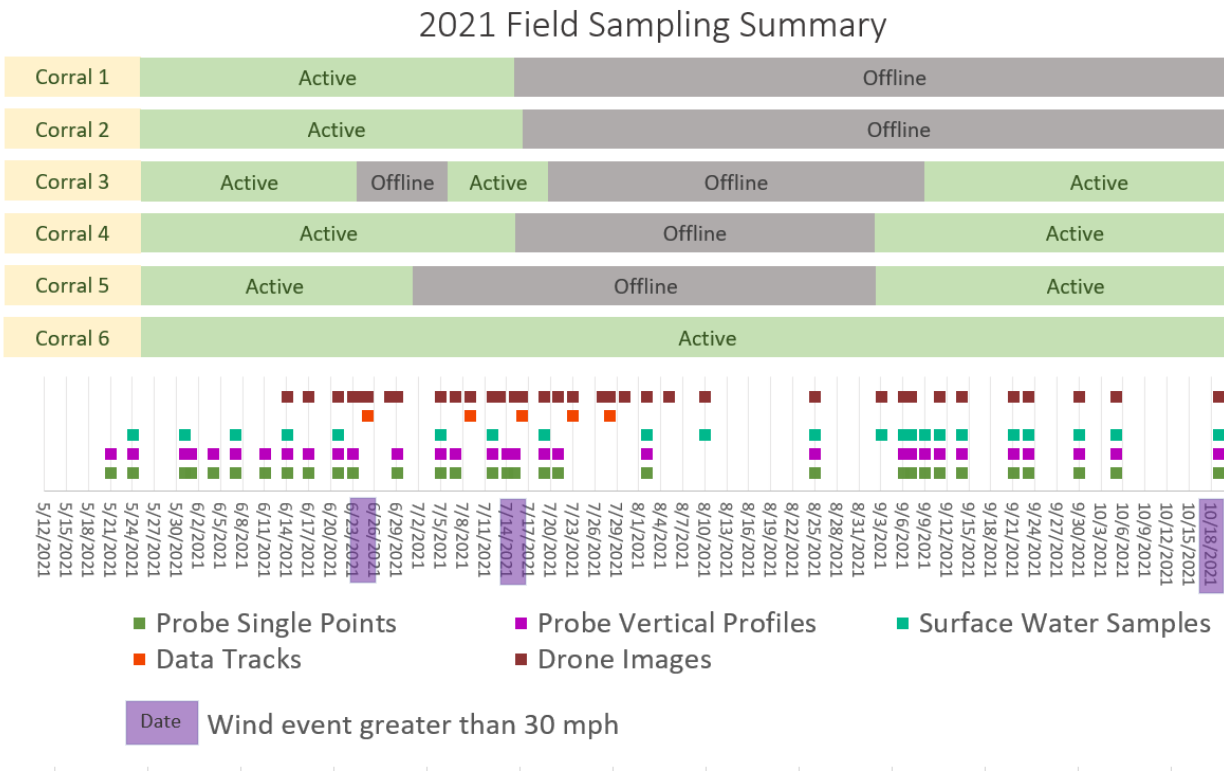

Figure 3 Field sampling schedule for 2021 which includes limnocorral status; corral placed but skirt raised (light green), corral placed, and skirt lowered (green), or corral was not present or was damaged (grey). During the grey periods, the corral was not able to isolate the water column and was not sampled.

To address the structural problems we encountered with the corrals and prevent similar difficulties with future corral function, we made adjustments to our original design prior to field deployment in the Phase II studies in the summer of 2022. TSSD developed structural reinforcement modification and anchoring for the corrals that should withstand the wind and wave action in UL for the duration of the sampling season. This TSSD-developed reinforced limnocorral design will be deployed in 2022. Preliminary versions of this design and anchoring were deployed late in 2021 for evaluation, and the results indicated that the design is effective.

The Phase I effort on limnocorral operation and design was a success in that we determined how to better design and operate the limnocorrals to provide the data required to characterize UL processes. We found that the commercial limnocorral design was not applicable to UL; some of the data sets collected during Phase I were compromised or incomplete as a result of limnocorral failures.

During Phase I, we collected a total of 153,327 probe data points and 126 water samples which we analyzed for over 35 parameters, depending on the sample. We provide detailed information on laboratory analysis and data in Section 4, and complete tables of data points in Appendix A. We were able to perform these analyses at a laboratory cost of ~$67 per sample, not including labor costs. Costs were low because we were able to certify our

students to perform most of the laboratory work in the TSSD, EAL, and Geology certified labs, so most of the costs were associated with laboratory consumables.

Overall, Phase I field data collection and laboratory analysis was a success: we determined a feasible sampling schedule, developed sampling and analysis methods, collected baseline data on UL geochemistry, and determined how to better design and operate the limnocorrals to provide the required sampling and experimentation environment.

1.6. Ecological Sampling and Results

Dr. David Richards led the ecological work performed during Phase I of this study and will continue to lead these efforts in Phase II. Because of the nature of the data and experiments, those data and results are not reported here, but are provided as separate reports:

- Richards, David C. and Blake Wellard (2022) *Macrophyte Restoration Utah Lake: A Success Story: Technical Memoranda"*, prepared by OreoHelix, LTD, prepared for the Timpanogas Special Service District, April 16, 2022.
- Richards, David C. (2022) *Plankton Biomass, Diets, Production biomass Ratios, And Ecotrophic Efficiency Estimates for Utah Lake Foodweb Model Development: Is It Raining Algae On The Benthos In The Summer?,* prepared by OreoHelix, LTD, prepared for the Wasatch Front Water Quality Council, Salt Lake City, UT, January 2, 2022.
- Richards, David C. (2021) Chlorophyll A Trends In Utah Lake From 1989 To 2019: technical memo, prepared by OreoHelix, LTD, prepared for the Wasatch Front Water Quality Council, Salt Lake City, UT, January 2, 2022.

2. Limnocorrals

2.1. Limnocorral Background

Limnocorrals are a type of *in situ* mesocosm which consist of floats that attach to large, flexible, tarp-like material extending from the surface of the water to the lakebed with a semi-tight seal at the bottom (Figure 4). The isolated water column created by the limnocorral simplifies some of the complexities of a natural ecosystem. The isolation allows researchers to better test theories and study lake processes, such as nutrient cycling and complex water chemistry, under controlled conditions.

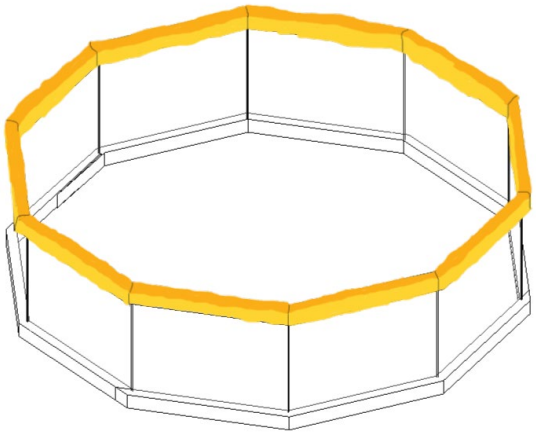

Figure 4 Drawing of the limnocorrals showing the floats (yellow) and the skirts extending down to the lake sediments.

In general, mesocosms are used by researchers for a variety of purposes, including studying dose response relationships, measuring natural variables (such as temperature or salinity), testing hypotheses, and developing theories [7]. As previously mentioned, mesocosms are used to test scientific hypotheses and develop future theories [8, 9]. Advantages of a mesocosm study include the relatively low cost, the speed of data collection, and the ability to replicate results [8, 10]. Trends, processes, and relationships identified in mesocosm experiments can be extrapolated to larger systems [11, 12]; however, quality data can only be obtained if the mesocosm study is appropriately designed in scale and duration [13, 14]. If these requirements cannot be met, data may be harder to extrapolate, and the results might be skewed. To prevent unreliable data, mesocosm studies should be designed using available background knowledge of the system and verified with subsequent whole-ecosystem studies [14].

Figure 5 Deep water corrals were located roughly one mile offshore of TSSD outfall.

2.2. Limnocorral Deployment

In April of 2021, TSSD staff, contractors from Hazelett Marine, and BYU students assembled and anchored six limnocorrals at the Northeast end of UL near the Timpanogos Special Service District outfall (Figure 5). Five of these corrals were anchored in 2-meter-deep (6.5 feet) water 235 meters (770 feet) from shore at the time of placement, and one limnocorral was anchored 60 meters (200 feet) from the shore in 1-meter-deep (3 feet) water. The limnocorrals have a decagonal shape with a diameter of approximately 10 meters (30 feet) (Figure 4).

Initially, the volume of water in the deep-water corrals (when the water was approximately 2.5 meters (8 feet) deep) was approximately 200 cubic meters (~50,000 gallons), and the shallow corral, at a depth of 1 meter, had a volume of about 80 cubic meters (~20,000 gallons). The large volume of water in the limnocorral is crucial for ensuring that processes within the corrals mimic those in the lake and data from the corrals can be extrapolated to the general lake system. This large volume allows the mesocosm to control study parameters while retaining some of the natural complexity of the lake.

As the summer progressed, lake levels decreased, lowering the surface water elevation by about 1 meter (3 feet). Due to this lake level change, the volume of water in the limnocorrals decreased by about half in both the deep-water and shallow-water corrals (Figure 6).

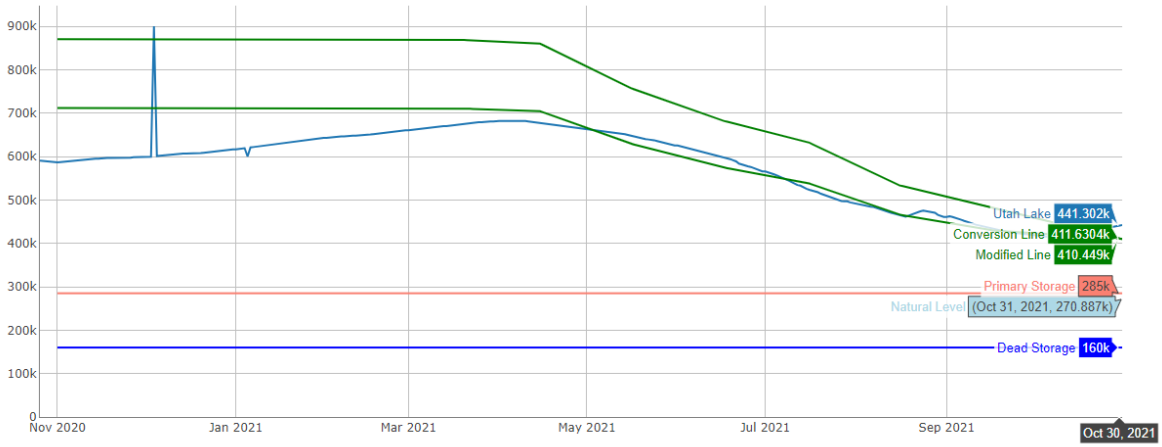

Figure 6 Utah Lake levels during 2021 in feet above sea level [15]. Light blue line indicates the surface of the lake, and clearly shows the significant decline in water levels during the summer.

Despite the decrease in water volume over the summer, we continued to take samples from the corrals. This drop in water level had the most significant impact on the shallow corral, which was in less than 0.5 meters (1.6 feet) of water at the end of the summer, resulting in an isolated mesocosm that did not appear to replicate the environment of the surrounding lake, because the skirts on the corrals were designed for a higher water level. It is possible that upon experiencing the extreme drop in water level, the long skirt on the corral served to isolate the internal water column from the rest of the lake more than anticipated. The deep corrals continued to mimic the lake for parameters such as temperature and nutrient levels, but clearly isolated the water column as indicated by variation in turbidity. Some preliminary analysis of these data and processes are provided in Section 4.

2.3. Limnocorral Construction and Structural Issues

2.3.1. Construction and Placement

Each of the six corrals was constructed using ten six-inch wide yellow floats, a six-foot long flexible woven skirt, a heavy chain roughly the circumference of the corral, ten metal struts, and a center float. The six-inch-wide yellow floats provided a buoyant force to hold the flexible woven skirt to the surface of the water and the long metal chain secured the skirt at the bottom to ensure a tight seal. The center float and metal struts were intended to hold a mesh cover to prevent fish from jumping in or out of the corral. For ease of sampling, we decided to forgo the cover. The reinforced design, which will be used for new corrals in Phase II and subsequent phases, uses 8-inch floats and a reinforced metal brace structure holding the floats together. Some of the corrals from Phase I will be reused with their six-inch floats. This new design eliminates the struts, as they are not necessary without the cover, and were prone to failures.

Figure 7 Limnocorral construction on the shore of Utah Lake.

Figure 8 Towing the limnocorrals from the shore assembly site to the experimental site.

Figure 9 SCUBA diver and others working to anchor the corrals to the experimental site.

We assembled the corrals on the north shore of UL (Figure 7) and then towed them into place with the BYU pontoon boat (Figure 8). A scuba diver from Hazelett Marine then placed anchors into the lakebed and attached the corrals (**Figure 9**).

Figure 10 Corral structural damage after high winds and waves in Utah Lake.

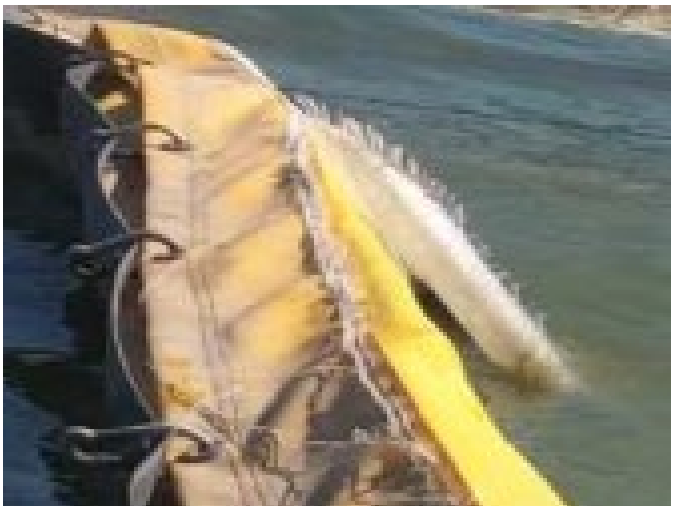

Figure 11 Corral damage after high winds and waves in Utah Lake.

2.3.2. Structural Issues

We anchored the corrals in place by April 29th, 2021, but due to the high winds on UL (the highest reached 51 mph) and large waves, the corrals began to exhibit structural damage in June, disrupting the desired isolated water column. The struts detached and fell from the yellow floats and began to rip the corral skirts (Figure 10). As time passed, the struts ripped bigger and bigger holes in the corral skirts and eventually embedded themselves into the lakebed. When strong winds blew, the corrals could not move with the waves properly due to the embedded struts, and water from the lake entered the corrals. The stitching on the corral skirts also began to unravel, causing the skirts to detach from the floats (Figure 11), which further increased water exchange between the limnocorrals and the lake.

The structural damage to the corrals made it difficult to take samples within the corrals and the lack of an isolated water column disrupted planned experiments.

Originally, we tried to take samples from each quadrant shown in Figure 13, but as the corrals fell apart, the quadrants no longer existed, and we could not take samples this way. Because of corral failure, we stopped sampling in the broken corrals until they could be fixed. The broken corrals meant that the corrals were no longer isolated from the lake and would mimic the nutrient cycling of the lake itself. During this time, we continued sampling activities in the functional corrals.

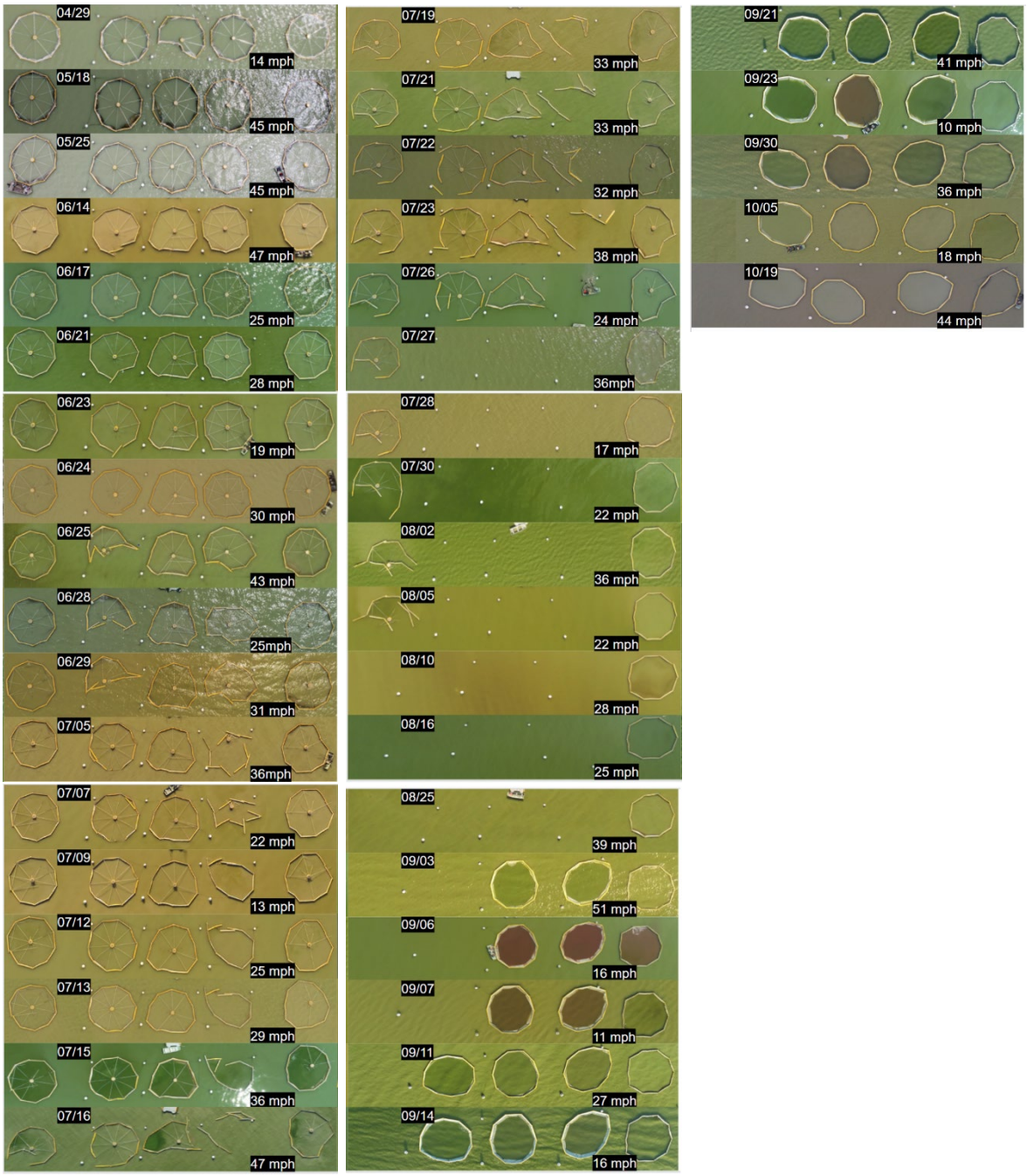

Figure 12 Behavior of limnocorrals over time showing the impact of waves and wind. The text on the images indicates the date the image was taken and the highest wind speed that occurred since the previous image.

TSSD developed a preliminary reinforcement design to help the corrals withstand the force of UL, and with their help we installed reinforced corrals. Figure 12 shows that damaged corrals were removed from the lake in mid-July, reinforced, and partially replaced in September. This new design removed the ten struts and the center float from

the corrals and added reinforcement beams around the corral perimeter as well as a reinforced anchoring system.

To remove the corrals, where possible, we gathered the damaged skirts and tied them to the yellow floats with rope, then towed the damaged corrals to shore and installed structural reinforcements on land. In some cases where the skirts were completely detached from the floats, we removed the skirts from the lake first, then disassembled the corral in the lake and hauled the pieces to shore on the boat. The skirts were later completely replaced with shorter two-meter (six and a half feet) skirts due to the shallower lake conditions and to decrease the amount of billowing in the skirts.

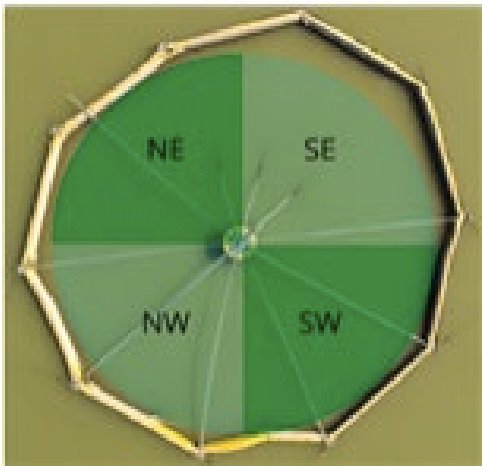

Figure 13 Limnocorral Quadrants

Figure 12 shows the corrals throughout the entirety of the sampling season (April to October 2021). The corrals were removed starting on July 26th and were reinstalled starting the end of August. The new design can be seen from September 3rd to October 19th in Figure 12. The new design ensured the integrity of the corrals until the end of the sampling season in mid-October. The Phase II reinforcement design is an extension of this original effort, with better reinforcement and better integration into the corral design.

2.3.3. Preliminary Limnocorral Results

2.3.3.1. Spatial Variation in the Limnocorrals.

To determine the spatial variability within the corrals, and whether sampling procedures could be streamlined in subsequent years, we took probe samples to determine water characteristics in each quadrant of the corrals (Figure 13). We then performed a statistical analysis of the quadrant sampling data.

We used an ANOVA analysis to compare the distribution of each quadrant and determine if it was statistically different from the samples from another quadrant. We performed the initial ANOVA analysis using a simple Tukey test for chlorophyll. Dr. Dennis Eggett of the BYU Statistics Department subsequently performed a more in-depth statistical analysis using a mixed effects model to estimate the variance in conductivity data between the quadrants in the corrals. Our analysis indicated that there was no statistically significant difference in data obtained from the different quadrants (Table 5).

Since most of the corrals experienced various failures and were removed from service for a significant period, we ran this analysis on Corral 6 data, as it was in service for the longest time and stayed relatively competent with no major failures. We only used data from days where we sampled each quadrant to avoid skewing the results toward any one quadrant. The results from both the preliminary Tukey test and the more advanced mixed effects model showed that the corrals are well mixed laterally and that we do not need to sample all four quadrants. Rather, a single sample represents the corrals well.

The mixed effect model found the largest variance was approximately 5 µS/cm--0.25% of the mean of the conductivity data, which was on the order of 2,000 µS/cm. This low variance is within the water quality sonde's 8-10 µS/cm accuracy range, meaning that the variation among the quadrants is not significant or even technically measurable.

These statistical tests indicated that water within the limnocorrals was well-mixed, and that a single sampling location per corral can be used in subsequent work. We plan to perform a more complete statistical analysis before the field season begins in 2022, but initial Phase II plans are to only take one sample per corral, rather than multiple samples in the different quadrants.

2.3.3.2. Lake Background

We compared the data taken within the limnocorrals to that taken at two nearby background locations within the lake. The background locations were near Buoy C6W and Buoy C2W (Figure 15). The preliminary results indicated that baseline data that described the lake, such as temperature, obtained from corrals two through six were similar to the values measured in the lake background locations. This comparison of corral temperature values with background temperature values used data only from periods when no experiments were active in the limnocorrals. Data describing processes that we expect the corrals to isolate, such as turbidity, were different between the lake and the corrals. Additional analysis of this data is provided in Section 5.6.

Table 1 Least squares mean test results for the different quadrants.

Effect	Quad	Est.	Std Error	DF	t	Pr > |t|
Quadrant	NE	2048.54	750.25	21	2.73	0.0125
Quadrant	NW	2051.17	750.25	21	2.73	0.0124
Quadrant	SE	2051.57	750.25	21	2.73	0.0124
Quadrant	SW	2046.21	750.25	21	2.73	0.0126

This analysis supports the idea that mesocosms created in the corrals are representative of the lake and can be used to study and understand lake processes. The shallow corral, Corral 1, did not match lake background data, but that was not unexpected given its extremely shallow depth later in the summer. Corral 1 was located at a water depth of 0.3 meters (~1 feet) to 1 meter (3 feet), depending on overall lake depth variation over the summer, while the lake background samples were taken in significantly deeper water: 1.5 (4 feet) – 2.5 (8 feet) meters in depth. Aerial images and data show that this shallow enclosure created a separate environment warmer environment that did not match the surrounding lake area as closely.

These Phase I findings helped us to create an improved limnocorral design, develop more efficient sampling methods for Phase II, and demonstrated that, when intact, the limnocorrals are behaving as designed and can be used for experiments and analysis of UL processes.

3. Field Sampling and Laboratory Methods

3.1. Sampling Procedures

3.1.1. Pre-Lake Procedure.

Weather and wind can make UL a dangerous, difficult place to work. At the beginning of each week, we held a scheduling meeting to review the weekly schedule and evaluate weather forecasts. Each day, before leaving for a sampling trip, the teams evaluated the current weather forecast to determine if it was safe and feasible to proceed. At the lake, we evaluated current weather conditions before leaving the harbor; if wind speeds were greater than 10 miles an hour, we did not go out on the boat or fly the drones.

Before we left for UL, we confirmed that our equipment was ready for sampling. This included checking battery charges on drones and YSI ProDDS equipment (probes), verifying we had sufficient new sample bottles, that sampling equipment was cleaned and calibrated, and that we had the required field notebooks and data collection equipment as stated in the checklists.

3.1.2. Lake Safety

For safety and record keeping, we completed the boat authorization spreadsheet each day and followed the pontoon boat launch procedure checklist for each trip. The trip leader reviewed safety procedures and rules at the beginning of every trip.

3.1.3. Lake Sampling

We performed sampling according to established checklists (Section 12: Appendix) that were developed and updated over the course of the summer. We developed different sampling trip types: we collected water and single-point probe samples on Corral Water Sampling trips, we took probe vertical profiles on Probe-Only Sampling trips, and we took probe surface samples over a large area on Probe Data Tracks Sampling trips. We attempted to take drone aerial images of the Lake during all sampling trips, but due to wind and equipment issues, this was not always possible. During the Probe Data Tracks sampling, we collected gridded images from the drones, mapping water appearance across the entire sampling area (Figure 14). In Figure 14, the darker water at the TSSD outfall is actually clear and the darker color is sediments in the Lake. The majority of the lake is the light tan color seen immediately to the left of the outfall which has significant suspended solids, while the outfall has very low suspended solids.

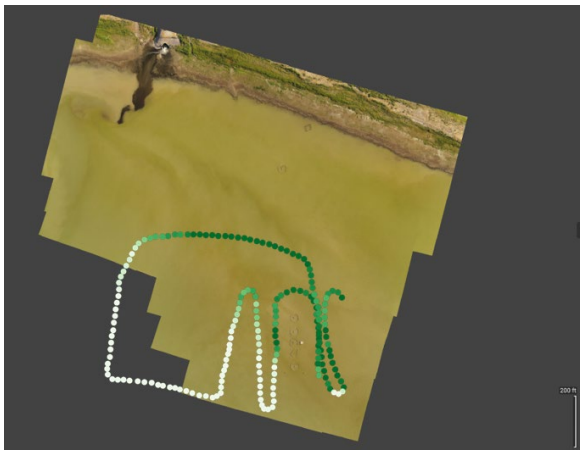

Figure 14 Visualization of data tracks superimposed on an image of the study area generated by a gridded flight with the drone.

Type Of Sampling Trip	Details of Sampling Trip
Water and Single Point Probe	Water samples and single-point surface probe samples were taken in all corrals and two lake locations.
Vertical Profile	The YSI probes were put into a "continuous" mode where samples were taken every second. For each corral and two lake locations the probe was slowly lowered to the lakebed so that the probe parameters could be compared with depth. Samples in each location were taken both on the way down and the way up.
Probe Data Tracks	The YSI probes were placed into a "continuous" mode where samples were taken every 10 seconds. The probes were then fastened onto the boat and dragged around the corrals and through the TSSD effluent plume.

We followed predetermined procedures for all sampling activities, including gridded drone flights, probe sampling, and water sampling. We recorded sampling activities in research journals by sample type and the time at which each sample was taken. The research journals are summaries of what we did, which lab member performed each task, and any notes on the task if required. Probe data were recorded on internal data loggers that were later downloaded. We distributed water samples to various laboratories for further analysis. We used a waterproof GoPro camera and personal cell phones to record sampling events and any specific issues or interesting features.

3.1.4. Sampling Locations

Although the specific measurements taken on each sampling trip varied somewhat throughout the sampling season due to weather complications, time constraints, equipment malfunction, and corral failures, we adhered as closely as possible to the following methods.

On the first sampling trip of each week, we collected a water sample from the southwest quadrant of each corral (diagram of quadrants shown in Figure 13) and probe single point measurements or vertical profile measurements from all four quadrants.

As previously mentioned, we also collected a water sample and a probe measurement from Buoy C6W, Buoy C2W, or both as background lake data. The locations of these buoys are shown in Figure 15.

On the second sampling trip of the week, we took single point and/or vertical profile probe measurements in all four quadrants of each corral and the buoy locations but did not collect water samples.

On the third sampling trip of the week, we took data tracks around the study area by dragging the probes behind the boat at very low speed. The purpose of the Probe Data Tracks was to explore how the probe parameters change with location in UL. More specifically we wanted to see the change in probe measurements around the corrals and through the TSSD effluent plume. A visualization of the data collected, superimposed on an image from the study area generated by a gridded flight with the drone, is shown in Figure 14. The path we followed for collecting data tracks varied significantly between trips because we wanted to explore several different paths in order to determine which one would provide the most interesting and useful data. We have not yet analyzed these data to develop a feasibility set of data tracks, this will be done in Phase II.

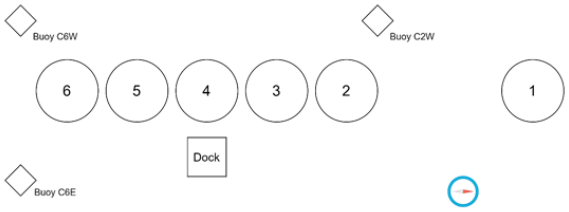

Figure 15 Diagram of limnocorral study area (not to scale)

3.1.5. Post-Sampling Procedures

We analyzed or froze water samples within 24 hours of collection. We filtered samples and ran the majority of our analyses in the TSSD's laboratory, then collected syringes of each sample (both filtered and unfiltered) to distribute to the BYU Environmental Analytical Laboratory (EAL), the BYU Geology Lab, and the BYU Environmental Engineering Lab for additional tests. We uploaded the results of the laboratory analyses onto the project SharePoint site (maintained by Jacobs Engineering), which provided access to all project members. We uploaded the probe data into a shared drive which was regularly synced with the SharePoint file, and uploaded pictures taken by the drones, on our GoPro, and on phones to the SharePoint file and to our shared drive. After uploading data, we recharged the drone batteries, cleaned the probes, and recorded the sampling trip in the research journal. We also checked the probe handhelds and put them on the charger when their batteries were below a 50% charge. A description and some preliminary characterization of these data in presented in Section 4.

3.1.6. Probe Calibration

We calibrated probe sensors at the beginning of every week. Table 2 presents the data we collected on probe sensor drift between weekly calibration events. We examined the sensor drift of each probe sensor to determine whether weekly calibrations were sufficient. We found that conductivity, Chl-a, pH, ORP, turbidity, and DO sensors exhibited the most drift in calibration, but that the drift was relatively minor (Table 2). The turbidity sensor had the most drift, with corrections on the order of 0.9 NTU, (the median value for UL was approximately 100 NTU), which is well within the manufacturer's specifications for calibration and very small compared to variation among field samples. We determined that these results justified weekly rather than daily calibration, we calibrated the probes weekly, just before the first sampling trip. Weekly calibration resulted in a significant cost and time saving.

Probe samples, weather permitting, were taken on Monday and Tuesday, so that the probes were calibrated no more than one day prior to a sampling event.

Table 2 Probe calibration drift.

Sensor	Avg Drift	Median Drift	Accuracy range
Conductivity	0.03	0.023	Conductivity - 0.001, 0.01 or 0.1 µS/cm (range dependent) Specific Conductance - 0.001, 0.01, 0.1 mS/cm
Chl-a (Phycocyanin)	0.10	0.033	Linearity: r2 = 0.999 for Rhodamine WT across full range
Chl-a (Chlorophyll)	0.04	0.027	Linearity: r2 = 0.999 for Rhodamine WT across full range
pH (pH7)	0.05	0.010	pH - ±0.2 pH units
pH (pH10)	0.04	0.003	pH - ±0.2 pH units
ORP	0.01	0.01	ORP - ±20 mV
Turbidity (0 FNU)	0.88	0.11	0 to 999 FNU: 0.3 FNU or ±2% of reading, whichever is greater 1000 to 4000 FNU: ±5% of reading
Turbidity (12.4/124 FNU)	0.90	0.14	0 to 999 FNU: 0.3 FNU or ±2% of reading, whichever is greater 1000 to 4000 FNU: ±5% of reading
Turbidity (1010 FNU)	0.09	0.04	0 to 999 FNU: 0.3 FNU or ±2% of reading, whichever is greater 1000 to 4000 FNU: ±5% of reading
DO	0.0042	0.0041	0 to 200%: ±1% of reading or 1% saturation, whichever is greater 200 to 500%: ±8% of reading 0 to 20 mg/L: ±0.1 mg/L or 1% of reading, whichever is greater 20 to 50 mg/L: ±8% of reading

3.2. Laboratory Procedures

3.2.1. Data Quality Objectives

For all sample analysis we followed the draft Data Quality Objectives (DQOs) for the ULNCS as presented in the request for proposal [2] and TSSD's 2022 Framework for the Utah Lake Solutions Program. These draft DQOs follow the United States Environmental Protection Agency's (EPA's) seven-step DQO process. The 2022 framework provides the most recent summary of objectives for this work. We view these DQO's and the 2022 framework as an essential component of the project as they provide the background, purpose, problem statement, conceptual model, and define the framework and requirements for the project. We followed DQOs and procedures from both the BYU Environmental Analytical Laboratory (EAL) and the TSSD Analytical Laboratory for the sample analysis performed in these laboratories.

3.2.2. Analytes

We measured pH, dissolved oxygen, temperature, specific conductance, turbidity, phycocyanin, chlorophyll-a, and depth *in situ* using YSI DSS Pro water quality sondes. This included point samples in the water sample locations, and vertical profiles in each deep corral.

We used the TSSD laboratory to analyze water samples for the constitutes listed in Table 3. The TSSD laboratory is certified for NH_3, NO_3, NO_2, TP, TDS, TSS, PO_4^{3-}, TOC, and COD, and is not certified, but is equipped to run VSS using EPA-compliant methods. Table 3 lists the tests and methods we ran on water samples at the TSSD laboratory.

Table 3 TSSD Laboratory methods and analysis

Item	Model/Method	Filtered/Unfiltered	Equipment
NH3	TNTplus 830 Ammonium	Filtered (0.45 µ)	DR 3900 Hach Spectrophotometer
NO3	TNTplus 835 Nitrate	Filtered (0.45 µ)	DR 3900 Hach Spectrophotometer
NO2	TNTplus 839 Nitrite	Filtered (0.45 µ)	DR 3900 Hach Spectrophotometer
TP	TNTplus 844 Phosphate	Unfiltered	DR 3900 Hach Spectrophotometer
PO43-	TNTplus 844 Phosphate	Filtered (0.45 µ)	DR 3900 Hach Spectrophotometer
PO43-	TNTplus 844 Phosphate	Unfiltered	DR 3900 Hach Spectrophotometer
TOC	TNTplus 810 TOC	Unfiltered	DR 3900 Hach Spectrophotometer
COD	Method 8000	Unfiltered	DR 3900 Hach Spectrophotometer
TDS	Method 2540 C	Unfiltered	VWR 1350FM Oven
TSS	Method 2540 D & E	Unfiltered	Binder ED53 Oven
VSS	Method 2540 D & E	Unfiltered	Thermolyne F62700 Oven

Table 4 EAL methods and analysis

Item	Method	Filtered/Unfiltered	Equipment
ICP metals	0.45 µ filter	Filtered (0.45 µ)	Thermo Scientific™ 7400 ICP-OES Radial Analyzer
ICP metals	Digestion Modified EPA 3051	Digested	Thermo Scientific™ 7400 ICP-OES Radial Analyzer
SRP	ASTM 4500-P G-199	Filtered (0.45 µ)	FIAlab FIAlyzer-2000

We used the BYU EAL (https://pws.byu.edu/eal) to analyze metals using inductively coupled plasma (ICP) spectroscopy and soluble reactive phosphorous (SRP). Our students were trained and certified by EAL staff to perform laboratory procedures. We followed the QA/QC processes and documents provided by the EAL for all SRP and ICP laboratory analysis. Our methods and analysis in the EAL are summarized in Table 4.

We only processed limited samples for SRP because of difficulties with sample preparation, costs, and machine reliability. We conducted both dissolved (i.e., filtered on a 0.45 µ filter) and digested ICP analysis for aluminum (Al), arsenic (As), boron (B), barium (Ba), calcium (Ca), cadmium (Cd), cobalt (Co), chromium (Cr), copper (Cu), iron (Fe), potassium (K), magnesium (Mg), manganese (Mn), molybdenum (Mo), sodium (Na), nickel (Ni), phosphorous (P), lead (Pb), sulfur (S), selenium (Se), silicon (Si), strontium (Sr), titanium (Ti), vanadium (V), and zinc (Zn). Detection limits vary by element but are on the order of a few µg/L or part-per-billion (ppb) level for these metals. For example, detection limit values for Al, Fe, and P are 1.51, 0.80, and 5.66 µg/L (ppb), respectively.

For anion analysis, we used the BYU Geological Sciences Laboratory (GSL) Dionex ICS-90 for Ion Chromatography (IC). We used the GSL's and the manufacturers' sample preparation and operation protocols and QA/QC procedures. We measured the major anions: fluoride (F^-), chloride (Cl^-), phosphate (PO_4^{3-}), and sulfate (SO_4^{2-}) in the water column. Our methods in the GSL are summarized in Table 5.

Table 5 GSL methods

Item	Method	Filtered/Unfiltered	Equipment
Anions (F^-, Cl^-, PO_4^{--}, SO_4^{--})	0.45 µ filter	Filtered (0.45 µ)	Dionex ICS-90

During Phase II, we will continue to define and review laboratory QA/QC procedures. Each individual workplan includes a Quality Assurance Plan and is reviewed by TSSD and the ULWQS Science Panel.

4. Preliminary Data Review

Table 6 TSSD Laboratory Results Summary

Analyte	N	Mean	Median	Std Dev	Range	Max	Variance
Nitrite (NO2-N)	104	0.047	0.042	0.039	0.167	0.160	0.002
Nitrate (NO3-N)	99	0.460	0.419	0.354	1.679	1.570	0.125
Ammonia (NH4-N)	105	0.154	0.119	0.117	0.475	0.503	0.014
Unfiltered React P (PO4-P)	109	0.369	0.134	0.614	4.088	4.110	0.377
Filtered React P (PO4-P)	78	0.068	0.044	0.067	0.294	0.296	0.004
Total P	89	0.266	0.242	0.116	0.584	0.666	0.013
Total N	13	3.421	3.010	1.537	5.680	7.200	2.364
COD	99	29.616	27.000	11.914	54.000	61.000	141.953
Filtered TOC	34	9.950	9.750	1.181	4.990	12.900	1.395
Unfiltered TOC	73	14.544	14.800	4.524	18.890	26.800	20.470
TSS	94	71.053	63.002	38.146	164.876	178.243	1455.106
VSS	105	0.314	0.438	0.205	0.479	0.479	0.042
TDS	94	1257.651	1243.286	113.930	531.390	1503.141	12980.083

COD – chemical oxygen demand *TSS – total suspended solids* *TDS – total dissolved solids*
TOC – total organic carbon *VSS – volatile suspended solids*

4.1. Laboratory Analysis

As discussed in Section 3.2, we performed sample analysis in three separate laboratories: we analyzed nutrient and water quality data in the TSSD Laboratory, we examined metal and reactive phosphorous in the BYU Environmental Analytical Laboratory (EAL), and we conducted anion analysis in the BYU Geology Laboratory. This section provides a brief overview of the data that were collected. It is organized by laboratory, as the analysis done at each laboratory is limited to specific areas with little overlap. The exception is nutrient data, which we analyzed at both the TSSD laboratory and in the soluble reactive phosphate (SRP) measurements taken at the BYU EAL laboratory. The ICP data also include total phosphorous. However, because of equipment issues, there are limited SRP data and ICP data are for total phosphorous, which may not be bioavailable.

We are currently performing an analysis of the data collected during Phase I which will include: evaluating how the different corrals behaved during the summer, correlations among the various parameters, and an initial effort to characterize lake processes using these data. Data analysis is complicated by the fact that most of the corrals lost structural integrity during the summer at different dates and for different periods of time, making it difficult to understand the reliability of data.

The summary statistics presented in this section provide a general overview of the dataset. They do not represent the actual behavior of the corrals as the reported statistics are computed using the entire data set which means that variation between the corrals and the lake present as larger variation in the data. This can be seen from the large standard deviation values compared to mean and median values. These values are not representative of the behavior of UL or of any individual corrals but do provide a good overview of the collected data and provide an initial check on data integrity. If the corrals are performing as expected, then the data from that corral would come from a different population than data from another corral or from the lake. By analyzing these data as a single data set, the standard deviation and variance are exaggerated. We are continuing data analysis to examine the impacts of the corrals and use the corral isolation to start to characterize lake processes.

Table 7 YSI Sondes measurement summary for surface probe readings taken in coordination with water samples.

Analyte	N	Mean	Std Dev	Range	Variance	Median
Barometer (mm Hg)	135	648.607	1.110	5.000	1.231	648.800
Conductivity (µS/cm) x	121	2077.036	154.336	629.000	23819.525	2128.700
Specific Conductivity (µS/cm)	124	2056.273	127.565	504.800	16272.917	2053.850
Salinity (psu)	124	1.047	0.067	0.260	0.005	1.040
TDS (mg/L)	124	1336.556	82.961	328.000	6882.493	1335.000
Temperature (°F)	123	77.774	2.746	11.000	7.538	77.200
Resistivity (ohms-cm)	120	483.313	37.031	146.400	1371.331	468.950
pH	108	8.727	0.282	1.190	0.079	8.635
ORP (mV)	114	115.432	18.290	89.100	334.509	113.800
Pressure (psi a)	138	0.202	0.214	0.732	0.046	0.133
Depth (m)	138	0.414	0.151	0.515	0.023	0.366
Conductivity (µS/cm) y	144	2022.549	226.104	1082.500	51123.222	2067.500
Chlorophyll (RFU)	138	0.526	0.291	1.310	0.085	0.480
Chlorophyll (µg/L)	138	15.879	9.340	42.570	87.228	14.070
Phycocyanin (RFU)	146	0.580	0.422	1.710	0.178	0.450
Phycocyanin (µg/L)	145	3.475	3.125	12.590	9.767	2.310
Turbidity (FNU)	124	105.027	57.111	218.800	3261.683	100.760
ODO (%)	147	107.204	33.438	186.200	1118.082	97.300
ODO (mg/L)	147	9.011	2.713	14.350	7.359	8.250

We present these summary statistics to provide an overview of the Phase I data and the number of analytes and samples measured. The statistics provide a good description of the general environment provided by the Lake and the corrals.

The data reported in this section have been refined by removing outlier values more than 1.5 times the range of the parameter. This may be too aggressive; we plan to develop more advanced methods to clean the data set taking into account that fact that data from different corrals come from different populations. This effort is completed by corral integrity as described. We will begin by computing the range for each corral or location, then use that value to clean the data for that location, rather than using the range of the entire data set. Using the range for entire data set may be excluding values for individual locations that are valid.

In the more in-depth, data-specific analysis we are currently conducting, we will present and define how outliers were defined and computed and what circumstances may have caused them. In addition, we will conduct a more thorough review of the outlier data before exclusion. For this initial data review, we used a more automated approach based on the entire data set for each parameter.

Report readers should understand these limitations on the statistics provided in this section.

4.2. Nutrient and Water Quality Data Overview

Table 6 presents a summary of the laboratory analyses we completed at TSSD. We analyzed between 75 and 100 samples for most constituents. We only analyzed 34 filtered TOC samples and 13 samples for Total N. As discussed above, the standard deviation and variance of these data are large, but provide a range of the conditions encountered during Phase I.

TDS values are large, with a large standard deviation of approximately 10% of the mean value. The mean value for total phosphorous (Total P) is 0.27 mg/L with a standard

deviation of 0.12 mg/L. The mean and median values are similar, indicating that the statistical distribution is not strongly skewed. Unfiltered reactive phosphorous has a higher mean value of 0.37 mg/L, with a lower standard deviation of 0.13. We attribute these values to the test methods: each of which extracts the phosphorous present in the sample in a different manner.

4.3. Probe Sample Overview

Table 7 provides summary statistics for the probe samples taken in coordination with the water samples. These samples were taken at a depth of about 0.4 meters (~ 1 foot). We took between 100 and 150 probe samples. The other probe samples, both vertical profiles and data tracks, were taken to help identify and characterize surface and vertical spatial patterns. We have started to analyze these data but do not yet have preliminary results.

While we have not performed a formal analysis, the TDS values estimated by the probe, which are based on conductivity measurements, are similar to the TDS values measured at the TSSD laboratory. The TSSD and probe measurements have mean values of 1,257 and 1,337 mg/L, respectively. This gives us some additional confidence in the probe estimation of TDS values as the TSSD values were generated by evaporating a filtered sample.

Dissolved oxygen in these near surface samples was high, with a mean of ~9 mg/L, which is approximately 107% of saturation at these temperatures. Dissolved oxygen is often over-saturated due to photosynthesis, which can significantly increase dissolved oxygen levels. The pH measurements were alkaline, with a mean value of 8.7±0.28, which is a relatively low variation over the entire summer season. The water temperature was relatively high, with a mean value of 78 degrees Fahrenheit and a standard deviation of only 2.7 degrees. Most of the sampling season was during the warm summer months.

We collected probe samples for several analytes. Many were measured indirectly, including dissolved oxygen (ODO), turbidity, chl-a, and phycocyanin (PC) using optical methods. Other analytes were estimated from conductivity measurements, such as TDS, and a few were measured directly, including conductivity, temperature, and pressure.

Probe data included surface samples at specific points, vertical profiles in each limnocorral, and probe tracks where we collected surface data as we dragged the probe through the experimental area. This resulted in between 3,000 and over 9,500 samples for each parameter. The number of probe samples per analyte is summarized in Table 8.

Table 8 YSI Sondes measurement summary including vertical profiles and data track measurements.

Analyte	# Data Points
Pressure (psi a)	9,546
Conductivity (uS/cm)	9,546
Specf Conductance (uS/cm)	9,546
Salinity (psu)	9,546
nLFCond (uS/cm)	9,546
TDS (mg/L)	9,546
Temperature (C)	9,546
Resistivity (ohms-cm)	9,546
Sigma-T (s t)	9,546
Sigma (s)	9,546
Chlorophyll (RFU)	5,229
Chlorophyll (ug/L)	5,229
PC (RFU)	5,229
PC (ug/L)	5,229
Turbidity (FNU)	3,466
TSS (mg/L)	3,466
ODO (% Saturation)	5,229
ODO (mg/L)	5,229
pH	3,084
pH (mV)	3,084
ORP (mV)	3,847

Table 9 BYU Geology Lab data summary

Analyte	N	Mean	Std Dev	Median	Max	Range	Variance
Fluoride	12	0.810	0.062	0.807	0.895	0.210	0.004
Chloride	10	13.270	5.554	11.309	21.765	16.042	30.845
Nitrate (NO3-N)	12	0.685	0.647	0.311	2.002	1.784	0.419
Phosphate	13	0.041	0.027	0.032	0.103	0.093	0.001

4.4. Anion Data Overview

Table 9 summarizes the anion data measured for the 29 samples analyzed in the BYU Geology laboratory; however, many of these samples were discarded because they were outliers based on our current data cleaning procedures. For this data set we believe these procedures are eliminating too many data and are reviewing and updating our approach. We analyzed for 5 different anions using ion chromatography in the BYU Geology laboratory but did not retain any of the sulfate data. We believe this was because of a non-numeric field in the database. We expect to update this soon.

One check we performed to determine the quality of the IC data was to compare with other data measuring the same analyte. The mean Nitrate (NO3-N) value using IC methods was 0.69, while the mean value measured at the TSSD laboratory using the Hach methods was 0.46. Both laboratories have large standard deviations associated with the measurement. Phosphate measurements are also similar, with median values of 0.03 and 0.04, for the IC and TSSD laboratory results, respectively again with both measurements having significant variation over the summer.

While we report these IC values, they are uncertain. Many of these values were generated while we were training students on the laboratory methods. During Phase II, we will have trained students and expect better data.

4.5. ICP Metal Data Overview

4.5.1. Background

To assist in producing a comprehensive chemical profile of the lake, we used Inductively Coupled Plasma Optical Emission Spectroscopy (ICP-OES) to evaluate total and dissolved concentrations of 25 metal elements. From June to October of 2021, we took multiple lake samples weekly and analyzed 169 of those samples using the EAL ICP machine. We analyzed both total and dissolved concentrations for 25 different metal elements, which resulted in 8,450 data points in total. Table 10 presents a statistical summary of these data with between about 70 and 90 points per analyte. The other values were excluded as outliers which had values greater or less than 1.5 times the range of the parameter. Again, we are reviewing the routines we developed for outlier identification as we feel that the number of data points rejected is significantly too high.

Table 10 presents a brief, preliminary statistical summary, like those presented for the other analysis methods. However, we present a more in-depth analysis of the ICP metal data in Section 5.3. The data presented in Section 5.3 have not had any outliers removed, so the distributions and whisker plots contain all the data as opposed to the summary presented in Table 10.

To our knowledge, this large data set for heavy metals in UL at the ppb-level is unique and we intend to perform additional ICP analysis to learn more.

Table 10 Summary of BYU Environmental Analytical Laboratory (EAL) ICP measurements

Dissolved Metals						Total Metals					
Analyte	**ICP**	**N**	**Mean**	**Std Dev**	**Median**	**Analyte**	**ICP**	**N**	**Mean**	**Std Dev**	**Median**
Dissolved Al	Al1670-D	89	0.98892	0.64868	0.83520	Total Al	Al1670-F	84	0.00664	0.00748	0.00405
Dissolved As	As1890-D	89	0.01094	0.00417	0.01090	Total As	As1890-F	85	0.01508	0.00353	0.01540
Dissolved B	B_2497-D	82	0.30726	0.03103	0.31405	Total B	B_2497-F	87	0.42010	0.05009	0.43570
Dissolved Ba	Ba4554-D	89	0.08748	0.01613	0.08760	Total Ba	Ba4554-F	86	0.09526	0.02361	0.08950
Dissolved Ca	Ca3179-D	89	53.61494	12.74124	55.49000	Total Ca	Ca3179-F	89	48.94618	11.95894	48.87000
Dissolved Cd	Cd2265-D	89	0.00017	0.00028	0.00020	Total Cd	Cd2265-F	87	0.00006	0.00023	0.00000
Dissolved Co	Co2286-D	89	0.00043	0.00042	0.00040	Total Co	Co2286-F	89	0.00034	0.00043	0.00030
Dissolved Cr	Cr2835-D	89	0.00377	0.00132	0.00380	Total Cr	Cr2835-F	88	0.00022	0.00055	0.00020
Dissolved Cu	Cu3247-D	76	0.01088	0.00967	0.00850	Total Cu	Cu3247-F	79	0.00303	0.00242	0.00230
Dissolved Fe	Fe2382-D	88	0.78421	0.47736	0.69685	Total Fe	Fe2382-F	83	0.00424	0.00362	0.00310
Dissolved K	K_7664-D	87	18.04839	2.18570	18.24000	Total K	K_7664-F	87	27.09989	3.81472	27.71000
Dissolved Mg	Mg2852-D	86	59.03826	6.76172	60.64000	Total Mg	Mg2852-F	87	79.75138	10.02429	82.32000
Dissolved Mn	Mn2576-D	88	0.02648	0.01536	0.02435	Total Mn	Mn2576-F	86	0.00118	0.00046	0.00110
Dissolved Mo	Mo2020-D	81	0.00580	0.00126	0.00570	Total Mo	Mo2020-F	85	0.00688	0.00123	0.00690
Dissolved Na	Na5895-D	85	161.14824	19.18025	164.30000	Total Na	Na5895-F	85	233.55529	29.38809	236.70000
Dissolved Ni	Ni2316-D	78	0.00448	0.00301	0.00330	Total Ni	Ni2316-F	89	0.00136	0.00096	0.00140
Dissolved P	P-1782-D	84	0.22848	0.09008	0.20275	Total P	P_1782-F	83	0.08518	0.04468	0.07190
Dissolved S	Pb2203-D	88	0.00363	0.00440	0.00415	Total S	Pb2203-F	89	0.00276	0.00269	0.00290
Dissolved Pb	S-1820-D	85	73.91506	7.93301	76.66000	Total Pb	S_1820-F	87	100.43379	13.12065	103.40000
Dissolved S	Se1960-D	88	0.00149	0.00349	0.00165	Total S	Se1960-F	89	0.00187	0.00303	0.00170
Dissolved Se	Si2516-D	89	8.52249	2.33873	7.93600	Total Se	Si2516-F	72	7.82846	0.67973	7.95550
Dissolved Si	Sr4077-D	87	0.97764	0.12701	1.00600	Total Si	Sr4077-F	89	1.10025	0.19960	1.10400
Dissolved Sr	Ti3088-D	89	0.02798	0.01884	0.02420	Total Sr	Ti3088-F	88	0.00001	0.00059	-0.00010
Dissolved Ti	V_3093-D	87	0.12706	0.01655	0.12840	Total Ti	V_3093-F	87	0.15756	0.02339	0.15960
Dissolved Zn	Zn2138-D	73	0.01362	0.00857	0.01290	Total Zn	Zn2138-F	81	0.01125	0.01016	0.01020

5. Experiments and Analysis

5.1. Overview

In this section we present some preliminary data analysis and results from some early experiments. These results and descriptions are not complete, and we expect to refine this analysis. Unless otherwise specified, only data collected from functional corrals and the open lake sampling sites was used for analysis.

5.2. Serial Filtration

5.2.1. Experiment Motivation

To examine the effectiveness of different laboratory procedures, we evaluated filtration methods by comparing the performance of a few different filter sizes (0.45µm, 0.7µm, and 1.5µm) when analyzing water samples. For example, the technical definition of "dissolved" in geochemical analyses is particles smaller than 0.45µm, but most standard methods for determining Total Suspended Solids suggest using a 1.5µm filter. Filtering at 0.45µm also requires more expensive filters and significantly more time. We performed a small serial filtration experiment using lake water collected near the corrals to obtain insights into what filtration level would be most useful for our studies, and to give us some preliminary information about the sorption status of various constituents in the lake water. Larger filters are easier to use; this set of simple experiments demonstrated how filter size might affect our results.

5.2.2. Methods

We conducted a small serial filtration experiment to examine filter size effects on metals in UL water. After filtration, the samples were digested before analysis.

On August 11th, we collected a large (roughly 1 gallon) surface sample near the corrals in a clean Nalgene jar, then immediately brought the sample to the EAL for analysis. We collected three mixed, homogenous splits from the sample, and successively filtered each split through a 1.5µm glass fiber filter, a 0.7µm glass fiber filter, and a 0.45µm membrane filter, using new filters for each split, with approximately 20mL of sample reserved for analysis at each step, including a 20mL sample of unfiltered water from each split.

This resulted in three replicates of sample at each four filter sizes, unfiltered, 1.5µm, 0.7µm, and 0.45µm. We performed a microwave digestion on these samples, and then analyzed all the samples on the ICP in the EAL.

Since this was a preliminary analysis, we did not repeat this analysis for other samples.

In this section, we present preliminary experimental data on the metals of particular interest in our work: phosphorous, aluminum and sodium, though we have ICP data for all 25 metals.

5.2.3. Results

Overall, we found that filter size had a significant impact on measured sample concentrations in the ppb range, but the ppb range is not significant for TSS as the amount of colloidal material in the 1.5 to 0.45 µm range is small. Based on these results, we decided to use a 0.45µm membrane filter for all filtered samples. For geochemical

analysis, 0.45µm is considered the boundary between dissolved and suspended or colloidal particles.

Prior to obtaining these results, some of the samples collected in early summer were run on 1.5µm or 0.7µm glass fiber filters because we did not have any 0.45 µm filters. For the second half of the sampling campaign, we ran all the filtered nutrient analyses using a 0.45µm membrane filter. We started using 0.45 µm filters before we analyzed these results to support this decision.

Our results are further explained in terms of their relevance for specific metals in the rest of this section.

5.2.3.1. Phosphorus Concentrations.

Phosphorus concentrations ranged from about 0.5 to 0.7 ppm (mg/L). There was slight variation in the results between the three replicates, but overall, there is a clear decrease in concentration at every level of filtration, as shown in **Figure 16**. The largest change in concentration occurred between no filtration and filtration on a 1.5µm filter. The overall magnitude of the decrease, however, is quite small—the complete filtration process only reduced the average concentration by 0.1064 ppm (from 0.6260 ppm to 0.5196 ppm). Since filtration at 0.45µm removed only about 17% of the phosphorus compared to the unfiltered samples, for this sample most of the phosphorus in the water column can be considered dissolved and likely present as PO_4.

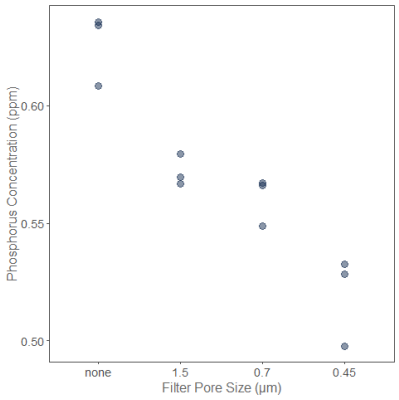

Figure 16 [Phosphorus] concentration versus filter pore size shows that phosphorus is associated with suspended solids.

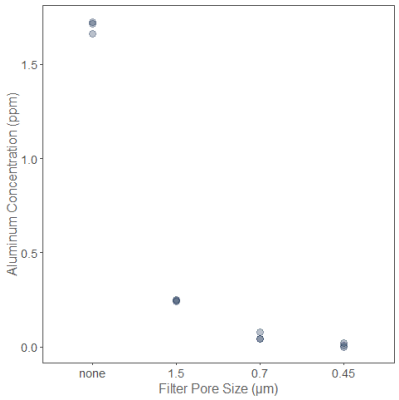

Figure 17 [Aluminum] versus filter pore size shows that aluminum is associated with suspended solids, but not as much with colloidal particles (<1.5µm).

5.2.3.2. Aluminum Concentrations.

In contrast to phosphorus, filtration removed a significant amount of aluminum from the samples, with the largest decrease occurring between no filtration and filtration on a 1.5µm filter, as shown in Figure 17. The first filtration (i.e., 1.5µm filter) reduced the concentration by 1.4562 ppm (from 1.7007 ppm to 0.2445 ppm), or approximately 85%. Full filtration reduced the initial concentration by 1.6923 ppm, over 99%. Since filtration removed 99.5% of the aluminum in the sample, with most being removed at the 1.5µm step, it is likely that most of the aluminum in the water column is found on particles larger than 1.5µm and very little of it could be considered dissolved, but rather present as colloidal or larger size particles.

5.2.3.3. Sodium Concentrations.

We observed an unexpected result with Sodium, which increased after filtration on a 1.5µm filter, as shown in

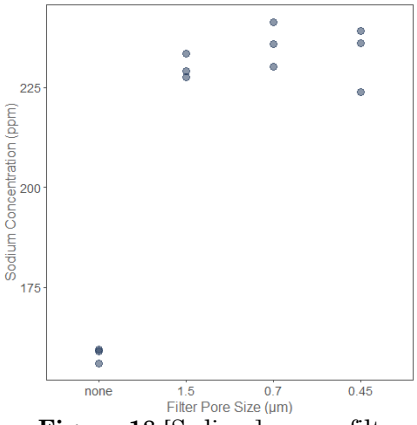

Figure 18 [Sodium] versus filter pore size shows an unexpected result of increasing concentrations with decreasing pore sizes. This suggests that the filters were adding sodium to the sample that passed through them.

(Figure 18). The most likely explanation for this is that the sample extracted sodium from the glass fiber filters. Unfortunately, we neglected to run a DI water blank in this experiment, so we cannot be certain.

The same trend of increasing concentration after filtration was also observed, at varying degrees of intensity, with Vanadium, Sulfur, Molybdenum, Magnesium, Potassium, and Boron. We suspect that this is due to salts on the filters. This is an effect that needs to be investigated with further experiments during the summer 2022 field sampling campaign. As part of our 2022 investigation, we will filter distilled water as a blank to determine if the filters have salt contamination.

5.2.4. Discussion.

Although this experiment was very small and not especially rigorous, the results suggest some interesting observations that confirm other data collected during the summer 2021 sampling campaign and warrant further research in 2022.

Aluminum appears to be present in the water column mostly as large, undissolved particles, while phosphorus appears to be present mostly as dissolved particles smaller than 0.45µm.

Further work is needed to fully investigate and describe the effects of different filter sizes on our analyses and determine the particle sizes of various constituents in the water column. Most importantly, we will run a DI water blank as a control in our 2022 analyses. It would also be useful to do a similar experiment with other tests in addition to the ICP, including the nutrient tests performed at TSSD's lab, and to test additional filter pore sizes.

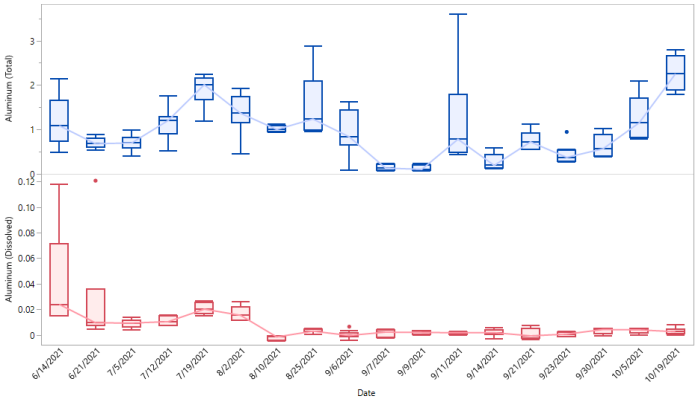

Figure 19 Time series of total (above) and dissolved (below) aluminum (Al) concentrations (ppm or mg/L) of lake samples.

5.3. ICP Analysis of Heavy Elements Preliminary Results

5.3.1. Background

As discussed in Section 4.5, we used Inductively Coupled Plasma Optical Emission Spectroscopy (ICP-OES) to evaluate total and dissolved concentrations of 25 metal elements: aluminum (Al), arsenic (As), boron (B), barium (Ba), calcium (Ca), cadmium (Cd), cobalt (Co), chromium (Cr), copper (Cu), iron (Fe), potassium (K), magnesium (Mg), manganese (Mn), molybdenum (Mo), sodium (Na), nickel (Ni), phosphorous (P), lead (Pb), sulfur (S), selenium (Se), silicon (Si), strontium (Sr), titanium (Ti), vanadium (V), and zinc (Zn). From June to October of 2021, we took multiple lake samples weekly and analyzed them using the ICP machine at BYU's Environmental Analytical Laboratory (EAL): a total of 169 data points for each element over a period of 19 weeks. We present a brief analysis of this unprecedented data set.

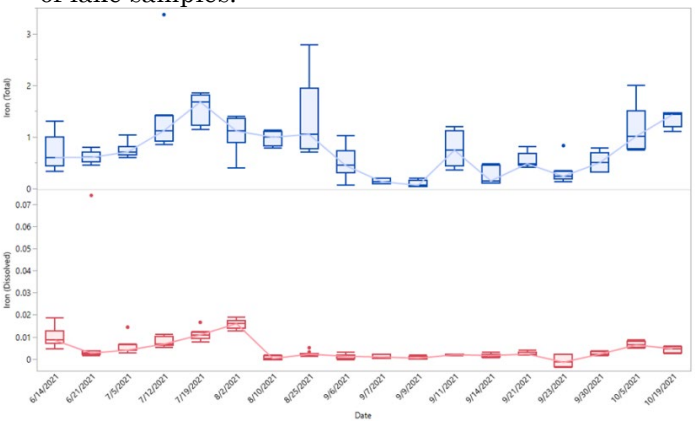

Figure 20 Time series of total (above) and dissolved (below) iron (Fe) concentrations (ppm or mg/L) of lake samples.

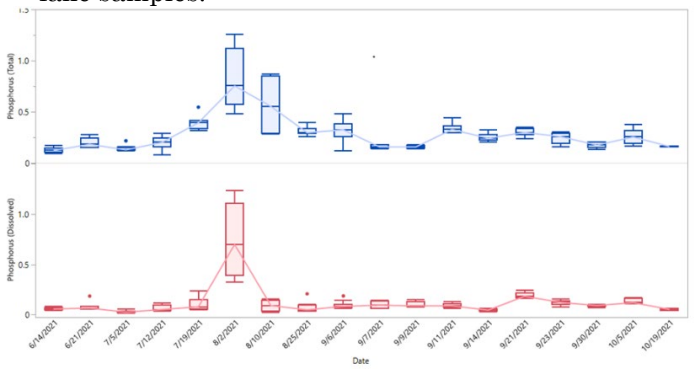

Figure 21 Time series of total (above) and dissolved (below) Phosphorus (P) concentrations (ppm or mg/L) of lake samples.

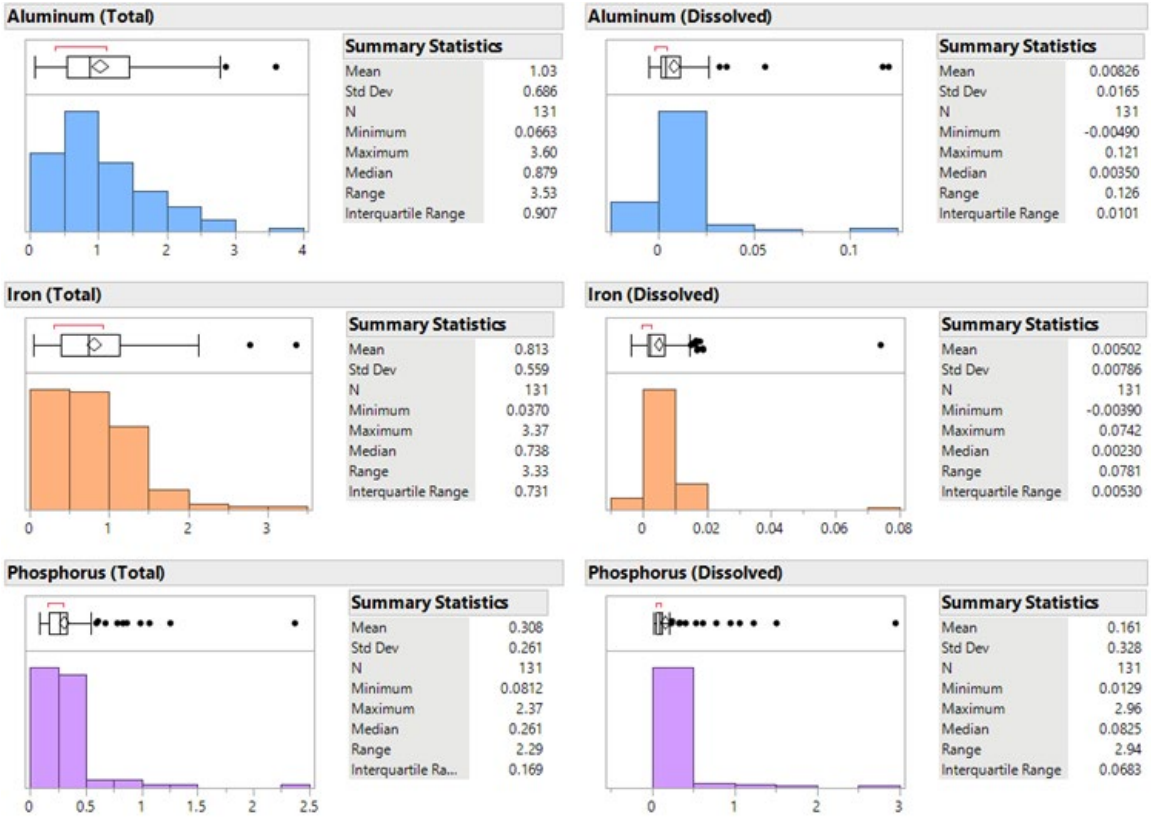

Figure 22 Median concentrations of total and dissolved aluminum (Al), iron (Fe), and Phosphorus (P) concentrations (ppm or mg/L) of lake samples.

5.3.2. Methods

We measured total concentrations by performing a microwave digestion using the EAL's modified EPA 3051 method on each sample before ICP analysis. We measured dissolved concentrations by filtering the sample through a 0.45 µm filter before analysis. We analyzed all samples with the ICP-OES at BYU's EAL.

QA/QC methods for this study include bottle blanks in the field as well as a standard and process blank for sample preparation. The ICP machine at BYU's EAL runs a quality check (QC) every 15 samples on known standards. If an element fails a QC check, the machine reanalyzes all samples completed after the last passed QC. In addition to the bottle blanks, standard, and process blank, we ran an additional standard and two ultrapure water samples through the ICP to provide more stringent QC data.

5.3.3. Results

Our study generated data for all 25 elements analyzed, but in the interest of time, we will only present detailed data for aluminum (Al), iron (Fe), and phosphorus (P). Phosphorus is of particular interest because it is essential to algal growth. These samples were taken both from the Lake and from the various limnocorrals. Depending on the time of year, the corrals provided some isolation from Lake processes. Drone images, laboratory data, and probe measurements show there was a range of turbidity or

suspended solids in the corrals. In this analysis we do not attempt to separate results by corral but to present the data as a single population over time.

Time series plots, histograms, and summary statistics for all 25 analytes are available in sections 9 and 10 in the appendix.

Figure 19, Figure 20, and Figure 21 present time series of total and dissolved concentrations for Al, Fe, and P, respectively. Figure 22 presents histograms for median concentrations of total and dissolved Al, Fe, and P in mg/L. Note that the y-axis concentration scale is significantly different for the total and dissolved concentrations for most of the plots.

Median values for total and dissolved Al are 879 µg/L and 3.5 µg/L, respectively. The 75th percentile and maximum excluding outlier concentrations for total Al are about 1.5 mg/L (1,500 µg/L) and 2.8 mg/L (2,800 µg/L), respectively. The total Al concentrations are very high. We attribute these high total concentrations and low dissolved concentrations to suspended clay particles meaning that this aluminum is probably not bioavailable. Median values for total and dissolved Fe are 738 µg/L and 2.3 µg/L, respectively. Median values for total and dissolved phosphorus are 261 µg/L and 8.25 µg/L, respectively.

Dissolved Al concentrations in UL samples were at levels expected for surface waters, but total Al in lake samples was routinely above 1000 µg/L. The state of Utah's regulation for Al is 750 µg/L, but there is some ambiguity as to whether this regulation refers to dissolved aluminum or "total recoverable" aluminum. If the latter, it is necessary to specify what methods should be used to obtain total recoverable aluminum as total digestion results in the UL being significantly out of compliance. During Phase II, we plan on exploring other extraction and filtration methods to determine the partitioning of Al in the lake and determine a useful, clear standard for aluminum.

These data show that Al is mostly associated with larger suspended solids with a reduction in the mean concentration of approximately 99.6% between total and dissolved concentrations. These results are based on 169 data points taken over the entire summer. The variation in both the total and dissolved sample sets is relatively small indicating that this result is valid. Figure 19 shows that mean total Al concentrations varied over the course of the summer, with variations on any given day also changing. While we have not yet analyzed the data, we expect these concentrations and variations are correlated with TSS and turbidity. The mean dissolved Al concentrations show very little variation and almost 2 orders of magnitude smaller than the total concentrations. Except for the first sample date, concentrations on any given day also show little variation.

Median values for total and dissolved Fe are 738 µg/L and 2.3 µg/L, respectively (Figure 22). Figure 20 shows that dissolved Fe concentrations were relatively constant over the summer, while total Fe concentrations showed more variation, though not as much as did the Al samples. We expect that total Fe concentrations are correlated with TSS and turbidity. Dissolved Fe concentrations show a rise and a peak through July and into early August (Figure 20). Fe has a much higher solubility limit in anoxic or anerobic water. We expect, though we have not yet done the analysis, that these higher values might be correlated with DO levels and may also be related to which corral was sampled,

as the corrals restricted mixing and may have resulted in reduced DO in this warmer period near the sediment-water interface when the corrals were functioning.

Median values for total and dissolved phosphorus were 261 µg/L and 8.25 µg/L, respectively (Figure 22). Dissolved concentrations showed little variation over time, except for the sample on August 2 (Figure 21). Total phosphorous concentrations were also relatively stable, with a peak in concentration at the beginning of August (Figure 21). There does not appear to be much correlation with Al concentration for this August peak in phosphorous.

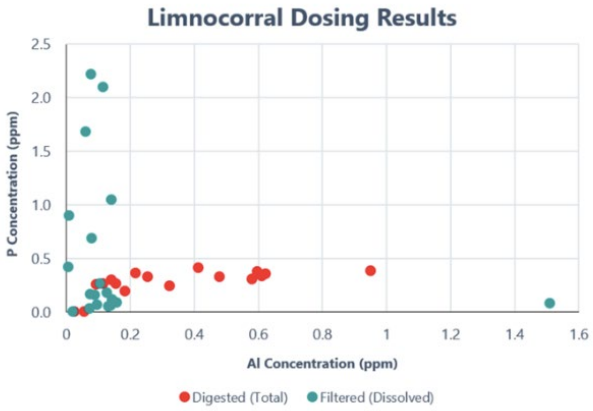

Figure 23 Preliminary data that is currently being analyzed from the limnocorral dosing experiments.

Continuing into 2022, we plan to integrate more rigorous QA/QC methods, potentially including matrix spikes to evaluate filtering activities.

5.4. Aluminum Dosing Experiment

5.4.1. Background

We performed an initial bench-scale experiment to evaluate the ability of aluminum to co-precipitate phosphorous and remove it from the water column. This process has been used in other water bodies, but the extremely high dissolved solids content and the total aluminum content of UL water could interfere with the process.

To characterize phosphorous sequestration using aluminum salts we took a large sample of UL water and ran a series of jar tests to evaluate the effectiveness of this approach in UL water. We treated jars using an aluminum chlorohydrate (ACH) solution and used target Al:P ratios ranging from 0.25:1 to 6:1. On July 30th, we dosed samples in triplicate using Al:P ratios of 0.25:1, 0.5:1, 1:1, and 5:1. On August 3rd, we dosed with a single ratio of 1:1, again in triplicate. Out of curiosity we also acidified and dosed three samples on that date.

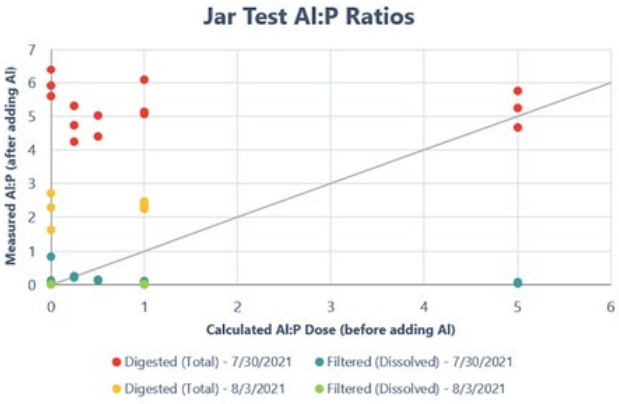

Figure 24 Preliminary jar test data indicated that UL has high aluminum concentrations which likely affected the experimental dosing process.

We evaluated the controls and dosed samples for both total and dissolved Al and P content using ICP-OES at BYU's EAL. Total Al and P was determined using a microwave digestion, and dissolved Al and P was determined by filtering the sample through a 0.45-micron filter.

Figure 25 Dyed corrals with 10 mL of rhodamine per corral.

5.4.2. Results

Preliminary jar test data indicates that over time, the lake was able to compensate for added Al, resulting in no discernable increase in Al concentration after later doses. This suggests that UL's high Al concentrations may have affected the experimental dosing process. The jar tests showed no discernable decrease in phosphorous concentration. However, in other water bodies, the molar concentration of aluminum added to sequester the phosphorous was on the same order as the molar concentration of phosphorous in the water column. In the case of UL water, this was an insignificant amount of aluminum, less than the variation of total aluminum concentration among the samples. Figure 24 shows that the ratio of total aluminum to total phosphate in unfiltered samples is on the order of 7, while that of the filtered samples is significantly lower.

We intend to extend these jar tests during Phase II to determine if aluminum salts could effectively sequester phosphorous in UL. This approach may work in waters with significantly lower TDS values, such as wastewater treatment plant outfall, but preliminary data indicate that will probably not be effective in UL.

5.5. Limnocorral Mixing/Dilution Studies

5.5.1. Rhodamine Dosing

5.5.1.1. Background

One challenge associated with the use of limnocorrals to study the lake water column is uncertainty regarding the level of isolation produced by the limnocorral mesocosm. Complete isolation is not desirable, as this would produce an environment very different from that of the general lake water. By the same token, too much flow in and out of the mesocosms would diminish the experimental quality. In response to these concerns, we need to understand the flushing rate of the corrals. The flushing rate is a measure of how long it takes for the water inside a corral to be completely replaced by outside lake water due to natural movement of water in and out of the corral. Especially in experiments that involve dosing, such as our Aluminum Salts and Phosphorus Addition Experiments, it is essential for us to have a quality estimate of the flushing rate within the corrals. To better understand this, we dosed the corrals with rhodamine water tracer dye on two separate occasions and measured the resulting dilution time by collecting water samples from the corrals and analyzing the concentration of rhodamine using a fluorescence spectrophotometer.

5.5.1.2. Methods

Rhodamine is a fluorescent water tracer dye used for many water studies. Rhodamine is non-toxic and inert, so it does not affect biological and chemical processes in the water column. It is visible to the naked eye at concentrations greater than 100 ppb and it is observable on a fluorometer at concentrations as low as .006 ppb. We used the Fluorometric Procedures for Dye Tracing manual from the USGS to design our dilution experiment and a Perkin-Elmer LS55 Luminescence Spectrometer. To dose each corral, we filled 3-gallon pressurized sprayers with DI water and added 10mL of rhodamine, then applied the mixture evenly to the surface of the water inside the corral, this took approximately 20 minutes per corral, so the added tracer was better mixed in the water (Figure 26). We dosed corrals 4, 5, and 6 on the morning of September 6th, 2021 and took measurements every 2 hours for the rest of the day, and then daily for the next 7 days.

Figure 26 Dosing the corrals with rhodamine with the top panel showing the garden sprayer, and the bottom panel showing application to the middle corral.

5.5.1.3. Results

Although we added just 10mL of rhodamine to the 100,000L corrals, the water was noticeably pink for several days after dosing (Figure 25). The day we dosed was very calm and warm, and the water column was not well-mixed. The dye remained in the top two feet of water in the corrals for most of the day, which means the concentrations we measured that day did not accurately reflect the concentration in the entire corral. By the next day, however, the water was thoroughly mixed, and successive sampling yielded reasonable dilution curves.

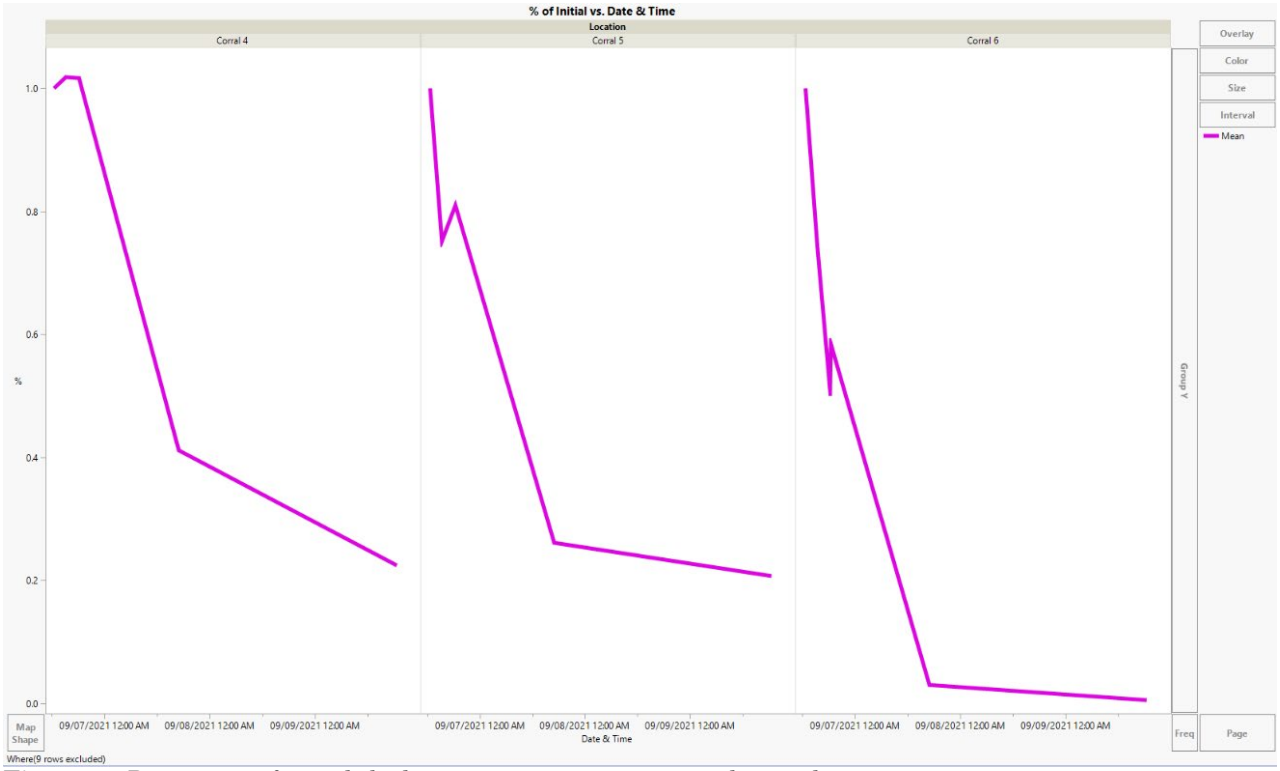

Figure 27 Percentage of initial rhodamine concentration versus date and time

Figure 27 shows the results of the samples collected on the day of dosing and for two days afterwards. Values are plotted as percentages of the initial concentration, as measured on the fluorometer according to the USGS procedure.

These results indicate that the flushing time in the corrals is quite short. Because the corrals were stratified on the first day, however, the concentrations in the surface water from which samples were collected were artificially high. This made the concentrations appear lower on successive days when the dye was mixed throughout the entire corral. Another known issue is that sunlight degrades rhodamine, which means that in the small portion of light-penetrated water at the top of the corral, rhodamine concentration levels likely decreased faster than if they had decreased solely through dilution.

These two factors mean that the actual flushing time in the corrals is likely longer, possibly around 3-6 days. The difference in the shape of the dilution curve for Corral 6 could be due to the fact that the turbidity of the water in Corral 6 was significantly higher for the duration of this experiment, which could have impacted the sorption of the dye and the readings on the fluorometers. The significant difference between Corral 6 and the rest of the corrals is explained by a later discovered 20% failure of the C6 skirt: 2 out of 10 sides were open to the lake.

5.5.1.4. Conclusions

Further dilution experiments are necessary to get a more accurate estimate of the flushing rate in the corrals, but for now we know that complete turnover of water in the corrals occurs in a matter of days, thanks to the permeable skirt. This is a good flushing rate for the experiments we are conducting because it is not so slow that the

environment inside the corrals significantly affects water column processes, but not so fast that our experiments are diluted before we can measure their effects.

5.6. Turbidity Observations

We have not attempted to perform an in-depth analysis of the turbidity or TSS data yet, but we have completed a preliminary first look to verify that the data are within expected values based on prior knowledge of Utah Lake geochemistry. This preliminary analysis highlights some interesting features.

Table 11 Summary statistics for probe data on turbidity (units of FNU).

100.0%	Max	222.47	Mean	105.03
99 .5%		222.47	Std Dev	57.11
97.5%		216.00	Std Err Mean	5.13
90.0%		172.93	Upper 95% Mean	115.17
75.0%	quartile	149.86	Lower 95% Mean	94.87
50.0%	median	100.76	N	124
25.0%	quartile	60.08	Variance	3261.68
10.0%		7.52	Skewness	-0.20
2.5%		5.55	Kurtosis	-0.85
0.5%		3.67	Range	218.80
0.0%	min	3.67	Interquartile Range	89.79

Table 11 and Figure 28 present the summary statistics and distribution plots for the probe data characterizing turbidity in Formazin Nephelometric Units (FNU), which are equivalent to more commonly reported Nephelometric Turbidity Units (NTU). The summary statistics imply that the data are nearly Gaussian: of normal distribution with low skew and kurtosis values. The histogram, however, indicates that the data consists of two or three different populations, each approximately Gaussian. This shown by the three peaks, each showing an approximately Gaussian shape.

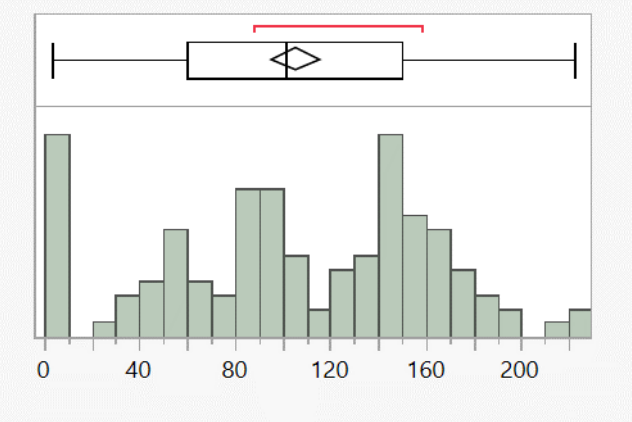

Figure 28 Distribution of all turbidity measurements collected with the probes over the entire sampling period. This includes measurements from the open lake, deep water corrals, and shallow corrals.

In addition to the probe FNU data, we also have aerial images that show the water in the corrals differs in turbidity from the lake, and that turbidity levels differ among the corrals. We attribute the difference in turbidity among the corrals to the varying integrity of the corral floats and skirts. Figure 29 shows aerial images from September 3rd and 11th, 2022. In these images the two corrals on the right (top panel, September 3rd) and the rightmost corral (bottom panel, September 11th) have been removed from the Lake, reinforced, and replaced. The images show that the water in the reinforced corrals is less turbid than the lake water. Corral 6, the leftmost corral in both panels, was repaired in the lake but still had significant tears in the skirt which allowed more water transfer between the lake and corral, and the images show the turbidity levels in the corral are similar to the lake water. It appears that the reinforced corrals, which are better able to isolate the water column, have lower turbidity levels than the lake. The distinct data populations shown in **Figure 28** may be due to this phenomenon, where functioning corrals isolate the water column from the lake and have different turbidity values than the lake and the broken corrals.

Figure 29 Aerial image from September 3 and 11, 2022 on the top and bottom, respectively, showing different turbidity in the reinforced corrals, which are the corrals on the right. The left-most corral is Corral 6, which was not removed and reinforced. These corrals appear to also have higher algal concentrations based on the green color.

Because our Phase I data includes relatively few measurements from fully functioning corrals that were properly isolating the water column, it is difficult to compare data from the corrals to background data. When the corrals were not fully functional, the differences are minor. We will continue to work with the Phase I data to develop better analyses, but we expect that the Phase II data will be better suited for analysis thanks to refined corral design and other experiment and testing procedures. The YSI probes measure both chlorophyll (chl-a), which is a pigment present in many phytoplankton groups, and phycocyanin (PC), which is a pigment mainly produced by cyanobacteria. This allows us to investigate how water column turbidity influences populations of phytoplankton in general and, specifically, the cyanobacteria that cause toxic algal blooms.

Figure 31presents example data indicative of turbidity and algal concentration from September 6th and 11th, which includes both chl-a and PC concentrations. These data match the aerial images in **Figure 29**, but with data from September 6th rather than September 3rd, as this is the closet match. We excluded data from Corral 6 (C6) on September 11th, as the readings were anomalous, with turbidity in the 30 NTU range. The temperature measured in the corrals is very similar to the measured temperature in the lake. This indicates that, while the corrals are isolating the water column from some processes, the general environment in the corrals is similar to that in the lake.

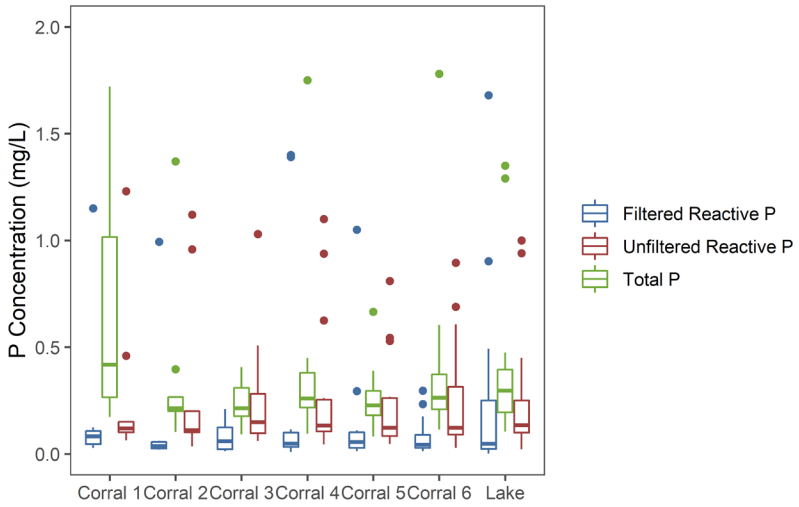

Figure 30 Filtered reactive phosphorous (blue), unfiltered reactive phosphorous (red), and total phosphorus (green), by Location

5.7. Geochemical Variation

One of the primary nutrients that influences algae growth is phosphorous, so understanding the distribution of phosphorous is an important aspect of the statistical analysis. Figure 30 shows a breakdown of the three phosphorous tests that were run at TSSD, separated by location. On the plot, green represents Total Phosphorous, blue represents Filtered Reactive Phosphorous, and red represents Unfiltered Reactive Phosphorous. This plot shows that total phosphorous is higher than unfiltered reactive phosphorous, which is higher than filtered phosphorous. This is an expected result. There are several outliers that need to be examined prior to further analysis, but this initial analysis shows what we expected to see.

Looking at total phosphorous by location (**Figure 30**), differences between locations become readily apparent. Corral 1 was consistently higher in many nutrients, including phosphorous. This can be seen in **Figure 30**, which indicates that Corral 1 did not effectively mirror the conditions of the rest of the lake. While Corral 1 did not mirror the lake well, the rest of the corrals proved to be a good estimation of the lake. **Figure 30** shows that both lake locations and the five other corrals all have similar median total phosphorus levels. This lack of similarity between Corral 1 and the lake seems to be connected to its lack of depth, since that is the only significant difference between it and the other corrals.

Figure 31 presents box plots of total suspended solids (TSS) and total dissolved solids (TDS) broken up by location. The red box plots are TDS, and the blue are TSS. It is clear to see that TDS is significantly higher than TSS, regardless of location. This is another expected result. Other studies from universities as far as Iraq and China have found that fact to be the same in numerous other lakes [16, 17].

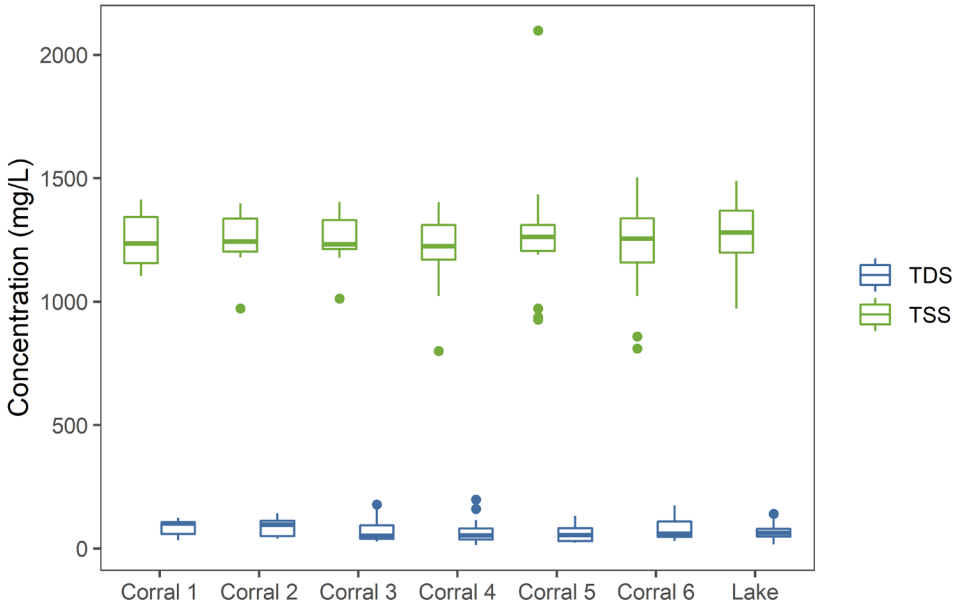

Figure 31 TSS and TDS by Location

6. Sediment and Porewater Chemistry

6.1. Introduction

Dr. Greg Carling from the Department of Geological Sciences conducted work in three categories during Phase I: 1) sediment chemistry; 2) pore water chemistry; and 3) geochemical modeling. His methods and results follow.

6.2. Sediment Chemistry

To characterize phosphorus chemistry of sediments, Dr. Carling measured P speciation through sequential extractions and will quantify mineralogy for six sediment samples during work in Phase II. Dr. Carling's group collected samples for this work inside and outside of the shallow vegetated limnocorral and from the open water (two samples per location) (Figure 33).

Figure 32 Student helpers displaying peeper samples and sediment samples collected outside the vegetated limnocorral.

The goal of the sequential extractions was to determine phosphorus speciation in sediment. He measured phosphorus fractions including:

- loosely bound P (Step 1),
- Fe-P (Step 2),
- exchangeable P (Step 3),
- Ca-P (Step 4), and
- refractory P (Step 5)

following the protocol of Hupfer, et al. [18] and Randall, et al. [19]. Dr. Carling's team's results show that most P is bound in the calcite (Step 4) and refractory (Step 5) fractions (Figure 34). This contrasts with previous work in other parts of UL that showed a substantial amount of Fe-bound P [19, 20]. Further work is needed to better understand spatial variability in P speciation in sediments across the lake and in the limnocorrals.

Figure 33 Sample locations for sediment and peeper samples. The samples are labeled "open water", "outside limnocorral", and "inside limnocorral".

During Phase II, Dr. Carling will measure sediment mineralogy by x-ray diffraction using the Rigaku Miniflex XRD at BYU. The mineralogy will provide information on the major mineral phases in UL sediments that may contribute to phosphorus cycling. The mineralogy results should be ready by the spring of 2022.

6.3. Pore Water Chemistry

Dr. Carling installed peepers to measure *in situ* pore water chemistry at the same three locations where sediment samples were collected (Figure 33). The peepers provided a two-week time-integrated snapshot of pore water chemistry. His team built the peepers following the design of Johnston, et al. [21]. These were the same type of peepers used by Dr. Josh LeMonte in a separate study funded by the Utah DWQ.

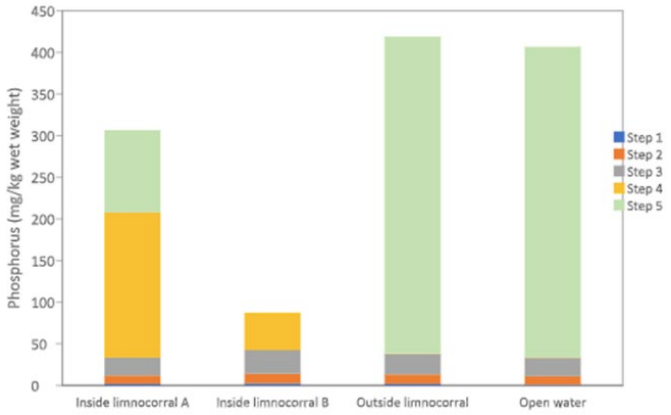

Figure 34 Sequential extraction results showing P concentrations in each leaching step expressed as mg/kg wet weight. The concentrations will be higher once corrected for dry weight, but the relative amount of P in each fraction will remain the same. Note that Step 5 was not measured for "Inside limnocorral B" because the sediment sample was lost during preparation.

The peepers provided measurements of total phosphorus, iron, orthophosphate, aluminum and other metals, major cations, and major anions at 4 cm increments from just above the sediment-water interface to a depth of about 20 cm below the sediment. They collected their samples during October 2021.

The results show a dynamic system with variable concentrations of nutrients (total dissolved P and nitrate) and other redox-sensitive compounds (total dissolved Fe and sulfate). In all three peepers, sediment pore water showed relatively high P, high Fe, low NO_3, and low SO_4 concentrations relative to the water column. In this proof of concept, the results show that the peepers are able to capture variable water chemistry of short distances in the water column and sediment. Additional work is needed to investigate seasonal variability across multiple limnocorrals.

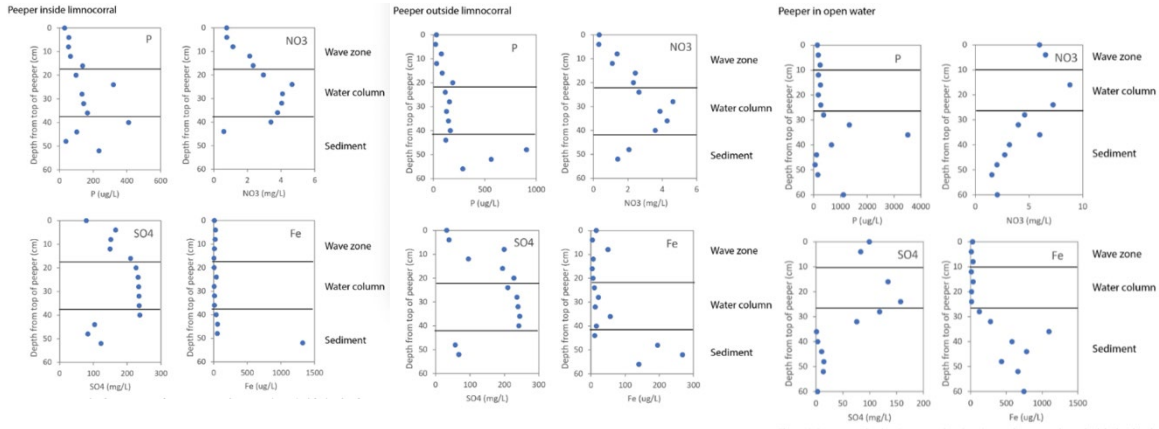

Figure 35 Peeper results showing water chemistry in sampling ports above the lake level in the wave zone, in the water column, and in sediment pore water for the peeper placed outside the limnocorral for the peeper inside the limnocorral, outside the limnocorral, and in open water in the left, middle, and center panels, respectively.

6.4. Geochemical Modeling

For Phase II, Dr. Carling will build equilibrium geochemical models for the water column and pore water to determine potential reactions involving phosphorus and aluminum. He will develop these models using Phreeqci and/or The Geochemist's

Workbench. To develop a realistic model, he will need accurate orthophosphate, major anion, and alkalinity concentrations in addition to the major cation and metals data already being collected from the limnocorrals. In Phase II, Dr. Carling's team will use the ion chromatograph (IC) in the geology department to analyze major anion concentrations (fluoride, chloride, nitrate, orthophosphate, and sulfate) and measure alkalinity by acid titrations. These measurements will be made on enough samples to provide confidence in the model for each limnocorral treatment. The models have not been developed.

7. Conclusions

7.1. Phase I Objective Accomplishment

As stated, the research goals for Phase I were to develop sampling and analysis methods to characterize geochemistry in UL, to determine how to place and operate limnocorrals, and to, if possible, start to develop baseline data. We have accomplished our objectives by developing a sampling schedule, characterizing problems with our limnocorrals, and identifying solutions. Through close coordination with the TSSD, we were also able to obtain preliminary data regarding lake geochemistry that has given us context for our studies and data use moving forward. Through our partnership with TSSD, we were also able to develop more sophisticated limnocorrals to produce more durable mesocosms that will allow us to gain more consistent data over the course of future project phases. With our Phase I goals met, we are confident in our ability to progress to Phase II.

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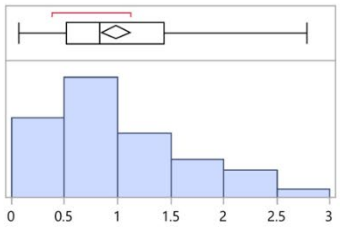
9. Appendix A: ICP Metals Distribution Plots

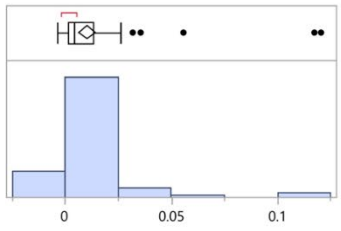
9.1. Aluminum through Cadmium Distributions

Aluminum (Total, ppm)



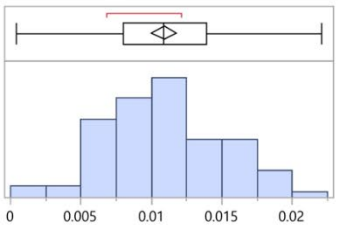
Summary Statistics	
Mean	0.989
Std Dev	0.649
N	89
Minimum	0.0663
Maximum	2.79
Median	0.835
Range	2.72
Interquartile Range	0.921

Aluminum (Dissolved, ppm)



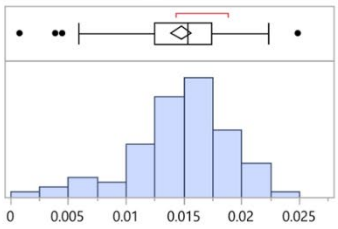
Summary Statistics	
Mean	0.0103
Std Dev	0.0193
N	89
Minimum	-0.0037
Maximum	0.121
Median	0.0045
Range	0.124
Interquartile Range	0.0117

Arsenic (Total, ppm)



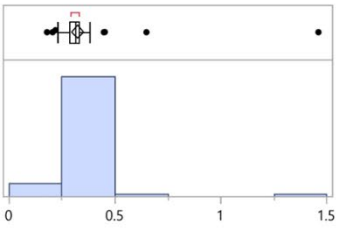
Summary Statistics	
Mean	0.0109
Std Dev	0.00417
N	89
Minimum	0.0004
Maximum	0.0221
Median	0.0109
Range	0.0217
Interquartile Range	0.0059

Arsenic (Dissolved, ppm)



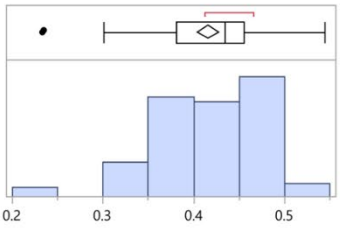
Summary Statistics	
Mean	0.0148
Std Dev	0.00423
N	89
Minimum	0.0008
Maximum	0.0249
Median	0.0153
Range	0.0241
Interquartile Range	0.0049

Boron (Total, ppm)



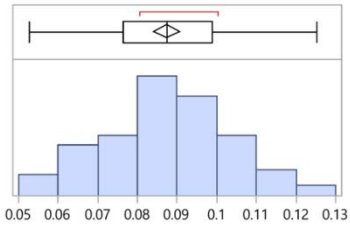
Summary Statistics	
Mean	0.324
Std Dev	0.134
N	89
Minimum	0.182
Maximum	1.46
Median	0.315
Range	1.28
Interquartile Range	0.0424

Boron (Dissolved, ppm)



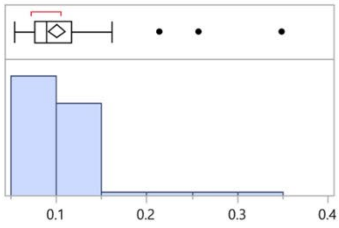
Summary Statistics	
Mean	0.416
Std Dev	0.0567
N	89
Minimum	0.234
Maximum	0.545
Median	0.435
Range	0.311
Interquartile Range	0.0751

Barium (Total, ppm)



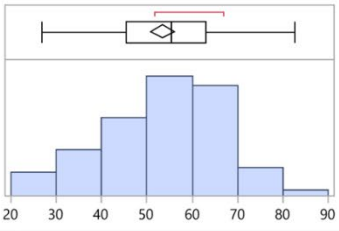
Summary Statistics	
Mean	0.0875
Std Dev	0.0161
N	89
Minimum	0.0526
Maximum	0.125
Median	0.0876
Range	0.0727
Interquartile Range	0.0225

Barium (Dissolved, ppm)



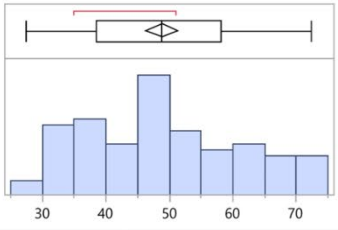
Summary Statistics	
Mean	0.101
Std Dev	0.0412
N	89
Minimum	0.0542
Maximum	0.349
Median	0.09
Range	0.295
Interquartile Range	0.0401

Calcium (Total, ppm)



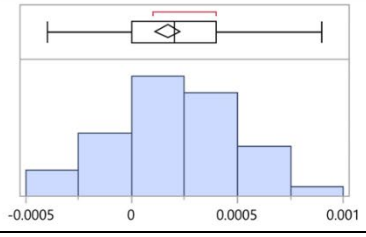
Summary Statistics	
Mean	53.6
Std Dev	12.7
N	89
Minimum	26.8
Maximum	82.6
Median	55.5
Range	55.8
Interquartile Range	17.5

Calcium (Dissolved, ppm)



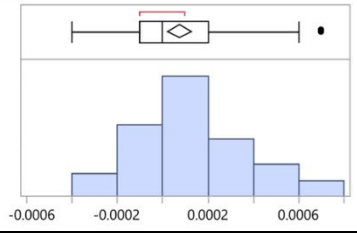
Summary Statistics	
Mean	48.9
Std Dev	12
N	89
Minimum	27.5
Maximum	72.4
Median	48.9
Range	44.9
Interquartile Range	19.7

Cadmium (Total, ppm)



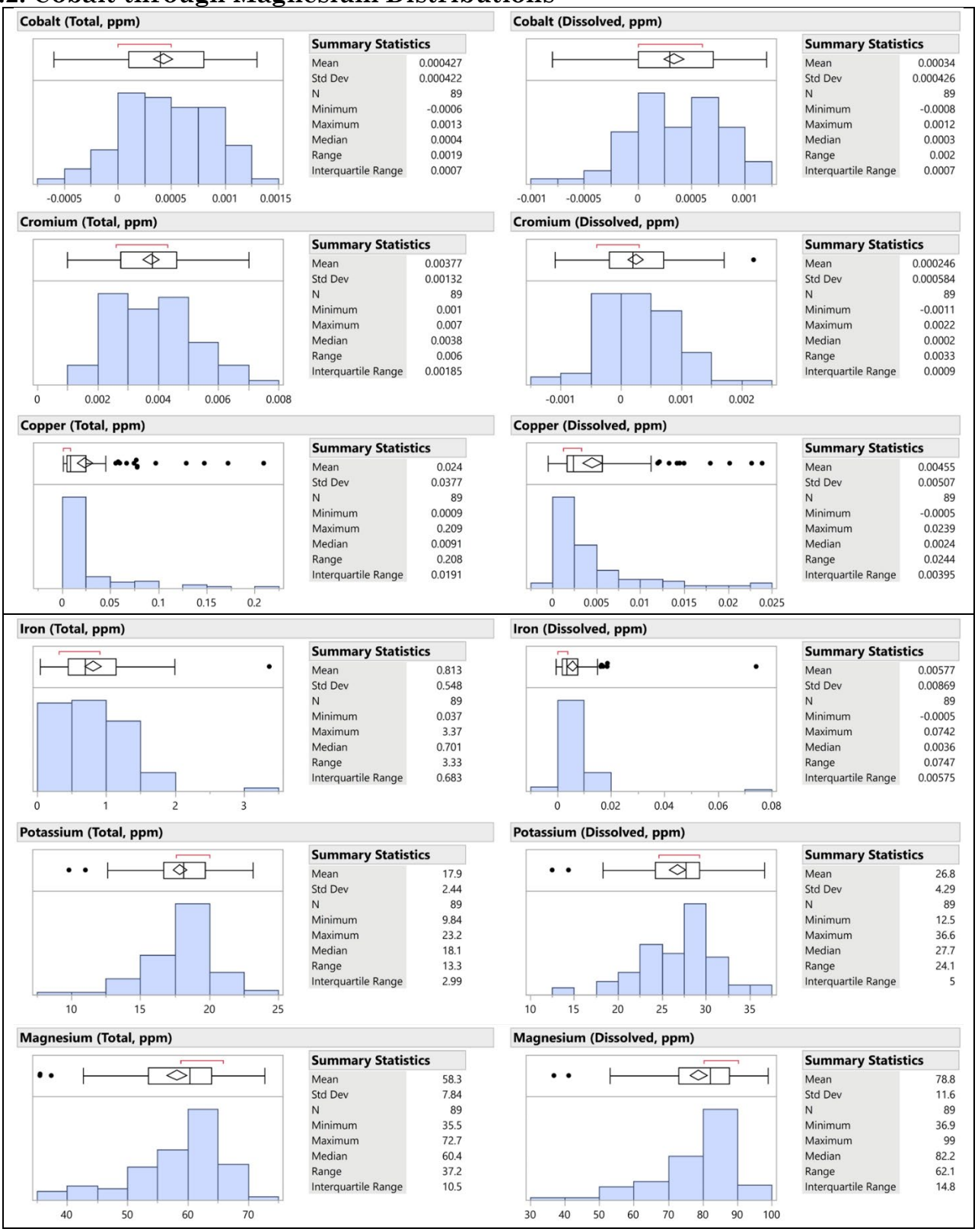
Summary Statistics	
Mean	0.000173
Std Dev	0.000276
N	89
Minimum	-0.0004
Maximum	0.0009
Median	0.0002
Range	0.0013
Interquartile Range	0.0004

Cadmium (Dissolved, ppm)



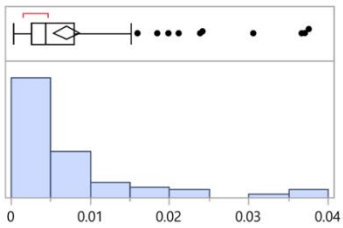
Summary Statistics	
Mean	0.0000764
Std Dev	0.000243
N	89
Minimum	-0.0004
Maximum	0.0007
Median	0
Range	0.0011
Interquartile Range	0.0003

9.2. Cobalt through Magnesium Distributions



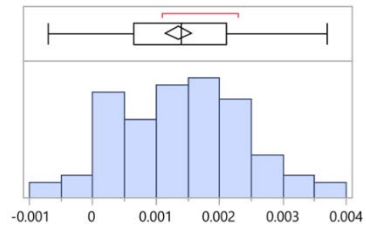
9.3. Nickel through Silicon Distributions

Nickel (Total, ppm)



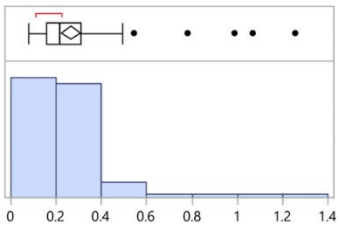
Summary Statistics	
Mean	0.00708
Std Dev	0.00805
N	89
Minimum	0.0003
Maximum	0.0376
Median	0.0043
Range	0.0373
Interquartile Range	0.00535

Nickel (Dissolved, ppm)



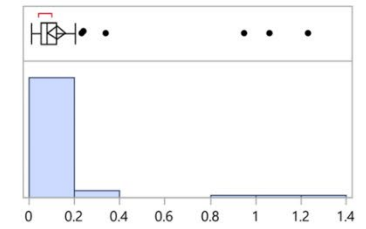
Summary Statistics	
Mean	0.00136
Std Dev	0.000964
N	89
Minimum	-0.0007
Maximum	0.0037
Median	0.0014
Range	0.0044
Interquartile Range	0.00145

Phosphorus (Total, ppm)



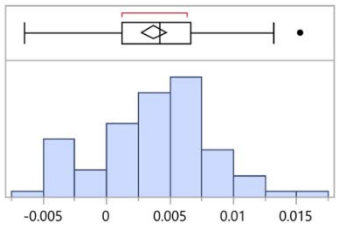
Summary Statistics	
Mean	0.268
Std Dev	0.193
N	89
Minimum	0.0812
Maximum	1.26
Median	0.217
Range	1.18
Interquartile Range	0.151

Phosphorus (Dissolved, ppm)



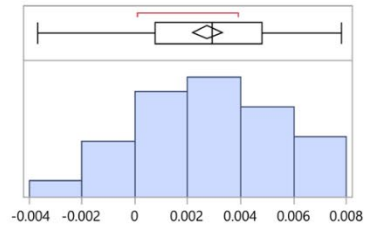
Summary Statistics	
Mean	0.125
Std Dev	0.189
N	89
Minimum	0.0129
Maximum	1.23
Median	0.081
Range	1.22
Interquartile Range	0.0687

Lead (Total, ppm)



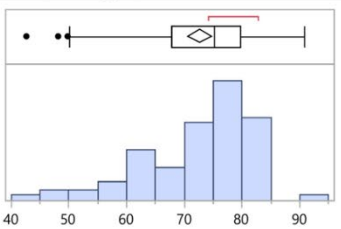
Summary Statistics	
Mean	0.00376
Std Dev	0.00454
N	89
Minimum	-0.0065
Maximum	0.0153
Median	0.0042
Range	0.0218
Interquartile Range	0.0054

Lead (Dissolved, ppm)



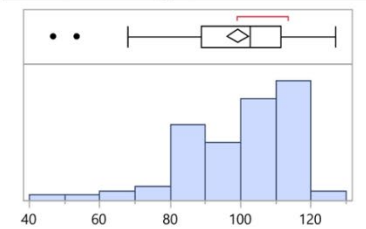
Summary Statistics	
Mean	0.00276
Std Dev	0.00269
N	89
Minimum	-0.0037
Maximum	0.0078
Median	0.0029
Range	0.0115
Interquartile Range	0.00405

Sulfur (Total, ppm)



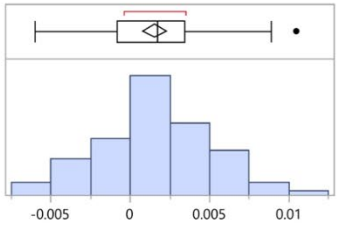
Summary Statistics	
Mean	72.7
Std Dev	9.5
N	89
Minimum	42.7
Maximum	90.9
Median	75.2
Range	48.2
Interquartile Range	11.9

Sulfur (Dissolved, ppm)



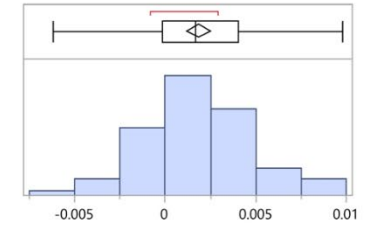
Summary Statistics	
Mean	99.3
Std Dev	15
N	89
Minimum	46.9
Maximum	127
Median	103
Range	79.9
Interquartile Range	22.3

Selenium (Total, ppm)



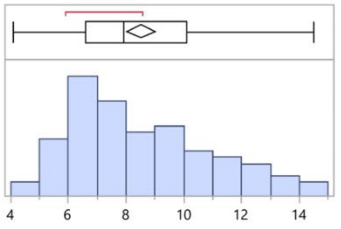
Summary Statistics	
Mean	0.00159
Std Dev	0.0036
N	89
Minimum	-0.006
Maximum	0.0105
Median	0.0017
Range	0.0165
Interquartile Range	0.0042

Selenium (Dissolved, ppm)



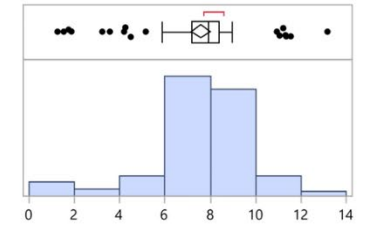
Summary Statistics	
Mean	0.00187
Std Dev	0.00303
N	89
Minimum	-0.0062
Maximum	0.0098
Median	0.0017
Range	0.016
Interquartile Range	0.00415

Silicon (Total, ppm)



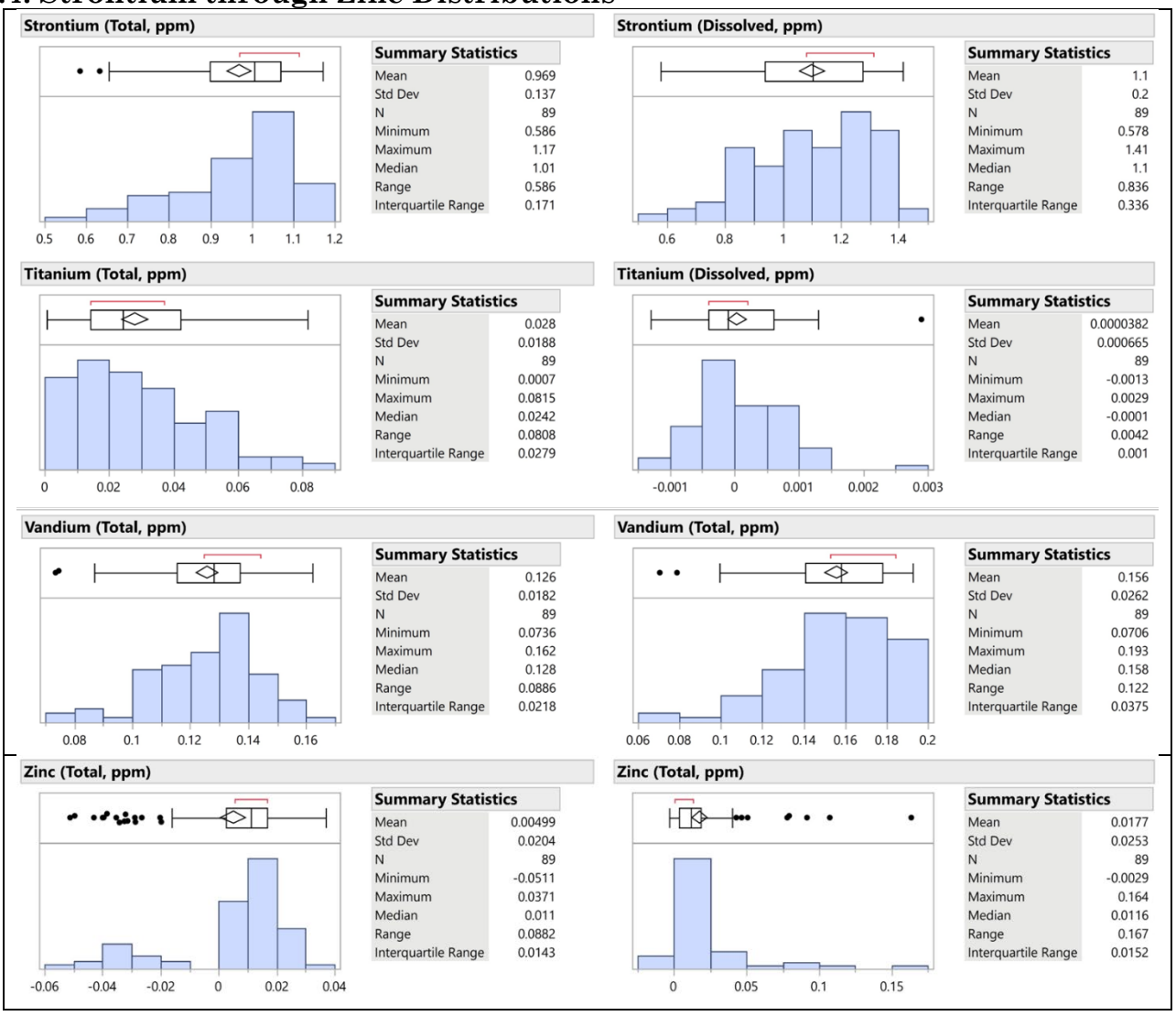
Summary Statistics	
Mean	8.52
Std Dev	2.34
N	89
Minimum	4.11
Maximum	14.5
Median	7.94
Range	10.4
Interquartile Range	3.5

Silicon (Dissolved, ppm)



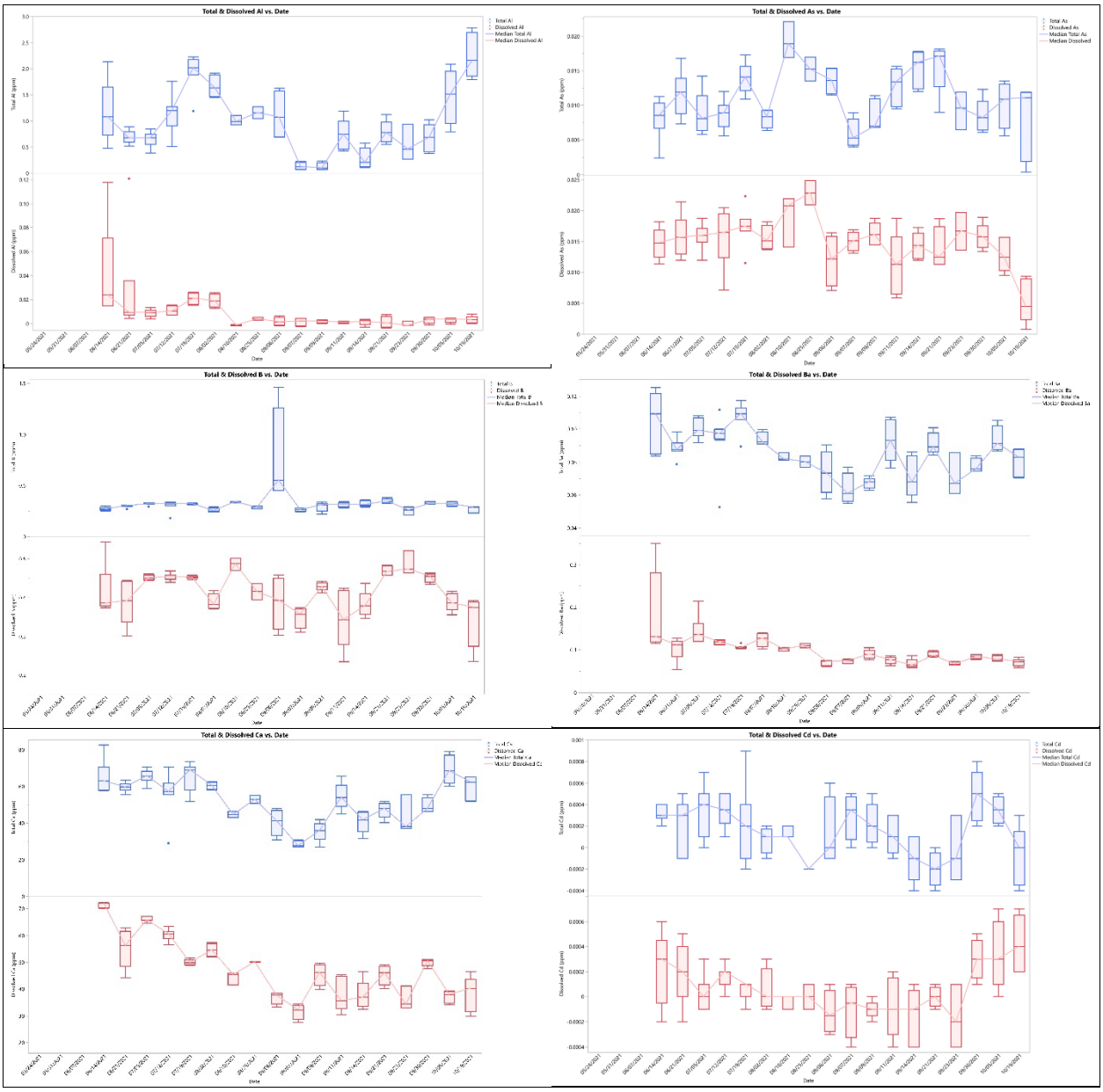
Summary Statistics	
Mean	7.59
Std Dev	2.03
N	89
Minimum	1.29
Maximum	13.2
Median	7.93
Range	11.9
Interquartile Range	1.21

9.4. Strontium through Zinc Distributions

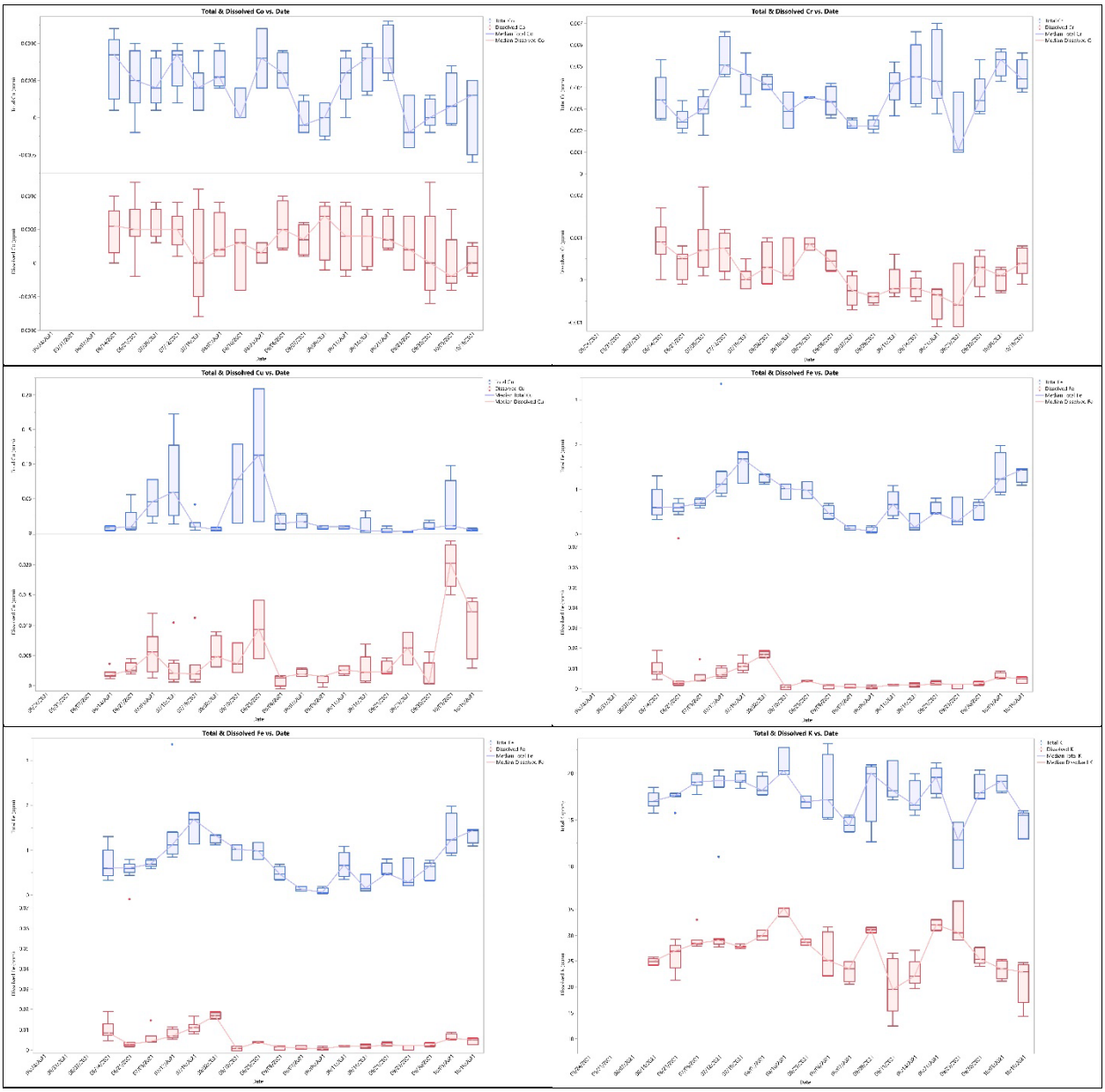


10. Appendix B: ICP Metals Time Series Box Plots

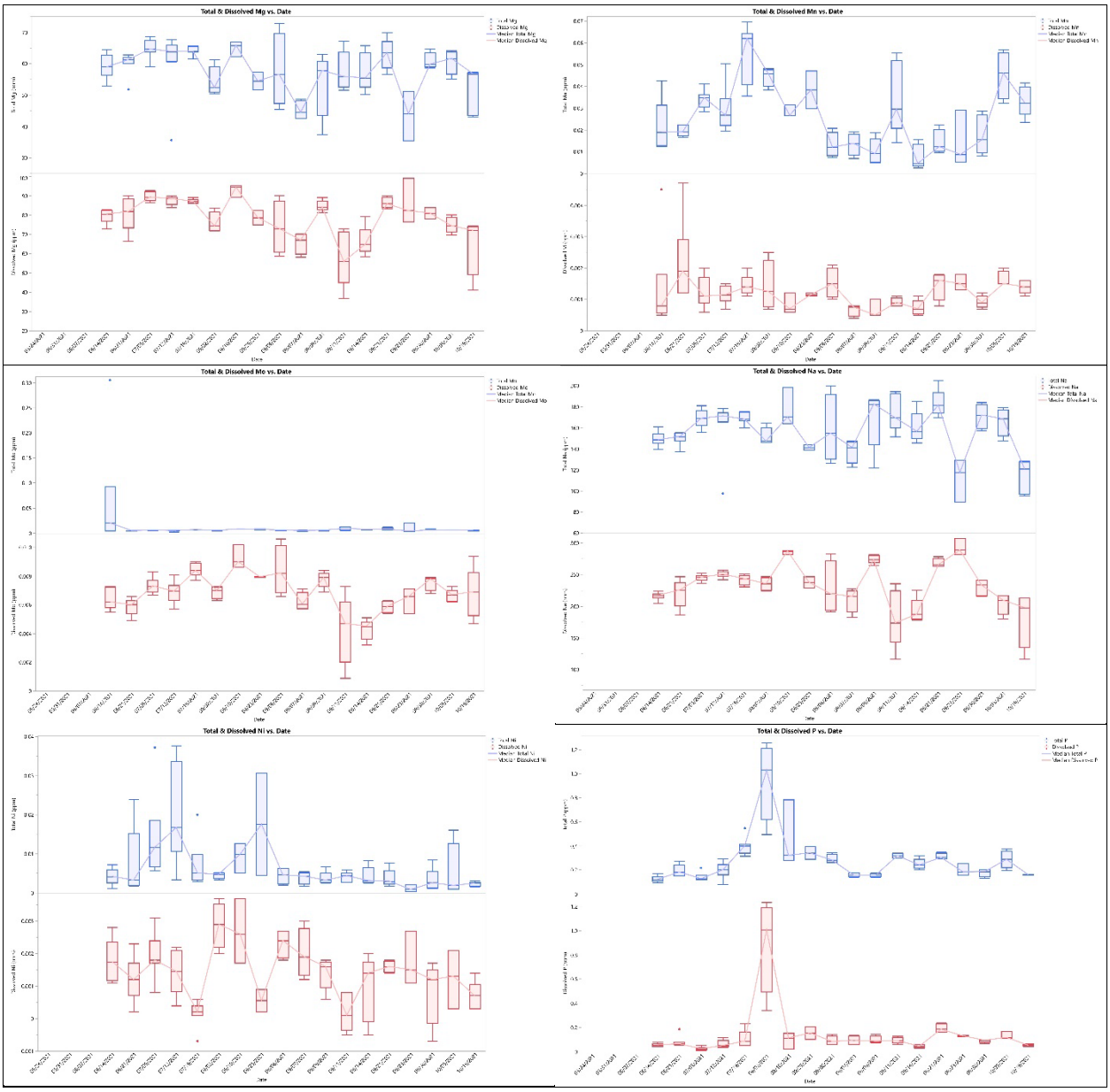
10.1. Time Series Plots – Aluminum through Cadmium, Total (blue), dissolved (red)



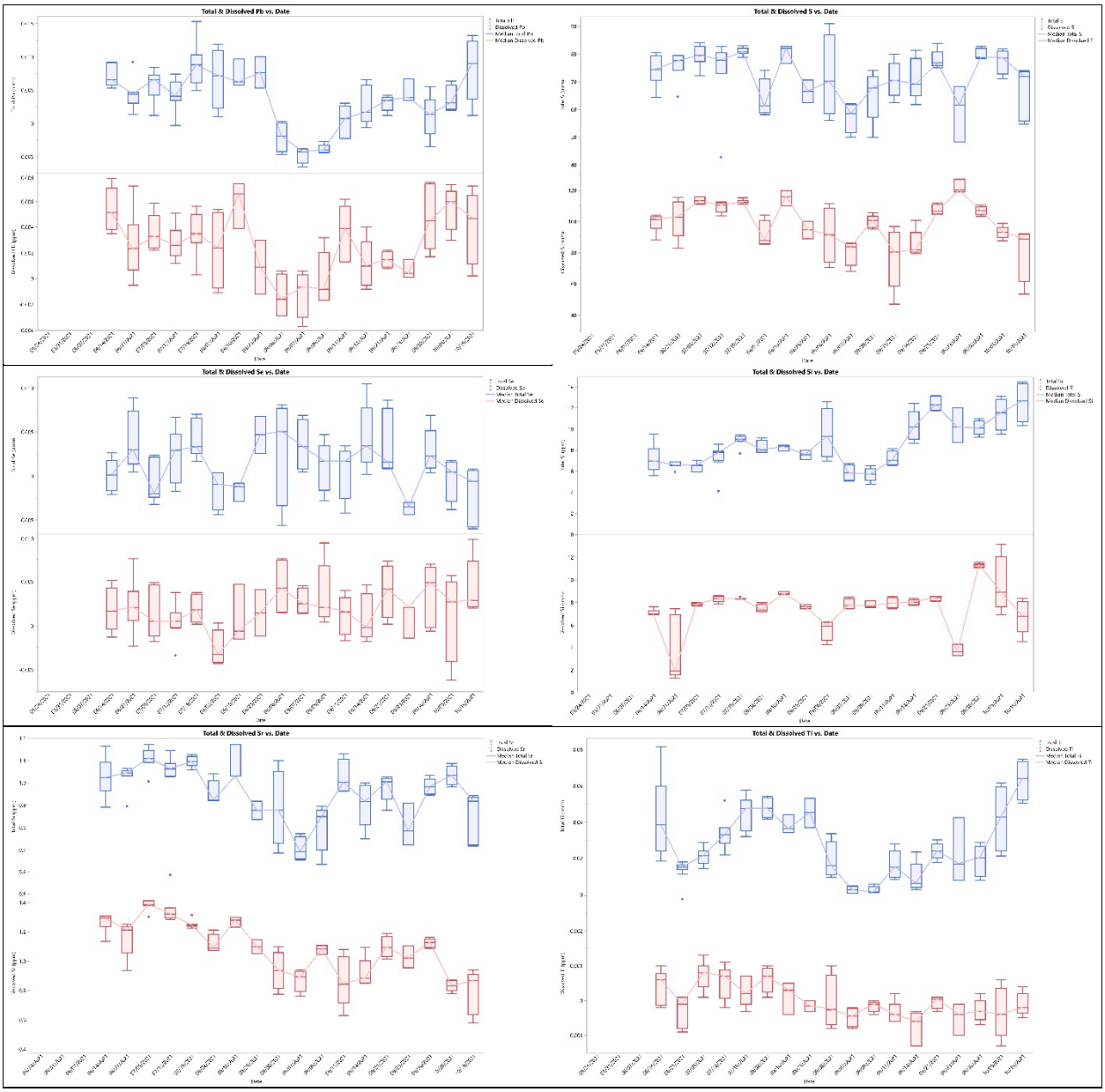
10.2. Time Series Plots – Cobalt through Potassium, Total (blue), dissolved (red)



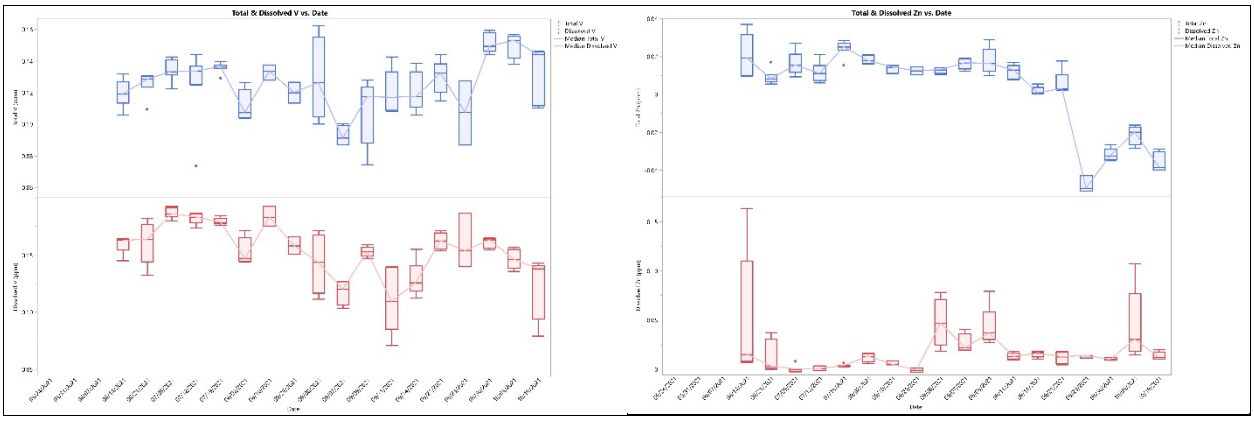
10.3. Time Series Plots – Magnesium through Phosphorous, Total (blue), dissolved (red)



10.4. Time Series Plots – Lead through Titanium, Total (blue), dissolved (red)



10.5. Time Series Plots – Vanadium through Zinc, Total (blue), dissolved (red)



11. Appendix C: Sample Totals

TSSD

Parameter	# of data points
Nitrite (NO2-N)	124
Nitrate (NO3-N)	124
Ammonia (NH4-N)	124
Unfiltered Reactive P (PO4-P)	124
Filtered Reactive P (PO4-P)	102
Total P (P)	116
Total Nitrogen	16
COD	95
TSS	95
VSS	95
TDS	95
TOC	56

YSI Sondes

Parameter	# of data points
Barometer	9546
Conductivity (µS/cm)	9546
Sp Conductivity (µS/cm)	9546
Sal (psu)	9546
nLFCond (µS/cm)	9546
TDS (mg/L)	9546
Temp (F)	9546
Resistivity (ohms-cm)	9546
Sigma-T (s t)	9546
Sigma (s)	9546
Chlorophyll (RFU)	5229
Chlorophyll (µg/L)	5229
PC (RFU)	5229
PC (µg/L)	5229
Turbidity (FNU)	3466
TSS (mg/L)	3466
ODO (% Saturation)	5229
ODO (mg/L)	5229
Pressure (psi a)	9546
pH	3084
pH (mV)	3084
ORP (mV)	3847

BYU Environmental Analytical Lab

Parameter	# of data points
Al1670-Digested	169
Al1670-Filtered	169
As1890-Digested	169
As1890-Filtered	169
B_2497-Digested	169
B_2497-Filtered	169
Ba4554-Digested	169
Ba4554-Filtered	169
Ca3179-Digested	169
Ca3179-Filtered	169
Cd2265-Digested	169
Cd2265-Filtered	169
Co2286-Digested	169
Co2286-Filtered	169
Cr2835-Digested	169
Cr2835-Filtered	169
Cu3247-Digested	169
Cu3247-Filtered	169
Fe2382-Digested	169
Fe2382-Filtered	169
K_7664-Digested	169
K_7664-Filtered	169
Mg2852-Digested	169
Mg2852-Filtered	169
Mn2576-Digested	169
Mn2576-Filtered	169

Parameter	# of data points
Mo2020-Digested	169
Mo2020-Filtered	169
Na5895-Digested	169
Na5895-Filtered	169
Ni2316-Digested	169
Ni2316-Filtered	169
P-1782-Digested	169
P-1782-Filtered	169
Pb2203-Digested	169
Pb2203-Filtered	169
S-1820-Digested	169
S-1820-Filtered	169
Se1960-Digested	169
Se1960-Filtered	169
Si2516-Digested	169
Si2516-Filtered	169
Sr4077-Digested	169
Sr4077-Filtered	169
Ti3088-Digested	169
Ti3088-Filtered	169
V_3093-Digested	169
V_3093-Filtered	169
Zn2138-Digested	169
Zn2138-Filtered	169
PO4-P (Lachate)	16

BYU Geology Lab (IC)

Parameter	# of data points
Chloride	29
Nitrate	29
Fluoride	29
Sulfate	29
Phosphate	29

163,104+ datapoints on Utah Lake 05/25-09/27

12. Appendix D: Checklists

12.1. Field Checklists

12.1.1. Anafi Drone Flight

Shallow Water Corrals

- Take Photo of all 5 corrals in 1 shot
- Drone should be around 170 ft above the water
- Include the Buoys in the shot
- Make sure the camera is at 90 Degrees (aimed straight down)
- Make sure that the camera is facing North (towards TSSD)
- Make sure that the camera is perpendicular to the corrals. We want the photo to show a straight horizontal line NOT a diagonal line.
- Take both visual and thermal image
- Take Individual Photos of Each Corral
- Start with corral 1 then corral 2,3 etc…
- Drone should be about 35-40 ft in elevation
- Make sure drone is facing North when you take the individual photos
- Take both visual and thermal photos

Deep Water Corral

- Take Photo of All Corrals in One Shot
  - Drone should be at about 170 ft above the water
  - Include Buoys in the picture
  - Take both visual and Thermal photos
  - Face East (towards BYU)
  - Make sure the camera is at 90 Degrees
  - The Corrals should be in a horizontal line in the photo NOT diagonal (see picture in 1. Shallow water corrals).
- Take Individual Photos of Each Corral
  - Start with corral 6 then corral 7,8, etc ….
  - Face East (BYU)
  - Make sure the camera is at 90 Degrees
  - Take both visual and Thermal Photos

Effluent

- Take photos of the effluent. Fit in as much as possible.
- Camera should be at 90 Degrees
- Follow the effluent tail if you can
- We can stitch pictures together later
- Look for the effluent hitting the corrals. Take pictures if applicable.

Scenic Photos

- Scenic photos are always great!

Working Photos

- Take pictures of the people in the skiff especially when they are taking samples with probes, dip sampler etc.

12.1.2. Lake Water Sampling and Data Collection SOP

- Collect 250 or 125mL per sample
- Do a bottle blank

- Triple rinse amber transfer bottle used to collect chl-a sample with site water on the opposite side of the boat from where samples will be taken
- Chl-a samples should be place on ice in the dark immediately while on the lake between sites and the shore before processing
- Do not take samples near the boat anchor or near the boat motor.

Supplies needed:

- Copy of this SOP
- Boat or floaties
- Chl-a sampling kit
- Depth finder
- Secchi disk
- Labeled sample bottles
- Clean 2000 mL amber bottles for collecting Chl-a water samples to be filtered
- Clean or triple-rinsed ½ gallon "transfer bottles" for collecting constituents to be filtered
- DI water
- Cooler with ice
- Lab sheets/field notebook
- pens/markers
- Sample bottle labels: current date/time, corral number, sampler's initials
- Take an equipment blank that you treat exactly the same as the other samples except you fill it with DI water instead of lake water

Sample types:

- Nitrate
  - Collect in a clean plastic container and analyze the day of collection, store at 4C if sample is to be analyzed within 24 to 48 hours (TSSD SOP)
- Nitrite
  - Collect in clean plastic container and analyze the day of collection, store at 4C if sample is to be analyzed within 24 to 48 hours
- Ammonia
  - Collect in a clean plastic container and analyze the day of collection
  - If not analyzed day of collection, preserve with 2mL HCL and refrigerate at 4C until analysis. Must be analyzed within 28 days of sampling and brought to room temperature and neutralized to pH 7.0 before analysis.
- DO
  - Collect in 500mL plastic container and brought directly to laboratory
- TDS
  - Collect in clean plastic container, refrigerate at 4C until analysis, analyze within 7 days of sampling
- TKN
  - Analyze within the day sampled or preserve samples with 2mL sulfuric acid, store at 4C and analyze within 28 days. Before analysis, neutralize preserved samples with 5N NaOH and warm to room temperature.
- Total Phosphorus
  - Collect in clean plastic containers and analyze the day of collection.

  - For samples to be analyzed for total phosphorus: preserve with 2mL sulfuric acid, store at 4C and analyze within 28 days. Before analysis, neutralize preserved samples with 5N NaOH and warm to room temperature
  - For samples to be analyzed for reactive phosphorus: store at 4C and analyze within 48 hours.
- Total and Volatile SS
  - Collect in clean plastic containers and refrigerate at 4C until time of analysis. Analyze within 7 days of sampling, bring to room temp before analysis.

12.1.3. Post-Sampling Checklist

Equipment

- Clean probes
- Check handheld battery level, if below 50% put on charger
- Empty cooler and put ice back in freezer
- Charge drone batteries and controller if necessary (make sure drone batteries don't stay on the charger for more than 3 hours!)

Data Entry

Water Samples

- Fill out the Sample Tracking and Metadata sheet
- Fill out any relevant Chain of Custody forms (if you dropped samples off at a lab):
- TSSD
- Secchi Disk Readings
- Environmental Analytical Lab
- Environmental Engineering Lab
- Geology Lab

Probe Data

- Upload probe data just from that day to the Google Drive using the Probe Upload Instructions

Images

- Upload all drone and GoPro images to "ByDate" folder in "Project Photos" folder on the shared J drive
- Copy any gridded flight photos and corral timelapse photos to the designated folders on the J drive
- Tag pictures after uploading using the picture tagging guide

Miscellaneous (Everyone)

- Update Research Journal
- Fill out Field Logbook

12.1.4. Probe Sampling

- Sources:
  - DWQ Calibration, Maintenance, and Use of YSI Multiprobes SOP
  - DWQ Calibration, Maintenance, and Use of Multiparameter Water Quality Sondes SOP
- Calibrate at least once daily during use (G SOP)

- Maintain on a regular maintenance schedule and on an as-needed basis
- Reschedule or stop sampling if conditions are unsafe
- ALWAYS keep the sensor guard on during use
- The sonde should always be stored wet--if it will be a short period of time (a day or less), fill the calibration cup with no more than a ½ inch of tap water. For a storage period longer than a day, fill it with no more than ½ inch of pH4 buffer solution.
- Make sure it is stored carefully for transport
- Sondes should not be exposed to temperatures below 33F or above 122F(so don't leave in a hot car!)
- Don't lower sondes into bottom sediments and collect readings after any disturbed sediment has been cleared by the current or settled.
- Supplies Needed:
    - Copy of this SOP (can be on your phone)
    - Copy of project-specific SAP (on your phone)
    - Sonde
    - Charging cables
    - Maintenance tool kit
    - Tap water
    - DI water
    - pH calibration standard solutions (7 and 10)
    - Lint-free cloth (kimwipes)
    - Handheld
    - Field laptop
- Before trip:
    - Review the SAP to confirm sampling locations and target conditions
    - Charge the handhelds and field laptop
    - Calibrate in the morning before sampling and anytime there is a reading that's unusual
- At the sampling site:
    - Remove calibration cup
    - Lower the sonde into the water UPSTREAM of any other sampling activity
    - Avoid laying the sonde in bottom sediments
    - Observe the water quality readings displayed on the handheld to make sure they're reasonable
    - Record the readings on the Trip Sheet to use as back up and for QA
- After sampling, rinse the sonde with clean tap water and store the sensors in tap water or pH4 buffer. Do NOT store in DI water, do NOT allow sensors to dry out, and do NOT allow storage medium to freeze around the sensors.

- After Sampling Trip
    - Download data from PDR
    - Format the parameters into the DWQ hydrofile format
    - Verify that values, site IDs, and sample date are accurate
- Weekly Maintenance:
    - Clean sondes with soap and water. Clean casing with a sponge and sensors with an extra soft toothbrush and cotton swabs. Clean calibration cups and sensor guards with soap and water.
- Monthly Maintenance

- Soak sensors and sensor guards in vinegar for 20 minutes if hard water stains are present. Clean the outside of all sensors with isopropyl alcohol, except the LDO sensor, which should be cleaned only with soap.

12.1.5. Sampling Trip Supply Checklist

- Water Sampling
  - Backpacks(probes)
  - DI Water (Squeeze Bottle)
  - Extra DI Water
  - Tap Water (Squeeze Bottle)
  - Dip Sampler (Confirm that it went through acid bath)
  - GoPro (Check batteries)
  - Drones (Check batteries)
  - Sample Bottles (Get as many as there are corrals, plus four)
  - Wagon
  - Ice
  - Cooler
- Vertical Profile/Data Tracks
  - Backpacks (Probes)

12.2. Lab Checklists

12.2.1. EAL Procedures

Testing Session Checklist

- Do ICP
- Do SRP
- Clean

Test-Specific Checklists

Microwave Digestion

- Setup
  - Put on gloves and a lab coat.
  - Lay down some paper towels on the lab bench, and put out the sample bottles.
  - Set out a numbered safety shield (brown) for each sample, plus two more for the water standard and process blank. #1 has to be the water standard, #4 is broken, and #9 is missing.
  - Place a white teflon tube inside each of the safety shields.
  - Tube #1 is the water standard. Pipette 20 mL of ultrapure water (0.10 ppm) water into tube #1.
  - Shake the sample vigorously, and pipette 20 mL of sample into their respective tubes. Use a different pipette tip for each sample. On the data sheet, record what sample or standard or blank each tube number contains. At this point, there should be one empty tube for the process blank.
  - Working under the fume hood, pour some 69.6% nitric acid (in the cabinet under the fume hood) into a beaker. Place the tubes in the fume hood, and, using a new pipette tip, pipette 3 mL of nitric acid into each tube. Pour any unused nitric acid back in the container, and put it back in the cabinet. Triple rinse the beaker with DI water.
  - DO NOT DO THIS STEP UNLESS OTHERWISE TOLD: Get the pipette with 50% hydrogen peroxide from the outside fridge, set it to 1 mL, and using a little beaker test to check that it's working. Then add 1 mL of the hydrogen peroxide to each tube. Dump the hydrogen peroxide from the beaker into the aqueous waste bottle under the fume hood.

- On the lab bench, stretch out the white Teflon caps using the reshaping tool, then put a Teflon cap on each of the tubes. Make sure they fit well.
- Place a safety valve (brown lid) on each of the tubes except #1. As you tighten the lid, the little thing will pop out.
- Put the thermometer cover thing (thermowell?) through the Teflon cap in Tube #1, the water standard. Tube #1 has a special cap that has a hole all the way through, look on the underside. Take the cover off the thermometer, slide it through that cap, and put a the washer on. Then put the thermometer and cap into Tube #1. Slide the ATC sensor through the safety valve (make sure the valve does not have a TFM foil in it). Slide the ATC sensor through a TFM foil. Place the sensor into the thermowell and tighten the safety valve.
- Open the microwave, take out the turntable, and put Tube #1 into the #1 spot on the turntable. Put the rest of the tubes on the outside ring of the turntable. The order doesn't really matter for the rest, but make sure the weight is balanced around the turntable. Rotate the tubes such that the white hole on the sides of the tubes is turned outward (gas and stuff). If you have any tubes on the inside ring, turn those white holes towards the center of the carousel.
- Put the top of the turntable on top of the tubes. If it doesn't want to pop on, rotate the tubes around a bit.
- Attach the thermometer end to the microwave.
- Put the turntable back in the microwave and wiggle it around a bit until you feel it lock into place.
- To start the microwave, flip the switch, put in the passcode ("123"). Then hit some buttons. Once it starts, look in the microwave to make sure the temperature cord isn't kinking.
- Come back in 1.5 hours!

Sample Transfer

- Collect the following items:
  - Purple centrifuge rack
  - Small distilled water wash bottle
  - 2 L 10% Nitric acid bath
  - Pressure release tool
  - Vessel removing tool
  - Washable marker
- Retrieve the carousel containing samples from the microwave digester and place in the fume hood.
- Number the centrifuge tubes using the washable marker.
- Working inside the hood, use the pressure relief tool to release excess pressure from the safety valve.
- Carefully remove the safety valve from the first brown safety shield.
- Using the vessel removing tool, pop the vessel out of the shield by lining up one of the two holes in the bottom of the shield with the two projections in the tool and pressing down.
- Remove the white Teflon cap from the vessel.
- Using distilled water, quantitatively transfer the contents of the vessel to the corresponding centrifuge tube.
- Dilute the sample up to the 25 ml mark on the centrifuge tube, also mark the dilution on the sample sheet.
- Cap the tube and invert it 3 times. Repeat with remaining samples.

Analysis:

- Run samples on ICP-OES under the 'Phosphorus' method (iCAP 7400, Thermo Scientific, Madison, WI).

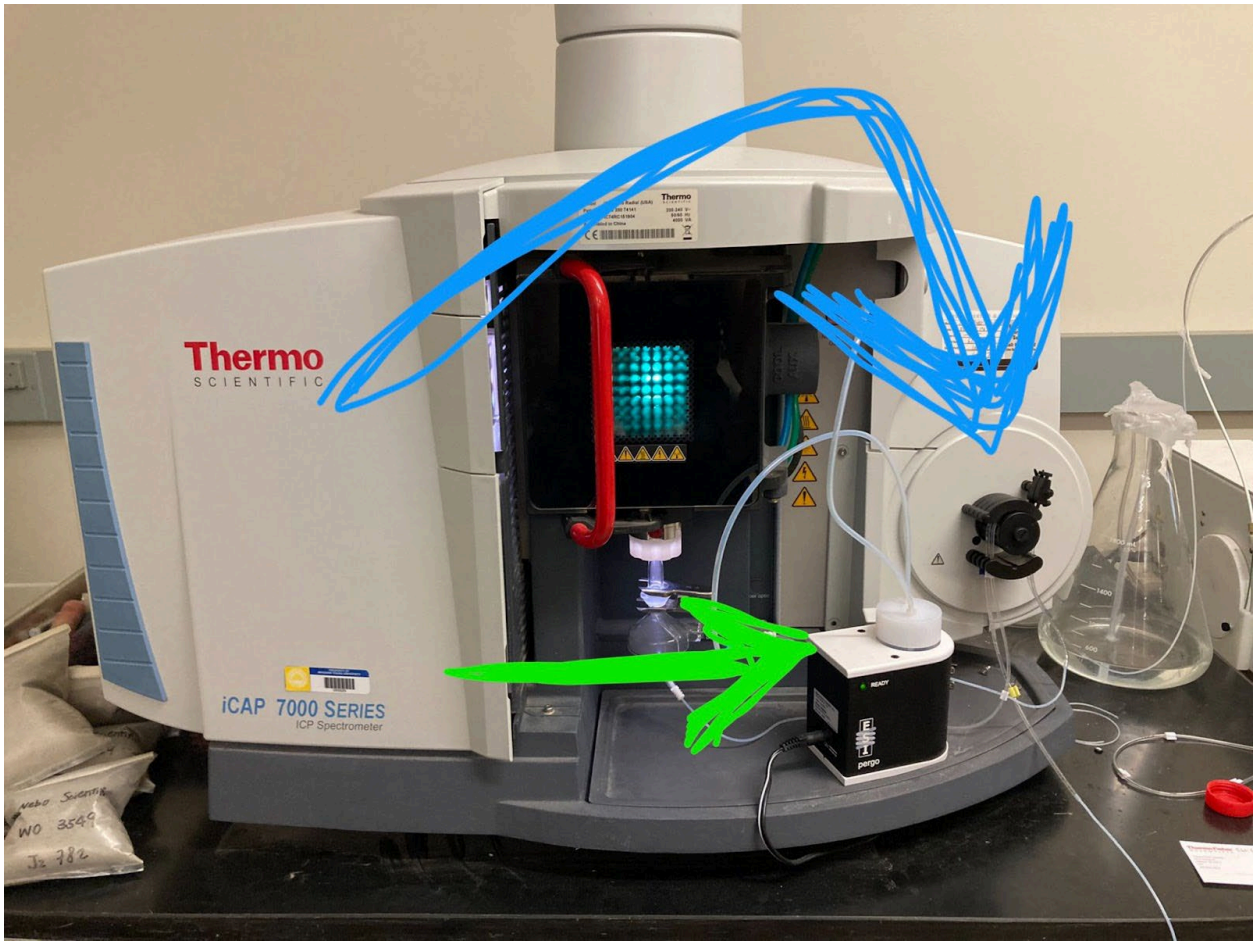
Clean up:

- Place white Teflon vessels and caps in a 10% Nitric acid bath (let sit for 30 minutes- 1 hour).
- Rinse the white Teflon vessels and caps with DI water.
- Rinse safety shields and safety valves with DI water be sure to tap additional water out of safety valves.
- Return sample carousel to the microwave digester.

ICP (Inductive Couple Plasma)

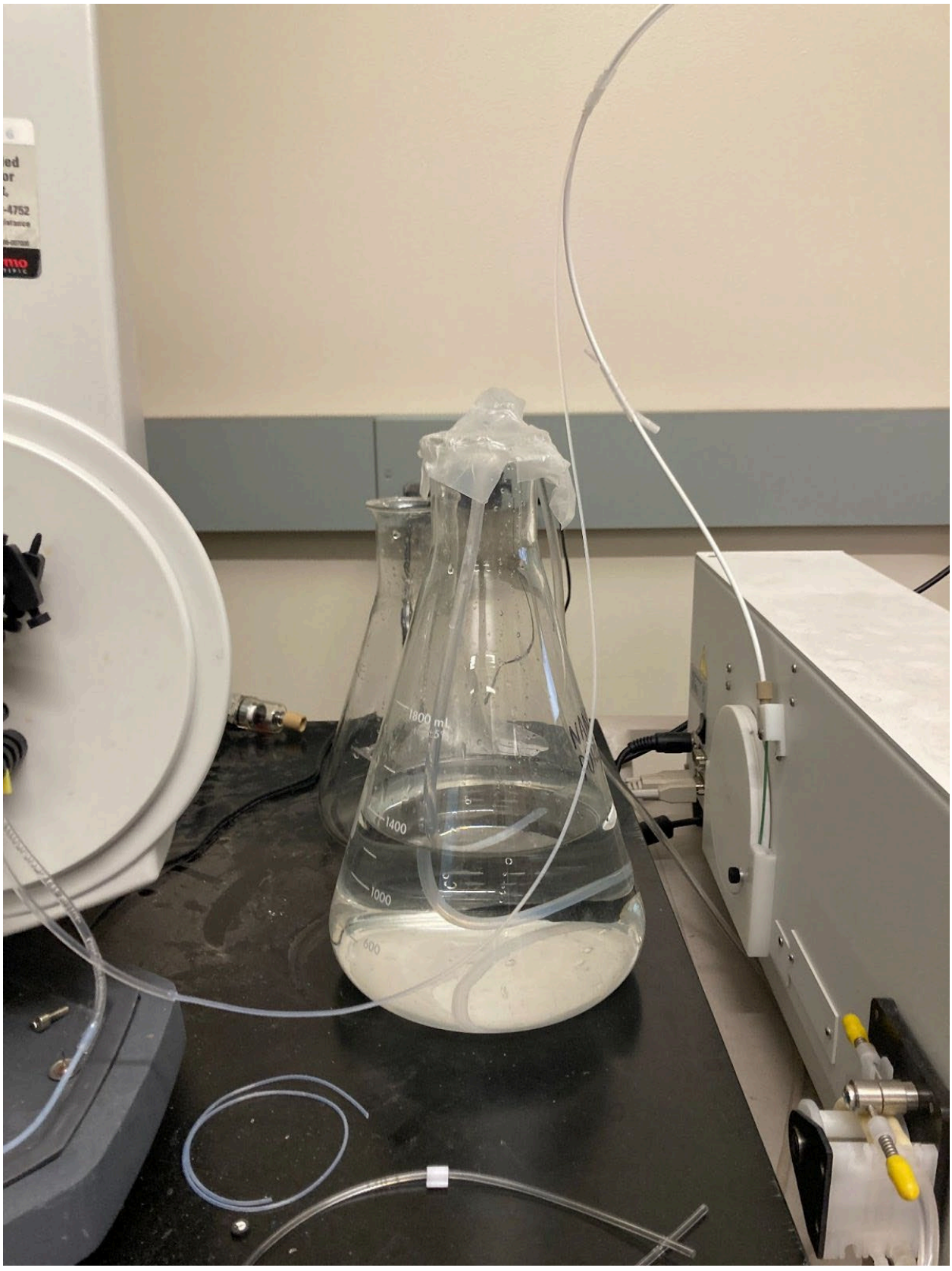
- Running the Machine
- **Before beginning:**
  - Turn on cooler (it's really important to protect the detector) (yellow arrow)

- Check water level in argon humidifier and turn it on (should be between min and max lines) (green arrow)

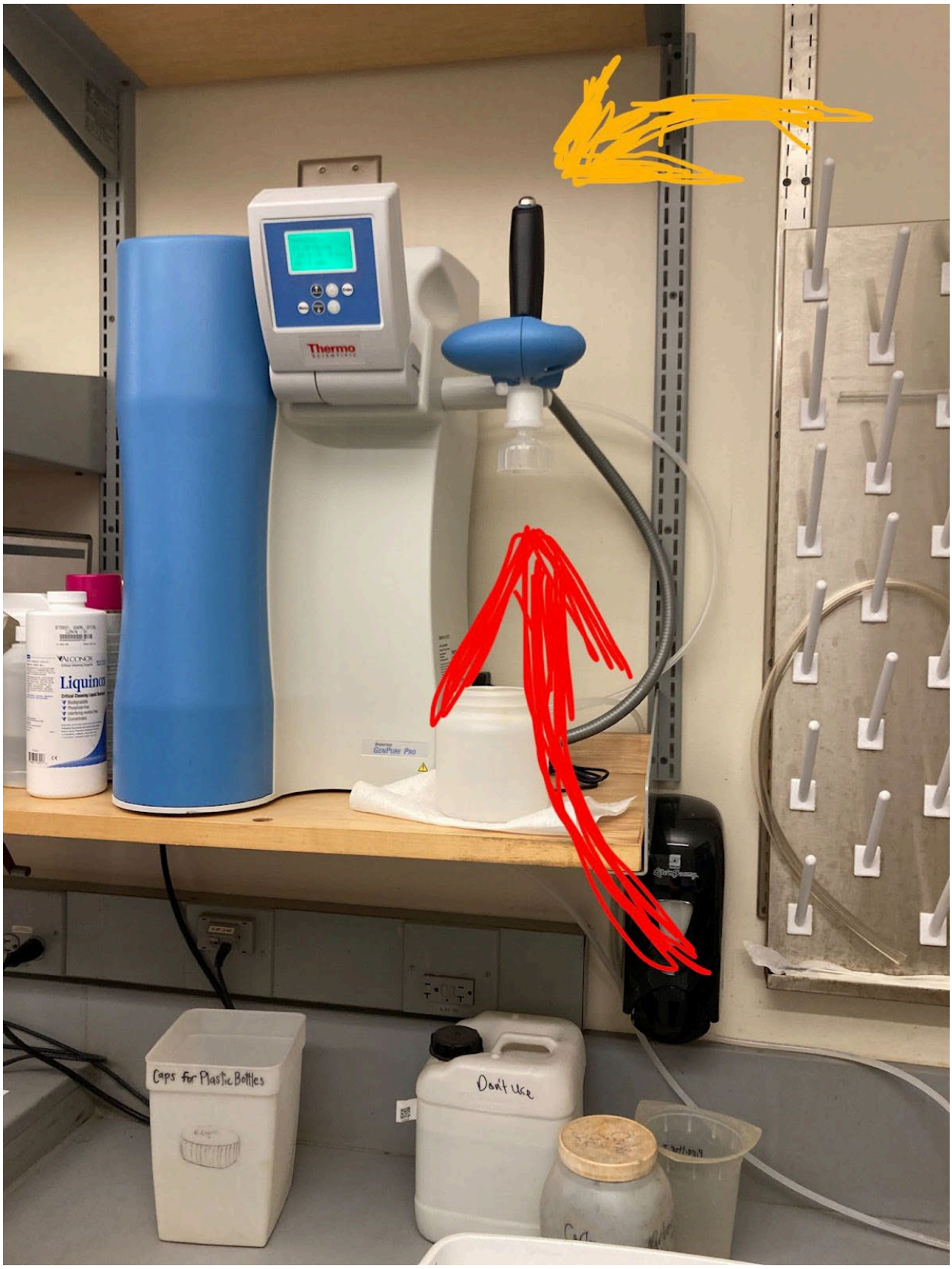


- Check pressure in Argon tank (regulator should be set to between 90-100 PSI)

- Open pressure builder if necessary
- Make sure there is a good amount of nanopure water in the large beaker to the right of the ICP machine (reservoir for the autosampler)


1800 mL
1400
1000
600

- Obtain nanopure water from the machine by the sink at the back of the main lab - press “nonstop” right below the display, then hold the thing that looks vaguely like a gasoline dispenser (red arrow) over the sink before pressing the button (orange arrow) on the handle twice to start the water flow. Use another beaker to get the water and pour it into the reservoir beaker.

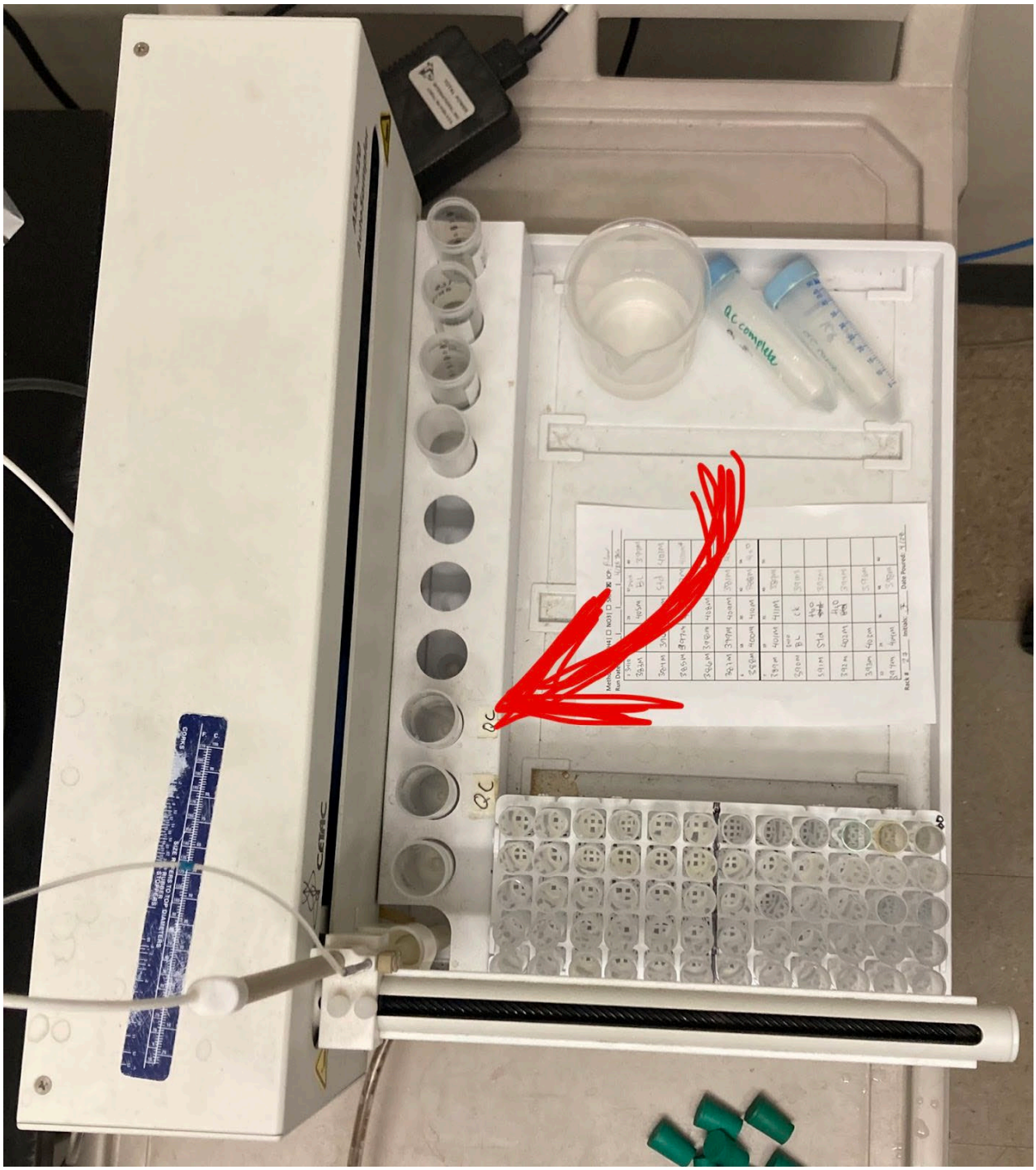


- **Selecting a method:**

- If the computer is off, start the computer.
- Type in "admin" for user name. There is no password.
- On the iTEVA screen, click the "analyst" icon.
- Select method
- If you are running multiple methods, you can add another session with a different method.
- Click "OK"

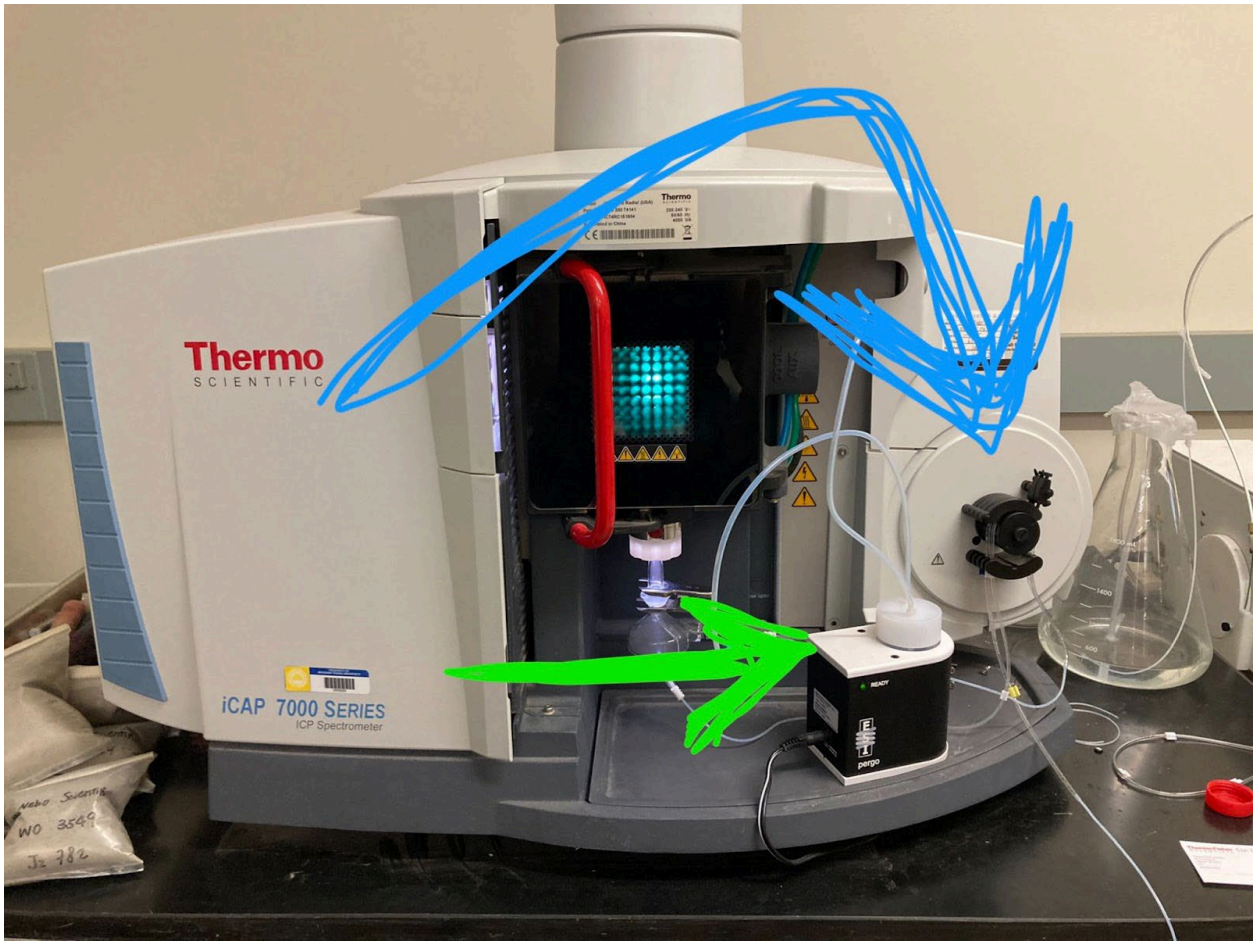
- **Running/Mapping samples using the auto sampler:**
  - *Refer to the instruction sheet next to the computer.*
  - Click on "Sequ" at the bottom left hand of the screen.
  - Click on "auto-session" in the top menu bar, then click, "New auto-sampler…".
  - Check "Ignite plasma" >> Set the plasma warmup for 30 minutes and set it to "shutdown plasma" automatically at the end of the set.
  - To enter samples, click "new"
  - Right click on the map ("Untitled…") and click "add sequence".
  - If you are running multiple methods, repeat the running/mapping samples procedure starting from this step to add another sequence.
  - Enter the number of samples, including checks, standard samples, and two water samples.
  - Put a low check after the last sample, followed by two water blanks.
  - If you need to adjust the sample list later, you can add or remove samples.
  - The instrument will automatically run the QC standard every 15 or so samples (depending on the method)
  - Enter the first sample number in the "Sample Name" box, taking into account the checks and standards. For example, if your first sample is #235 and you have one check and one standard, enter number 233 in the sample name box.
  - The program will auto-increment this number.
  - Click "OK".
  - Click "OK" again, a diagram of the autosampler will appear.
  - Click on "Untitled (CETAC ASX-520 5X12 by Col Racks)" followed by a right click on the same icon.
  - Click "auto-locate all".
  - Standards and sample positions are shown. Make certain they are correct:
    - S-1 position is a blank, be sure this is always nanopure water straight from the filtration apparatus by the back sink in the main lab area.
    - S-2 position is the high standard
  - S-3 & S-4 positions are QC standards. Use the high standard as the QC standard
  - The standards go in the large tubes at the top of the rack (red arrow). Obtain the special containers for the standards from the tub on the shelves by the main entrance to the lab (orange arrow) - make sure that you only use tubes that say, "ICP only" NOT "FIA only".
  - Standards can be found on the shelf by the computer.

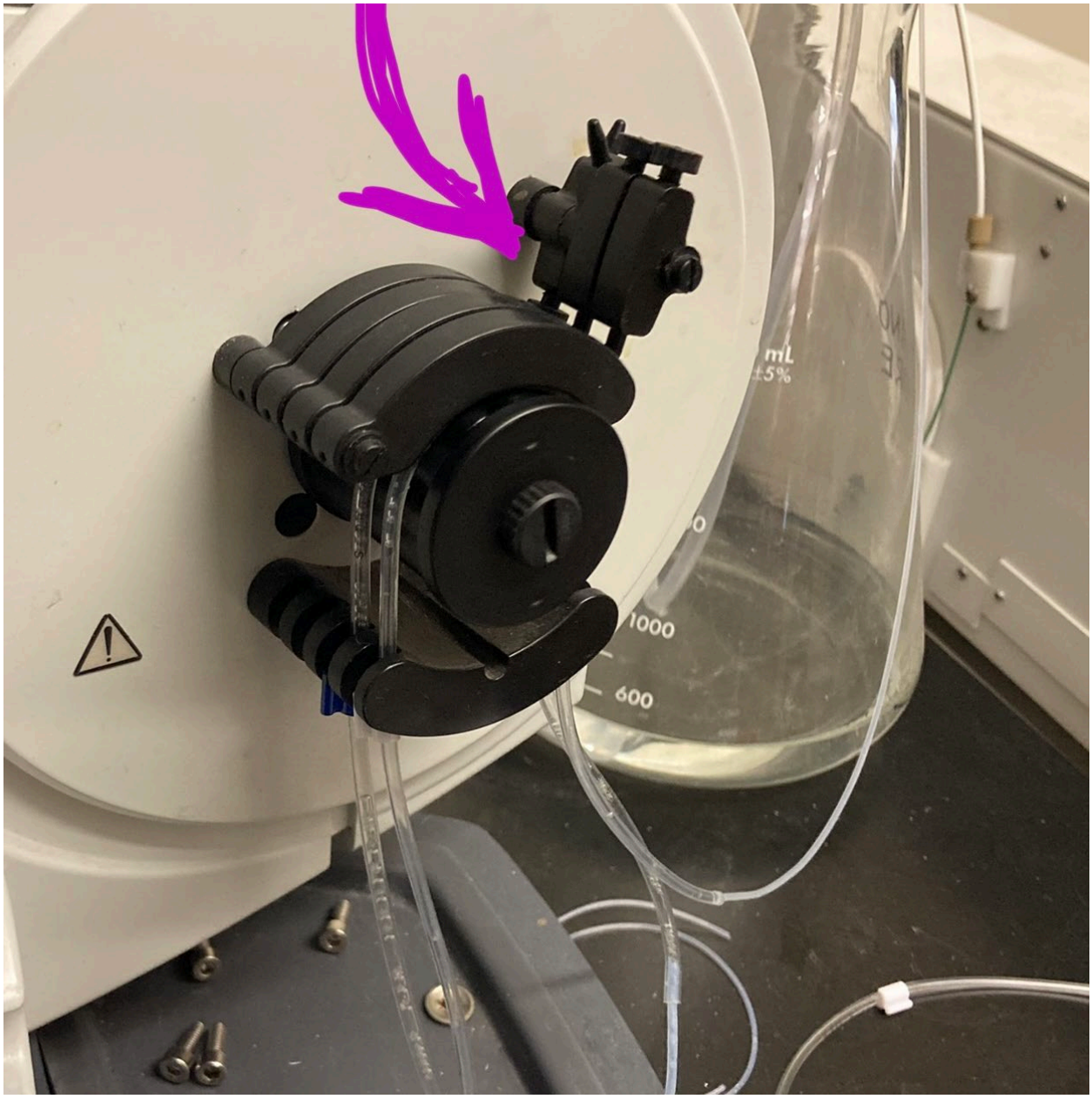

P
High st/QC
1 ppm
Al - 1.0
As - 1.0
B - 1.0
Ba - 1.0
Ca - 200
Cd - 1.0
Co - 1.0
Fe - 2.0
High St
ITPA (QC)
Zn 2.0
Fe 10.0
Mn 5.0
Cu 2.0
As, B, Se
(set) QC
in H2O
As 10.0
B 1.0
Se 10.0
Exchangeables
High st/QC
in NH4OAc
Ca 200 ppm
Mg 50
Na 20
Sulfates
high/QC
in 500 ppm P
Sulfates
(in kcl)
Check
S - 2.0


QC complete
QC
QC


500 mL Vials
ICP*

- To rename your samples: switch to "List View" next to the yellow play button on the computer. You can add samples using the Tube+ icon and remove them with the Tube- icon. Click on a cell on the table to type directly into it.
- Check to make sure the autosampler is turned on. If not, push the red button at the back of the autosampler.
- Click on the "connect auto sampler to PC & initialize" icon, farthest to the right on the toolbar (lightning bolt).
- Make sure camera temp is -45°C:
- Click the little gray pyramid button ("plasma status") in the bottom-right next to "eal" to see instrument status
- Click "instrument status"
- At the bottom of the new window that appears, you can see the camera temperature drop.
- DO NOT turn on the plasma until the camera temperature is -45°C!
- Click on the "run auto-session" icon: yellow arrow in the middle of the toolbar (play button).
- Instrument should ignite. (snapping sound after a few seconds)
- Click pump tubes in place and make sure tension is correct.
- Clamp tubes (blue arrow)
- The first tube goes in the first slot, the second tube goes in the second slot, and then push the clamp down (provide support for the narrow clamp piece (purple arrow) by placing your finger on it as you push it down so it doesn't break off).

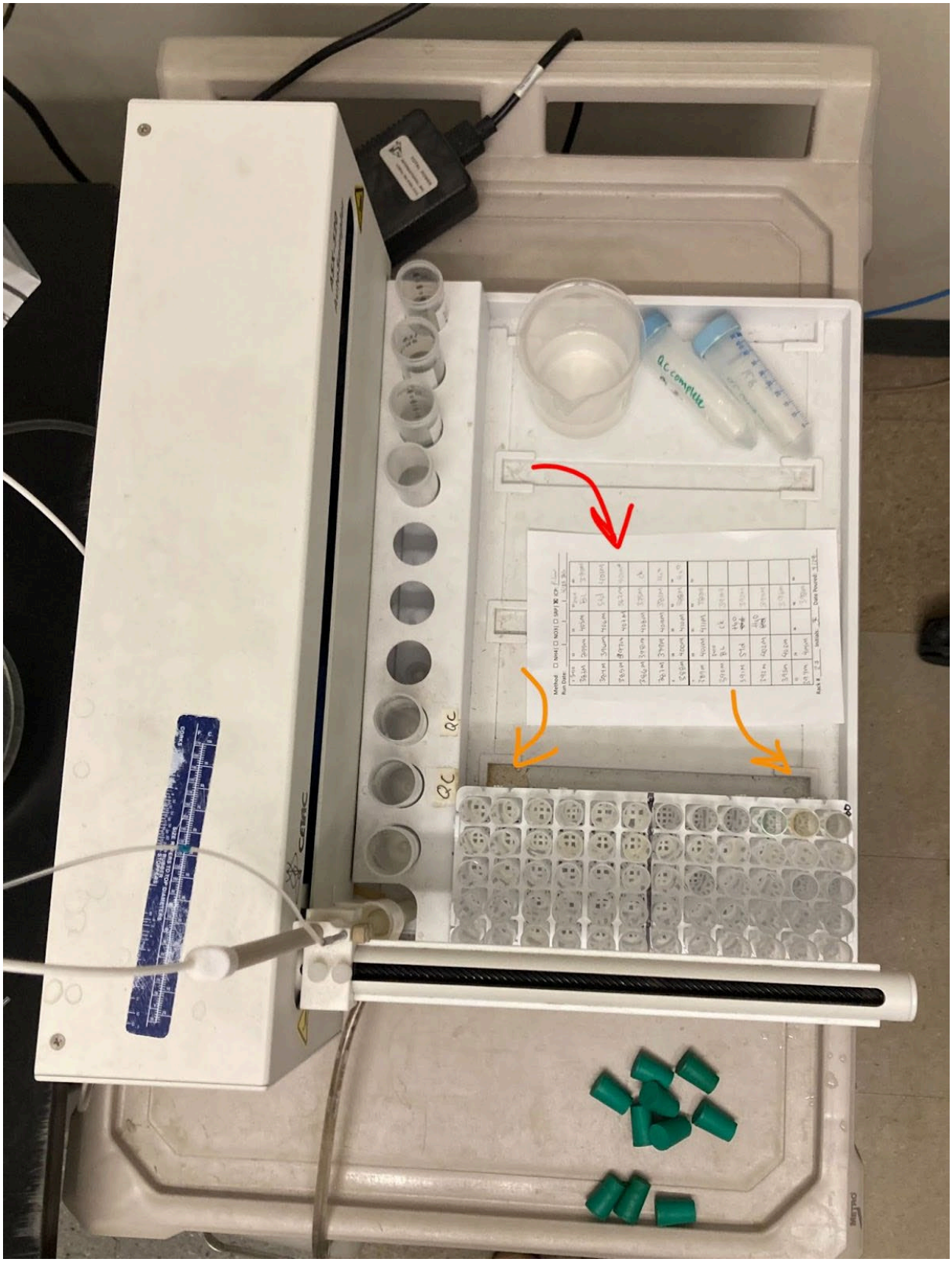

Thermo
SCIENTIFIC
iCAP 7000 SERIES
ICP Spectrometer
READY
pergo



- Make sure vapor can be seen in the nebulizer and that solution is being pumped from the bottom of the nebulizer (at the wide base of the green arrow above).
- If you haven't already (details below in Setting up the Test Tube Rack):
- Place standards and samples in the rack. Place a check in the first tube and in the tube following the samples. Place two water samples at the end of the run.
- Place the rack on the autosampler. Check that it is securely in place with the bottom tabs fitting into the grooves.
- After warm up time, samples will run until the batch is complete.
- Be sure to fill out the test run record sheet for billing information
- To see sample data, click on "Anal…".
- When the run is complete and plasma has shut down, *release pump tubes* and turn off the cooler.
- If the autosampler does not shutdown, turn it off.

- **Setting up the Test Tube Rack:**

- In most cases, digest the samples in the microwave.
- Use a map to keep track of sample placement in the test tube rack. (Red arrow)
- Rinse the test tubes with each sample before filling them ¾ to ⅘ of the way to the top.
- Use gloves while filling the test tubes because during the digestion, you made each sample into a dilute acid.
- Secure the feet of the test tube rack on the machine. (Orange arrows)



- **Printing or saving results:**
  - Click on the word "method".

- Exit
- Click on “Publisher”.
- Click on “Horizontal table report” followed by “New horizontal table report”.
- Select all instruments.
- Click on button to the right of “Method names”.
- Select desired method, click “OK”.
- Select “Find samples analyzed”.
- Select “Between” and add the desired dates.
- Click “Search”.
- Click “Select all” or select the desired samples by highlighting the samples and then right clicking. You can then either select or deselect the samples.
- Check that the Q/C matches within ten percent the number on its bottle (yellow arrow).

- Click “OK”.
- Click on the export icon (floppy disk with gear) at the top of the screen.
- Change the file format to Excel data only and save it to your USB.
- To save, click the envelope icon, select Excel data only and save to your desired location (USB).

12.2.2. TSSD Checklist

Testing Session Checklist

- Label each sample **bottle**. Do NOT just label the lid, as the bottle and sample can easily be mismatched) with a capital letter (A, B, C . . .). Preferably, label the Corral 1 sample bottle “A”, Corral 2 sample bottle “B”, and so on.

- Enter your sample labels into the TSSD data binder and the share point on the sample info sheet. Enter the date, the sample label/ID, and the letter assigned to the sample in the previous step. Use capital letters to avoid confusion between "i" and "l." The individual parameter sheets will auto-populate with the information you enter in the "Main" sheet.
- Put on gloves
- Label the pipettes with the sample letters too and use them for every test.
- Start COD, TOC, Total Phosphorus, and TSS first since those need to cook for a while. Have one person start COD, TOC, and Total P while another person starts TSS.

Order of Tests

Order	Person 1	Person 2	Person 3
1.	COD -Unfiltered -2 hr cook time	TSS/VSS	Reactive Phosphorus -Unfiltered -Filtered
2.	Total Phosphorus -Unfiltered -1 hr cook time	Filter Samples	Nitrite -Filtered
3.	TOC -Unfiltered -Filtered -2 hr cook time	TDS -Filtered	Nitrate -Filtered
4.			Ammonia -Filtered

COD (Chemical Oxygen Demand)

- **Overview:** Use unfiltered sample, cooks for 2 hours.
- Turn on the heating block to 150°C
- There is a program preinstalled on the heating block called COD, just check that it's up and hit "start."
- Lay out a **low-range** COD test vial for every sample and two spikes. Label the spike vials "A spk 1" and "A spk 2"
- These vials can be found in a cabinet on the central lab counter near the sink. The cabinet is labeled COD vials
- A matrix spike for COD is just spike 1 and 2 of sample A
- There are standard and blank solutions but they stay the same. Do NOT throw the Standard and Blank spikes away, they stay good until we open the next box of COD vials.
- **Invert** each sample bottle before pipetting part of the sample into each vial
- Pipette 2mL of **unfiltered** sample into each vial and recap
- Make sure that the labeled caps and vials line up with the sample put in the vial
- Shake well (be careful not to burn yourself, the vials will heat up)
- Do a matrix spike for sample A
- The spike is 1 ml of sample A (from corral 1) and 1 ml of COD Standard
- The COD standard is found in the refrig erator in the back room
- QC set is the standard, the blank, and the two spikes
- The standard and the blank solutions stay the same. Do NOT throw the Standard and Blank spikes away, they stay good until we open the next box of COD vials.
- Shake well

- Compare the liquid levels in the vials to each other
- They should all be the same. If not, something went wrong. Figure out what went wrong and redo the test
- Place the vials with samples and the two spikes in the heating block
- The standard and the blank are NOT cooked again. Do NOT place them in the heating block!!!
- Once the temperature has reached 150C, press start
- The block should beep when ready. If not, the temperature is displayed
- The two hour timer is programmed into the block. Make sure when you press start it starts counting down.
- Shake the samples as you take them out of the heating block. Do this as soon as the timer beeps because they'll cool faster on the counter than in the block
- Once the vials have cooled, gently clean all of the vials with a chem wipe to make sure the results are accurate.
- Calibrate 0 in the machine by reading the blank first and setting that as 0
- Read the results, make sure NOT to shake the vials when you read the results.
- To read the results you need to change the program on the spectrometer. The COD program is preinstalled under the "favorites" tab. You will need to press "start" each time you read a vile
- Record the results in the TSSD Lab Binder
- This data will later be copied to the Sharepoint
- Clean up by throwing all vials still filled with liquid (except for the Blank and Standard) into the bin labeled COD
- This is in our back room
- **Do NOT pour COD down the sink!!!!**

Total Phosphorus

- **Overview:** Cooks for 1 hour, unfiltered.
- Heat the heating block to 100°C
- There should be a preset program at 100°C for an hour
- Lay out and label **Low Range** vials for the samples and **High Range** vials for the QC and Matrix spikes.
- Carefully remove the protective foil lid from the vial. Unscrew the cap from the vial
- **Invert** each sample bottle before pipetting part of the sample into each vial
- For **Low Range**, pipette 2.0mL of **unfiltered** sample into the reagent vial
- Reminder, low range is used for the sample, High range is used for the QC and matrix spikes
- Flip the cap over so that the reagent side on the cap faces into the vial. Screw the cap tightly onto the vial.
- Shake the capped vial 2-3 times to dissolve the reagent in the cap. Verify the reagent has dissolved by looking down through the open end of the cap lid.
- Prepare the Matrix spikes and QC. Use the **High Range** Vials
- The QC is Standard, OPR, and Blank
- Place 0.5 ml of Standard, OPR, and Blank in their respective vials
- The OPR standard used for Total Phosphorus and Reactive Phosphorus are the same
- The standard and OPR are found on the central counter near the pipette tips
- The Matrix spikes of sample A (1 and 2)
- Pipette 0.25 ml of unfiltered sample into the vial
- Then pipette 0.25 ml of T-phosphorus **standard** into the same vial
- Flip the cap over so that the reagent side on the cap faces into the vial. Screw the cap tightly onto the vial
- Shake the capped vial 2-3 times to dissolve the reagent in the cap. Verify the reagent has dissolved by looking down through the open end of the cap lid
- You can't shake the vial too much, but you can shake it too little!

- Check all the low range and high range vials. Make sure that each group has the same amount of liquid in them
- If they do not, figure out what went wrong and redo the necessary vials test
- Insert the vials (all the samples, QC, and Matrix spikes) into the reactor/heating block and start the timer
- Should be at 100°C. for one hour. Close the protective cover and heat for one hour at 100°C
- After the timer expires, carefully remove the hot vial from the rector. Insert it in the cooling rack and allow to cool to room temperature
- Pipette 0.2mL of reagent B into the cooled vial. Immediately close the reagent B container
- Screw the grey cap C onto the vial.
- Invert the capped vial 2-3 times to dissolve the reagent in the grey cap.
- It's okay to shake the bottle. The most important thing is that the reagent in the cap is dissolved into the solution.
- You can't shake the bottle too much but you can shake it too little
- Set a timer for 10 minutes.
- When the timer expires, invert the vial again 2-3 times.
- The reagent cannot stay in too long, but it can stay in for too short of a time. Make sure that the vials sit undisturbed for at least 10 minutes
- Wipe the outside of the vials with a chem wipe
- Clean the outside of each vial before inserting them in the reader.
- Make sure that the units are PO_4^{3-}-P
- Record the results on a piece of paper or on the Google Sheets
- Clean up by dumping out the contents of the vial down the sink in our back room and throwing the vials and caps in the garbage

TOC (Total Organic Carbon)

- Read the instructions on the box or follow the steps below
- These are both **Unfiltered** and **Filtered** Samples
- Turn on the heating block for 100 °C
- Label the vials with the respective sample label and F for filtered or U for unfiltered.
- Pipette 2 mL of sample into the clear vial
- Place the clear vial into the shaker for 5 minutes without a lid
- Be sure to push the vial all the way into the holder
- The shaker has its own 5 min timer just turn it on
- Take the green lid off the purple/blue vials and place the clear lid on with the barcode down
- Once the clear vials are done shaking attach the clear vials to the blue/purple vials with the clear cap
- Place the vials in the heating block, clear vial down, for 2 hours
- When the timer goes off, take the vials out and let them cool, clear vial still down
- **Do NOT mix the vials**
- When the vials are cool, gently tip them over (Blue vial should be on the bottom)
- Clean the blue vial with a kimwipe
- Read the blue vial and record the data in both the TSSD data binder and the Sharepoint file online
- To clean up throw the whole vial (blue and clear) in the trash
- **DO NOT dump the liquid down the sink**

TSS & VSS (Total & Volatile Suspended Solids)

- Get a 0.45 micron 47mm diameter filter for each sample and label each filter with the sample letter on the very edge (use a sharpie). Don't handle filters with your bare hands--wear gloves and/or use forceps.
- Place filters (wrinkled side up) in watch glasses and put them in the muffle furnace (500 +/- 50° C) for 5 minutes.

- Tare the scale, and then weigh each filter using the analytical balance (the one with a glass cover) and record the weight on the "TSS" spreadsheet under "Filter Weight."
- Next, set up the 3-part vacuum assembly. Clean the filter funnels with a kimwipe and attach them to the bases as shown in the picture. The bases and filters snap together with a magnet.
- Place one filter on the funnel base with the wrinkle side up.
- Turn on the vacuum pump using the switch below the counter.
- Thoroughly **mix the sample by inverting the sample bottle** several times, then fill a class A graduated cylinder with 100mL of sample. You can use a different volume if you're low on sample, but always record the volume you used on the "TSS" spreadsheet under "Sample Volume."
- If there's large chunks in the sample volume you poured out, discard it and pour a new volume.
- Turn on the vacuum by rotating the lever clockwise and empty the graduated cylinder into the funnel.
- Use a squeeze bottle to rinse the sides of the cylinder with DI water, then pour the rinse water into the funnel. Also rinse the inside of the funnel with a squeeze bottle while the vacuum is running.
- Leave the vacuum running for approximately three minutes. The filtration is done when the filter paper no longer looks wet.
- Remove the filters from the filtration apparatus and return them to the watch glasses. Dry the filters on the watch glass in the Thermo oven for a minimum of 1 hour at 103-105 °C. Note the time that you put them in the oven so you can record how long they were in for.
- Cool on a piece of paper
- Remove the filters from the oven and record the time in the oven on the "TSS" sheet under "Time." Weigh each filter on the analytical balance and record the weight under "1st Weight."
- Place the filters into the Thermo Oven **overnight**
- Take the filters out of the oven, let them cool and then read the 2nd weight reading
- Place filters in the one small white bowl (found on the muffle furnace) and place in the muffler furnace (500 +/- 50° C) for 20 to 30 min
- Weigh filters on the analytical balance, and record the results on the "VSS" sheet under "Final Weight" and in the TSSD data Binder
- Check that the spreadsheet has filled in the "Results" column for both the "TSS" and "VSS" sheets, and enter your initials in the "Analyst Initials" column of both spreadsheets.

TDS (Total Dissolved Solids)

- Grab as many mini beakers as there are samples.
- We should have a stash of 9 beakers in the back room (A-I) for us to use
- Weigh beaker A and record the weight in the TSSD data Binder or online
- Tare the beaker weight of beaker A on the scale then pour the **filtered** sample A into the beaker and place on the scale.
- This give you the sample weight
- Record the sample weight in the TSSD data notebook and on the Sharepoint
- Repeat the above three steps for the rest of the samples.
- Be sure to zero out the scale between each sample measurement- the scale has a tendency to drift
- Place all of the beakers in the 105C oven overnight
- When the samples have dissolved, take the beakers out and allow them to cool
- Reweigh the beakers and record the weight in the TSSD data Binder and in the Sharepoint file online

- Clean the beakers by scrubbing out the dissolved nutrients and placing them in the HCL Bath near the back corner sink

Reactive Phosphorus

- **Overview:** 10-minute wait time, unfiltered and filtered samples (two separate tests with the various filtered and unfiltered samples)
- Do steps 2 and 6-9 on the box.
- Lay out and label **Low Range** vials for the samples and for the QC and Matrix spikes.
- Label with the sample letter as well as F for filtered and U for unfiltered
- Carefully remove the protective foil lid from the vial. Unscrew the cap from the vial and throw the green caps away
- Pipette 2.0mL of **unfiltered** sample into the reagent vial
- Reminder, low range is used for the sample, High range is used for the matrix spikes
- Prepare the Matrix spikes and QC.
- The QC is Standard, OPR, and Blank
- Place 2 ml of Standard, OPR, and Blank in the vials
- The OPR standard used for Total Phosphorus and Reactive Phosphorus are the same
- The standard are found on the central counter near the tips
- The Matrix spikes of sample A (1 and 2)
- Pipette 1 ml of unfiltered sample into the vial
- Then pipette 1 of O-phosphorus **standard** in to the same vial
- Pipette 0.2mL of reagent B into the cooled vial. Immediately close the reagent B container
- Screw the grey cap C onto the vial.
- Invert the capped vial 2-3 times to dissolve the reagent in the grey cap.
- It's okay to shake the bottle. The most important thing is that the reagent in the cap is dissolved into the solution.
- You can't shake the bottle too much but you can shake it too little
- Set a timer for 10 minutes.
- When the timer expires, invert the vial again 2-3 times.
- The reagent cannot stay in too long, but it can stay in for too short of a time. Make sure that the vials sit undisturbed for at least 10 minutes
- Wipe the outside of the vials with a chem wipe
- Clean the outside of each vial before inserting them in the reader.
- Make sure that the units are in PO_4^{3-}-P
- Record the results in the TSSD data Binder and on the Google Sheets
- Clean up by dumping out the contents of the vial down the sink in our back room and throwing the vials and caps in the garbage

Nitrite (NO_2^-)

- Follow the instructions on the box lid or the checklist below
- Lay out and label **Low Range** for the sample bottles, QC, and matrix spikes
- Carefully remove the protective foil lid from the cap of the TNT vial. Unscrew the cap from the vial and place on the counter
- **Invert** each sample bottle before pipetting part of the sample into each vial
- Pipette 2mL of sample into the reagent vial
- For the matrix spikes pipette 1 mL of sample A and 1 mL of Nitrite standard
- For the QC pipette 2 mL of Nitrite Standard in one vial, 2 mL of OPR in another vial, and 2 mL of DI water.
- Immediately flip the cap over so that the reagent side faces inside the vial.
- Shake the capped vial 2-3 times to dissolve the reagent in the cap. Verify the reagent has dissolved by looking down the through the open end of the vial lid.
- Wait 10 minutes.
- Wipe the vials.

- Shake the vials before reading them. To read the vials insert into the cell holder. The instrument reads the barcode, then selects and performs the correct test. Results are in mg/L NO_2-N.
- Continue to read the remaining samples and record results in the Sharepoint excel file.

Ammonia as N (NH_3-N)

- Follow the instructions on the box lid or the checklist below
- Lay out and label **Low Range** for the sample bottles, QC, and matrix spikes
- Carefully remove the protective foil lid from the cap of the TNT vial. Unscrew the cap from the vial
- Pipette 5mL of sample into each sample vial
- For the matrix spikes pipette 2.5 mL of sample A and 2.5 mL of Ammonia standard
- For the QC pipette 5 mL of Ammonia Standard in one vial, 5 mL of OPR in another vial, and 5 mL of DI water.
- Flip the cap over so that the reagent side faces the vial. Screw the cap tightly onto the vial.
- Shake the capped vial 2-3 times to dissolve the reagent in the cap. Verify that the reagent has dissolved by looking down through the open end of the cap. Wait 15 minutes.
- Verify that all of the vials have the same level of liquid in them. If they are not, figure out what happened and redo the test if need be
- After 15 minutes, invert the sample an additional 2-3 times to mix.
- Thoroughly clean the outside of the vial with a kimwipe.
- Insert the prepared vial into the cell holder. The instrument reads the barcode and performs the correct test. Results are in mg/L NH_3N.
- Continue to read the remaining samples and record the results on paper or on the sharepoint excel file.

Nitrate (NO_3^-)

- Follow the instructions on the box lid or the checklist below
- Lay out and label **Low Range** for the sample bottles, QC, and matrix spikes
- Carefully remove the protective foil lid from the cap of the TNT vial. Unscrew the cap from the vial
- Pipette 1mL of sample into the reagent vial
- For the matrix spikes pipette 0.5 mL of sample A and 0.5 mL Nitrate standard
- For the QC pipette 1 mL of Nitrate Standard in one vial, 5 mL of OPR in another vial, and 5 mL of DI water.
- Pipette 0.2mL of solution A into the vial. Do these two at a time and cap and shake immediately cause this test is a little more finicky.
- Cap and invert the reaction tube 2-3 times until no more streaks can be seen in the reaction tube solution.
- Wait 15 minutes.
- Shake the vials
- Wipe the vials with a chem wipe.
- Read the vials. The instrument reads the barcode, then selects and performs the correct test. Results are in mg/L NO_3-N. Record the data on the sharepoint excel file.
- Continue to read the remaining samples and record results

Group 1

- TSS/VSS - Filter 250 ml of the sample
- TDS - filtered sample

Group 2

- COD - unfiltered sample
- Total Phosphorus- unfiltered
- Reactive Phosphorus - unfiltered and filtered

Group 3

- Nitrate- filtered

- Nitrite- filtered
- Ammonia- filtered

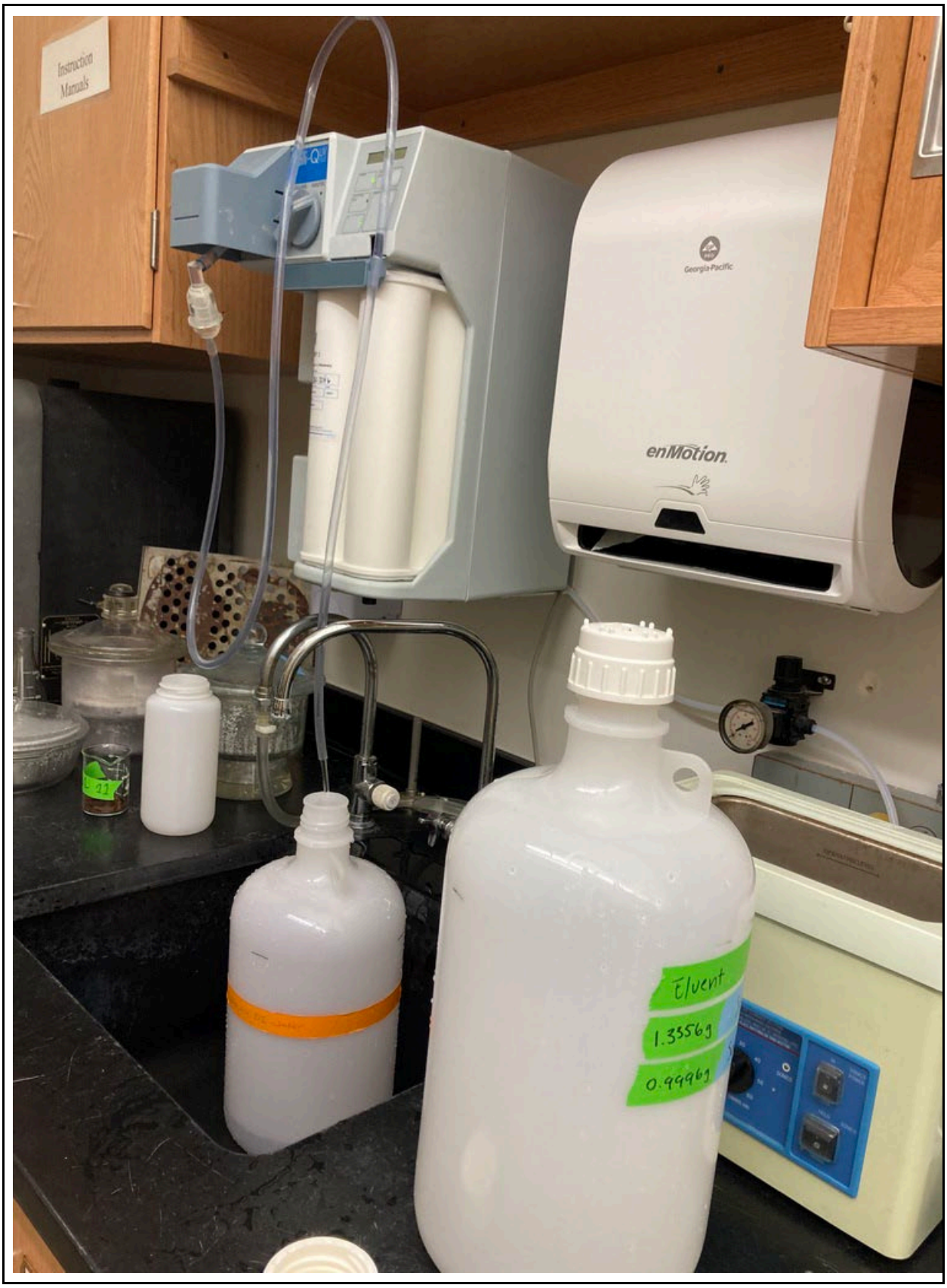
Total Nitrogen

- Pipette 1.3 mL into each bottle (large glass bottles). 5 QC samples: a spike, a2 spike, blank, standard, ORP
- Use same standards for this that are used for TKN
- Add 1.3 mL of reagent with blue lid to every sample
- Add a white tablet from the vial to each bottle
- Put the caps on and put them in a heating block for 30 minutes at 120C
- Take them out, shake them, let them cool, then take 5mL from those bottles and put them into each of the green-lidded vials, add the reagent, and read on the spectrophotometer

12.2.3. Ion Chromatography

Operating the Milli-Q Machine

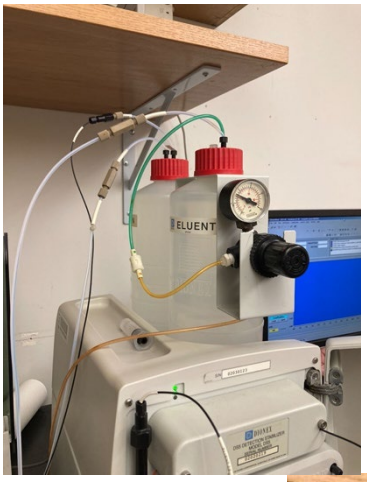
- The milli-Q water machine is above the sink closest to the main lab door.
    - Turn it on by flipping the switch on the top in the back left corner.
    - Make sure the screen reads: Product 18.2 mOhm cm
        - If it doesn't say this:
            - Press the "operate/standby" button until it changes.

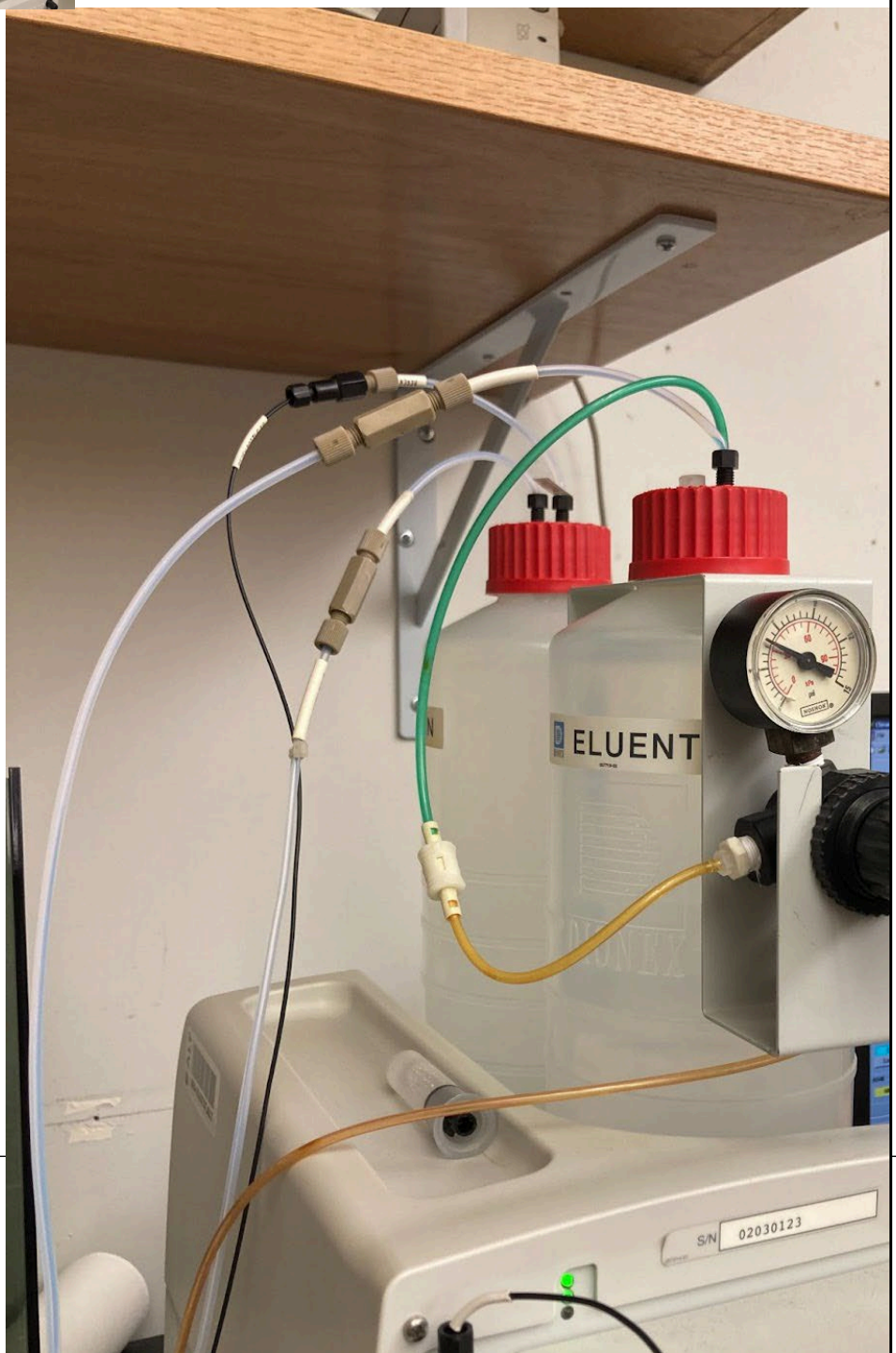

Instruction Manuals
Georgia-Pacific
enMotion
Effluent
1.3556g
0.9996g

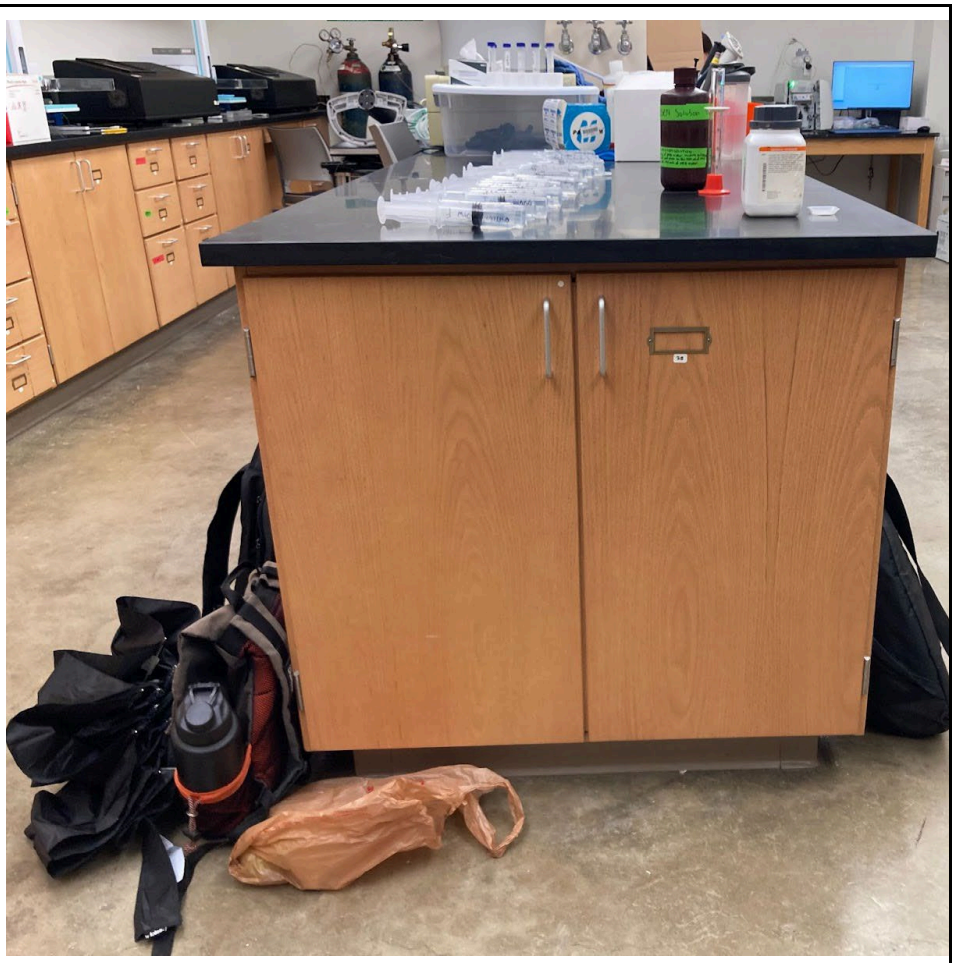
Milli-Q water machine

Setting Up the Machine

- Every run, you **MUST** replenish the "Eluent" and "Regenerate" solutions in the 2-liter bottles:
    - Pull out the spare Regenerate and Eluent solution, which is kept in 7-liter jugs in the cabinet on the end of the middle lab bench closest to the door.
        - Spare Regenerate solution is good for a few weeks, but **spare Eluent solution is only good for two days**. If it has been two days since the spare Eluent solution was made, you have to toss it out and make a new solution. Instructions on making new solutions are in the next section.


ELUENT


ELUENT
S/N 02030123



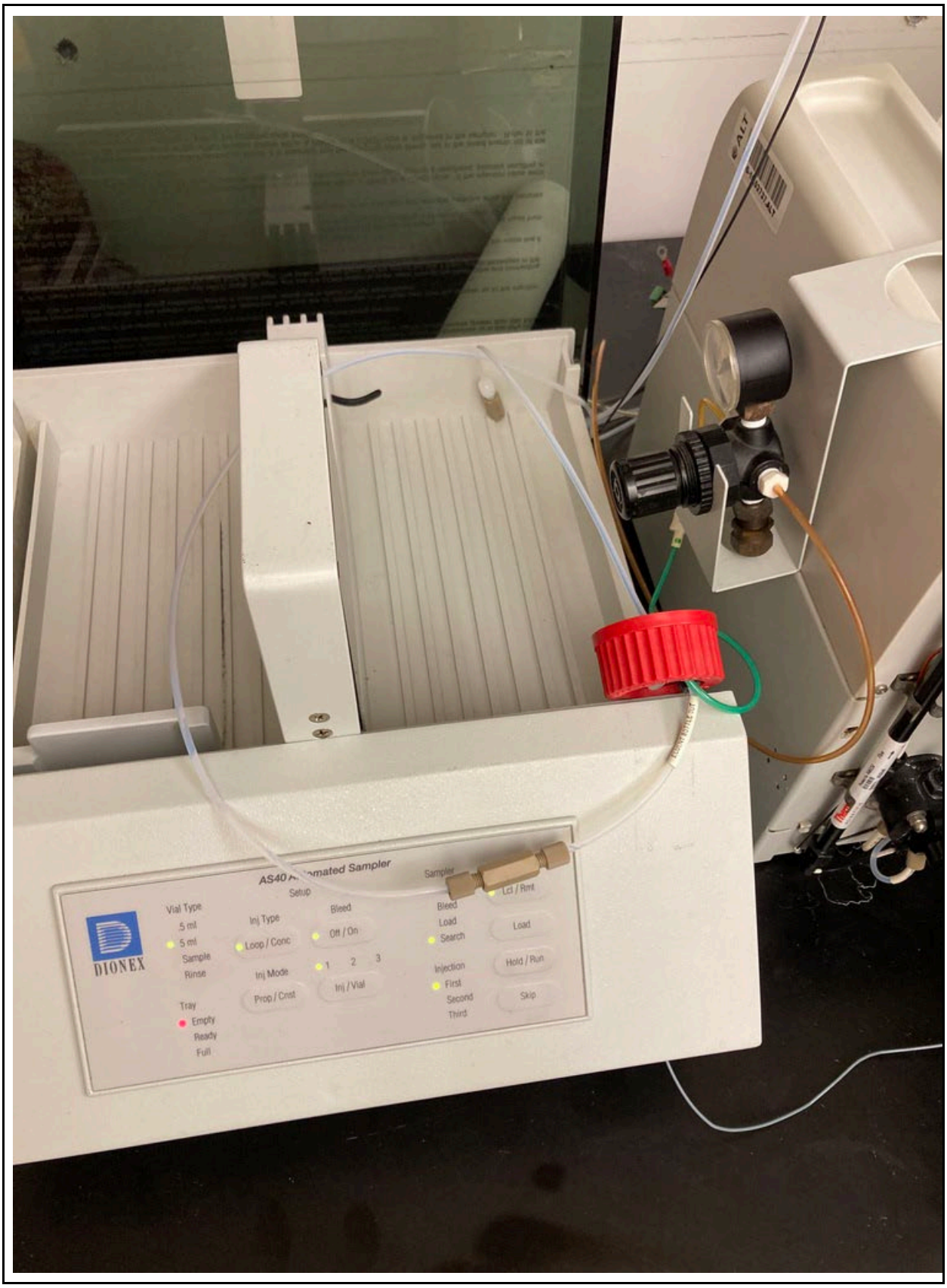
Eluent and regenerate solutions (2-liter bottles) in the IC machine
Cabinet where spare solutions are stored

- Close the **rightmost** nitrogen gas cylinder by twisting the silver knob. **DO NOT** touch any of the other knobs on the cylinder.


NITROGEN
FORT AUDERD

Nitrogen gas tank

- After the cylinder is closed, you may lift the regulator (silver necktie looking thing with a pressure gauge on it) from where it sits on the eluent 2-liter bottle and loosen the red cap on the eluent bottle. Wait for the hissing sound to stop, then remove the cap entirely. The stick hanging down from the inside of the cap should not touch your hands because the oils on your hands will contaminate it.
    - Place the cap to the side so that the stick is resting on the autosampler tray or on a paper towel.


DIONEX
Vial Type
5 ml
5 ml
Sample
Rinse
Tray
Empty
Ready
Full
Setup
Inj Type
Loop / Conc
Inj Mode
Prop / Cnst
Bleed
Off / On
1 2 3
Inj / Vial
Sampler
Lct / Rmt
Bleed
Load
Search
Load
Injection
First
Second
Third
Hold / Run
Skip

Cap in autosampler.

- Remove the Eluent bottle from the machine.
- Dump excess Eluent solution from the 2-liter bottle down the sink.
- Triple rinse with milli-Q water.
- Refill Eluent 2-liter bottle to the shoulder from the fresh spare solution in the 7-liter jug.
- Put the Eluent 2-liter bottle back in the machine, topped with the regulator and red cap.
- Turn the rightmost nitrogen gas cylinder back on by twisting the silver knob back one full turn (until the regulator is at about 40 psi).
- Take off the cap of the Regenerate bottle, handling it the same way you did with the Eluent bottle.
- Remove the Regenerate bottle from the machine
- Dump excess Regenerate solution from the 2-liter bottle down the sink.
- Triple rinse with milli-Q water
- Refill Regenerate 2-liter bottle to within ¼ inch of the very top from the fresh spare solution in the 7-liter jug.
- Put the Regenerate 2-liter bottle back into the machine, topped with the red cap.

Making Fresh Regenerate and Eluent Solutions

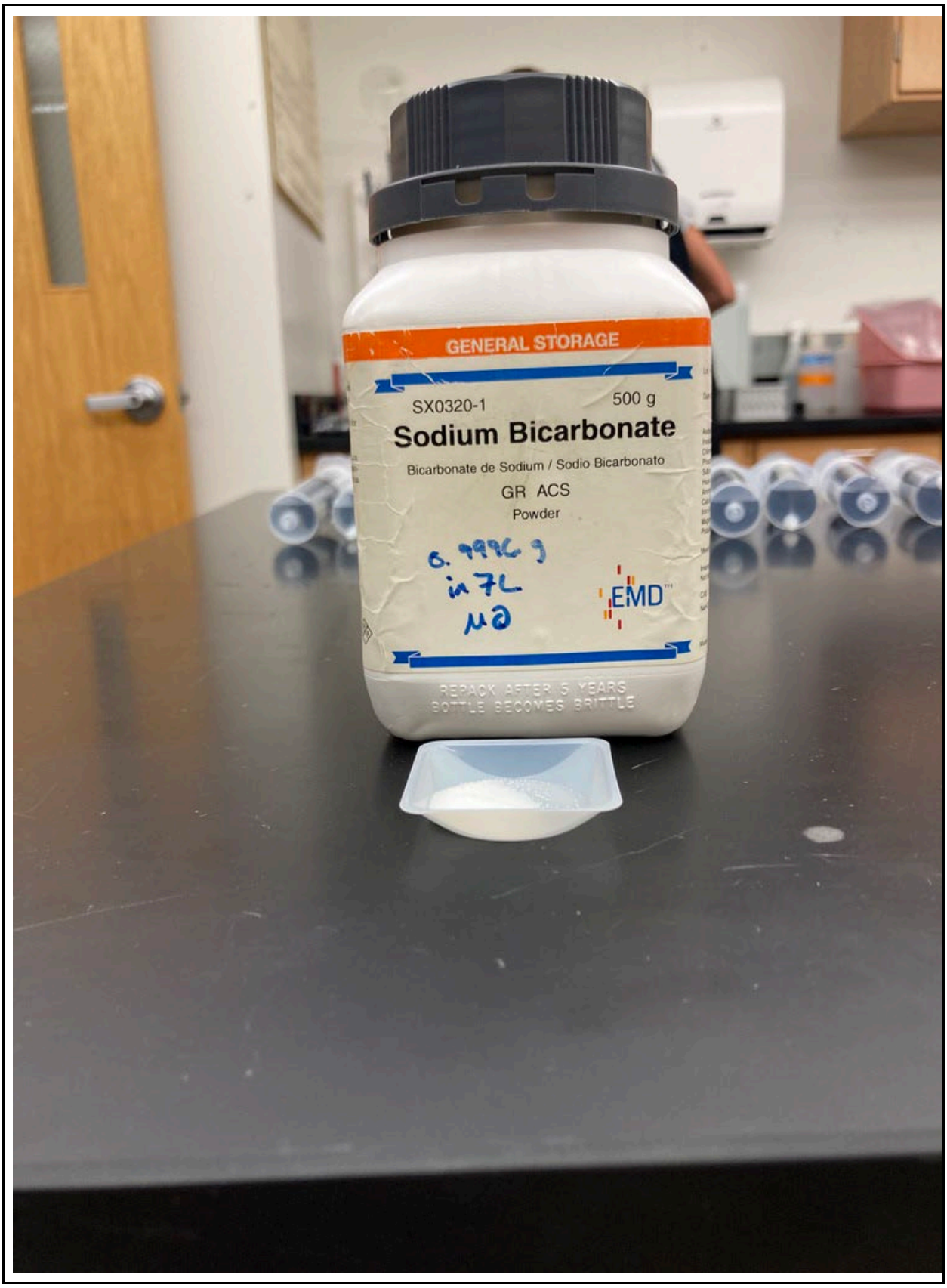
- **Never mix regenerate and eluent solutions. The machine is very delicate and mixing them, however carefully, will mess it up.**
    - If there is extra regenerate or eluent (there will usually be about 1 liter left in the jugs, since each machine bottle holds 2 liters), dump it down the sink.
    - Triple rinse the jugs with the cap using milli-Q water, but **don't** touch the inside of the jugs with your fingers or with the end of the milli-Q hose. Don't let the hose touch the jug.
- Fill the jugs to the Sharpie line with milli-Q water.
- Get the raw material to make the regenerate and eluent solutions from the cupboard across from the cabinet.

Raw materials cupboard.

- The recipe for each solution is written on the tape on the jugs and on the raw materials containers. The two solids are for making eluent, and the regenerate solution is liquid. **You must wear gloves and use a fume hood when handling the regenerate solution.**
  - Make the eluent solution:
    - The balance and instruments are on the table down and left from the raw materials cupboard. Wipe off all the instruments with a Kimwipe before, between, and after using them to measure solid chemicals.
    - Get a weigh boat from the same cupboard as the raw materials.
    - Tare the balance to 0.000g.
- Don't touch the inside of the solid material containers with your fingers, but you don't need to use gloves while measuring the solid material.
- The weight of the raw material must be accurate to 4 decimal places.
  - Start with less raw material, and measure out each of the two ingredients to 4 decimal places accuracy.
  - Give the balance ample time to catch up in between addition/subtraction of materials to/from the weigh boat.

Measuring out raw material on the balance.

- Put extra raw material on the spatula in the trash.


GENERAL STORAGE
SX0320-1
500 g
Sodium Bicarbonate
Bicarbonate de Sodium / Sodio Bicarbonato
GR ACS
Powder
EMD
REPACK AFTER 5 YEARS
BOTTLE BECOMES BRITTLE

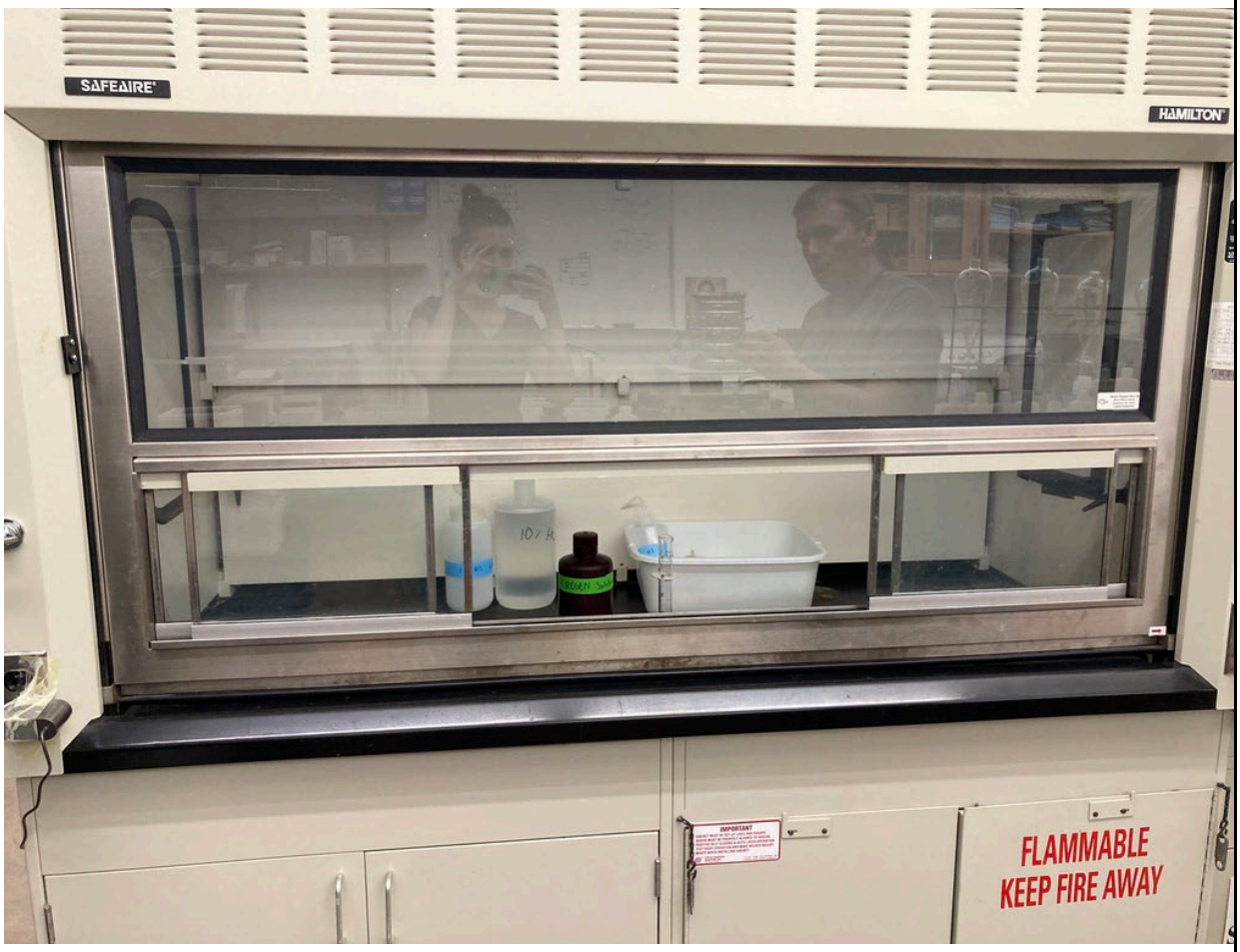
One of the raw materials containers with a weigh boat.

- To put the solid chemicals into the jug, pinch the opposing corners of the weigh boat to make a funnel and pour each chemical in separately. If some stays on the weigh boat, shake it off by tapping the topmost corner of the weigh boat.


XL

Funneling raw material into the jug.

    - Cap the jug and invert it several times to mix.
- Make the regenerate solution:
- **Use gloves.**
    - Take the bottle out to the fume hood in the main lab area.
    - Measure out the regenerate solution into a graduated cylinder (stored in the raw materials cupboard).

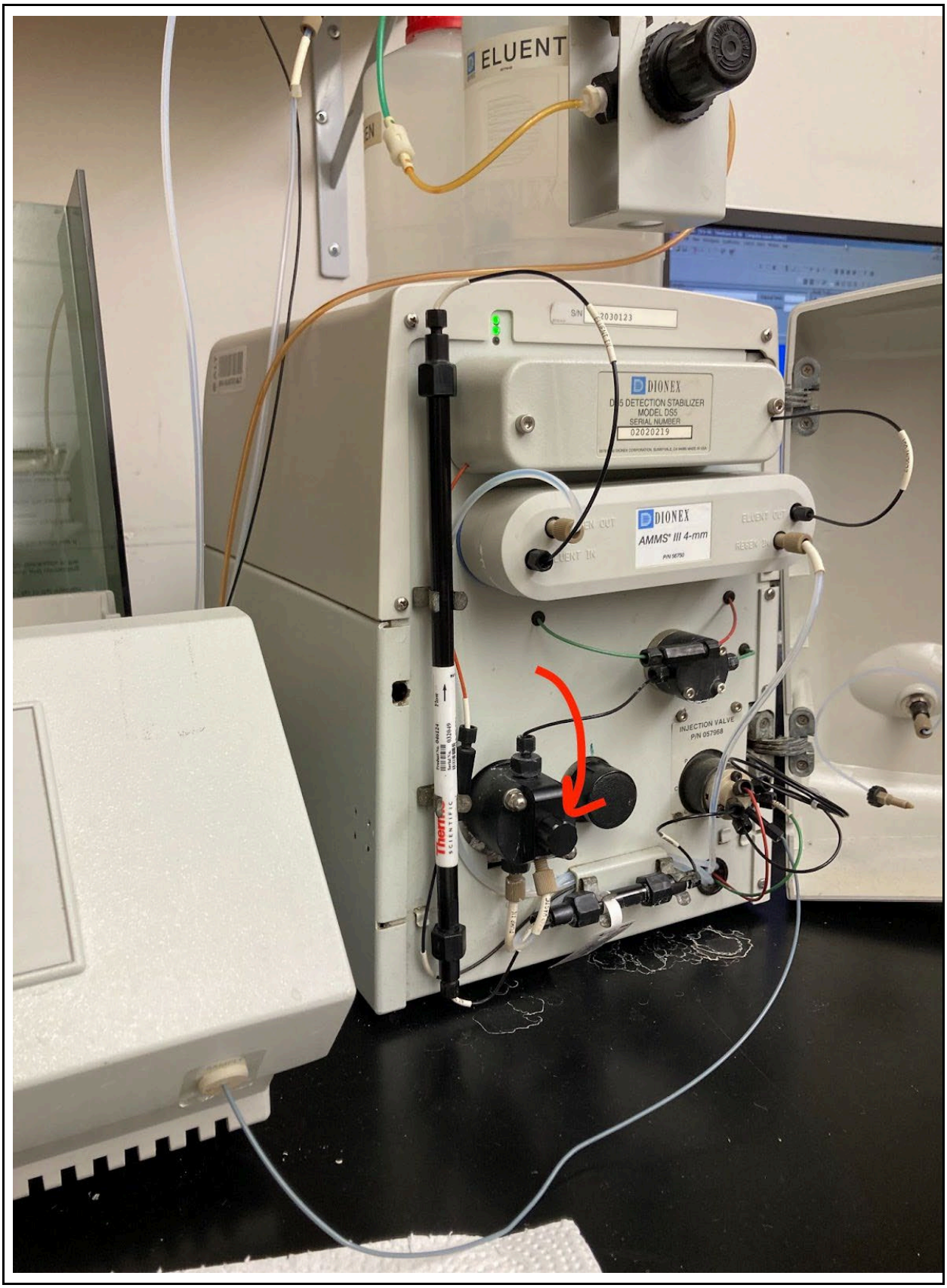


Fume hood with regenerate solution and graduated cylinder inside.

    - Pour it into the regenerate jug.
    - Cap the jug and invert it several times to mix.

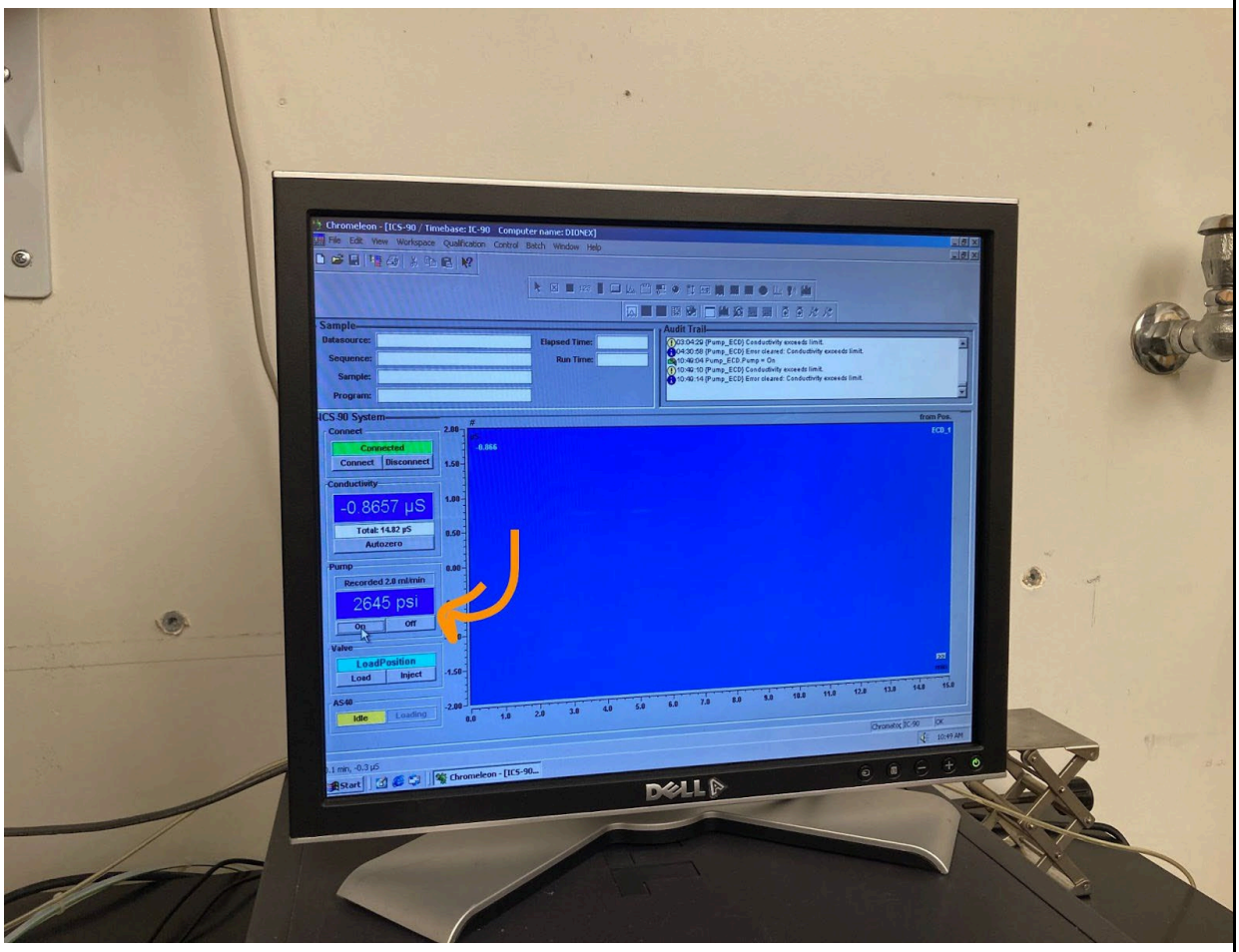
Start the Machine

- Bleed the bypass pump: Open the little black (pump bypass) knob half a turn and watch for a bubble coming out of the eluent tubes- leave the pump bypass open for 15 seconds, then close it.


ELUENT
DIONEX
DS5 DETECTION STABILIZER
MODEL DS5
SERIAL NUMBER
02020219
DIONEX
AMMS* III 4-mm
INJECTION VALVE
P/N 057968

Red arrow points to pump bypass knob.

- In the time base window, click “on” in the pump box.



The orange arrow points to the pump box. When it is off, the box with the numbers on it is red. When it is on, the box has numbers on it. The screen shown is the time base window.

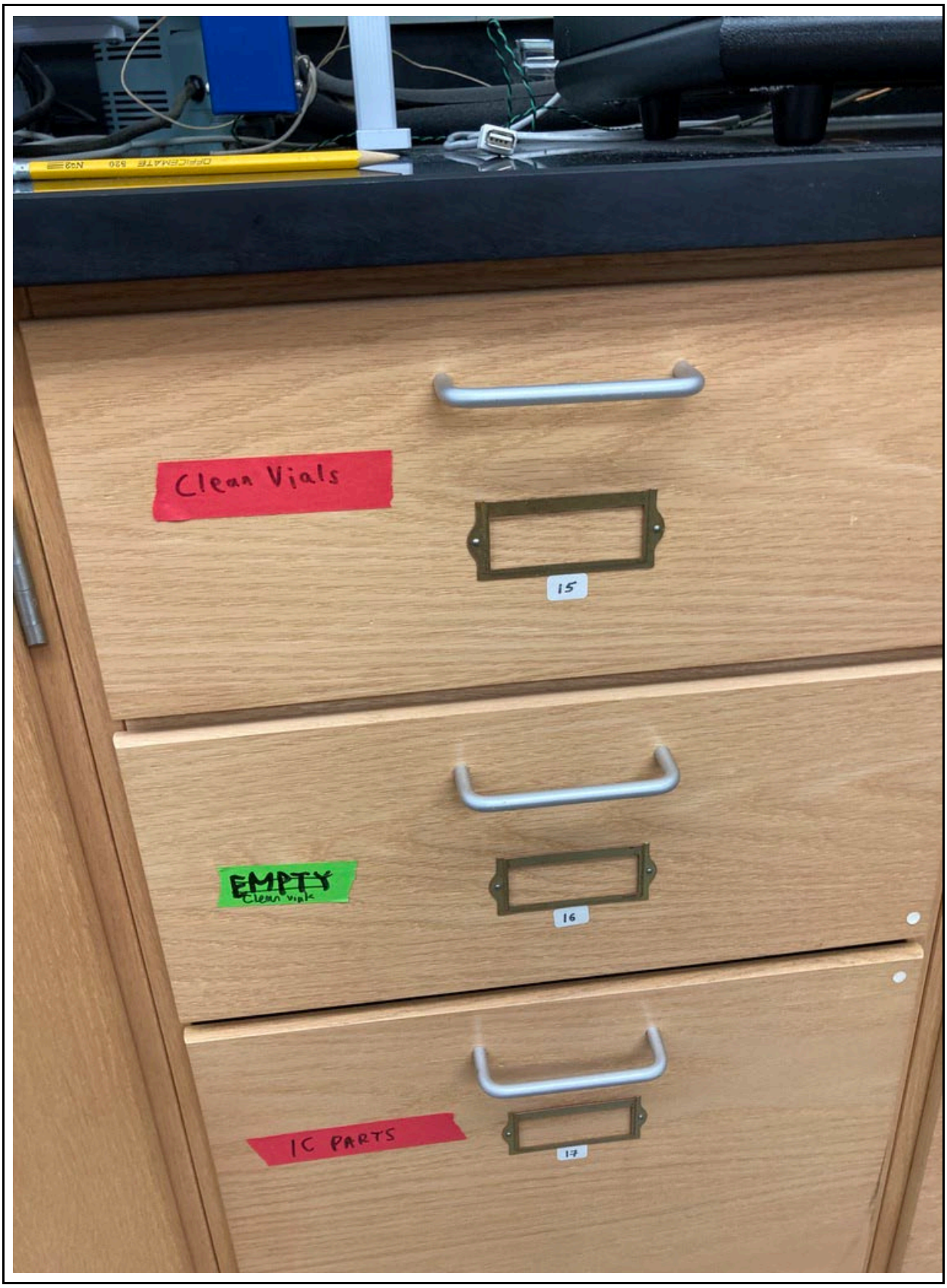
- Bleed the pump bypass knob again with the pump running. When the pressure drops to 500 (as shown in the pump box on the time base window), close the bypass pump again.
- Let the pump run for 10-15 minutes before starting anything else.

Set up the autosampler

- The trays for the autosampler are in the drawer on the same side of the wall as the door, labeled “IC trays”.

Drawer for IC trays

- Vials and vial caps are in the drawer on the same side of the wall as the door, labeled “Clean vials”.


Clean Vials
15
EMPTY
Clean sink
16
IC PARTS
17

Drawer for clean vials

- The autosampler can hold up to eight trays at once.
- Fill the first tray with standards. The standards are in the fridge on the bottom shelf of the door.


VWT

The standards are on the bottom shelf.

- The first spot (marked by a black dot) is for a vial of milli-Q water.
- The second through sixth spots are for the standards, 1-5. Keep track of where each standard is placed.
- The seventh spot (first spot in the second tray) is for a vial of milli-Q water.
- The last spot in the total sequence you will be running is for a vial of milli-Q water.

Milli-Q water spots are marked in blue, standard spots are marked in purple, sample spots are everywhere else. If you do not fill all eight rows, put the milli-Q vial in the spot after the last sample.

- Don't touch the inside of the vials or the underside of the caps while preparing them. Even in gloves, it will contaminate them and mess up the readings.
- Fill vials to within a quarter-inch from the top.
- Set cap in the vial, then use the black tool by the sink by the door to insert the cap.
    - Over the sink, push the cap down using the slit side of the tool.
    - Use the other side to push the cap down until the top of the spike is flush with the lip of the vial.
        - Try not to have air bubbles in the vial.


Non Potable Water
(Industrial Water)
DO NOT DRINK

Filling the vial with sample, using the tool to push the cap in, and placing in tray.

- Place the samples in the autosampler by setting the trays in the left side with the black dot towards the center. Put the side of the trays with teeth towards the teeth on the black belt on the inside of the front wall of the machine.

The conveyor belt is marked by the green line.

Set up the Computer

- From the window, go to the browser.
- Click on all-2022, then 2022 template.
- Click “File” > Save as > *date**month abbreviation**year abbreviation*_tssd > Save
    - Example of file name: 15jul22_tssd
- Click “Highlight unknowns”
- Right click “insert samples” to make more unknowns.
- Stop command at end.
- POS numbers don’t matter.
- Select “name” and start typing to change the name.
- Number 7 and the milli-Q at the end are “rinses”.
- Save the sequence (file > save) once everything is labeled.
- Click “batch” > start > find your run > ready check > okay
- Close the autosampler lid
- Press the “hold run” button on the autosampler.
- When the green light is lit, press “start”.

13. Contact Information

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