The Aquatic Monitoring Program for the Hunting Creek Area of the Tidal Freshwater Potomac River 2015

FINAL REPORT
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by

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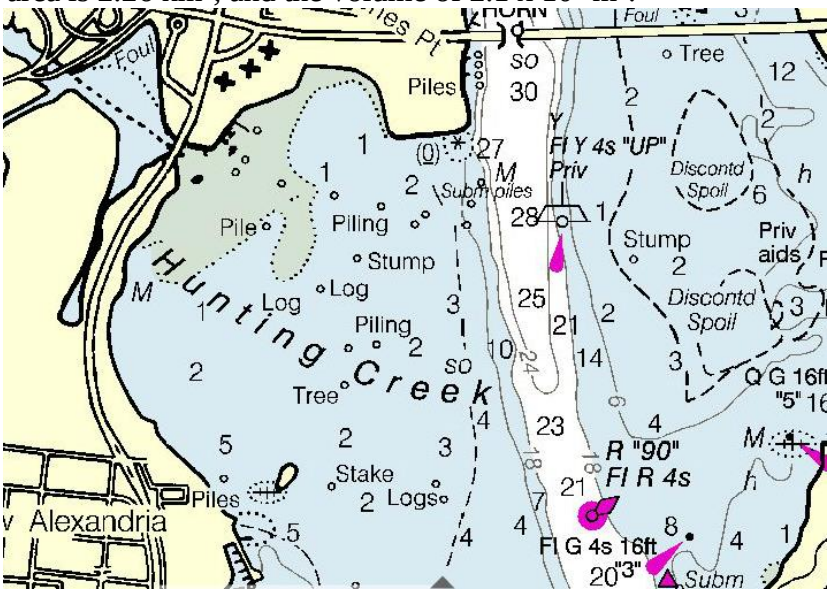
INTRODUCTION

This section reports the results of the third year of an aquatic monitoring program for Alexandria Renew Enterprises conducted by the Potomac Environmental Research and Education Center (PEREC) in the College of Science at George Mason University. Three other sections of the report include an anadromous fish study of Cameron Run, a study of the incidence of PCB's and endocrine disrupting chemicals in Hunting Creek, and a survey of *Escherichia coli* levels in the Hunting Creek area of the tidal Potomac River.

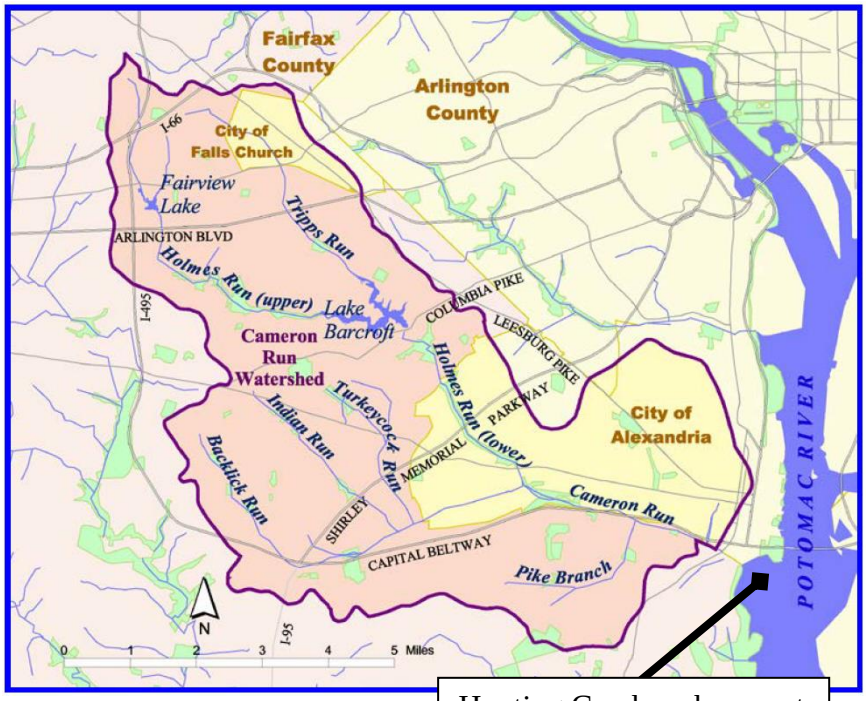
This work was in response to a request from Karen Pallansch, Chief Executive Officer of Alexandria Renew Enterprises (Alex Renew), operator of the wastewater reclamation and reuse facility (WRRF) which serves about 350,000 people in the City of Alexandria and the County of Fairfax in northern Virginia. The study is patterned on the long-running Gunston Cove Study which PEREC has been conducting in partnership with the County of Fairfax Department of Public Works and Environmental Services since 1984. The goal of these projects is to provide baseline and on-going trend analysis of the ecosystems receiving reclaimed water from wastewater treatment facilities with the objective of adaptive management of these valuable freshwater resources. This will facilitate the formulation of well-grounded management strategies for maintenance and improvement of water quality and biotic resources in the tidal Potomac. A secondary but important educational goal is to provide training for Mason graduate and undergraduate students in water quality and biological monitoring and assessment.

Setting of Hunting Creek

Hunting Creek is an embayment of the tidal Potomac River located just downstream of the City of Alexandria and the Woodrow Wilson Bridge. Waters are shallow with the entire embayment having a depth of 2 m or less at mean tide. According to the "Environmental Atlas of the Potomac Estuary" (Lippson et al. 1981), the mean depth of Hunting Creek is 1.0 m, the surface area is 2.26 km^2, and the volume of 2.1 x 10^6 m^3.



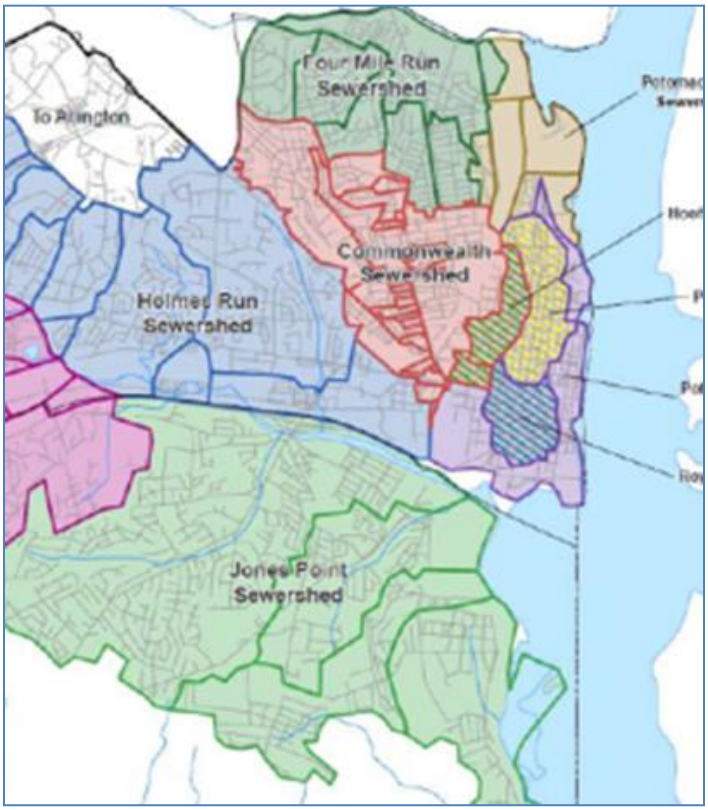
On the left is the Hunting Creek embayment. The Woodrow Wilson Bridge spans the tidal Potomac River at the top of the map. The Potomac River main channel is the whitish area running from north to south through the middle of the map. Soundings (numbers on the map) are in feet at mean low water. For the purposes of this report "Hunting Creek" will extend to the head of tide, roughly to Telegraph Rd.



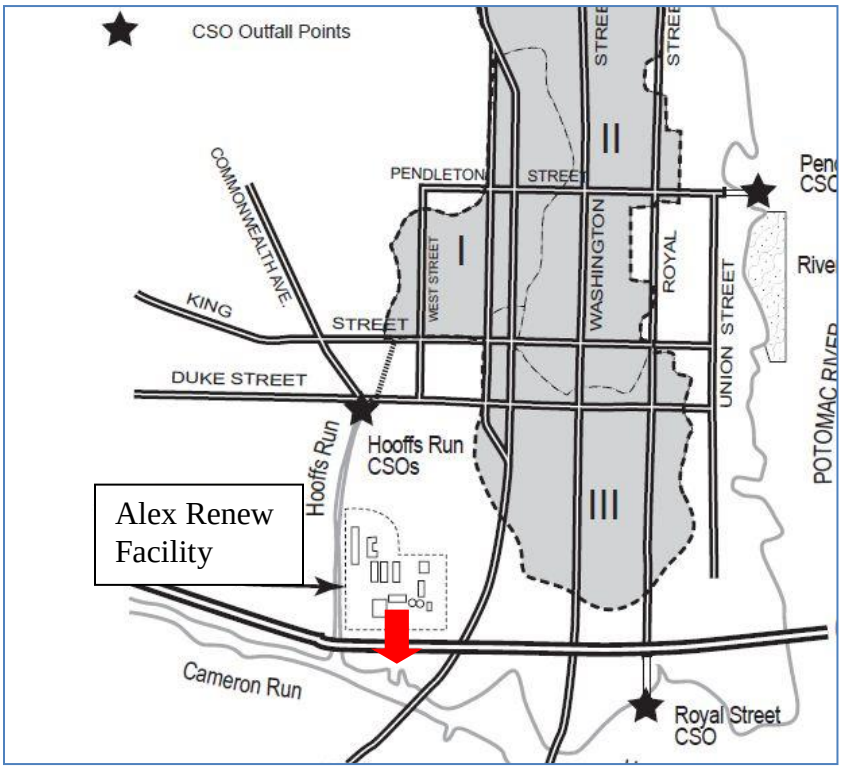
Hunting Creek embayment

On the left is a map of the Hunting Creek watershed. Cameron Run is the freshwater stream which drains the vast majority of the watershed of Hunting Creek. The watershed is predominantly suburban in nature with areas of higher density commercial and residential development. The watershed has an area of 44 square miles and drains most of the Cities of Alexandria and Falls Church and much of east central Fairfax County. A major aquatic feature of the watershed is Lake Barcroft. The suburban land uses in the watershed are a source of nonpoint pollution to Hunting Creek.

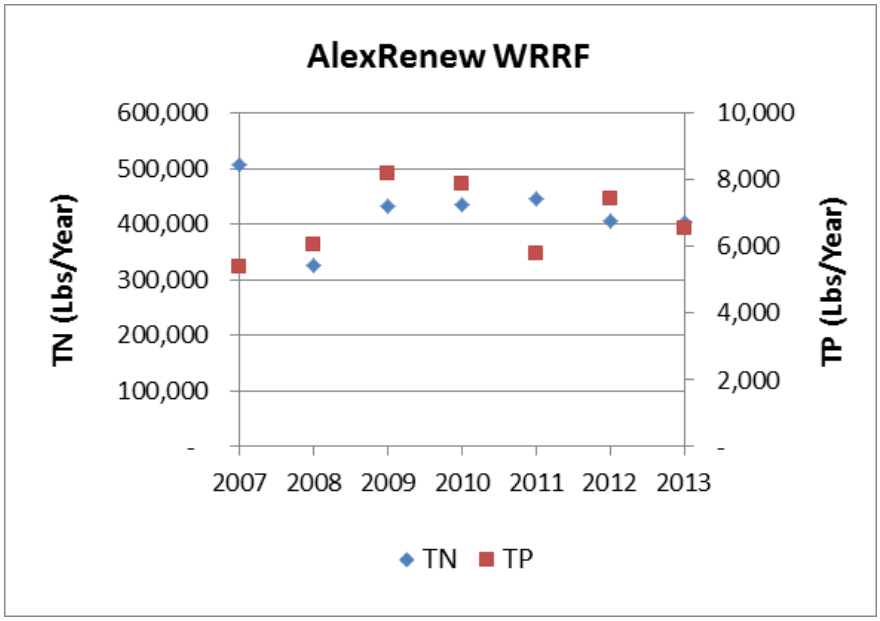
The Alex Renew WRRF serves an area similar in extent to the Cameron Run watershed with the addition of some areas along the Potomac shoreline from Four Mile Run to Dyke Marsh. The effluent of the Alexandria Renew Enterprises plant enters the upper tidal reach of Hunting Creek under the Rt 1/I-95 interchange.



The map at the left shows the sewersheds which contribute to the AlexRenew WRRF. Of particular note are the shaded areas within the City of Alexandria. These sewersheds (Hooff Run, Pendleton, and Royal St.) all contain combined sewers meaning that domestic wastewater is co-mingled with street runoff. Under most conditions, all of this water is directed to the AlexRenew WRRF for treatment. But in extreme runoff conditions (like torrential rains), some may be diverted directly into the tidal Potomac via a Combined Sewer outfall (CSO).



The map at the left is an enlargement of the area where the Alex Renew WRRF is found and where the discharge sites of the CSO's are located. Note the close proximity of two of the CSO's to the Alex Renew WRRF discharge (shown as red arrow).



The graph at the left shows the loading of nitrogen and phosphorus from the Alexandria Renew WRRF for the last seven years. Loadings of both nutrient elements have remained fairly constant at about 400,000 lb/yr (181,000 kg/yr) for nitrogen and 7,000 lb/yr (3175 kg/yr) for phosphorus.

Ecology of the Freshwater Tidal Potomac

The tidal Potomac River is an integral part of the Chesapeake Bay tidal system and at its mouth the Potomac is contiguous to the bay proper. The tidal Potomac is often called a subestuary of the Chesapeake Bay and as such it is the largest subestuary of the bay in terms of size and amount of freshwater input. The mixing of freshwater with saltwater is the hallmark of an estuary. While the water elevation in an estuary is "sea level", the water contained in an estuary is not pure sea water such as found in the open ocean. Pure ocean sea water has a salt concentration of about 35 parts per thousand (ppt). Water in Chesapeake Bay ranges from about 30 ppt near its mouth to 0 ppt in the upper reaches where there is substantial freshwater inflow such as in the upper tidal Potomac River. Salinity at a given location is determined by the balance between freshwater input and salt water mixing in from the ocean. It generally varies with season being lower in spring when freshwater inflows are greater and higher in summer when there is less freshwater inflow. In the Hunting Creek study area, the salinity is essentially 0 yearround.

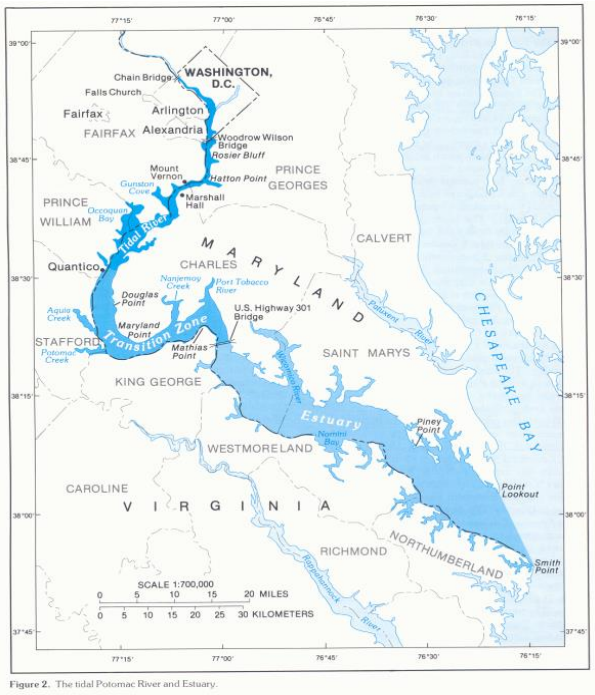


Figure 2. The tidal Potomac River and Estuary.

(map courtesy USGS)

The tidal Potomac is generally divided into three salinity zones as indicated by the map to the left:
-Estuarine or Mesohaline zone (6-14 ppt)
-Transition or Oligohaline zone (0.5-6 ppt)
-Tidal River or Tidal Fresh zone (<0.5 ppt)
Hunting Creek is in the upper part of the Tidal River/Tidal Fresh zone and as such it never experiences detectable salinity

Within the tidal freshwater zone, the flora and fauna are generally characterized by the same species that would occur in a freshwater lake in this area and the food web is similar. Primary producers are freshwater species of submersed aquatic vegetation (SAV) such as native taxa *Vallisneria americana* (water celery), *Potomogeton* spp, (pondweeds), and *Ceratophyllum* (coontail) as well as introduced species such as *Hydrilla verticallata* (hydrilla) and *Myriophyllum spicatum* (water milfoil). Historical accounts indicate that most of the shallow areas of the tidal freshwater Potomac were colonized by SAV around 1900 (Carter et al. 1985).

The other group of important primary producers are phytoplankton, a mixed assemblage of algae and cyanobacteria which may turn over rapidly on a seasonal basis. The dominant

groups of phytoplankton in the tidal freshwater Potomac are diatoms (considered a good food source for aquatic consumers) and cyanobacteria (considered a less desirable food source for aquatic consumers. For the latter part of the 20th century, the high nutrient loadings into the river favored cyanobacteria over both diatoms and SAV resulting in large production of undesirable food for consumers. In the last decade or so, as nutrient reductions have become manifest, cyanobacteria have decreased and diatoms and SAV have increased.

The biomass contained in the cells of phytoplankton nourishes the growth of zooplankton and benthic macroinvertebrates which provide an essential food supply for the juvenile and smaller fish. These in turn provide food for the larger fish like striped bass and largemouth bass. The species of zooplankton and benthos found in the tidal fresh zone are similar to those found in lakes in the area, but the fish fauna is augmented by species that migrate in and out from the open interface with the estuary.

Resident fish species include typical lake species such as sunfish (*Lepomis* spp.), bass (*Micropterus* spp.), and crappie (*Pomoxis* spp.) as well as estuarine species such as white perch (*Morone americana*) and killifish (*Fundulus* spp.). Species which spend part of their year in the area include striped bass (*Morone saxitilis*) and river herrings and shad (*Alosa* spp.). Non-native fish species have also become established in the tidal freshwater Potomac such as northern snakehead (*Channa argus*) and blue catfish (*Ictalurus furcatus*).

Larval fishes are transitional stages in the development of juvenile fishes. They range in development from newly hatched, embryonic fish to juvenile fish with morphological features similar to those of an adult. Many fishes such as clupeids (herring family), white perch, striped bass, and yellow perch disperse their eggs and sperm into the open water. The larvae of these species are carried with the current and termed “ichthyoplankton”. Other fish species such as sunfish and bass lay their eggs in “nests” on the bottom and their larvae are rare in the plankton.

After hatching from the egg, the larva draws nutrition from a yolk sack for a few days time. When the yolk sack diminishes to nothing, the fish begins a life of feeding on other organisms. This post yolk sack larva feeds on small planktonic organisms (mostly small zooplankton) for a period of several days. It continues to be a fragile, almost transparent, larva and suffers high mortality to predatory zooplankton and juvenile and adult fishes of many species, including its own. When it has fed enough, it changes into an opaque juvenile, with greatly enhanced swimming ability. It can no longer be caught with a slow-moving plankton net, but is soon susceptible to capture with the seine or trawl net.

METHODS

A. Profiles and Plankton: Sampling Day

Sampling was conducted on a semimonthly basis at stations representing both Hunting Creek and the Potomac mainstem (Figure 1). One station (AR 1) was located near the mouth of Cameron Run in the small bay located just west of the George Washington Parkway bridge. Sampling was generally conducted at AR 1 from the Parkway bridge. Two stations (AR 2 & 3) were located in the Hunting Creek embayment proper. A fourth station was located in the river channel about 100 m upstream from Buoy 90. Dates for sampling as well as weather conditions on sampling dates and immediately preceding days are shown in Table 1. Note that certain dates such as April 21 and July 2 had significant rainfall in days preceding sampling which may have impacted conditions in Hunting Creek due to it shallow nature and relatively large watershed contributing runoff.

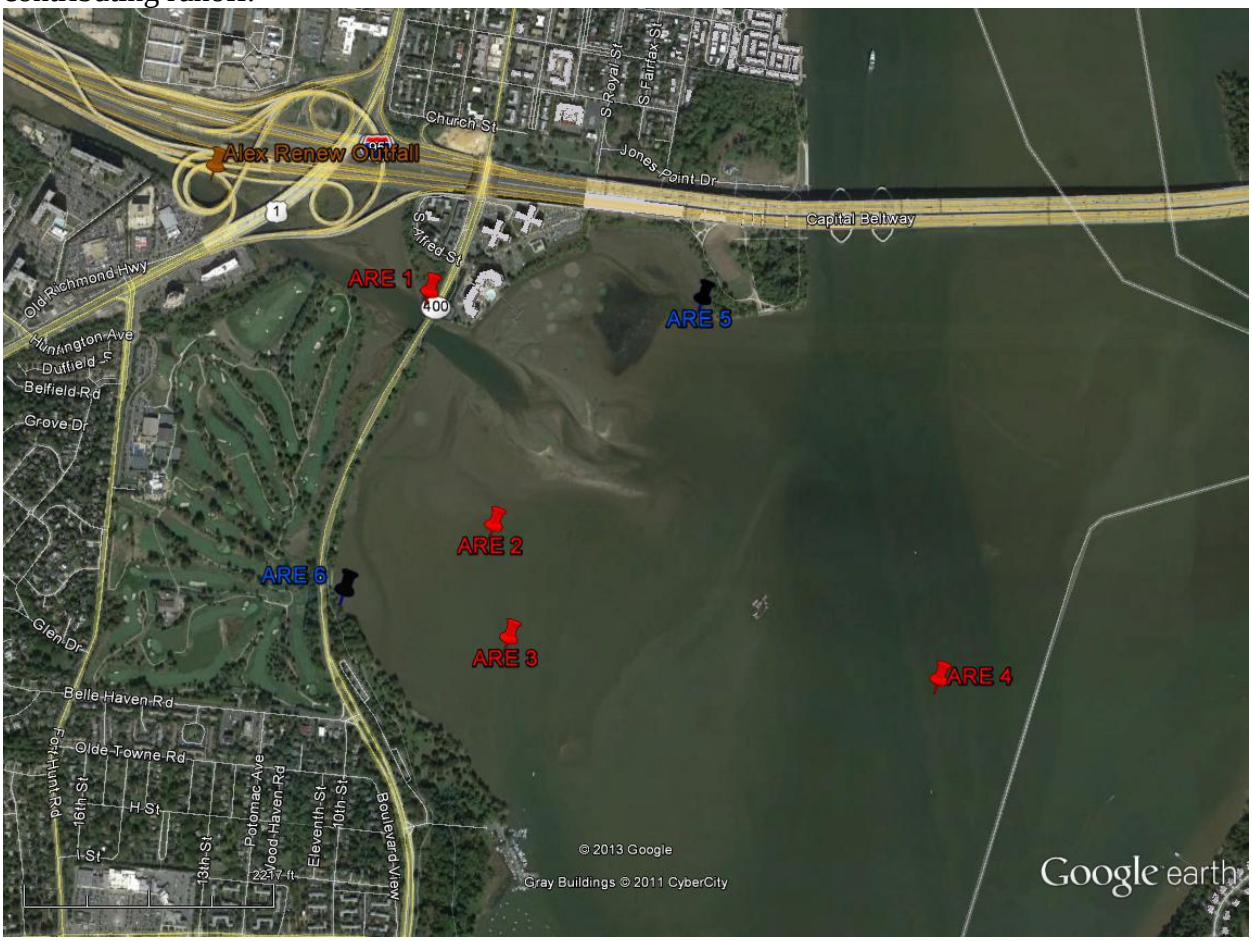


Figure 1. Hunting Creek area of the Tidal Potomac River showing sampling stations. AR 1, 2, 3, and 4 represent water quality stations, AR 2 and 4 are the phytoplankton and zooplankton stations, AR 3 and 4 are the fish trawl stations, and AR 5 and 6 are the fish seine stations.

Table 1
Hunting Creek Study: Sampling Dates and Weather Data for 2015

	Type of Sampling					Avg Daily Temp (°C)		Precipitation (cm)	
Date	WP	B	D	T	S	1-Day	3-Day	1-Day	3-Day
April 21				T	S	17.2	18.3	0.41	3.30
April 22	WP					17.2	18.5	T	2.37
May 5				T	S	23.9	22.0	0.13	0.13
May 6	WP					23.3	22.8	T	0.14
May 20	WP					20.6	25.2	0	1.93
May 27				T	S	28.3	26.3	0.03	0.03
June 5	WP					20.0	17.8	0.08	1.04
June 10				T	S	24.4	25.6	0	1.65
June 18	WP					27.8	28.1	1.35	1.80
June 30				T	S	26.7	24.6	0.03	0.04
July 2	WP					22.8	25.2	0.05	3.38
July 8				T	S	29.4	28.7	2.84	2.90
July 16	WP					25.0	26.7	0	0.04
July 22				T	S	27.2	29.8	0	T
July 30			D			29.4	29.6	1.35	1.35
Aug 3	WP					28.3	28.1	0	T
Aug 12				T	S	25.6	25.9	0	1.24
Aug 17	WP					28.9	27.6	0	0
Aug 19			D			28.9	28.5	0	0
Aug 20				T	S	26.7	27.8	0.43	0.43
Sept 1	WP					28.3	27.0	0	0
Sept 16				T	S	22.8	21.7	0	0
Sept 29	WP					24.4	23.0	4.19	4.20

Type of Sampling: WP: Water quality (samples to AlexRenew Lab), profiles and plankton, B: benthos (station numbers indicated), D: dataflow (water quality mapping), T: fish collected by trawling, S: fish collected by seining. *seining started (one site), but not completed due to boat problems; completed Aug 8.
T under Precipitation equals “trace”.

Sampling was initiated about 10:00 am. Four types of measurements or samples were obtained at each station: (1) depth profiles of temperature, conductivity, dissolved oxygen, pH, and irradiance (photosynthetically active radiation) measured directly in the field; (2) water samples for GMU lab determination of chlorophyll *a* and phytoplankton species composition and abundance; (3) water samples for determination of N and P forms, BOD, COD, alkalinity, hardness, suspended solids, chloride, and pH by the Alexandria Renew Enterprises lab; (4) net sampling of zooplankton and ichthyoplankton.

Profiles of temperature, conductivity, and dissolved oxygen were conducted at each station using a YSI 6600 datasonde with temperature, conductivity, dissolved oxygen and pH probes. Measurements were taken at 0.3 m increments from surface to bottom at the embayment stations. In the river measurements were made with the sonde at depths of 0.3 m and 2.0 m increments to the bottom. Meters were checked for calibration before and after sampling. Profiles of irradiance (photosynthetically active radiation, PAR) were collected with a LI-COR underwater flat scalar PAR probe. PAR measurements were taken at 10 cm intervals to a depth of 1.0 m. Simultaneous measurements were made with a terrestrial probe in air during each profile to correct for changes in ambient light if needed. Secchi depth was also determined. The readings of at least two crew members were averaged due to variability in eye sensitivity among individuals. If the Secchi disk was still visible at the bottom or if its path was block by SAV while still visible, a proper reading could not be obtained.

A 1-liter depth-composited sample for GMU lab work was constructed from equal volumes of water collected at each of three depths (0.3 m below the surface, middepth, and 0.3 m off of the bottom) using a submersible bilge pump. A 100-mL aliquot of this sample was preserved immediately with acid Lugol's iodine for later identification and enumeration of phytoplankton at stations AR2 and AR4. The remainder of the sample was placed in an insulated cooler with ice. A separate 1-liter surface sample was collected from 0.3 m using the submersible bilge pump and placed in the insulated cooler with ice for lab analysis of surface chlorophyll *a*.

Separate 2-liter samples were collected monthly at each station from just below the surface (0.3 m) and near the bottom (0.3 m off bottom) at each station using the submersible pump. This water was promptly delivered to the nearby Alexandria Renew Laboratory for determination of nitrogen, phosphorus, BOD, TSS, VSS, pH, total alkalinity, and chloride.

At stations AR2 and AR4, microzooplankton was collected by pumping 32 liters from each of three depths (0.3 m, middepth, and 0.3 m off the bottom) through a 44 µm mesh sieve. The sieve consisted of a 12-inch long cylinder of 6-inch diameter PVC pipe with a piece of 44 µm nitex net glued to one end. The 44 µm cloth was backed by a larger mesh cloth to protect it. The pumped water was passed through this sieve from each depth and then the collected microzooplankton was backflushed into the sample bottle. The resulting sample was treated with about 50 mL of club soda and then preserved with formalin containing a small amount of rose bengal to a concentration of 5-10%.

At stations AR2 and AR4, macrozooplankton was collected by towing a 202 µm net (0.3 m opening, 2 m long) for 1 minute at each of three depths (near surface, middepth, and

near bottom). Ichthyoplankton (larval fish) was sampled by towing a 333 µm net (0.5 m opening, 2 m long) for 2 minutes at each of the same depths at Stations AR2 and AR4. In the embayment, the boat traveled from AR2 toward AR3 during the tow while in the river the net was towed in a more linear fashion along the channel. Macrozooplankton tows were about 300 m and ichthyoplankton tows about 600 m. Actual distance depended on specific wind conditions and tidal current intensity and direction, but an attempt was made to maintain a constant slow forward speed (approximately 2 miles per hour) through the water during the tow. The net was not towed directly in the wake of the engine. A General Oceanics flowmeter, fitted into the mouth of each net, was used to establish the exact towing distance. During towing the three depths were attained by playing out rope equivalent to about 1.5-2 times the desired depth. Samples which had obviously scraped bottom were discarded and the tow was repeated. Flowmeter readings taken before and after towing allowed precise determination of the distance towed and when multiplied by the area of the opening produced the total volume of water filtered.

Macrozooplankton were preserved immediately with formalin to a concentration of 5-10%. Rose bengal formalin with club soda pretreatment was used for macrozooplankton. Ichthyoplankton was preserved in 70% ethanol. Macrozooplankton was collected on each sampling trip; ichthyoplankton collections ended after July because larval fish were normally not found after this time.

Benthic macroinvertebrate samples were collected monthly at stations AR2, AR3, and AR4. Three samples were collected at each station using a petite ponar grab. The bottom material was sieved through a 0.5 mm stainless steel sieve and resulting organisms were preserved in rose bengal formalin for lab analysis.

Samples for water quality determination were maintained on ice delivered to the Alexandria Renew Enterprises (AlexRenew) Laboratory by 2 pm on sampling day and returned to GMU by 3 pm. At GMU 10-15 mL aliquots of both depth-integrated and surface samples were filtered through 0.45 µm membrane filters (Gelman GN-6 and Millipore MF HAWP) at a vacuum of less than 10 lbs/in^2 for chlorophyll a and pheopigment determination. During the final phases of filtration, 0.1 mL of $MgCO_3$ suspension (1 g/100 mL water) was added to the filter to prevent premature acidification. Filters were stored in 20 mL plastic scintillation vials in the lab freezer for later analysis. Seston dry weight and seston organic weight were measured by filtering 200-400 mL of depth-integrated sample through a pretared glass fiber filter (Whatman 984AH).

Sampling day activities were normally completed by 5:30 pm.

B. Profiles and Plankton: Follow-up Analyses

Chlorophyll *a* samples were extracted in a ground glass tissue grinder to which 4 mL of dimethyl sulfoxide (DMSO) was added. The filter disintegrated in the DMSO and was ground for about 1 minute by rotating the grinder under moderate hand pressure. The ground suspension was transferred back to its scintillation vial by rinsing with 90% acetone. Ground samples were stored in the refrigerator overnight. Samples were removed from the

refrigerator and centrifuged for 5 minutes to remove residual particulates.

Chlorophyll *a* concentration in the extracts was determined fluorometrically using a Turner Designs Model 10 field fluorometer configured for chlorophyll analysis as specified by the manufacturer. The instrument was calibrated using standards obtained from Turner Designs. Fluorescence was determined before and after acidification with 2 drops of 10% HCl. Chlorophyll *a* was calculated from the following equation which corrects for pheophytin interference:

$$\text{Chlorophyll } a\ (\mu g/L) = F_s R_s (R_b - R_a)/(R_s - 1)$$

where F_s=concentration per unit fluorescence for pure chlorophyll *a*
R_s=fluorescence before acid/fluorescence after acid for pure chlorophyll *a*
R_b=fluorescence of sample before acid
R_a=fluorescence of sample after acid

All chlorophyll analyses were completed within one month of sample collection.

Phytoplankton species composition and abundance was determined using the inverted microscope-settling chamber technique (Lund et al. 1958). Ten milliters of well-mixed algal sample were added to a settling chamber and allowed to stand for several hours. The chamber was then placed on an inverted microscope and random fields were enumerated. At least two hundred cells were identified to species and enumerated on each slide. Counts were converted to number per mL by dividing number counted by the volume counted. Biovolume of individual cells of each species was determined by measuring dimensions microscopically and applying volume formulae for appropriate solid shapes.

Microzooplankton and macrozooplankton samples were rinsed by sieving a well-mixed subsample of known volume and resuspending it in tap water. This allowed subsample volume to be adjusted to obtain an appropriate number of organisms for counting and for formalin preservative to be purged to avoid fume inhalation during counting. One mL subsamples were placed in a Sedgewick-Rafter counting cell and whole slides were analyzed until at least 200 animals had been identified and enumerated. A minimum of two slides was examined for each sample. References for identification were: Ward and Whipple (1959), Pennak (1978), and Rutner-Kolisko (1974). Zooplankton counts were converted to number per liter (microzooplankton) or per cubic meter (macrozooplankton) with the following formula:

$$\text{Zooplankton } (\#/L \text{ or } \#/m^3) = NV_s/(V_c V_f)$$

where N = number of individuals counted
V_s = volume of reconstituted sample, (mL)
V_c = volume of reconstituted sample counted, (mL)
V_f = volume of water sieved, (L or m^3)

Larval fish were picked from the ethanol-preserved ichthyoplankton samples with the aid of a stereo dissecting microscope. Identification of ichthyoplankton was made to family

and further to genus and species where possible. If the number of animals in the sample exceeded several hundred, then the sample was split with a plankton splitter and the resulting counts were multiplied by the subsampling factor. The works Hogue et al. (1976), Jones et al. (1978), Lippson and Moran (1974), and Mansueti and Hardy (1967) were used for identification. The number of ichthyoplankton in each sample was expressed as number per 10 m^3 using the following formula:

Ichthyoplankton (#/10m^3) = 10N/V
where N = number ichthyoplankton in the sample
V = volume of water filtered, (m^3)

C. Adult and Juvenile Fish

Fishes were sampled by trawling at stations AR3 and AR4, and seining at stations AR5 and AR6 (Figure 1). For trawling, a try-net bottom trawl with a 15-foot horizontal opening, a ¾ inch square body mesh and a ¼ inch square cod end mesh was used. The otter boards were 12 inches by 24 inches. Towing speed was 2-3 miles per hour and tow length was 5 minutes. The trawls were towed upriver parallel to the channel at AR4, and following the curve of the 'cove' away from the channel at AR3. The direction of tow should not be crucial. Dates of sampling and weather conditions are found in Table 1.

Seining was performed with a bag seine that was 50 feet long, 3 feet high, and made of knotted nylon with a ¼ inch square mesh. The bag is located in the middle of the net and measures 3 ft^3. The seining procedure was standardized as much as possible. The net was stretched out perpendicular to the shore with the shore end right at the water line. The net was then pulled parallel to the shore for a distance of 100 feet by a worker at each end moving at a slow walk. Actual distance was recorded if in any circumstance it was lower than 100 feet. At the end of the prescribed distance, the offshore end of the net was swung in an arc to the shore and the net pulled up on the beach to trap the fish. Dates for seine sampling were the same as those for trawl sampling (Table 1).

After the catch from each of these two gear types was hauled in, the fishes were measured for standard length and total length to the nearest mm. Standard length is the distance from the front tip of the snout to the end of the vertebral column and base of the caudal fin. This is evident in a crease perpendicular to the axis of the body when the caudal fin is pulled to the side. Total length is the distance from the tip of the snout to the tip of the longer lobe of the caudal fin, measured by straightening the longer lobe toward the midline.

If the identification of the fish was not certain in the field, a specimen was preserved in 70% ethanol and identified later in the lab. Fishes kept for chemical analysis were kept on ice wrapped in aluminum foil until frozen in the lab. All fishes retained for laboratory analysis or identification were first euthanized by submerging them in an ice sludge conforming AICUC protocol. Identification was based on characteristics in dichotomous keys found in several books and articles, including Jenkins and Burkhead (1983), Hildebrand and Schroeder (1928), Loos et al (1972), Dahlberg (1975), Scott and Crossman (1973), Bigelow and Schroeder (1953), Eddy and Underhill (1978), Page and Burr (1998), and

Douglass (1999).

D. Submersed Aquatic Vegetation

Data on coverage and composition of submersed aquatic vegetation (SAV) are generally obtained from the SAV webpage of the Virginia Institute of Marine Science (http://www.vims.edu/bio/sav). Information on this web site was obtained from aerial photographs near the time of peak SAV abundance as well as ground surveys which were used to determine species composition.

E. Benthic Macroinvertebrates

Benthic macroinvertebrates were sampled monthly using a petite ponar sampler at AR2, AR3, and AR4. Triplicate samples were collected at each station monthly. Bottom samples were sieved on-site through a 0.5 mm stainless steel sieve and preserved with rose bengal formalin. In the laboratory benthic samples were rinsed with tap water through a 0.5 mm sieve to remove formalin preservative and resuspended in tap water. All organisms were picked, sorted, identified and enumerated.

F. Water Quality Mapping (Dataflow)

On two additional dates in 2015 (July 30 and August 19) *in situ* water quality mapping was conducted by slowly transiting through much of the Hunting Creek study area as water was pumped through a chamber containing a YSI 6600 sonde equipped with temperature, specific conductance, dissolved oxygen, pH, turbidity, and chlorophyll probes. Readings were recorded at 15 second intervals along with simultaneous GPS position readings. Every 2 minutes water samples were collected for chlorophyll and turbidity calibration. Some areas of the Hunting Creek embayment could not be surveyed due to shallow water or heavy SAV growth. These surveys allowed a much better understanding of spatial patterns in water quality within the Hunting Creek area which facilitated interpretation of data from the fixed stations. This approach is in wide use in the Chesapeake Bay region by both Virginia and Maryland under the name “dataflow”.

G. Data Analysis

Data for each parameter were entered into spreadsheets (Excel or SigmaPlot) for graphing of temporal and spatial patterns. SYSTAT was used for statistical calculations and to create illustrations of the water quality mapping cruises. JMP v8.0.1was used for fish graphs. Other data analysis approaches are explained in the text.

RESULTS

A. Climatic and Hydrologic Factors

In 2015 air temperature was substantially above average from April through September (Table 2). July was the warmest month, but May was the most above normal. There were 41 days with maximum temperature above 32.2°C (90°F) during 2015 compared with 4 in 2004, 18 in 2005, 29 in 2006, 33 in 2007, 31 in 2008, 16 days in 2009, 62 in 2010, 42 in 2011, 42 in 2012, 27 in 2013, and 20 in 2014. Precipitation was below normal during May, but over three times normal in June. It was slightly above normal in July and well below normal in August and September. The largest daily rainfall totals were all in the very wet month of June: 6.32 cm (2.49 in) on June 1, 6.02 cm (2.37 in) on June 20 and 6.99 cm (2.75 in) on June 27.

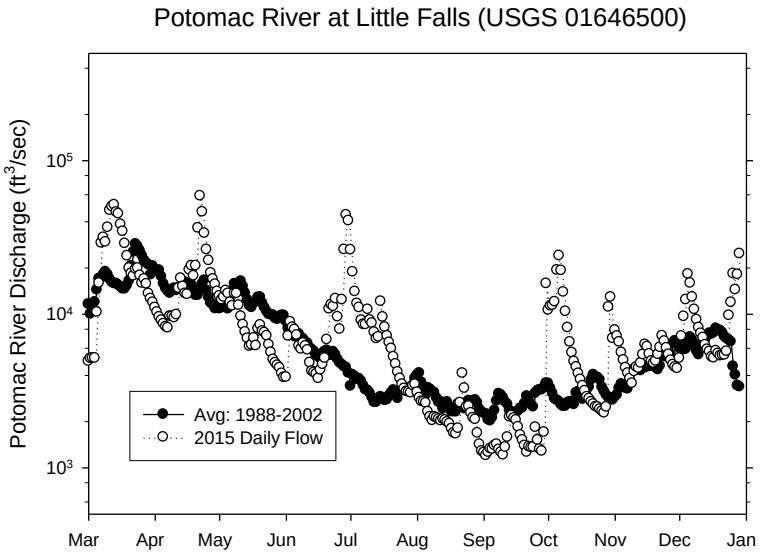
Table 2. Meteorological Data for 2015. National Airport. Monthly Summary.

	Air Temp (°C)		Precipitation (cm)	
MONTH				
March	7.4	(8.1)	10.3	(9.1)
April	15.2	(13.4)	8.7	(7.0)
May	22.9	(18.7)	4.9	(9.7)
June	25.2	(23.6)	30.3	(8.0)
July	27.4	(26.2)	12.7	(9.3)
August	26.3	(25.2)	2.9	(8.7)
September	23.8	(21.4)	5.5	(9.6)
October	14.9	(14.9)	8.9	(8.2)
November	12.0	(9.3)	5.3	(7.7)
December	10.7	(4.2)	12.3	(7.8)

Note: 2014 monthly averages or totals are shown accompanied by long-term monthly averages (1971-2000).
Source: Local Climatological Data. National Climatic Data Center, National Oceanic and Atmospheric Administration.

Table 3. Monthly mean discharge at USGS Stations representing freshwater flow into the study area. (+) 2015 month > 2x Long Term Avg. (-) 2015 month < ½ Long Term Avg.

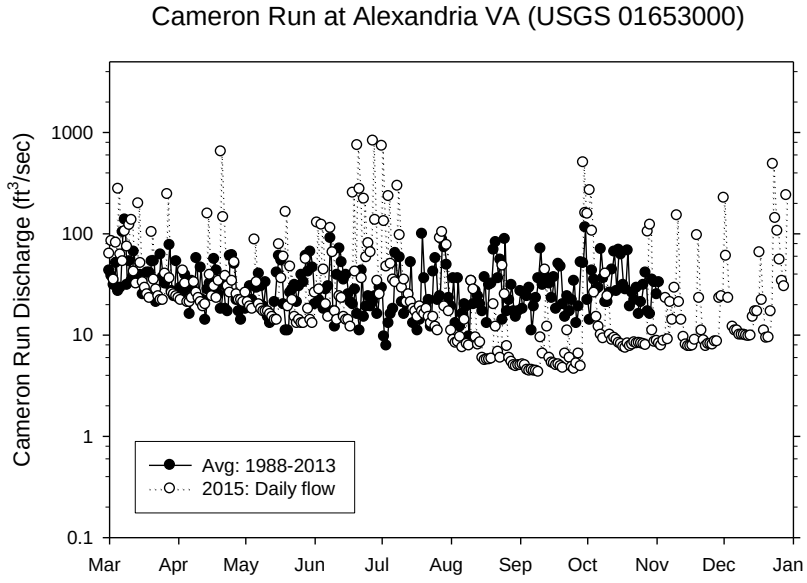
	Potomac River at Little Falls (cfs)		**Cameron Run at Wheeler Ave (cfs)**	
	2015	**Long Term Average**	**2015**	**Long Term Average**
January	7971	13700	41.9	41
February	6149	16600	21.5	45
March	23816	23600	69.4	55
April	18146	20400	56.5	42
May	8526	15000	30.5	41
June	10105	9030	116.1 (+)	38
July	8020	4820	73.4 (+)	31
August	2245	4550	11.3 (-)	28
September	1983 (-)	5040	28.7	38
October	7504	5930	33.9	33



In a tidal freshwater system like the Potomac River, river flow entering from upstream is important in maintaining freshwater conditions and also serves to bring in dissolved and particulate substances from the watershed. High freshwater flows may also flush planktonic organisms downstream and bring in suspended sediments that decrease water clarity. The volume of river flow per unit time is referred to as "river discharge" by hydrologists. Note the general long term seasonal pattern of higher discharges in winter and spring and lower discharges in summer and fall.

Figure 2. Mean Daily Discharge: Potomac River at Little Falls (USGS Data). Month tick is at the beginning of the month.

Potomac River discharge during 2015 was elevated during mid-March, late April, and late June-early July (Table 3, Figure 2). The late June-early July levels were unusual as flow is usually decreasing at that time. Potomac Rive discharge was consistently below average in August and September. Cameron Run flows were about average during April and May. However, in late June and early July, an extended period of flows well above average was observed due to several large rainfall events during this period. Water quality/plankton sampling dates that may have been particularly affected by immediately prior storm events include June 5, June 18 and July 2 (Table 1).



In the Hunting Creek region of the tidal Potomac, freshwater discharge is occurring from both the major Potomac River watershed upstream (measured at Little Falls) and from immediate tributaries, principally Cameron Run which empties directly into Hunting Creek. The gauge on Cameron Run at Wheeler Avenue is located just above the head of tide and covers most area which contributes runoff directly to the Hunting Creek embayment from the watershed. The contributing area to the Wheeler Ave gauge is 33.9 sq mi. (USGS)

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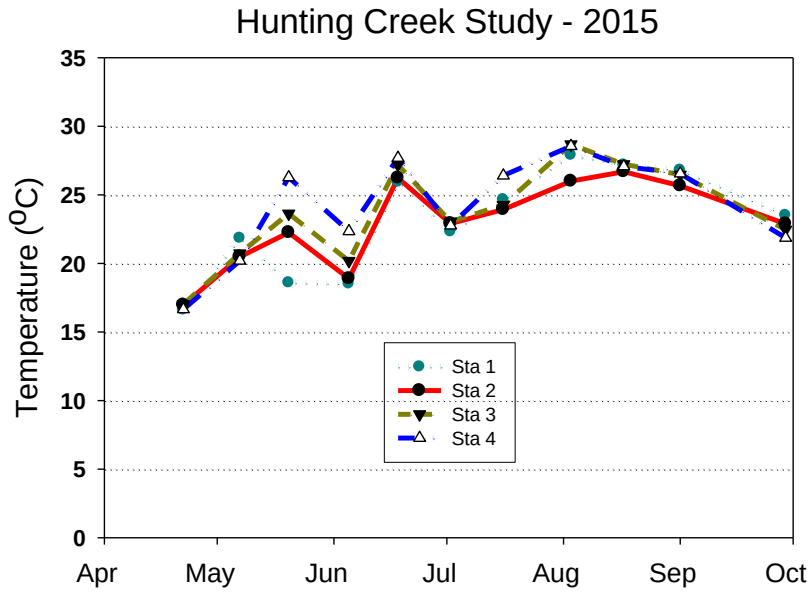
Figure 3. Mean Daily Discharge: Cameron Run at Alexandria (Wheeler Ave) (USGS Data).

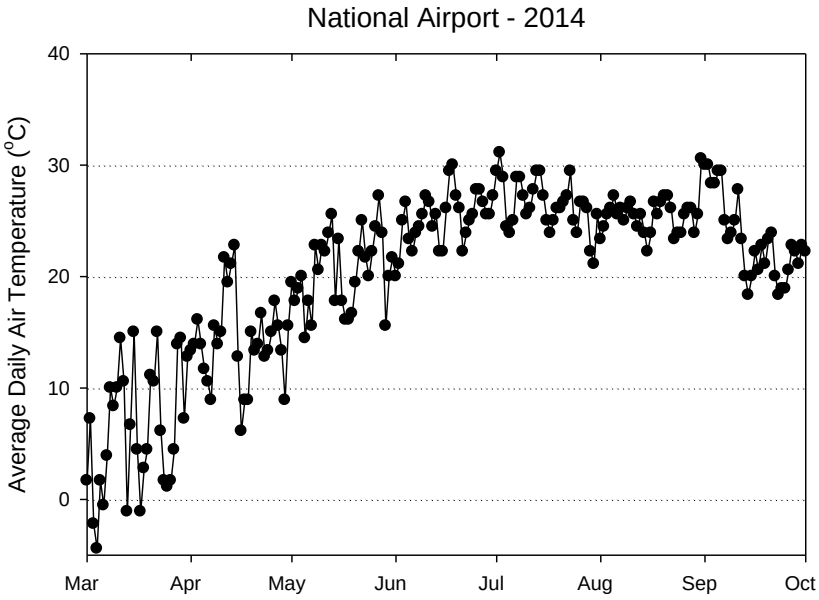
B. Physico-chemical Parameters – 2015



Water temperature is an important factor affecting both water quality and aquatic life. In a well-mixed system like the tidal Potomac, water temperatures are generally fairly uniform with depth. In a shallow mixed system such as the tidal Potomac, water temperature often closely tracks daily changes in air temperature.

Figure 4. Water Temperature (ºC). GMU Field Data. Month tick is at first day of month.

In 2014, water temperature followed the typical seasonal pattern at all stations (Figure 4). Values generally increased from April through early August, but there were a couple of reversals in early June and early July. In each case these followed a cold front which knocked air temperatures down to 20-22°C on sampling days. Maximum temperatures were between 25 and 30°C at all sites and were generally present from late June to early August. Mean daily air temperature showed similar patterns (Figure 5)



Mean daily air temperature (Figure 5) was a good predictor of water temperature (Figure 4).

Figure 5. Average Daily Air Temperature (ºC) at Reagan National Airport.

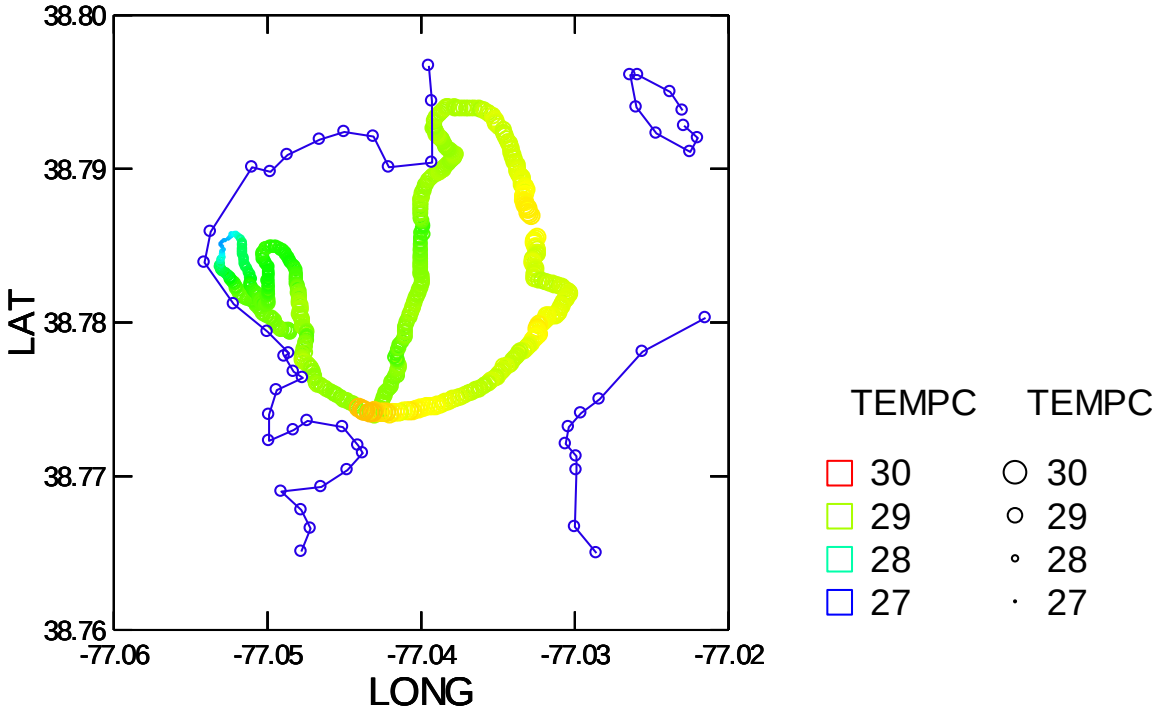


Figure 6a. Water Quality Mapping. July 30, 2015. Temperature (°C).

Mapping of water temperature was conducted on two dates in 2015: July 30 and August 19. In July temperatures were noticably cooler in Hunting Creek than in areas nearer the river mainstem whereas in August temperatures were higher in the shallow portions of Hunting Creek (Figure 6a&b).

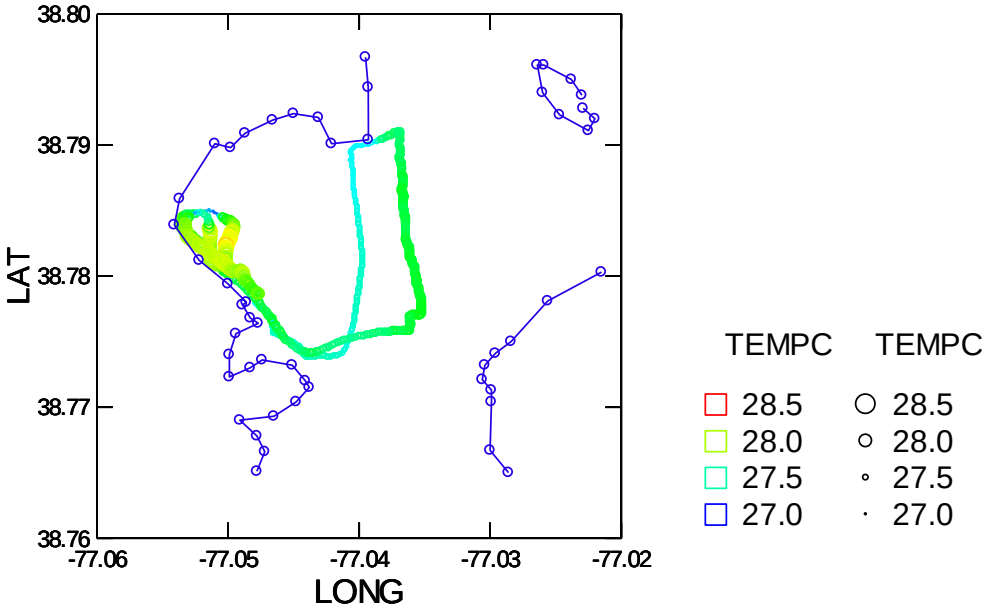
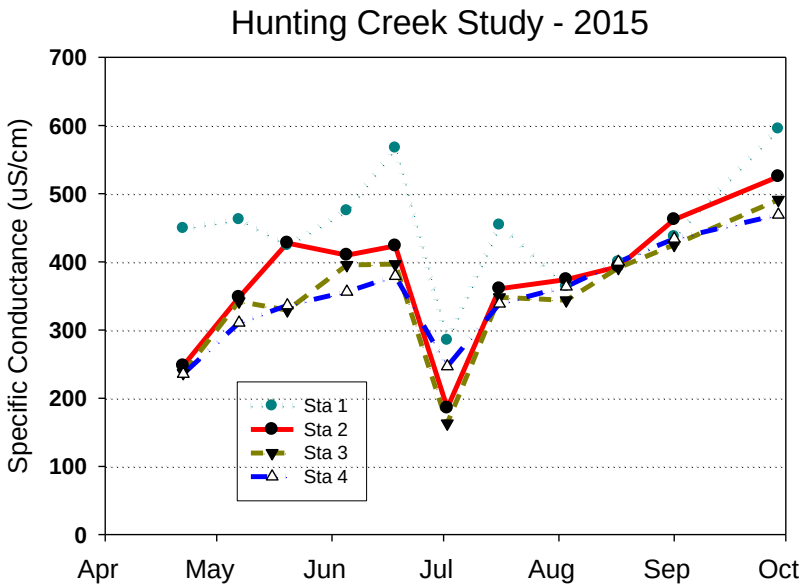


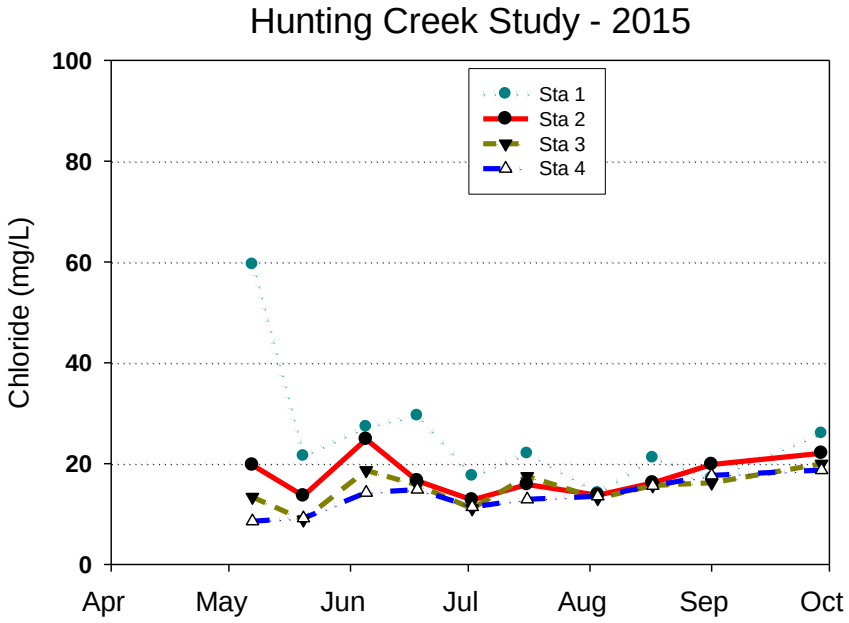
Figure 6b. Water Quality Mapping. August 19, 2015. Temperature (°C).



Specific conductance measures the capacity of the water to conduct electricity standardized to 25°C. This is a measure of the concentration of dissolved ions in the water. In freshwater, conductivity is relatively low. Ion concentration generally increases slowly during periods of low freshwater inflow and decreases during periods of high freshwater inflow. Sewage treatment facilities can be a source of elevated conductivity. In winter road salts can be a major source of conductivity in urban streams.

Figure 7. Specific Conductance (µS/cm). GMU Field Data. Month tick is at first day of month.

During most of 2015, specific conductance (Figure 7) showed a general pattern of slow increase with time over the entire study area. A major exception to this pattern occurred in early July with a very marked decline in specific conductance being observed at all stations. The corresponded to major increases in runoff in both the Potomac mainstream and in the Cameron Run tributary. Chloride (Figure 8) was elevated in April and generally slightly higher at AR1 consistent with their closer proximity to the AlexRenew outfall. Only a minor decrease was found in chloride associated with the early July flow event.



Chloride ion (Cl-) is a principal contributor to conductance. Major sources of chloride in the study area are sewage treatment plant discharges, road salt, and brackish water from the downriver portion of the tidal Potomac. Chloride concentrations observed in the Hunting Creek area are very low relative to those observed in brackish, estuarine, and coastal areas of the Mid-Atlantic region. Chloride may increased slightly in late summer or fall when brackish water from down estuary may reach the area as freshwater discharge declines.

Figure 8. Chloride (mg/L). Alexandria Renew Lab Data. Month tick is at first day of month.

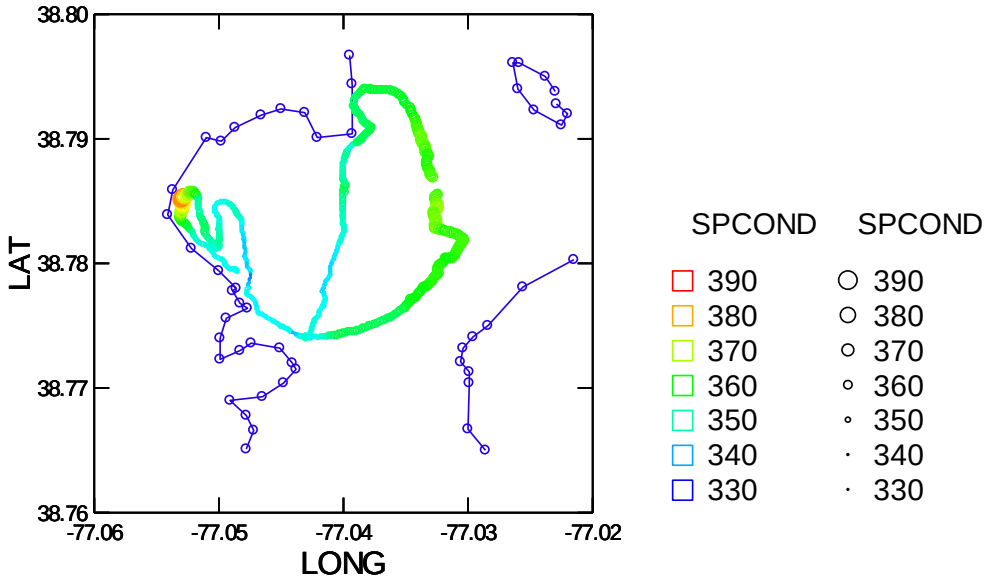


Figure 9a. Water Quality Mapping. July 30, 2015. Specific conductance (µS).

Mapping of specific conductance on both dates showed highest values along the Hunting Creek shoreline (Figure 9a&b). This was consistent with the proximity of this area to inflows from Cameron Run and Alex Renew. The other area of somewhat elevated conductivity values is the river mainstem. This is probably due to the influence of Blue Plains effluent from immediately upstream.

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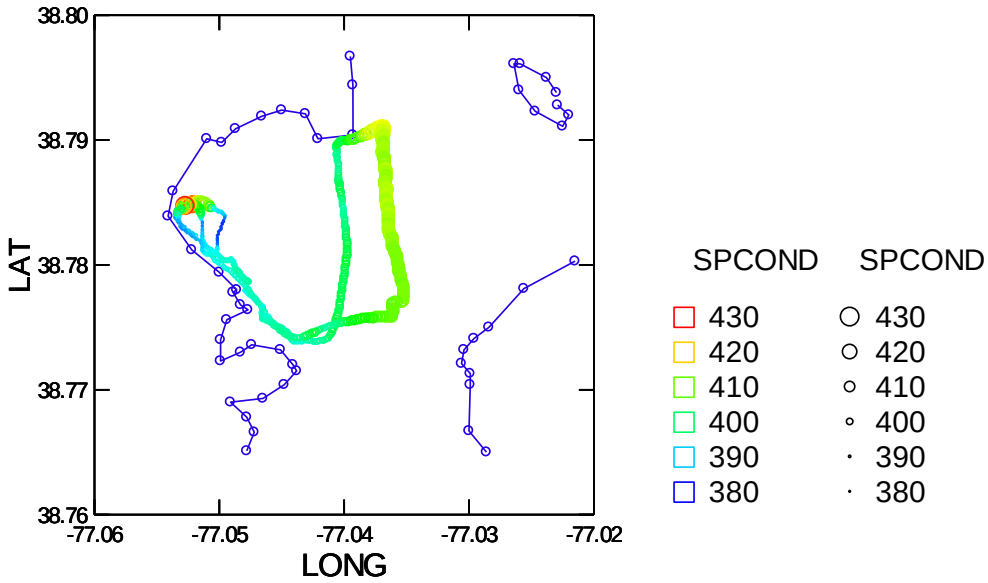
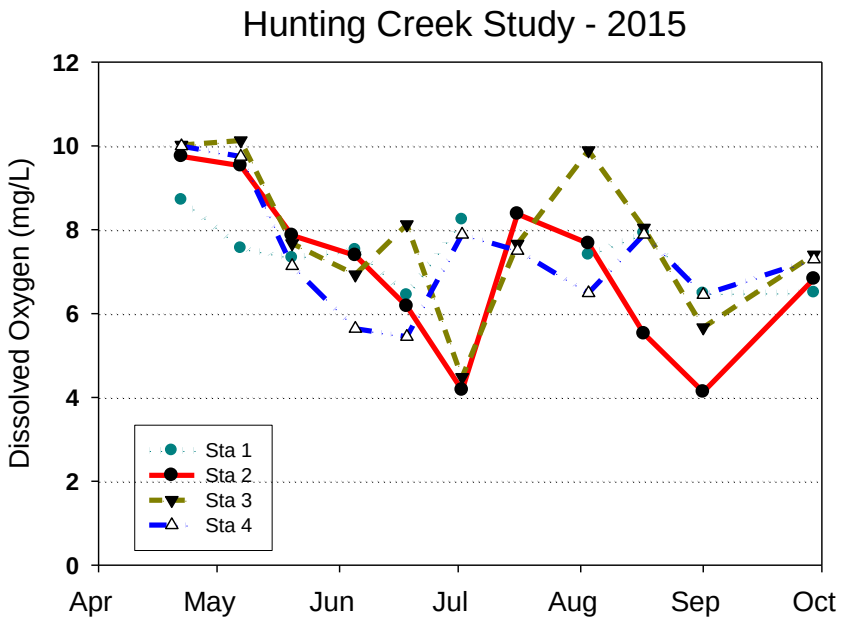


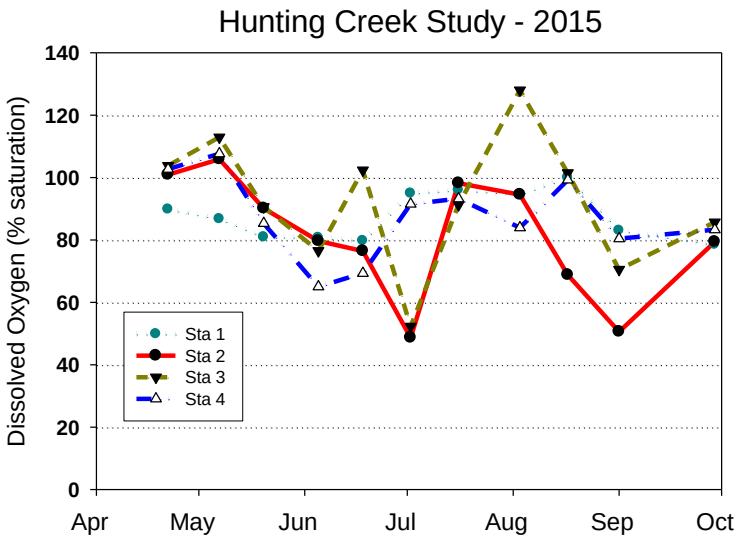
Figure 9b. Water Quality Mapping. August 19, 2015. Specific conductance (µS).



Oxygen dissolved in the water is required by freshwater animals for survival. The standard for dissolved oxygen (DO) in most surface waters is 5 mg/L. Oxygen concentrations in freshwater are in balance with oxygen in the atmosphere, but oxygen is only weakly soluble in water so water contains much less oxygen than air. This solubility is determined by temperature with oxygen more soluble at low temperatures.

Figure 10. Dissolved Oxygen (mg/L). GMU Field Data. Month tick is at first day of month.

The general pattern for dissolved oxygen (mg/L) was a seasonal decline from April through June and steady values for the rest of the year (Figure 10). Values at AR1 and AR4 increased in early July whereas at AR 2 and AR 3 there were distinct declines. Looking at DO as percent saturation (Figure 11), the seasonal pattern was less pronounced due to temperature changes in saturation capacity of water. The high value of about 130% observed at AR 3 in early August indicates high rates of photosynthesis due to the thick beds of SAV at this site.



The temperature effect on oxygen concentration can be removed by calculating DO as percent saturation. This allows examination of the balance between photosynthesis and respiration both of which also impact DO. Photosynthesis adds oxygen to the water while respiration removes it. Values above 120% saturation are indicative of intense photosynthesis while values below 80% reflect a preponderance of respiration or decomposition.

Figure 11. Dissolved Oxygen (% saturation). GMU Field Data. Month tick is at first day.

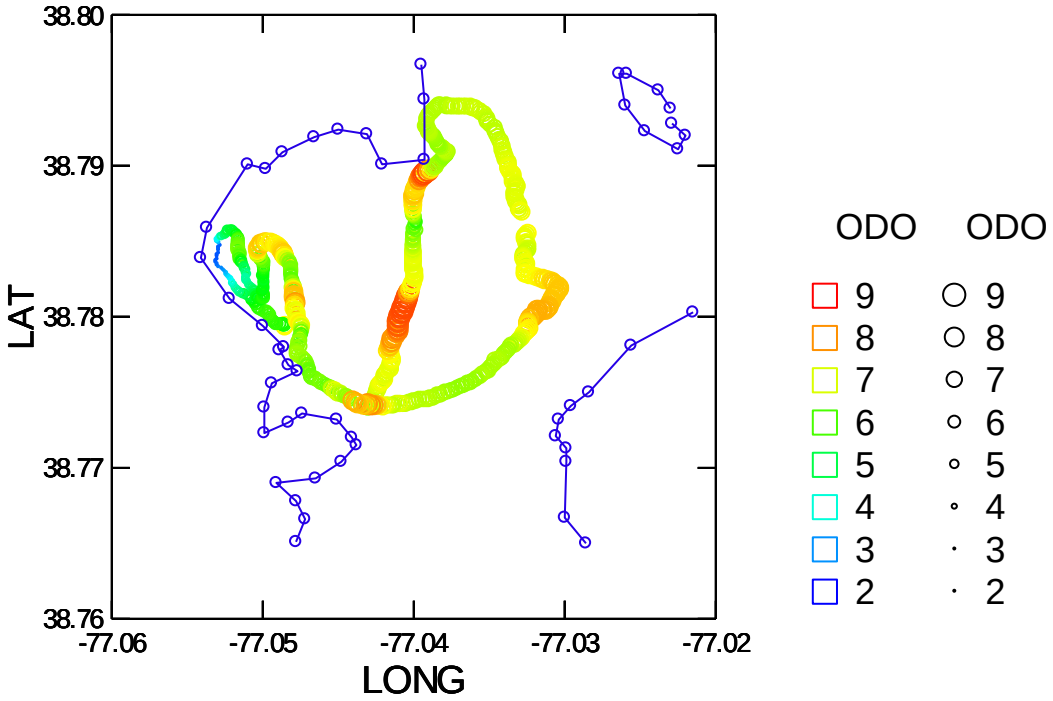


Figure 12a. Water Quality Mapping. July 30, 2015. Dissolved oxygen (mg/L).

Dissolved oxygen was highest along the interface between Hunting Creek and the river mainstem (Figures 12a&b). This was a little surprising given that SAV was considered the dominant source of photosynthesis; one would have expected highest levels in Hunting Creek proper. Part of this may be due to sampling occuring early in the day and the inability to sample directly in the SAV beds.

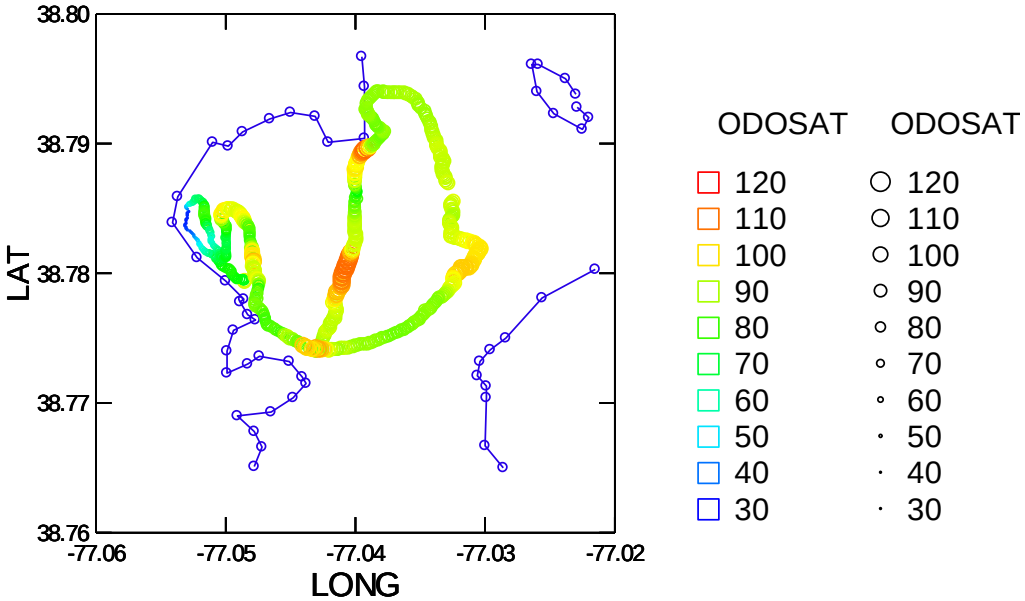


Figure 12b. Water Quality Mapping. July 30, 2015. Dissolved oxygen (percent saturation).

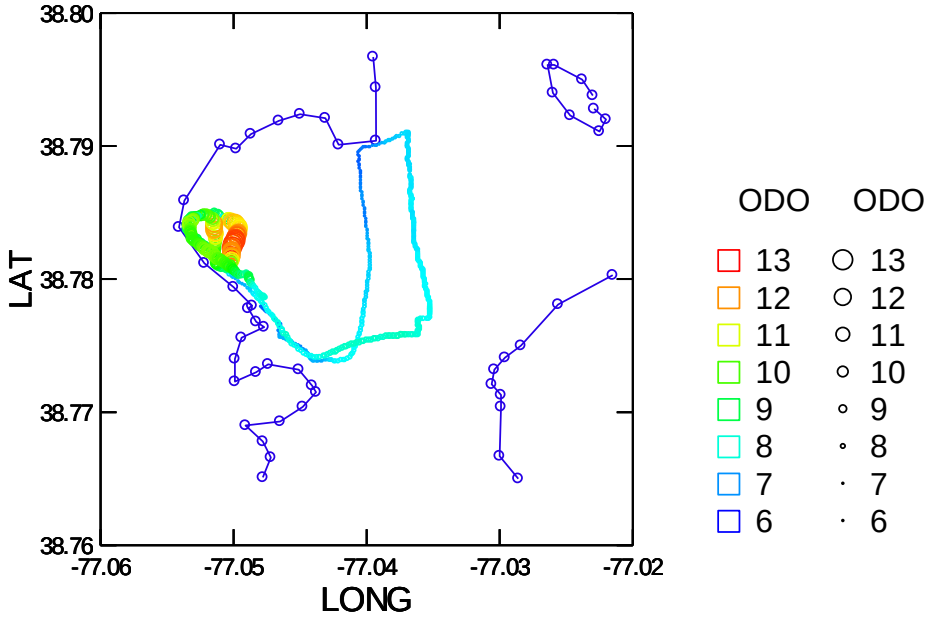


Figure 13a. Water Quality Mapping. August 19, 2015. Dissolved oxygen (mg/L).

In the August data mapping cruise highest levels were observed in the Hunting Creek embayment in shallow water near the SAV beds (Figures 13a & 13b). Values in the channel area were at or very near saturation and clearly lower than in the embayment.

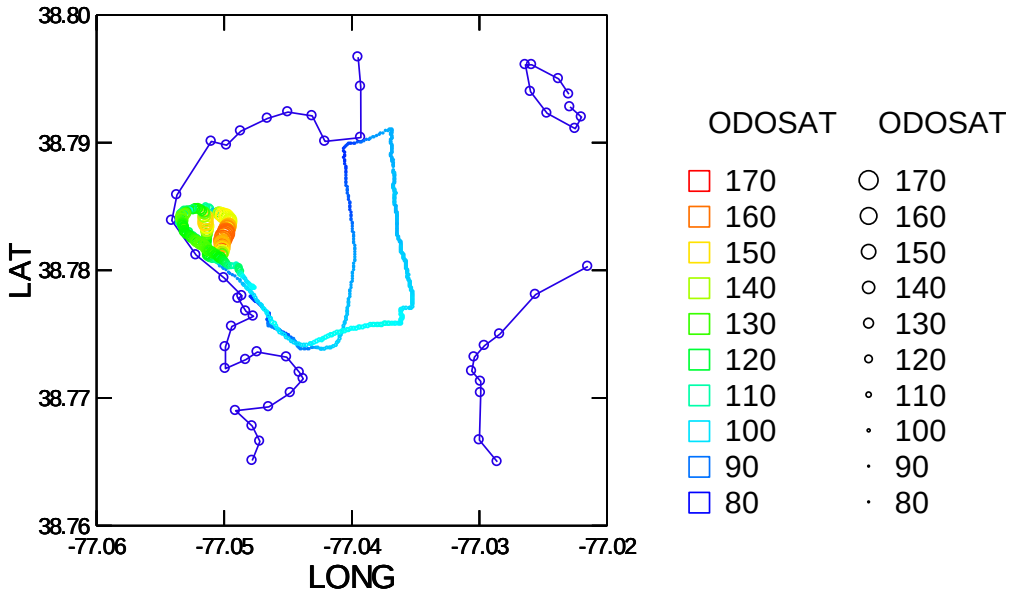


Figure 13b. Water Quality Mapping. August 19, 2015. Dissolved oxygen (percent saturation).

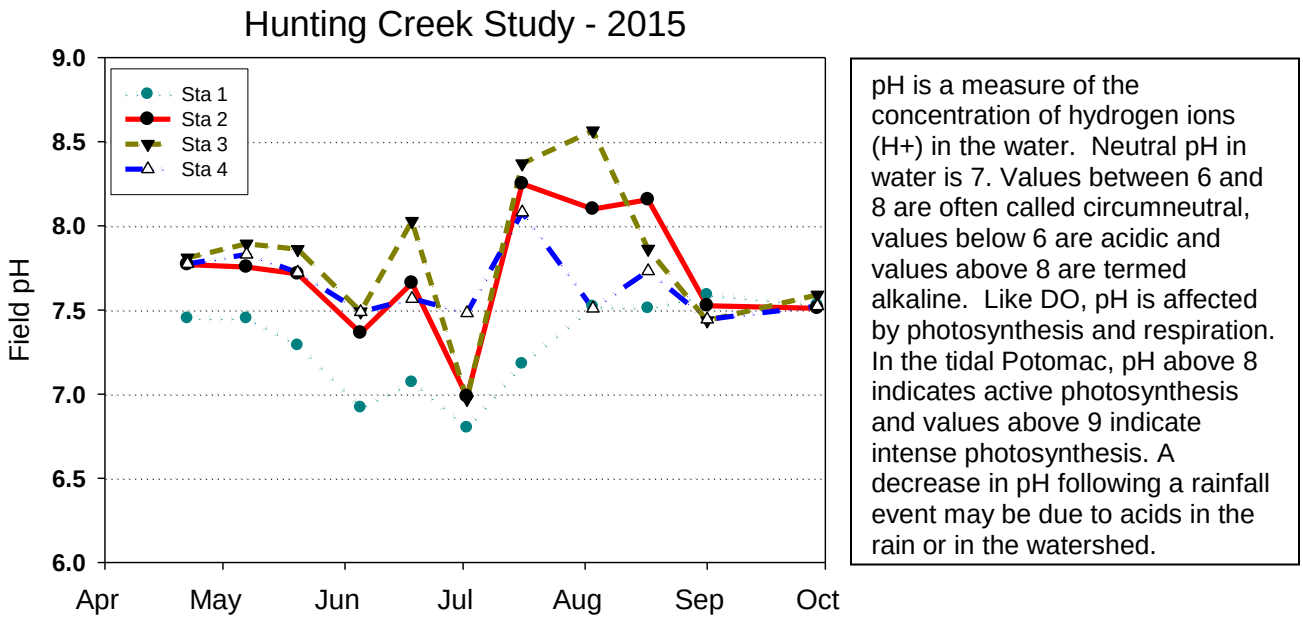


Figure 14. pH. GMU Field Data. Month tick is at first day of month.

Field pH and lab pH showed consistent seasonal and spatial patterns in 2015 (Figure 14, 15). The river mainstem site (AR 4) was fairly constant through time, generally in the 7.5-8.0 range. Values at the upstream site AR1 were generally lower than at the other sites during most of the year; this trend was more marked in the field data. At the two sites in Hunting Creek embayment, AR2 and AR3, there was a strong decline in early July, probably due to the large runoff volumes on that date. A subsequent recovery in late July and early August is attributable to intense photosynthesis from SAV beds.

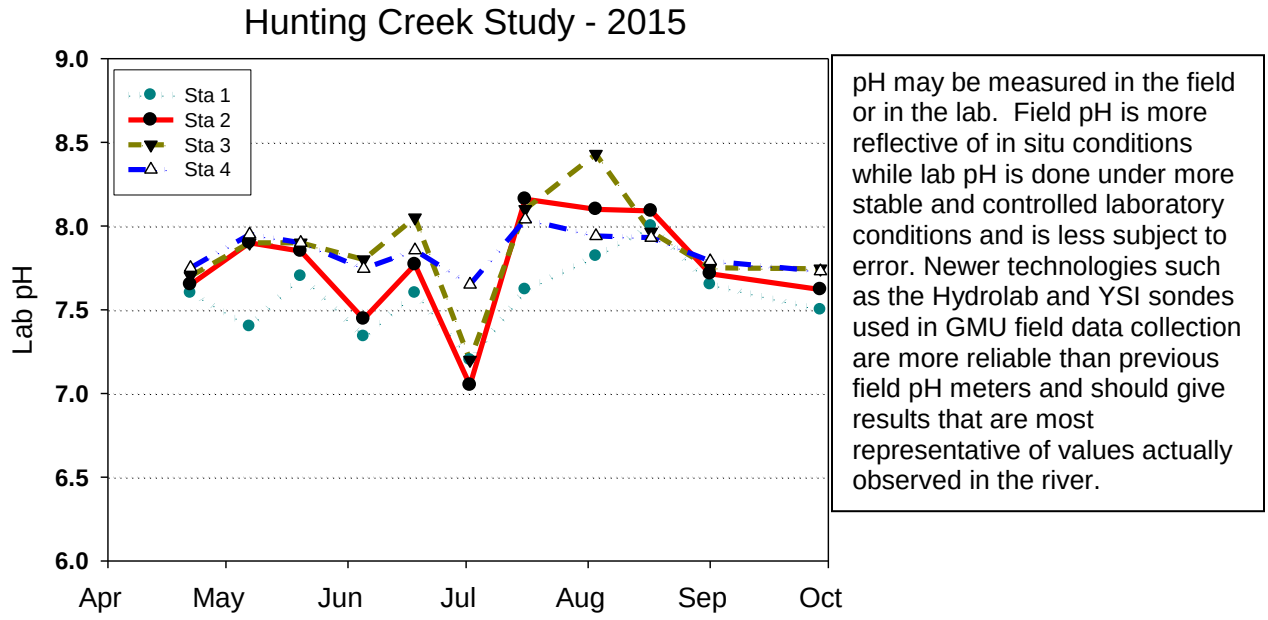


Figure 15. pH. AlexRenew Lab Data. Month tick is at first day of month.

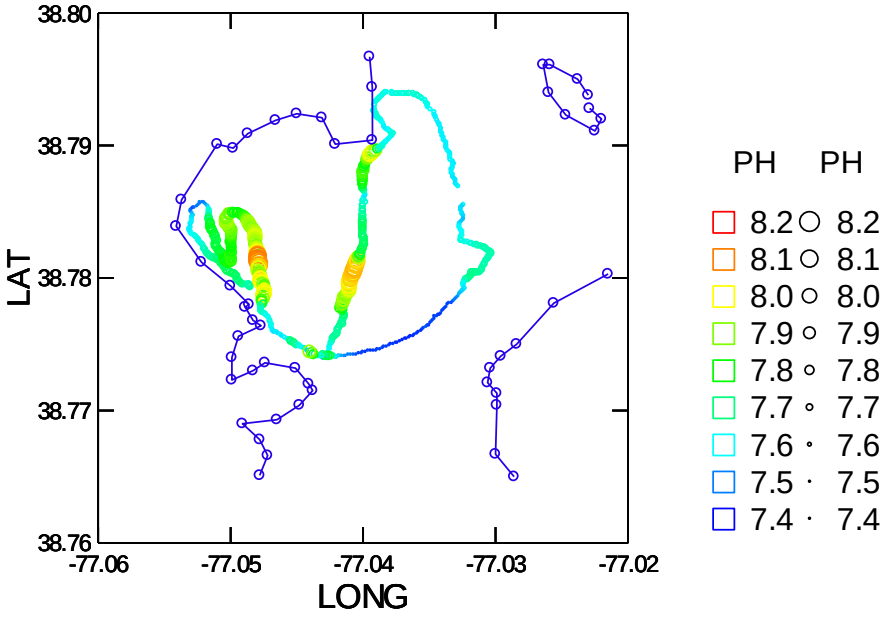


Figure 16a. Water Quality Mapping. July 30, 2015. pH.

Water quality mapping on both dates showed elevated pH in the Hunting Creek embayment due to heavy SAV growth (Figure 16a&b). Values were particularly high in August reaching a value of 9; this was consistent with the elevated DO values also observed in the embayment in August.

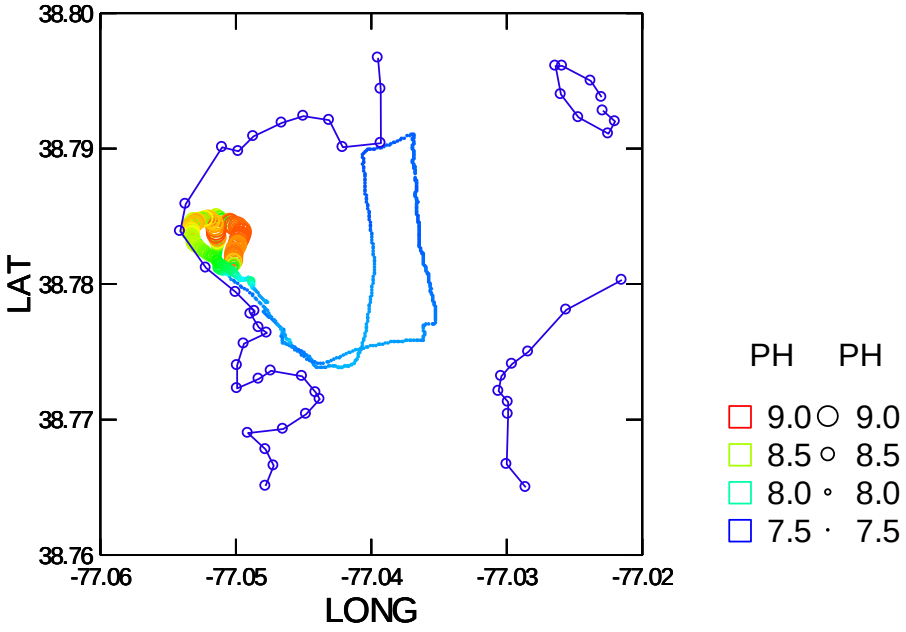
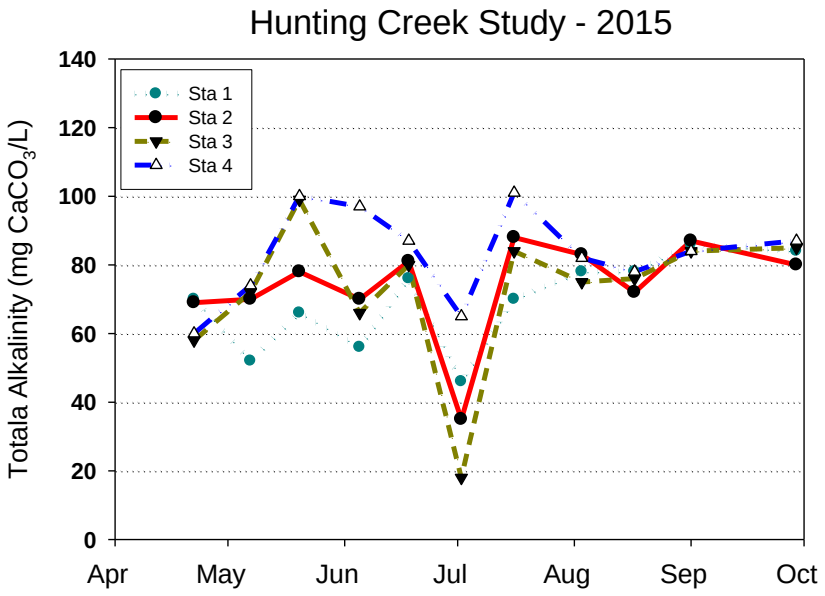


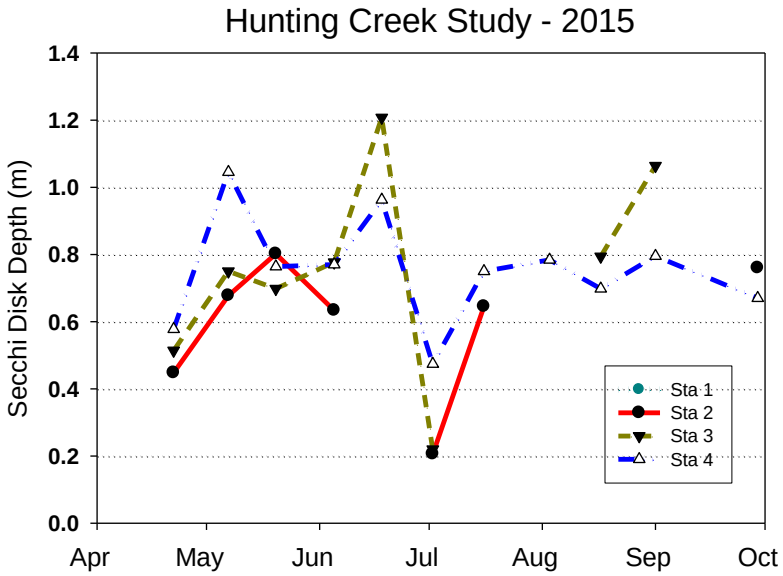
Figure 16b. Water Quality Mapping. August 19, 2015. pH.



Total alkalinity measures the amount of bicarbonate and carbonate dissolved in the water. In freshwater this corresponds to the ability of the water to absorb hydrogen ions (acid) and still maintain a near neutral pH. Alkalinity in the tidal freshwater Potomac generally falls into the moderate range allowing adequate buffering without carbonate precipitation.

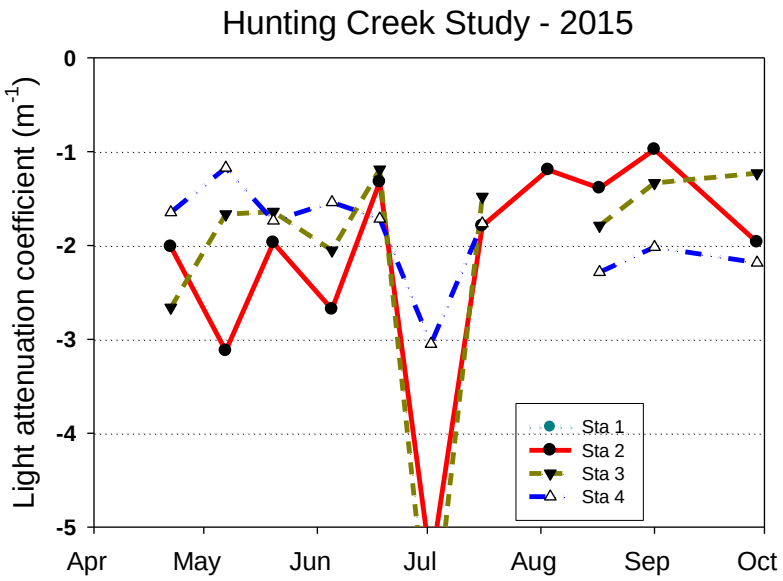
Figure 17. Total Alkalinity (mg/L as $CaCO_3$). AlexRenew Lab data. Month tick is at first day.

Total alkalinity was generally in the range 60-100 mg/L as $CaCO_3$. The major exception to this pattern was in early July when a major decline was observed in alkalinity corresponding to the large flow event (Figure 17). The decline was most marked at AR 2 and AR 3 which dropped to less than half their normal value. Water clarity as reflected by Secchi disk depth varied markedly over the year (Figure 18). Values generally increased in the spring indicating increasingly clear water, but then dropped markedly as a result of the early July runoff event. Missing values in Hunting Creek sites are due to the fact that SAV beds were about 0.5 m from the surface and, since the Secchi disk could still be seen at that depth, valid measurements were not possible.



Secchi Depth is a measure of the transparency of the water. The Secchi disk is a flat circle of thick sheet metal or plywood about 6 inches in diameter which is painted into alternate black and white quadrants. It is lowered on a calibrated rope or rod to a depth at which the disk disappears. This depth is termed the Secchi Depth. This is a quick method for determining how far light is penetrating into the water column. Light is necessary for photosynthesis and thereby for growth of aquatic plants and algae.

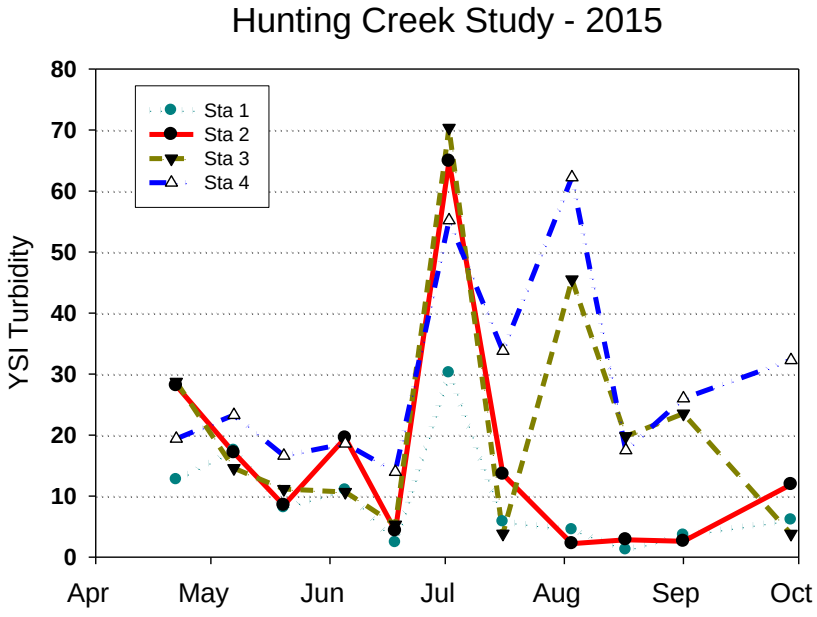
Figure 18. Secchi Disk Depth (m). GMU Field Data. Month tick is at first day of month.



Light Attenuation is another approach to measuring light penetration. This is determined by measuring light levels at a series of depths starting near the surface. The resulting relationship between depth and light is fit to a semi-logarithmic curve and the resulting slope is called the light attenuation coefficient. This relationship is called Beer's Law. It is analogous to absorbance on a spectrophotometer. The greater the light attenuation, the faster light is absorbed with depth. More negative values indicate greater attenuation. Greater attenuation is due to particulate and dissolved material which absorbs and deflects light.

Figure 19. Light Attenuation Coefficient (m^{-1}). GMU Field Data. Month tick is at first day of month.

Light attenuation coefficient data generally fell in the range -1.0 to -3.0 m^{-1} (Figure 19). A major exception to this occurred in early July when values plunged in Hunting Creek due to the particulate matter brought in by the runoff event. Values recovered and remained in the -1.0 to -2.0 range in Hunting Creek for the remainder of the year. In the river there was a smaller drop in early July. Turbidity also showed a strong response to the early July flows and there were also some elevated values through the rest of the summer at AR3 and AR4 (Figure 20).



Turbidity is yet a third way of measuring light penetration. Turbidity is a measure of the amount of light scattering by the water column. Light scattering is a function of the concentration and size of particles in the water. Small particles scatter more light than large ones (per unit mass) and more particles result in more light scattering than fewer particles.

Figure 20. Turbidity (NTU). GMU Lab Data. Month tick is at first day of month.

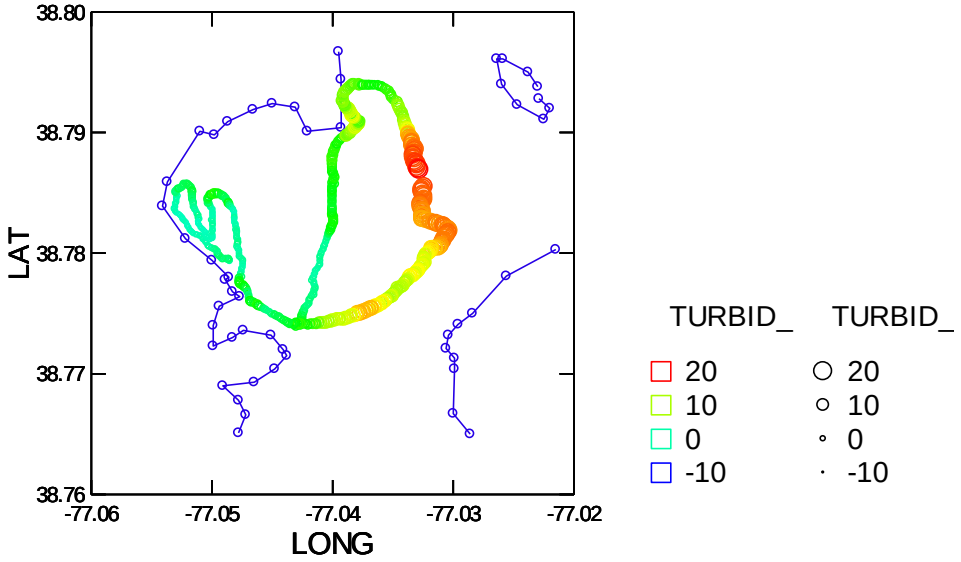


Figure 21a. Water Quality Mapping. July 30, 2015. Turbidity YSI.

On the July mapping cruise there was a marked difference in turbidity levels between Hunting Creek and the Potomac mainstem with much higher levels observed in the mainstem (Figure 21a). Within Hunting Creek values were generally in the 0-10 NTU range moving gradually to 20-30 NTU in the river mainstem. In August the differences persisted (Figure 21b).

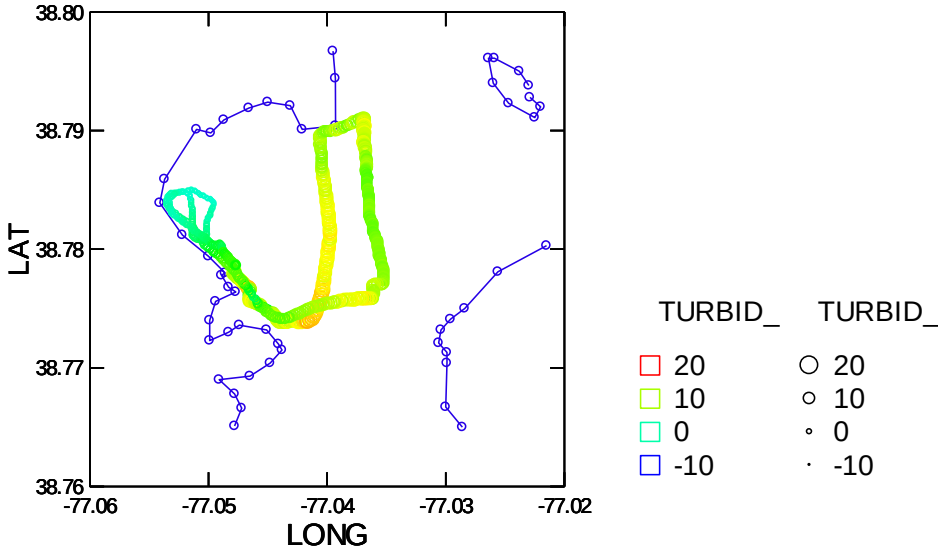
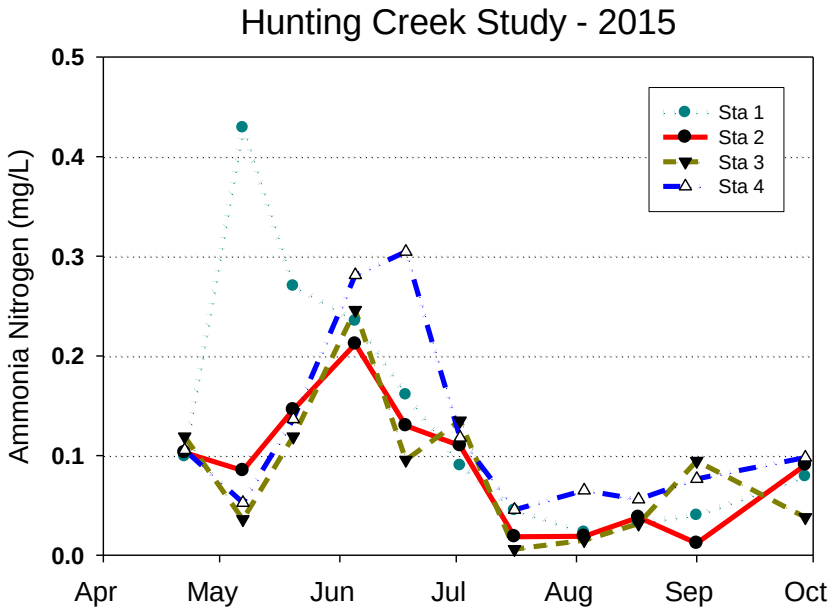


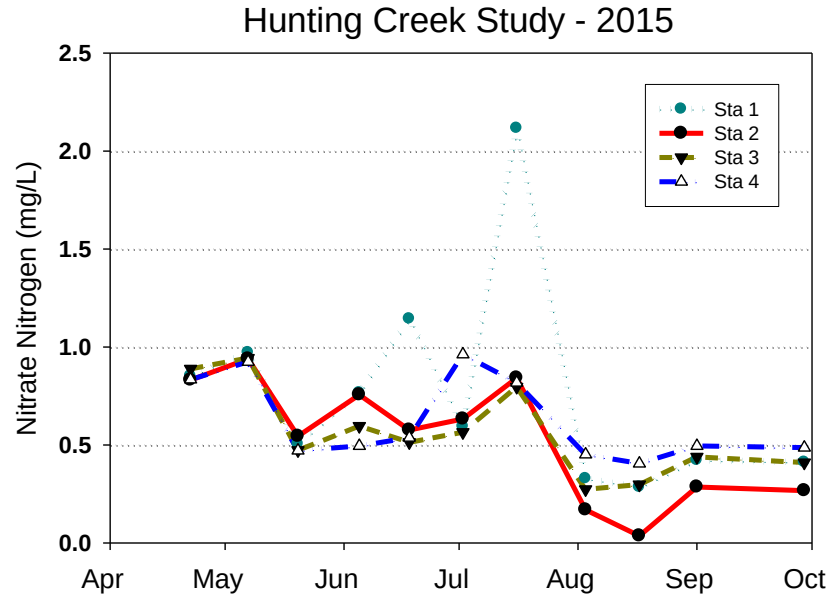
Figure 21b. Water Quality Mapping. August 19, 2015. Turbidity YSI.



Ammonia nitrogen measures the amount of ammonium ion (NH_4^+) and ammonia gas (NH_3) dissolved in the water. Ammonia nitrogen is readily available to algae and aquatic plants and acts to stimulate their growth. While phosphorus is normally the most limiting nutrient in freshwater, nitrogen is a close second. Ammonia nitrogen is rapidly oxidized to nitrate nitrogen when oxygen is present in the water so high ammonia levels suggest proximity to a source.

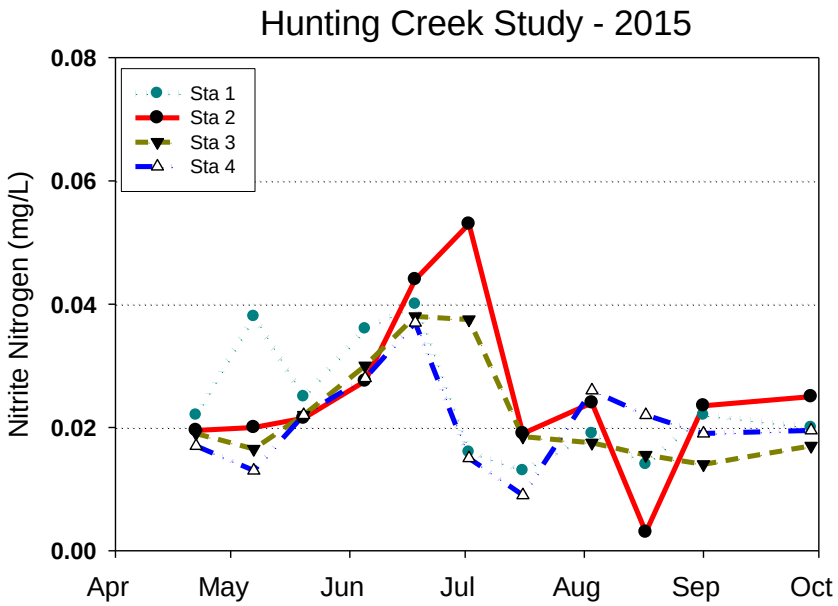
Figure 22. Ammonia Nitrogen (mg/L). AlexRenew Lab Data. Month tick is at first day of month.

Ammonia nitrogen was consistently low (<0.2 mg/L) for most of the study period (Figure 22). A clear seasonal pattern was seen at most sites with an increase from early May into June followed by a decline to low levels by mid July. Nitrate nitrogen levels showed a general seasonal decline at all sites (Figure 23). Nitrate levels were elevated in two samples from at AR1.



Nitrate Nitrogen refers to the amount of N that is in the form of nitrate ion (NO_3^-). Nitrate ion is the most common form of nitrogen in most well oxidized freshwater systems. Nitrate concentrations are increased by input of wastewater, nonpoint sources, and oxidation of ammonia in the water. Nitrate concentrations decrease when algae and plants are actively growing and removing nitrogen as part of their growth.

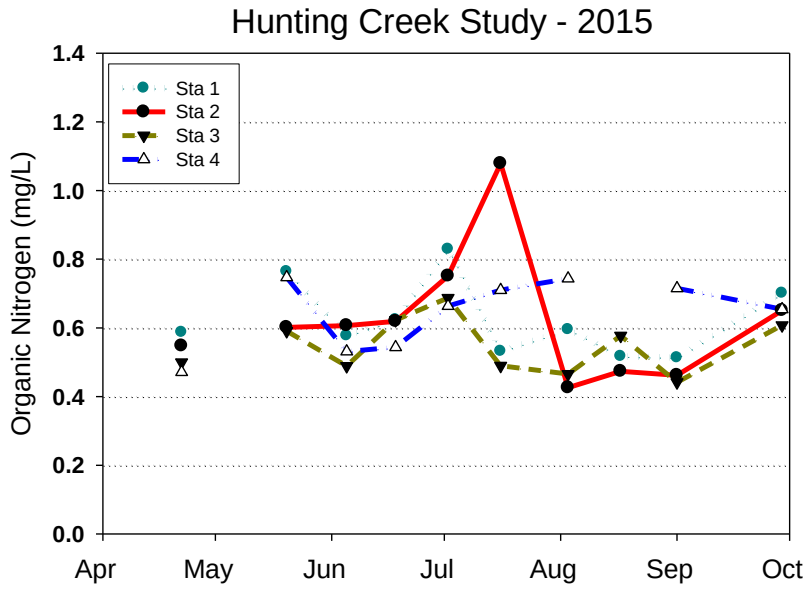
Figure 23. Nitrate Nitrogen (mg/L). AlexRenew Lab Data. Month tick is at first day of month.



Nitrite nitrogen consists of nitrogen in the form of nitrite ion (NO_2^-). Nitrite is an intermediate in the oxidation of ammonia to nitrate, a process called nitrification. Nitrite is usually in very low concentrations unless there is active nitrification.

Figure 24. Nitrite Nitrogen (mg/L). AlexRenew Lab Data. Month tick is at first day of month.

Nitrite nitrogen was generally low (<0.04 mg/L) throughout the year (Figure 24). Values were slightly higher at AR2 in June. Organic nitrogen values were generally in the range of 0.4-0.8 mg/L (Figure 25). At AR2 there was a peak in mid July.



Organic nitrogen measures the nitrogen in dissolved and particulate organic compounds in the water. Organic nitrogen comprises algal and bacterial cells, detritus (particles of decaying plant, microbial, and animal matter), amino acids, urea, and small proteins. When broken down in the environment, organic nitrogen results in ammonia nitrogen. Organic nitrogen is determined as the difference between total Kjeldahl nitrogen and ammonia nitrogen.

Figure 25. Organic Nitrogen (mg/L). AlexRenew Lab Data. Month tick is at first day of month.

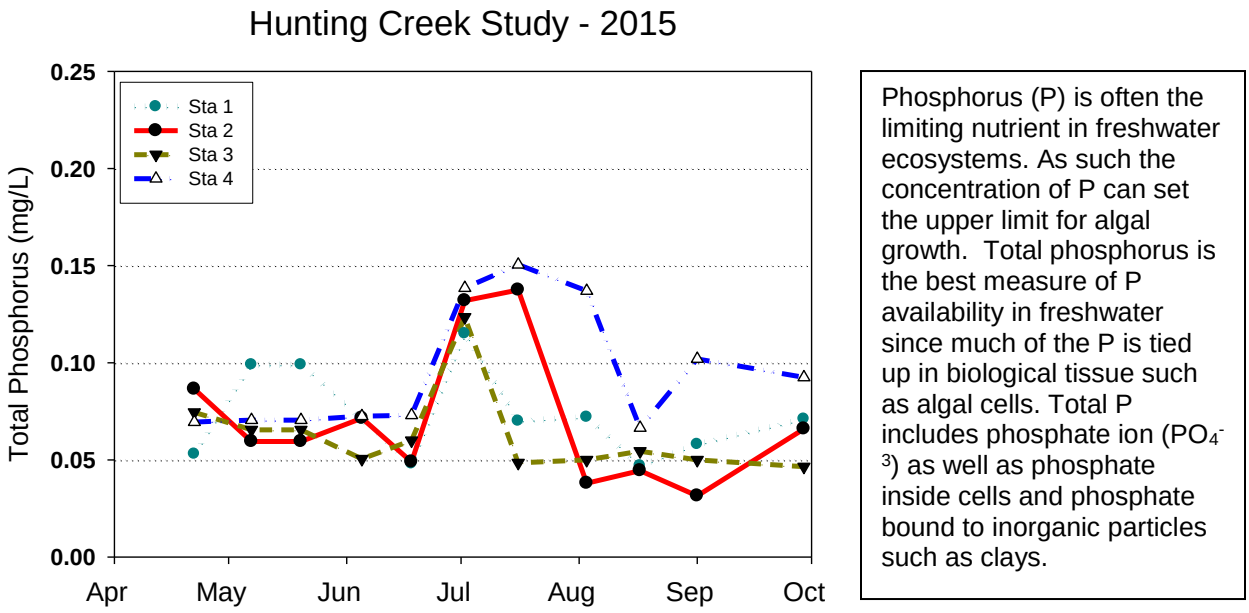


Figure 26. Total Phosphorus (mg/L). AlexRenew Lab Data. Month tick is at first day of month.

Total phosphorus showed a distinct seasonal pattern with elevated values in midsummer at all sites (Figure 26). There was little difference between river and embayments in the first half of the year, but the river mainstem was consistently somewhat higher in the later samples. Ortho-phosphorus showed a general decline throughout the year with little difference among the sites (Figure 27).

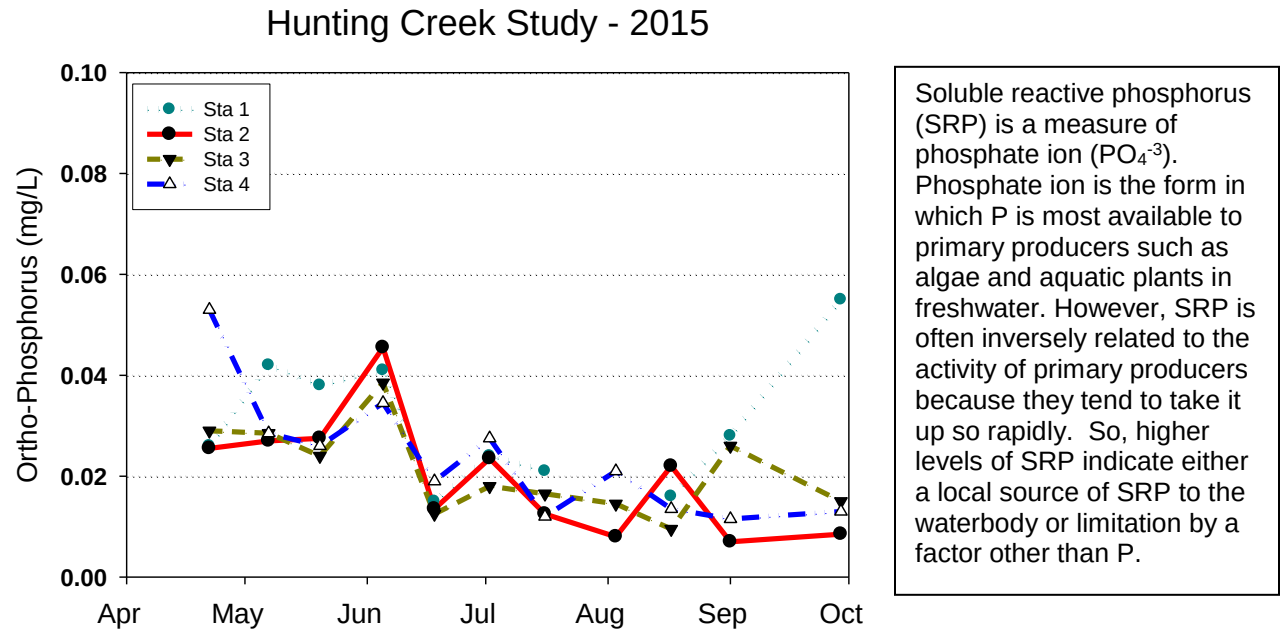
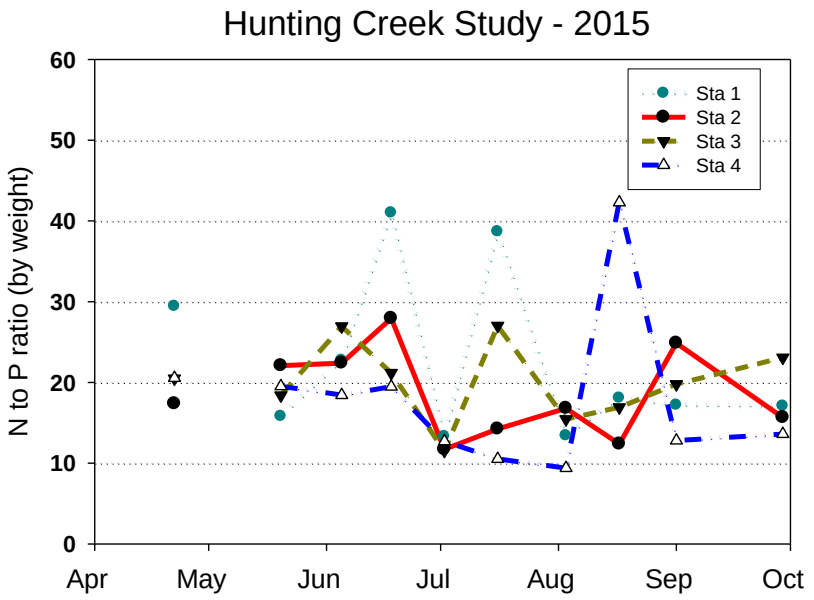


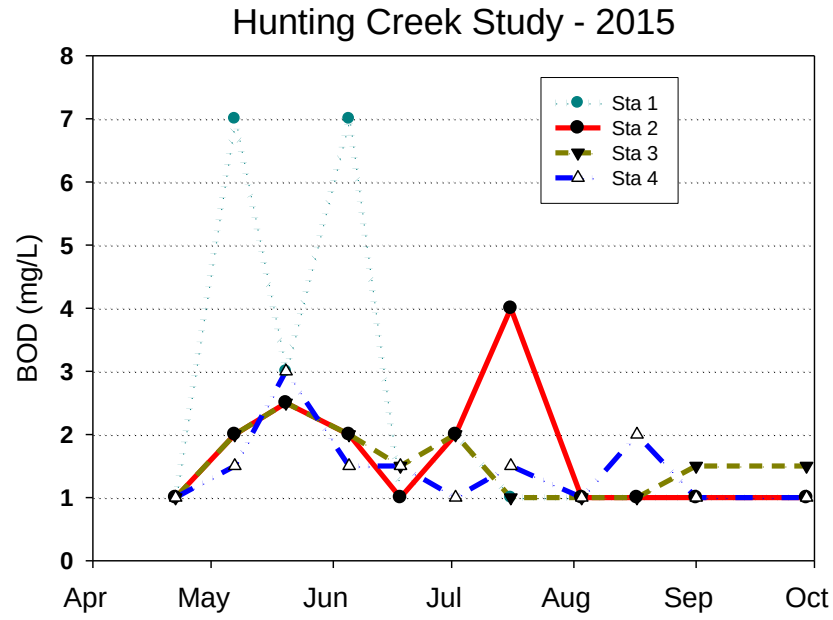
Figure 27. Soluble Reactive Phosphorus (mg/L). AlexRenew Lab Data. Month tick is at first day of month.



N:P ratio is determined by summing all of the compon-ents of N (ammonia, nitrate, nitrite, and organic nitrogen) and dividing by total P. This ratio gives an indication of whether N or P is more likely to be limiting primary production in a given freshwater system. Generally, values above 7.2 are considered indicative of P limitation while values below 7.2 suggest N limitation. N limitation could lead to dominance by cyanobacteria who can fix their own N from the atmosphere.

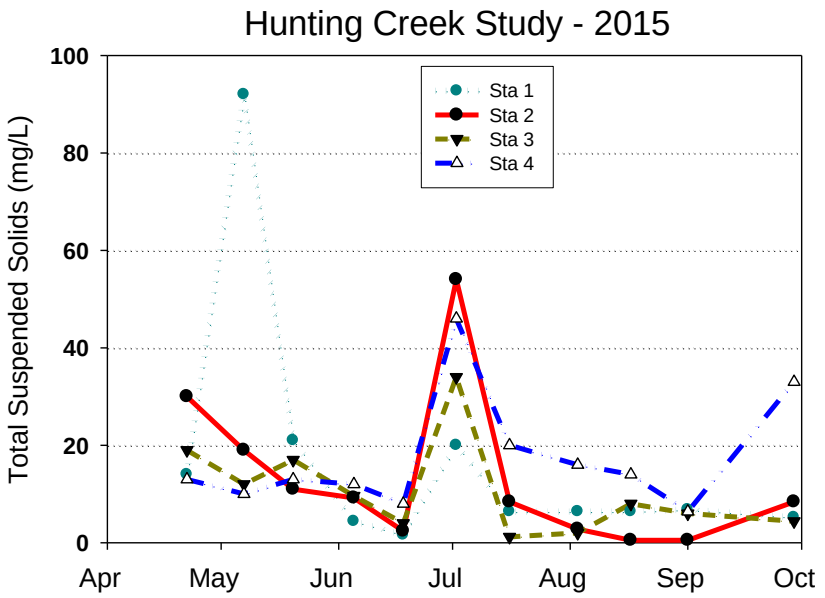
Figure 28. N/P Ratio (by mass). AlexRenew Lab Data. Month tick is at first day of month.

N/P ratio consistently pointed to P limitation being greater than 7.2 at all times (Figure 28). Values were most consistent at stations AR 2 and AR 3. Biochemical oxygen demand (BOD) was often 1 or 2 mg/L at all stations (Figure 29). Exceptions to this occurred at AR1 in the spring and at AR2 in mid July.



Biochemical oxygen demand (BOD) measures the amount of decomposable organic matter in the water as a function of how much oxygen it consumes as it breaks down over a given number of days. Most commonly the number of days used is 5. BOD is a good indicator of the potential for oxygen depletion in water. BOD is composed both dissolved organic compounds in the water as well as microbes such as bacteria and algae which will respire and consume oxygen during the period of measurement.

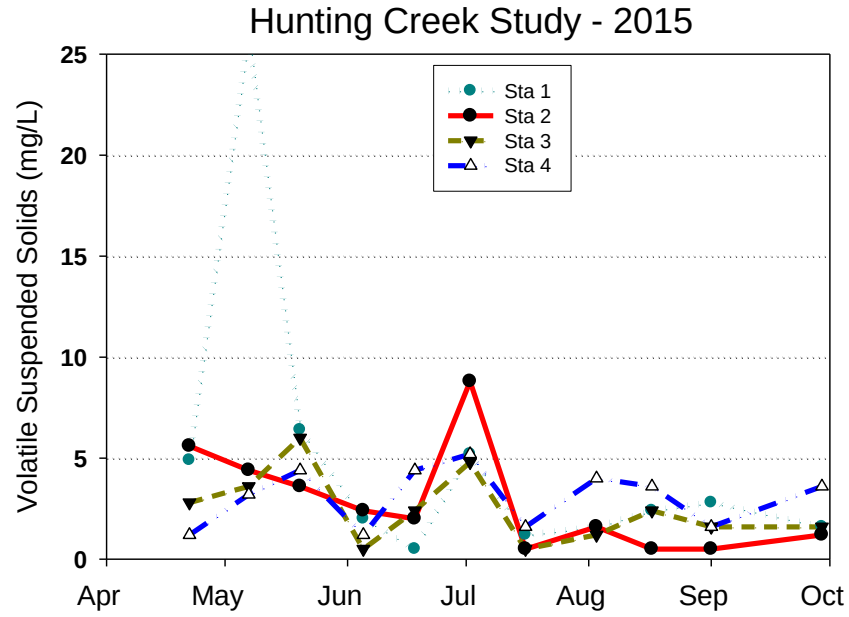
Figure 29. Biochemical Oxygen Demand (mg/L). AlexRenew Lab Data. Month tick is at first day of month.



Total suspended solids (TSS) is measured by filtering a known amount of water through a fine filter which retains all or virtually all particles in the water. This filter is then dried and the weight of particles on the filter determined by difference. TSS consists of both organic and inorganic particles. During periods of low river and tributary inflow, organic particles such as algae may dominate. During storm flow periods or heavy winds causing resuspension, inorganic particles may dominate.

Figure 30. Total Suspended Solids (mg/L). AlexRenew Lab Data. Month tick is at first day of month.

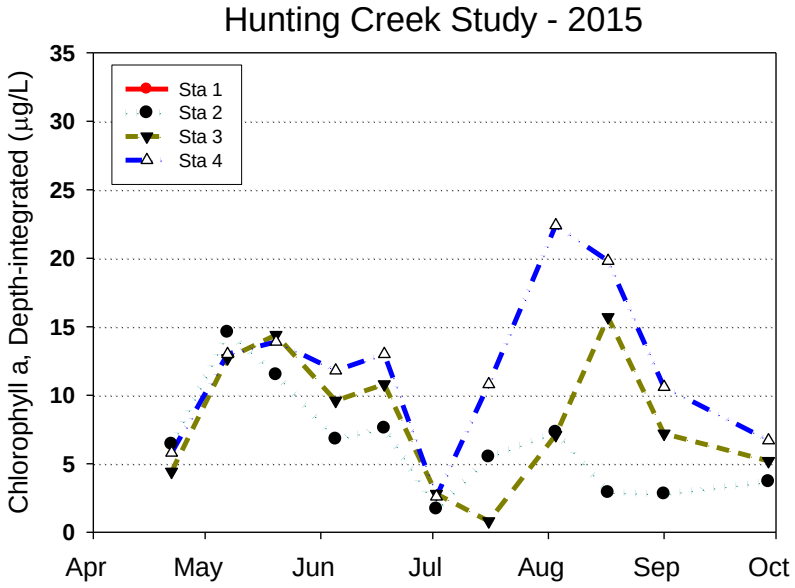
Total suspended solids was generally in the range 0-20 mg/L (Figure 30). The major exception to this was in early July when values were clearly elevated at all stations due to the runoff event. Values were also generally higher in the river in the later part of the year, due to scouring in the bottom samples. VSS showed a similar pattern generally, the early July flow event did not stand out as much (Figure 31). Causes for the early May peak at AR 1 were not readily apparent. (Note: some bottom TSS/VSS values were not included here as the samples may have included resuspended sediments).



Volatile suspended solids (VSS) is determined by taking the filters used for TSS and then ashing them to combust (volatilize) the organic matter. The organic component is then determined by difference. VSS is a measure of organic solids in a water sample. These organic solids could be bacteria, algae, or detritus. Origins include sewage effluent, algae growth in the water column, or detritus produced within the waterbody or from tributaries. In summer in Gunston Cove a chief source is algal (phytoplankton) growth.

Figure 31. Volatile Suspended Solids (mg/L). AlexRenew Lab Data. Month tick is at first day of month.

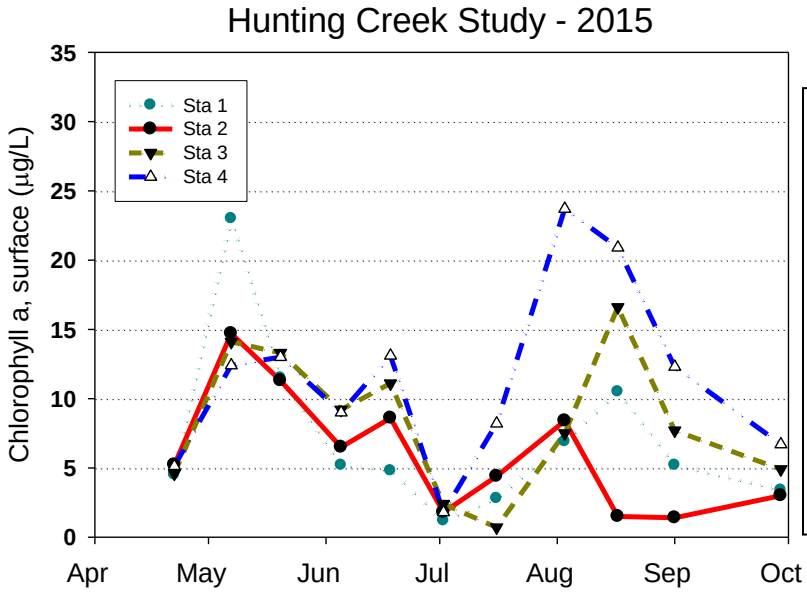
C. Phytoplankton -2015



Chlorophyll *a* is a measure of the amount of algae growing in the water column. These suspended algae are called phytoplankton, meaning "plant wanderers". In addition to the true algae (greens, diatoms, cryptophytes, etc.) the term phytoplankton includes cyanobacteria (sometimes known as "blue-green" algae). Both depth-integrated and surface chlorophyll values are measured due to the capacity of phytoplankton to aggregate near the surface under certain conditions.

Figure 32. Chlorophyll *a* (µg/L). Depth-integrated. GMU Lab Data. Month tick is at the first day of month.

Chlorophyll *a* increased from April to May and remained fairly stable at 10-15 ug/L at all stations through mid June (Figure 32). Values then declined sharply due to flushing from the flow event. A major increase was then observed at the river mainstem site AR4 reaching a peak of nearly 25 ug/L in August. Values also increased at this time at AR 3, but never recovered at AR2. Surface chlorophyll showed similar spatial and temporal patterns (Figure 33).



In the tidal freshwater Potomac generally, there is very little difference in surface and depth-integrated chlorophyll levels because tidal action keeps the water well-mixed which overcomes any potential surface aggregation by the phytoplankton. Summer chlorophyll concentrations above 30 ug/L are generally considered characteristic or eutrophic conditions.

Figure 33. Chlorophyll *a* (µg/L). Surface. GMU Lab Data. Month tick is at first day of month.

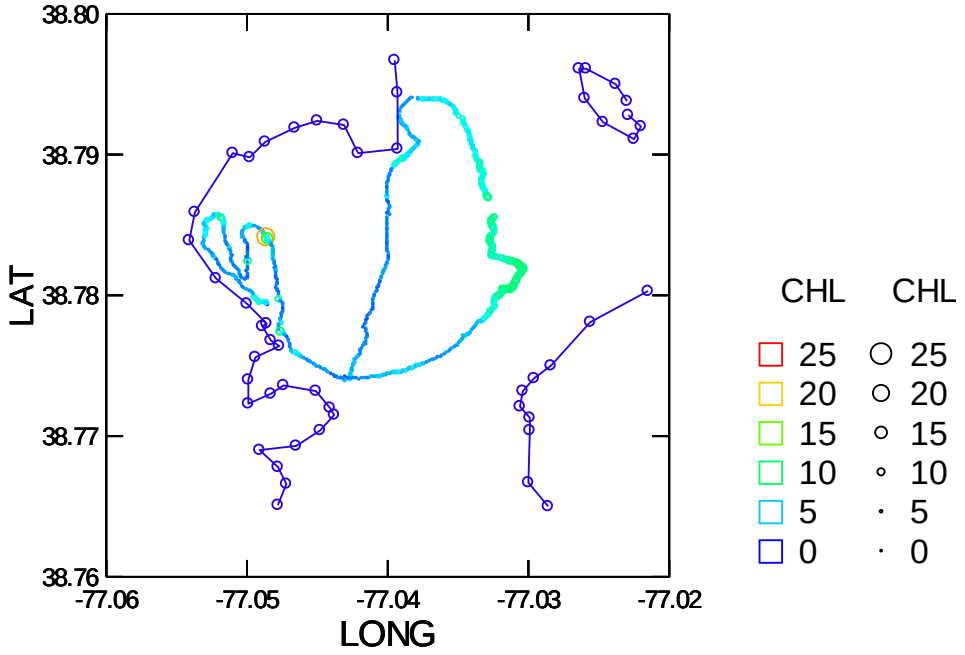


Figure 34a. Water Quality Mapping. July 30, 2015. Chlorophyll YSI (mg/L).

Both water quality mapping cruises indicated higher chlorophyll level in and near the river mainstem (Figure 34a&b). Values were in the 0-10 ug/L range in SAV-laden Hunting Creek and 10-20 ug/L in the river mainstem.

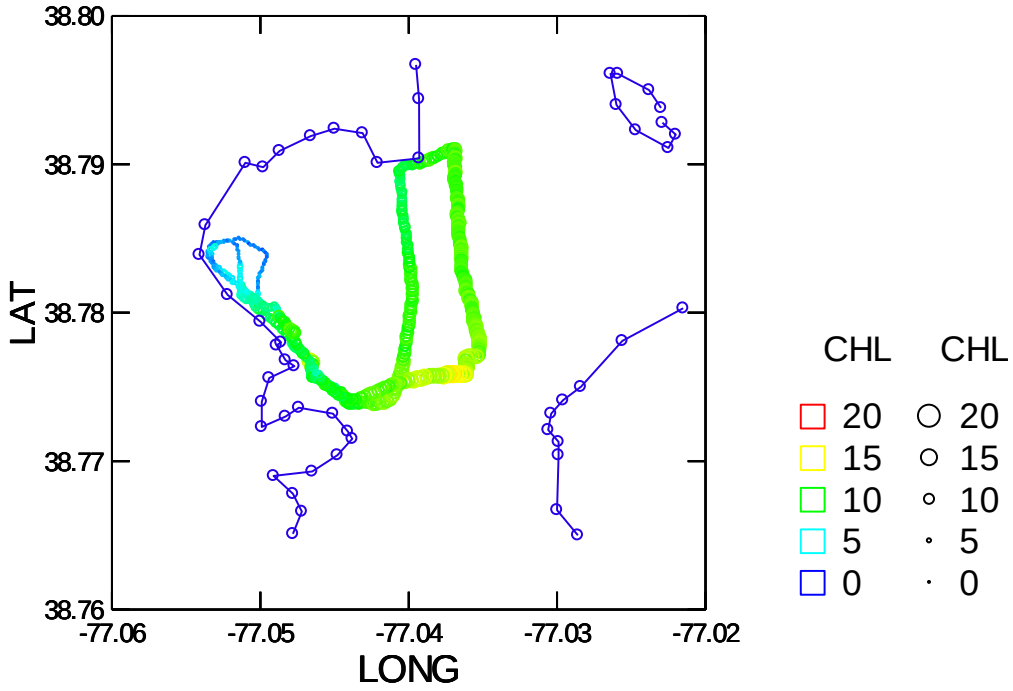


Figure 34b. Water Quality Mapping. August 29, 2015. Chlorophyll YSI (mg/L).

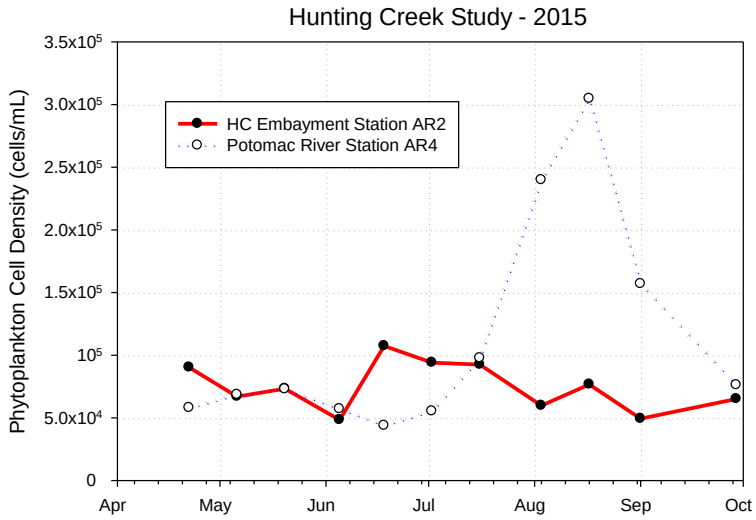


Figure 35. Phytoplankton Density (cells/mL).

Phytoplankton cell density provides a measure of the number of algal cells per unit volume. This is a rough measure of the abundance of phytoplankton, but does not discriminate between large and small cells. Therefore, a large number of small cells may actually represent less biomass (weight of living tissue) than a smaller number of large cells. However, small cells are typically more active than larger ones so cell density is probably a better indicator of activity than of biomass. The smaller cells are mostly cyanobacteria.

Phytoplankton density was generally low from April through early July in both embayment and river (Figure 35). At the river station, a clear rise was observed in mid July with high values continuing through August. Values generally declined from mid June on in Hunting Creek (AR2). Total biovolume exhibited substantial variability from sampling to sampling (Figure 36).

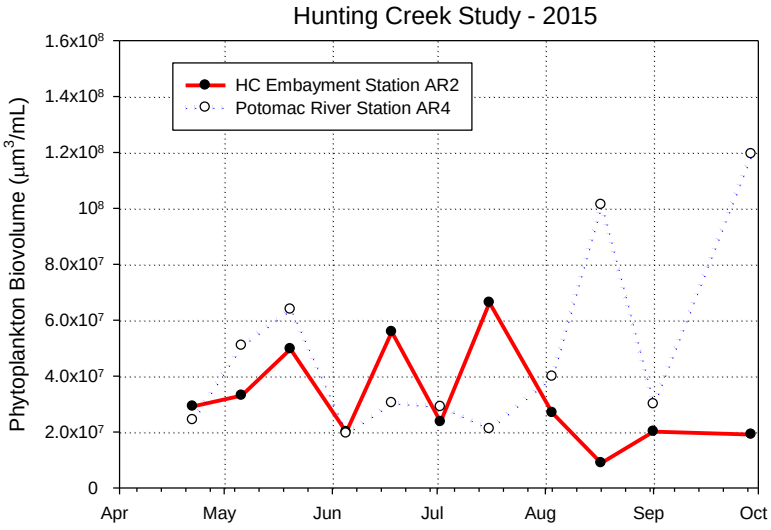
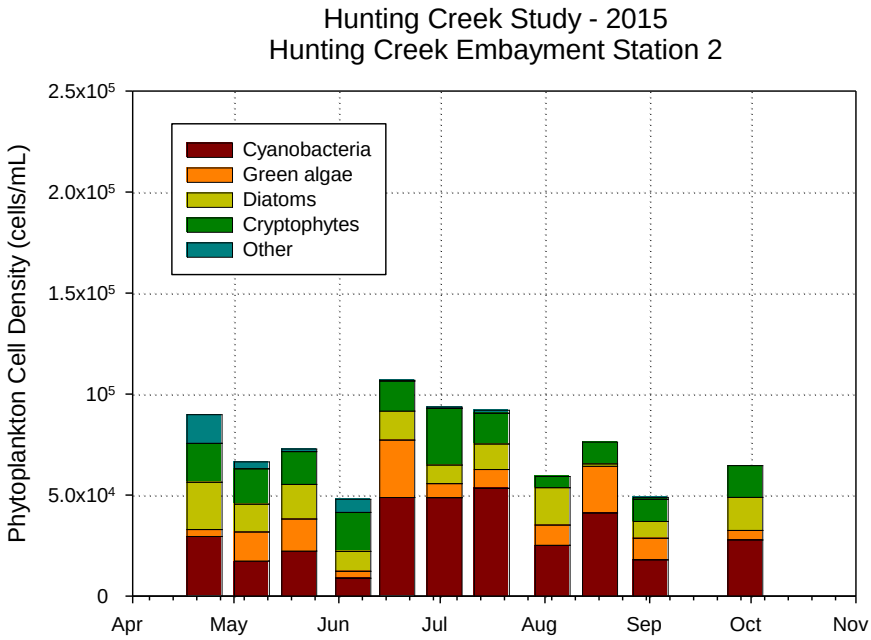


Figure 36. Phytoplankton Biovolume (um³/mL).

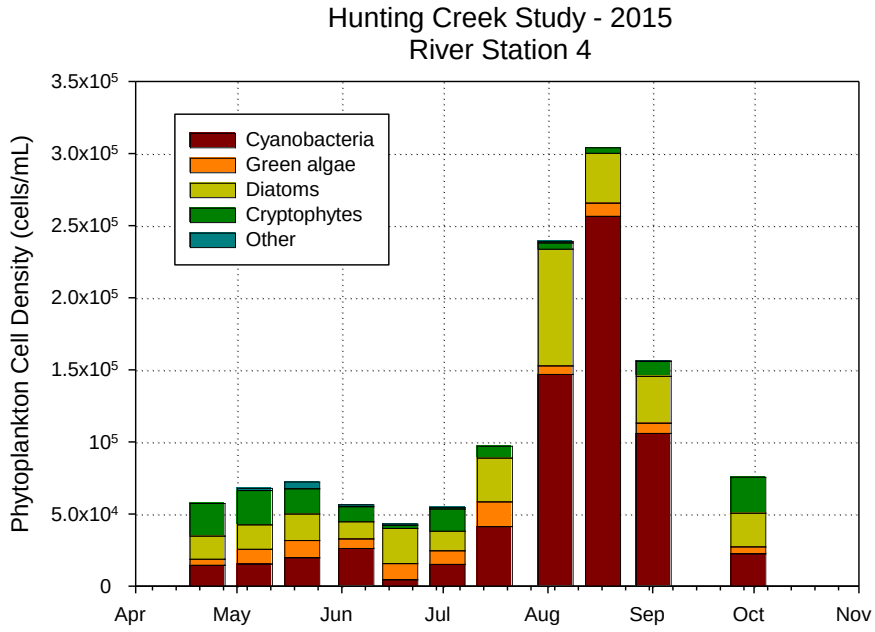
The volume of individual cells of each species is determined by approximating the cells of each species to an appropriate geometric shape (e.g. sphere, cylinder, cone, cube, etc.) and then making the measurements of the appropriate dimensions under the microscope. Total phytoplankton biovolume (shown here) is determined by multiplying the cell density of each species by the biovolume of each cell of that species. Biovolume accounts for the differing size of various phytoplankton cells and is probably a better measure of biomass. However, it does not account for the varying amount of water and other nonliving constituents in cells.



Total phytoplankton cell density can be broken down by major group. **Cyanobacteria** are sometimes called "blue-green algae". **Other** includes euglenoids and dinoflagellates. Due to their small size cyanobacteria typically dominate cell density numbers. Their numbers are typically highest in the late summer reflecting an accumulation of cells during favorable summer growing conditions.

Figure 37. Phytoplankton Density by Major Group (cells/mL). Hunting Creek.

Cyanobacteria were generally the most numerous major group of phytoplankton in Hunting Creek (Figure 37). Depending on the date, cryptophytes, diatoms, and green algae were also important. Similar patterns in dominance were observed in the river, but the main difference was the greatly elevated total densities in August and early September attributable mainly to large increases in cyanobacteria (Figure 38). The early July runoff event did not strongly impact phytoplankton densities at either site.



In the river cyanobacteria normally follow similar patterns as in the embayments, but may attain lower abundances. This is probably due to the deeper water column which leads to lower effective light levels and greater mixing. Other groups such as diatoms and green algae tend to be more important on a relative basis than in the embayments.

Figure 38. Phytoplankton Density by Major Group (cells/mL). River.

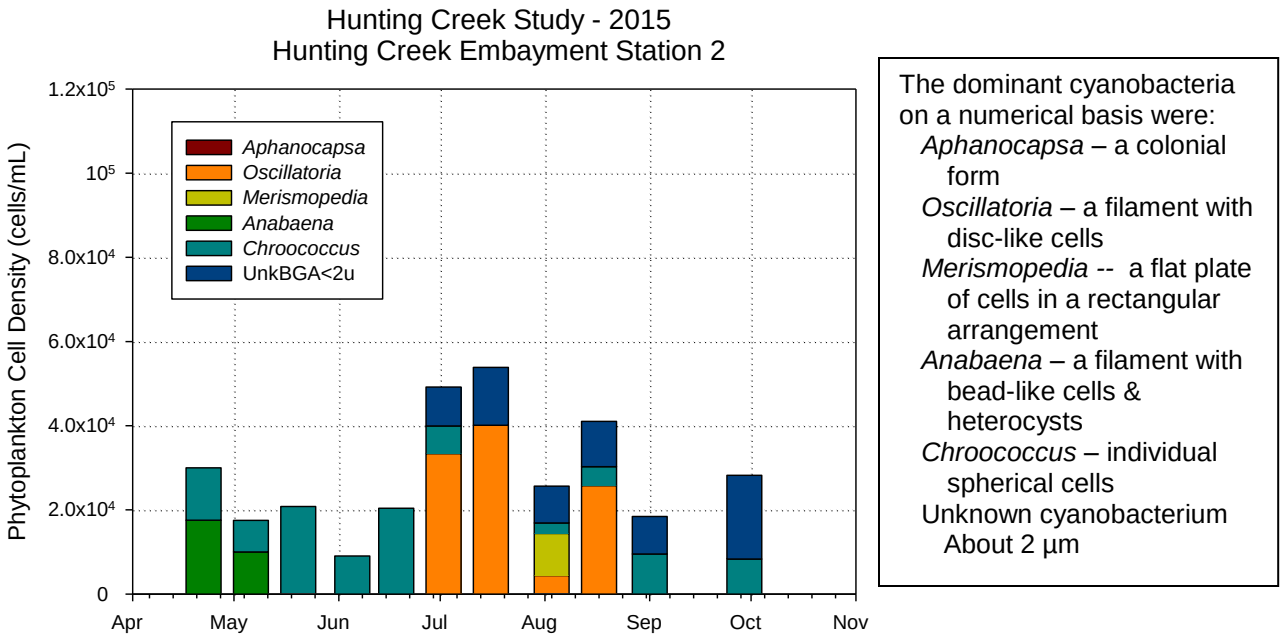


Figure 39. Phytoplankton Density by Dominant Cyanobacteria (cells/mL). Hunting Creek.

Oscillatoria was the dominant cyanobacterium in Hunting Creek in summer. *Anabaena* was most abundant in spring and *Chroococcus* was found through the entire period (Figure 39). In the river cyanobacteria were more numerous than in the cove in August and early September. *Oscillatoria*, *Merismopedia*, and *Aphanocapsa* were the dominants responsible for these elevated levels (Figure 40).

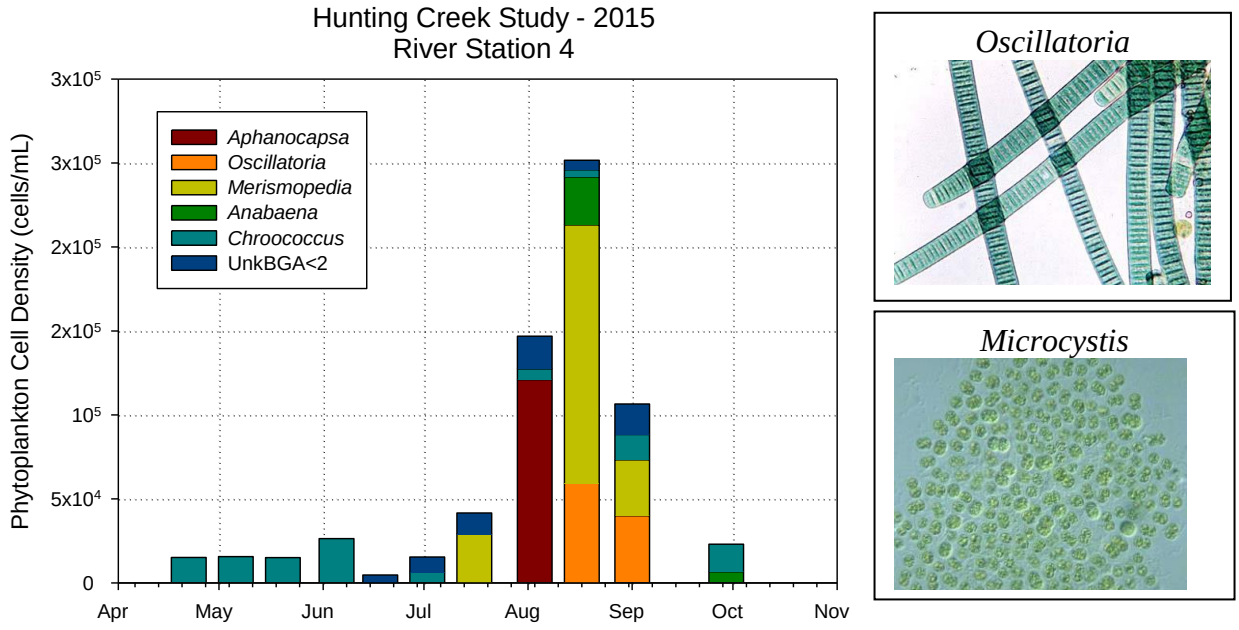


Figure 40. Phytoplankton Density by Dominant Cyanobacteria (cells/mL). River.

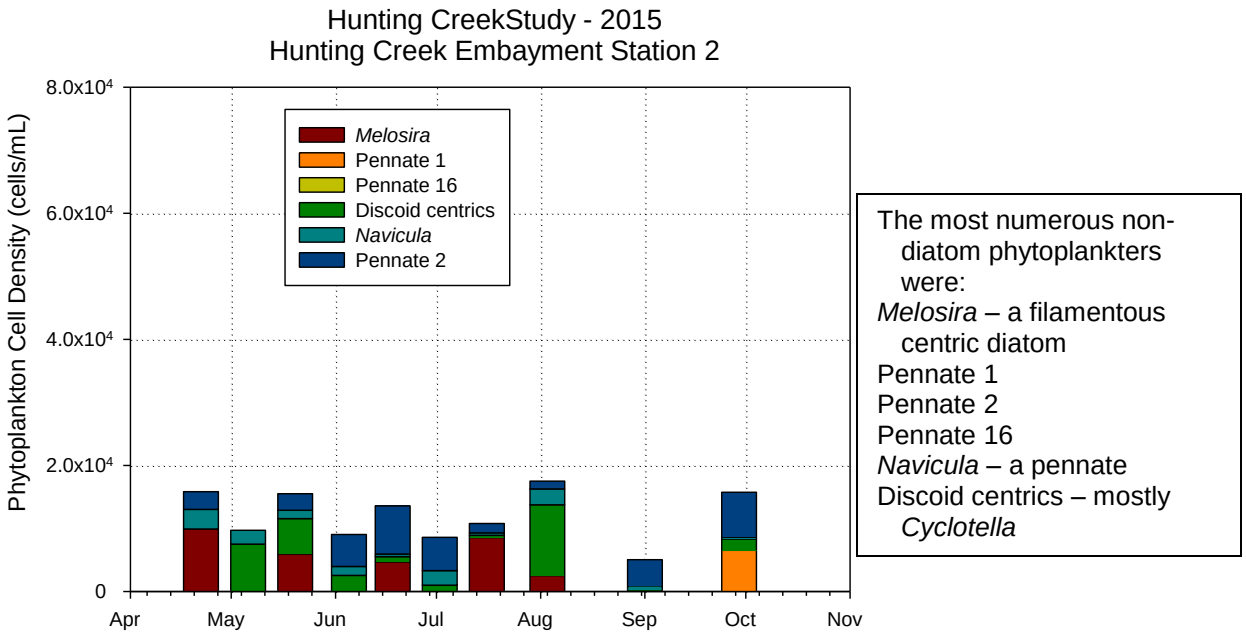


Figure 41. Phytoplankton Density (#/mL) by Dominant Diatom Taxa. Hunting Creek.

Melosira, discoid centrics, and Pennate 2 were generally the greatest contributors to cell density at the Hunting Creek station (Figure 41). In the river diatom densities were generally similar to those in the cove with similar taxonomic makeup (Figure 42). The exception was in early August when pennate diatoms were unusually abundant. This may have been due to resuspension of cells from the sediments. This issue will be discussed later.

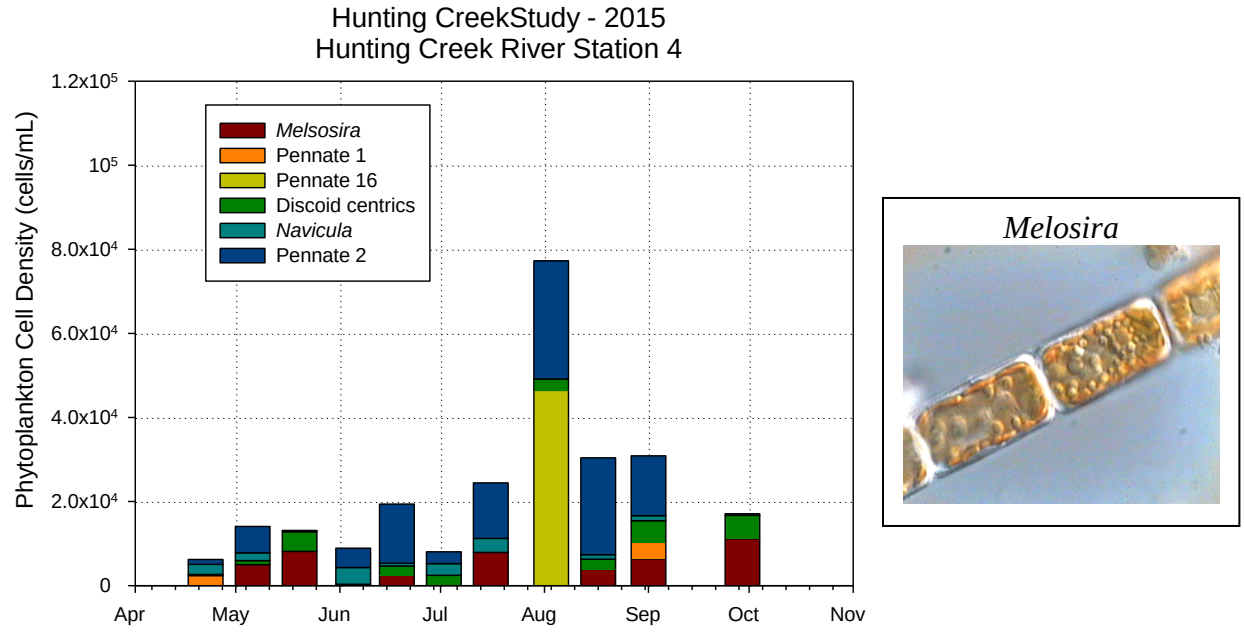


Figure 42. Phytoplankton Density (#/mL) by Dominant Diatom Taxa. River.

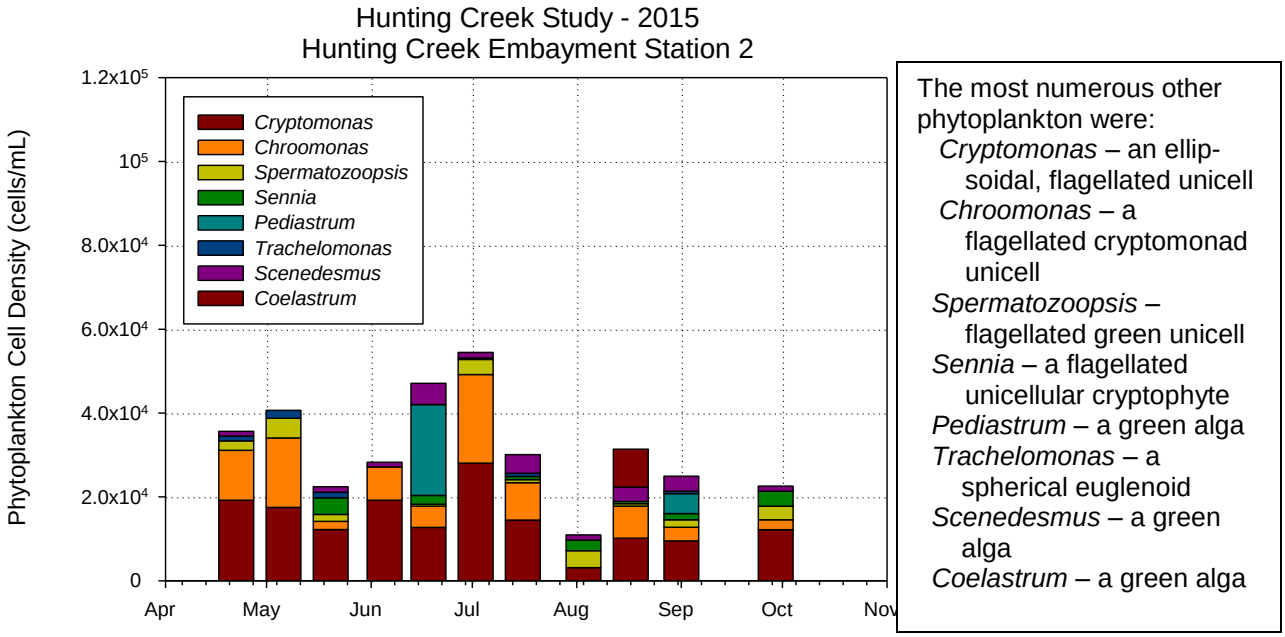


Figure 43. Phytoplankton Density (#/mL) by Dominant Other Taxa. Hunting Creek.

Phytoplankton species that were neither cyanobacteria nor diatoms were grouped together as "other" for these graphs; these included most numerous taxa of green algae, cryptophytes, euglenoids, and dinoflagellates. At AR2 the cryptophytes *Cryptomonas* and *Chroomonas* were consistently the most numerous (Figure 43). A similar pattern in the abundance of other taxa was found in the river with *Trachelomonas* and *Scenedesmus* also being important on some dates (Figure 44).

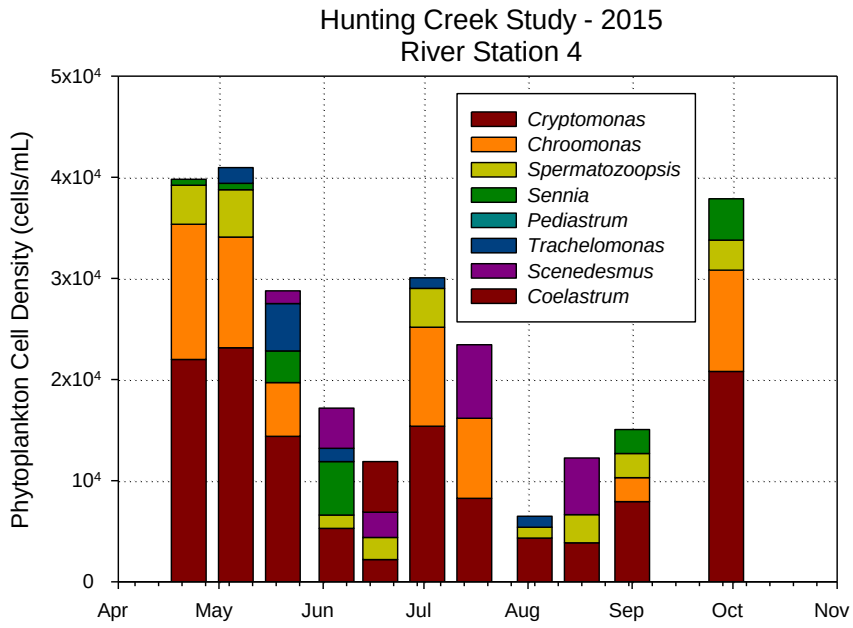


Figure 44. Phytoplankton Density (#/mL) by Dominant Taxa. River.

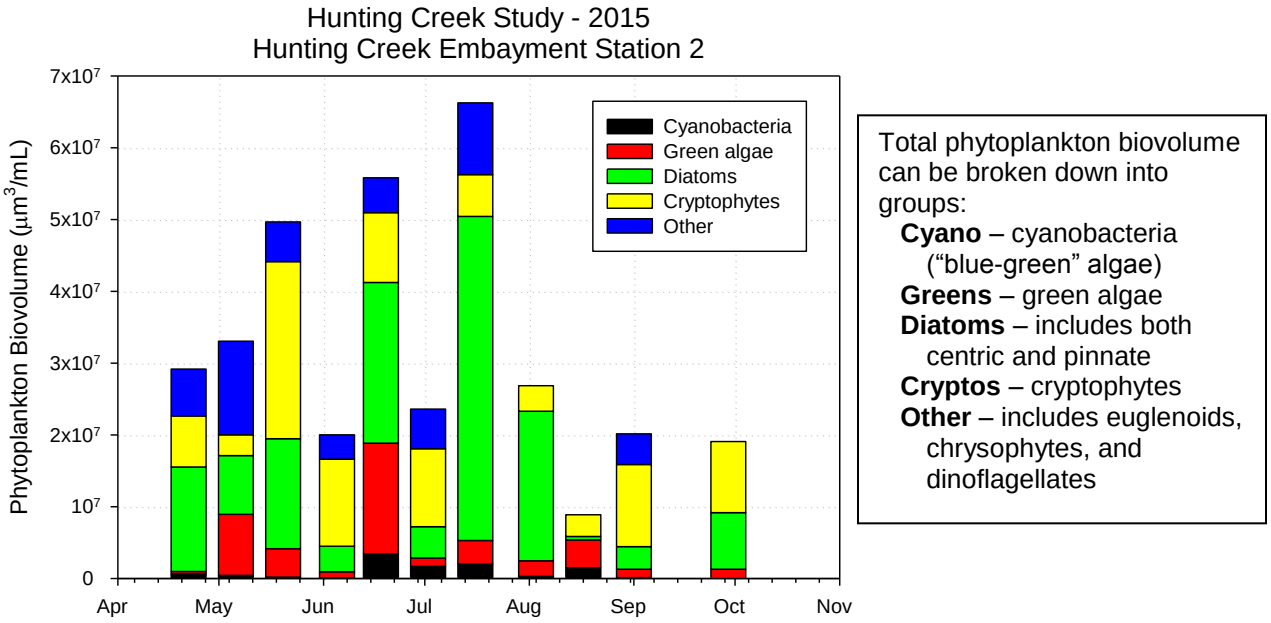


Figure 45. Phytoplankton Biovolume (um³/mL) by Major Groups. Hunting Creek.

In the cove diatoms were dominant in biovolume in most samples (Figure 45). Cryptophytes and other algae consistently made substantial contributions on some dates. Despite their dominance of cell density, cyanobacteria were generally negligible in cell biovolume given their small size. In the river, diatoms were overwhelming in their dominance (Figure 46). Cryptophytes and other algae made substantial contributions on some dates.

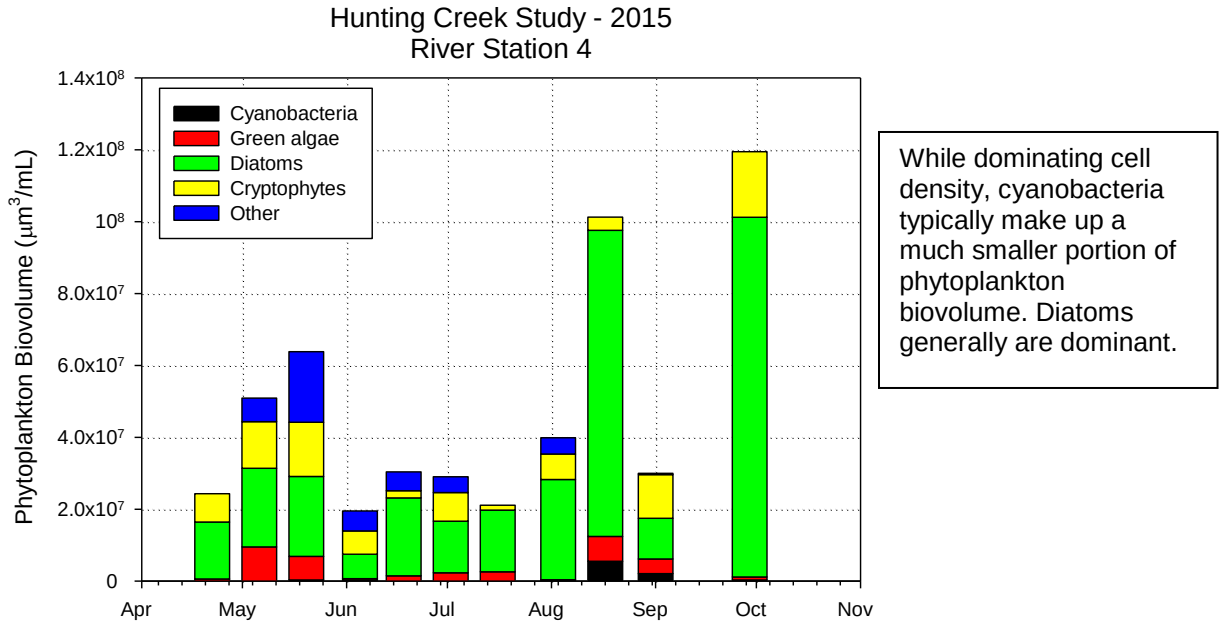


Figure 46. Phytoplankton Biovolume (um³/mL) by Major Groups. River.

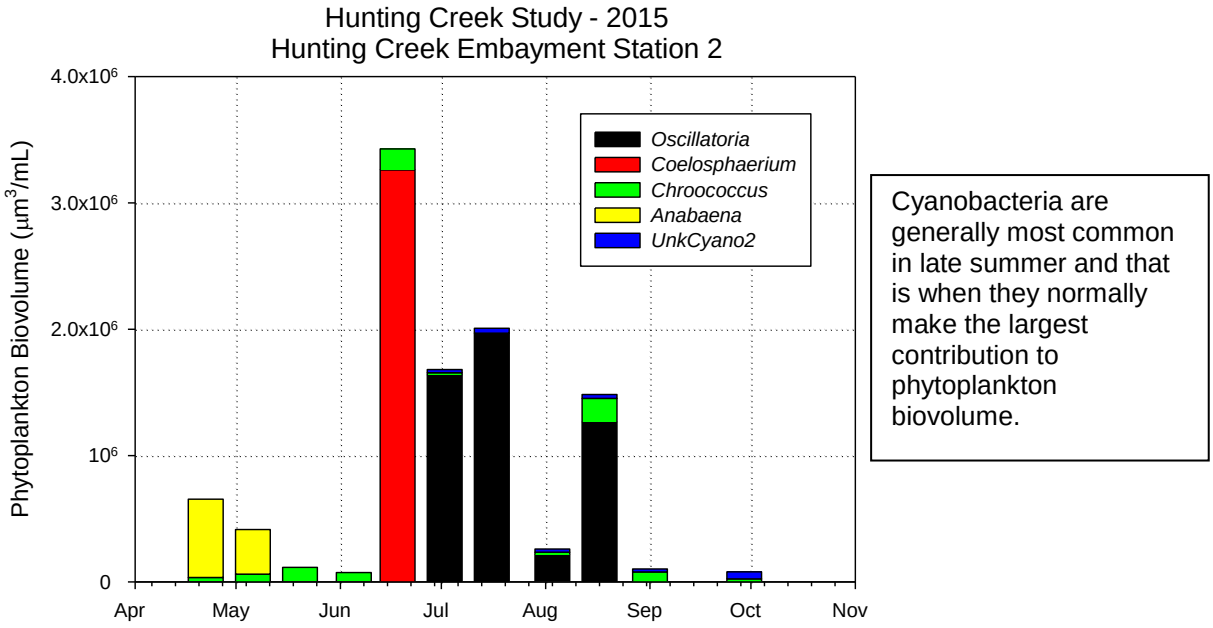


Figure 47. Phytoplankton Biovolume (um^3/mL) by Cyanobacteria Taxa. Hunting Creek.

In Hunting Creek *Oscillatoria* was dominant in cyanobacteria biovolume during most of the summer. *Coelosphaerium* had high densities on one date and *Anabaena* was most important in early spring (Figure 47). *Oscillatoria* was particularly dominant in late August and early September with contributions from *Chroococcus* and *Anabaena* in mid August (Figure 48).

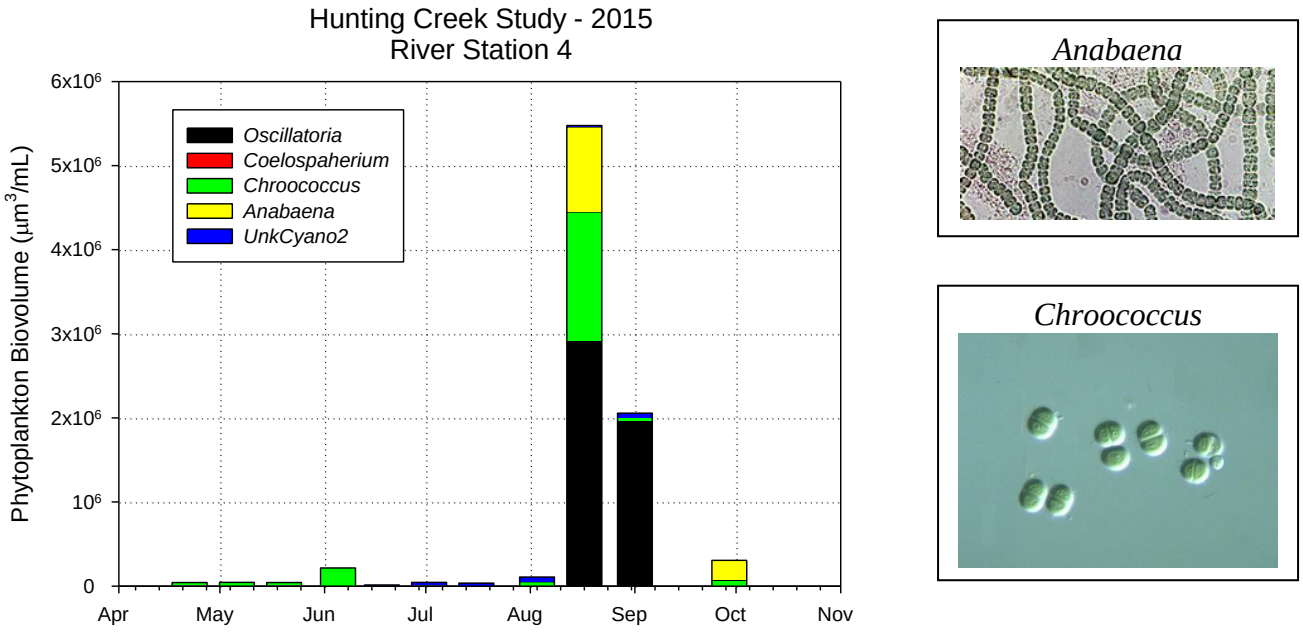


Figure 48. Phytoplankton Biovolume (um^3/mL) by Cyanobacterial Taxa. River.

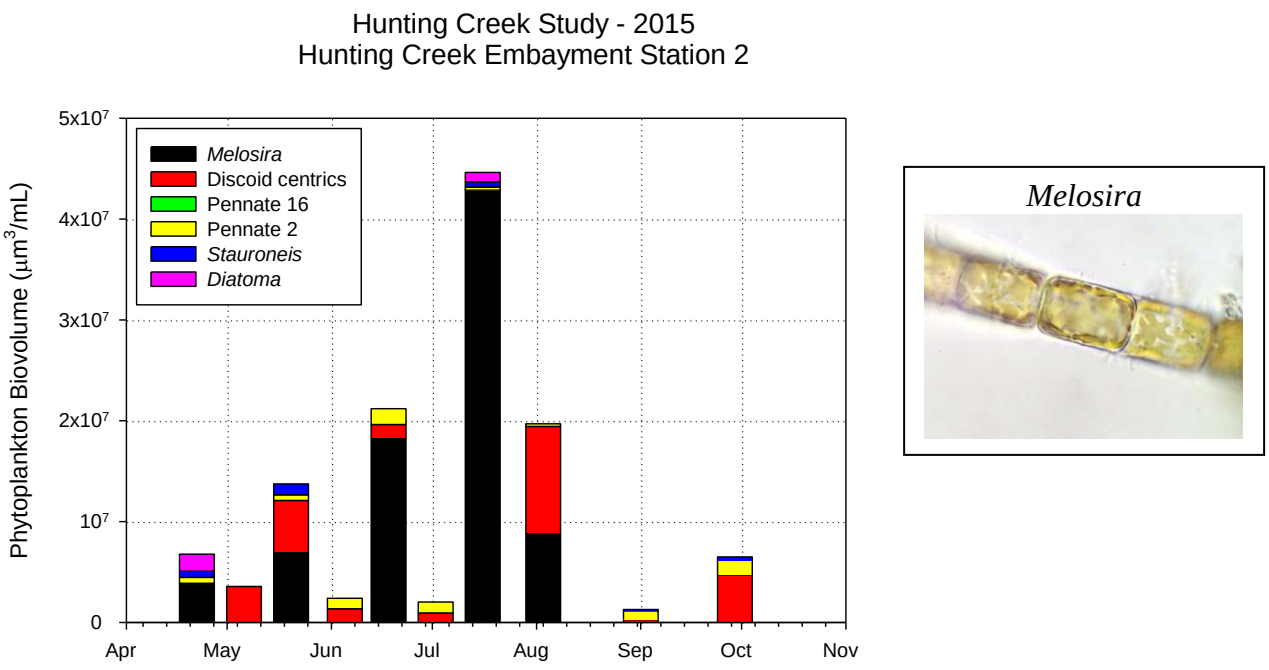


Figure 49. Phytoplankton Biovolume (um³/mL) by Dominant Diatom Taxa. Hunting Creek.

On many dates in 2015, *Melosira* was the overwhelming dominant in biovolume with discoid centrics sometimes co-dominant (Figure 49). In the river, *Melosira* was again dominant when it was present (Figure 50). Pennates were dominant in early August.

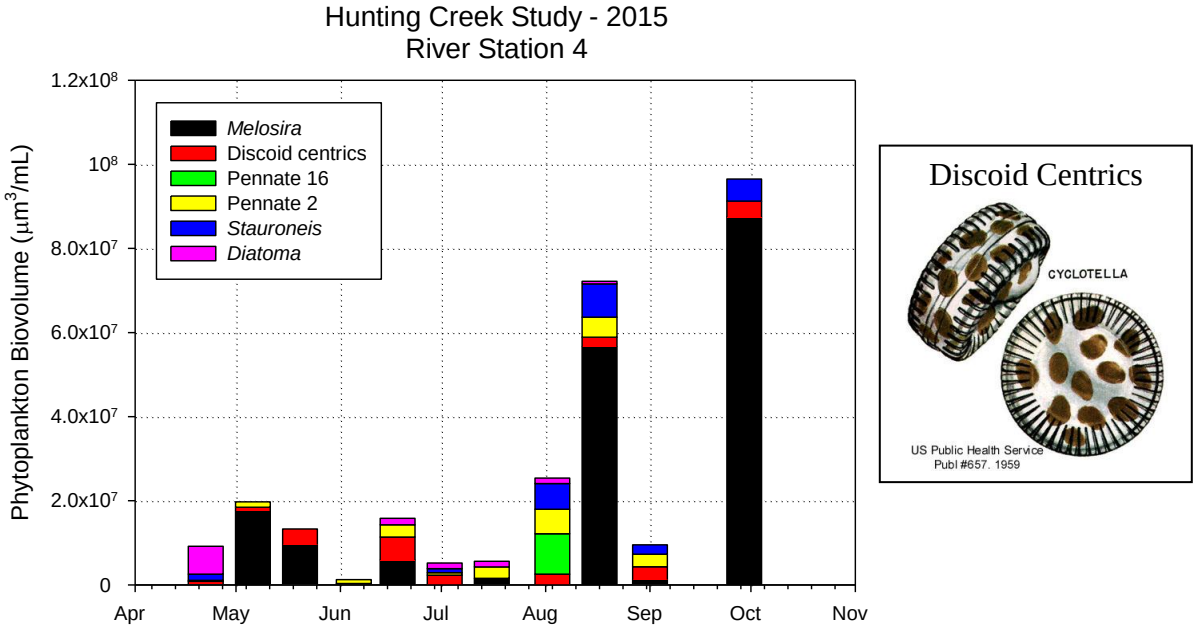


Figure 50. Phytoplankton Biovolume (um³/mL) by Dominant Diatom Taxon. River.

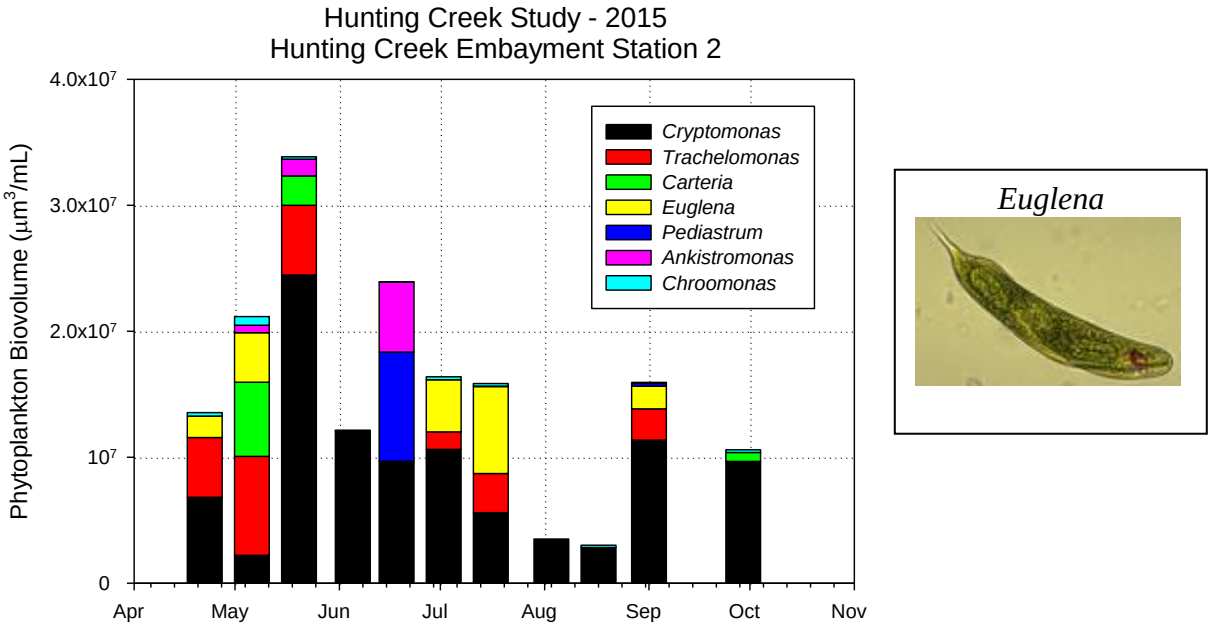


Figure 51. Phytoplankton Biovolume (um³/mL) by Dominant Other Taxa. Hunting Creek.

The cryptophyte *Cryptomonas* was the most important component of biovolume in the Other phytoplankton in Hunting Creek during 2015 (Figure 51). *Trachelomonas*, *Carteria*, *Euglena*, *Pediastrum* and *Ankistrodesmus* were important on some dates. In the river *Cryptomonas* was much less dominant (Figure 52). *Trachelomonas* and *Carteria* were dominant on some occasions.

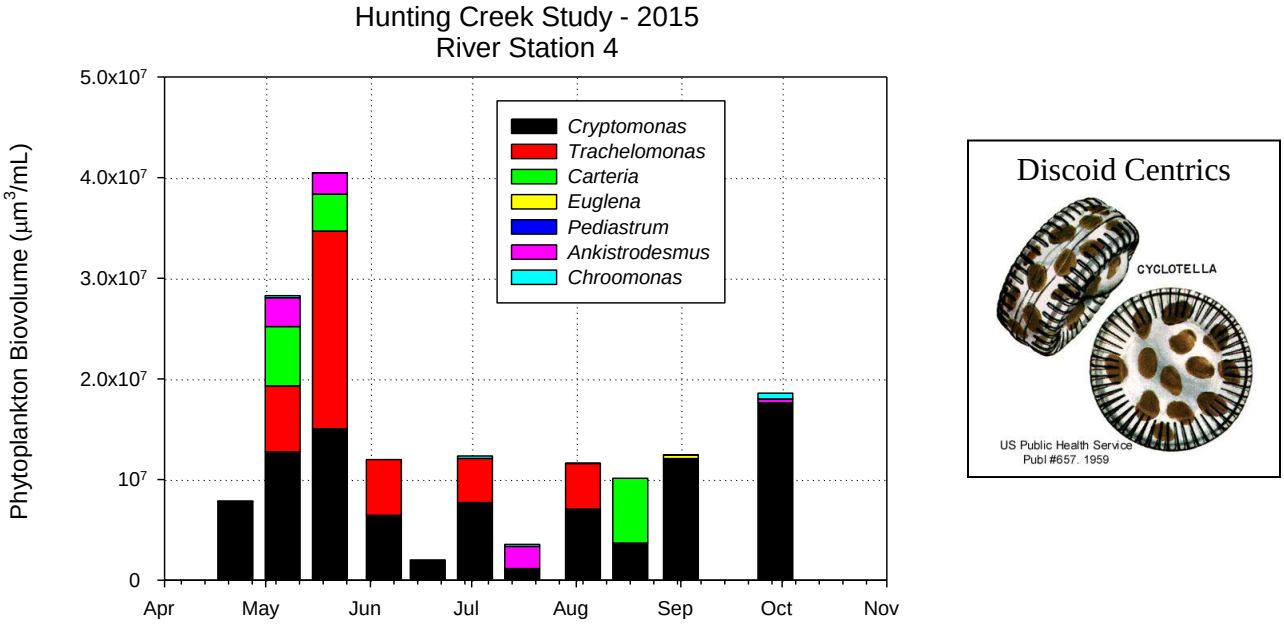


Figure 52. Phytoplankton Biovolume (um³/mL) by Dominant Other Taxon. River.

D. Zooplankton – 2014

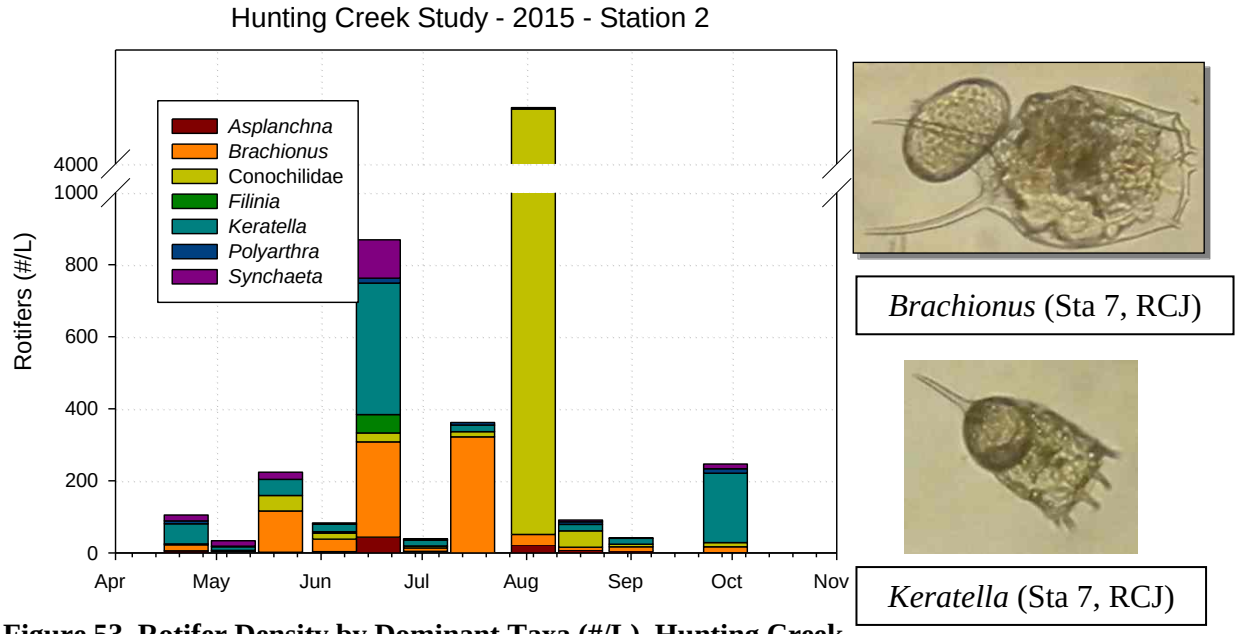


Figure 53. Rotifer Density by Dominant Taxa (#/L). Hunting Creek.

In Hunting Creek, rotifers exhibited a general, but not consistent increase through mid June. They decreased strongly in early July corresponding to the high flow event (Figure 53). *Brachionus* was generally dominant during this period and immediately after in late July. In August there was a strong surge of Conochilidae. In the river *Brachionus* and *Keratella* were co-dominant all year (Figure 54). Again, a strong decline was observed in early July corresponding with the high flow event.

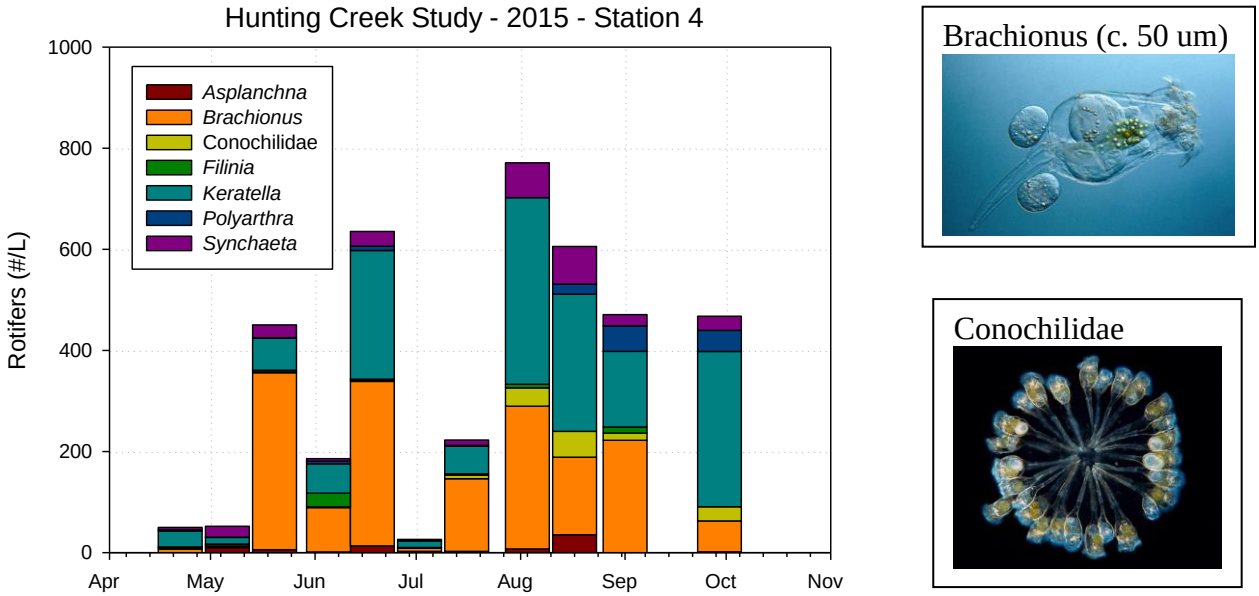
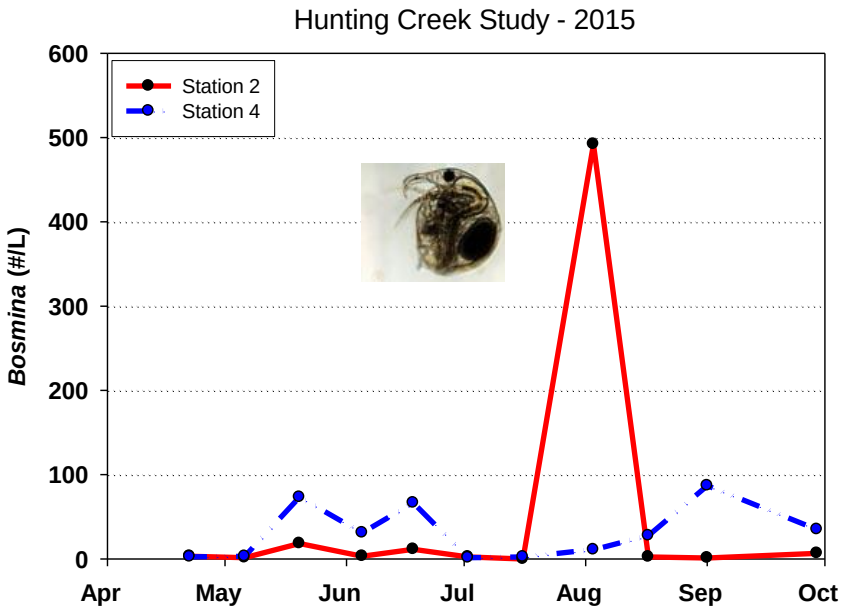


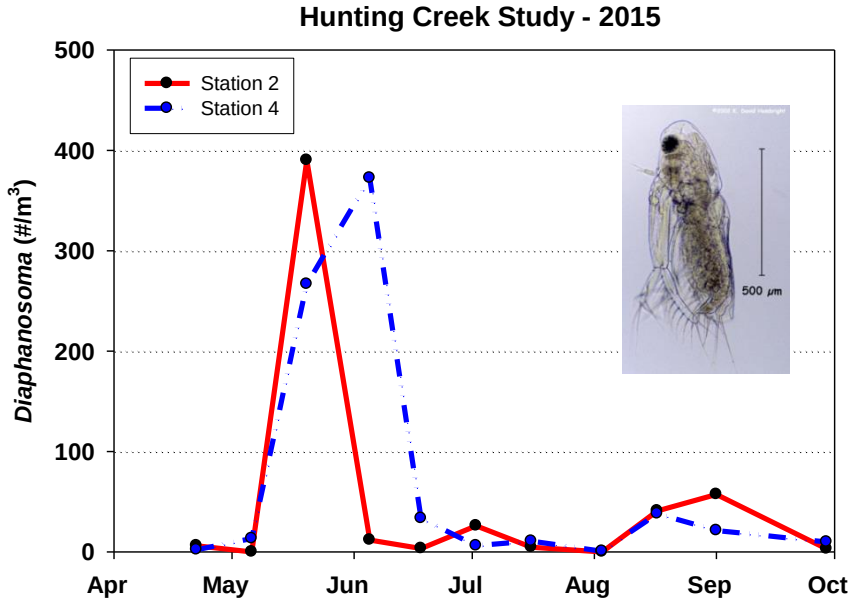
Figure 54. Rotifer Density by Dominant Taxa (#/L). River.



Bosmina is a small-bodied cladoceran, or "waterflea", which is common in lakes and freshwater tidal areas. It is typically the most abundant cladoceran with maximum numbers generally about 100-1000 animals per liter. Due to its small size and relatively high abundances, it is enumerated in the micro-zooplankton samples. *Bosmina* can graze on smaller phytoplankton cells, but can also utilize some cells from colonies by knocking them loose.

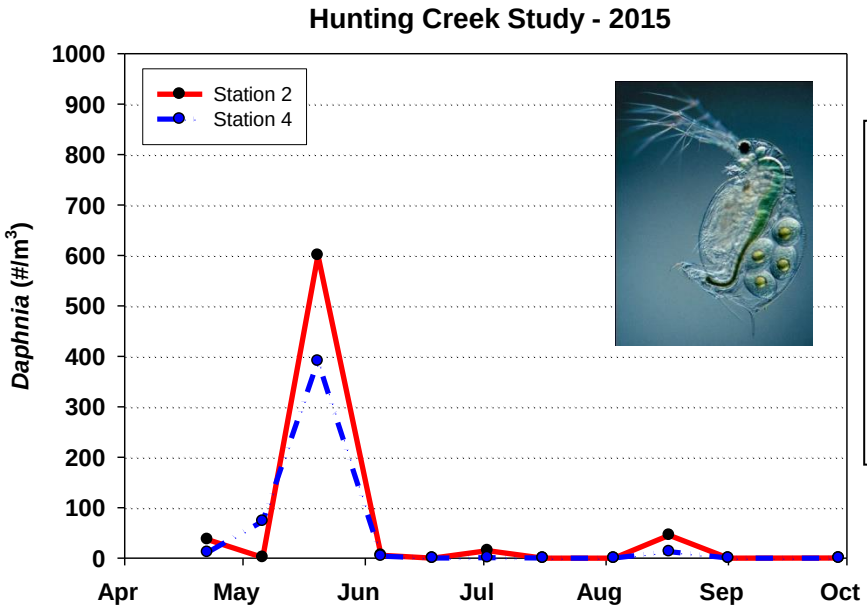
Figure 55. *Bosmina* Density by Station (#/L).

In 2015 the small cladoceran *Bosmina* was generally present at low densities at both stations (Figure 55). The exception was a strong surge in densities in early August at the embayment station. *Diaphanosoma*, typically the most abundant larger cladoceran in Gunston Cove, was present at relatively low densities peaking at about 400/m^3 (Figure 56). At the embayment site the peak occurred in late May, but the higher levels extended into early June in the river mainstem.



Diaphanosoma is the most abundant larger cladoceran found in the tidal Potomac River. It generally reaches numbers of 1,000-10,000 per m^3 (which would be 1-10 per liter). Due to their larger size and lower abundances, *Diaphanosoma* and the other cladocera are enumerated in the macrozooplankton samples. *Diaphanosoma* prefers warmer temperatures than some cladocera and is often common in the summer.

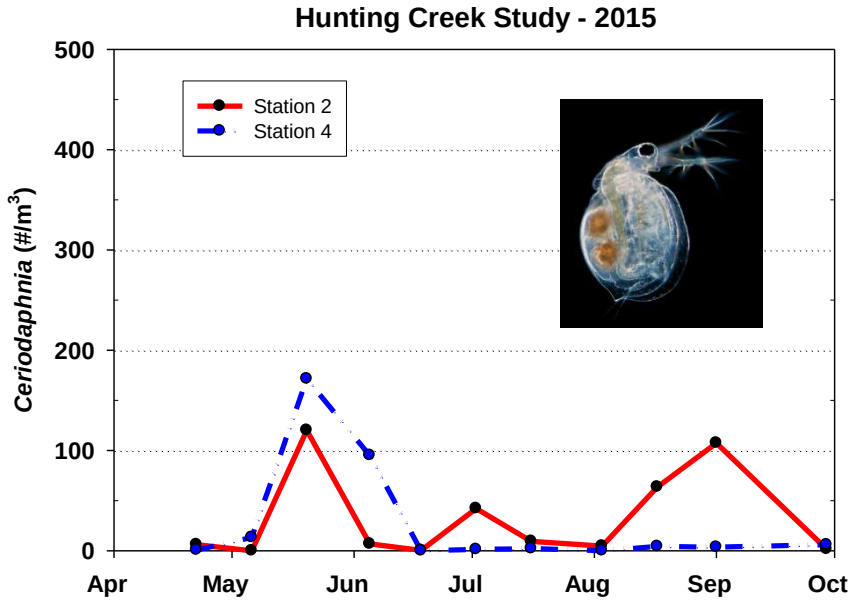
Figure 56. *Diaphanosoma* Density by Station (#/m^3).



Daphnia, the common waterflea, is one of the most efficient grazers of phytoplankton in freshwater ecosystems. In the tidal Potomac River it is present, but has not generally been as abundant as *Diaphanosoma*. It is typically most common in spring.

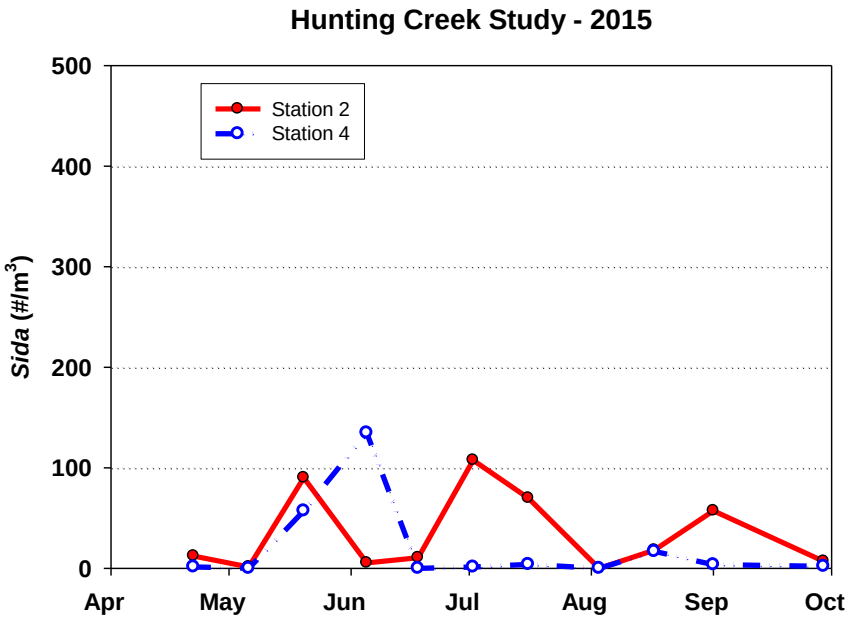
Figure 57. *Daphnia* Density by Station (#/m^3).

Daphnia was moderately abundant in the study area in late May 2015. In Hunting Creek peak values 600/m^3 with about 500/m3 observed in the river mainstem (Figure 57). *Ceriodaphnia* was also present in the study area, but at lower densities (Figure 58). Levels peaked at 100-200/m^3 in mid May and early September in Hunting Creek and at a similar level in the river in mid May.



Ceriodaphnia, another common large-bodied cladoceran, is usually present in numbers similar to *Daphnia*. Like all waterfleas, the juveniles look like miniature adults and grow through a series of molts to a larger size and finally reach reproductive maturity. Most reproduction is asexual except during stressful environmental conditions.

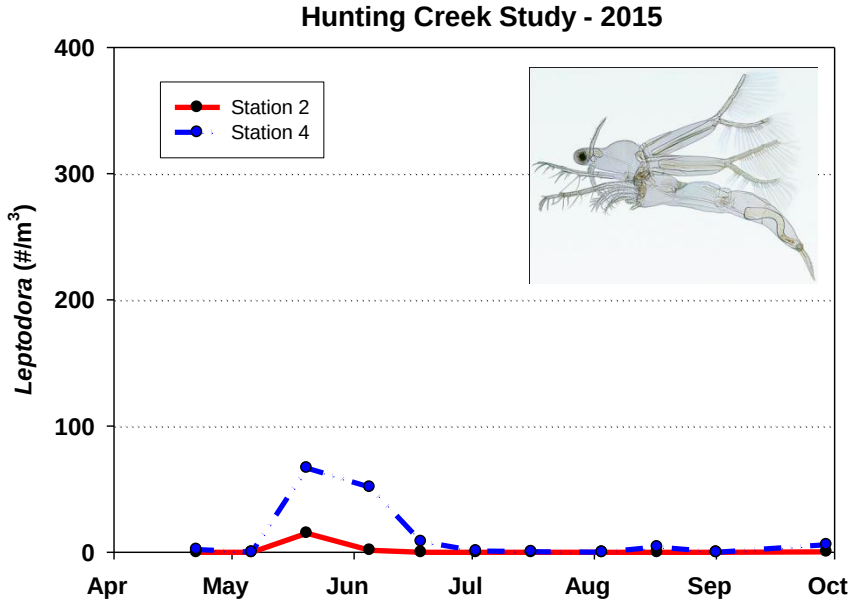
Figure 58. *Ceriodaphnia* Density by Station (#/m^3).



Moinadaphnia is another waterflea that is often observed in the tidal Potomac River. Like the other cladocera mentioned so far, *Moinadaphnia* grazes on phytoplankton to obtain its food supply.

Figure 59. *Sida* Density by Station (#/m^3).

Sida was found at moderate densities on several occasions at the Hunting Creek embayment station (Figure 59). It was also present in moderate numbers in the river mainstem in early June. *Leptodora*, the large cladoceran predator, was very scarce in Hunting Creek, but attained appreciable densities in the river in late May (Figure 60).



Leptodora is substantially larger than the other cladocera mentioned. Also different is its mode of feeding – it is a predator on other zooplankton. It normally occurs for brief periods in the late spring or early summer.

Figure 60. *Leptodora* Density by Station (#/m^3).

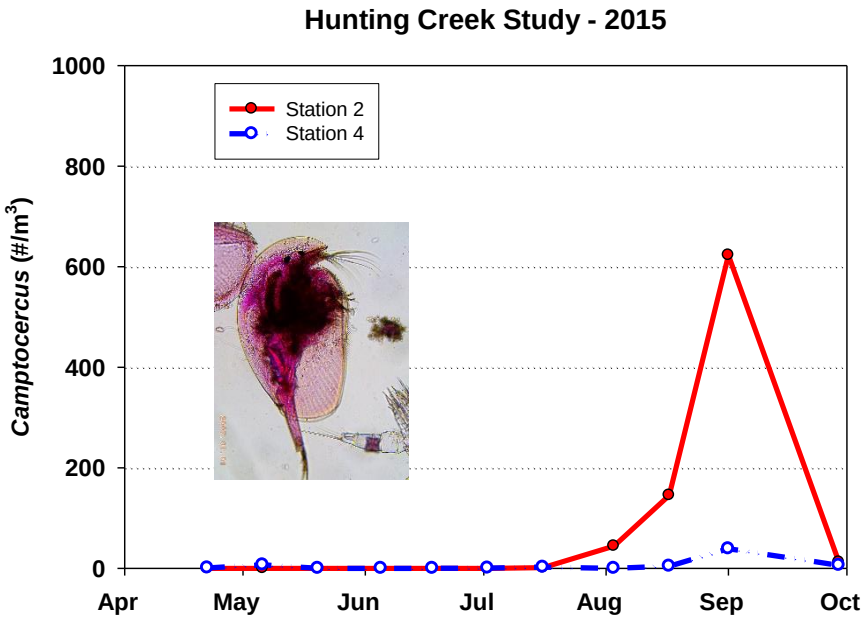


Figure 61. *Camptocercus* Density by Station (#/m^3). (photo: L. Birsa from HC samples)

Two new cladoceran taxa are being highlighted in 2015 data (Figures 61 and 62). Both are taxa associated with submersed aquatic vegetation (SAV). Both were found almost exclusively at the Hunting Creek embayment (Station 2) and both were mainly observed in late August and early September when SAV is most extensive and dominates the entire tidal creek area.

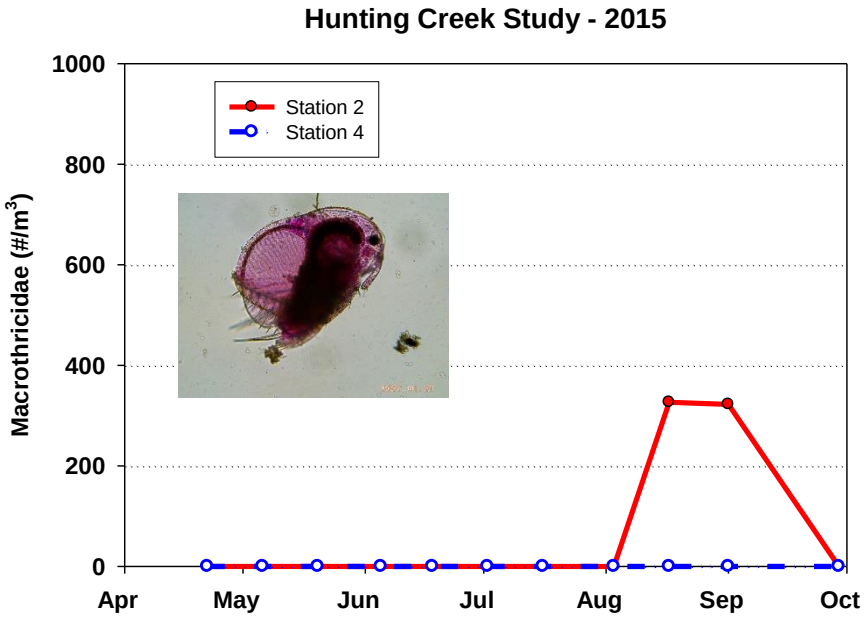
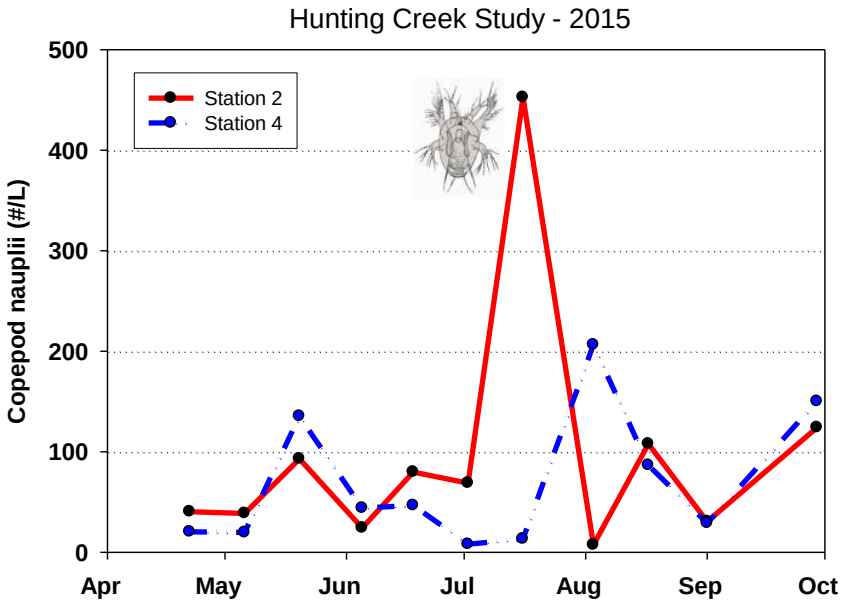


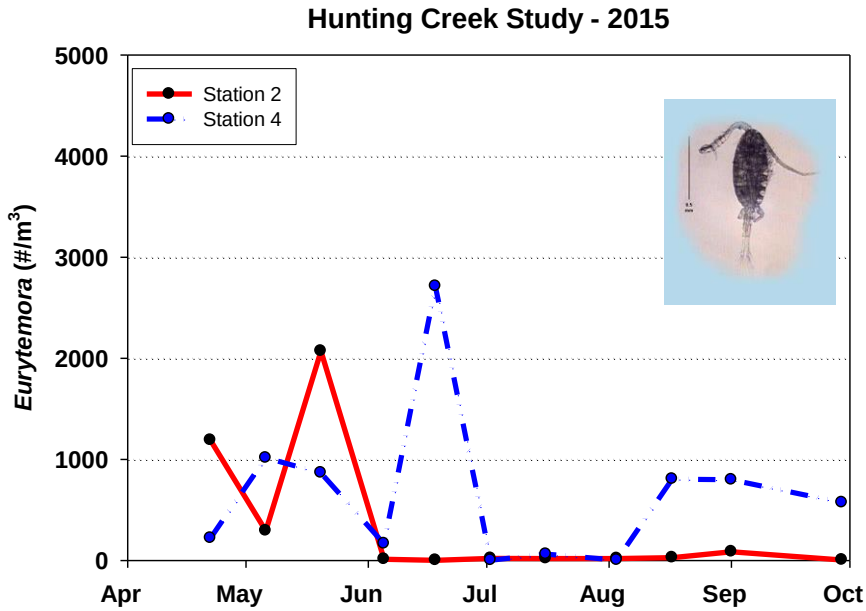
Figure 62. Macrothricid Density by Station (#/m^3). (photo: L. Birsa from HC samples)



Copepod eggs hatch to form an immature stage called a nauplius. The nauplius is a larval stage that does not closely resemble the adult and the nauplii of different species of copepods are not easily distinguished so they are lumped in this study. Copepods go through 5 naupliar molts before reaching the copepodid stage which is morphologically very similar to the adult. Because of their small size and high abundance, copepod nauplii are enumerated in the micro-zooplankton samples.

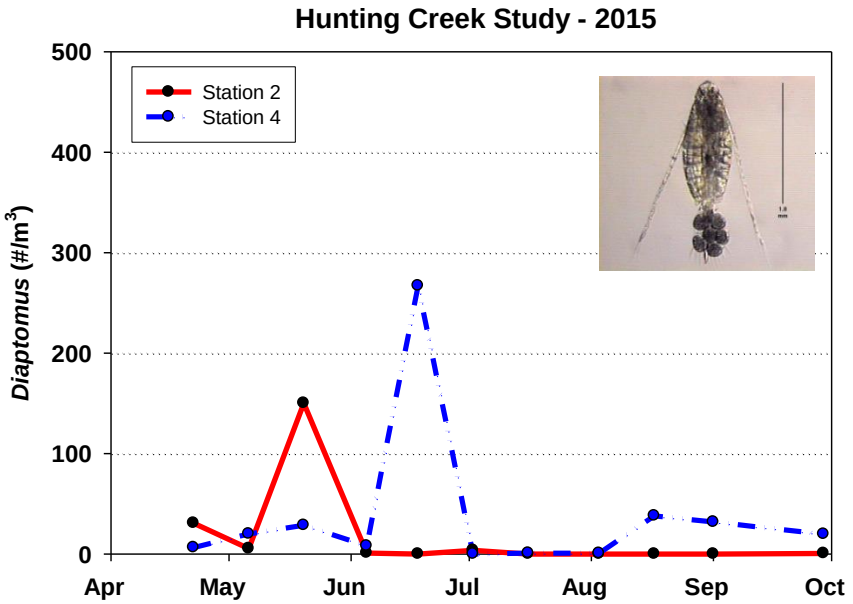
Figure 63. Copepod Nauplii Density by Station (#/L).

Copepod nauplii were present at low to moderate levels in April, May, and June at both sites (Figure 61). An increase in Hunting Creek in mid July to over 400/L was followed by a peak at the river mainstem station of 200/L in early August. *Eurytemora* was common in Hunting Creek in April and May, but declined thereafter. In the river, densities were highest in late June at about 3000/m^3 (Figure 62).



Eurytemora affinis is a large calanoid copepod characteristic of the freshwater and brackish areas of the Chesapeake Bay. *Eurytemora* is a cool water copepod which often reaches maximum abundance in the late winter or early spring. Included in this graph are adults and those copepodids that are recognizable as *Eurytemora*.

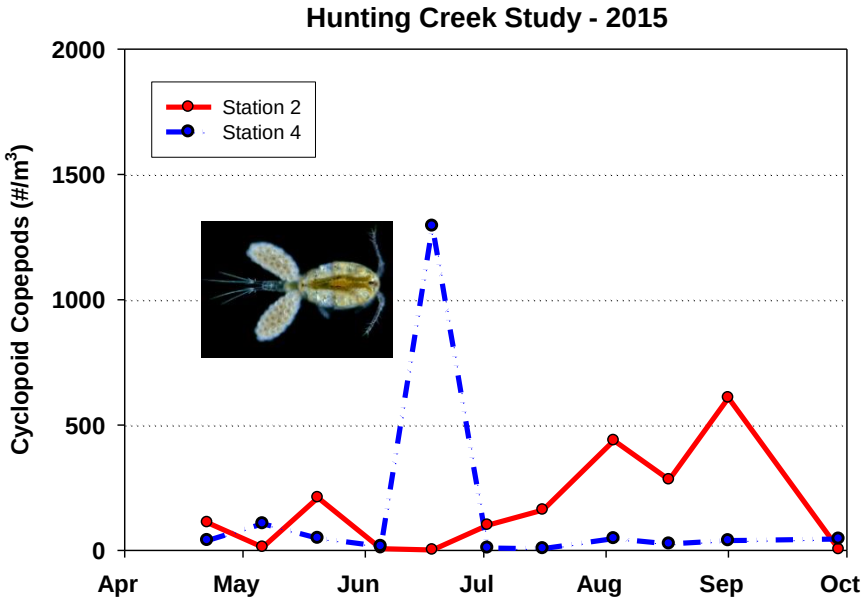
Figure 64. *Eurytemora* Density by Station (#/m^3).



Diaptomus pallidus is a calanoid copepod often found in moderate densities in the Gunston Cove area. *Diaptomus* is an efficient grazer of algae, bacteria, and detrital particles in freshwater ecosystems Included in this graph are adults and those copepodids that are recognizable as *Diaptomus*.

Figure 65. *Diaptomus* Density by Station (#/m^3).

Diaptomus was more common in Hunting Creek in May and in the river in June (Figure 63). Cyclopoid copepods exhibited a seasonal peak at both sites attaining highest values in mid June in the river and in early September in Hunting Creek (Figure 65).



Cyclopoids are the other major group of planktonic copepods. Cyclopoids feed on individual particles suspended in the water including small zooplankton as well as phytoplankton. In this study we have lumped all copepodid and adult cyclopoids together.

Figure 66. Cyclopoid Copepods by Station (#/m^3).

E. Ichthyoplankton - 2015

We collected 14 samples (7 at Station ARE 2 and 7 at Station ARE 4) during the months April through July and found an average larval density of 2138 larvae per 10 m^3 (Table 4). The dominant species was gizzard shad (*Dorosoma cepedianum*) with an average larval density of 562 larvae per $10m^3$. The taxon Clupeidae had the second highest density with 552 larvae per $10m^3$, which is comprised of clupeids (*Alosa* or *Dorosoma* sp.) that could not be identified to a lower taxonomic level. *Alosa aestivalis* (blueback herring) and *Alosa pseudoharengus* (alewife) were present in high densities as well: 473 and 393 larvae per $10m^3$ respectively. Other clupeids present that could positively be identified to the species level are hickory shad at 30.28 per $10m^3$, and American shad at 3.04 larvae per $10m^3$. A different taxon with relatively high representation is white perch with 24.08 larvae per $10m^3$. In addition *Morone* sp., which is either white perch or striped bass, was found at 16.31 larvae per $10m^3$; most if not all of those are likely white perch.

Table 4. The average larval density (#/$10m^3$) in Hunting Creek (Sta. 2) and the Potomac River (Sta. 4) in 2015.

Taxon	Species	Station 2	Station 4	Average
Alosa aestivalis	blueback herring	619.19	327.03	473.11
Alosa mediocris	hickory shad	45.66	14.89	30.28
Alosa pseudoharengus	alewife	504.28	282.54	393.41
Alosa sapidissima	American shad	4.07	2.00	3.04
Cyprinidae sp.	carp sp.	5.55	1.58	3.57
Clupeidae	herring or shad	861.18	243.60	552.39
Dorosoma cepedianum	gizzard shad	283.11	841.14	562.13
Lepomis sp.	sunfish	4.90	0	2.45
Menidia beryllina	inland silverside	16.28	1.78	9.03
Morone americana	white perch	19.45	28.71	24.08
Morone sp.	perch or bass	9.38	23.23	16.31
Egg	Unidentified	3.40	4.48	3.94
Unidentified	Unidentified	73.30	56.04	64.67

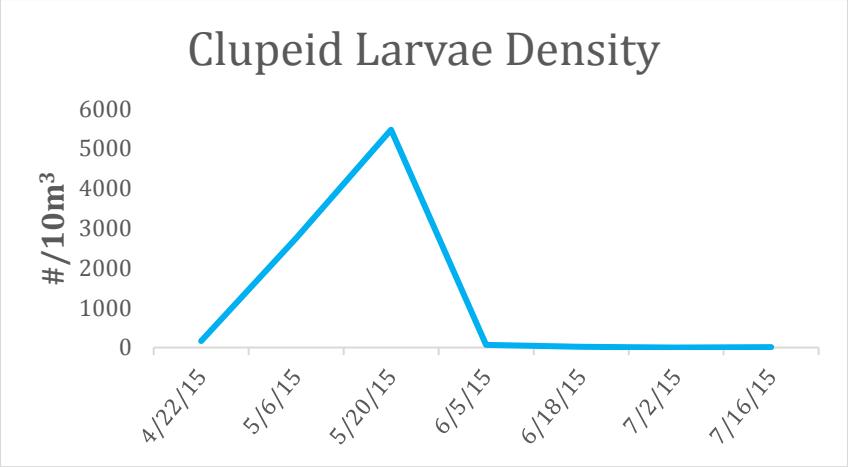


Figure 67. Density of clupeid larvae per $10m^3$.

Clupeid larvae in Figure 67 include blueback herring, hickory shad, alewife, American shad and gizzard shad. These have similar spawning patterns so they are lumped into one group for this analysis. Clupeids started to appear in the sample at the end of April, increased to a maximum of 5472 larvae per 10 m^3 in mid-May, and decreased again at the start of June (Figure 67). Of these clupeids, alewife and blueback herring are the two species that make up river herring, of which we describe the spawning population study at the end of this report. White perch and (potentially) striped bass larvae attained highest density in mid-May as well (Figure 68). The group of larvae that are not clupeids or morone species are dominated by unidentified larvae (Figure 69. Highest densities of unidentified larvae were found in May as well. The unidentified larvae were not intact unknown species, but larvae too mangled for proper identification. Because of the high density of clupeid larvae, most unidentified larvae are likely to be clupeids as well.

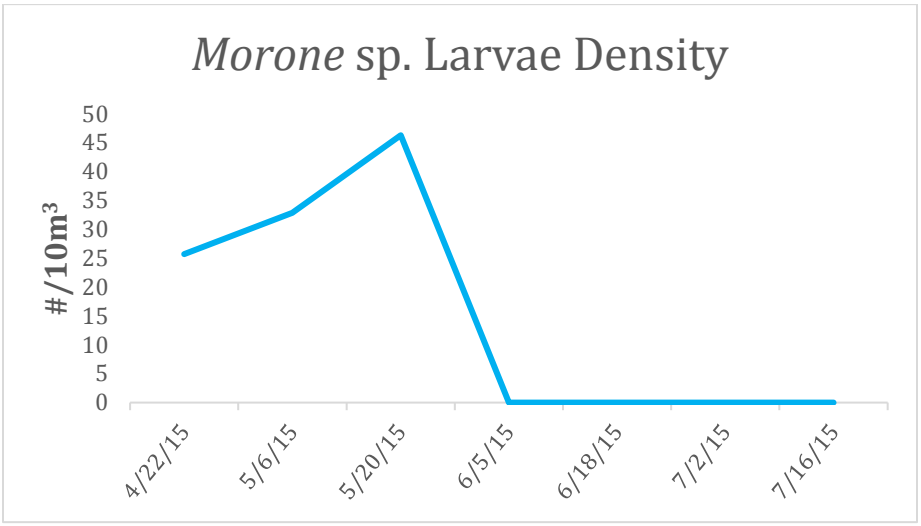


Figure 68. Density of *Morone* sp. (white perch and striped bass) per 10m^3.

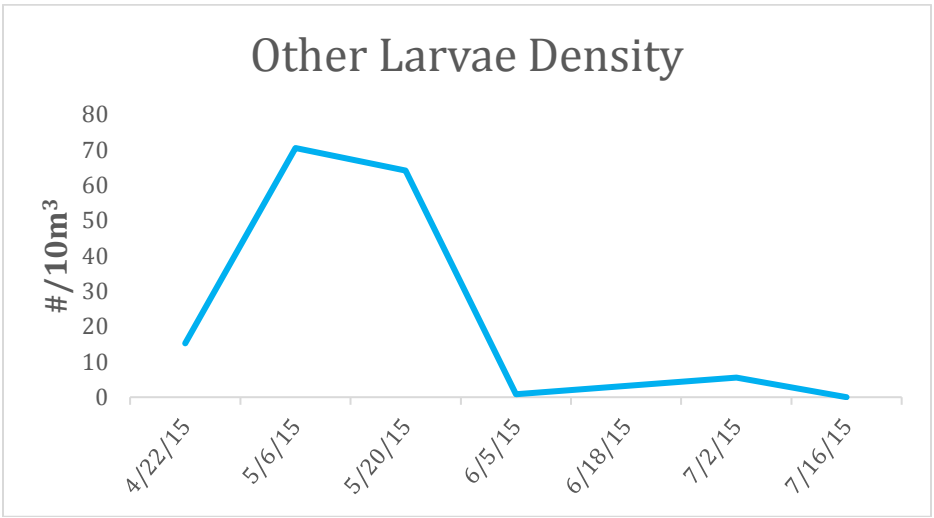


Figure 69. Density of other larvae per 10m^3.

F. Adult and juvenile fishes – 2015

Trawls

Trawl sampling was conducted between April 21 and September 16 at station 3 and 4. A total of 821 fishes comprising at least 22 species were collected with trawls (Table 5). Collections were dominated by white perch (63%). The second most abundant species caught was American shad (6.7%). Other abundant species (annual total >2%) included: spottail shiner (6.1%), tessellated darter (4.6%), alewife (3.3%), and bluegill (2.3%). Other species were observed at lower abundances (Tables 5 and 6).

Table 5. Adult and juvenile fish collected by trawling. Hunting Creek - 2015

Species	Common name	Abundance	% total
Alosa aestivalis	blueback herring	2	0.2
Alosa pseudoharengus	alewife	27	3.3
Alosa sapidissima	American shad	55	6.7
Alosa sp.	herring or shad	42	5.1
Ameiurus nebulosus	brown bullhead	3	0.4
Anchoa mitchilli	bay anchovy	7	0.9
Carassius auratus	goldfish	1	0.1
Dorosoma cepedianum	gizzard shad	2	0.2
Etheostoma olmstedi	tessellated darter	38	4.6
Fundulus diaphanus	banded killifish	13	1.6
Ictalurus furcatus	blue catfish	4	0.5
Ictalurus punctatus	channel catfish	2	0.2
Lepomis auritus	redbreast sunfish	1	0.1
Lepomis gibbosus	pumpkinseed	8	1.0
Lepomis macrochirus	bluegill	19	2.3
Lepomis microlophus	redear sunfish	2	0.2
Lepomis sp.	sunfish	7	0.9
Menidia berylina	inland silverside	8	1.0
Morone americana	white perch	520	63.3
Morone saxatilis	striped bass	1	0.1
Notemigonus crysoleucas	golden shiner	1	0.1
Notropis hudsonius	spottail shiner	50	6.1
Perca flavescens	yellow perch	7	0.9
Pomoxis nigromaculatus	black crappie	1	0.1
	Total	**821**	**100**

Table 6. Adult and juvenile fish collected by trawling. Hunting Creek study - 2015

		4/21	5/5	5/27	6/10	6/30	7/8	7/22	8/12	8/20	9/16	**Total**
Alosa aestivalis	blueback herring	0	0	0	0	0	0	0	1	1	0	2
Alosa pseudoharengus	alewife	0	0	11	14	2	0	0	0	0	0	27
Alosa sapidissima	American shad	0	0	20	18	0	7	4	0	1	5	55
Alosa sp.	herring or shad	0	0	42	0	0	0	0	0	0	0	42
Ameiurus nebulosus	brown bullhead	1	1	0	0	1	0	0	0	0	0	3
Anchoa mitchilli	bay anchovy	0	0	0	0	0	0	0	0	0	7	7
Carassius auratus	goldfish	0	0	0	0	1	0	0	0	0	0	1
Dorosoma cepedianum	gizzard shad	0	0	0	2	0	0	0	0	0	0	2
Etheostoma olmstedi	tessellated darter	0	0	7	20	11	0	0	0	0	0	38
Fundulus diaphanus	banded killifish	3	1	5	4	0	0	0	0	0	0	13
Ictalurus furcatus	blue catfish	0	0	0	0	3	0	0	1	0	0	4
Ictalurus punctatus	channel catfish	0	0	0	0	0	0	0	0	0	2	2
Lepomis auritus	redbreast sunfish	0	0	1	0	0	0	0	0	0	0	1
Lepomis gibbosus	pumpkinseed	0	1	0	6	1	0	0	0	0	0	8
Lepomis macrochirus	bluegill	1	0	0	0	2	0	16	0	0	0	19
Lepomis microlophus	redear sunfish	0	1	0	0	1	0	0	0	0	0	2
Lepomis sp.	sunfish	0	0	0	0	0	0	7	0	0	0	7
Menidia berylina	inland silverside	0	0	0	8	0	0	0	0	0	0	8
Morone americana	white perch	13	23	233	177	33	3	28	6	4	0	520
Morone saxatilis	striped bass	0	0	0	0	0	0	0	1	0	0	1
Notemigonus crysoleucas	golden shiner	0	0	0	1	0	0	0	0	0	0	1
Notropis hudsonius	spottail shiner	10	0	8	11	4	2	9	0	6	0	50
Perca flavescens	yellow perch	1	1	0	4	0	0	1	0	0	0	7
Pomoxis nigromaculatus	black crappie	0	0	1	0	0	0	0	0	0	0	1
	Total	**29**	**28**	**328**	**265**	**59**	**12**	**65**	**9**	**12**	**14**	**821**

The highest catch occurred on May 27, and was due to the high abundance of white perch in that trawl sample (Tables 6). Most catches by far occurred at site 3, which means species actively pursue the shallower habitat in Hunting Creek from the Potomac River mainstem (Table 7). In total numbers and species richness of fish, station 3 dominated station 4 with 758 individuals from at least 20 species. Stations 4 had 63 individuals from 7 species. There were no centrarchids (sunfishes) in station 4, which are species know to be associated with submerged aquatic vegetation, while 37 individuals from at least 4 species of sunfish were found in station 3. Of the invasive blue catfish one was found in station 3, and 3 were found in station 4.

Table 7. Adult and juvenile fish collected by trawling. Hunting Creek study - 2015

Species	Common name	Station 3	Station 4
Alosa aestivalis	blueback herring	0	2
Alosa pseudoharengus	alewife	27	0
Alosa sapidissima	American shad	38	17
Alosa sp.	herring or shad	42	0
Ameiurus nebulosus	brown bullhead	1	2
Anchoa mitchilli	bay anchovy	0	7
Carassius auratus	goldfish	1	0
Dorosoma cepedianum	gizzard shad	2	0
Etheostoma olmstedi	tessellated darter	38	0
Fundulus diaphanus	banded killifish	13	0
Ictalurus furcatus	blue catfish	1	3
Ictalurus punctatus	channel catfish	0	2
Lepomis auritus	redbreast sunfish	1	0
Lepomis gibbosus	pumpkinseed	8	0
Lepomis macrochirus	bluegill	19	0
Lepomis microlophus	redear sunfish	2	0
Lepomis sp.	sunfish	7	0
Menidia beryllina	inland silverside	8	0
Morone americana	white perch	490	30
Morone saxatilis	striped bass	1	0
Notemigonus crysoleucas	golden shiner	1	0
Notropis hudsonius	spottail shiner	50	0
Perca flavescens	yellow perch	7	0
Pomoxis nigromaculatus	black crappie	1	0
	Total	**758**	**63**

The pattern of abundance shows one dominant species, white perch, with about six other species having moderately high abundance (Figure 70 A and B). At both stations, white perch made up a significant proportion of the total catch, and was by far the species with the highest abundance in station 3. The six most abundant species after white perch varied in representation across the two stations (Figure 70 A and B). Station 3 was overall the most productive site of the two, with a total abundance more than 10 times higher than station 4.

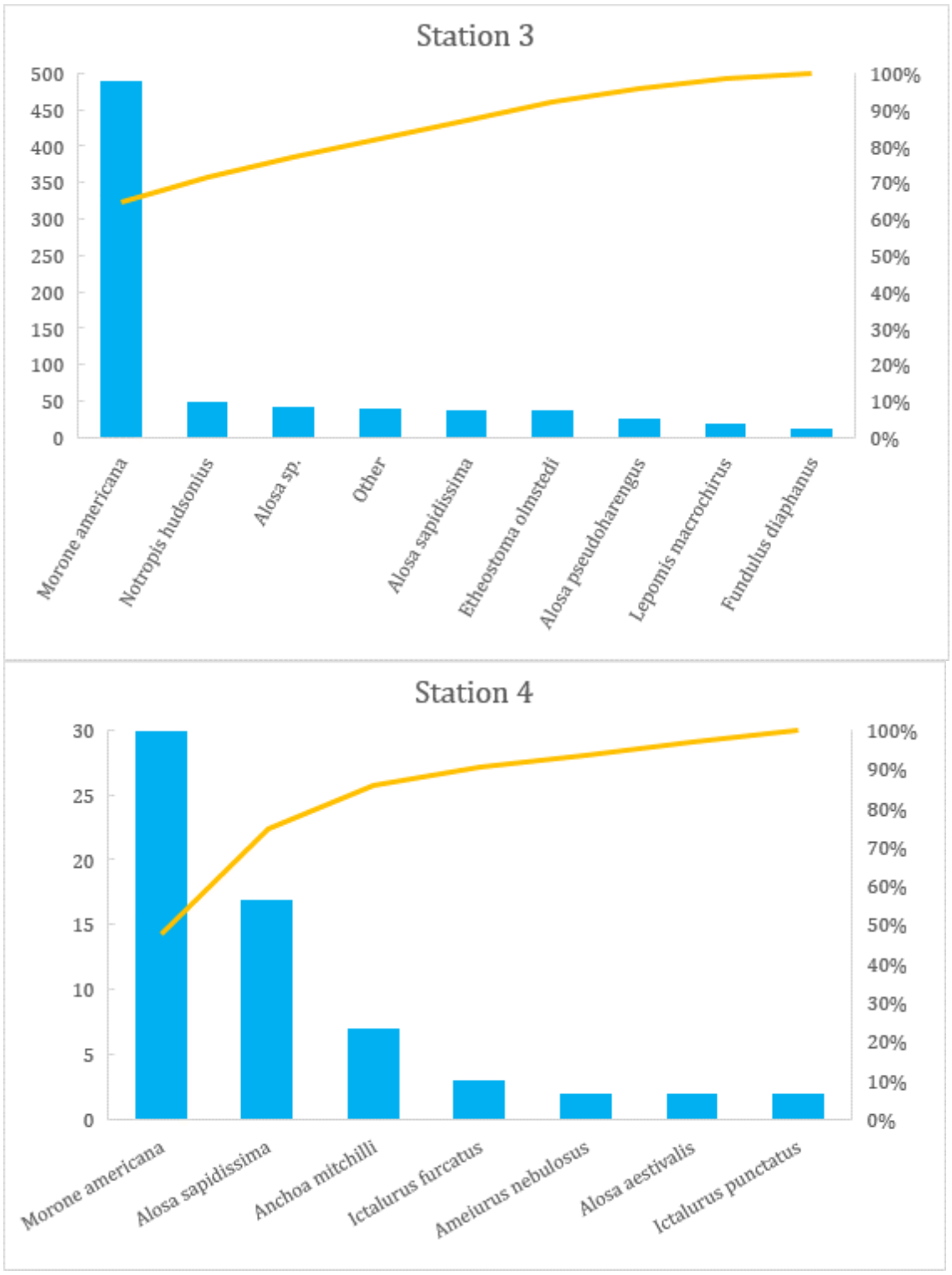


Figure 70A and B. Pareto chart of adult and juvenile fishes collected by trawling. Dominant species by station in total abundance and cumulative percentage of total for Station 3 (top) and Station 4 (bottom).

When the six most dominant species are displayed as catch per month, it can be seen again that white perch was the most common species, and was present from April to August (Figure 71 A and B). Spottail shiner was common in catches too and present from April to August, but less abundant than spottail shiner, and with a different distribution. The most productive months by far were May and June, mostly due to high catches of white perch.

White perch (*Morone americana*) is the dominant species in Hunting Creek, and continues to be an important commercial and popular game fish. Adults grow to over 30 cm long. Sexual maturity begins the second year at lengths greater than 9 cm. As juveniles they feed on zooplankton and macrobenthos, but as they get larger consume fish as well.

Spottail shiner (*Notropis hudsonius* is a common fish in the open waters of Hunting Creek. Spawning occurs throughout the warmer months. It reaches sexual maturity at about 5.5 cm and may attain a length of 10 cm. They feed primarily on benthic invertebrates and occasionally on algae and plants.

Trawling collects fish that are located in the open water near the bottom. Due to the shallowness of Hunting Creek, the volume collected is a substantial part of the water column. However, in the river channel, the near bottom habitat through which the trawl moves is only a small portion of the water column. Fishes tend to concentrate near the bottom or along shorelines rather than in the upper portion of the open water.

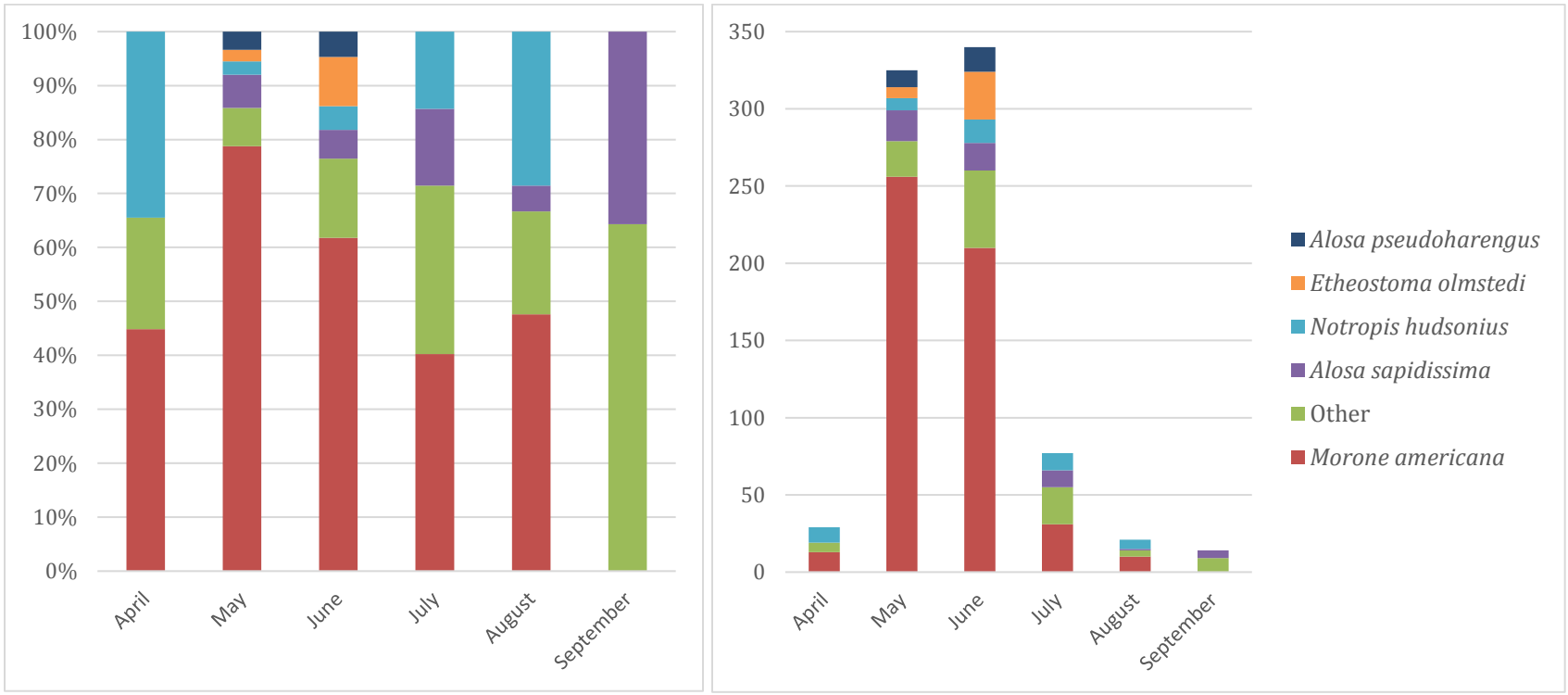


Figure 71 A&B. Adult and juvenile fishes collected by trawling. Dominant species by month in percentage of total (A) and total abundance (B).

Seines

Table 8. Adult and juvenile fish collected by seining. Hunting Creek study – 2015

Species	Common name	Abundance	% Total
Alosa aestivalis	blueback herring	10	0.30
Alosa pseudoharengus	alewife	1	0.03
Alosa sapidissima	American shad	108	3.20
Alosa sp.	herring or shad	13	0.39
Anguilla rostrata	American eel	2	0.06
Carassius auratus	goldfish	1	0.03
Carpiodes cyprinus	quilback	2	0.06
Cyprinella spiloptera	spotfin shiner	1	0.03
Cyprinus carpio	common carp	1	0.03
Dorosoma cepedianum	gizzard shad	1	0.03
Etheostoma olmstedi	tessellated darter	1	0.03
Fundulus diaphanus	banded killifish	2710	80.34
Fundulus heteroclitus	mummichog	174	5.16
Gambusia holbrooki	eastern mosquitofish	19	0.56
Hybognathus regius	eastern silvery minnow	31	0.92
Lepisosteus osseus	longnose gar	3	0.09
Lepomis gibbosus	pumpkinseed	3	0.09
Lepomis macrochirus	bluegill	2	0.06
Lepomis microlophus	redear sunfish	3	0.09
Menidia beryllina	inland silverside	65	1.93
Micropterus dolomieu	smallmouth bass	9	0.27
Morone americana	white perch	166	4.92
Morone saxatilis	striped bass	1	0.03
Notemigonus crysoleucas	golden shiner	12	0.36
Notropis hudsonius	spottail shiner	28	0.83
Pomoxis nigromaculatus	black crappie	3	0.09
Strongylura marina	Atlantic needlefish	3	0.09
	Total	**3373**	**100**

Seine sampling was conducted between April 21 and September 16 at Station 5 and Station 6. As planned, one sampling trip per month was performed in April and September, and two sampling trips per month from May to August.

The two seines stations (Station 5 and 6; Figure 1) were selected as sites with shallow sloping shorelines that would enable us to tow a beach seine. The net was towed up onto the beach unless high water completely submerged the beach. In those cases, the net was towed into the boat.

A total of 20 seine samples were conducted (10 per station), comprising 3373 fishes of at least 26 species (Table 8). By far the most dominant species in seine catches was banded killifish (80.34%), followed by mummichogs (5.16%), which are both killifishes. Other species that were relatively abundant were white perch (4.92%), American shad (3.2%), and silverside (1.93%). Other species occurred at low abundances (Table 8).

Banded killifish was present and dominant in all months sampled (Table 9; Figure 73A&B). The most productive month was May due to the high numbers of banded killifish present that month. In both stations the high majority of all fish collected was banded killifish (Table 10). The Pareto charts of station 5 and 6 (Figure 72 A&B) indicate with very shallow slopes of the cumulative percent of the catch that banded killifish is highly dominant, with relatively small contributions of other species. Other species with relatively high abundance (> 10 specimens) were mummichog, white perch, American shad, inland silverside, eastern silvery minnow, spottail shiner, eastern musquitofish, and golden shiner.

Table 9. Adult and juvenile fish collected by seining. Hunting Creek study - 2015

Species	Common name	4/21	5/5	5/27	6/10	6/30	7/8	7/22	8/12	8/20	9/16	**Total**
Alosa aestivalis	blueback herring	0	0	0	0	4	0	0	6	0	0	10
Alosa pseudoharengus	alewife	0	0	0	0	0	0	0	1	0	0	1
Alosa sapidissima	American shad	0	0	0	4	14	10	9	58	9	4	108
Alosa sp.	herring or shad	0	0	0	1	0	0	0	12	0	0	13
Anguilla rostrata	American eel	1	0	1	0	0	0	0	0	0	0	2
Carassius auratus	goldfish	0	0	0	0	0	0	1	0	0	0	1
Carpiodes cyprinus	quilback	0	0	0	2	0	0	0	0	0	0	2
Cyprinella spiloptera	spotfin shiner	0	0	0	0	0	1	0	0	0	0	1
Cyprinus carpio	common carp	1	0	0	0	0	0	0	0	0	0	1
Dorosoma cepedianum	gizzard shad	1	0	0	0	0	0	0	0	0	0	1
Etheostoma olmstedi	tessellated darter	0	1	0	0	0	0	0	0	0	0	1
Fundulus diaphanus	banded killifish	12	657	1172	544	15	113	47	40	63	47	2710
Fundulus heteroclitus	mummichog	0	11	14	5	2	0	55	87	0	0	174
Gambusia holbrooki	eastern mosquitofish	0	0	0	0	0	1	0	17	1	0	19
Hybognathus regius	eastern silvery minnow	0	1	0	0	1	1	0	0	0	28	31
Lepisosteus osseus	longnose gar	0	0	0	0	2	0	1	0	0	0	3
Lepomis gibbosus	pumpkinseed	1	1	0	0	0	0	0	0	0	1	3
Lepomis macrochirus	bluegill	0	0	0	0	0	0	1	0	1	0	2
Lepomis microlophus	redear sunfish	0	0	3	0	0	0	0	0	0	0	3
Menidia beryllina	silverside	19	38	2	1	1	4	0	0	0	0	65
Micropterus dolomieu	smallmouth bass	0	0	0	0	0	2	2	2	1	2	9
Morone americana	white perch	0	1	0	108	14	14	0	0	7	22	166
Morone saxatilis	striped bass	1	0	0	0	0	0	0	0	0	0	1
Notemigonus crysoleucas	golden shiner	8	3	0	0	0	1	0	0	0	0	12
Notropis hudsonius	spottail shiner	2	23	0	1	0	1	0	1	0	0	28
Pomoxis nigromaculatus	black crappie	0	1	0	0	2	0	0	0	0	0	3
Strongylura marina	Atlantic needlefish	0	0	0	3	0	0	0	0	0	0	3
	Total	**46**	**737**	**1192**	**669**	**55**	**148**	**116**	**224**	**82**	**104**	**3373**

Table 10. Adult and juvenile fish collected by seining. Hunting Creek study – 2015

Species	Common name	Station 5	Station 6
Alosa aestivalis	blueback herring	10	0
Alosa pseudoharengus	alewife	1	0
Alosa sapidissima	American shad	94	14
Alosa sp.	herring or shad	13	0
Anguilla rostrata	American eel	2	0
Carassius auratus	goldfish	0	1
Carpiodes cyprinus	quilback	0	2
Cyprinella spiloptera	spotfin shiner	1	0
Cyprinus carpio	common carp	1	0
Dorosoma cepedianum	gizzard shad	0	1
Etheostoma olmstedi	tessellated darter	0	1
Fundulus diaphanus	banded killifish	709	2001
Fundulus heteroclitus	mummichog	86	88
Gambusia holbrooki	eastern mosquitofish	16	3
Hybognathus regius	eastern silvery minnow	31	0
Lepisosteus osseus	longnose gar	2	1
Lepomis gibbosus	pumpkinseed	3	0
Lepomis macrochirus	bluegill	1	1
Lepomis microlophus	redear sunfish	3	0
Menidia beryllina	silverside	3	62
Micropterus dolomieu	smallmouth bass	7	2
Morone americana	white perch	104	62
Morone saxatilis	striped bass	0	1
Notemigonus crysoleucas	golden shiner	1	11
Notropis hudsonius	spottail shiner	9	19
Pomoxis nigromaculatus	black crappie	2	1
Strongylura marina	Atlantic needlefish	0	3
	Total	1099	2274

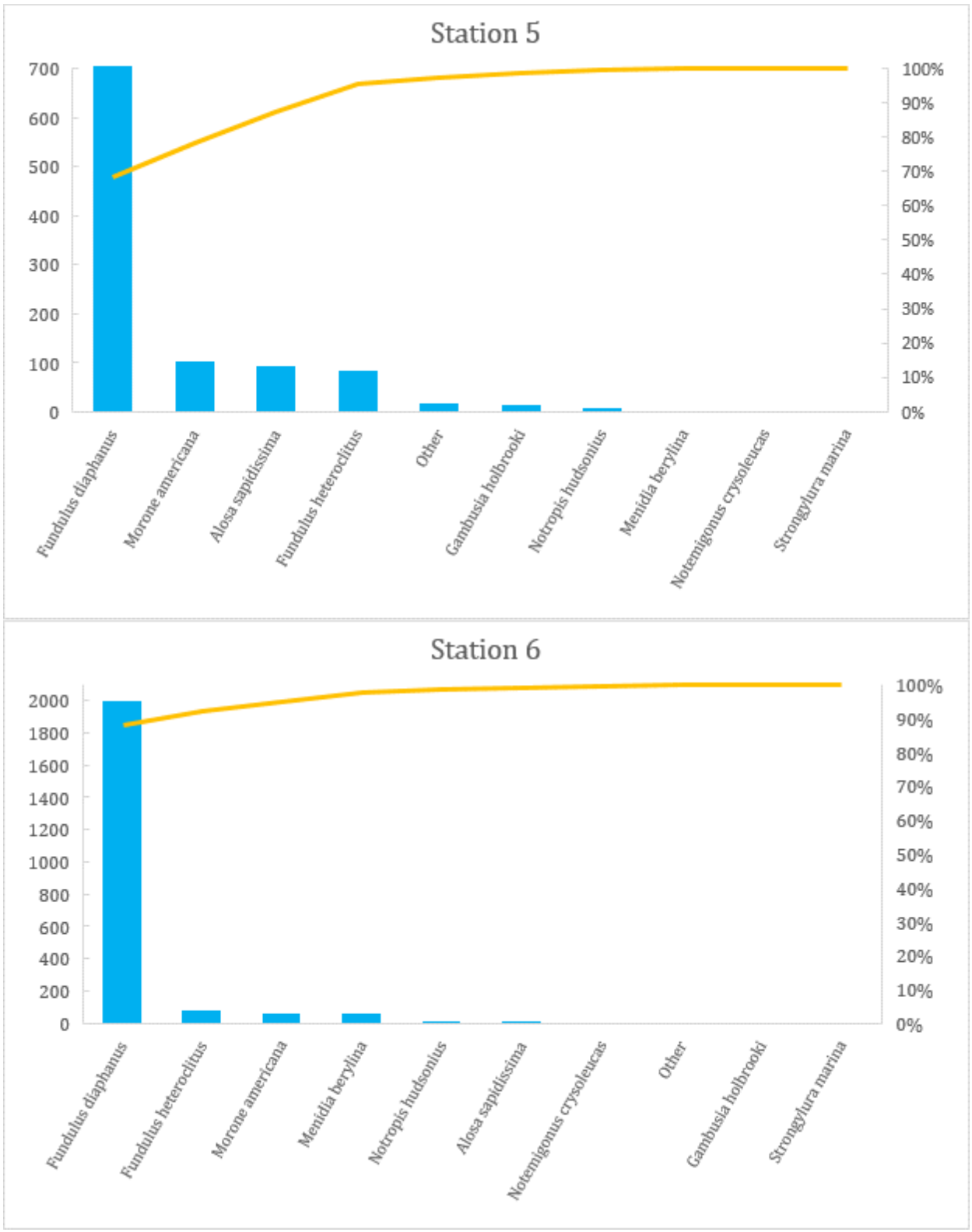


Figure 72 A and B. Pareto chart of adult and juvenile fishes collected by seining. Dominant species by station in total abundance and cumulative percentage of total for Station 5 (top) and Station 6 (bottom).

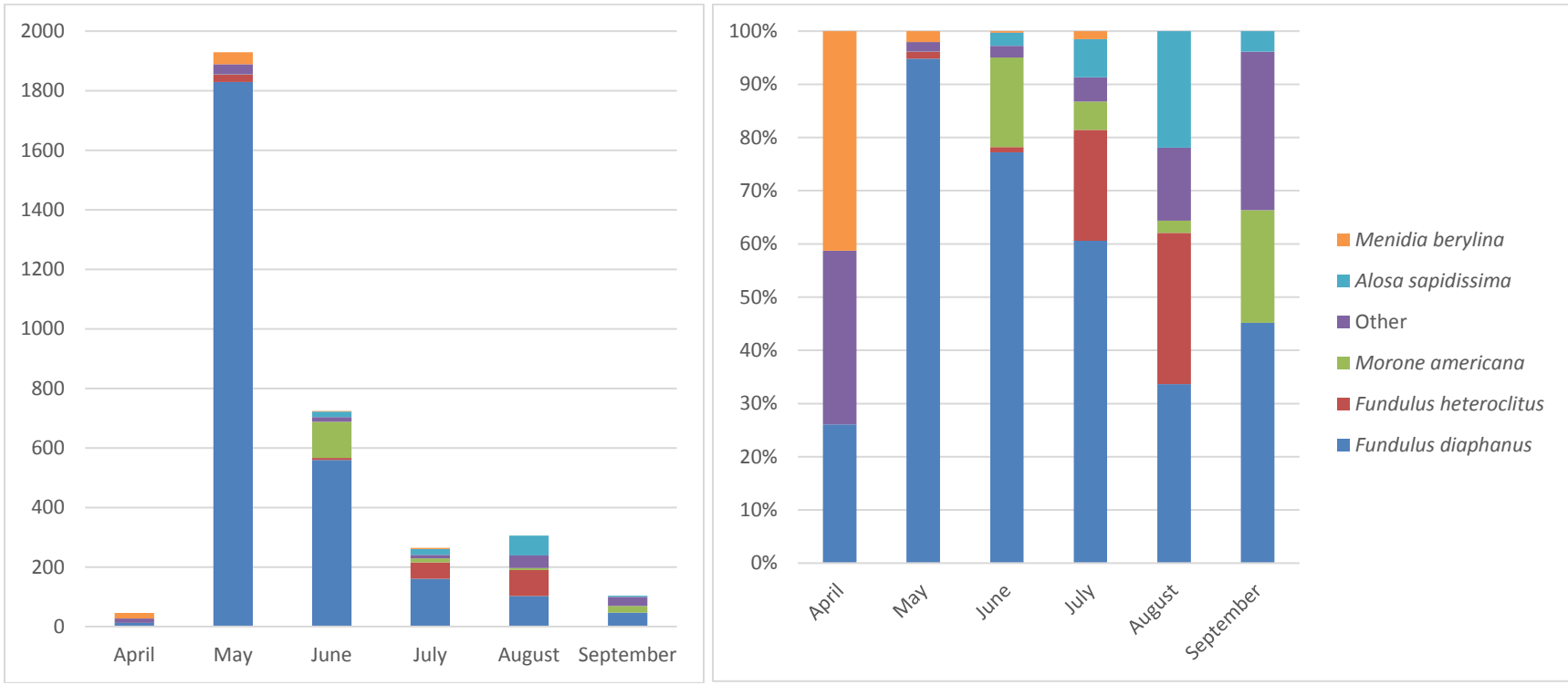


Figure 73A and B. Adult and juvenile fish collected by seining. Dominant species by month in percentage of total (A) and total abundance (B).

Banded killifish (*Fundulus diaphanus*) is a small fish, but the most abundant species in shoreline areas. Individuals become sexually mature at about 5 cm in length and may grow to over 8 cm long. Spawning occurs throughout the warmer months over vegetation and shells. They feed on benthic invertebrates, vegetation, and very small fishes.	White perch (*Morone americana*), which was discussed earlier in the trawl section, is also a common shoreline fish as juveniles collected in seines. The juveniles of white perch are attracted to the littoral zone as habitat where their predation risk is lower and potential food intake is higher.	Seining is conducted in shallow water adjacent to the shoreline. Some fish minimize predation by congregating along the shoreline rather than disperse through the open water. The high abundance of fish in seine tows, while seines sample a smaller volume of water than trawls, emphasizes the higher densities of fish along the shoreline.

F. Submersed Aquatic Vegetation – 2015

SAV data overflights by VIMS were conducted in 2015 and Figure 74 depicts the area covered by SAV that was detectable by aerial remote sensing. As can be seen from this map, the entire surface area of the Hunting Creek embayment was colonized by SAV. Some photographs are also shown below (Figure 75).

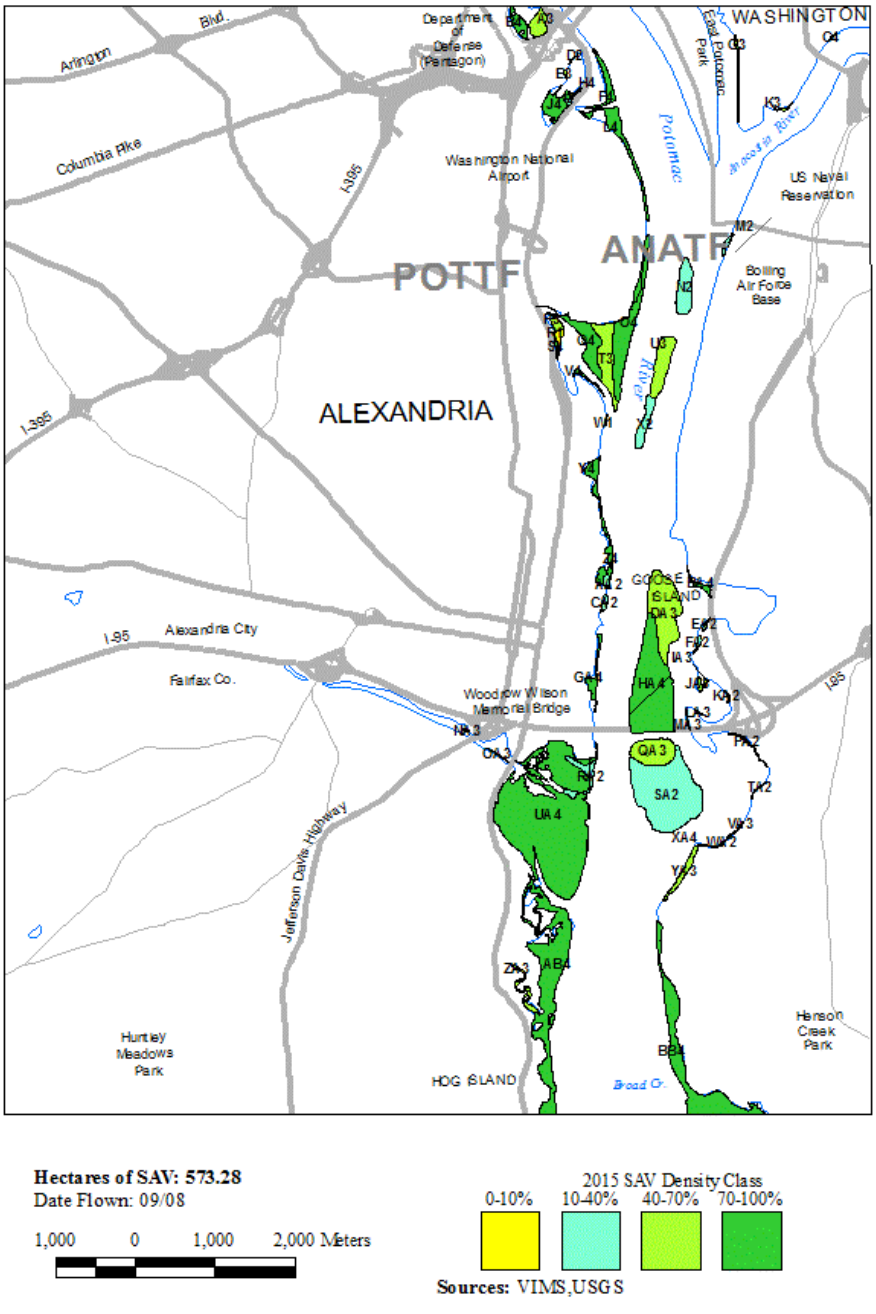


Figure 74. Distribution and density of Submersed Aquatic Vegetation (SAV) in the Hunting Creek area in 2013. VIMS (http://www.vims.edu/bio/sav/index.html).

Figure 75. Photos of SAV beds in Hunting Creek - 2015

H. Benthic Macroinvertebrates - 2015

Triplicate petite ponar samples were collected AR2, AR3, and AR4 monthly from May through September. Averages over samples collected at each station are shown in Figure 76. Oligochaetes were the most common invertebrates collected in these samples ranging from 118-276 per petite ponar (Figure 76a). Oligochaete densities were highest at Hunting Creek AR2 and AR3 and substantially lower in the river. Chironomid (midge) larvae made up a substantial portion of the remaining organisms at most stations ranging from 6 to 66 per petite ponar. Substantial numbers of other taxa were found at each station (Figure 76b). Gastropods were very abundant at embayment stations AR2 and AR3. Amphipods were the second most numerous of these "other" group, being found frequently at all three stations. Bivalves were found at lower densities at all three stations and leeches and isopods were present at some sites.

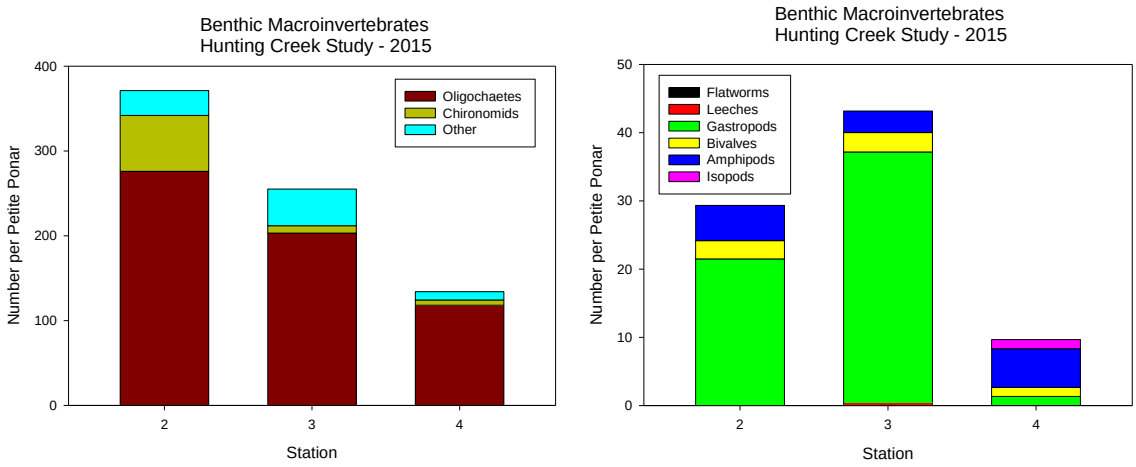


Figure 76. Average abundance of various benthic macroinvertebrate taxa in petite ponar samples collected in 2015. (a) dominant taxa. (b) "other" group from (a) broken out by taxa.

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