

Road salt deposition and retention in the riparian
zones: High impact in the High Peaks?



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ABSTRACT

Anthropogenic application of road salt (NaCl) negatively impacts natural ecosystems in the Adirondack Park (AP). Increased road salt application from climate change will exacerbate Na and Cl concentrations in surface waters and potentially degrade riparian soils. The purpose of this study was to assess the impact of road salt on riparian zone soils in the AP because riparian zones are vital to protecting AP waterways. We used soil samples from two comparable riparian zones: Johns Brook Tributary (JBT), which was not exposed to winter salting, and Cascade Brook (CB), which was exposed to winter salting. We collected soil cores from both sites three times during the salting season to measure fluctuations in Na and Cl concentrations. There was a significant difference in these concentrations between our sites, with CB having consistently higher concentrations of both. We also found spatial variance within our sites each month in relation to distance from the stream edge. To test the impact of Na on soil structure, we wet-sieved samples from each site to measure macroaggregate content. With our limited sample size, there was no statistically significant difference in macroaggregates between sites. We also investigated the influence of various Cl concentrations on soil microbial communities. We did not observe a clear relationship between Cl concentration and microbial biomass. Finally, we analyzed soil samples for available ammonium and phosphate to estimate the impact on available nutrients. We found significantly higher concentrations of both nutrients in JBT, although we cannot conclude road salt accounts for our observed differences. These results highlight the need for future studies, so we can understand road salt impacts more holistically.

INTRODUCTION

Since 1980, nearly 6.4 million metric tons of road salt have been applied to the Adirondack Park (AP) (AsRA, 2017). Each year, 98,000 metric tons of road deicers are applied to state roads in the AP from mid-October to mid-April (Laxson et al., 2016; Viggiani, 2018). Deicers are used during winter months to ensure safe driving conditions during winter storms, precipitation, and sub-freezing temperatures. As a result of climate change, areas in the AP will experience an increase in precipitation, an increase in more extreme weather events, and an increase in freeze/thaw events (Rosenzweig, 2011). Because of this, we predict there will be an increase in road salt application throughout the AP.

The road deicers used in the AP, specifically Essex County, primarily contain sodium (Na) and chloride (Cl). After application, these ions enter the surrounding ecosystem by direct deposition from plowing, wind deposition, or through runoff (Lundmark & Olofsson, 2007). This salt can persist in water and soils for decades, influencing AP ecosystems that naturally have low concentrations of Na and Cl ions (Kelting et al., 2012). In fact, the concentrations of Na and Cl in AP lakes is directly correlated to and dependent on road density in a lake's watershed (Kelting et al., 2012). The road salt likely impacts other aspects of the AP ecosystem, such as riparian zones.

Riparian zones are critical components of watersheds because they protect the AP's most important resource--its waterways. Riparian zones are the interface between aquatic and terrestrial systems in inland areas (González et al., 2017). They act as the transition zone of material, energy, and information between land and water (Fu et al., 2016). Riparian zones provide ecosystem services and fulfill many ecological roles, including habitat for regional biodiversity, climate regulation, flood buffering, water and nutrient filtering, and stream channel

shading (González et al., 2017). Riparian zones face many threats of degradation from increased precipitation, substantial flooding, and erosion. These threats exacerbate degradation along rivers by overturning banks, compromising aquatic habitat, and carving new channels for water flow. Since riparian zones are vital for protecting AP waterways, understanding any threats is essential for maintaining the overall health of the AP.

The goal of this study was to investigate the impact of road salt on riparian soils in the AP. To assess riparian zone health, we measured extractable soil Na and Cl concentrations, soil aggregates, and available ammonium and phosphate. We also analyzed the influence of Cl on microbial biomass. High concentrations of Na destroy aggregates, which are essential to riparian zone health (Rillig et al., 1999; Bronick et al., 2005). A healthy soil structure allows for percolation which aids in flood buffering, pore water retention, and nutrient filtration (Helliwell et al., 2014). Soil structure also provides a medium for the cation exchange. Additionally, high concentrations of Cl can decrease soil microbial populations, which may influence the availability of vital nutrients such as, ammonium and phosphate (Xiao et al., 2017; Bastviken et al., 2007, Bedard Haughn et al., 2003).

METHODS

Field collection sites

We collected soil from two comparable riparian zones in Essex County, NY: a tributary to Johns Brook (JBT) and Cascade Brook (CB). JBT was isolated from roads, and therefore, we expected it to not be exposed to road salt. CB was adjacent to route 73 and was exposed to road salt (Figure 1). These sites were both historically classified as the same “Hermon gravelly loamy sand” in the

USDA SSURGO Soils database. Both sites have southern aspects and are characterized by a temperate climate.

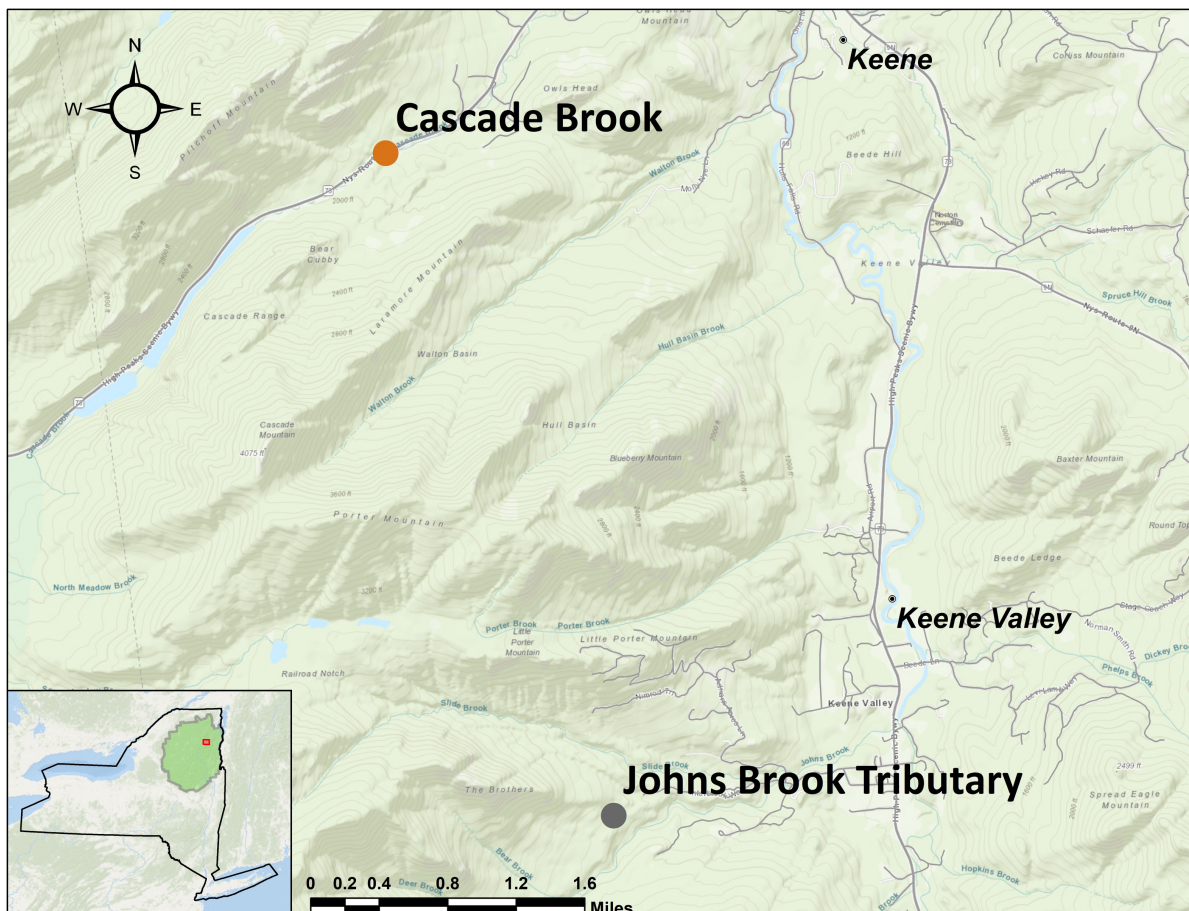


Figure 1: Map of our two field collection sites. Our sites are in the AP near Keene Valley, 6 kilometers apart from each other.

Field methods

We collected soil samples three times throughout the year: November 2017 (the start of the salting season), January 2018 (the middle of the salting season), and April 2018 (the end of the salting season). Within each site, we collected samples along three parallel transects 5 meters apart, and perpendicular to the stream. We collected three composite soil samples at distances

1m, 3m, 6m, 8m, and 10m from the stream along each transect (Figure 2). We collected soil cores using an 8 cm wide, 15 cm deep pvc core. Before further analyses, we sieved the soil samples through a 2 mm mesh sieve and refrigerated them.



Figure 2: Example of transect methodology in relation to stream bank and route 73 at CB site (Not to scale).

Extractable Na and Cl methods

We added 25.0 mL of 0.01 M CaNO_3 to 10.0 g of soil. We left the samples at room temperature for 48 hours. We resuspended the samples and left them at room temperature for an additional hour, and then filtered them. Samples were diluted to fit a standard curve of 0-5 ppm. We measured the Na concentration using an Atomic Absorption Spectrometer, and Cl

concentrations using an Ion Chromatograph. We ran two-way ANOVA tests on logarithmically transformed data to compare Na and Cl by site and season.

Aggregate distribution methods

We determined the aggregate distribution between the two riparian zones using 30 g of unsieved air-dried soil samples collected from our sites in November of 2017. We wet-sieved the samples in water through a series of 2000- μm , 250- μm , and 53- μm sieves (Grandy & Robertson, 2006). Material remaining on the 2000 μm sieve was treated as excess material. We dispersed the dried subsamples from the 250 μm sieve in sodium hexametaphosphate solution (20 g/L) and shook them for 48 hours on a rotary shaker at 190 rpm to separate mineral sand. We sieved the remaining sediment to determine sand content, which we used to correct for actual macroaggregate content.

Microbial community experiment

We tested the impact of road salt on microbial communities in a controlled lab experiment. We placed composite soil samples from JBT in closed Ball jars at room temperature. We used soils randomly taken from JBT in order to control for any alterations predicted higher Cl concentrations might have had on microbial communities in CB. The samples were exposed once to Cl concentrations of 0ppm, 20ppm, 40ppm, 80ppm, and 100ppm. NaCl was dissolved in DI water prior to the application to simulate the input of road salt from runoff. Higher concentrations were meant to mimic conditions closer to roadways. The Cl concentrations we used were based off of observed Cl concentrations in our CB November samples. We took soil

subsamples from each jar two, three, and four weeks after Cl exposure to estimate microbial biomass.

Microbial biomass method

We used the Substrate Induced Respiration (SIR) method to estimate microbial biomass in the subsamples from the microbial community experiment. Similar to Fierer (2003), we took 10 g of soil (fresh weight) and put it into 50 mL centrifuge tubes equipped with gas-tight lids that had rubber septa for gas sampling. We prepared yeast solution by adding 3g autolyzed yeast extract to 250 mL DI water. We added 7 mL of this yeast solution to each tube. We shook the tubes horizontally at 190 rpm at room temperature for 4 hours. We measured the headspace CO₂ concentrations 20 minutes after we added the yeast solution (T0 time point), and again 2 hours and 4 hours after the T0 time point. We measured the headspace CO₂ concentrations by taking a 1 mL air sample from each tube and measuring the cumulative CO₂ concentrations on an Infrared Gas Analyzer (IRGA) equipped for static CO₂ analysis. We calculated the slope of the line relating CO₂ concentrations to time. The average respiration rate over the 4-hour incubation period was an index of the SIR-responsive microbial biomass, which we used to see relative differences in our treatment soils.

Nutrient analysis methods

We analyzed plant available ammonium and phosphate by extracting them from soils with KCl. We distributed 20 g of moist soil and 50 mL of 1M KCl into each extraction cup. After every 20 samples, we inserted a reagent blank of 50 mL of KCl. We shook each sample for one minute and

let the samples sit at room temperature overnight. We resuspended the samples the next day and filtered the extractions an hour later. We analyzed these samples using a Biotek Plate reader. Both ammonium and phosphate were compared by site and season by performing a two-way ANOVA test on logarithmically transformed data.

RESULTS

Extractable sodium and chloride

Na concentrations varied significantly between sites (Figure 3). JBT soil Na concentrations ranged from 29.758 mg/kg soil to 32.742 mg/kg soil in November (Figure 4). Concentrations peaked in January, ranging from 59.353 mg/kg soil to 129.695 mg/kg soil (Figure 5). They then dropped in April between 33.411 mg/kg soil to 84.537 mg/kg soil (Figure 6). Na concentrations were significantly greater in Cascade Brook compared to Johns Brook Tributary. Na concentrations in Cascade Brook in November ranged from 71.479 mg/kg soil to 542.369 mg/kg soil (Figure 4). They also peaked in January, ranging from 151.212 mg/kg soil to 1023.935 mg/kg soil (Figure 5). Overall, Na concentrations dropped in April between 132.796 mg/kg soil to 1783.175 mg/kg soil (Figure 6).

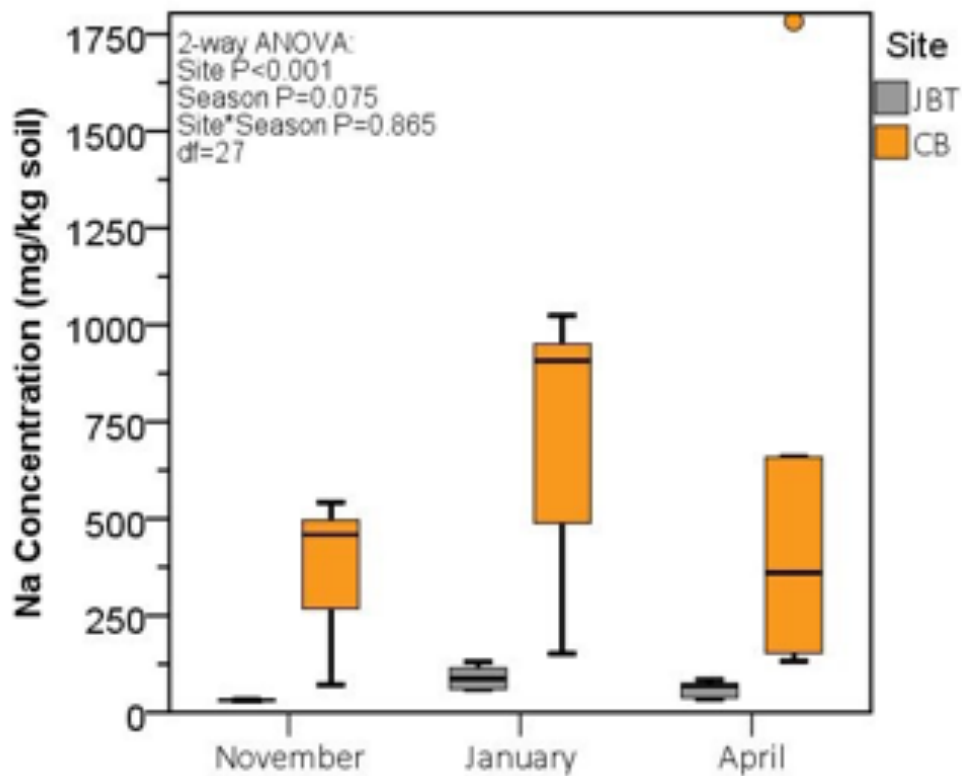


Figure 3: The average concentration of extractable Na found in both study sites. The average concentration was calculated from soil samples taken at 1m, 3m, 6m, 8m, and 10m from the stream along the sampling transect. A two-way ANOVA test showed a significant difference between sites ($p < 0.001$, $n = 28$).

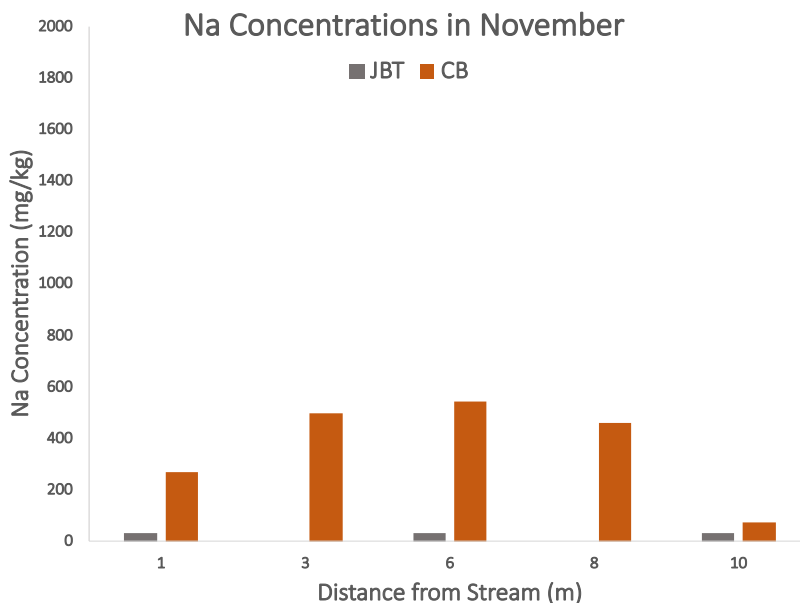


Figure 4: Extractable Na concentrations of the November soil samples collected along field sampling transects at Johns Brook Tributary and Cascade Brook. The concentrations at Johns Brook Tributary and Cascade Brook were found using a formula derived from the standard curve: $y = 3.8759x - 0.0507$ ($R^2 = 0.9994$). The Na concentrations displayed are averages of two subsamples and are corrected for soil moisture. Note that 3m and 8m soil samples were not collected in November at JBT.

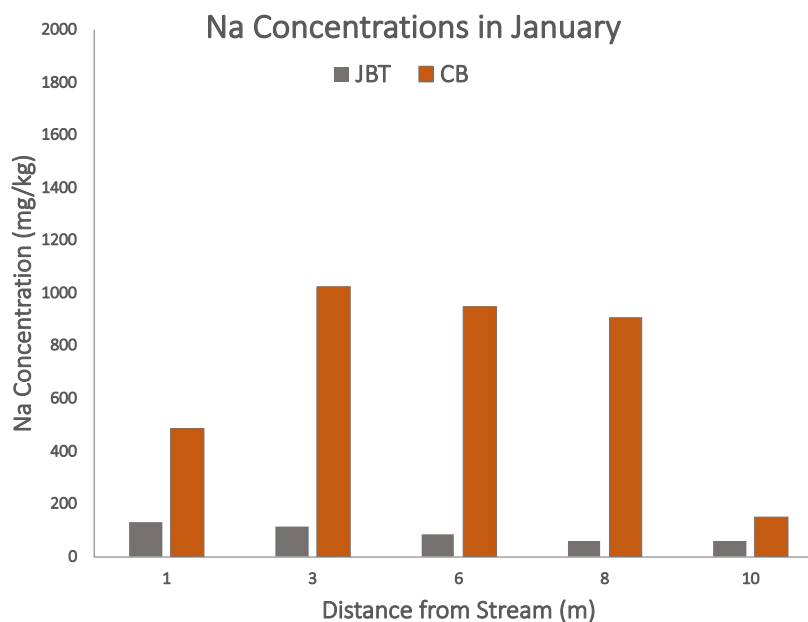


Figure 5: Extractable Na concentrations of the January soil samples collected along field sampling transects at Johns Brook Tributary and Cascade Brook. The concentrations at Johns Brook Tributary and Cascade Brook were found using a formula derived from the standard curve: $y = 3.8759x - 0.0507$ ($R^2 = 0.9994$). The Na concentrations displayed are averages of two subsamples and are corrected for soil moisture.

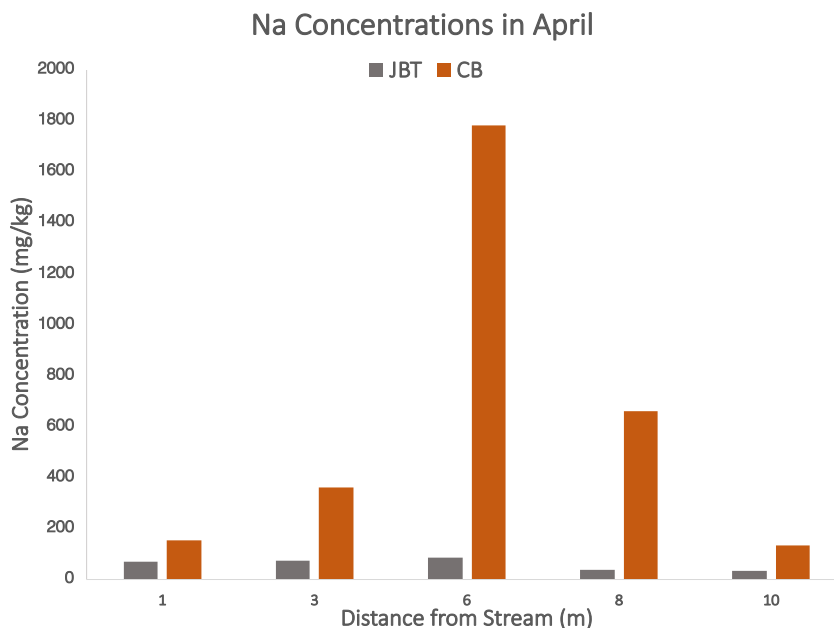


Figure 6: Extractable Na concentrations of the April soil samples collected along field sampling transects at Johns Brook Tributary and Cascade Brook. The concentrations at Johns Brook Tributary and Cascade Brook were found using a formula derived from the standard curve: $y = 3.8759x - 0.0507$ ($R^2 = 0.9994$). The Na concentrations displayed are averages of two subsamples and are corrected for soil moisture.

Extractable Cl concentrations varied significantly between our sites (Figure 7). At JBT, Cl concentrations ranged from 16.838 mg/kg soil to 26.295 mg/kg soil in November (Figure 8). Then Cl concentrations peaked in January, ranging from 41.684 mg/kg soil to 113.867 mg/kg soil (Figure 9). Cl concentrations were significantly greater in CB than those in JBT. Cl concentrations in CB in November ranged from 11.391 mg/kg soil to 505.097 mg/kg soil (Figure 8), then they peaked in January, ranging from 57.923 mg/kg soil to 1126.300 mg/kg soil (Figure 9). Cl concentrations in our April samples had contamination issues and were not included in these results (Appendix 1).

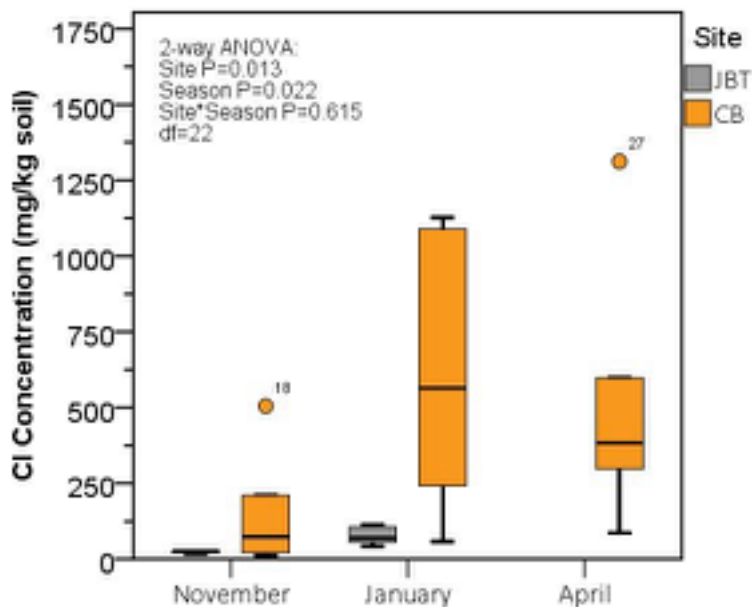


Figure 7: The average concentration of extractable Cl found in both study sites. The average concentration was calculated from soil samples taken at 1m, 3m, 6m, 8m, and 10m from the stream along the sampling transect. A two-way ANOVA test showed a significant difference between sites ($p < 0.013$, $n = 23$) and season ($p < 0.022$, $n = 23$), result do not include JBT April data, but we included CB April data for context.

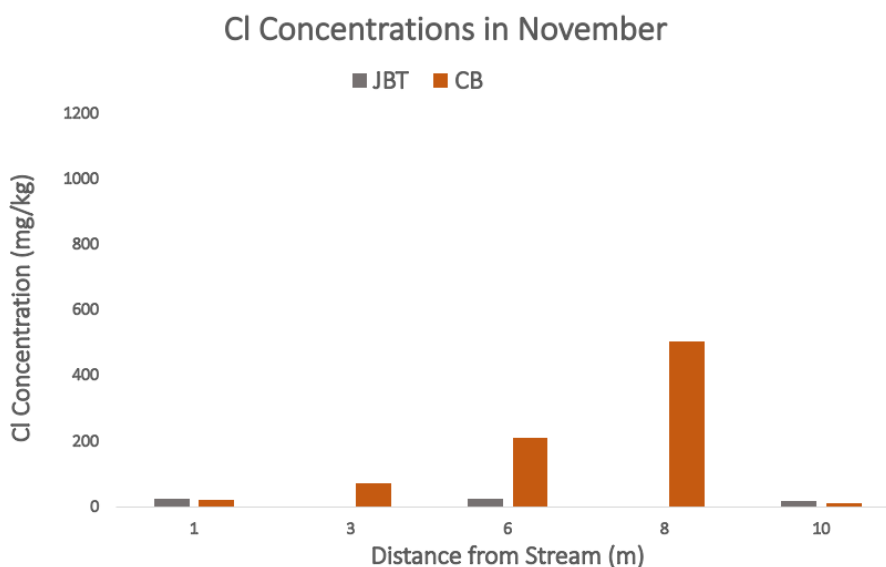


Figure 8: Extractable Cl concentrations of the soil samples collected in November along the field sampling transects at the Johns Brook Tributary and Cascade Brook sites. The concentrations at Johns Brook Tributary and Cascade Brook were found using a formula derived from the standard curve: $y = 3.2149x + 0.0383$ ($R^2 = 0.9994$). The Cl concentrations displayed are averages of two subsamples and are corrected for soil moisture. Note that 3m and 8m soil samples were not collected in November at JBT.

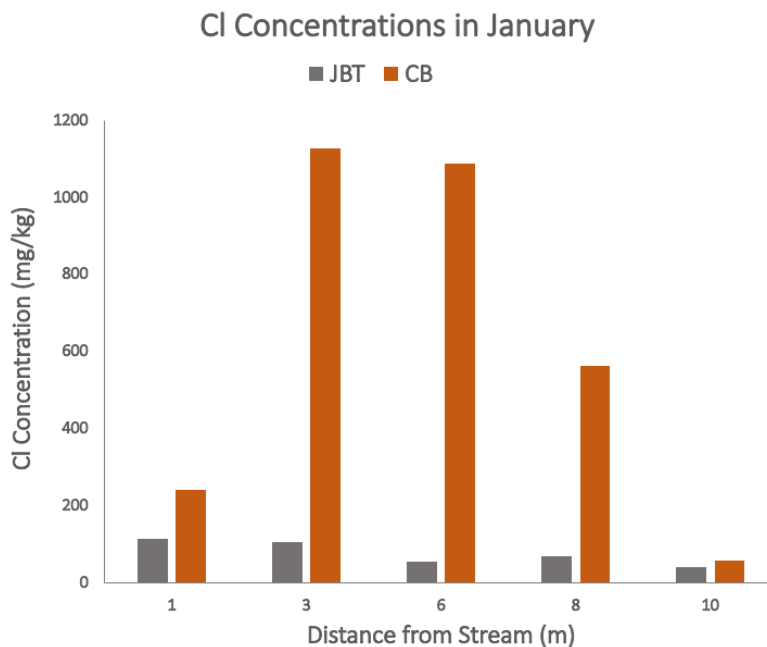


Figure 9: Extractable Cl concentrations of the soil samples collected in January along the field sampling transects at the Johns Brook Tributary and Cascade Brook sites. The concentrations at Johns Brook Tributary and Cascade Brook were found using a formula derived from the standard curve: $y = 3.2149x + 0.0383$ ($R^2 = 0.9994$). The Cl concentrations displayed are averages of two subsamples and are corrected for soil moisture.

Aggregate distribution

We calculated the aggregates at each sieve size (250 μ m, 53 μ m, and <53 μ m) using percent by mass. 5.65% to 15.68% of soil samples from JBT and 3.15% to 7.70% of samples from CB remained on the 250 μ m sieve. 3.56% to 14.21% of soil samples from JBT and 6.69% to 24.17% of samples from CB remained on the 53 μ m sieve. 12.18% to 15.47% of soil samples from JBT and 6.98% to 21.06% of samples from CB were calculated to be the remaining particles <53 μ m (Figure 10).

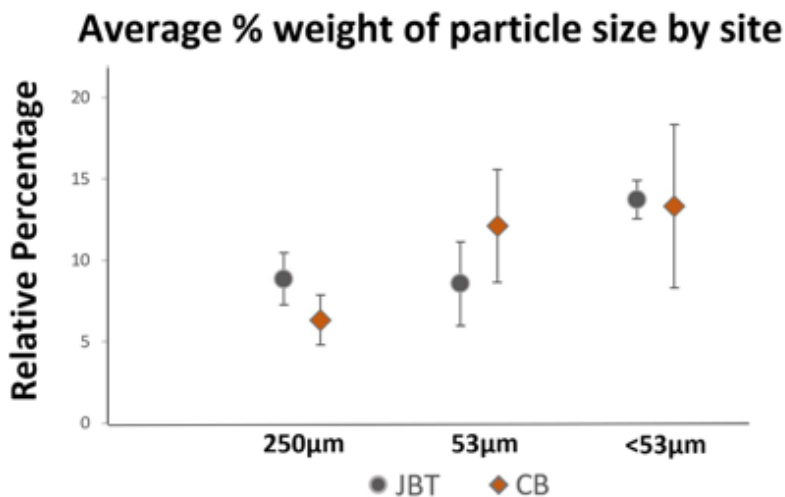


Figure 10: Aggregate distribution by clast size/relative percentage of aggregates by dry weight. A two-way ANOVA test determined there was no significant difference between either site for any of the aggregate sizes tested ($p > 0.05$, $n = 8$).

Microbial biomass

The average rate of CO_2 production remained fairly consistent over time across various Cl concentrations (Figure 11). The average respiration rate over the 4-hour incubation period, measured 2, 3, and 4 weeks after initial exposure to NaCl, is an index of the SIR-responsive microbial biomass (Fierer, 2003). We calculated the slope of each soil sample over the four-week incubation (First week 2 to week 3, then week 3 to week 4) to determine respiration change over time. The overall CO_2 produced ranged from $0.3249\mu\text{g C-CO}_2$ to $0.68\mu\text{g C-CO}_2$. All soil samples had the highest rate of CO_2 production in week two, with the dry soil having the highest rate at $0.68\mu\text{g C-CO}_2$ and 80ppm soil the lowest at $0.5286\mu\text{g C-CO}_2$. Three weeks after exposure to the NaCl, CO_2 production ranged from $0.3249\mu\text{g C-CO}_2$ to $0.5463\mu\text{g C-CO}_2$. Finally, four weeks after exposure, the production of CO_2 had a similar range as the other weeks: $0.3491\mu\text{g C-CO}_2$ to

0.5159 $\mu\text{g C-CO}_2$. In sum, all soil samples had the greatest production of CO_2 two weeks after exposure to Cl^- , and fairly consistent trends for the duration of our study.

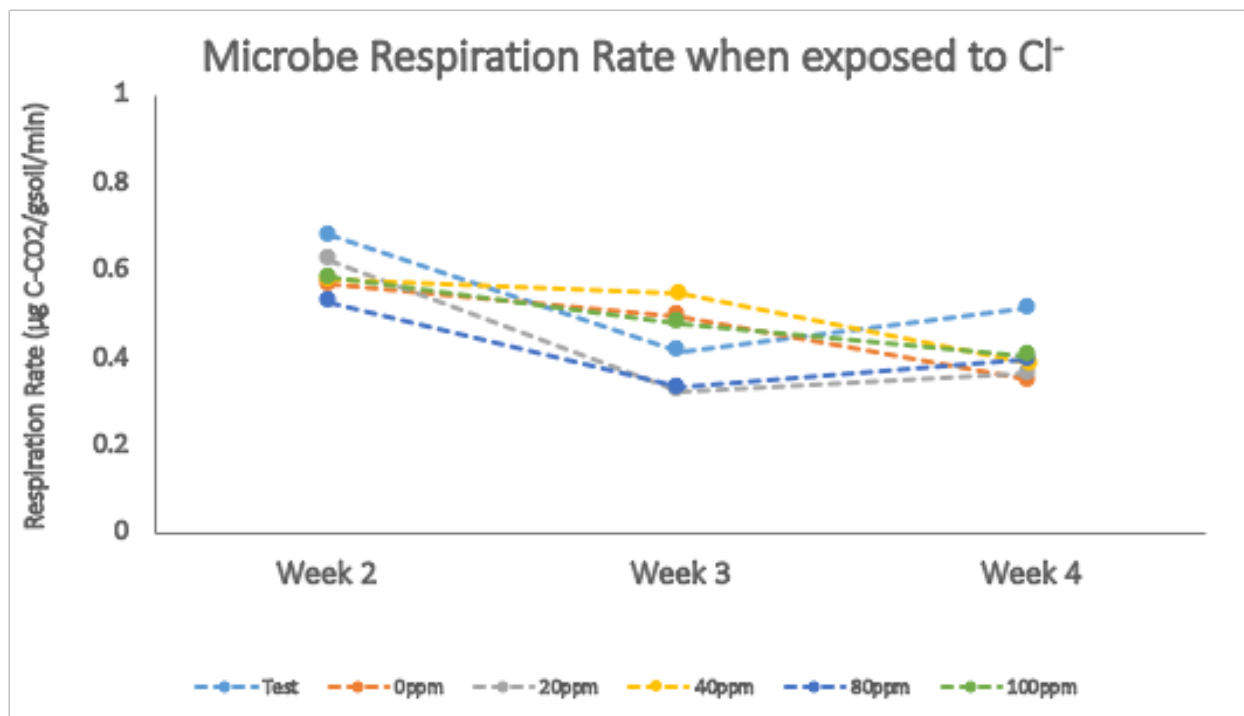


Figure 11: The rate of $\mu\text{g CO}_2$ produced during the 4-hour incubation period for each Cl^- concentrations. This was measured 2, 3, and 4 weeks after exposure to Cl^- . The test soil had no liquid added to it.

Nutrient analyses

Composite soil samples from JBT and CB showed a significant difference in available ammonium.

JBT had significantly higher concentrations than CB in November, January, and April (Figure 12).

Available ammonium in JBT ranged from 10.28 to 57.15 mg of available ammonium per kg soil

with the highest concentration in the January samples, and the lowest concentration in the

November samples. Contrary to this trend, the available ammonium in CB was fairly consistent,

ranging from 4.36 to 8.012 mg of available ammonium per kg soil. The highest concentration in

the CB samples was in November, and the lowest was in April.

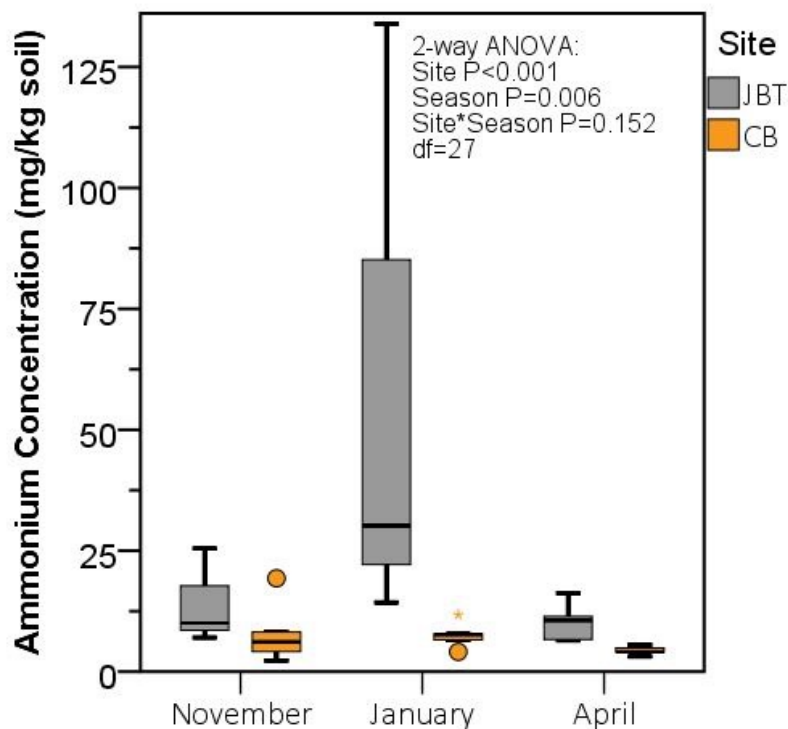


Figure 12: The concentration of available ammonium found in both study sites. The concentration is an average concentration calculated from soil samples taken at 1m, 3m, 6m, 8m, and 10m from the stream along the sampling transect. The values are corrected for soil moisture and were found using the following standard curves: $y = 2.5177x - 0.1807$ ($R^2 = 0.9996$) for the November and January samples, and $y = 2.6109x - 0.2057$ ($R^2 = 0.9994$) for the April samples. A two-way ANOVA test showed a significant difference between site ($p < 0.001$, $n = 28$) and season ($p = 0.006$, $n = 28$).

Similar to ammonium, phosphate was significantly higher in JBT throughout our sampling period (Figure 13). Concentrations at JBT ranged from 0.83 to 8.13 mg of available phosphate per kg soil, with concentrations increasing from November to April. Concentrations in CB remained consistent compared to JBT. Phosphate concentrations ranged from 0.31 to 1.00 mg of available phosphate per kg soil, and these samples increased from November to April.

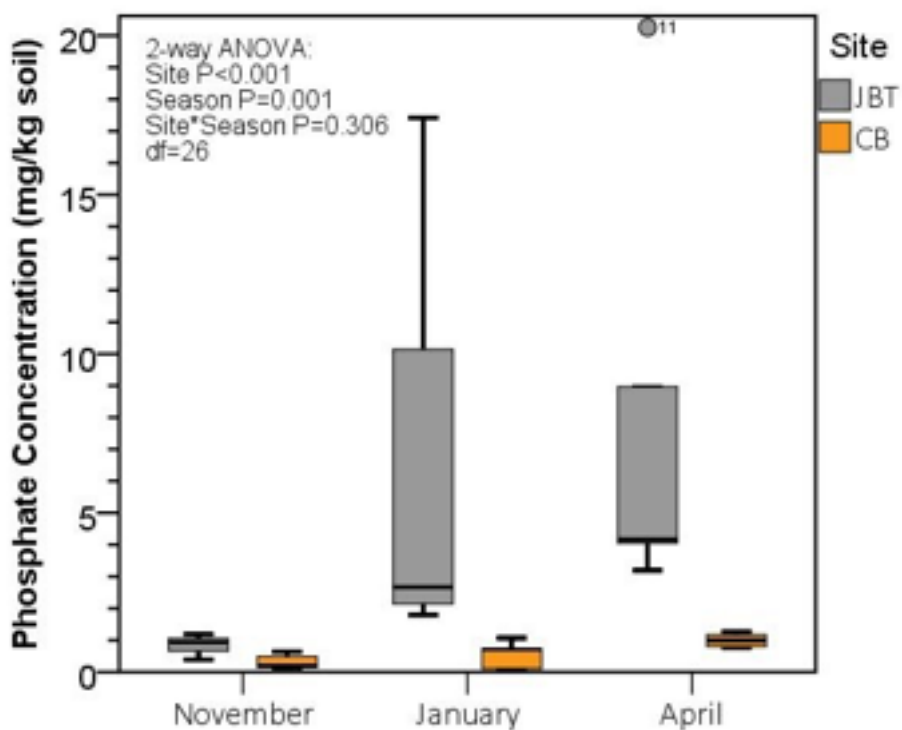


Figure 13: The concentration of available phosphate found in both study sites. The concentration is an average concentration calculated from soil samples taken at 1m, 3m, 6m, 8m, and 10m from the stream along the sampling transect. The values are corrected soil moisture and were found using the following standard curves: $y = 0.7501x - 0.0217$ ($R^2 = 0.9984$). A two-way ANOVA test showed a significant difference between site ($p < 0.001$, $n = 27$) and season ($p = 0.001$, $n = 27$).

DISCUSSION

Extractable Sodium

From November to January we saw an increase in Na concentrations at both JBT and CB. This coincides with the continued application and deposition of NaCl on roads as the winter season progressed. Na concentrations at CB increased as a result of its proximity to route 73. We believe that wind deposition contributed to the Na concentration increase at the JBT site. Na concentrations declined at JBT in April because there was likely less NaCl application this late in the salting season. At CB, we saw a decrease in Na concentrations at all transects except 6m.

Beginning in April, melting and rain events increased the amount of runoff, which pooled at the CB 6m location, as a result of the site's slope. Na concentrations decreased below the 6m transect potentially as a result of stream flooding.

Extractable Chloride

Similar to the patterns we observed in Na, Cl concentrations increased at both sites from November to January and CB had higher concentrations of Cl. This increase in Cl can be attributed to the increased application and deposition of road salt. Due to a contamination error, we were unable to obtain Cl concentrations for April, however, we expect that it would have followed a similar topographic trend as Na concentrations in April.

Aggregate distribution

We confirmed the assumptions in the USDA SSURGO Soils database. These soils were comparable based on structural characteristics. There was not a significant difference between the sites in relative aggregate mass by clast size. These masses were corrected for mineral sand-sized particles at the 250 μ m level, which Cascade Brook had more of. Although our sample size was too small to make definitive conclusions, we did begin to see differences in the percentage of macroaggregates between sites, with JBT having more than CB. We expected this trend because CB is exposed to larger amounts of Na. Therefore, we predict that future studies with a greater sample size may see a significant difference between the two sites.

Microbial biomass

Soil microbial respiration remained fairly consistent after one exposure to various concentrations of Cl. This observation suggests that high chlorine environments are able to support consistent microbial activity shortly after Cl inundation. Other studies have found that native microbial communities are altered in the presence of Cl, however, we did not see a difference in respiration rates (Gryndler et al., 2007). This lack of change indicates there may be microbial uptake by AP microbes, which is one mechanism for Cl retention after application (Robinson et al., 2017; Bastviken et al., 2007). Microbial retention of Cl usually delays the release of the Cl to the ecosystem by about one month, which can help explain fluctuations of surface water concentrations, and the consistently higher concentrations throughout the year in AP lakes (Robinson et al., 2017; Kelting et al., 2012). Although we did not quantify the diversity or type of microbial communities present before and after Cl exposure, our study suggests that native soil microbial communities were able to tolerate high Cl concentrations of at least 100 ppm, or that alterations in microbial communities occur extremely rapidly after inundation and did not ultimately change the total respiration rate. Conversely, Cl induced alterations may not take place after one exposure in a short amount of time, indicating a need to replicate this experiment over a longer time period.

Microbial retention of Cl could potentially have impacts on other ecosystem services and overall productivity because microbes are regulators of plant productivity. There is evidence that Cl can eventually lead to a decline in microbial communities (Bastviken et al., 2007). Microbes are responsible for mineralizing soil nitrogen (N) from its organic form to its inorganic form. This inorganic form is what plants use and is often a limiting nutrient. Since about 95% of soil N is in

the organic form, a decline in microbial productivity and biomass could limit plant growth (Bedard Haughn et al., 2003). If this were to happen, vital riparian zone ecosystem services, such as erosion prevention, water filtration, water temperature control, and other services would be adversely impacted.

Future studies would ideally replicate our experiment but expose soil microbes to higher concentrations of Cl more frequently in order to replicate roadside conditions. Since the concentrations we used were based off of observed Cl concentrations from our CB November soil samples, we underestimated the concentrations microbes would have to face in natural ecosystems during heavier salting periods, such as January and February. Additionally, consistent application of dissolved NaCl should be applied to the soil samples. Although our results show microbes ability to maintain their productivity after an increase in Cl concentration, our study did not investigate whether or not productivity was maintained after multiple NaCl inputs at the same concentrations, which is the mechanism most natural ecosystems would have to tolerate. Finally, future studies should see how native microbial communities respond to increased Cl at different temperatures. Our study was completed at a constant temperature, 23°C. However, this temperature is well above temperatures road salt would be applied in natural ecosystems (typically temperatures below 0°C), and is unrealistic because winter temperatures fluctuate rapidly, at times within a day or less. This variable may be increasingly important to investigate. Sorensen et al. (2018) found that changes in winter temperatures, and an increase in winter freeze thaw cycles, has led to a decrease in microbial biomass that is not compensated for by overall warming of soils. This has led to changes in C and N cycling (Sorensen et al., 2018), and could possibly impact Cl retention as well.

These findings, although limited and preliminary, demonstrate a need to further investigate microbial Cl retention in the AP. Understanding this process will help with riparian zone restoration in the AP, which has been more necessary as devastating precipitation events, such as Hurricane Irene, have become more prevalent as a result of climate change (Rosenzweig, 2011). If native AP microbes can tolerate increased Cl, selecting vegetation that can as well will be vital for riparian zone restoration. If further studies conclude that microbial biomass and productivity is lost, it will be important to ensure plants added to restored riparian zones can survive in low N environments.

Nutrients

JBT had significantly higher concentrations of both available ammonium and phosphate throughout our sampling period. Although these concentrations varied over time at JBT, we attribute this fluctuation to changes in the temperature. Lower temperatures in November and January resulted in decreased nitrification, which allowed ammonium concentrations to accumulate. Once temperatures increased in April, nitrification resumed and ammonium concentrations decreased. We did not measure other soils properties, such as organic matter, that would have allowed us to explain these changes in detail. These results indicate that the riparian zone at JBT may be healthier than the riparian zone at CB. We consider JBT to be healthier because both of these nutrients are necessary for plant growth, which provides many ecosystem services in riparian zones. Both of these findings were expected but leave questions as to whether road salt is the cause of these changes.

Formation of ammonium is positively related to microbial biomass (Lamb et al., 2014). Since Cl is known to be used by microbes, and excess inputs can decrease microbial communities, we expected ammonium to be higher in JBT due to reductions in microbial biomass in CB (Bastviken et al., 2007; Robinson et al., 2017). Further, since ammonium has a positive charge, we expected it would not be retained by the CB soils since these soils were inundated with other cations, mainly Na and Mg, during the salting season. These other cations would knock off and replace ammonium on the soil cation exchange, resulting in consistently low amounts during the salting season in CB compared to JBT. This trend was observed, but the fact that our microbial experiment did not show a correlation between Cl concentration and microbial respiration indicates that the low amount of ammonium in CB is possibly due to salt cation deposition. Since ammonium remained consistently low from the start of our sampling time, we cannot conclude whether ammonium was present after the summer season. If ammonium was never there, perhaps due to limited microbial biomass, cation salt deposition could not be responsible for the low concentrations in CB because the salt cations could not have removed what was not there.

Other factors also influence the amount of ammonium found in soils, such as the soil type and the amount of organic matter. CB soils were noticeably sandier than soils at JBT, perhaps suggesting a lower cation exchange capacity (CEC). A lower CEC would limit the amount of cations, such as ammonium, the soils in CB would retain. This would suggest that the lower concentrations of ammonium found at CB are controlled by soil particle content, not salting. Additionally, soil organic matter has a CEC and can retain cations, just as negatively charged soil particles can. If CB had a significantly lower amount of organic matter in its soils, that could also

explain why ammonium was lower there, and perhaps confirm that road salt is not the main driver of this observed difference.

Future studies should aim to address these limitations by quantifying the CEC and soil organic matter in the two riparian zones. Obtaining this information would allow for more definitive conclusions about the impact road salt has on ammonium concentrations in the AP. Additionally, studies should continue to monitor ammonium concentrations throughout the year, and over many years, to see how comparable the concentrations are outside of the salting season. This will help us understand nutrient cycling at these two sites and whether it is possible to assume the salt cation deposition controls ammonium availability. This information will also allow us to understand if the concentration is different during the growing season, when nutrients matter the most for plant health.

Similarly, phosphate displayed the predicted results, but we were unable to conclude that road salt caused the observed differences. We expected road salt to influence phosphate by impacting microbes. Microbes play an integral part in making soil phosphorous available to plants primarily through mineralization and solubilization (Richardson & Simpson, 2011). Additionally, plant interactions and their relationships with microbes play a large role in regulating the available phosphorus in an area (Richardson & Simpson, 2011). Therefore, we expected that the Cl impact on microbes and the differences in vegetation between the two sites, would lead to higher phosphate concentrations in JBT. While we expected this, we did not quantitatively assess the difference in vegetation, which such be looked at further in other studies. As was the case with ammonium, our expected trend was observed, but conclusions were limited because of our microbial experiment results. We would need to measure microbial

biomass in our sites or observe a negative relationship between Cl and microbial biomass in another experiment to attribute differences in phosphate to road salt.

Other factors impact available phosphorus in soils, which further complicates our conclusions. Soil pH is a large driver of available phosphorus because it controls conditions in which phosphorus is converted to a useful, inorganic form. Additionally, the main way phosphate is lost from soils is through erosion. Therefore, since CB is closer to paved roads, it will likely have greater levels of erosion due to runoff from paved areas. These variables alone could explain the observed differences in phosphate between our two sites.

To better understand road salts' impact on available phosphate, future studies should address the following issues. First, studies should identify the vegetation cover in each riparian zone. This will allow us to understand the phosphate concentration through relationships between plants and microbes. For example, different mycorrhizal associations are known to impact available phosphate (Richardson & Simpson, 2011; Phillips et al., 2013). Further, studies should examine soil erosion at the two sites to quantify the amount of soil removed at each site. Next, soil pH and cations should be measured, since they are the main factors influencing phosphate availability. Soil pH is a measurement of Hydrogen ions on the cation exchange. Na from road salt could potentially replace Hydrogen ions on the exchange, and lead to a higher pH at CB. Although this could happen, we observed more sand at CB. Sand has a low CEC, meaning the soil there is limited in the amount Hydrogen ions it can hold. This might suggest that the pH in CB would be higher due to its inability to hold a large amount of Hydrogen ions. Alternatively, this could suggest that the additional Na plays even more of a role in increasing the pH at CB. While we are unsure if this would significantly change pH, it would help explain the relationship

between road salt, pH, and available phosphate. If the pH were higher in CB, it would result in lower phosphorus fixation (USDA, 2013). Finally, phosphate concentrations should be measured throughout the year and compared outside of the salting season. This information will allow us to understand long term soil impacts and will help us see if these nutrients are different during the growing season, when they matter the most. There is still a gap in knowledge regarding how road salt impacts riparian zones as indicated by our research.

CONCLUSION

Na and Cl both entered the ecosystem through road salt, followed patterns of topography, and were found outside of their deposition area. Na and Cl were found 25 m away from the road at CB, and over 25 m from the nearest road in JBT. While we did not have a big enough sample size to confirm aggregate distribution was impacted by Na, we did begin to see a difference. We believe a bigger sample size over a longer period of time would reveal a significant difference between the percentage of macroaggregates at both sites. Additionally, we believe we did not see significant differences in our microbial lab experiment because we only applied Cl to our soils once. With more frequent application and higher concentrations of Cl, we believe future studies may observe a difference in microbial biomass. Finally, while we saw a significant difference in ammonium and phosphate concentrations between sites and seasons, we cannot definitively contribute this to road salt. In the future, we need to investigate other ecosystem components, such as those previously discussed, in order to understand the impacts of road salt on AP ecosystems holistically.

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APPENDIX 1

Cl April data

When analyzing our Cl data for the sampling period in April, the JBT soils had a spike in Cl much larger than anything we had seen in any of the CB samples (Figure 14). Contamination seems most likely because subsamples were extremely inconsistent after multiple tests with the IC.

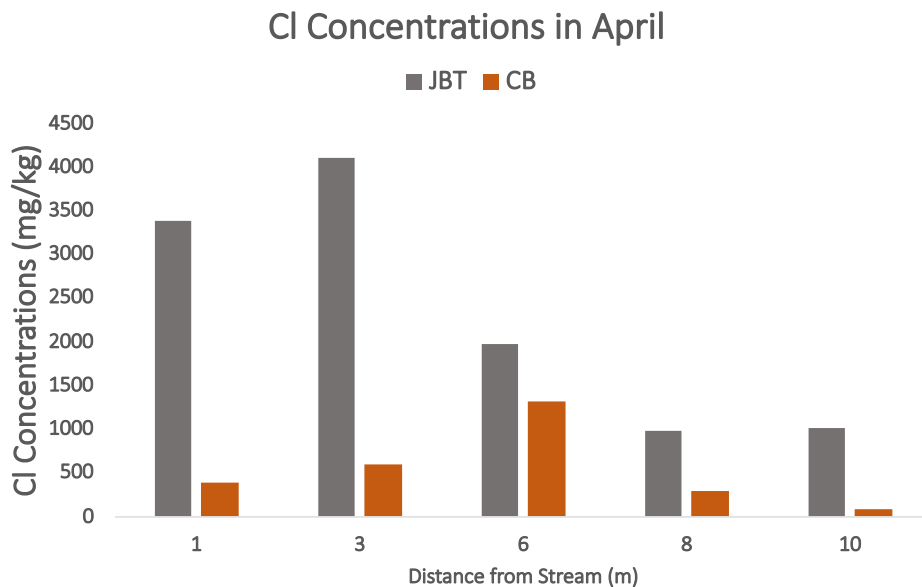


Figure 14: Extractable Cl concentrations of the soil samples collected in April along the field sampling transects at the Johns Brook Tributary and Cascade Brook sites. The concentrations at Johns Brook Tributary and Cascade Brook were found using a formula derived from the standard curve: $y = 1.9664x - 0.363$ ($R^2 = 0.9996$). The Cl concentrations displayed are averages of two subsamples and are corrected for soil moisture. Data not included in final results due to expected contamination error.

APPENDIX 2

Mg data

Essex County uses treated road salt which contains a mix of magnesium chloride ($MgCl_2$) and NaCl. Therefore, we measured Mg concentrations in the soil using the same methodology that we used to measure Na concentrations in the soil. Again, there was likely a contamination issue with our April JBT soil samples since the concentrations measured were greater than those in CB (Figure 15). We did notice similar trends in increased Mg content between sites to what we saw with Na content, but because of the clear contamination did not run statistics to prove the difference (Figure 16). We suspect that we may not have diluted the JBT samples as we thought we did, although the concentrations would still be higher than expected.

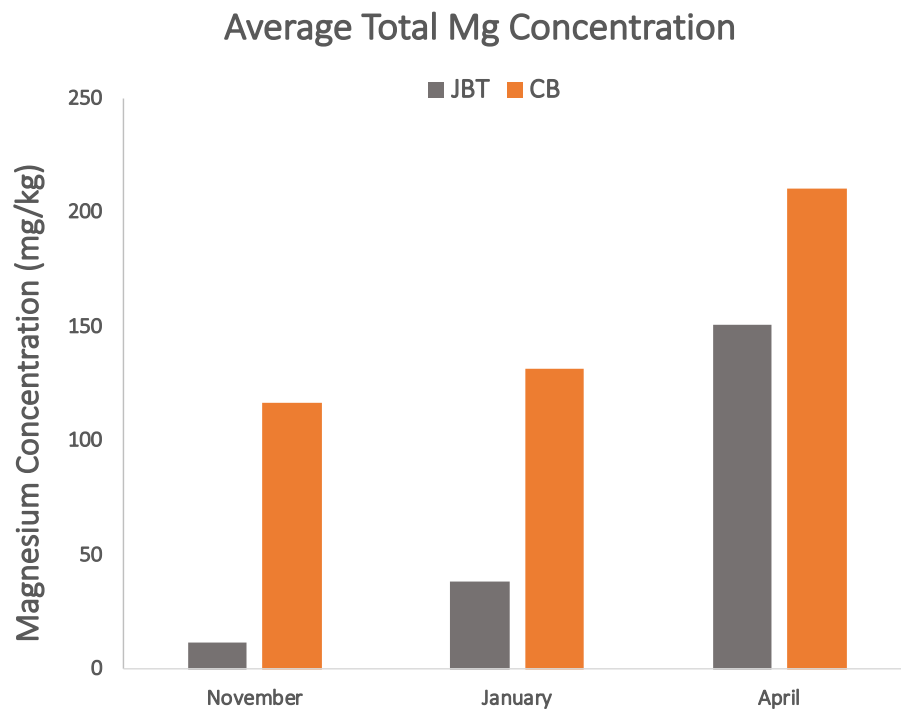


Figure 15: Extractable Mg concentrations of the soil samples in both study sites. The concentration is an average concentration calculated from soil samples taken at 1m, 3m, 6m, 8m, and 10m from the stream along the sampling transect. The values are corrected soil moisture and were found using the following standard curve: $y = 3.066x + 0.0752$ ($R^2 = 0.9666$). Data not included in final results due to expected contamination error.

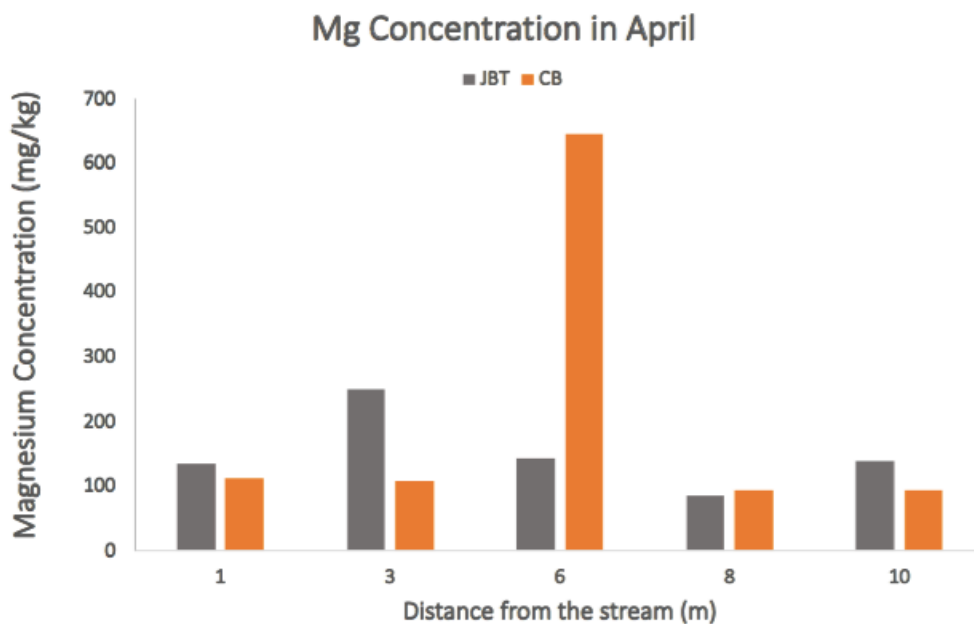


Figure 16: Extractable Mg concentrations of the soil samples collected in April along the field sampling transects at the Johns Brook Tributary and Cascade Brook sites. The concentrations at Johns Brook Tributary and Cascade Brook were found using a formula derived from the standard curve: $y = 3.066x + 0.0752$ ($R^2 = 0.9666$). The Mg concentrations displayed are averages of two subsamples and are corrected for soil moisture. Data not included in final results due to expected contamination error similar to that observed in Cl April data.

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