Biofouling Impacts on Eastern Oyster (*Crassostrea virginica*) Growth and Oyster Condition Index

Megan Reed[(1)(2)], Katie Lankowicz[(3)], Alex Ascher[(2)(4)], Isabelle Sée[(2)]

[(1)]Eckerd College

[(2)]Quahog Bay Conservancy

[(3)]Gulf of Maine Research Institute

[(4)]Woods Hole Oceanographic Institute

Abstract:

Aquaculture is a growing global industry. The Gulf of Maine has seen a consistent increase in eastern oyster (*Crassostrea virginica*) aquaculture in recent years, which has brought millions of dollars of profit to working waterfronts in Maine. Maine is near the northern range limit of oysters, and the colder waters in this area limit the duration of the annual growing season. In order to successfully grow oysters, negative impacts on oyster growth, like biofouling, must be kept to a minimum. This study investigated the impact of biofouling on oyster growth and condition over the summer of 2025. Though growth rate and condition were not significantly different between oysters grown in clean and biofouled bags, oysters grown in biofouled bags had a high variance of achieved size by the end of the growing season. It is important to note that biofouling can cause spatial heterogeneity of competition for food and speed of water flow within a single bag, resulting in higher variation of oyster growth rates than for oysters that are grown in clean bags. This can make it more difficult for oyster farmers to grow a uniform product, potentially hurting their profits.

Introduction

With the decline in traditional fishing yields due to overfishing and other anthropogenic factors (Maine Aquaculture Association, 2025), the aquaculture industry is expected to continue growing to meet global demand for seafood. In 2020, the global aquaculture industry was valued at $281.5 billion (Food and Agriculture Organization of the United Nations, 2025). Opportunities in aquaculture can help economically diversify coastal communities and support working waterfronts. In Maine, the aquaculture industry brought in $73.4 million in 2014 (Maine Aquaculture Association, 2025). Shellfish aquaculture has become vital to many Maine coastal communities. In 2018, there were 190 active aquaculture leases for finfish, shellfish, and kelps (Maine Aquaculture Association, 2025), operating in Maine's coastal waters, which covered 1,319 acres of coastal waters. This area grew to 1,558 acres in 2019 (Maine Aquaculture Association, 2025). Maine oyster aquaculture is a quickly growing industry providing an additional and more stable source of income for communities facing declines and distribution shifts in wild fisheries. In 2024, Maine oyster aquaculture brought in close to $15 million, making it the 3rd most valuable fishery in the state (Department of Marine Resources, 2025).

The eastern oyster (*Crassostrea virginica*) is the primary species cultivated in Maine's oyster aquaculture industry, though Maine is near the northern edge of the species' native range. Eastern oysters' optimal growth temperature range is between 10-30 °C (Sanibel - Captiva Conservation Foundation, 2025). Oysters are typically grown in bays, estuaries, and rivers which have summer temperature ranges of 12 °C to 23 °C (Jiang et al., 2022). This means that the growing season in Maine is very short, with temperatures only optimal for growth from June

through September. Maine aquaculturists must therefore optimize conditions to take full advantage of the short growing season. Biofouling, defined as the unwanted accumulation of non-target organisms on or near cultured species and infrastructure, is a common challenge (Fitridge et al., 2012).

Biofouling can cause a number of problems in oyster aquaculture. Fouling can result in physical damage to the oyster. Biofouling organisms like barnacles can attach to oyster shells, and if growing on the hinge or lip of the shell, may impact filter feeding (Fitridge et al., 2012). This harms the organism and may either significantly stunt its growth or result in death. Filter-feeding biofouling organisms create competition for food resources (Fitridge et al., 2012). As oysters are sessile and dependent on currents and other processes to circulate water containing food near them, biofouling that reduces water flow within the structures on which oysters grow can also be detrimental.

In addition to biological impacts, biofouling puts increased stress on the farming equipment (Fitridge et al., 2012). The added weight of organisms can greatly increase the overall weight of the equipment, making it difficult for aquaculture farmers to move and maintain the equipment. The increased time needed to tend to biofouled equipment could increase labor costs. Over time, the impacts of biofouling on aquaculture equipment, such as the cages and bags that oysters are grown in, can compromise its integrity, increasing repair costs and decreasing the lifespan of the equipment. If more money has to be put into repairing and replacing aquaculture equipment, there will be less profit for oyster farmers.

In this study, we monitored oyster growth over the summer, the optimal growth season, to quantify the impact of biofouling on oyster growth and condition. It is expected that oysters in non-biofouled treatments will have higher growth rates, higher survival, and better condition throughout the summer than oysters in biofouled conditions.

Methods

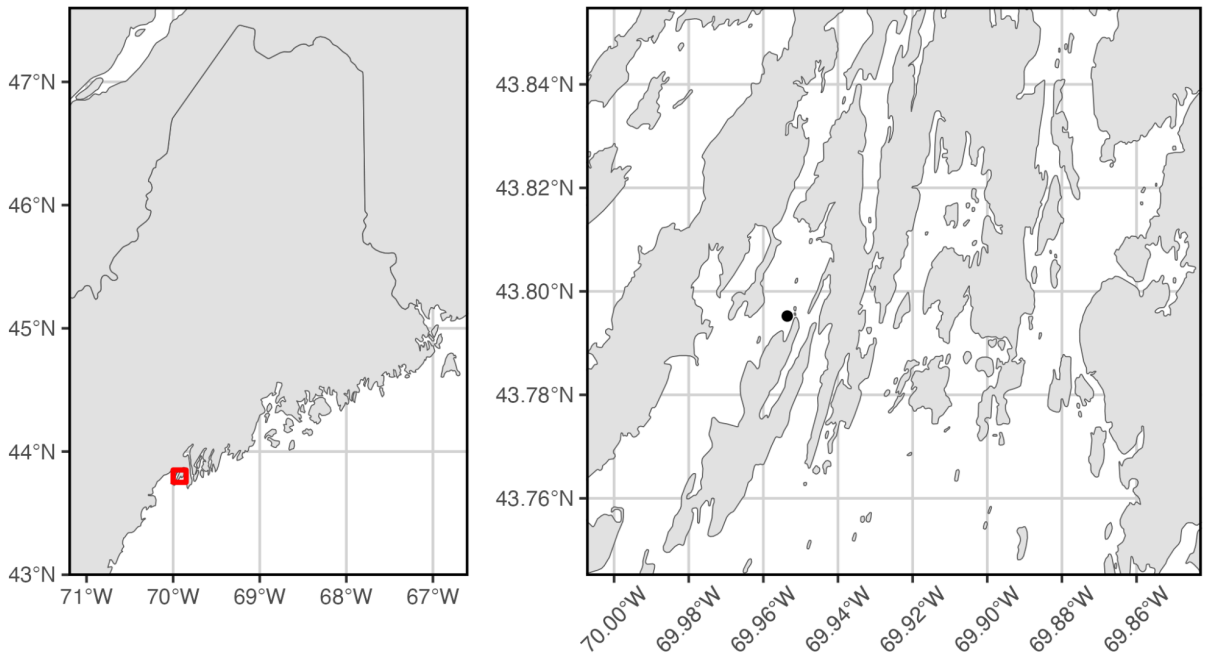

Figure 1: Map of Quahog Bay, Harpswell Maine. The black dot represents the location of the Dogs Head lease site located in Harpswell Sound.

This study was conducted at Snow Island Oysters' Dogs Head lease site in Harpswell Sound, Harpswell, Maine (Figure 1). This lease uses a floating cage design, where cages are floated at the surface in rows and anchored on either end of the row to the substrate. Within each floating cage there are plastic mesh bags that the oysters live within. Oysters were selected from a batch

of freshly tumbled year-old oysters. Tumbling of oysters is a process in which oysters are passed through a rotating metal tube. This process helps remove biofouling. The chipping of the growing shell edge during tumbling is also hypothesized to redirect energy into growing deeper, rather than longer shells, which results in a more desirable product for consumers. After the tumbling process, no biofouling organisms were present on the experimental oysters. The oysters selected for the experiment were all of similar sizes at the beginning of the experiment. A Levene test for homogeneity of variance was used to confirm that in week 1, there was no significant variance in oyster lengths between the individuals in the clean and biofouled treatments (p-value = 0.383).

For each treatment, 40 oysters were placed in each of two bags. The biofouled bags had large amounts of fouling growth when the study was initiated (Figure 2). The clean bags started with no growth and were swapped out midway through the experimental period to minimize potential fouling growth. Both bags for each treatment were placed into a floating cage. To ensure consistent environmental conditions between treatments, the cages were placed adjacent to each other. For both treatments, one bag was considered “active” and the second bag was “inactive”. The active bags were equipped with HOBO data loggers to record temperature and light intensity every 5 minutes. Loggers were affixed to the active bags with the photoreceptor oriented upward towards the sky. The inactive bags held extra oysters, which served as replacements in case of mortality in the active bag. Each bag contained 5 oysters marked with a zip tie attached with marine glue to the left (or cup) shell and paint marker numbers on the right (or flat) shell. In all bags, marked oysters were measured weekly using digital calipers, with measurements recorded to the nearest hundredth millimeter.

Figure 2: Biofouled and clean bags used in the experiment. Images on the top show biofouled condition bags. Bottom images show bags used in the clean treatment. Colored zip ties show hobo attachment and indicate the bottom side of the bag.

We selected 3 oysters from both treatments' "active" bags each week to calculate Oyster Condition Index (OCI) (Rainer et al., 1992). OCI measures the ratio of dry meat mass to wet meat mass. Higher OCI values correspond to higher meat content, which is more desirable for consumers and may offer insight into the health of the oyster. To calculate OCI, the length and width of each oyster were measured in millimeters using digital calipers. Wet weights of the

whole oyster, shell, and meat were recorded to the nearest hundredth gram. Then the oysters were placed in a 55 °C drying oven for 48 to 72 hours to allow all moisture to evaporate. Then, dry weights of the shell and meat were taken. Two methods of calculating the condition index were considered:

(dry meat weight (g)/ whole weight (g) - wet shell weight(g)) x 100 = Index 1

dry meat weight (g) x 100/ dry shell weight (g) = Index 3

(Rainer et al., 1992)

To limit the potential impact of inaccurate weight measurements, index 3 was selected as the index of choice and used in all OCI analyses.

All OCI and length data were digitized and analyzed in R. OCI values across the experimental period were grouped at the treatment level. A Mann-Whitney-U test was used to identify differences in mean OCI between the biofouled and clean treatments.

Linear models were used to regress length measurements of individual marked oysters to week. The slope of each regression is therefore an estimate of the weekly growth rate of an individual oyster. Oysters that died over the course of the experiment, had negative growth rates, or had insignificant relationships between length and week were removed from the dataset. It was assumed that these oysters had one or more instances of a measurement or handling issue throughout the experiment, which would confound the relationship between length and time elapsed. Once the models were finalized, we used a Mann-Whitney-U test to identify any difference in the mean growth rate between the clean and biofouled treatments. For both

treatments, a Levene test for homogeneity of variance was used to test for differences in the variance of oyster lengths in the first and final weeks of the experimental period.

A generalized additive model was used to identify the relationship between oyster condition index and experimental treatment, water temperature, fluorescence, current speed, colored dissolved organic material (CDOM), and rainfall. Because our data came from a field experiment conducted at a working oyster farm, this modeling step was necessary to contextualize how fluctuating environmental conditions may have affected our results. Temperature data came from the on-site HOBO loggers, which provided fine-scale resolution to identify existing temperature differences between the experimental oyster cages. Oyster feeding rates and metabolic processes are temperature-dependent; therefore, temperature was considered a crucial parameter. Data for fluorescence, CDOM, and current speed were sourced from the Bowdoin College Land/Ocean Biogeochemical Observatory (LOBO) Buoy in Harpswell Sound, which is approximately 4 km downstream from the Dogs Head oyster lease (http://bowdoin.loboviz.com/). Fluorescence, commonly used as a proxy for the amount of Chlorophyll a in the water, is useful to estimate the availability of phytoplankton food resources. The sensor used in the Bowdoin College LOBO interprets fluorescence values to estimate the concentration of photosynthetic material in the water column (ug/L). Another proxy used is CDOM, a proxy for riverine input (QSDE). Current speed may also affect food availability. Oysters are sessile organisms and have a limited ability to draw water in. They rely on the constant circulation of water to renew food resources where they are settled, or else they will filter out all available food. Salinity was considered as another potential covariate, but was ultimately not included in modeling efforts because it was correlated with temperature. Daily total rainfall data were sourced from a privately-owned personal weather

station within 1 km of the Dogs Head oyster lease (https://ambientweather.net/dashboard/d3b2e4f5e2cdc190fabd235f9468f5f2/tiles).

Results

The overall trend in weekly oyster lengths for both the clean and biofouled treatments was positive (Figure 3, 4). This is what we would expect to see as the experimental oysters grow over time. Linear models indicated that growth rates were similar between treatments. Mann-Whitney-U tests provided evidence that there was no statistically significant difference between the biofouled and clean treatment growth rates (p-value = 1) (Figure 5).

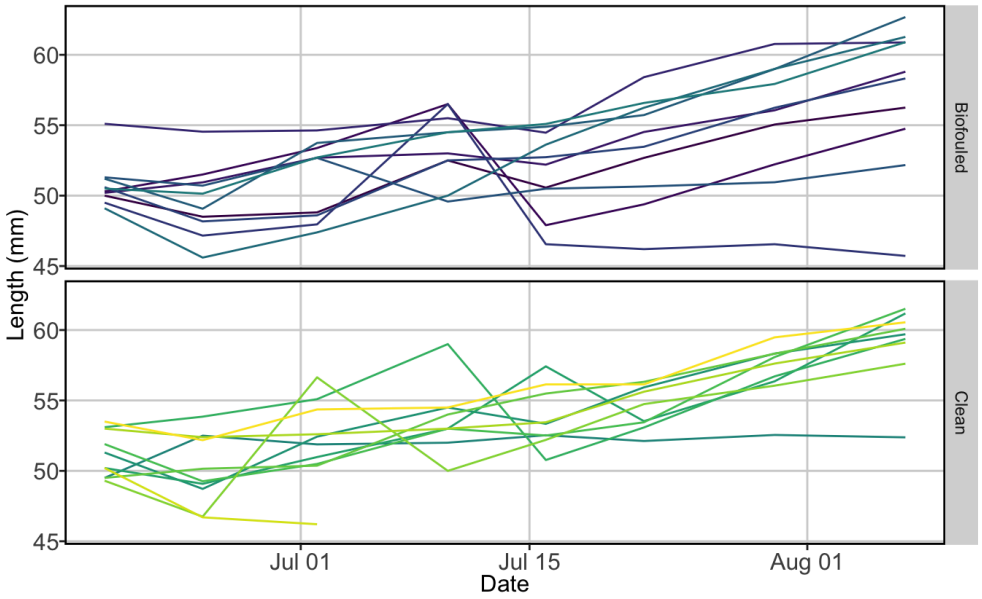

Figure 3: Marked oyster length(mm) over the course of the study. Each color represents an individual oyster. The top panel shows oysters in the biofouled treatment (n=10), and the bottom panel shows oysters in the clean treatment (n=10).

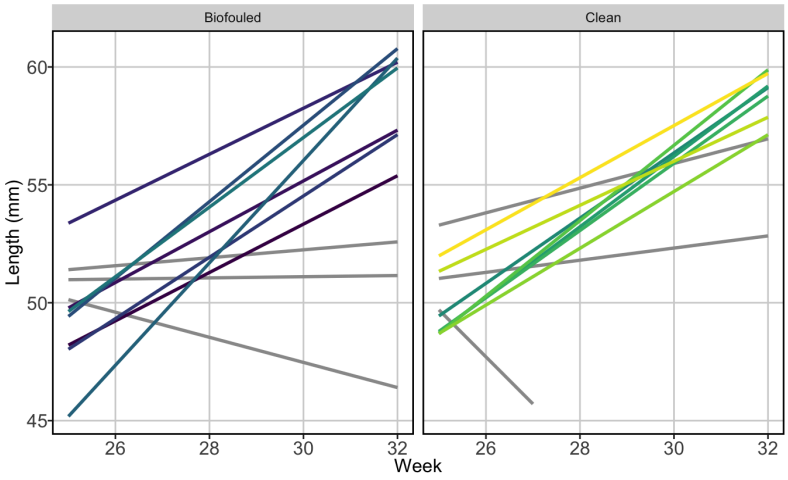

Figure 4: Oyster length in each week as modeled from the growth rates extracted from the linear models. Oysters in the biofouled treatment are represented in the left panel, and oysters in the clean treatment are represented in the right panel. Colored lines indicate individual oysters. Gray lines indicate oysters removed from growth rate analysis due to mortality or inconsistent precision of measurements.

However, the data indicated a trend of more consistent growth rates for oysters in the clean treatment. For the clean treatment, most of the oysters grew to similar sizes as each other (Figure 3). Levene tests for homogeneity of variance indicated that there was no significant difference in the variance of individual oyster lengths between the first and final weeks of the experimental period for the clean treatment (p-value = 0.753). For the biofouled treatment, the final size of the oysters was much more variable (Figure 3). The Levene test for this treatment indicated a significant difference in the variance of oyster lengths in the first vs. the last week of the experimental period (p-value = 0.038).

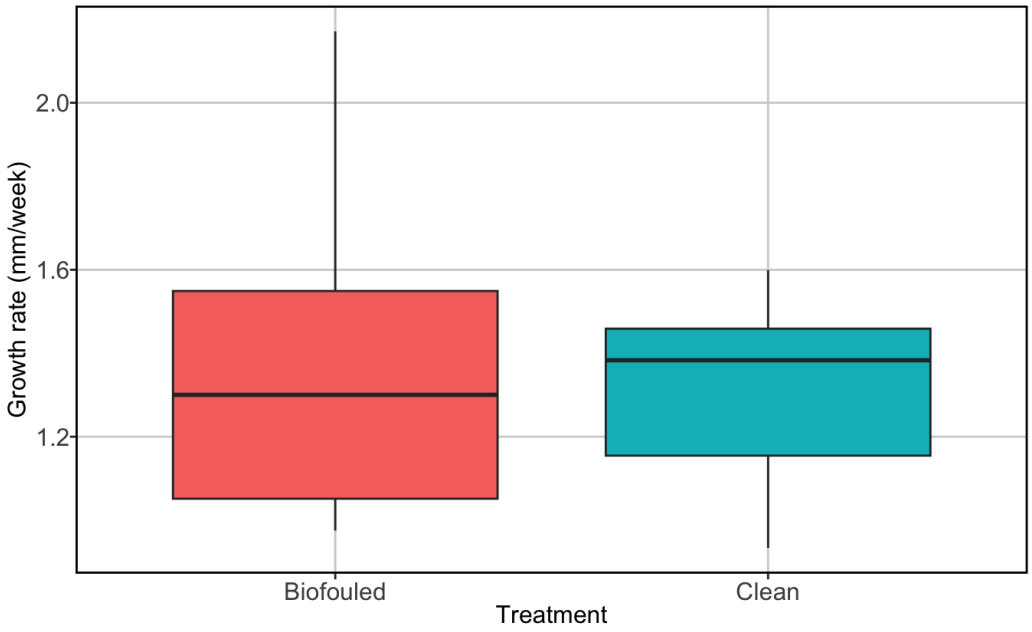

Figure 5: Distribution of oyster growth rates (mm/week) for both the biofouled and clean treatments.

The environmental conditions over the course of this study varied. Current speed varied with prevailing wind direction and speed, as well as diurnal and monthly tidal cycles. At the beginning of the experiment, fluorescence had a very large spike above 30 ug/L, and then declined and stayed consistent around 10 ug/L (Figure 7). Both of these impact the food available to the oysters. The spike in fluorescence in early June also coincided with a spike in colored dissolved organic matter (Figure 6). This is a proxy for fresh water runoff seen with rain events. Though the daily rainfall record has a gap during early June, there is at least one day in this period with significant rainfall (Figure 7). Another way we can see this rain input is in the dip in salinity down to 25 ppt in early June (Figure 6). It is likely that in this period, the rainfall event was a stronger influence on the conditions of the surface water than tidal flushing. Additionally, we saw a slight increase in temperature over the course of the study from around 16 °C to around 19 °C (Figure 6), which is within the range preferred by *C. virginica*.

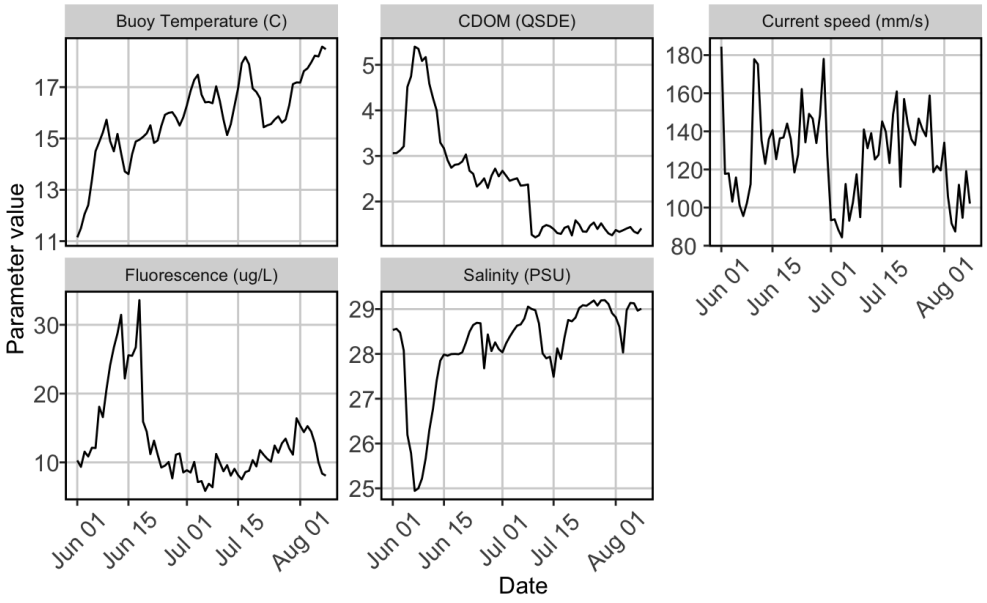

Figure 6: Environmental conditions over the course of the study. Top row, from left to right: surface temperature at the Bowdoin LOBO (°C), Colored dissolved organic matter (QSDE), Current speed (mm/s), Fluorescence (ug/L), and Salinity (ppt).

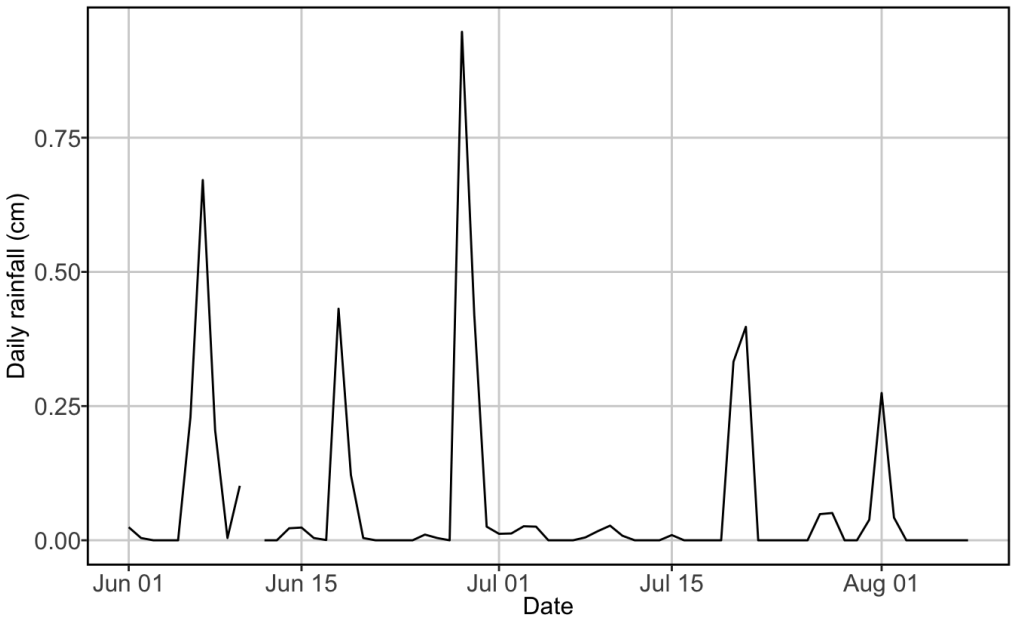

Figure 7: Daily rainfall (cm/day) at the Dogs Head Ambient Weather personal weather station over the course of the study period.

Throughout the summer the OCI values for both clean and biofouled showed a positive trend (Figure 8). Notably, the clean treatment showed a greater rate of increase in OCI as compared to the biofouled treatment, though this is not a statistically significant difference (Figure 8). Mann-Whitney-U test results indicated that, when OCI values for each treatment were grouped across the summer, there was no significant difference between treatments (p value = 0.709) (Figure 9).

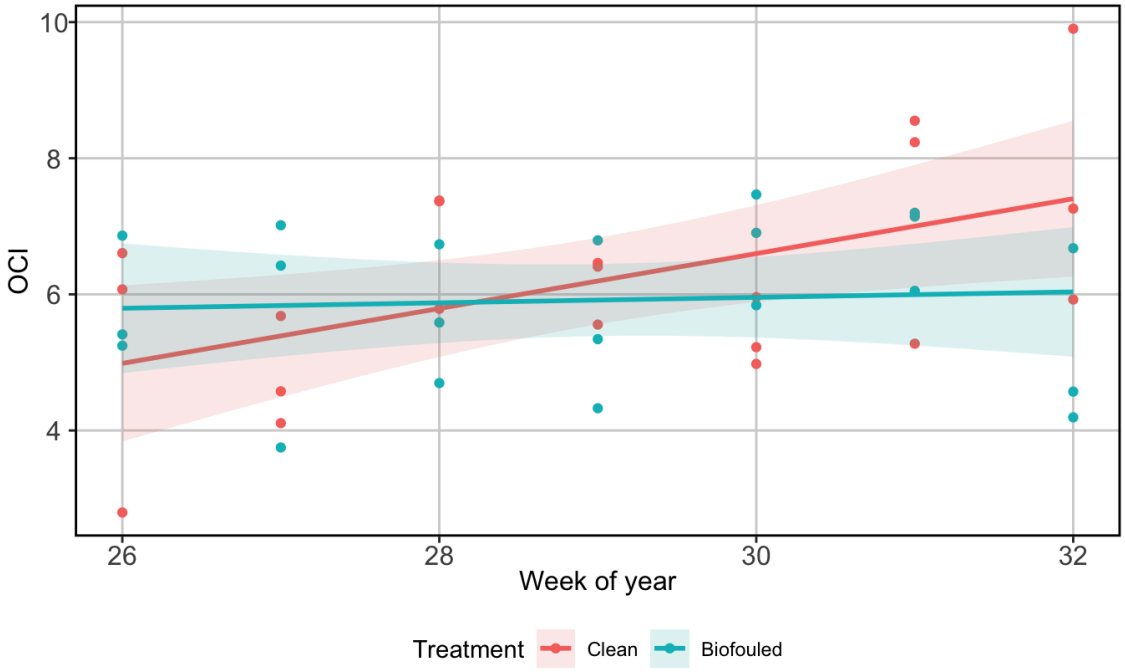

Figure 8: Weekly OCI trends for clean and biofouled treatments. Dots represent individual OCI values, lines represent average OCI per week.

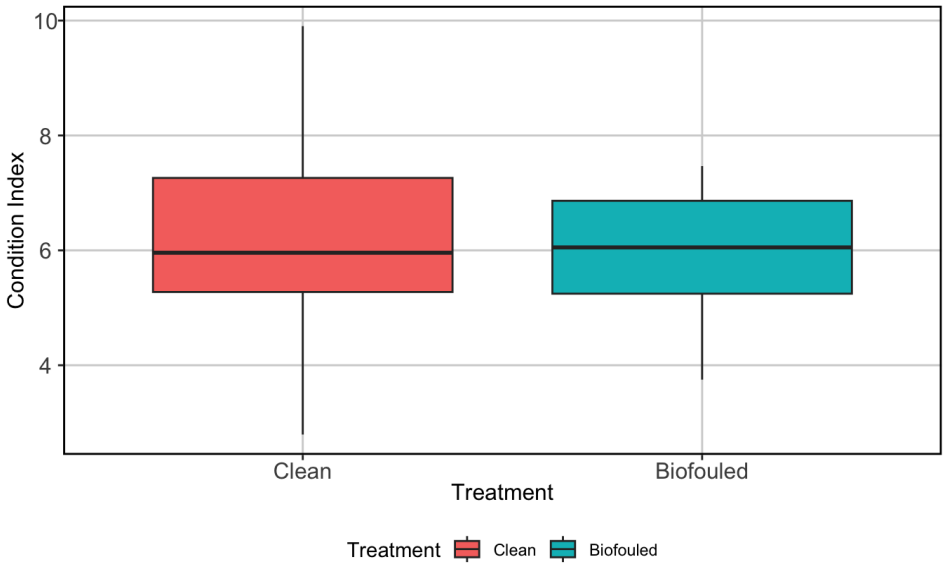

Figure 9: Distribution of oyster condition index values for clean and biofouled treatments.

No significant relationship was detected between OCI and any of the explanatory environmental variables (Table 1, Figure 10). This means that there is no significant relationship showing that these covariates impact OCI. Additionally, this model explained very little of the variation in oyster condition index (R^2=0.137).

Table 1: P values for relationships between environmental covariates and oyster condition index.

Environmental Covariate	**P - Value**
Week	0.099
HOBO temperature: clean treatment (℃)	0.517
HOBO temperature: biofouled treatment (℃)	0.579
Colored Dissolved Organic Matter (QSDE)	0.747
Current speed (mm/s)	0.899
Fluorescence (ug/L)	0.556

Daily Rain (cm)	0.188

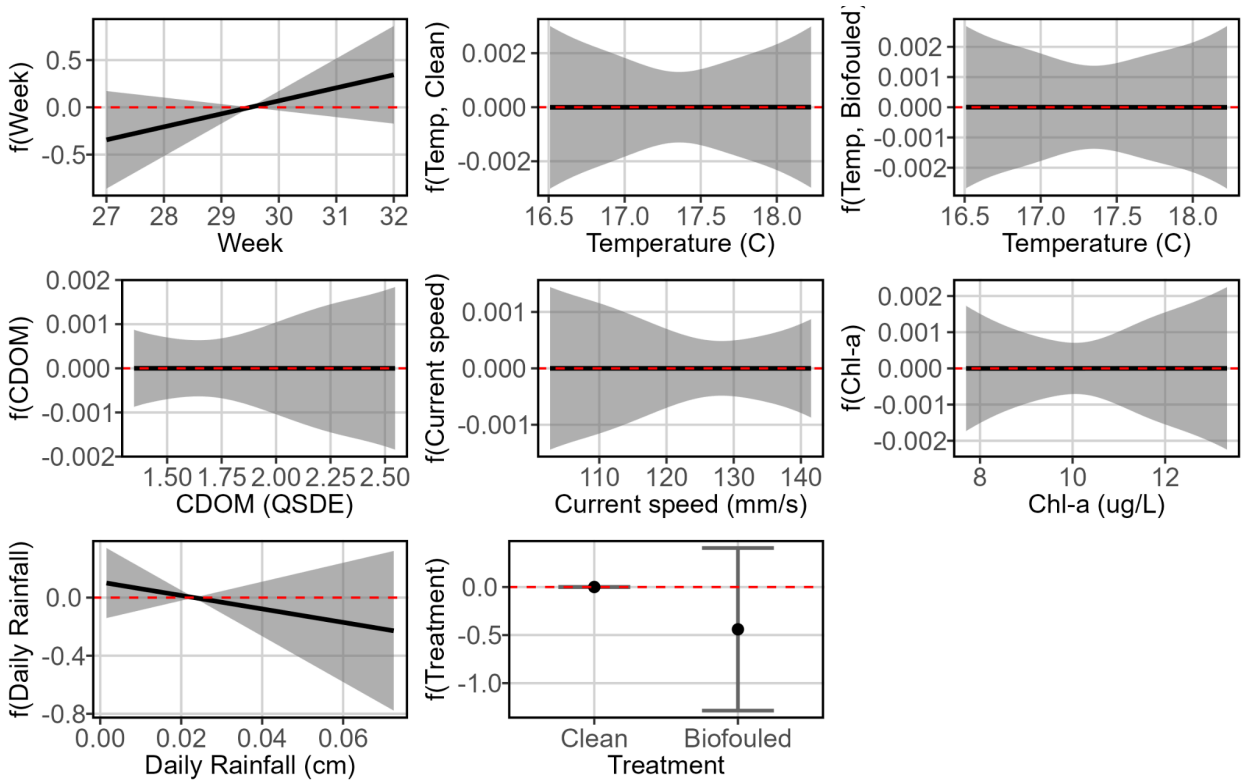

Figure 10: Modeled environmental covariates' effects on OCI. Gray shaded areas represent 95% confidence intervals, and red dashed lines show no relationship.

Discussion

Overall, there was no significant difference in oyster growth rates or condition index between oysters grown in biofouled or clean conditions. Additionally, no environmental covariates had significant relationships to the oyster condition index. A variety of factors relating to our experimental setup may have confounded our results. It is possible that the calipers used in the study did not have the necessary precision to measure the weekly change in oyster lengths, as this was often on the scale of tenths of a millimeter. Extracting the oysters from their growing

bags each week to measure them may have resulted in unintentional chipping of the shell edge, which would explain the instances of negative weekly growth. Oysters may also chip naturally within the growing bags, as currents or boat wakes jostle the individuals.

Another important note is the small sample size used in this study. Only 10 oysters per treatment were measured weekly, but only 7 individuals per treatment could be used to create growth models. Because of the small sample size, we were limited to using non-parametric rank-sum comparisons (Mann-Whitney-U tests) to test for differences in growth rates between groups. The lack of specificity in this test and the low sample size resulted in a p-value of 1 for the comparison of modeled oyster growth rates. Though the test was applied correctly and the raw data suggest little to no ecologically relevant difference in growth rates between treatments, little confidence should be placed in these results.

It is possible that environmental conditions experienced by the oysters throughout the study did not vary enough to reveal the actual relationships between different parameters and the condition index. Visualizations from the GAM outputs illustrate that no environmental covariates have significant relationships to OCI at the ranges experienced by the experimental oysters (Figure 10). The short time scale of this study may also have impacted the results. This study was run from June through early August, which is fully within the optimal growing season. Critically, the temperature was consistently warm (16-19 °C). This does not reflect the shifting of temperature conditions that oysters grown in Maine truly experience over a year. Oysters grow relatively slowly (Mallet et al., 2009), so there was not much potential growth to observe over such a short time interval. Running the experiment over months to years in a more controlled environment

would be necessary to reveal the cumulative effects of biofouling on oyster growth. It is also possible that other, unmeasured environmental covariates may impact OCI values. And as in the growth rate analyses, small sample sizes may have limited the statistical power of the study.

Despite these difficulties, we still observed negative impacts of biofouling on oysters. In this experiment, biofouling was found to result in uneven growth rates across individual oysters. Oysters within the clean bags exhibited similar growth rates. On the other hand, oysters within the biofouled bags exhibited significant variation in their growth rate, leading to a large variety of sizes at the end of the experimental period. This could be attributed to the clean bag being a more uniform environment for the oysters to grow in, resulting in little within-bag variation in growth rates. But the biofouling bag has many species of fouling organisms colonizing different areas. Oysters residing in different parts of the bag may be subject to varying levels of competition from barnacles, tunicates, or mussels. They may also have reduced water flow in certain heavily fouled parts of the bag, preventing adequate water flow to supply food for their growth. Inconsistent products are not a good thing for oyster farmers. If growth is inconsistent, as seen in the biofouled bags, they will need to spend more time sorting oysters by size because of the variation in size. Overall, even if biofouling does not result in the death of oysters, it can impact the uniformity of their growth and make it more difficult for oyster farmers to create a uniform product for market.

Acknowledgements:

First, I would like to acknowledge and thank Pat Scalan for founding Quahog Bay Conservancy and creating the opportunity for this internship program. This program allows undergrads to work on a diverse array of projects and gain significant knowledge about working in a scientific setting. Next, I would like to thank Isabelle See and Tait Nygaard for their large amount of support to our internship program this summer. This project and internship would not have been possible without your endless support. Next I would like to thank Dr. Katie Lankowicz for her support in my project, your hours of help with everything from the experimental design, to analysis of data and writing of this paper was endlessly helpful to this project. Additionally, I would like to thank Dr. Alex Ascher for his help and feedback throughout this project. I would also like to thank Dr. Walt Golet and the University of Maine Pelagic Fisheries Lab for the opportunity to tour his lab, work with his interns and for giving us an offshore charter experience. Lastly, I would like to thank my fellow interns, Eliza Chance, Caroline Salvatierra, and Annabella Williams for their support on this project and all of our other projects throughout the summer.

Works Cited

Department of Marine Resources. (2025). Maine 2024 Commercial Fisheries Value Increases by More than $74 Million.

Fitridge, I., Dempster, T., Guenther, J., & de Nys, R. (2012). The impact and control of biofouling in marine aquaculture: a review. *Biofouling*, *28*(7), 649–669.

Food and Agriculture Organization of the United Nations. (2025). The state of world fisheries and aquaculture.

Jiang, Binbin., Boss, Emmanuel., Kiffney, Thomas., Hesketh, Gabriel., Bourdin, Guillaume., Fan, Daidu., Brady, Damian C. (2022). Oyster aquaculture site selection using high-resolution remote sensing: A case study in the gulf of Maine, United States. Frontiers in Marine Science.

Maine Aquaculture Association. (2025). Frequently asked questions.

Mallet, André L., Carver, Claire E., Hardy, Matthew. (2009). The effect of floating bag management strategies on biofouling, oyster growth and biodeposition levels. Aquaculture, Vol 287, 312-323.

Rainer, Julia S., Mann, Roger. (1992). A comparison of methods for calculating condition index in eastern oysters, *Crassostera virginica* (Gmelin, 1791). Journal of Shellfish Research, Vol 11, No1, 55-58.

Sanibel - Captiva Conservation Foundation. (2025). Eastern Oysters *Crassostrea virginica.*