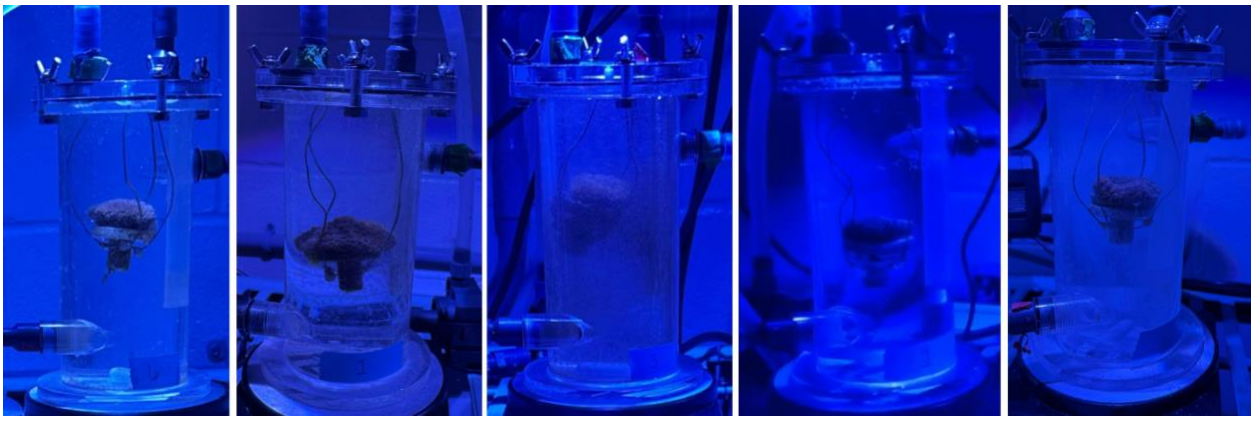
Source-Specific Impacts of Suspended Sediments on *Orbicella faveolata* Health Benchmarks



Visual Comparison of the five turbidity (NTU) treatments *Orbicella faveolata* were exposed to during chronic turbidity exposure experiments.

Source-Specific Impacts of Suspended Sediments on *Orbicella faveolata* Health Benchmarks

Final Report

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June 2025

Completed in Fulfillment of Agreement # C42415 for

Florida Department of Environmental Protection

Coral Protection and Restoration Program

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This report should be cited as follows:

Bahr, K., Bretzing, R., Coleman, M., and Willans, J. 2025. The effects of suspended sediments on susceptible Florida corals . Florida Department of Environmental Protection Coral Protection and Restoration Program. pp 1- XX.

This report was prepared for the Florida Department of Environmental Protection's (DEP) Coral Protection and Restoration Program by Texas A&M University-Corpus Christi. Funding was provided by the DEP Award No. # C42415. The views, statements, findings, conclusions, and recommendations expressed herein are those of the authors and do not necessarily reflect the views of the State of Florida or Florida Department of Environmental Protection.

MANAGEMENT SUMMARY

This project, a collaboration between Texas A&M University-Corpus Christi and the Florida Department of Environmental Protection (DEP), investigated the impact of suspended sediments from coastal development activities on the health of *Orbicella faveolata*, a threatened coral species. The primary objective was to assess the physiological responses of corals exposed to turbidity stress and sediment contaminants, providing data that can inform water quality management and permitting decisions.

Corals were exposed in a controlled laboratory setting to two types of sediment—one collected from a natural carbonate reef and another from Port Everglades, a heavily trafficked port area with known organic and heavy metal contamination. Each sediment type was tested at two turbidity levels (4 and 15 NTU) over 30 days, after which coral recovery was monitored for an additional five weeks. Throughout the exposure and recovery periods, coral health was assessed using physiological indicators, including oxygen consumption, photosynthetic efficiency, calcification rates, protein concentration, chlorophyll content, and symbiont density.

Findings showed that port sediments had significantly higher levels of organic matter and elevated concentrations of heavy metals, including arsenic, chromium, and copper. Although corals did not experience mortality or visible bleaching, measurable physiological stress occurred across all treatments. Corals exhibited reduced oxygen production, lower protein and chlorophyll levels, and variable calcification responses, with some corals exposed to port sediment showing elevated calcification potentially driven by higher energy input per symbiont. However, these responses likely reflect a compensatory mechanism in response to metabolic stress. Notably, several corals failed to recover to pre-exposure photosynthetic efficiency even after five weeks, particularly those subjected to high turbidity and port sediment, indicating potential long-term impacts.

The study demonstrates that even moderate turbidity levels, especially when combined with poor sediment quality, can impose sublethal but significant physiological stress on vulnerable coral species. These early warning indicators, not detectable through visual surveys alone, should be integrated into environmental assessments and permitting frameworks. Incorporating sublethal physiological benchmarks will enable more accurate risk assessments for coastal development and dredging activities near coral reefs, supporting DEP's goals of enhancing resilience in Florida's coral reef ecosystems.

Orbicella faveolate post-experimental photo showing signs of tissue loss and skeleton degradation.

EXECUTIVE SUMMARY

Coral reefs are increasingly threatened by coastal development activities that elevate turbidity and resuspend sediment. This project, conducted by Texas A&M University-Corpus Christi (TAMU-CC) in partnership with the Florida Department of Environmental Protection (DEP), evaluated the sublethal impacts of suspended sediments on *Orbicella faveolata*, a threatened and ESA-listed coral species particularly vulnerable to sediment stress and stony coral tissue loss disease (SCTLD).

Using controlled laboratory experiments, the study exposed *O. faveolata* fragments to suspended sediments collected from two sources: a natural reef (carbonate-dominated) and a port channel (fine-grained, contaminated). The experiments were conducted at two turbidity levels (4 and 15 NTU). Corals were assessed over 30 days for metabolic performance (via oxygen consumption), calcification (using total alkalinity anomaly), and photosynthetic efficiency (via PAM fluorometry). A subsequent recovery period was monitored to assess resilience following stressor exposure.

Key findings include:

- Sediment characterization revealed that port sediments had a higher organic content (66.2% vs. 24.0%) and elevated concentrations of heavy metals, including arsenic, copper, and zinc, as well as altered microbial communities with potential pathogenic risks.
- Sublethal physiological impacts were observed across treatments. Corals exhibited trends of reduced photosynthetic efficiency, protein concentration, and symbiont density—despite no visible bleaching—highlighting subtle but meaningful biological stress.
- Calcification and growth rates differed significantly across treatments. Surprisingly, corals exposed to port sediments exhibited higher net calcification in some cases, potentially due to a greater chlorophyll-to-symbiont ratio that supported photosynthesis despite turbidity stress.
- Respiration rates (oxygen consumption) fluctuated over time, indicating metabolic strain. Increased oxygen demand early in exposure may serve as a useful early-warning metric of coral stress.
- Recovery varied by genotype. Some corals failed to return to their pre-stress physiological conditions after five weeks, particularly those from genotypes that showed the most significant photophysiological decline during exposure.

Multivariate analyses revealed that sediment source and associated contaminants (especially heavy metals) were primary drivers of coral response, along with treatment turbidity and coral genotype. While direct mortality was not observed, the cumulative effects of reduced metabolic and photobiological function indicate potential long-term vulnerability under repeated exposure scenarios. These findings provide critical evidence to inform DEP and other regulatory agencies in updating water quality criteria and sediment management practices. The study supports the implementation of sublethal physiological endpoints (e.g., respiration, protein loss, and photosynthetic decline) into environmental permitting and coral reef impact assessments. It also emphasizes the need for stricter turbidity thresholds and post-disturbance monitoring during and after coastal construction activities. In summary, this work advances our understanding of how

chronic, moderate turbidity affects coral health, identifies key sediment-related risk factors, and highlights the importance of incorporating sensitive physiological metrics into coral reef conservation and resilience planning.

ACKNOWLEDGMENTS

Special acknowledgments are extended to Dr. Cheryl Woodley's research group and Dr. Mark Ladd's research group for their valuable contributions to this area of research. We are grateful for their efforts and collaboration throughout this study.

Furthermore, we would like to express our gratitude to the team from the Florida Department of Environmental Protection for their invaluable project feedback and assistance in shaping our experimental design. Their expertise and guidance were instrumental in ensuring the scientific rigor of our study.

A special thanks to David Gilliam and his research team at the CRRAM Lab at Nova Southeastern University for collecting and providing us with the OFAV nursery corals used throughout this project. His team's commitment to coral conservation and Caribbean coral reef rehabilitation is inspiring and the timeline of this project would have been very different without his and his team's help.

TABLE OF CONTENTS

LIST OF FIGURES

LIST OF TABLES

1. INTRODUCTION

1.1 Suspended sediments

Coral reefs are among the most ecologically and economically valuable ecosystems on the planet, providing essential services such as shoreline protection, food security, biodiversity support, and marine-based tourism (Knowlton et al., 2010). These reef systems provide direct ecosystem services to humans by acting as natural wave barriers, significantly reducing coastal erosion and protecting infrastructure (Elliff & Silva, 2017). Approximately half the global human population resides within 200 km of a coast (Kummu et al., 2016), placing increasing pressure on coastal ecosystems, especially coral reefs. Anthropogenic stressors such as dredging, beach nourishment, and land-based runoff have escalated in intensity and frequency, posing substantial threats to reef health (Good & Bahr, 2021; Miller et al., 2016). These activities increase sedimentation and turbidity, key drivers of coral decline by suspending fine particles and contaminants into the water column, reducing water clarity, and altering the photic environment critical for coral photosynthesis (Walker et al., 2012).The situation is particularly concerning in regions like Florida, where port expansion and dredging projects frequently occur in proximity to vulnerable coral habitats. Turbidity, quantified in nephelometric turbidity units (NTU), directly impairs coral photosynthesis, feeding, and settlement. Thresholds for coral stress and mortality have been identified across various studies. In Florida, turbidity levels above 10 NTU have been correlated with significant coral mortality (Miller et al., 2016; Walker et al., 2012), while levels exceeding 30–40 NTU have caused widespread reef degradation in other global regions (Fabricius, 2005). Sensitivity varies among species; some exhibit moderate turbidity tolerance, while others, particularly slow-growing or ESA-listed species, are highly vulnerable (Duckworth et al., 2017; Piniak, 2007; Weber et al., 2006). Elevated turbidity not only affects coral health directly but also increases coral susceptibility to disease, especially under prolonged exposure to suboptimal light conditions and sediment stress (Gilmour, 1999; Pollock et al., 2014; Studivan et al., 2022). To effectively mitigate these risks, it is essential to go beyond descriptive studies and establish biologically relevant benchmarks for coral resilience under turbid conditions. Such benchmarks can guide policy and best practices for sediment management in coastal development projects and provide data to update turbidity standards for state and regional coastal activities.

This research aims to close critical knowledge gaps by examining the physiological responses of a sensitive coral species to turbidity using sediment collected from two distinct locations in Port Everglades, Florida. These findings will inform state and federal water quality standards, support the implementation of Florida's Coral Reef Resilience Action Plan (2021–2026), and improve restoration site selection, planning, and adaptive management strategies. Ultimately, protecting coral reef health supports not only marine biodiversity but also the socio-economic well-being of coastal communities worldwide.

1.2 Sediment Locality and Characterization

Marine sediments in port and coastal development zones are a significant and often localized stressor for coral reef ecosystems. In natural reef systems, sediments are typically coarser, carbonate-based, and generally lower in pollutants due to constant water movement and their distance from industrial sources. In contrast, port sediments are often fine-grained, organically rich, and have a long history of contaminant accumulation from industrial runoff, vessel discharge,

sewage, and stormwater outflows (Bartley et al., 2014). These sediments are frequently disturbed during dredging, a common practice to maintain navigation channels and expand port infrastructure. When dredging occurs, large volumes of previously settled material are resuspended into the water column, often spreading beyond the immediate work area due to ocean currents (Miller et al., 2016). This process mobilizes previously buried contaminants and can dramatically alter water quality conditions over both spatial and temporal scales (Ikenaga et al., 2010). Comprehensive sediment characterization in these two locations allows for a robust understanding of the physical parameters (e.g., grain size, porosity), chemical constituents (e.g., metal concentrations, nutrient levels), and biological content (e.g., microbial communities) (Ikenaga et al., 2010). It is essential to understand site-specific risks and differences in sediment composition and contaminant load between natural and developed coastal zones on the short term (acute responses) and long-term (recovery to disturbance) sustainability of reef ecosystems (Macdonald et al., 1996).

1.3 Heavy Metal Contamination and Microbial Communities

One of the most serious risks associated with dredging is the release of heavy metals that have bound to sediment particles (Giarikos et al., 2023). Elements such as mercury (Hg), lead (Pb), copper (Cu), zinc (Zn), cadmium (Cd), and others can desorb from particles when environmental conditions change and become bioavailable to marine organisms (Giarikos et al., 2023). Coral tissues and their endosymbiotic algae are particularly sensitive to heavy metal toxicity, which can impair photosynthetic function, disrupt calcification, and compromise immune responses. Heavy metals such as copper can directly damage the photosynthetic apparatus in *Symbiodiniaceae* (further referred to as symbionts), reducing the energy available for essential functions like growth and reproduction (El-Sorogy et al., 2012; Glynn et al., 1989; Guzmán & Jiménez, 1992). These sublethal stresses, especially when combined with elevated temperatures or disease exposure, reduce coral resilience and increase the likelihood of mortality or bleaching. Simultaneously, dredging releases sediment-associated microbial communities that can include pathogenic, opportunistic, or invasive microbes. The sediment microbiome, shaped by organic matter accumulation, low oxygen conditions, and nutrient enrichment, often harbors taxa not typically found in healthy reef waters (Ikenaga et al., 2010). Upon resuspension, these microbes can interact with corals in harmful ways, disrupting the native coral microbiome and initiating dysbiosis—an imbalance associated with increased disease risk, particularly in species already susceptible to stony coral tissue loss disease (SCTLD) (Meyer et al., 2019).

<u>Research Questions</u>

1. What are the impacts of different sediments on coral health?
2. How do coral biological benchmarks vary with sediments from different sources?
3. What characteristics of suspended sediment contribute to the observed impact on coral health?

<u>Research Objectives</u>

1. Conduct turbidity exposure experiments of sediments on selected coral species.
2. Examine the effect of turbidity treatments on the metabolism, health, growth and recovery of selected coral species.
3. Define the specific sediment characteristics primarily contributing to the negative impact on selected coral species.

2. MATERIALS AND METHODS

2.1 Coral Acquisition and Maintenance

Twenty-four fragments of *Orbicella faveolata* (OFAV) were obtained from NSU (David Gilliam; SAL-24-2454-SCRP). Corals were packaged and shipped to TAMU-CC following protocols and insights derived from the "Florida Reef Tract Rescue Project". These methods included: 1. Corals will be packed into Uline® plastic bags filled with filtered seawater and oxygen, and 2. Bags will be packed into insulated foam shipping boxes within larger cardboard boxes. Corals fragments were delivered by air cargo to reduce stress associated with handling via mail carriers and were shipped to arrive the next day.

Upon arrival at the holding facility, corals were slowly acclimated to the temperature and chemistry of the water at the facility via slow mixing of water from the tank into the cooler or bag. After the temperature had reached 0.5°C and water chemistry was within an acceptable range, corals were placed into the holding tank, where they were held for a 30-day quarantine period. OFAV fragments were given 10-minute antiseptic prophylactic baths using Lugol's iodine solution, then rinsed with clean aquarium water before placing in the tank. Upon arrival, the initial condition of each coral will be assessed to determine its health status. Corals were recorded as "Healthy" or "Unhealthy" upon arrival at our facility. Examples of "Unhealthy" include excess mucus, broken, abraded, tissue loss, etc. Photographs of each oral (with its ID tag) were taken within the first 24 hours of arrival. Daily water quality parameters and coral health status (Healthy/Unhealthy) were taken every day during the 30-day quarantine period, and then weekly until the start of experimental trials.

2.2 Water Quality and Husbandry

The aquarium system in which the corals were kept consisted of two 110L tanks and a 155L sump (375L total). Each tank is serviced by a pump (Sicce Syncra ADV 40W, Sicce, Pazzoleone, Italy), with a maximum output of 1450 gph, allowing for a possible turnover of 50 times per hour. Two AI Hydra 64HD models provide lighting for each tank, with LED lights positioned 17cm above the water's surface. They are programmed to give a 12-hour photoperiod from 6 am to 6 pm and PAR readings of between 150 and 200 PAR at the base of the corals (Aqua Illumination, Bethlehem, PA). The life support system also features a 1 kW double quartz heater to maintain the water's temperature (Hygger, Chino, CA). The water quality parameter ranges chosen for this system were chosen to closely match those of the location from which the corals came. Parameters maintained include temperature (26-27°C), salinity (35 ppt), pH (8.1-8.3), total alkalinity (2500-2998 $\mu mol\ kg^{-1}$), ammonia (0 ppm), nitrites (0 ppm), nitrates (0-20 ppm), phosphates (0-0.3 ppm) calcium (390-420 ppm) and magnesium (1250-1350 ppm) (Enoch er al. 2018). To ensure that the parameters remain within range, water quality tests were performed multiple times a week, and 25% water changes were performed weekly, using artificial saltwater composed of RODI water mixed with Red Sea salt (Red Sea Fish, Tel Aviv, Israel). Physical parameters, such as

temperature, salinity, pH, and alkalinity, are tested daily. In contrast, biological parameters (ammonia, nitrites, nitrates, phosphates, calcium, and magnesium) are tested once a week. These tests were performed using a variety of methods, including Profi colorimetry test kits to assess ammonia, nitrite, nitrate, and phosphate levels (Salifert, Holland), Hanna colorimeter checkers for magnesium, calcium, and alkalinity, and in-water probes for pH (Orion Star A111, Thermo Scientific, Waltham MA), temperature and salinity (YSI pro Quattro multiparameter meter, YSI Inc., Yellow Springs, OH). To maintain nutrient levels within range, the system was dosed with Bulk Reef Supply Soda Ash, Calcium Chloride, and Magnesium Mix as needed (Bulk Reef Supply, Golden Valley, MN), alongside daily additions of Tropic Marin +NP. Supplementary feeding was also provided to the corals twice weekly using a mix of phytoplankton species and coral food (ReefRoids, Polyp Lab, Quebec, Canada).

2.3 Experimental Approach

Pretrial: Before the start of the experiment, each individual was assigned an ID and pretrial measurements for buoyant weight, wet weight, volume displacement, and Pulse Amplitude Modulated-fluorometry (PAM; Diving PAM 2.0; Heinz Walz GmbH, Effeltrich, Germany) were collected. An initial photo was also taken for each individual. Corals were dark acclimated for a minimum of 30 minutes before collecting PAM readings. PAM was used to measure the photosynthetic efficiency of coral symbionts. Before placing corals in their chambers, each oxygen sensor was calibrated to 0% and 100% oxygen. Once pretrial measurements were obtained and sensors were calibrated, corals were placed in their respective chambers, which were assigned using a random number generator, and allowed to acclimate for 24 hours.

Days 1-30: After acclimation, the individuals were held at a turbidity level of either 4 or 15 Nephelometric Turbidity Units (NTU) using sediment obtained from either the Port Everglades or a reef located off the coast of Florida. This was done to test the differences between sediment types and NTU on the calcification rates of the individuals. To ensure that turbidity levels were being maintained, water samples were taken during the flush period every 2 hours between 8:00 and 17:00 Monday through Friday and analyzed using a HACH 2100Q Portable Turbidimeter (HACH Company, Loveland, CO). Samples were also collected at 10:00 and 12:00 on Saturday and Sunday to ensure that the turbidity level was being held over the weekend. Sumps were dosed with their respective sediment types when needed based on NTU readings. Water samples were collected on Monday, Wednesday, and Friday at 12:00 to measure the TA of the chamber water using a Metrohm Eco titrator. At the midway point (Day 15), the individuals were removed from their chambers and allowed to dark acclimated for a minimum of 30 minutes in order to acquire PAM values. Midpoint values were also collected for buoyant weight, wet weight, and volume displacement. A photo was also taken of each coral. The coral plugs and chambers were scrubbed to remove any algae growth. The individuals were then placed back into their chambers and the experiment continued.

Post-trial: Once the 30-day incubation concluded, the individuals were removed from their chambers and dark acclimated for a minimum of 30 minutes in order to acquire endpoint PAM measurements. Endpoint values were also acquired for buoyant weight, wet weight, and volume displacement. The coral fragments were then cut in half using a diamond band saw (Gryphon Diamond Band Saw Model C-40, Gryphon Corporation, Sylmar, CA). One half was placed back into the holding tank to recover, while the other was frozen for destructive endpoint analysis.

Recovery measurements (PAM, buoyant weight, wet weight, volume displacement, and a photo) were taken once a week for four weeks and then biweekly until the individuals recovered to their pretrial PAM value.

Table 1. Experimental approach outlining coral individuals, which treatments they were exposed to, and the custom respirometry chamber they were assigned to during each trial. Chambers were spaced evenly across two metal racks with the "Rack" column indicating their position.

Trial	Coral ID	Rack	Treatment	Chamber number
1	OFAV69.1	A	Port 4	1
	OFAV69.3	A	Port 15	3
	OFAV3.4	A	Port 15	4
	OFAV3.2	A	Port 4	5
	OFAV69.4	A	Control	6
	OFAV3.3	B	Control	7
	OFAV69.2	B	Reef 4	8
	OFAV3.1	B	Reef 15	9
	OFAV3.5	B	Reef 4	10
	OFAV69.5	B	Reef 15	11
2	OFAV2.3	A	Port 4	1
	OFAV2.2	A	Port 15	3
	OFAV1.2	A	Port 15	4
	OFAV1.5	A	Port 4	5
	OFAV2.1	A	Control	6
	OFAV1.3	B	Control	7
	OFAV1.4	B	Reef 4	8
	OFAV2.5	B	Reef 15	9
	OFAV2.4	B	Reef 4	10
	OFAV1.1	B	Reef 15	11

2.4 Timeline

Turbidity threshold experiments were conducted November 6th – December 8th, 2023, with a quality assurance experiment conducted April 8th - April 12th, 2024, to compare treatment manipulation methods. Multi-stressor experiments were conducted January 22nd, 2024 – February 16th, 2024, with a quality assurance experiment conducted April 1st – April 5th, 2024, to compare treatment manipulation methods. All experimental weeks consisted of 5 days with 3 days (72 hours) of treatment exposure.

Table 2. Experimental schedule for turbidity exposure experiment trial 1 from Wednesday February 12th, 2025 – Friday March 14th, 2025. Columns PAM, Buoyant weight, Turbidity, Total alkalinity, Water quality, and Photo indicate processes used to assess biological condition, with an "X" indicating which day these metrics were measured during the 30-day trial.

Experiment	Date	Weekday	Day	PAM	Buoyant weight	Turbidity	Total alkalinity	Water quality	Photo
1	2/12/2025	Wed	0	X	X				X
	2/13/2025	Thurs	1			X			
	2/14/2025	Fri	2			X	X	X	
	2/15/2025	Sat	3			X			
	2/16/2025	Sun	4			X			
	2/17/2025	Mon	5			X			
	2/18/2025	Tue	6			X	X	X	
	2/19/2025	Wed	7			X			
	2/20/2025	Thurs	8			X			
	2/21/2025	Fri	9			X	X	X	
	2/22/2025	Sat	10			X			
	2/23/2025	Sun	11			X			
	2/24/2025	Mon	12			X	X	X	
	2/25/2025	Tue	13	X	X	X			X
	2/26/2025	Wed	14			X	X	X	
	2/27/2025	Thurs	15			X			
	2/28/2025	Fri	16			X	X	X	
	3/1/2025	Sat	17			X			
	3/2/2025	Sun	18			X			
	3/3/2025	Mon	19			X	X	X	
	3/4/2025	Tue	20			X			
	3/5/2025	Wed	21			X	X	X	
	3/6/2025	Thurs	22			X			
	3/7/2025	Fri	23			X	X	X	
	3/8/2025	Sat	24			X			
	3/9/2025	Sun	25			X			
	3/10/2025	Mon	26			X	X	X	
	3/11/2025	Tue	27			X			
	3/12/2025	Wed	28			X	X	X	
	3/13/2025	Thurs	29			X			
	3/14/2025	Fri	30	X	X	X			X

Table 3. Experimental schedule for turbidity exposure experiment trial 2 from Monday March 24th, 2025 – Wednesday April 23th, 2025. Columns PAM, Buoyant weight, Turbidity, Total alkalinity, Water quality, and Photo indicate processes used to assess biological condition, with an "X" indicating which day these metrics were measured during the 30-day trial.

Experiment	Date	Weekday	Day	PAM	Buoyant weight	Turbidity	Total alkalinity	Water quality	Photo
2	3/24/2025	Mon	0	X	X				X
	3/25/2025	Tue	1			X	X	X	
	3/26/2025	Wed	2			X			
	3/27/2025	Thurs	3			X			
	3/28/2025	Fri	4			X	X	X	
	3/29/2025	Sat	5			X			
	3/30/2025	Sun	6			X			
	3/31/2025	Mon	7			X	X	X	
	4/1/2025	Tue	8			X			
	4/2/2025	Wed	9			X	X	X	
	4/3/2025	Thurs	10			X			
	4/4/2025	Fri	11			X	X	X	
	4/5/2025	Sat	12			X			
	4/6/2025	Sun	13			X			
	4/7/2025	Mon	14	X	X	X	X	X	X
	4/8/2025	Tue	15			X			
	4/9/2025	Wed	16			X	X	X	
	4/10/2025	Thurs	17			X			
	4/11/2025	Fri	18			X	X	X	
	4/12/2025	Sat	19			X			
	4/13/2025	Sun	20			X			
	4/14/2025	Mon	21			X	X	X	
	4/15/2025	Tue	22			X			
	4/16/2025	Wed	23			X	X	X	
	4/17/2025	Thurs	24			X			
	4/18/2025	Fri	25			X	X	X	
	4/19/2025	Sat	26			X			
	4/20/2025	Sun	27			X			
	4/21/2025	Mon	28			X	X	X	
	4/22/2025	Tue	29			X			
	4/23/2025	Wed	30	X	X	X			X

2.5 Sediment Characterization

A cumulative 50lbs of reef sediments was collected into 5 Teflon bags from an aggregate reef site, North 3 (3N), and a cumulative 50lbs of port sediment was collected into 5 Teflon from a port site, Dania Cutoff Channel (DCC) (Figure 1). Sediments were shipped to Texas A&M University-Corpus Christi (TAMUCC) frozen in cold shipping boxes overnight via FedEx. Once received at TAMUCC, a small (~5mg) subsample from three random bags for each sediment locale were taken for heavy metal analysis. A second (~5mg) subsample was taken from one random bag from each

locale for microbial community metabarcoding. After subsamples were taken, bags were kept frozen until processing. To prepare sediment samples for experimentation, bags were thawed at room temperature. Sediment was transferred to a 15L plastic container and suspended in 30ppt artificial saltwater. For the port sediments, a slotted spoon was used to remove any floating detritus.

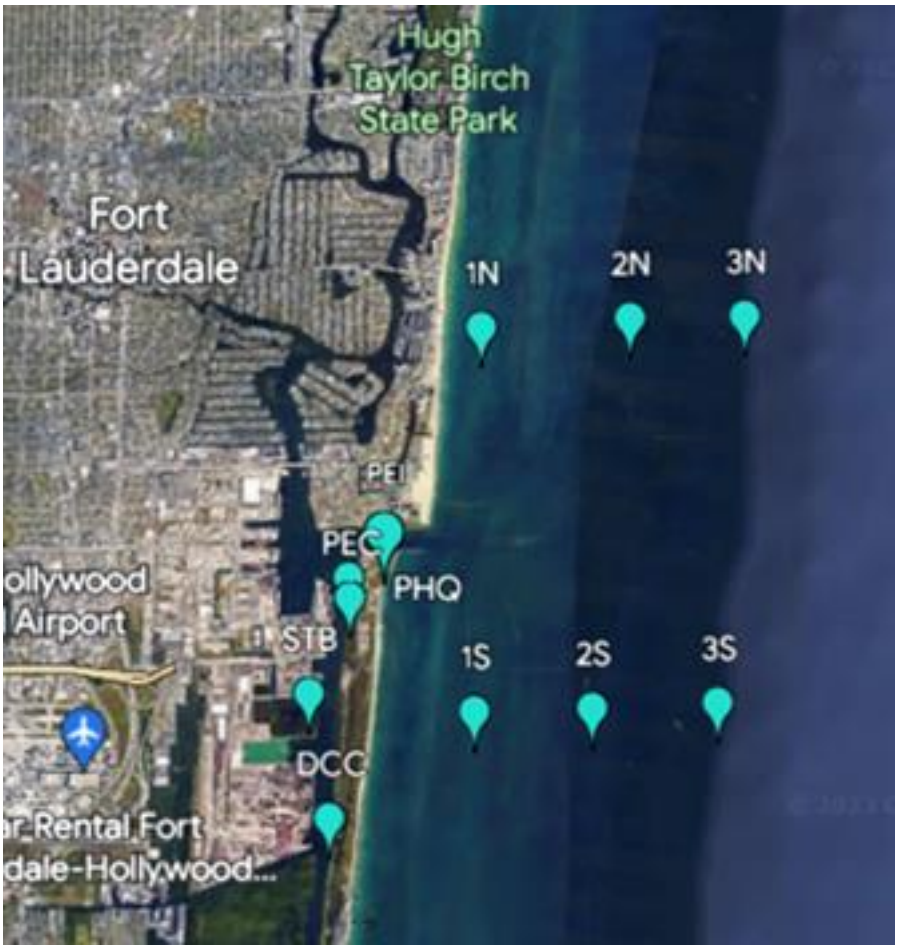

Figure 1. Sampling sites within and outside Port Everglades, Florida, with Dania Cutoff Canal (DCC) and 3N as the chosen sites for sediment sampling.

2.6 Treatment Manipulation

Post-processed sediment was homogenized and stored in 5L glass jars at -80ºC until use. A rate of loss trial was conducted to estimate turbidity loss over time (measured in NTU) across an 8-hour period. A predetermined weight (10mg) of homogenized reef and port sediment were individually added to 15L of artificial seawater at 35ppt in a 20L plastic food storage container. An overhead stirrer (LAB FISH) was added to both containers and set to 500rpm for the duration of the trial. Turbidity measurements were taken every hour from both containers to monitor turbidity loss. The values produced were graphed and fit with a line of best fit to obtain an equation describing the rate of turbidity loss for both sediment samples (Figure 2, Figure 3 respectively). These equations were then applied to estimate dosing rates to maintain experimental treatments during the 30-day stress experiments (Table 4). Two-experimental treatment levels were identified by funding managers for these experiments at 4 and 15 NTU for both port and reef sediment locales. During

experimental days 1-30, sediment was weighed and added to an 8L container and mixed with 35ppt artificial seawater and hand-dosed to the sumps to maintain turbidity targets using a sterile pipette. For the duration of the trials, treatment levels were measured every 2 hours in the sumps and chambers to assess for turbidity drift between the two locations. Dosing was implemented subsequent to measurements to ensure the sediment could homogenize in the sump before the next flush period where the live coral would be exposed to the target treatment level. Water changes were also implemented every Tuesday and Friday during the trials to maintain low microbial build up in the chamber and sump, as well as provide fresh saltwater to the organisms. New saltwater was dosed with the appropriate amount of sediment after the water changes to ensure turbidity targets were maintained throughout the experiments.

Table 4. Target NTUs and corresponding masses of sediment for turbidity treatments.

Target NTU	Seawater (L)	Port sediment (g)	Reef sediment (g)
0	15	0	0
4	15	0.09200	0.94576
15	15	0.79912	1.66160

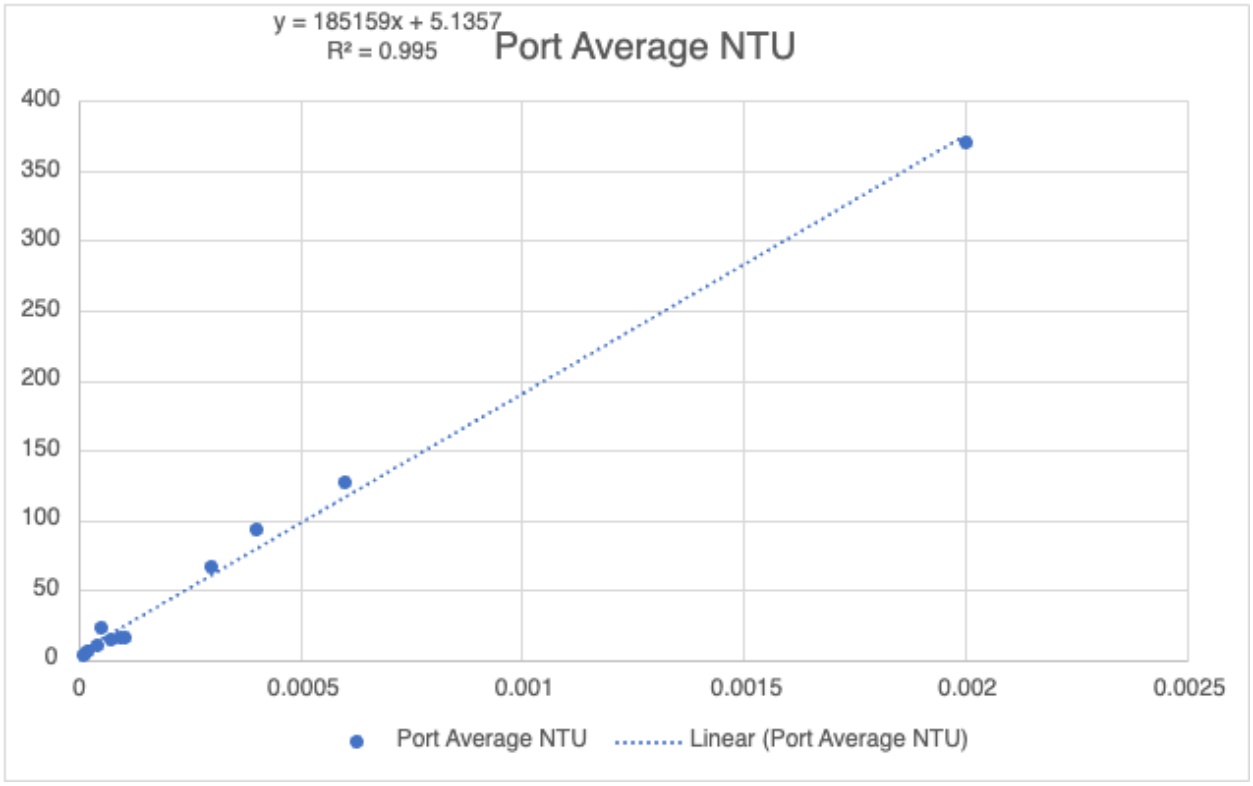

Figure 2. Average port turbidity (NTU) measured over 8 hours. A linear equation was produced by fitting a regression line to the measured data points.

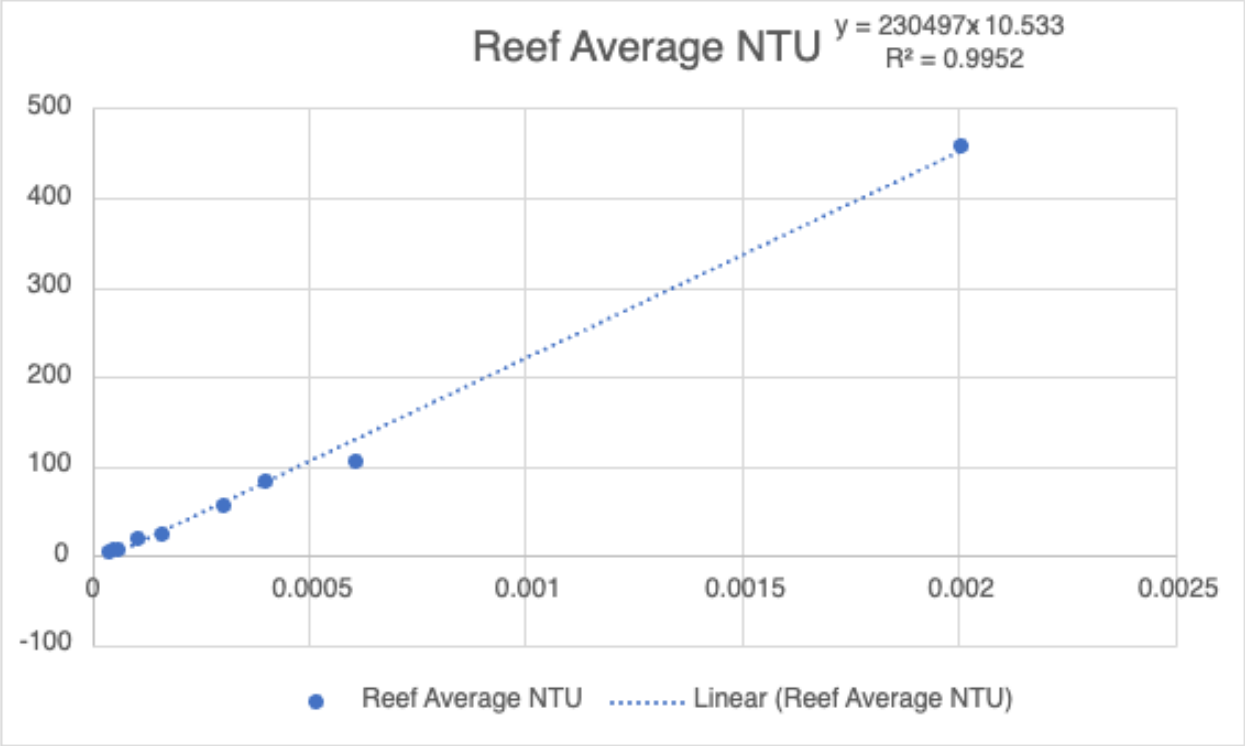

Figure 3. Average reef turbidity (NTU) was measured over 8 hours. A linear equation was produced by fitting a regression line to the measured data points.

2.7 Respirometry Chamber System

Custom-made 630 mL, 7 cm x 15 cm, cylindrical respirometry chambers (Loligo Systems, Viborg, Denmark) were connected to a 20 L sump via 10 mm tubing and a second pump line containing an oxygen sensor (Witrox 4, Loligo Systems, Viborg, Denmark) via 8 mm tubing (Figure 4). External pumps with dimensions 5.7 x 7.9 x 3.7 inches (Eheim Universal 300 Pump, Eheim GmbH & Co.KG, Deizisau, Germany) were used for both the respirometry and sump flush connections. The chambers rested on a stir plate so that a stir bar could resuspend sediments, settling within the chamber. Corals were placed on a wire pedestal at a height in the chamber so that the stir bar did not directly disturb the individual and to reduce sediment buildup on the coral plug. A single LED light (Prime 16 HD LED, Aquaillumination) was suspended above each chamber to supply light to the corals (150 - 200 mmol of photons m^{-2} s^{-1}). Intake and outflow flush tubing were secured deep in the sump water using suction cups to prevent air from disturbing the respirometry readings. The sumps consisted of a twenty-liter clear cylindrical container placed in a water bath. Each water bath was outfitted with a digital thermometer for temperature control. Two adjacent sumps fit in each water bath, allowing two systems to be placed on each rack. The room temperature was consistently 26˚C. An overhead stirrer was placed in each sump to disturb settling sediment, and an air stone was placed near the surface to supply oxygen without allowing bubbles to enter the chamber system. Air was supplied to each rack by a 4-channel air pump (95 air pump 4-way, Fedour).

Figure 4. Respirometry setup: A full sump and chamber are set up without coral (Right), and a full chamber is set up with coral exposed to treatment after a flush period (left).

2.8 Coral Metabolism

The chamber (630 mL) system was designed intermittent flow respirometry to measure metabolic oxygen consumption (MO_2 mg $h^{-1}g^{-1}$) without accumulating waste products like CO_2 (Svendsen et al., 2016). Polymer optical fiber sensors were connected to an oxygen transmitter (witrox-4) and used to collect oxygen readings every second (Loligo Autoresp 3.0). A two-point calibration was done prior to experimentation with oxygen-saturated water (100%) and 10 grams of sodium sulfite (0%). Each oxygen sensor had an associated temperature probe measuring the temperature of the sump continuously to calculate the oxygen consumption (mg L^{-1}) (Fig. 2). One complete measurement cycle consists of three timing periods: (1) Flush (5 min) - The chamber is open, allowing water to flow through the system. During this phase, a pump (Eheim 600) flushes out the chamber water into the sump while simultaneously pumping in new seawater from the sump; (2) Wait (1 min) – Flush pump turns off, allowing the newly introduced water to circulate and stabilize. The chamber is now considered closed; (3) Measure (154 min) - The chamber remains closed, and the oxygen consumption measurement takes place (Svendsen et al. 2016).

2.9 Photosynthetic Efficiency

PAM measurements were obtained (DIVING-PAM 2.0, Heinz Walz GmbH, Effeltrich, Germany), which is used to measure changes in algal symbiont activity and photosynthetic efficiency. Corals were assessed with PAM before going into the chambers, after the conclusion of an experiment, and weekly post-experiment to assess recovery. Corals were dark acclimated for at least 20 minutes before PAM measurements were taken. All corals were measured three times with PAM and averaged to account for potential variation in PAM readings across the colony. The PAR sensor was situated 5-10mm from the surface of the coral using a marked sensor cap and was not moved while a measurement was being taken. Only Fv/Fm was measured and reported, as this is the general value for photosynthetic efficiency, or “health” of algal symbionts (Ralph et al., 2015).

2.10 Calcification

The total alkalinity (TA) anomaly technique (Kinsey, 1978; Smith & Kinsey, 1978) was used to determine the net calcification rates of corals over the course of the experiment. Water samples were collected from the sump and chamber in 150 mL borosilicate glass bottles. The initial TA was collected from the sump prior to the flush. The final TA was collected from the water exiting the chamber during the flush. After collection, samples were placed in a water bath at room temperature (25ºC) and then weighed out on a scale (VWR-224AC) and run on a Metrohm Compact Sample Changer and EcoTitrator. Duplicates (w/ in 5 µmol) were run for each chamber sample and then averaged together. A pH benchtop (Thermoscientific) was used to verify the pH of each sample. Net calcification (G_{net}) in µmol $CaCO_3$ • g bwt^{-1} • h^{-1} were calculated from changes in TA (ΔTA) based on the following equation (McNicholl & Koch, 2021).

$$Gnet = -0.5\rho_w \frac{\Delta TA \cdot v}{(BW \cdot 1.54)\cdot t}$$

2.11 Post-experimental processing

After the experiment, various biological analyses were conducted on the coral fragments. These included measuring the concentration of total protein and chlorophyll (*a*, C^2, total), determining the abundance of symbiotic algae (*Symbiodinium spp.*), assessing the bulk skeletal density, and calculating the surface area of each halved coral fragment. To begin, the coral tissue was removed using an airbrush and phosphate buffer solution (PBS) using a Paasche Airbrush Co. (Kenosha, WI). The resulting mixture (25 mL) was then sonicated for twenty seconds using a sonicator ultrasonic processor (Qsonica, LLC). The sonicated slurry was divided into separate sample sets for protein, symbiont, and chlorophyll analysis. This was done by using a vortex mixer (Four E's Scientific) and a centrifuge (VWR International, LLC. Radnor, PA). The abundance of algal symbionts (*Symbiodinium spp.*) cells was determined by counting them using a hemocytometer (Bright-Line, Hausser Scientific, Horsham, PA) and a microscope at 10X magnification (ICC50W, Leica Microsystems Inc., Deerfield IL). Protein and chlorophyll absorbance was measured using a spectrophotometer (Spectromax M3, Molecular Devices, LLC., San Jose, CA), with PBS and 100% acetone as a blank, respectfully. Next, the coral skeletons were bleached (10% bleach) and then dried for four hours at 60˚C using the Drying Oven DX302C (Yomato Scientific America Inc., Santa Clara, CA). Following this, the coral skeletons were weighed using a VWR-4002B2 balance (VWR International, Radnor, PA). The skeletal density of each coral fragment was determined by dividing the dry mass of the coral, and the volume found using water displacement. Three-dimensional scans of the coral skeletons were generated and edited using the Einscan-SE 3D Scanner (Hangzhou Shining 3D Tech Co., LTD., Hangzhou, China) and MeshLab software (National Research Council and Institute of Information Sciences and Technology, Pisa, Italy). These scans were used to calculate the total surface area of each coral fragment. Finally, all the biological results obtained for the individual coral fragments were standardized to their respective surface areas. This allowed for the determination experiments of the total abundance and concentration of symbionts, chlorophyll, and protein in the corals' tissue.

2.12 Recovery

Recovery was starting one week post experimental exposure to allow for coral individuals to acclimate to housing conditions. PAM, buoyant weight, and a picture were taken for each coral on a weekly basis. Recovery was assessed and defined at the moment a coral individual's photo synthetic activity met or exceeded their baseline measurement in the pre-experimental phase.

2.13 Statistical Approach

Linear models including, but not limited to, ANOVA, ANCOVA, linear regression, and mixed-affect linear regression as well as non-linear models including logistic regression were used to assess coral responses to stressors. All destructive endpoint coral responses were calculated as a percent change (final – initial/final *100) using the pre-sacrificial corals as the initial timepoint, and all values collected were standardized to controls for each genotype. Physiological responses (metabolic oxygen (MO_2) and calcification (G_{net})), photobiological parameters (Chl-a and symbiont density), and other health metrics (Total insoluble protein and Fv/Fm) were assessed within treatment by averaging all individuals exposed to the same treatment. Treatment levels, and variation within treatment NTU were not significantly different between trials; therefore, all biological data produced during the two independent trials were analyzed together as one large data cohort.

3. RESULTS

3.1 Sediment Characterization

The port sediment (DCC) was primarily comprised of organic material (66.15%) followed by terrigenous (16.73%), carbonate (15.92%), and moisture (1.19%) while the reef sediment was primarily comprised of carbonate material (62.37%) followed by organic (23.95%), terrigenous (12.78%), and moisture (0.89%) (Table 5). Grain size analysis of the port sediment was coarse sand or organic detritus from surrounding organic matter (83.93%), with the second largest portion being coarse sand (8.29%). Detritus was floated in a super-saturated salt solution and scooped out to account for detrital material in the port sediment samples. The reef sediment was primarily gravel (68.32%), with the next largest portion medium sand (11.03%). A significant fraction of large shells and coral skeletons also accounted for the high percentage of gravel in the samples. A small percent (2.88%) of the sample was silt-sized particles (Table 6). Eight heavy metal analytes: arsenic (As), cadmium (Cd), chromium (Cr), mercury (Hg), nickel (Ni), lead (Pb), and zinc (Zn) were present in the port and reef samples. The port site exhibited higher concentrations of arsenic (As), chromium, copper, nickel, and zinc compared to the reef site. Chromium became overrepresented in the port cryomilled samples compared to the raw sediment sample whereas zinc was overrepresented in the reef raw sediment sample compared to the cryomilled sample (Figure 5). The number of raw reads recovered from sequencing for the microbiome of the sediment was highest in the manually sifted reef sediment (3N-1-250) and raw port sediment (Port-1) with the lowest number of reads in the cryomilled reef sediment (Reef-Cryo). The manually sifted port sediment (Port) yielded too low DNA concentration (< 0.5 ng/mL) for sequencing.

Table 5. Sediment Composition fractions determined by controlled burning of dried sediment samples.

Crucible	Location	Moisture	Organic	Carbonate	Terrigenous
1	Port (DCC)	1.34%	66.15%	9.53%	22.98%
2		1.19%	72.79%	7.26%	18.76%
3		1.19%	60.00%	19.46%	19.35%
4		1.16%	62.41%	22.98%	13.45%
5		1.08%	69.42%	20.38%	9.11%
	Average	1.19%	66.15%	15.92%	16.73%
6	Reef (3N)	0.89%	25.03%	63.96%	10.12%
7		0.86%	23.84%	69.01%	6.30%
8		1.00%	20.22%	57.95%	20.83%
9		0.87%	26.84%	57.23%	15.06%
10		0.84%	23.84%	63.71%	11.61%
	Average	0.89%	23.95%	62.37%	12.78%

Table 6. Relative grain size composition of each sediment collection site.

Site location	Size class	Size (μm)	Percent composition
Port (DCC)	Gravel/Organic	>2000	83.93%
	Coarse sand	500-2000	8.29%
	Medium sand	250-500	1.90%
	Fine sand	63-250	5.41%
	Silt/clay	<63	0.469%
Reef (3N)	Gravel /Organic	>2000	68.32%
	Coarse sand	500-2000	9.92%
	Medium sand	250-500	11.03%
	Fine sand	63-250	7.85%
	Silt/clay	<63	2.88%

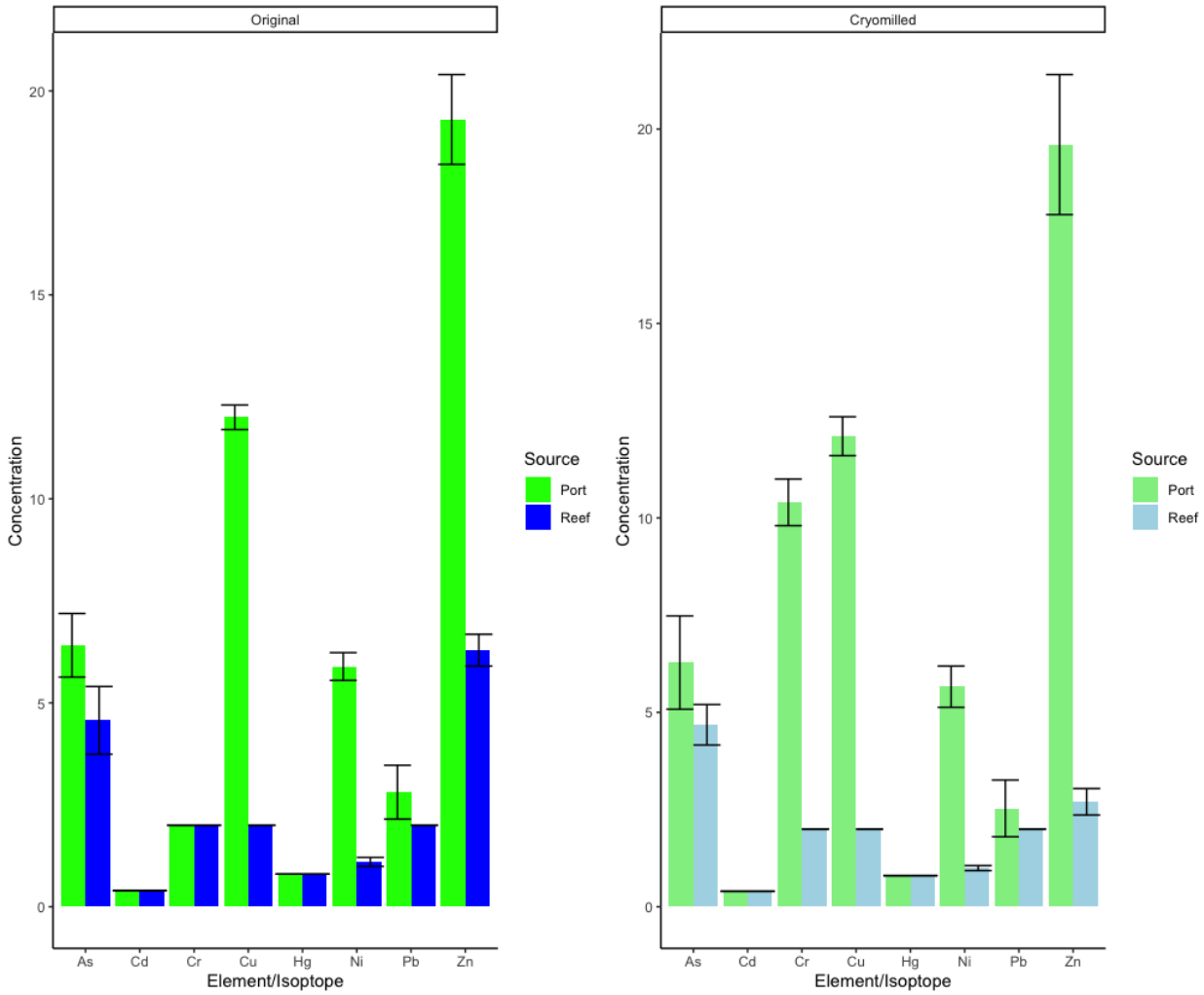

Figure 5. Comparison of heavy metal concentrations between initial and cryo-milled sediment samples from the Port and Reef sites.

Table 7. Raw reads produced via Illumina Next Generation Sequencing for each sediment sample. Sample IDs are: 3N-1-250, manually sifted reef sediment, Port-1, raw port sediment, Port-Cryo, cryomilled port sediment, Reef-1, raw reef sediment, Reef-Cryo, cryomilled reef sediment, and Port-1-250, manually sifted port sediment.

Sample ID	# Reads
3N-1-250	3,707,187
Port-1	2,066,539
Port-Cryo	1,172,073
Reef-1	1,052,999
Reef-Cryo	972,648
Port-1-250	0

3.2 Experimental Treatments

Turbidity threshold experiments: The control treatments had readings between 0 and a maximum of 4.45 NTU every time readings were taken. Turbidity readings in 15 NTU treatments were between 5 and 15 NTU 83% of the time. Turbidity readings in 29 NTU treatments were between 19 and 39 NTU 61% of the time. Finally, turbidity readings in 50 NTU treatments were between 40 and 60 NTU 46% of the time. All mean treatment NTUs were within 2 NTUs of desired levels, showing relatively low standard error. A Kruskal-Wallis test had a p-value less than $2.2x10^{-16}$, confirming a significant difference between treatments, and a Dunn's test showed that all experimental turbidity treatments had turbidities significantly different from each other (p-value less than 10^{-5} across all comparisons between treatments; Figure 4).

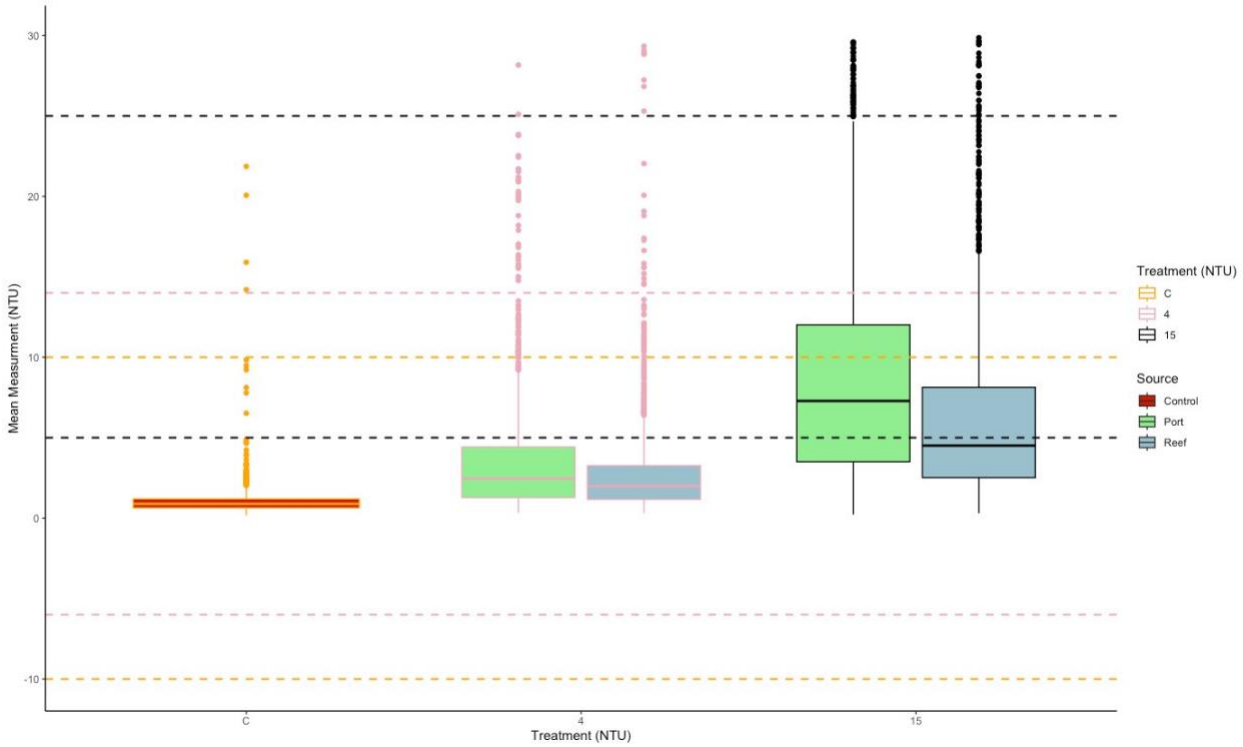

Figure 6. Average NTU for turbidity threshold maintenance over 30-days of experiment. Treatment included control (0 NTU), port sediment at 4 NTU, port sediment at 15 NTU, reef sediment at 4 NTU, and reef sediment at 15 NTU. Dotted lined indicates +/- 10 NTU error.

3.3 Respirometry (Oxygen Consumption)

There were no significant differences in oxygen consumption between treatments (p-value > 0.05; Figure 8), but trends in oxygen consumption suggests an overall decrease in oxygen production and increase in oxygen consumption across all turbidity levels. Increased oxygen consumption under turbidity stress indicates that corals might be experiencing heightened metabolic demand as a response to decreasing photosynthetic activity. This response represents a sublethal energetic cost that may compromise long-term fitness, even in the absence of visible bleaching or mortality. The oscillating nature of the oxygen consumption during the 30-day exposure period suggest the corals go through periods of acclimation and stress even though the corals were maintained at the

same treatment levels throughout the experiment's duration. While O2 consumption seemed to decrease within the first couple days of exposure, the initial increase in oxygen consumption suggests this metric is a valuable early-warning indicator of stress and a strong argument for incorporating metabolic endpoints into turbidity threshold assessments for reef management.

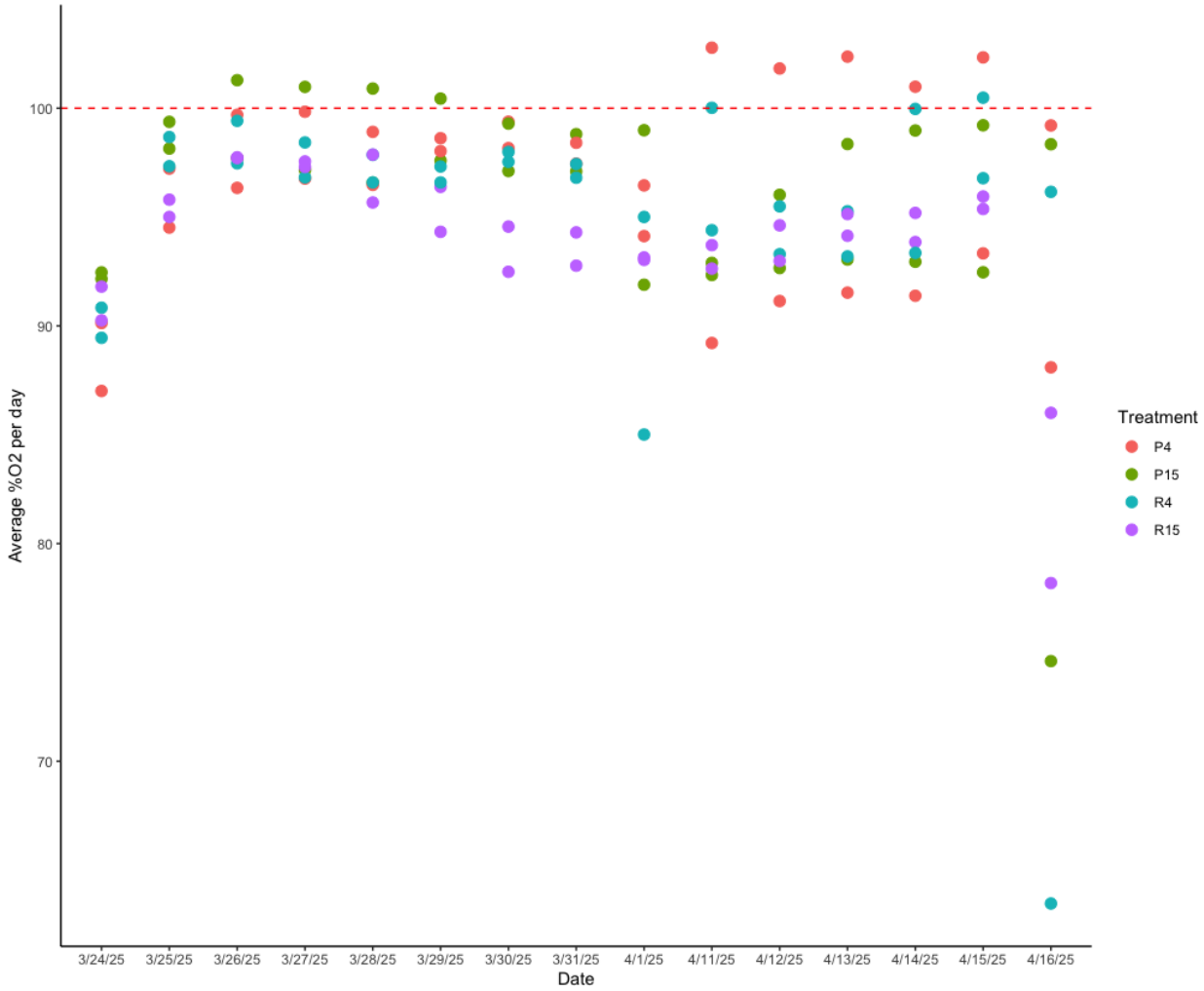

Figure 7. Scatter plot of average oxygen saturation per day across 30 days of stress exposure of *Orbicella faveolata*. Treatments included port sediment at 15 NTU (P15), port sediment at 4 NTU (P4), reef sediment at 15 NTU (R15), and reef sediment at 4 NTU (R4).

3.4 Calcification

Net calcification (G_{net}) did not significantly differ between genotypes of OFAV (p-value >> 0.05), but was highly significant between treatments (p-value << 0.05) (Figure 8). Over the 30-day exposure period, the average change in net calcification was not significantly different between Treatment when including Date as a fixed factor (p-value > 0.05). Change in buoyant weight was marginally significant between treatments (p-value = 0.091; Figure 9).

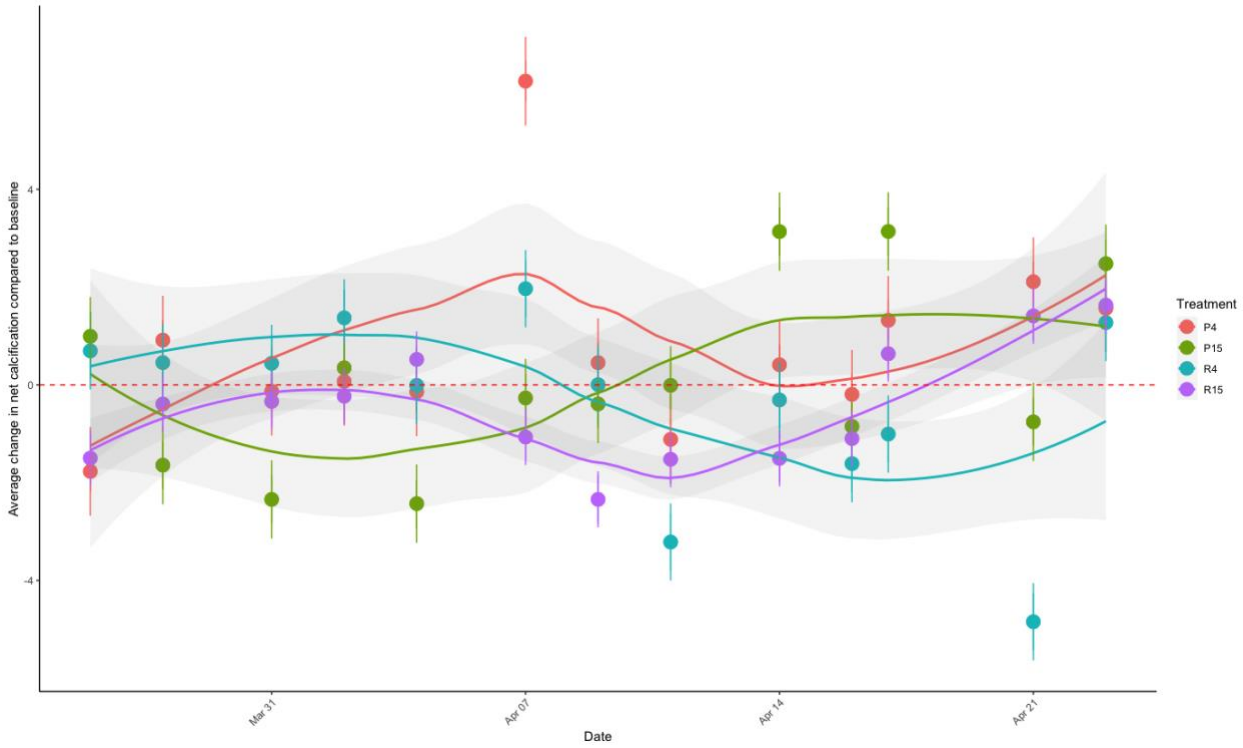

Figure 8. Time series of net calcification (G_{net}) over 30-day stress exposure of four genotypes of *Orbicella faveolata*. Treatments included port sediment at 15 NTU (P15), port sediment at 4 NTU (P4), reef sediment at 15 NTU (R15), and reef sediment at 4 NTU (R4).

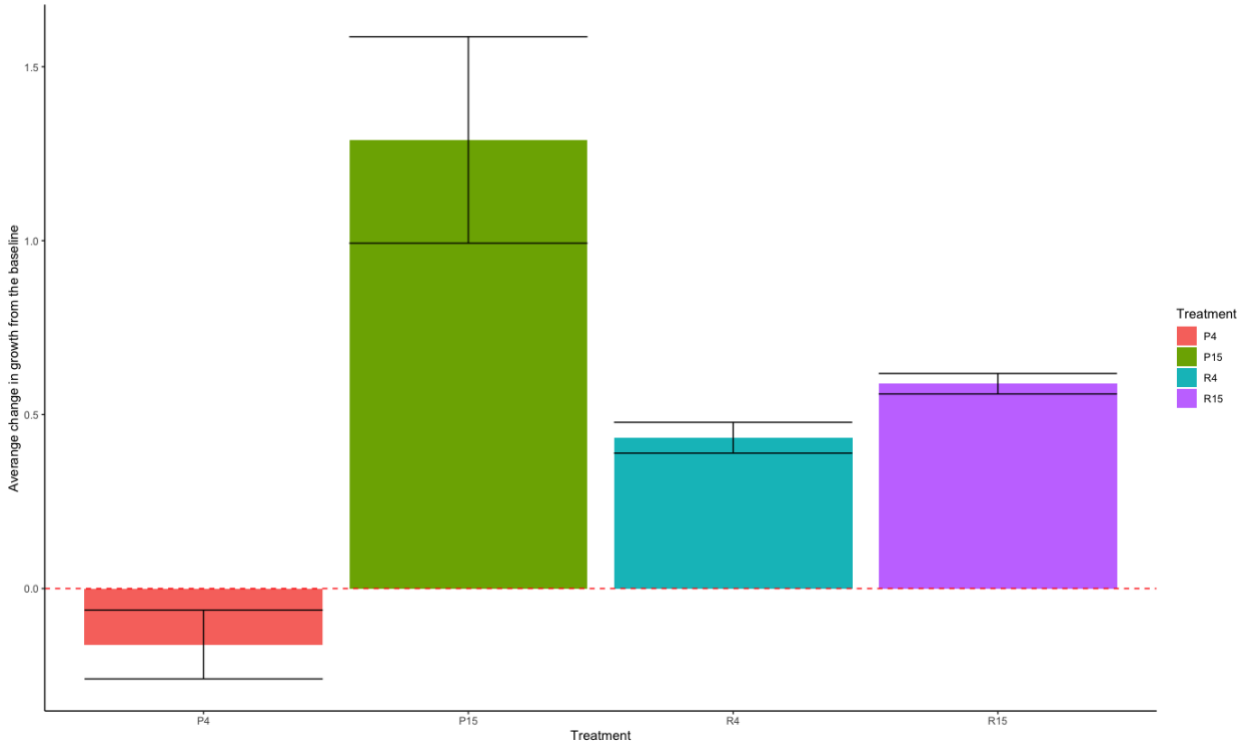

Figure 9. Average change in growth (mg • g-1 • day-1) standardized to control groups of *Orbicella faveolata* fragments exposed to four turbidity treatments. Treatments included port sediment at 15 NTU (P15), port sediment at 4 NTU (P4), reef sediment at 15 NTU (R

3.5 Photosynthetic efficiency & Recovery

Photosynthetic efficiency (Fv/Fm) was measured in triplicate for each coral individual at three time points; baseline (Pre, 0 days since the experiment started), midway (Mid, 15 days since the experiment started), and post-experimentation (Post, 30 days since the start of the experiment) (Figure 10). Coral exposed to all treatments generally decreased their Fv/Fm between the initial and midway points and then continued decreasing between the midway and post points except for corals exposed to reef 15 treatments. Corals exposed to port 4 NTU continued to decrease in Fv/Fm while corals exposed to port 15 NTU, reef 4 NTU, and reef 15 NTU increased their Fv/Fm during the recovery period. No corals have exhibited evidence of recovery at this point.

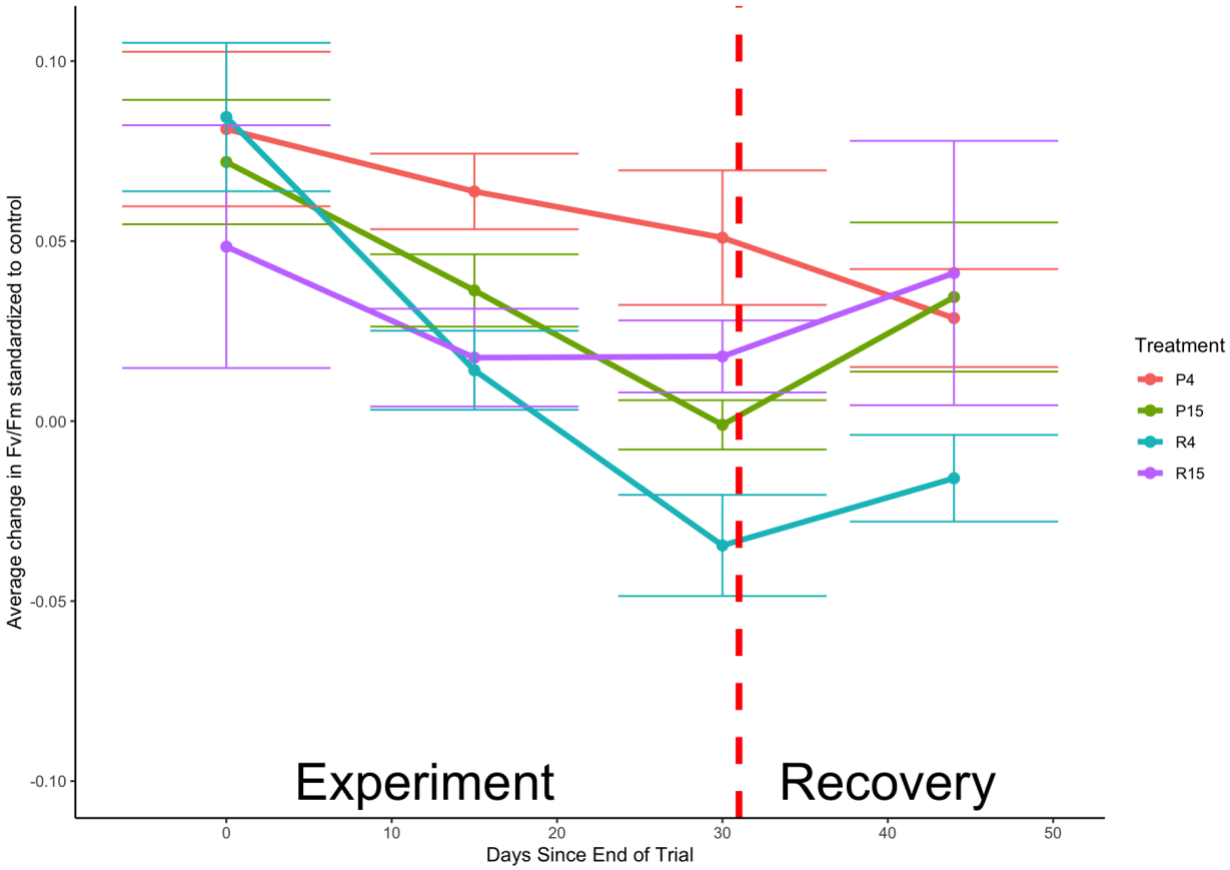

Figure 10. Photosynthetic efficiency (Fv/Fm) measured in triplicate for each coral individual at day 0, 15, and 30 during the experiment including weekly recovery measurements. Red line indicates the end of the experiment and the start of recovery period.

3.6 Biological variables

There were no significant differences among treatments for standardized change in chlorophyll a (p-value >> 0.05; Figure 11), Trends in chlorophyll concentration show varied responses to treatments within and across genotypes with chlorophyll a concentration in general being reduced in response to all treatments except for corals exposed to Reef 4 NTU treatment. Standard error shows overlap among all treatments illustrating varied responses within genotype (Figure 11). Symbiont density was shown to decrease in all corals exposed to all treatments with overlap in standard error across all treatments with no significant differences in standard percent change of symbiont density across treatments (p-value >> 0.05; Figure 13). Trends in chl-a concentrations standardized to symbiont density shows corals exposed to port sediment treatments had positive changes in chl-a per symbiont compared to a decrease in chl-a per symbiont in reef treatments with overlap in standard error bars (Figure 12). Standard change in total insoluble protein was not significantly different between treatments (p-value >>> 0.05) with a notable negative change in across all corals exposed to all treatments (Figure 14).

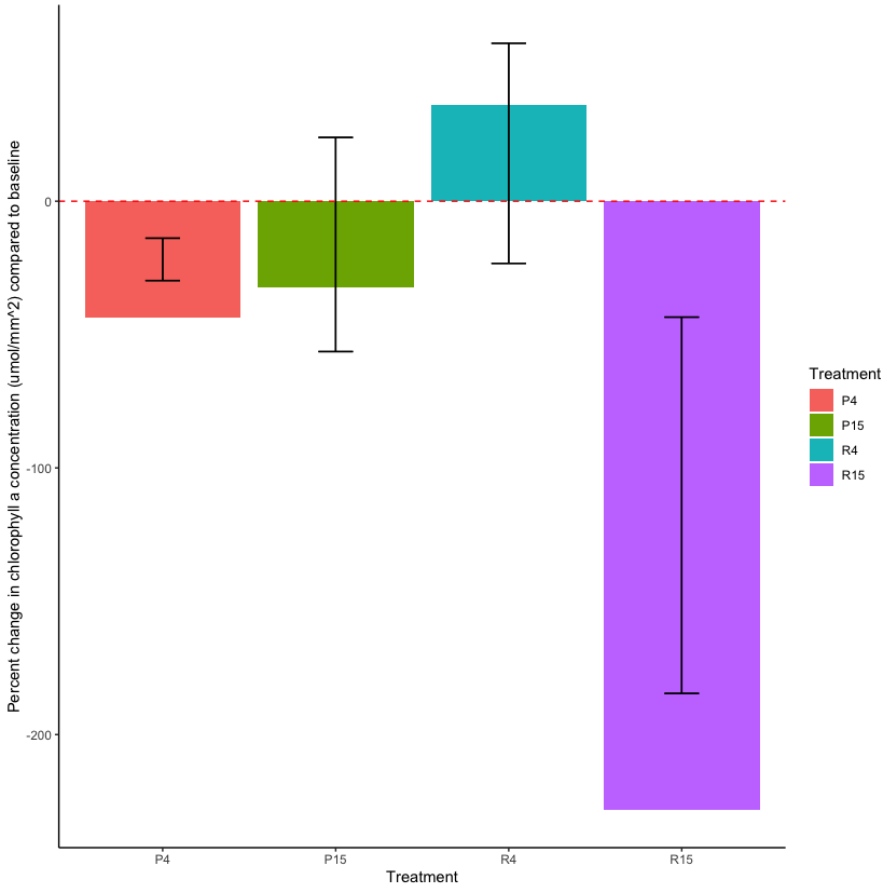

Figure 11. Average change in chl-a concentration to control treatments of *Orbicella faveolata*. Treatments included port sediment at 15 NTU (P15), port sediment at 4 NTU (P4), reef sediment at 15 NTU (R15), and reef sediment at 4 NTU (R4).

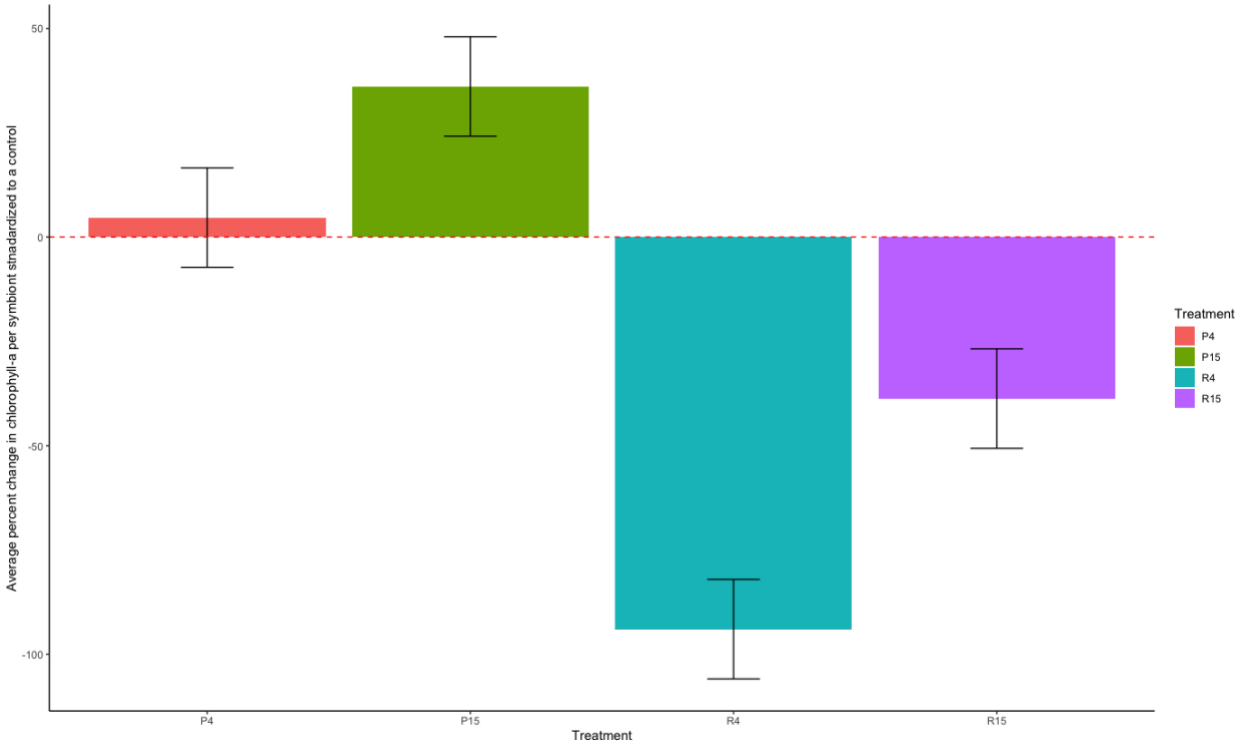

Figure 12. Average change in chl a concentration per symbiont standardized to control treatments of *Orbicella faveolata*. Treatments included port sediment at 15 NTU (P15), port sediment at 4 NTU (P4), reef sediment at 15 NTU (R15), and reef sediment at 4 NTU (R4)

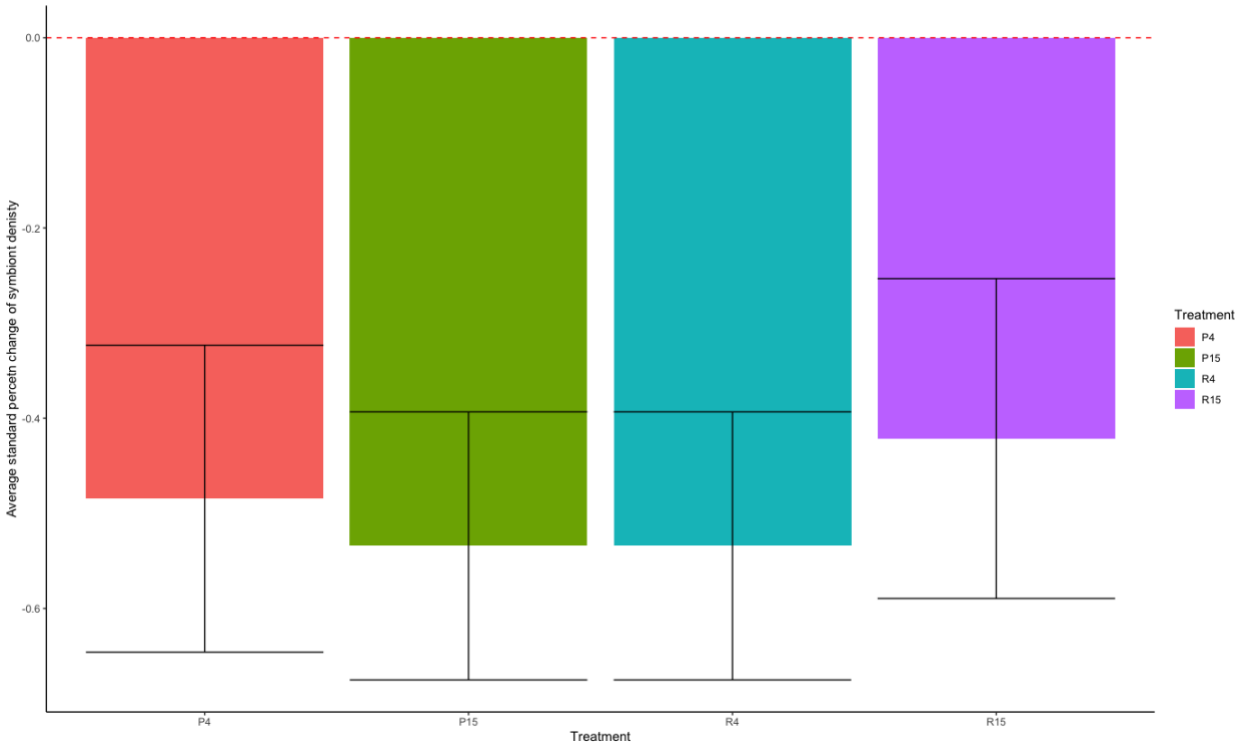

Figure 13. Average change in symbiont density standardized control treatments of *Orbicella faveolata*. Treatments included port sediment at 15 NTU (P15), port sediment at 4 NTU (P4), reef sediment at 15 NTU (R15), and reef sediment at 4 NTU (R4).

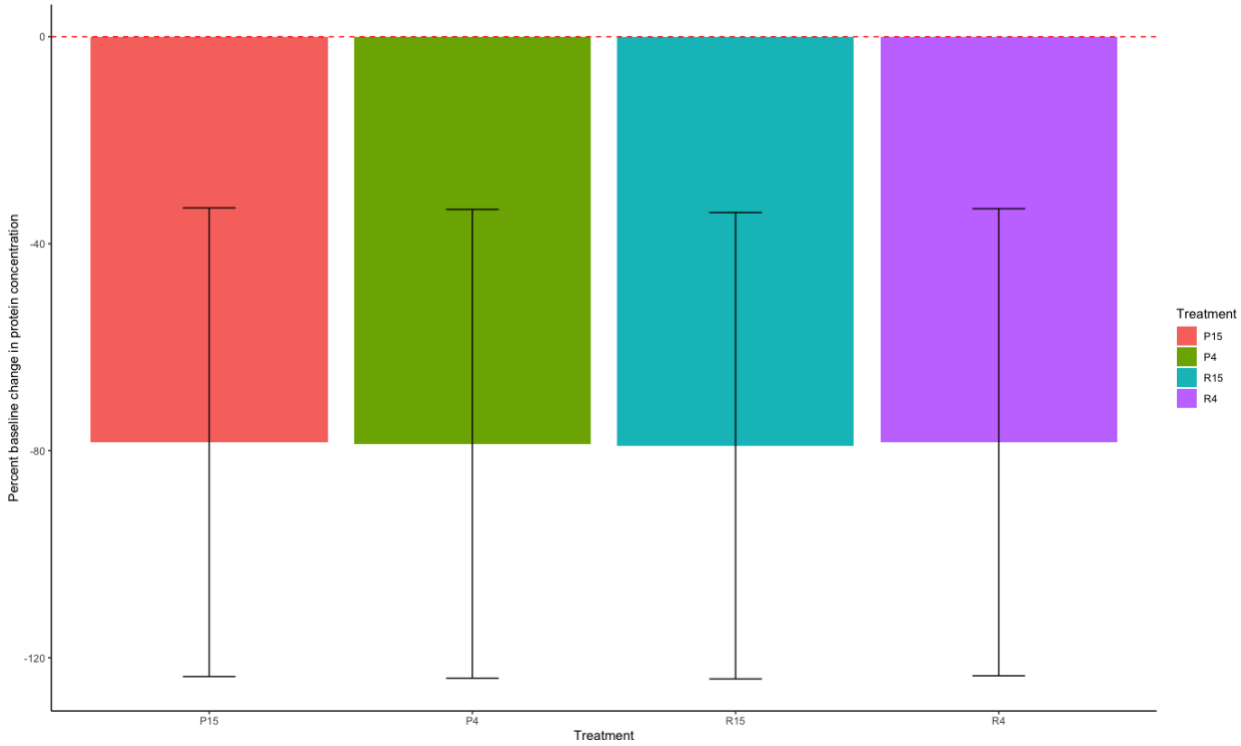

Figure 14. Average change in total insoluble protein standardized to control treatments of *Orbicella faveolata*. Treatments included port sediment at 15 NTU (P15), port sediment at 4 NTU (P4), reef sediment at 15 NTU (R15), and reef sediment at 4 NTU (R4).

3.7 Multivariate approaches

The greatest variation in the data was contributed by differences within genotype (54.5%) and was most closely associated with factors such as heavy metal concentrations (excluding cadmium) and sediment source. Whereas differences among genotypes only explained 23.1% of the variation within the data, and factors most associated with between genotype variation were Treatment NTU, cadmium heavy metal concentration, buoyant weight, average Fv/Fm, net calcification, oxygen saturation, protein concentration, symbiont density, and chlorophyll concentration (Figure 15). Redundancy analysis showed no significant differences in biological responses when tested against environmental factors (p-vale >>> 0.05; Figure 16), but RDA1 explained 99% of the variance, primarily influenced by chlorophyll concentration, which was strongly aligned with genotype 69. Genotype 3 showed higher influence from sediment source and NTU treatment, although these axes explain much less variance (1% on RDA2). Biological response (O2) and environmental predictor variables (heavy metal concentration and sediment source) showed limited association with the environmental gradient.

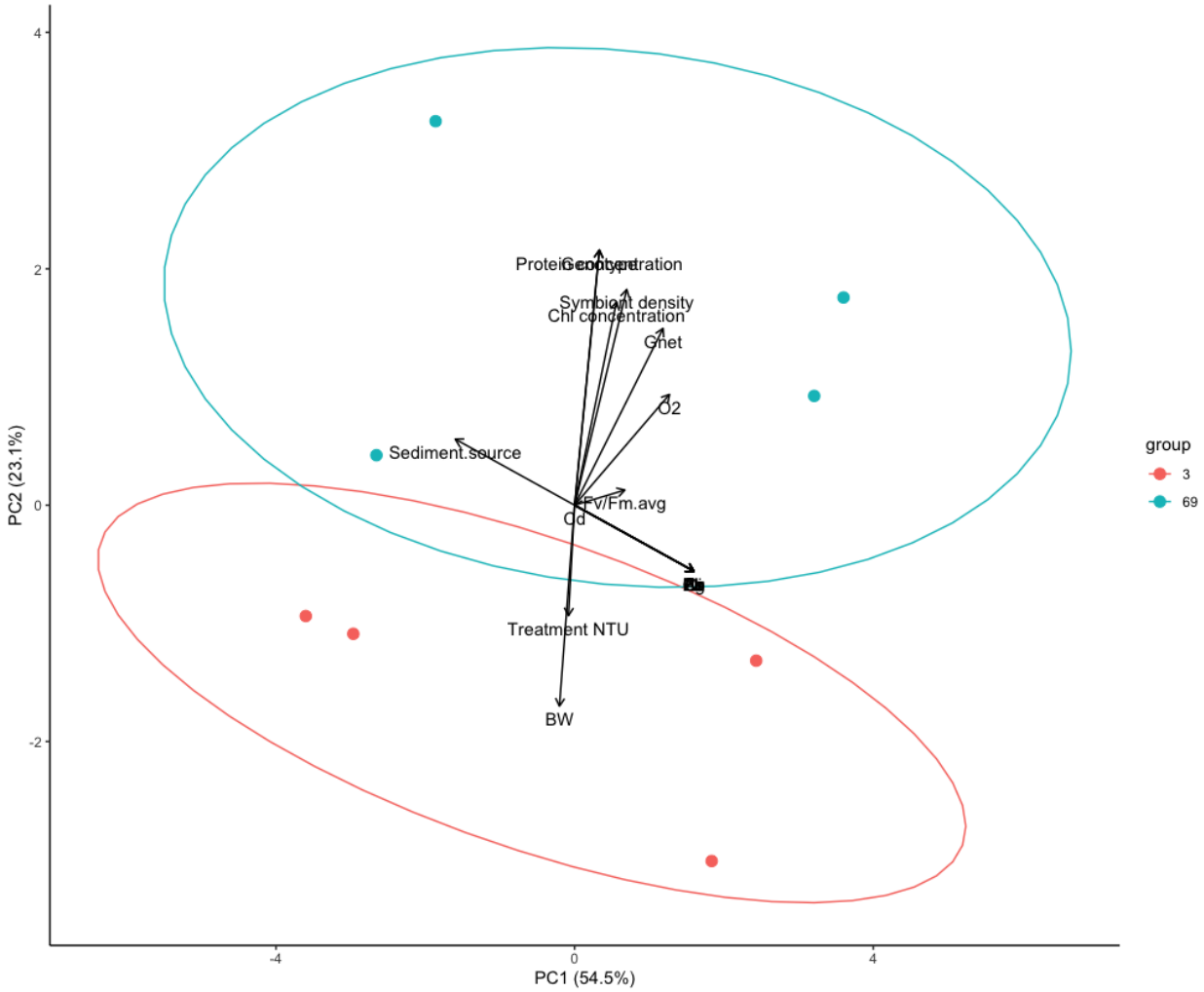

Figure 15. Unsupervised clustering with principal component analysis of biological response variables (Protein concentration, Chl-a concentration, Symbiont density, Average Fv/Fm, Percent O2 saturation, net calcification, and Buoyant weight) and environmental predictors (Treatment NTU, Sediment source, and heavy metal concentrations). The percent variation explained is included on the axes. Ellipses denote the spread of variation between groups.

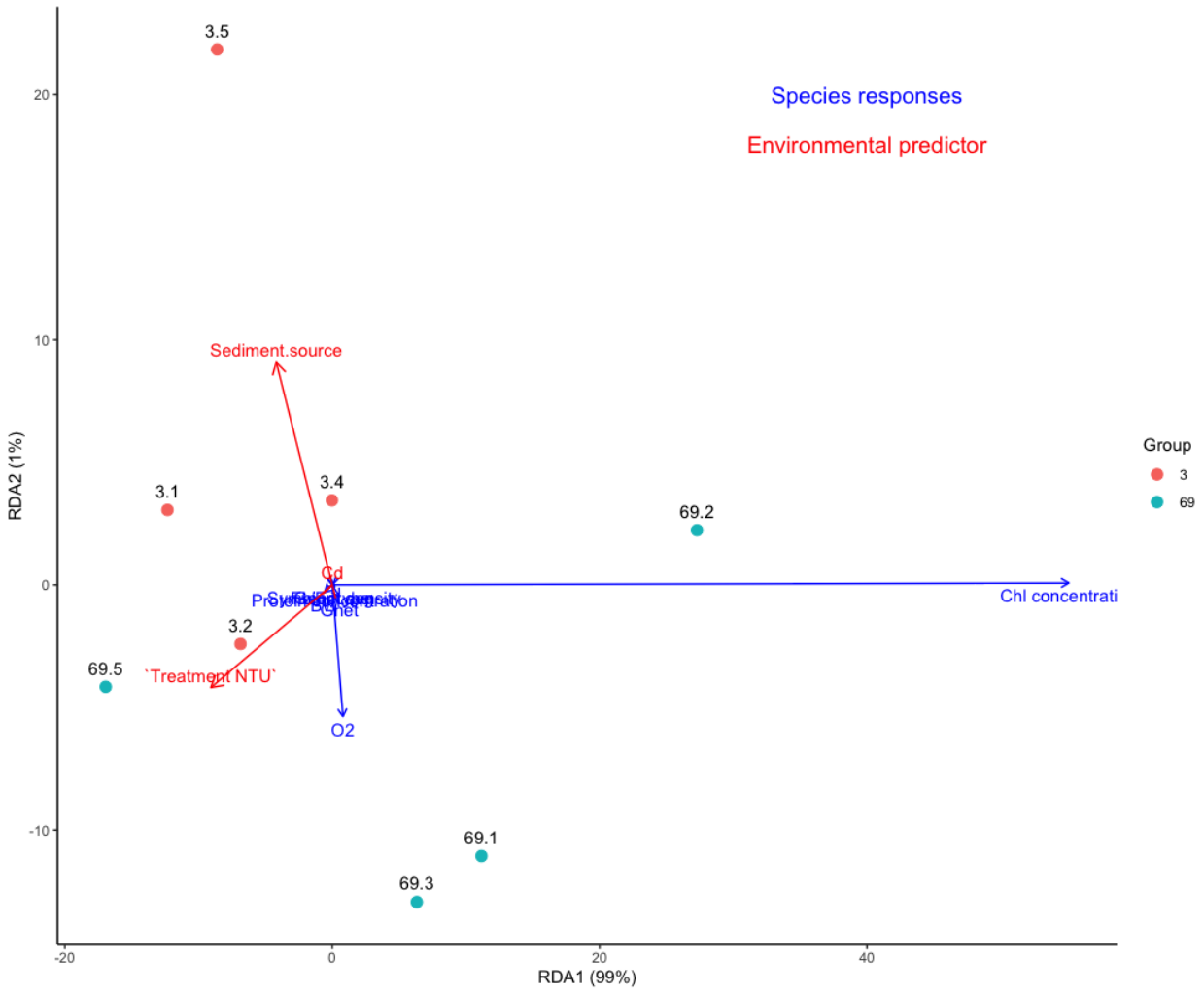

Figure 16. Redundancy analysis of biological response variables (Protein concentration, Chl-a concentration, Symbiont density, Average Fv/Fm, Percent O2 saturation, net calcification, and Buoyant weight) tested against environmental predictors (Treatment NTU, Sediment source, and heavy metal concentration. The percent variation explained for each component is included on the axes.

4. DISCUSSION

4.1 Role of Sediment Factors

Sediment stress appears to influence physiological and host/symbiont responses in a way that is detectable but subtle. This is evident where sediment source (port vs. reef) and Treatment NTU (4 & 15) seem to affect genotypic response the most, with genotype 3 aligning more with variation explained by these factors. This suggests that sediment stress may not drastically change photobiological activity (chlorophyll-a concentration) but does impact sublethal physiological parameters such as oxygen consumption. Sublethal effects like reduced symbiont density, protein content, and O_2 could indicate early signs of coral stress before outright bleaching or mortality and can be important in the context of identifying a benchmark for tolerable stress duration. While no

evidence suggested heavy metal contamination had an outright effect on coral physiology and health, the implication of heavy metal contaminates being in higher concentrations within the port sediment (arsenic, chromium, copper, lead, zinc, and nickel) does not rule out the likelihood that these heavy metals might assimilate into coral tissue during prolonged exposure, or after repeat exposures (Berry et al., 2013). This could be a focus point of future studies looking at sediment sources and heavy metal contamination.

4.2 Physiological Thresholds

Sublethal stress testing is important because it reveals early, non-lethal impacts of turbidity on corals, such as reduced growth or respiration (physiological responses) before irreversible damage or mortality occurs (Jones et al., 2020). By maintaining sublethal stress, the duration of the stress can persist, allowing for a greater exposure period for collecting physiological data. For this project, net calcification, growth, oxygen saturation, and protein concentration were used as physiological benchmarks. There were no significant differences in protein concentration changes between treatments during the duration of the experiment, but overwhelming negative changes in protein concentration suggest that the coral host is responding negatively. Responses and implications of decreased protein concentration can include tissue degradation, a shift in activity from growth and repairing tissue to survival functions, breaking down tissue for energy conservation, or symbiont loss (Lesser, 2013). While no significant differences were seen in oxygen consumption over the 30-day stress period, there were significant differences in growth response to treatment, with port treatments facilitating net calcification more compared to reef treatments. The disparity between the carbonate composition of the port and reef sediments wouldn't lead us to believe the port sediments would facilitate calcification over the reef sediment, but chl-a concentration per symbiont was greatest in the port treatments, lending to the idea that the corals exposed to port treatments calcified more because they had a greater energy budget for calcification compared to reef sediment exposed corals leading us to believe the difference in calcification performance was due to energy limitation and not substrate availability in the water. Overall, the trend in oxygen saturation leans towards a consumption-dominant system across all coral exposed to all treatments, with the lowest oxygen concentrations occurring at the beginning and end of the experiment. The oscillating nature of oxygen saturation values across the duration of the experiments could suggest coral individuals overcompensating for energy using cellular respiration when the photobiological component of the coral ecosystem is performing sub-optimally during the stressor event. Overall, the trends in growth, net calcification, oxygen consumption, and protein concentration suggest that while there might not be a physiological threshold in response to treatment for these corals that were tested, there might be a length of exposure threshold where the cumulative days of exposure to turbidity stress might be the driving factor in coral performance regardless of environmental factors (sediment source and treatment NTU), and that turbidity imposes metabolic stress on corals, likely by limiting light and reducing energy input, which in turn negatively affects coral health and physiology.

4.3 Photobiology

The corals varied responses could indicate genotypic responses to turbidity stress, where in this experiment, we see a trend in coral exposed to port sediments losing chlorophyll at a higher rate than algal symbionts, but corals exposed to reef sediment lost algal symbionts at a higher rate than chlorophyll. This could imply that corals exposed to port sediments may be selectively expelling weaker or damaged symbionts while retaining healthy, photosynthetically active ones. In contrast,

corals exposed to reef sediment are expressing dysfunctional or photodamaged symbionts, which may suggest more severe stress on the photosystem. This type of photo-physiological damage could suggest those corals were in a condition preceding bleaching. The physical aspects of light limitation can also contribute to reduced photosynthetic activity, leaning towards the actual shading of light during photolytically active periods, becoming an important contributor to these trends (Bessell-Browne et al., 2017).

4.4 Recovery

As sublethal stress duration increases, it would be amicable to expect corals to take longer to recover to their pre-stress condition (Jones et al., 2020). Using Fv/Fm (photosynthetic efficiency) in the field of coral biology is well-cited as one of the best non-destructive indicators of overall coral colony health. Using this proxy as a pre-stress baseline allows for a finite determination of recovery and allows for a better prediction of recovery timeline post-stress testing. While we did not see evidence of recovery across all corals exposed to turbidity stress, as stated above, this is understandable given the length of duration of the sublethal treatments. It is also important to note that future studies using similar systems of measurements and turbidity stress should take into consideration collecting destructive endpoint samples during the duration of the recovery period to have a better understanding of changes and adaptations in coral health metrics as a direct result of turbidity exposure. This could include heavy metal contaminant assimilation in coral host tissue, permanent damage to photo-physiological systems, or compromises in coral host tissue health and integrity (Berry et al., 2013). Overall, this study shows the importance of monitoring these corals during a recovery timeline to produce information and data that would help managers understand the timeframe required by these organisms to recover before repeat exposure to turbidity stress occurs.

5. CONCLUSION AND FUTURE DIRECTIONS

This study highlights the critical role of sublethal turbidity levels as a stressor affecting coral physiology and health dynamics. Through multivariate analyses, we observed that sediment-related variables such as turbidity and sediment source influence subtle but important shifts in coral physiological responses, including reductions in insoluble protein concentration and alterations in chlorophyll-to-symbiont density ratios. These patterns suggest that even in the absence of visible bleaching or mortality, corals experience measurable physiological stress under turbid conditions. Notably, corals that retained chlorophyll while losing symbionts at a higher rate may be exhibiting a more adaptive response, whereas those showing chlorophyll degradation in retained symbionts likely face more severe stress. These findings underscore the value of sublethal stress testing in detecting early indicators of coral health decline before irreversible damage occurs. For coastal management, particularly in regions considering dredging or sediment-disturbing activities near coral habitats, our results advocate for stricter turbidity thresholds and monitoring protocols that account for early physiological stress, not just visible degradation. Moving forward, integrate physiological response data into risk assessment models to define turbidity exposure limits that can inform permitting and dredging operations near coral reefs, incorporating sublethal endpoints into environmental assessments, and testing whether different coral species or morphologies (e.g., branching vs. bouldering) exhibit distinct

physiological or photobiological responses to turbidity will be essential to developing more effective, science-based protections for coral reefs in coastal zones.

REFERENCES

Bartley, R., Bainbridge, Z. T., Lewis, S. E., Kroon, F. J., Wilkinson, S. N., Brodie, J. E., & Silburn, D. M. (2014). Relating sediment impacts on coral reefs to watershed sources, processes and management: A review. *Science of The Total Environment*, *468–469*, 1138–1153. https://doi.org/10.1016/j.scitotenv.2013.09.030

Berry, K. L. E., Seemann, J., Dellwig, O., Struck, U., Wild, C., & Leinfelder, R. R. (2013). Sources and spatial distribution of heavy metals in scleractinian coral tissues and sediments from the Bocas del Toro Archipelago, Panama. *Environmental Monitoring and Assessment*, *185*(11), 9089–9099. https://doi.org/10.1007/s10661-013-3238-8

Bessell-Browne, P., Negri, A. P., Fisher, R., Clode, P. L., Duckworth, A., & Jones, R. (2017). Impacts of turbidity on corals: The relative importance of light limitation and suspended sediments. *Marine Pollution Bulletin*, *117*(1–2), 161–170. https://doi.org/10.1016/j.marpolbul.2017.01.050

Duckworth, A., Giofre, N., & Jones, R. (2017). Coral morphology and sedimentation. *Marine Pollution Bulletin*, *125*(1), 289–300. https://doi.org/10.1016/j.marpolbul.2017.08.036

Elliff, C. I., & Silva, I. R. (2017). Coral reefs as the first line of defense: Shoreline protection in face of climate change. *Marine Environmental Research*, *127*, 148–154. https://doi.org/10.1016/j.marenvres.2017.03.007

El-Sorogy, A. S., Mohamed, M. A., & Nour, H. E. (2012). Heavy metals contamination of the Quaternary coral reefs, Red Sea coast, Egypt. *Environmental Earth Sciences*, *67*(3), 777–785. https://doi.org/10.1007/s12665-012-1535-0

Fabricius, K. E. (2005). Effects of terrestrial runoff on the ecology of corals and coral reefs: Review and synthesis. *Marine Pollution Bulletin*, *50*(2), 125–146. https://doi.org/10.1016/j.marpolbul.2004.11.028

Giarikos, D. G., White, L., Daniels, A. M., Santos, R. G., Baldauf, P. E., & Hirons, A. C. (2023). Assessing the ecological risk of heavy metal sediment contamination from Port Everglades Florida USA. *PeerJ*, *11*, e16152. https://doi.org/10.7717/peerj.16152

Gilmour, J. (1999). Experimental investigation into the effects of suspended sediment on fertilisation, larval survival and settlement in a scleractinian coral. *Marine Biology*, *135*(3), 451–462. https://doi.org/10.1007/s002270050645

Glynn, P. W., Szmant, A. M., Corcoran, E. F., & Cofer-Shabica, S. V. (1989). Condition of coral reef cnidarians from the northern Florida reef tract: Pesticides, heavy metals, and histopathological examination. *Marine Pollution Bulletin*, *20*(11), 568–576. https://doi.org/10.1016/0025-326X(89)90359-7

Good, A. M., & Bahr, K. D. (2021). The coral conservation crisis: Interacting local and global stressors reduce reef resiliency and create challenges for conservation solutions. *SN Applied Sciences*, *3*(3), 312. https://doi.org/10.1007/s42452-021-04319-8

Guzmán, H. M., & Jiménez, C. E. (1992). Contamination of coral reefs by heavy metals along the Caribbean coast of Central America (Costa Rica and Panama). *Marine Pollution Bulletin*, *24*(11), 554–561. https://doi.org/10.1016/0025-326X(92)90708-E

Ikenaga, M., Guevara, R., Dean, A. L., Pisani, C., & Boyer, J. N. (2010). Changes in Community Structure of Sediment Bacteria Along the Florida Coastal Everglades Marsh–Mangrove–Seagrass Salinity Gradient. *Microbial Ecology*, *59*(2), 284–295. https://doi.org/10.1007/s00248-009-9572-2

Jones, R., Giofre, N., Luter, H. M., Neoh, T. L., Fisher, R., & Duckworth, A. (2020). Responses of corals to chronic turbidity. *Scientific Reports*, *10*(1), 4762. https://doi.org/10.1038/s41598-020-61712-w

Knowlton, N., Brainard, R. E., Fisher, R., Moews, M., Plaisance, L., & Caley, M. J. (2010). Coral Reef Biodiversity. In *Life in the World's Oceans* (pp. 65–78). John Wiley & Sons, Ltd. https://doi.org/10.1002/9781444325508.ch4

Kummu, M., Guillaume, J. H. A., de Moel, H., Eisner, S., Flörke, M., Porkka, M., Siebert, S., Veldkamp, T. I. E., & Ward, P. J. (2016). The world's road to water scarcity: Shortage and stress in the 20th century and pathways towards sustainability. *Scientific Reports*, *6*(1), 38495. https://doi.org/10.1038/srep38495

Lesser, M. P. (2013). Using energetic budgets to assess the effects of environmental stress on corals: Are we measuring the right things? *Coral Reefs*, *32*(1), 25–33. https://doi.org/10.1007/s00338-012-0993-x

Macdonald, D. D., Carr, R. S., Calder, F. D., Long, E. R., & Ingersoll, C. G. (1996). Development and evaluation of sediment quality guidelines for Florida coastal waters. *Ecotoxicology*, *5*(4), 253–278. https://doi.org/10.1007/BF00118995

McNicholl, C., & Koch, M. S. (2021). Irradiance, photosynthesis and elevated pCO2 effects on net calcification in tropical reef macroalgae. *Journal of Experimental Marine Biology and Ecology*, *535*, 151489. https://doi.org/10.1016/j.jembe.2020.151489

Meyer, J. L., Castellanos-Gell, J., Aeby, G. S., Häse, C. C., Ushijima, B., & Paul, V. J. (2019). Microbial Community Shifts Associated With the Ongoing Stony Coral Tissue Loss Disease Outbreak on the Florida Reef Tract. *Frontiers in Microbiology*, *10*. https://doi.org/10.3389/fmicb.2019.02244

Miller, M. W., Karazsia, J., Groves, C. E., Griffin, S., Moore, T., Wilber, P., & Gregg, K. (2016). Detecting sedimentation impacts to coral reefs resulting from dredging the Port of Miami, Florida USA. *PeerJ*, *4*, e2711. https://doi.org/10.7717/peerj.2711

Piniak, G. A. (2007). Effects of two sediment types on the fluorescence yield of two Hawaiian scleractinian corals. *Marine Environmental Research*, *64*(4), 456–468. https://doi.org/10.1016/j.marenvres.2007.04.001

Pollock, F. J., Lamb, J. B., Field, S. N., Heron, S. F., Schaffelke, B., Shedrawi, G., Bourne, D. G., & Willis, B. L. (2014). Sediment and Turbidity Associated with Offshore Dredging Increase Coral Disease Prevalence on Nearby Reefs. *PLOS ONE*, *9*(7), e102498. https://doi.org/10.1371/journal.pone.0102498

Studivan, M. S., Rossin, A. M., Rubin, E., Soderberg, N., Holstein, D. M., & Enochs, I. C. (2022). Reef Sediments Can Act As a Stony Coral Tissue Loss Disease Vector. *Frontiers in Marine Science*, *8*. https://doi.org/10.3389/fmars.2021.815698

Svendsen, M. B. S., Bushnell, P. G., & Steffensen, J. F. (2016). Design and setup of intermittent-flow respirometry system for aquatic organisms. *Journal of Fish Biology*, *88*(1), 26–50. https://doi.org/10.1111/jfb.12797

Walker, B., Gilliam, D., Dodge, R., & Walczak, J. (2012). Dredging and Shipping Impacts on Southeast Florida Coral Reefs. *Marine & Environmental Sciences Faculty Proceedings, Presentations, Speeches, Lectures*. https://nsuworks.nova.edu/occ_facpresentations/45

Weber, M., Lott, C., & Fabricius, K. E. (2006). Sedimentation stress in a scleractinian coral exposed to terrestrial and marine sediments with contrasting physical, organic and geochemical properties. *Journal of Experimental Marine Biology and Ecology*, *336*(1), 18–32. https://doi.org/10.1016/j.jembe.2006.04.007