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Lindsay A. Green-Gavrielidis  
*University of Rhode Island*, lindsaygreen@uri.edu

Fiona MacKechnie  
*University of Rhode Island*

Carol S. Thornber  
*University of Rhode Island*, thornber@uri.edu

Marta Gomez-Chiarri  
*University of Rhode Island*, gomezchi@uri.edu

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Bloom-forming macroalgae (*Ulva* spp.) inhibit the growth of co-occurring macroalgae and decrease eastern oyster larval survival

Lindsay A. Green-Gavrielidis\(^1\)*, Fiona MacKechnie\(^{1,2}\), Carol S. Thornber\(^1\), and Marta Gomez-Chiarri\(^3\)

\(^1\)Department of Natural Resources Science, University of Rhode Island, Kingston, RI 02881, U.S.A.

\(^2\)Present address: Scottish Association for Marine Science, Scottish Marine Institute, Oban, Argyll, PA37 1QA, United Kingdom

\(^3\)Department of Fisheries, Animal, and Veterinary Sciences, University of Rhode Island, Kingston, RI, 02881

*Corresponding author [lindsaygreen@uri.edu](mailto:lindsaygreen@uri.edu)

Macroalgal blooms have increased in frequency worldwide due to anthropogenic activities. Algal blooms can disrupt recreational activities, interfere with fisheries, and deplete oxygen during decomposition. Narragansett Bay has experienced macroalgal blooms dominated by blade-forming Ulva for over a century. Evidence from other systems has suggested that Ulva can negatively impact other organisms. The first objective of this study was to determine whether bloom-forming Ulva compressa and U. rigida inhibit the growth of co-occurring macroalgae, Gracilaria vermiculophylla, Cystoclonium purpureum, and Chondrus crispus, during co-culture via laboratory-based assays. We found that U. compressa and U. rigida significantly inhibited the growth of all three macroalgae. We were able to verify the negative effects of Ulva compressa, but not U. rigida on the growth of G. vermiculophylla in flow-through seawater tanks. Our second objective was to determine if Ulva exudate decreased the survival of eastern oyster larvae in laboratory challenge experiments. We documented a significant negative effect of Ulva exudate on oyster survival, which depended on both the Ulva species and the nutrient condition. The strongest effect on oyster larval survival was seen in larvae exposed to nutrient replete Ulva compressa exudate, which had less than 30% relative survival after one week. Our results indicate that bloom-forming Ulva has the potential to inhibit co-occurring macroalgae and cause oyster larval mortality.

**KEY WORDS**- Ulva compressa, Ulva rigida, macroalgal blooms, larval mortality.

**Introduction**

Macroalgal blooms, generally consisting of green ulvoid macroalgae (commonly referred to as “green tides”), have been increasing worldwide (Valiela et al. 1997, Nelson
et al. 2003, Teichberg et al. 2010, Liu et al. 2013, Smetacek & Zingone 2013) and are common occurrences on the northeastern coast of the United States (Bricker et al. 2008). Macroalgal blooms are typically driven by anthropogenic nutrient loading in shallow estuaries and can result in declines in seagrass (Valiela et al. 1997, McGlathery 2001), perennial algae, and overall community diversity (Worm & Lotze 2006). During bloom decomposition, macroinvertebrate abundance declines (Cummins et al. 2004) and dissolved organic nitrogen is released into the water column (Tyler et al. 2001) that can fuel further primary production (reviewed by Raffaelli et al. 1998). Macroalgal blooms are also costly to clean up (Atkins et al. 1993, Lapointe & Bedford 2007).

Narragansett Bay, Rhode Island, U.S.A is a 380 km² semi-diurnal, well mixed tidal estuary (Deacutis et al. 2006). The northern part of the bay is heavily populated and there are three major urban freshwater inflows that contribute anthropogenic nutrients to the system (Deacutis et al. 2006, Thornber et al. 2008). Greenwich Bay, a small sub-embayment on the western side of Narragansett Bay, has been plagued by persistent macroalgal blooms during the summer months for more than a century, dominated by Ulva compressa Linnaeus and U. rigida C. Agardh (Granger et al. 2000, Guidone et al. 2013, Thornber et al. 2017). For example, Granger et al. (2000) documented 100-400 g dry mass/m² (1015-4060 g wet mass/m² based on the conversion factors of Angell et al. 2012) of Ulva in Greenwich Bay in 1996, while Guidone & Thornber (2013) observed a maximum biomass of >1800 g wet mass/m² in 2010.

Although green macroalgal blooms can have significant deleterious impacts on coastal ecosystems (see Fletcher 1996), they have historically been considered non-toxic (Valiela & Cole 2002, Anderson 2009). Green macroalgae have been considered to be
less likely than red and brown macroalgae to inhibit or harm co-occurring organisms 
(Harlin 1987, Valiela et al. 1997), and therefore, competition between green macroalgae 
and co-occurring species has been investigated less than with red and brown macroalgae 
(Hurd et al. 2014). However, growing evidence has suggested that ulvoid species of green 
macroalgae (species in the Family Ulvophyceae) can inhibit the growth, germination, 
and/or development of co-occurring organisms (Nelson et al. 2003, Nan et al. 2008, 
Nelson & Gregg 2013, Van Alstyne et al. 2014, Van Alstyne et al. 2015; Table 1). 
Evidence of ulvoid species suppressing the growth of phytoplankton, especially species 
that cause harmful algal blooms, has been especially strong (e.g. Jin & Dong 2003, Nan 

While several researchers have reported positive and negative effects of ulvoid 
species on invertebrates (Nelson & Gregg 2013, Van Alstyne et al. 2014), to date there 
have been no reports of the potential effects of Ulva spp. on the economically important 
eastern oyster, Crassostrea virginica. Muñoz et al. (2012) showed that the presence of 
young Ulva thalli improved the post-larval growth rate of the commercially produced red 
abalone Haliotis rufescens, while Huggett et al. (2005) reported high settlement of the 
abalone H. rubra on two ulvoid species. Lamb (2015) noted that the presence of Ulva 
thalli in aquaculture bags resulted in slower growth of adult Pacific oysters, Crassostrea 
gigas. Currently there are 315 aquaculture farms that cultivate the eastern oyster C. 
virginica in the U.S. (USDA 2014), many of them in areas where Ulva is present.
Therefore, it is important to understand the interactions between bloom-forming Ulva and 
the eastern oyster.
Given the mounting evidence from other systems dominated by ulvoid macroalgae, we hypothesized that blade-forming species of *Ulva*, namely the bloom-forming *U. compressa* and *U. rigida*, inhibit the growth of co-occurring organisms in Narragansett Bay. The first objective of this study was to determine if *U. compressa* or *U. rigida* negatively affect the growth of co-occurring macroalgae. Our second objective was to determine if exudate from *U. compressa* or *U. rigida* affected the survival of eastern oyster larvae. Testing the impacts of *Ulva* exudate on oyster larvae was important for two reasons. First, *Ulva* blooms form in coastal ponds where eastern oyster populations co-occur and oyster cultivation is present (Thorne-Miller et al. 1983, Beutel 2017). Second, larvae should be included in assays because they are generally more sensitive to heavy metals and pollutants than adults (Connor 1972, His et al. 1999). We discuss our findings in light of increased coastal development and eutrophication, which will likely fuel increasing macroalgal blooms in the future.

**Materials and Methods**

**Genetic identification of *Ulva***

The genus *Ulva* contains many blade-forming species that appear morphologically similar, however, the cell shape and numbers of pyrenoids can be used to distinguish between *U. compressa* and *U. rigida* (Guidone et al. 2013). We examined each blade of *Ulva* and determined its identity based on the morphological characteristics detailed by Guidone et al. (2013). *Ulva compressa* cells are polygonal with rounded corners and contain a single pyrenoid, while *U. rigida* cells are polygonal with angular corners and contain 2-4 pyrenoids. However, *Ulva* morphology can be highly variable.
(Hofmann et al. 2010), so we also used DNA barcoding to verify the accuracy of our morphological identifications. We amplified a 678 bp segment from the rbcL gene of specimens used in these experiments following the methods of Guidone et al. (2013) except that we used a modified CTAB plant DNA extraction protocol based on Doyle and Doyle (1987). The raw sequence chromatograms were trimmed and proofread in 4Peaks (v.1.8, Nucleobytes) and sequences were aligned and assembled in Seq Man Pro (v.12, DNA Star Inc.).

**Genetic identification of Gracilaria**

Two species of *Gracilaria* occur in Narragansett Bay, the native *Gracilaria tikvahiae* McLachlan and the introduced *Gracilaria vermiculophylla* (Ohmi) Papenfuss (Nettleton et al. 2013). These two species have morphological characteristics that overlap, and therefore restricted fragment length polymorphism (RFLP) and selected DNA barcoding was performed to determine the species identification of material from the laboratory-based mesocosm trials.

DNA was extracted using the modified CTAB plant DNA extraction protocol based on Doyle and Doyle (1987). Polymerase chain reaction was performed in 50 μL volumes containing 10 μL of 5X GoTaq® Flexi DNA Polymerase (Promega Corporation), 7 μL of 25 mM Mg²⁺, 1 μL of 2.5 mM dNTP, and 4 μL of extracted DNA template (10-50 ng). A 307 bp segment from the mitochondrial gene COX1 was used for species identification and was amplified with the forward primer CO1F328 and the reverse primer CO1R634 (Nettleton et al. 2013). The PCR profile consisted of an initial denaturation at 95°C for 2 minutes, followed by 30 cycles of 57°C for 1 minute, 73°C for
1 minute, and 95°C for 1 minute followed by a final minute at 57°C and a final extension
2 at 73°C for 6 minutes. RFLP analysis was performed on the PCR samples after
3 amplification following the protocol of Nettleton (2012). In addition to the RFLP
4 analysis, three samples were chosen at random to be sequenced. PCR purification, sample
5 preparation, sequencing, and sequence analysis were performed as described above (see
6 “Genetic Identification of Ulva”).

Effects of Ulva on co-occurring macroalgae

In order to determine whether *Ulva compressa* or *U. rigida* suppress the growth of
other macroalgae, we performed a series of co-culture experiments. We co-cultured
isolated tips of three species that are common in Ulva blooms in Narragansett Bay
(Thornber, unpub. data), *Gracilaria vermiculophylla*, *Cystoclionium purpureum* (Hudson)
Batters, and *Chondrus crispus* Stackhouse from adult thalli, in separate trials, with the
bloom-forming *U. compressa* and *U. rigida*. We then conducted a series of semi-
controlled trials with *U. compressa*, *U. rigida*, and *G. vermiculophylla* in outdoor flow-
through seawater tanks.

All macroalgal material was collected in Narragansett Bay, Rhode Island during
low tide in the intertidal or shallow subtidal zone and transported to the laboratory on ice
for processing. Upon arrival at the laboratory, all material was cleaned with sterile
seawater to remove epiphytes. Following epiphyte removal, tips of *G. vermiculophylla*,
*C. purpureum*, and *C. crispus* were excised using sterile razor blades, rinsed three times
with sterile seawater (30-32 psu), and placed in 250 mL flasks with sterile Von Stosch
Enriched (VSE) natural seawater (Ott 1966) under acclimation conditions (20-23°C, 100
μmol photons m$^{-2}$ s$^{-1}$, and a 16:8 Light: Dark photoperiod with constant aeration; total acclimation to laboratory conditions occurred for at least 3 days, with at least 24 hours allowed for wound healing following tip cutting. Natural seawater was obtained from the Marine Science Research Center (MSRC) at the University of Rhode Island’s Narragansett Bay Campus, filtered to 0.2 μM, and autoclaved prior to use.

After the acclimation period, the blotted-dry wet mass of tips of *G. vermiculophylla* were taken and the tips were placed in individual 1 L mesocosms that were divided in half with mesh with 1 mm$^2$ openings and filled with 400 mL of sterile VSE seawater. On the other side of the mesh, 0.4 g of either *U. compressa* or *U. rigida* (=1 g/L) was added. Experimental culture conditions were equivalent to those provided during the acclimation period and light was supplied from the top to ensure no interspecific shading. In total, there were 21 mesocosms with seven replicates each of the *U. compressa* treatment, *U. rigida* treatment, and mesocosm control (*G. vermiculophylla* in mesh-divided mesocosm without *Ulva*) per trial. In order to prevent nutrient limitation, NO$_3^-$ was measured daily as a proxy for nutrient concentrations, and all VSE nutrients (NaNO$_3$, Na$_2$HPO$_4$·12H$_2$O, FeSO$_4$·7H$_2$O, MnCl$_2$·4H$_2$O, Na$_2$EDTA·2H$_2$O, Thiamine-HCl, Biotin, Vitamin B$_{12}$) were replenished based on nitrate depletion. Nitrate was measured using an API Nitrate Test Kit modified for a 1 mL sample. Nitrate was considered depleted if it was below 40 ppm (*i.e.* full VSE enrichment) and was replenished to this level daily. On average, we replenished nutrients in the *U. compressa* treatment every other day and the *U. rigida* treatment every 2.6 days.

On Days 2, 4, 6 and 8 of each trial, the blotted-dry wet mass of *G. vermiculophylla* tips was measured. Relative growth rate (RGR) was calculated using the
following equation: \( R_{GR} \% = 100 \times \left[ \ln \left( \frac{L_2}{L_1} \right) / (t_2 - t_1) \right] \), where \( L_2 \) and \( L_1 \) were the blade weight at times \( t_2 \) and \( t_1 \), respectively. A total of two \( G. \) *vermiculophylla* trials were performed. The same experimental design was used to conduct separate trials with \( Cystoclonium \) *purpureum* (2 trials) and \( Chondrus \) *crispus* (2 trials). Daily pH levels were determined for the first \( Chondrus \) *crispus* trial only using an EcoTestr™ pH meter (Oakton ®).

**Gracilaria vermiculophylla control trials**

To confirm that the observed results were due to the presence of *Ulva* and not simply due to the presence of another macroalga, two \( G. \) *vermiculophylla* control trials with the same experimental design as the co-culture trials described above were performed. Seven replicates each of two treatments, \( G. \) *vermiculophylla* and mesocosm control, were included in each trial. The \( G. \) *vermiculophylla* treatment had 0.4 g (=1 g/L) of \( G. \) *vermiculophylla* on one side of the mesh and a tip of \( G. \) *vermiculophylla* on the other side.

**Semi-controlled outdoor flow-through seawater tank trials**

In order to determine whether *Ulva* suppressed the growth of co-occurring macroalgae, semi-controlled trials in outdoor flow-through seawater tanks were conducted at the MSRC during July 2015 (n=4). \( G. \) *vermiculophylla* (0.85 ±0.04 g) was co-cultured with 1.5 g of either *U. compressa*, *U. rigida*, or \( G. \) *vermiculophylla* (control) in separate flow-through tanks (n=3). *Ulva compressa*, *U. rigida*, or \( G. \) *vermiculophylla* (1 g/L) were placed in individual mesocosms (16 x 11.9 x 7.62 cm) covered with mesh on
all sides (mesh size=1.6 cm²) that was connected with cable ties to a mesocosm containing *G. vermiculophylla*. The mesocosm pairs were arranged so that *Ulva* was upstream of *G. vermiculophylla*. The mass of *G. vermiculophylla* was measured on Day 0 and Day 7 and relative growth rate (RGR) was calculated. Due to space limitations, four individual trials were conducted with a single replicate from each treatment in each trial. Water temperature was measured using HOBO Tidbit v2 water temperature loggers (Onset Computer Corporation) and averaged 23.6°C (individual tanks ranged from 23.16°C ± 0.09 to 27.28°C ± 0.51; mean ± SE).

**Effects of *Ulva* on oyster larvae**

In order to determine whether exudate from *U. compressa* or *U. rigida* affected the survival of eastern oyster larvae, a series of challenge experiments were conducted. *Ulva compressa* and *U. rigida* (5 g/L) were cultured in nutrient replete (*i.e.* supplied full VSE nutrients) or nutrient deplete (*i.e.* no nutrients supplied) seawater for 2-3 days, under the same conditions outlined above, to produce *Ulva* exudate. This concentration of *Ulva* was chosen to reflect those present in *Ulva* blooms. Bloom biomass can exceed 8,000 g/m³ in the subtidal and 3,000 g/m² in the intertidal (Thornber et al. 2017). In the nutrient replete cultures, NO₃⁻ was measured daily as a proxy for nutrient concentrations, and all VSE nutrients were replenished based on nitrate depletion. However, exudate was not collected for use in the challenge experiments until all NO₃⁻ was depleted in the nutrient replete cultures, since nitrate can be toxic to juvenile and adult shellfish (Epifanio & Srna 1975).

At the end of the culture period, *Ulva* material was removed from the seawater
and the pH of exudate was adjusted to 7.9-8.0. The exudate was then filter sterilized (0.2 μM). Oyster larvae were obtained from the Blount Shellfish Hatchery at Roger Williams University and acclimated to laboratory conditions in sterile natural seawater on a shaker plate (40 rpm). Larvae were fed 2 mL/L of Shellfish Diet 1800® (Reed Mariculture, Campbell, CA) every other day while in the laboratory. At the start of the experiments oyster larvae were between 3 and 9 days old.

Challenge experiments (3 trials) were conducted in 6-well culture plates following a slight modification of previously developed protocols (Karim et al. 2013, Sohn et al. 2016). Oyster larvae (~50-100) were collected onto 45 μM nylon mesh, washed with filtered sterile seawater and placed into each well with 5 mL of the assigned treatment water. Treatments included *U. compressa* + nutrients, *U. compressa* – nutrients, *U. rigida* + nutrients, and *U. rigida* – nutrients. Each well plate contained three wells of a treatment and three wells of control (sterile seawater). Larval survival was assessed on Days 3, 5, and 7 by counting dead larvae (i.e. empty shells) in each well using an inverted microscope. At the end of the experiment, larvae were fixed by adding 70% ethanol to each well to obtain a total count. Percent survival of oyster larvae was calculated for each day using the following equation: % survival = (total - dead) ÷ total × 100. In instances where % survival was less than 0, due to human error in counting, survival was adjusted to 0% (8 out of 141 observations). The relative percent survival (of control) was calculated by randomly pairing each treatment well with a control well from the same plate using the following equation: relative percent survival = (% survival of treatment ÷ % survival of control) × 100.
Statistical Analysis

We used separate split-plot analysis of variance (ANOVA) tests to determine the effect of co-culture with *U. compressa* and *U. rigida* on the growth rate of *G. vermiculophylla*, *C. purpureum*, and *C. crispus* with treatment as the main plot (3 levels) and time as the sub-plot (4 levels); Trial (n=2) was included as a blocking factor. We also used a split-plot ANOVA to test the effect of co-culture with *G. vermiculophylla* on the growth of *G. vermiculophylla* tips (*G. vermiculophylla* control trial). We used a one-way ANOVA to test the effect of treatment (3 levels) on the growth rate of *G. vermiculophylla* in semi-controlled outdoor flow-through seawater tank trials, with Trial (n=4) included as a blocking factor. We used a two-way split-split-plot ANOVA to determine the effect of *Ulva* species (main plot, 2 levels), nutrients (sub-plot, 2 levels), and day (sub-sub plot, 3 levels) on the percent survival (of control) of oyster larvae from the challenge experiment. Trial (n=3) was used as a blocking factor.

Prior to analyses, all data were examined for normality and homogeneity of variances and transformed where appropriate; *G. vermiculophylla* growth rate was log transformed to ensure homogeneity of variances. Our growth and percent survival data did not meet the assumption of normality, even after transformation; however ANOVA is robust to deviations from normality when experiments have balanced designs and reasonable sample sizes (Underwood 1997). Post-hoc comparisons were made using Tukey’s Honestly Significant Differences tests. All statistical analyses were conducted using JMP (v.12.0.1, SAS Institute Inc.).

Results
Genetic identification of Ulva

All *U. compressa* specimens identified using morphological characteristics (n=14) in this study were verified by DNA barcoding using MegaAlign (v.12, DNA Star Inc.) to match *U. compressa* from the Northwest Atlantic (GenBank® Accession: KC582355.1). Although the holotype sequence for *U. compressa* is not currently available, our sequences agreed with the *U. compressa* concept identified by Guidone et al. (2013).

The topotype of *U. rigida* is labeled on GenBank® as *U. armorica* (Shimada et al. 2003; Guidone et al. 2013), which has since been synonymized with *U. rigida*. All *U. rigida* specimens identified using morphological characteristics in this study (n=14) were verified to match *U. rigida* from the Northwest Atlantic (GenBank® Accession: EU484395.1) and were 99% identical to the *U. rigida* topotype (GenBank® Accession: AB097630).

Genetic identification of Gracilaria

All *Gracilaria* specimens used in the laboratory co-culture experiments (n=6 for *Ulva* experiments and n=8 for *G. vermiculophylla* control) were identified through RFLP analysis as *G. vermiculophylla*. The three samples that were sequenced were identical to *G. vermiculophylla* from the Northwest Atlantic (GenBank® Accession: JQ675712.1) based on Nettleton et al. (2013).

Effects of Ulva on co-occurring macroalgae

The effect of treatment (*Ulva compressa*, *U. rigida*, and mesocosm control) on the relative growth rate of *Gracilaria vermiculophylla* was dependent on Day (Treatment x
Day: F_{6,153} = 2.2, p = 0.048; Figure 1a; Table S1). After eight days of co-culture, the RGR of *G. vermiculophylla* without *Ulva* (7.44 ± 1.35 % d⁻¹) was more than three times higher than *G. vermiculophylla* co-cultured with *U. rigida* (2.31 ± 0.69 % d⁻¹). *G. vermiculophylla* co-cultured with *U. compressa* had virtually no change in mass on Day 8 and grew significantly slower than *G. vermiculophylla* tips in the mesocosm control (p=0.004; Figure 1a).

Day significantly affected the relative growth rate of *Cystoclonium purpureum* (F_{3,151} = 8.2, p < 0.001) and was dependent on Treatment (Treatment x Day: F_{6,151} = 3.9, p = 0.001; Figure 1b; Table S2). There was no significant difference between the RGR of *C. purpureum* tips co-cultured with *U. rigida* (0.94 ± 1.28 % d⁻¹) and the mesocosm control (6.37 ± 0.96 % d⁻¹), after 8 days of co-culture. However, after 8 days of co-culture tips co-cultured with *U. compressa* grew significantly slower (-5.39 ± 1.22 % d⁻¹) than tips co-cultured with *U. rigida* (p=0.024) or alone (p<0.001; Figure 1b; Table 2).

The relative growth rate of *Chondrus crispus* was significantly affected by Treatment (*U. compressa*, *U. rigida*, and mesocosm control; F_{2,152} = 39.3, p = 0.025) with the effect of Treatment dependent on Day (Day: F_{3,151} = 8.7, p < 0.001; Treatment x Day: F_{6,151} = 2.6, p = 0.018; Figure 1c; Table S3). *C. crispus* thalli grown without *Ulva* grew significantly faster than thalli grown with *U. rigida* (p=0.025) or *U. compressa* (p=0.019) after six days of co-culture (Figure 1c; Table 3). On Day 8, *C. crispus* thalli in both *Ulva* treatments were losing mass while *C. crispus* cultured without *Ulva* was growing at a RGR of 5.7 ± 0.5 % d⁻¹ (Figure 1c, Table 3). In the first *C. crispus* trial, the pH levels were 8.1 ±0.03, 9.2±0.2, and 8.9±0.2 in the mesocosm control, *U. compressa* treatment, and *U. rigida* treatment, respectively.
There was no effect of co-culture with *G. vermiculophylla* on the relative growth rate of tips of *G. vermiculophylla* in the control trials (Treatment: $F_{1,101} = 3.2$, $p = 0.157$; data not shown). The average RGR of *G. vermiculophylla* cultured alone was $6.29 \pm 0.52$ % d$^{-1}$, while tips co-cultured with *G. vermiculophylla* had an average RGR of $3.79 \pm 0.90$ % d$^{-1}$.

The overall relative growth rate of *G. vermiculophylla* in outdoor flow-through seawater tank trials was significantly different among treatments ($F_{2,6} = 5.8$, $p = 0.0393$). There was no significant difference in the RGR of *G. vermiculophylla* between the mesocosm control ($8.5 \pm 1.7$ % d$^{-1}$) and *U. rigida* ($7.7 \pm 3.0$ % d$^{-1}$) treatments or between *U. rigida* and *U. compressa* ($3.9 \pm 0.9$ % d$^{-1}$) treatments. However, *G. vermiculophylla* in the mesocosm control grew significantly faster than *G. vermiculophylla* co-cultured with *U. compressa* ($p=0.043$).

**Effects of *Ulva* on oyster larvae**

Survival in the control oyster larvae wells was good throughout the 7-day challenge experiment. Mean survival in the controls was $97.8\% \pm 1.3\%$ on Day 3, $89.4\% \pm 2.5\%$ on Day 5 and $71.9\% \pm 3.8\%$ on Day 7 (mean $\pm$ SE, $n=48$). Relative percent survival of oyster larvae was significantly lower when larvae were cultured in exudate from *U. compressa* ($79.5 \pm 1.9\%$) than from *U. rigida* ($98.1 \pm 1.9\%$; $F_{1,2} = 47.4$, $p = 0.008$; Figure 2; Table S4). The effect of *Ulva* species on oyster larval survival was dependent on nutrients and time (*Ulva* species $\times$ nutrients $\times$ day=$F_{2,122} = 6.7$, $p = 0.002$; Table S4). Post hoc analysis revealed no difference between the treatments after 3 days of culture. However, oyster survivorship was significantly lower
when cultured in *U. compressa* + nutrients than *U. compressa* – nutrients (p<0.001) and
*U. rigida* + nutrients (p<0.001) after 5 days of culture (Figure 2). This pattern was
consistent after 7 days, when oyster survival in the *U. compressa* + nutrients treatment
was less than 30% (Figure 2).

**Discussion**

While green macroalgae have been traditionally thought of as non-toxic,
increasing evidence has shown that species of ulvoid macroalgae can inhibit co-occurring
phytoplankton (Nan et al. 2008; Tang & Gobler 2011), macroalgae (Gao et al. 2014), and
Here, we found that two dominant bloom-forming ulvoid species, *Ulva compressa* and *U.
rigida*, inhibit the growth of the co-occurring red macroalgae, *Gracilaria
vermiculophylla, Cystoclonium purpureum,* and *Chondrus crispus* at *Ulva* concentrations
that are observed during blooms (Thornber et al. 2017). Thornber et al. (2017)
documented blooms dominated by *Ulva* that reached a biomass of >3000g/m² in the
intertidal and >8000g/m³ in the subtidal zone; blooms with over 12,000 g/m³ were
recently documented in a coastal salt pond (Green-Gavrielidis et al. 2017). We were able
to validate the negative effect of *U. compressa* on the growth rate of *G. vermiculophylla*
through trials in outdoor flow-through seawater tanks.

Previous studies have reported that species of *Ulva* (e.g. *Ulva linza*; Gao et al.
2014) inhibited the growth and photosynthesis of *G. lemaneiformis* in co-culture
experiments, through a combination of chemical and nutrient competition. In our study,
we attempted to eliminate the effects of nutrient competition by replenishing nutrients
daily. It should be noted, however, that nitrate was used as a proxy for all nutrients in the seawater media and concentrations of other essential nutrients (e.g. phosphorus, trace minerals) were not measured. Despite this, we believe that nutrient limitation was unlikely since the uptake rate of nitrogen is generally several times higher than the uptake of other nutrients in macroalgae (Wallentinus 1984). Additionally, although previous studies have indicated that *Ulva* and *Gracilaria* have similar nitrogen uptake rates (Wallentinus 1984, Naldi & Wheeler 2002), we saw no negative effect on the growth rate of *G. vermiculophylla* in our control trials, which suggests that nitrogen limitation did not occur. However, we cannot completely eliminate the possibility that nutrient competition played a role in our study. Future studies should test the concentrations of all nutrients to eliminate nutrient competition as a mechanism.

We found that nutrient replete *U. compressa* caused significant mortality in oyster larvae, while nutrient deplete *Ulva* extract had no significant effect on larval mortality. Other studies have shown that bryozoan and hydroid larvae can be negatively impacted by brown algae (Schmitt et al. 1998), red algae can cause necrosis in soft corals (de Nys et al. 1991), and green algae can negatively affect the development of Pacific oyster larvae (Nelson & Gregg 2013), growth rate of adult Pacific oysters (Nelson et al. 2003, Nelson & Gregg 2013, Van Alstyne et al. 2014), and metamorphosis of crab larvae (Van Alstyne et al. 2014). Interestingly, several studies have reported that the toxicity of phytoplankton increased under nutrient limitation. For example, the haptophyte *Prymesium parvum* causes significant mortality in other phytoplankton species and the toxicity of *P. parvum* was enhanced under nutrient limited conditions (Granéli & Johansson 2003, Uronen et al. 2005, reviewed by Granéli et al. 2008). Ribalet et al.
(2007) reported that production of toxic polyunsaturated aldehydes (PUAs) by marine diatoms increased under nutrient limitation. Our results indicate that *U. rigida* grown under nutrient deplete conditions had a stronger negative effect on oyster larval survival, although this trend was not statistically significant. Contrastingly, Nan et al. (2008) showed that *Ulva lactuca* caused mortality in microalgae under nutrient replete conditions, similar to our findings for *U. compressa*.

Previous researchers have also demonstrated that the effect of macroalgae on co-occurring species is dependent on species-specific characteristics. For example, Accoroni et al. (2015) showed that co-culture with fresh thalli of the brown alga *Dictyota dichotoma* had a stronger negative effect on the growth of the benthic diatom *Ostreopsis cf. ovata* than co-culture with *U. rigida*. There are also species-specific effects within the Ulvales. Nelson et al. (2003) showed that extract from *U. obscura* more strongly inhibited the germination of *Fucus gardneri* than extract from *U. fenestrata*.

In our laboratory-based mesocosms studies, both *U. compressa* and *U. rigida* inhibited the growth of *G. vermiculophylla* and there was no significant difference in the growth of *G. vermiculophylla* between the *Ulva* treatments. However, our results from outdoor flow-through seawater tank trials showed that only *U. compressa* significantly suppressed the growth of *G. vermiculophylla*. Although there was a trend of reduced *G. vermico...phylla growth in the *U. rigida* treatment, there was no significant effect of co-culture with *U. rigida*, likely due to low replication. Furthermore, we documented consistent, contrasting responses of oyster larvae to exudate of *U. compressa* and *U. rigida*. Therefore, we hypothesize that the mechanisms responsible for the negative effects of *U. compressa* and *U. rigida* on co-occurring organisms are species specific.
The three co-occurring macroalgae tested here also responded differently to *U. compressa* and *U. rigida*. For example, the native *C. purpureum* began to lose mass or show negligible growth in the presence of *U. compressa* and *U. rigida* after 4 and 6 days of co-culture, respectively. *Chondrus crispus* grown in the presence of both *Ulva* species had lower growth rates, but only began to lose mass after 8 days of co-culture with *U. compressa*. Interestingly, the non-native *G. vermiculophylla* appeared to be the least affected of the three macroalgae tested; *G. vermiculophylla* did not experience a significant reduction in growth rate in either *Ulva* treatment until 8 days of co-culture had passed and never began to lose mass. Differences in the response of species to ulvoids have also been documented in phytoplankton (Tang & Gobler 2011) and could have ecological consequences for species presence and abundance in or near macroalgal blooms. In Narragansett Bay, *Gracilaria* spp. is very common in blooms (Thornber et al. 2017), perhaps owing to its ability to coexist with *U. compressa* and *U. rigida*. Species interactions shape ecological communities and the impacts of *U. compressa* and *U. rigida* on community composition require further research.

The responses documented here could be the result of allelopathy (*i.e.* chemical inhibition) by *U. compressa* and *U. rigida*. However, identifying chemically mediated interactions depends on detection of chemicals at or near the alga surface (Steinberg and de Nys 2002). Therefore, this hypothesis cannot be validated until allelochemicals are detected, isolated, and identified from *U. compressa* and *U. rigida*, and the effect of those isolated allelochemicals on target species is tested. Furthermore, it is important to note that *Ulva* can also compete with co-occurring macroalgae through other mechanisms such as nutrient competition (discussed above) or through pH alteration. For example,
Ulva intestinalis has been shown to raise the pH of rockpools to a level (>10) where seaweeds cannot utilize external carbonic anhydrase (CA) to covert HCO$_3^-$ to CO$_2$ for use in photosynthesis, and therefore become carbon limited (Bjork et al. 2004). Chondrus crispus utilizes HCO$_3^-$ only through external CA and becomes bleached when growing in rockpools dominated by U. intestinalis due to high pH (Bjork et al. 2004). Although pH was only measured in the first Chondrus crispus trial, we did document pH levels that were potentially high enough to interrupt external CA activity. Alterations in pH, however, cannot explain all of the results documented here. In particular, research has shown that species of Gracilaria and closely related Gracilariopsis use both external CA (sensitive to high pH) and a direct HCO$_3^-$ transporter (not sensitive to pH) simultaneously to take up inorganic carbon (Andría et al. 1999, Pérez-Lloréns et al. 2004), yet we documented a negative effect of U. compressa on the relative growth rate of Gracilaria vermiculophylla in closed mesocosms. Additionally, in the oyster larval survival assays, we adjusted the pH of the Ulva exudate to match control seawater (7.9-8.0) prior to use. If pH were responsible for the negative effects of Ulva on oyster larvae, we should have seen no difference in the survival between treatments.

We have demonstrated for the first time that U. compressa has a significant negative effect on the survival of eastern oyster larvae, an important aquaculture crop in the U.S. (USDA 2014), when cultured under eutrophic conditions. Approximately two-thirds of U.S. coastal waterways, including Narragansett Bay, are considered degraded by an excess of nitrogen (N) from anthropogenic influences (Howarth & Marino 2006). Excess nutrients are known to cause blooms of ulvoid macroalgae (Teichberg et al. 2010) and our results suggest that U. compressa can cause mortality in oyster larvae in these
systems especially when oyster spawning coincides with the occurrence of Ulva blooms. Interestingly, U. rigida did not cause significant mortality of oyster larvae, although it did inhibit the growth of co-occurring macroalgae. One important caveat of this study is the lack of validation of these effects in situ. While we did use ecologically relevant concentrations of Ulva in our study, these results are likely to change as a result of hydrodynamics (Steinberg et al. 2002). Further research is required to examine the effects of Ulva on other economically important bivalves (e.g. clams and scallops) and on post-larval eastern oysters and to verify these effects in situ.

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We would like to thank I. Burns, K. Carpentier, L. Dansereau, and C. Anderson for laboratory and/or logistical help. Funding was provided by the Rhode Island Science and Technology Council and the National Science Foundation EPSCoR Cooperative Agreement #EPS-1004057. This research is based in part upon work conducted using the Rhode Island Genomics and Sequencing Center and the Marine Life Sciences Facility. We are thankful to three anonymous reviewers for their insightful feedback that greatly improved this manuscript.

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Figure Legend

Figure 1. Mean relative growth rate (% d$^{-1}$) of a) Gracilaria vermiculophylla b) Cystoclonium purpureum and c) Chondrus crispus co-cultured with U. compressa, U. rigida, or alone (control). Error bars represent ±1SE. Asterisks (*) denote a statistically significant difference based on Tukey’s HSD post hoc comparisons. The results of posthoc comparisons for C. purpureum and C. crispus are available in Table 2 and 3, respectively.

Figure 2. Relative percent survival of oyster larvae exposed to exudate from U. compressa and U. rigida grown under nutrient replete (+ Nutrients) or nutrient deplete (- Nutrients) conditions. Bars with a letter in common are not statistically different based on Tukey’s HSD post hoc comparisons. Error bars represent ±1SE.
Figure 1.

(a) U. compressa, U. rigida, Control

(b) Mean G. vulpinus RGR (% d⁻¹)

(c) Mean C. purpureum RGR (% d⁻¹)

Day
Figure 2.
Table 1: Selected examples of studies documenting the effects of ulvoid macroalgae (Family Ulvophyceae) on co-occurring organisms.

<table>
<thead>
<tr>
<th>Location</th>
<th>Macroalgal Taxa</th>
<th>Documented Effects</th>
<th>Reference(s)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Sirolo, Italy</td>
<td><em>U. rigida</em></td>
<td>Inhibited growth of toxic benthic dinoflagellate</td>
<td>Accoroni et al. 2015</td>
</tr>
</tbody>
</table>
Table 2. Mean relative growth rate (RGR; % d⁻¹) of *Cystoclonium purpureum* tips cultured with *U. compressa, U. rigida*, or alone (mesocosm control). Means without a common superscript letter differ significantly (p<0.05) based on Tukey’s HSD post hoc comparisons.

<table>
<thead>
<tr>
<th>Days of Co-culture</th>
<th>Treatment</th>
<th>RGR (% d⁻¹) ±1SE</th>
</tr>
</thead>
<tbody>
<tr>
<td>2</td>
<td><em>U. compressa</em></td>
<td>4.89 ± 0.70abc</td>
</tr>
<tr>
<td></td>
<td><em>U. rigida</em></td>
<td>4.58 ± 1.08abc</td>
</tr>
<tr>
<td></td>
<td>Mesocosm control</td>
<td>5.38 ± 1.29ab</td>
</tr>
<tr>
<td>4</td>
<td><em>U. compressa</em></td>
<td>-0.60 ± 1.25cde</td>
</tr>
<tr>
<td></td>
<td><em>U. rigida</em></td>
<td>2.31 ± 1.02abc</td>
</tr>
<tr>
<td></td>
<td>Mesocosm control</td>
<td>5.42 ± 1.21ab</td>
</tr>
<tr>
<td>6</td>
<td><em>U. compressa</em></td>
<td>-4.40 ± 1.89de</td>
</tr>
<tr>
<td></td>
<td><em>U. rigida</em></td>
<td>0.22 ± 2.01bcd</td>
</tr>
<tr>
<td></td>
<td>Mesocosm control</td>
<td>5.36 ± 1.03ab</td>
</tr>
<tr>
<td>8</td>
<td><em>U. compressa</em></td>
<td>-5.39 ± 1.22c</td>
</tr>
<tr>
<td></td>
<td><em>U. rigida</em></td>
<td>0.94 ± 1.28abcd</td>
</tr>
<tr>
<td></td>
<td>Mesocosm control</td>
<td>6.37 ± 0.96a</td>
</tr>
</tbody>
</table>

Table 3. Mean relative growth rate (RGR; % day⁻¹) of *Chondrus crispus* tips cultured with *U. compressa, U. rigida*, or alone (mesocosm control). Means without a common superscript letter differ significantly (p<0.05) based on Tukey’s HSD post hoc comparisons.

<table>
<thead>
<tr>
<th>Days of Co-culture</th>
<th>Treatment</th>
<th>RGR (mg d⁻¹) ±1SE</th>
</tr>
</thead>
<tbody>
<tr>
<td>2</td>
<td><em>U. compressa</em></td>
<td>4.46 ± 0.82abc</td>
</tr>
<tr>
<td></td>
<td><em>U. rigida</em></td>
<td>3.76 ± 0.54abc</td>
</tr>
<tr>
<td></td>
<td>Mesocosm control</td>
<td>6.76 ± 0.50a</td>
</tr>
<tr>
<td>4</td>
<td><em>U. compressa</em></td>
<td>1.68 ± 0.72cd</td>
</tr>
<tr>
<td></td>
<td><em>U. rigida</em></td>
<td>1.50 ± 0.65cd</td>
</tr>
<tr>
<td></td>
<td>Mesocosm control</td>
<td>3.84 ± 0.98abc</td>
</tr>
<tr>
<td>6</td>
<td><em>U. compressa</em></td>
<td>1.34 ± 0.47cd</td>
</tr>
<tr>
<td></td>
<td><em>U. rigida</em></td>
<td>1.44 ± 0.62cd</td>
</tr>
<tr>
<td></td>
<td>Mesocosm control</td>
<td>5.37 ± 0.32ab</td>
</tr>
<tr>
<td>8</td>
<td><em>U. compressa</em></td>
<td>-1.52 ± 1.87d</td>
</tr>
<tr>
<td></td>
<td><em>U. rigida</em></td>
<td>1.91 ± 0.60bcd</td>
</tr>
<tr>
<td></td>
<td>Mesocosm control</td>
<td>5.73 ± 0.50a</td>
</tr>
</tbody>
</table>