



**HAL**  
open science

## Effect of CO<sub>2</sub>-induced ocean acidification on the early development and shell mineralization of the European abalone (*Haliotis tuberculata*)

Nathalie Wessel, Sophie Martin, Aïcha Badou, Philippe Dubois, Sylvain Huchette, Vivien Julia, Flavia Nunes, Ewan Harney, Christine Paillard, Stéphanie Auzoux-Bordenave

### ► To cite this version:

Nathalie Wessel, Sophie Martin, Aïcha Badou, Philippe Dubois, Sylvain Huchette, et al.. Effect of CO<sub>2</sub>-induced ocean acidification on the early development and shell mineralization of the European abalone (*Haliotis tuberculata*). *Journal of Experimental Marine Biology and Ecology*, 2018, 508, pp.52 - 63. 10.1016/j.jembe.2018.08.005 . hal-01912239

**HAL Id: hal-01912239**

**<https://hal.sorbonne-universite.fr/hal-01912239>**

Submitted on 5 Nov 2018

**HAL** is a multi-disciplinary open access archive for the deposit and dissemination of scientific research documents, whether they are published or not. The documents may come from teaching and research institutions in France or abroad, or from public or private research centers.

L'archive ouverte pluridisciplinaire **HAL**, est destinée au dépôt et à la diffusion de documents scientifiques de niveau recherche, publiés ou non, émanant des établissements d'enseignement et de recherche français ou étrangers, des laboratoires publics ou privés.

1 **Effect of CO<sub>2</sub>-induced ocean acidification on the early development**  
2 **and shell mineralization of the European abalone (*Haliotis***  
3 ***tuberculata*)**

---

4 Nathalie Wessel<sup>1,2</sup>, Sophie Martin<sup>3,8</sup>, Aïcha Badou<sup>1</sup>, Philippe Dubois<sup>4</sup>, Sylvain  
5 Huchette<sup>5</sup>, Vivien Julia<sup>1,6</sup>, Flavia Nunes<sup>6,7</sup>, Ewan Harney<sup>6</sup>, Christine Paillard<sup>6</sup> and  
6 Stéphanie Auzoux-Bordenave<sup>1,8\*</sup>

7 <sup>1</sup>UMR 7208 "Biologie des Organismes et Ecosystèmes Aquatiques" (BOREA),  
8 MNHN/CNRS/IRD/UPMC, Muséum national d'Histoire naturelle, Station de Biologie Marine de  
9 Concarneau, 29900 Concarneau, France

10 <sup>2</sup> Current address : Ifremer, Département Océanographie et Dynamique des Ecosystèmes (ODE), Rue de  
11 l'île d'Yeu, BP21105, 44311 Nantes Cedex 3, France

12 <sup>3</sup>UMR 7144 "Adaptation et Diversité en Milieu Marin" (AD2M), Station Biologique de Roscoff,  
13 29680 Roscoff Cedex, France

14 <sup>4</sup>Laboratoire de Biologie Marine, Université Libre de Bruxelles, CP160/15, 1050, Brussels, Belgium

15 <sup>5</sup> Ecloserie France-Haliotis, Kerazan, 29880 Plouguerneau, France

16 <sup>6</sup> Laboratoire des Sciences de l'Environnement Marin (LEMAR), UMR 6539  
17 CNRS/UBO/IRD/Ifremer, Institut Universitaire Européen de la Mer, University of Brest (UBO),  
18 Université Européenne de Bretagne (UEB), Place Nicolas Copernic, 29280 Plouzané, France

19 <sup>7</sup>Laboratory of Coastal Benthic Ecology (LEBCO), DYNECO, Ifremer Centre Bretagne, 29280,  
20 Plouzané, France

21 <sup>8</sup>Sorbonne Université, 4, place Jussieu, 75005 Paris, France

22  
23 \* Corresponding author: tel: + 33 2 98 50 42 88; fax: + 33 2 98 97 81 24,  
24 E-mail: [stephanie.auzoux-bordenave@mnhn.fr](mailto:stephanie.auzoux-bordenave@mnhn.fr)

25  
26  
27 **Abstract**

28 Ocean acidification is a major global stressor that leads to substantial changes in  
29 seawater carbonate chemistry, with potentially significant consequences for calcifying  
30 organisms. Marine shelled mollusks are ecologically and economically important  
31 species providing essential ecosystem services and food sources for other species.  
32 Because they use calcium carbonate (CaCO<sub>3</sub>) to produce their shells, mollusks are  
33 among the most vulnerable invertebrates to ocean acidification, with early  
34 developmental stages being particularly sensitive to pH changes. This study investigated  
35 the effects of CO<sub>2</sub>-induced ocean acidification on larval development of the European  
36 abalone *Haliotis tuberculata*, a commercially important gastropod species. Abalone  
37 larvae were exposed to a range of reduced pHs (8.0, 7.7 and 7.6) over the course of their

38 development cycle, from early-hatched trochophore to pre-metamorphic veliger.  
39 Biological responses were evaluated by measuring the survival rate, morphology and  
40 development, growth rate and shell calcification. Larval survival was significantly lower  
41 in acidified conditions than in control conditions. Similarly, larval size was consistently  
42 smaller under low pH conditions. Larval development was also affected, with evidence  
43 of a developmental delay and an increase in the proportion of malformed or unshelled  
44 larvae. In shelled larvae, the intensity of birefringence decreased under low pH  
45 conditions, suggesting a reduction in shell mineralization. Since these biological effects  
46 were observed for pH values expected by 2100, ocean acidification may have  
47 potentially negative consequences for larval recruitment and persistence of abalone  
48 populations in the near future.

49 **Keywords:** ocean acidification, abalone, larval development, shell mineralization

## 50 **1. Introduction**

51 Ocean acidification and warming are major concerns for marine ecosystems. By the  
52 end of the 21<sup>st</sup> century, global mean surface temperatures are expected to increase by 1  
53 to 3°C, while surface ocean pH is likely to decrease by 0.1 to 0.3 units (Gattuso et al.,  
54 2015). These changes will lead to alterations in seawater carbonate chemistry and a  
55 reduction in the degree of saturation with respect to calcium carbonate (Gattuso et al.,  
56 2015; IPCC, 2014). Since marine benthic ecosystems constitute a reservoir for  
57 biodiversity, several studies have focused on the evaluation of changing ocean  
58 conditions on marine biodiversity via the measurement of key physiological and  
59 ecological processes in marine organisms (for a review, see Widdicombe and Spicer,  
60 2008).

61 Changing ocean conditions are considered as major threats to marine species,

62 affecting early development, skeletal growth and key physiological functions, which can  
63 ultimately impact animal behaviour and species distribution (Kroeker et al., 2010;  
64 Widdicombe and Spicer, 2008). Reduced oceanic pH has been shown to impact a  
65 variety of calcifying species, such as corals, mollusks and echinoderms, leading to  
66 contrasting biological responses (Hendricks et al., 2010; Hofmann et al., 2010;  
67 Wittmann and Pörtner, 2013). Because calcium carbonate ( $\text{CaCO}_3$ ) is necessary for  
68 shell production, mollusks are among the most vulnerable invertebrates to ocean  
69 acidification, with larval and juvenile stages being particularly vulnerable (Gazeau et  
70 al., 2013; Orr et al., 2005; Przeslawski et al., 2015 Talmage and Gobler, 2010). Indeed,  
71 it is during larval development that marine mollusks initiate the deposition of calcium  
72 carbonate ( $\text{CaCO}_3$ ) to build their shell (Kurihara, 2008).

73 A number of studies have reported delays in development, reduced growth rate  
74 and/or shell abnormalities in larval mollusks that could potentially affect larval survival,  
75 metamorphosis and recruitment into adult populations (Parker et al., 2013; Ross et al.,  
76 2011). The impacts of ocean acidification may be particularly severe for bivalves and  
77 gastropods, which start to calcify at early developmental stages (Parker et al., 2013).  
78 Since many mollusk species are commercially important food sources, negative impacts  
79 of ocean acidification may also result in significant economic loss (Ekstrom et al., 2015;  
80 Gazeau et al., 2007). Several studies have recently focused on the impacts of elevated  
81 partial pressure of  $\text{CO}_2$  ( $p\text{CO}_2$ ) on embryonic and larval stages of shelled mollusks,  
82 especially cultivated bivalves (see reviews by Gazeau et al., 2013; Parker et al., 2013).  
83 In comparison to marine bivalves, our knowledge of the impacts of elevated  $p\text{CO}_2$  on  
84 early developmental stages in gastropods is based on fewer studies, covering only about  
85 four genera (*Cavolinia*, *Crepidula*, *Littorina* and *Haliotis*). In the gastropods studied to  
86 date, ocean acidification was shown to reduce larval survival, increase development  
87 time, alter morphology and/or impair shell formation and calcification (Byrne et al.,

88 2011; Comeau et al., 2010; Crim et al., 2011; Ellis et al., 2009; Guo et al., 2015; Kimura  
89 et al., 2011; Noisette et al., 2014; Zippay and Hofmann, 2010).

90 Among the species that have been considered in acidification studies, abalone  
91 (*Haliotis* spp.) are ecologically and economically important, acting as grazers in the  
92 marine ecosystem and as a food source for humans (Cook, 2014; Huchette and Clavier,  
93 2004). Many abalone species worldwide have experienced severe population declines  
94 due to both overfishing and the combined effects of environmental stressors such as  
95 elevated CO<sub>2</sub>, global warming and pathogen occurrence (Crim et al., 2011; Travers et  
96 al., 2009; Morash and Alter, 2015). Early-life-history stages of abalone appear to be  
97 negatively affected by elevated CO<sub>2</sub>, with a higher percentage of deformed larvae under  
98 low pH conditions than other intertidal marine mollusks such as oysters (Byrne et al.,  
99 2011; Crim et al., 2011; Guo et al., 2015; Kimura et al., 2011; Zippay and Hofmann,  
100 2010). For example, elevated seawater CO<sub>2</sub> concentrations impaired larval development  
101 and reduced larval survival in the northern abalone *Haliotis kamtschatkana* (Crim et al.,  
102 2011). In the abalone *H. coccoradiata*, embryos were dramatically affected by a  
103 combination of warm (+2 to +4°C) and acidified conditions (-0.4 to -0.6 pH units) with  
104 only a small percentage surviving, and with those embryos that did survive producing  
105 unshelled larvae (Byrne et al., 2011).

106 The European abalone *Haliotis tuberculata* is a commercially important mollusk  
107 species, for which the whole life cycle is completed under anthropogenic control  
108 (Courtois de Viçose et al., 2007). As for most marine invertebrates, abalone display a  
109 pelago-benthic life cycle with a larval planktonic stage followed by a critical  
110 metamorphosis into the benthic juvenile, making them highly sensitive to environmental  
111 changes (Byrne et al., 2011). Larval development and shell formation have been  
112 extensively studied in *H. tuberculata*, with deposition of amorphous calcium carbonate  
113 (ACC) in the early larval shell followed by deposition of aragonite in the juvenile and

114 adult shell (Auzoux-Bordenave et al., 2010). Since ACC and aragonite are relatively  
115 soluble forms of CaCO<sub>3</sub> (compared to calcite), the abalone shell is a relevant model for  
116 investigating the effects of ocean acidification. The controlled production of *Haliotis*  
117 *tuberculata* embryos and larvae provides a unique opportunity to study the impact of  
118 acidification on the early development of a marine calcifying species.

119 Here we investigated the effects of CO<sub>2</sub>-induced ocean acidification on survival,  
120 early development, growth and shell mineralization during the entire larval development  
121 of the European abalone *Haliotis tuberculata*. Abalone larvae, obtained from a  
122 controlled fertilization carried out at the 'France-Haliotis' hatchery, were exposed to  
123 three experimental pHs (8.0, 7.7 and 7.6) throughout their larval development.  
124 Biological responses of larvae were evaluated by measuring the survival rate,  
125 morphology and development, growth and shell calcification. Optical and SEM  
126 microscopy analyses were performed to assess whether reduced pH had an influence on  
127 larval shell morphology and microstructure.

128

## 129 **2. Materials and methods**

130

### 131 **2.1. Production of abalone larvae**

132 *Haliotis tuberculata* parental stock were collected from northwest Brittany (Roscoff,  
133 France) and conditioned in flowing seawater at the 'France-Haliotis' commercial  
134 abalone hatchery (Plouguerneau, France). Larvae were obtained from controlled  
135 fertilizations carried out in September 2013 at a water temperature of 17.0 ± 0.5°C,  
136 which led to approximately 6x10<sup>6</sup> larvae. Following fertilization, egg density was  
137 evaluated under a binocular microscope and the embryos were transferred to  
138 experimental tanks for acidification experiments.

## 139      **2.2. Experimental design**

140      The effects of lowered pH were investigated by exposing abalone larvae to three pH  
141 conditions, including one present-day control pH 8.0 ( $p\text{CO}_2 \approx 400 \mu\text{atm}$ ) and two levels  
142 of pH predicted under varying climate change scenarios: 7.7 ( $p\text{CO}_2 \approx 1000 \mu\text{atm}$ ) and  
143 7.6 ( $p\text{CO}_2 \approx 1400 \mu\text{atm}$ ), as outlined by Riebesell et al. (2010). Experiments were carried  
144 out in September 2013 at the ‘France-Haliotis’ hatchery, according to an experimental  
145 design adapted from Martin et al. (2011). Fertilized embryos were grown in 300 L tanks  
146 of flowing seawater at a density of  $5 \times 10^5$  per tank, under the three  $p\text{CO}_2$  conditions, and  
147 with two replicate tanks per condition. The effects of seawater acidification were  
148 investigated over the total duration of larval development (5 days), from early-hatched  
149 trochophore larvae to the pre-metamorphic stage. *Haliotis* larvae are lecithotrophic and  
150 were not fed during the experiment, avoiding any influence of diet on the biological  
151 responses.

152

### 153      *2.2.1. pH control and carbonate chemistry*

154      Larval tanks were kept in a temperature-controlled room and supplied with natural  
155 filtered seawater that was continuously aerated with ambient air. The temperature and  
156 salinity were measured daily using a conductimeter (3110, WTW, Germany). Low  
157 seawater pH was obtained by bubbling  $\text{CO}_2$  (Air liquide, France) into the tanks through  
158 electro-valves regulated by a pH-stat system (Aquastar, IKS Computer System,  
159 Germany). Seawater pH ( $\text{pH}_T$ , expressed on the total hydrogen ion concentration scale,  
160 Dickson, 2010) was adjusted to the desired level from ambient  $\text{pH}_T$  (8.0) to low  $\text{pH}_T$   
161 (7.7 and 7.6) to within  $\pm 0.05$  pH units. pH values on the National Bureau of Standards  
162 (NBS) scale obtained with the pH-stat system were adjusted daily with measurements of  
163  $\text{pH}_T$  for each tank using a pH meter (Metrohm 826 pH mobile, Metrohm, Switzerland)

164 with a glass electrode (Metrohm electrode plus) calibrated on the total scale using  
165 Tris/HCl and 2-aminopyridine/HCl buffer (Dickson *et al.*, 2007). pH (NBS) was  
166 recorded every 15 minutes in each tank by the pH-stat system and converted to pH<sub>T</sub> by  
167 using daily measurements.

168 Total alkalinity (A<sub>T</sub>) of seawater was measured twice on 100 ml samples taken from  
169 incoming water and from each experimental tank. Seawater samples were filtered  
170 through 0.7 µm Whatman GF/F membranes, immediately poisoned with mercuric  
171 chloride and stored in a cool dark place pending analyses. A<sub>T</sub> was determined  
172 potentiometrically using an automatic titrator (Titroline alpha, Schott SI Analytics,  
173 Germany) calibrated with the National Bureau of Standards scale. A<sub>T</sub> was calculated  
174 using a Gran function applied to pH values ranging from 3.5 to 3.0 as described by  
175 Dickson *et al.* (2007) and corrected by comparison with standard reference material  
176 provided by Andrew G. Dickson (CRM Batch 111). *p*CO<sub>2</sub> and other parameters of  
177 carbonate chemistry were determined from pH<sub>T</sub>, A<sub>T</sub>, temperature and salinity by using  
178 the CO<sub>2</sub>SYS software (Lewis and Wallace, 1998) using constants from Mehrbach *et al.*  
179 (1973) refitted by Dickson and Millero (1987).

180

### 181 *2.2.2. Larval sampling and measurements*

182 Larval development was monitored by daily observations with a binocular  
183 microscope. Larvae were sampled at four different development stages: trochophore  
184 (approximately 20 hours post fertilization, hpf), early veliger (30 hpf), post-torsional  
185 veliger (48 hpf) and pre-metamorphic stage (96 hpf). At each sampling time point, 10-  
186 12 L of seawater containing larvae were collected from each tank. Larval viability was  
187 calculated at each stage as the proportion of live larvae divided by the total number of  
188 larvae (n ≈ 200). Larvae were filtered on a 40 µm-sieve and aliquoted into 15 ml tubes.  
189 Larval samples were then fixed in either 70% ethanol for transmitted and polarized light



190 microscopy or in a 3% glutaraldehyde solution in Sørensen-sucrose buffer, adjusted to  
191 1100 mOsm for SEM analysis.

192

### 193 **2.3. Light and polarized microscopy**

#### 194 *2.3.1. Slide preparation and observation*

195 Microscopic slides were prepared with ethanol-fixed larvae from the different pH  
196 conditions (two slides per condition). Approximately 100 larvae were whole-mounted in  
197 a drop of glycerol, with the amount of ethanol transferred kept to a minimum. Slides  
198 were kept at room temperature for 5 to 10 min, allowing ethanol to evaporate and larvae  
199 to settle. Four spots of vacuum gel were deposited on the corners of a squared cover slip  
200 to prevent the larvae being crushed. After placing the cover slip over the glycerol, the  
201 slides were gently sealed with varnish. About 200 larvae per condition were observed  
202 and photographed under phase contrast and polarized light with an Olympus binocular  
203 microscope (Olympus, Hamburg, Germany) equipped with polarizing filters. All images  
204 were acquired with a digital camera (DS-R11, Nikon) at 100x magnification and 40 ms  
205 light exposition. Images were analyzed with NIS-element and Image-J software.

206

#### 207 *2.3.2. Morphometrical analysis*

208 Larval development stage, shell presence and size were determined under light  
209 microscopy (n = 100 larvae per tank, 200 larvae per pH condition). Maximal larval  
210 length and width of normal larvae were measured in specimens lying on the lateral side,  
211 as shown by the dotted black arrows in Figure 1. The product of length \* width was  
212 calculated for each larva and the mean of these values per tank was computed. A growth  
213 index per tank was calculated as the square root of each of these means.

214 Trochophore larvae were scored as one of four possible morphological groups,  
215 according to the presence/absence of larval shell, the occurrence of body abnormalities  
216 and/or delayed development:

- 217 1- normal shelled larvae (Fig 1A),
- 218 2- shelled larvae with abnormalities or delayed development,
- 219 3- unshelled larvae with normal body (Fig 1B),
- 220 4- unshelled larvae with abnormalities or delayed development,

221 For veliger stages, the presence/absence of shell, body abnormalities and/or delayed  
222 development were also recorded. Additional attributes like mantle formation,  
223 appearance of eyes or tentacles, and shell abnormalities were included for the  
224 assessment of morphological status.

225 According to these parameters, veliger larvae were scored as one of the following four  
226 morphological groups:

- 227 1- normal shelled larvae (Fig. 2A, D, G)
- 228 2- larvae with shell malformation (Fig. 2B, E)
- 229 3- larvae presenting body abnormalities or delayed development (Fig. 2H)
- 230 4- larvae with both shell and body abnormalities (Fig. 2C, F, I)

231

### 232 *2.3.3. Birefringence analysis*

233 The degree of CaCO<sub>3</sub> mineralization within the larval shell was evaluated by  
234 measuring the intensity of birefringence under cross-polarized light using an Olympus  
235 microscope, according to the method described by Jardillier et al. (2008). Cross-  
236 polarized light passing through calcium carbonate (an anisotropic material) is double  
237 refracted. As shells with higher calcium carbonate content double refract more light, the  
238 intensity of birefringence can be used as a proxy for the evaluation of shell

239 mineralization (Noisette et al., 2014). Measurements were obtained for 30, 48 and 96  
240 hpf larvae. Earlier larval stages were not considered because their shell lacked sufficient  
241 crystallized  $\text{CaCO}_3$  to calculate birefringence. Birefringence was determined for 40  
242 larvae per treatment using Image J software. The mean of grey-scale level (in pixels)  
243 was determined for each area of the larval shell showing birefringence (1 to 3 areas per  
244 larval shell). The values for each area were averaged into a global mean grey-scale  
245 value, providing the birefringence intensity (in %) for each larval shell (Fig S1,  
246 electronic supplementary material). Larvae were categorized as one of three types: fully  
247 mineralized (birefringence > 90%); partially mineralized (70% < birefringence < 90%)  
248 and less mineralized (birefringence < 70%). Finally, the birefringence intensity of larval  
249 shells grown under the different pH conditions was expressed as a percentage of larvae  
250 distributed in the three categories for each developmental stage.

251

#### 252 **2.4. Scanning electron microscopy (SEM)**

253 Four individuals per pH condition were used for SEM analysis. Larval samples were  
254 fixed in a 3% glutaraldehyde solution and then washed in Sørensen-sucrose buffer.  
255 Samples were subsequently dehydrated in a series of increasingly concentrated ethanol  
256 solutions and were critical point dried with liquid carbon dioxide. Finally, samples were  
257 carbon-coated and observed at the “Plateforme d’Imagerie et de Mesures en  
258 Microscopie” (PIMM, Université de Bretagne Occidentale, Brest, France) with a  
259 scanning electron microscope operating at 5 kV (Hitachi S-3200N).

260

261

## 262 **2.5. Statistical analyses**

263 All statistical analyses were performed with R software (R Development Core Team,  
264 2014) or Systat 12. In order to determine if larval viability was significantly different  
265 between pH treatments, an unpaired Student's t-test with Welch correction was  
266 performed. For morphological and developmental data, a homogeneity  $\chi^2$  test was  
267 performed in order to evaluate the effect of elevated  $p\text{CO}_2$  on larval phenotypes.  
268 Correlation between larval length and width across pH treatments was assessed using a  
269 Spearman's correlation test, allowing length and width to be combined as one parameter  
270 to estimate growth. Differences in larval growth across pH treatments were assessed by  
271 repeated measures ANOVA using the growth index per tank (pH: fixed crossed factor,  
272 time: fixed repeated factor) followed by post-hoc Tukey tests for multiple comparisons  
273 using the appropriate mean square error (Doncaster and Davey, 2007). To test the effect  
274 of pH on shell birefringence, a homogeneity  $\chi^2$  test was performed followed by a pair-  
275 wise Wilcoxon rank sum. For all tests, differences were considered significant at  $p <$   
276 0.05.

## 277 **3. Results**

### 278 **3.1. Seawater parameters**

279 Mean values of seawater carbonate chemistry parameters are reported in Table 1.  
280 The  $\text{pH}_T$  of experimental aquaria was maintained closed to nominal values throughout  
281 the experiment, respectively at a  $\text{pH}_T = 8.0$  ( $p\text{CO}_2$  of  $460 \mu\text{atm} \pm 3\mu\text{atm}$ ),  $\text{pH}_T = 7.68$   
282 ( $p\text{CO}_2$  of  $1055 \pm 3\mu\text{atm}$ ) and  $\text{pH}_T = 7.58$  ( $p\text{CO}_2$  of  $1331 \pm 10\mu\text{atm}$ ). In the following  
283 sections, rounded mean  $\text{pH}_T$  values are used namely pH 8.0, 7.7, and 7.6). The  
284 temperature was maintained at  $17.0 \pm 0.5^\circ\text{C}$  ( $n = 5$ ) and salinity at  $37.0 \pm 0.1$  ( $n = 5$ ) in  
285 all tanks and there were no significant differences across treatments. Total alkalinity

286 ( $A_T$ ) measured from incoming water and from experimental tanks differed only slightly  
287 and remained stable over the experiment (mean = 2344  $\mu\text{Eq.kg}^{-1}$ ).

288

### 289 **3.2. Larval viability**

290 Larval viability was measured for the three development stages in the different pH  
291 conditions (Fig. 3). In control seawater (pH 8.0) larval viability remained > 95% until  
292 the pre-metamorphic stage. A significant decrease in larval viability was observed in  
293 reduced pH treatments. At pH 7.7 reduced larval viability was observed in 30 and 48  
294 hpf larvae, although the decrease was not significant ( $p = 0.257$  and  $p = 0.125$   
295 respectively), but a 50% reduction in viability at 96 hpf was significant (unpaired  
296 Student's t-test,  $p < 0.001$ ). At pH 7.6, viability slightly decreased to 75% at 30 hpf ( $p =$   
297  $0.067$ ) and dropped to 56% and 47% at 48 and 96 hpf respectively (unpaired Student's t-  
298 test,  $p = 0.001$  and  $p = 0.008$  respectively). Abnormalities in larval morphology and  
299 hyperactive movements were observed in the two low pH conditions for all four larval  
300 stages (S. Auzoux-Bordenave, pers. observations).

### 301 **3.3. Morphology and development**

302 Larval developmental stage and shell presence were determined at each time point  
303 and compared between the different treatments, according to the four morphological  
304 groups defined in section 2.3.2. Larval distribution within these groups across the  
305 different pH treatments is shown for the trochophore stage (Fig. 4A) and later veliger  
306 stages (Figs. 4B-D). At each developmental stage, conformity  $\chi^2$  tests showed that the  
307 distribution within the groups was dependent on the pH ( $p < 0.001$ ).

308 At the first developmental stage (20 hpf trochophore), larval distribution into the  
309 different morphological groups was pH dependent (Fig. 4A;  $\chi^2 = 83.31$ ,  $df = 6$ ,  $p <$

310 0.001). Normal shelled larvae represented 60% of the total amount of larvae in the  
311 control group, while this proportion was less than 10% at the lowest pH ( $\chi^2 = 83.31$ ,  $df =$   
312 6,  $p < 0.001$ ). The percentage of shelled larvae, including those with normal and  
313 abnormal bodies, was significantly lower at pH 7.6 (47%) compared to controls (75%)  
314 ( $\chi^2 = 25.19$ ,  $df = 2$ ,  $p < 0.001$ ). In parallel, the percentage of unshelled larvae was  
315 significantly greater at pH 7.6 (53%) compared to controls (10%) ( $\chi^2 = 25.19$ ,  $df = 2$ ,  $p <$   
316 0.001). The presence of unshelled larvae with normal bodies in the control group  
317 probably occurred due to differences in the timing of shell morphogenesis among  
318 individuals.

319 The distribution of veliger stages into the different morphological groups was also  
320 pH dependent, and similar to that observed in trochophore larvae (Fig. 4B-D). At 30  
321 hpf, a strong decrease in the percentage of normal larvae was observed, from 60% in the  
322 control group to only 10% at the lowest pH (Fig. 4B;  $\chi^2 = 75.25$ ,  $df = 6$ ,  $p < 0.001$ .  
323 Meanwhile, the proportion of malformed or delayed larvae increased from 25 to 45%.  
324 An important impact of acidification was observed at 48 hpf, where the proportion of  
325 normal larvae decreased from 90% in the control condition to about 58% at pH 7.7 and  
326 less than 20% at pH 7.6. (Fig. 4C;  $\chi^2 = 133.77$ ,  $df = 6$ ,  $p < 0.001$ ). The proportion of  
327 larvae showing both shell and body abnormalities (cumulative effects) strongly  
328 increased when pH was reduced, from a negligible percentage in the control group, to  
329 above 65% at pH 7.6 ( $\chi^2 = 133.77$ ,  $df = 6$ ,  $p < 0.001$ ). At 96 hpf, the percentage of  
330 normal larvae also drastically decreased in the reduced pH treatments, from 70% in the  
331 control group to 25% at pH 7.7 and to 12% at pH 7.6 (Fig. 4D) ( $\chi^2 = 119.14$ ,  $df = 6$ ,  $p <$   
332 0.001). As a result, the amount of larvae with body malformations or delayed  
333 development increased significantly with seawater acidification, from 20% in control  
334 conditions to 45% at pH 7.6. For all three veliger stages, the proportion of larvae that  
335 showed both shell and body abnormalities (cumulative effects) strongly increased when

336 lowering the pH, while the proportion of larvae displaying only shell malformation  
337 remained < 15% and did not differ significantly among treatments.

338 Larval shells (48 hpf) grown at lower pH exhibited differences in the texture and  
339 porosity of the surface layer (Fig. 5). In control larvae, the mineralized protoconch  
340 almost completely covered the larval body (Fig. 5A). At higher magnification, the shell  
341 surface showed a uniform granular texture covered by a very thin homogeneous layer  
342 (Fig. 5B). In 48 hpf veliger larvae exposed to decreased pH (7.6), the protoconch  
343 exhibited an irregular surface, with differences in thickness, and the homogeneous outer  
344 layer was not as distinct as that of control larvae (Fig. 5C). The outer surface had a  
345 porous appearance with numerous small holes interspaced between the biominerals and  
346 remnants of the organic coating (Fig 5.D).

347

#### 348 **3.4. Larval growth**

349 Length and width of abalone larvae reared in the three pH conditions were measured.  
350 Only normal larvae were used in order to determine if growth rate differed according to  
351 pH. For each treatment, a Spearman's correlation was carried out to evaluate the  
352 relationship between larval length and width during the course of the experiment (Table  
353 2). The results showed a linear relationship between the two measures for each pH  
354 group (Fig. 6), allowing length and width to be combined as one parameter to estimate  
355 larval growth. The growth index (calculated as the square root of length \* width) of  
356 larvae exposed to the three pH conditions and according to time is presented in Figure  
357 7.

358 Both pH and time significantly affected the growth index (Table 3A;  $p = 0.009$  and  $p$   
359  $< 0.001$ , respectively), while the interaction term was not significant (Table 3A;  $p =$   
360  $0.088$ ). Larval size significantly increased between 20 hpf and 30 hpf and between 48

361 hpf and 96 hpf (Table 3B; Tukey,  $p = 0.002$  and  $p = 0.009$ , respectively). Larval size  
362 was significantly lower at pH 7.7 and 7.6 vs 8.0 (Tukey,  $p = 0.008$  and  $p = 0.031$ ,  
363 respectively) but did not differ significantly between the two low pH treatments (Table  
364 3B; Tukey,  $p = 0.105$ ).

365

### 366 **3.5. Shell calcification**

367 Under control pH conditions, larvae clearly exhibited the characteristic dark cross  
368 indicating a radial arrangement of the aragonite crystals within 2 to 3 areas of  
369 birefringence (Fig. 8B). Larvae grown at lower pH (7.6) typically showed a decrease in  
370 shell birefringence indicating reduced calcification (Fig. 8D). At 30 hpf, the distribution  
371 of larvae into the three categories (i.e. fully mineralized, partially mineralized and less  
372 mineralized) was not significantly different between the three pH treatments (Fig. 9A;  
373 Table 4A,  $\chi^2 = 9.28$ ,  $df = 4$ ,  $p = 0.055$ ). At 48 hpf, the number of fully mineralized  
374 larvae was strongly reduced at low pH conditions, while the number of partially and less  
375 mineralized larvae was significantly higher (Fig. 9B; Table 4B,  $p < 0.001$ ). Similar  
376 results were observed in 96 hpf larvae, with significant differences in larval  
377 birefringence between the control group and the low pH group 7.7. (Fig. 9C; Table 4B,  
378  $p < 0.001$ ). At pH 7.6, larval distribution within the three categories was close to that  
379 observed in the control group (Table 4B,  $p = 0.073$ ).



#### 380 4. Discussion

381 This study provides the first evidence that decreased pH negatively impacts  
382 survival, early larval development and shell calcification in the European abalone  
383 *Haliotis tuberculata*. Biological responses were measured over the total duration of  
384 larval development (from early-hatched trochophore to the pre-metamorphic stage),  
385 using near-future oceanic CO<sub>2</sub> levels. Our results are consistent with previous studies on  
386 mollusk larvae, in which near-future CO<sub>2</sub> concentrations reduced survival and impaired  
387 early development, suggesting potential negative consequences for larval recruitment  
388 and the persistence of abalone populations (see reviews by Gazeau et al., 2013; Ross et  
389 al., 2011).

390 Our results showed that *H. tuberculata* experienced a significant decrease in larval  
391 survival at pH 7.7 and 7.6 with a maximum decrease of 47% at pH 7.6 relative to the  
392 control. Furthermore, low pH conditions negatively affected larval morphology, inducing  
393 a developmental delay and a decrease in the proportion of shelled larvae. In addition to  
394 developmental delays and body abnormalities, seawater acidification resulted in  
395 decreased growth rate (length and width). These results are consistent with previous  
396 studies on other abalone species, such as those of Byrne et al. (2011), Crim et al. (2011)  
397 and Kimura et al. (2011) that found reduced survival in, respectively, *H. coccoradiata*,  
398 *H. kamtschatkana* and *H. discus hannai*. Moreover, these studies showed similar results  
399 with respect to increasing numbers of unshelled larvae at lower pHs (Byrne et al., 2011;  
400 Crim et al., 2011), as well as reduction in larval size (Crim et al., 2011; Kimura et al.,  
401 2011). Abalone appears to be particularly sensitive to ocean acidification at early life  
402 stages, which may lead to lower recruitment in these commercially and ecologically  
403 important species. Similarly, a recent comparison of two abalone species (*H. discus*  
404 *hannai* and *H. diversicolor*) together with the oyster *Crassostrea angulata* found

405 reduced size to be a common response to reduced pH (Guo et al., 2015). Among other  
406 species, deformities have been observed in the gastropod *Crepidula fornicata* under  
407 reduced pH (Noisette et al., 2014), and the oysters *Crassostrea gigas* and *Saccostrea*  
408 *glomerata* displayed reduced development rates and increasing levels of abnormalities  
409 in low pH treatments (Kurihara et al., 2007; Kurihara, 2008; Parker et al., 2009;  
410 Timmins-Schiffman et al., 2013; Watson et al., 2009). More generally, it seems that  
411 reduced survival, abnormal or prolonged development and reduced size represent a  
412 common response of many marine mollusk larvae exposed to elevated CO<sub>2</sub> (Gazeau et  
413 al., 2013; Ross et al., 2011).

414 In addition to reduced survival, development abnormalities and decreased growth  
415 rate, seawater acidification also resulted in reduced shell calcification of abalone larvae,  
416 as shown by the decrease in birefringence intensity under cross-polarized light. The  
417 reduction in shell calcification in larvae grown at lower pH treatments was highly  
418 significant at later stages (48 and 96 hpf). However, only a slight, but not significant,  
419 increase in the percentage of partially mineralized shells was observed at 30 hpf under  
420 low pH 7.7, and no significant differences were observed between control and pH 7.6.  
421 This could be related to low amounts of calcium carbonate in the earliest larval shell,  
422 which in combination with heterogeneous crystalline orientation, might have resulted in  
423 higher variability in the grey levels. In the future, attempts to calibrate grey scale levels  
424 with measures of shell mass may allow improvements in the use of birefringence as a  
425 proxy for determining shell mineralization.

426 Cross-polarized light and SEM observations of shelled larvae provided evidence of  
427 a lack of calcification under lower pH, suggesting that larvae deposited less calcium  
428 carbonate and produced a thinner shell. The impacts of ocean acidification on shell  
429 calcification have been well studied in juvenile and adult mollusks, showing that their

430 calcified shell is highly sensitive to elevated  $p\text{CO}_2$  (Gazeau et al., 2013). For example,  
431 in the marine bivalves *Mytilus edulis* and *C. gigas*, the calcification rate was negatively  
432 impacted by a short-term exposure to low pH (-0.2 units from ambient, Gazeau et al.,  
433 2007). Previous studies have reported a decrease in shell size and shell thickness as well as  
434 changes in morphology and shape of the larval shell under elevated  $p\text{CO}_2$  (Gazeau et al.,  
435 2010; Kurihara et al., 2007, 2008; Miller et al., 2009; Talmage and Gobler, 2009). Few  
436 studies have reported changes in larval shell calcification under OA stress by measuring  
437 birefringence intensity; however, our results on larval abalone shell are consistent with  
438 those of Kurihara *et al.* (2007) and Noisette *et al.* (2014), obtained respectively in  
439 *Crassostrea gigas* and *Crepidula fornicata*, showing a reduction in larval shell  
440 calcification when seawater pH is lowered.

441 Mollusk shell formation is a complex process starting at the trochophore stage in  
442 abalone. The larval shell is formed by a specialized group of ectodermal cells that form  
443 the shell gland and the organic periostracum. Primary mineralization takes place  
444 between the shell gland and the periostracum, producing the so-called protoconch in  
445 gastropods (Eyster, 1986). In abalone, the first deposition of  $\text{CaCO}_3$  occurs at an early  
446 veliger stage with the deposition of amorphous calcium carbonate (ACC), which is rapidly  
447 transformed into crystalline aragonite (Auzoux-Bordenave et al., 2010). Since ACC and  
448 aragonite are more soluble forms of  $\text{CaCO}_3$  than calcite in lower pH conditions, the larval  
449 shell is likely to be more susceptible to dissolution than juvenile or adult shell. In the  
450 present study, SEM images of larval shells reared at pH 7.6 revealed numerous small  
451 holes at the shell surface, suggesting that  $\text{CaCO}_3$  dissolution is likely to be a factor  
452 explaining reduced calcification in abalone larvae experiencing acidified conditions.  
453 Shell dissolution processes may arise as a result of a lower availability of carbonate ions at  
454 the site of calcification (a direct effect of the carbonate chemistry) or through indirect

455 physiological changes in ionic composition, matrix protein formation or enzymatic  
456 activities (Hüning et al., 2012).

457       Recent studies have suggested that lowered aragonite saturation state may be one of the  
458 key parameters controlling whether shell development in larval mollusks occurs normally  
459 (Thomsen et al., 2015; Waldbusser et al., 2015). In the oyster *C. gigas*, developmental  
460 success and growth rates were not significantly altered as long as carbonate ion  
461 concentrations were above aragonite saturation levels, but they strongly decreased when  
462 carbonate ion concentrations dropped below aragonite saturation levels (Gazeau et al.,  
463 2011). The authors suggested that the mechanisms used by oyster larvae to regulate  
464 calcification rates were not efficient enough to compensate for the low availability of  
465 carbonate ions under acidified conditions. Interestingly, recent studies suggest that initial  
466 shell formation in larvae can occur even when aragonite is reduced below saturation levels  
467 (Frieder et al., 2016), and that dissolution of the initial shell, at least in species such as *M.*  
468 *edulis*, only occurs at very high levels of OA (Ramesh et al., 2017). However, it is likely  
469 that resource allocation trade-offs between biomineralization and other vital biological  
470 processes will begin to occur as larval development progresses, as shown in sea urchins  
471 (Pan et al., 2015). This could help to explain why reduced birefringence of *H. tuberculata*  
472 larvae under elevated  $p\text{CO}_2$  was only observed at 48 and 96 hpf, and not at 30 hpf (this  
473 study).

474       As well as directly causing shell dissolution, a number of other biological processes  
475 responsible for larval shell calcification, such as matrix protein production, chitin  
476 synthesis and enzymatic control are influenced by changes in seawater  $p\text{CO}_2$  (Weiss et  
477 al., 2013). For example, the activity of carbonic anhydrase, an enzyme that catalyzes the  
478 reversible hydration of  $\text{CO}_2$  to  $\text{HCO}_3^-$  and  $\text{H}^+$ , reaches its maximum activity at the end  
479 of each developmental stage and has been correlated with larval shell biomineralization

480 (Gaume et al., 2011; Medakovic, 2000). In the mussel *M. edulis*, six months of  
481 incubation at 750  $\mu\text{atm } p\text{CO}_2$  (pH 7.5) significantly reduced carbonic anhydrase activity  
482 within the mantle tissue, explaining shell growth reduction (Fitzer et al., 2014b). More  
483 recently, evidence from proteomic studies suggests that elevated  $p\text{CO}_2$  influences a  
484 wide range of molecular pathways, including several associated with biomineralization,  
485 metabolism and the cytoskeleton, and which may correlate with calcification  
486 (Dineshram et al., 2015; Harney et al., 2016).

487 In order to discriminate between the effect of the aragonite saturation state and the  
488 complex physiological effects of pH decrease on mollusk shell formation, future studies  
489 should explicitly consider  $\text{CaCO}_3$  saturation state in experimental seawater and the ability  
490 of the species to maintain internal pH at the site of calcification. Failure to properly  
491 biomineralise at this early stage not only results in reduced survival and developmental  
492 problems for larvae (Byrne, 2012), but can also have carry-over effects later in life  
493 (Hettinger et al., 2013; Rühl et al., 2017), which can explain why larval stages represent  
494 such a major bottleneck for population persistence under changing environmental  
495 conditions (Przeslawski et al., 2015). In this study, fertilization was carried out under  
496 ambient conditions, and resulting embryos were transferred into experimental tanks  
497 with different  $p\text{CO}_2$  conditions, as is the case of most studies assessing the impacts of  
498 ocean acidification on mollusk larvae. It is now well established that exposure of adults  
499 to elevated  $\text{CO}_2$  during reproductive conditioning can result in positive or negative  
500 carry-over effects being transmitted from adults to their offspring, influencing the  
501 resilience of mollusks to ocean acidification (Fitzer et al., 2014; Parker et al., 2013). In  
502 the oyster *S. glomerata*, Parker et al. (2012) found that larvae from parents exposed to  
503 elevated  $\text{CO}_2$  during reproductive conditioning were larger and developed faster when  
504 they also experienced reduced pH, as compared to larvae from parents conditioned

505 under ambient pH. These results highlight the importance of assessing carry-over effects  
506 for determining species responses to ocean acidification. To overcome this problem,  
507 future projects should conduct long-term transgenerational experiments. Biological  
508 responses measured during long-term exposure to elevated  $p\text{CO}_2$ , from reproducing  
509 adults to larval and juvenile stages, will provide valuable information regarding  
510 acclimation and adaptation of abalone to changing ocean conditions. In addition, the  
511 exploration of multiple stressors known to interact with pH (e.g. temperature, salinity,  
512 pathogens and pollutants) will result in more ecologically realistic simulations of the  
513 impact of global environmental change, providing a greater understanding of ecological  
514 relationships (e.g. Ko et al., 2014). Indeed, consideration of multiple stressors is also  
515 crucial in fisheries and aquaculture to identify optimal conditions and adapt culturing  
516 practices for sustainable shellfish production.

517

## 518 **Acknowledgements**

519 N. Wessel was supported by a post-doctoral fellowship from the MNHN (Ministère  
520 de l'Enseignement Supérieur et de la Recherche, Paris, France). This work was financed  
521 in part by the ATM program "Biomínéralisation" of the MNHN funded by the Ministère  
522 délégué à l'Enseignement Supérieur et à la Recherche (Paris, France). V. Julia, F. Nunes  
523 and E. Harney were supported by a grant from the Regional Council of Brittany, from  
524 the European Funds (ERDF) and supported by the "Laboratoire d'Excellence"  
525 LabexMER (ANR-10-LABX-19) and co-funded by a grant from the French government  
526 under the program "Investissements d'Avenir". We thank Gérard Sinquin for his  
527 assistance in scanning electron microscopy (Plateforme d'Imagerie et de Mesures en  
528 Microscopie, Université de Bretagne Occidentale, Brest). P. Dubois is a Research  
529 Director of the National Fund for Scientific Research (Belgium).  
530 The experiments complied with the current French laws.

531 **Figures and tables**

532 **Figure 1.** Trochophore larvae grown in control condition (A) and at pH 7.6 (B). These  
533 larvae were respectively classified in the morphological groups 1 (normal shelled) and 3  
534 (unshelled larval phenotype). s: shell, pt: prototrochal ciliary band, at: apical tuft.  
535 Dotted black arrows indicate the length and width measurements.

536

537 **Figure 2.** Morphological variables measured in abalone veliger larvae grown in various  
538 pH conditions; the group in brackets refers to morphological groups described in section  
539 2.3.2. **A:** 30 hpf at pH 8.0 (group 1), **B:** 30 hpf at pH 7.6 (group 2), **C:** 30 hpf at pH 7.6  
540 (group 4). **D:** 48 hpf at pH 8.0 (group 1), **E:** 48 hpf at pH 7.6 (group 2), **F:** 48 hpf at  
541 pH 7.6 (group 4). **G:** 96 hpf at pH 8.0 (group 1), **H:** 96 hpf at pH 7.6 (group 3), **I:**  
542 96 hpf at pH 7.6 (group 4). Arrows indicate attributes: eyes (e), foot (f), mantle (m),  
543 shell (s); arrowheads indicate shell abnormalities (sa) or deformed shell (ds); dotted  
544 arrows indicate the length and width measurements.

545

546 **Figure 3.** Viability of abalone larvae exposed to different pH conditions at 30 hpf (light  
547 grey), 48 hpf (grey) and 96 hpf (dark grey). Errors bars represent standard deviations.  
548 Asterisks denote significant difference between control and low-pH condition (unpaired  
549 Student's t-test,  $p < 0.05$ ).

550

551 **Figure 4.** Morphology and development of abalone larvae exposed to different pH  
552 conditions at 20 hpf (A), 30 hpf (B), 48 hpf (C), and 96 hpf (D). Grey levels represent  
553 the morphological groups, according to the developmental stage. **A (trochophore**  
554 **larvae):** black: normal shelled larvae; dark grey : shelled larvae with abnormalities or  
555 delayed development; light grey: unshelled larvae with normal body; white: unshelled

556 larvae with abnormalities or delayed development; **B, C, D (veliger larvae)** : black:  
557 normal shelled larvae; dark grey larvae with shell malformation(s)/ normal body; light  
558 grey : shelled larvae with body abnormalities or delayed development; white: larvae  
559 with both shell and body abnormalities.

560

561 **Figure 5.** Scanning electron microscopy (SEM) images of abalone larvae grown under  
562 control (A, B) and low pH condition (C, D). Shell surfaces of 48 hpf veliger are shown.  
563 **A:** lateral view of a 48h-old veliger under control pH (8.0); the protoconch is well  
564 developed and covers almost completely the larval body; **B:** detail of the shell surface  
565 boxed in 5A showing a uniform granular texture covered by a very thin organic coating;  
566 **C:** lateral view of a 48h-old veliger exposed to low pH (7.6); the protoconch appears  
567 well developed but exhibits an heterogeneous surface; **D:** detail of the protoconch  
568 surface boxed in 5C showing numerous small holes interspaced between the  
569 biominerals and remnants of the organic coating.

570

571 **Figure 6.** Correlation between length and width for each pH treatment. Light grey  
572 represents the control pH (8.0), dark grey represents the pH 7.7, and black represents the  
573 pH 7.6. Results of the statistical analyses are reported in Table 2.

574

575 **Figure 7.** Effect of decreased pH on abalone larval growth, expressed as square roots  
576 of (length x width) at 20 hpf (A), 30 hpf (B), 48 hpf (C), and 96 hpf (D). Grey level of  
577 the boxes denote the different pH conditions. Centre lines of box plots show the  
578 medians; box limits indicate the first and third quartile respectively with lines  
579 encompassing data within 1.5 times the spread from the median ( $p < 0.05$ ). Results of  
580 the statistical analyses are reported in Table 3.

581



582 **Figure 8.** Morphology and shell birefringence of 48 hpf veliger larvae grown under  
583 control (A, B) and low pH conditions (C, D). Larvae were observed under phase  
584 contrast (A, C) and polarized microscopy (B, D). Normal larvae under control pH (8.0)  
585 showing the characteristic black cross of birefringence (A, B). Shell abnormalities in  
586 larvae exposed to low pH (7.7) result in a significant decrease of calcified areas (C, D).  
587

588 **Figure 9.** Shell mineralization of larval abalone determined by polarized light  
589 microscopy for each development stage (A: 30hpf, B: 48hpf, C: 96hpf). Grey levels  
590 represent the three categories of mineralization: black bars for fully mineralized  
591 (birefringence > 90%); stripe bars for partially mineralized (70% < birefringence <  
592 90%) and dotted bars for less mineralized shells (birefringence < 70%). Differences in  
593 larval distribution across pH treatments were tested using a homogeneity  $\chi^2$  test by  
594 treating birefringence as a categorical factor (n=40 larvae per pH condition). Results of  
595 the statistical analyses are reported in Table 4.

596

597 **Table 1.** Mean parameters of seawater carbonate chemistry during the experiment.  
598 Seawater pH on the total scale ( $\text{pH}_T$ ), temperature ( $17.0 \pm 0.5$  °C), salinity ( $37.0 \pm 0.1$ )  
599 and total alkalinity (mean  $2344 \mu\text{Eq.kg}^{-1}$ ) were used to calculate  $\text{CO}_2$  partial pressure  
600 ( $p\text{CO}_2$ ;  $\mu\text{atm}$ ), Dissolved Inorganic Carbon (DIC;  $\mu\text{mol.kg SW}$ ),  $\text{HCO}_3^-$ ,  $\text{CO}_3^{2-}$ ,  
601 aragonite saturation state ( $\Omega_{\text{ar}}$ ) and calcite saturation state ( $\Omega_{\text{calc}}$ ) by using the  
602  $\text{CO}_2\text{SYS}$  software.  $\text{pH}_T$  is the average value logged throughout the 5 days of experiment  
603 (every 15 mn) in the two tanks (n = 96/tank). Temperature and salinity were measured  
604 daily (n = 5); total alkalinity was measured twice (n = 2). Results are expressed as  
605 mean  $\pm$  SD.

606

607

608 **Table 2:** Results of the Spearman's correlation performed to evaluate the nullity of  
609 correlation between larval length and width for each pH treatment in the course of the  
610 experiment.

611

612 **Table 3.** Summary of statistics used to test the differences in larval growth.

613 **A.** Results of the repeated measures ANOVA on larval growth index (square root of  
614 length \* width); **B.** Multiple comparison Tukey tests testing the effects of pH (fixed  
615 crossed factor) according to time (fixed repeated factor). Significant results in bold ( $p <$   
616  $0.05$ ).

617

618 **Table 4.** Summary of statistics used to test the differences in shell birefringence.

619 **A.** Homogeneity  $\chi^2$  test used to test the effect of pH on shell birefringence, for each  
620 development stage. **B.** Pair-wise Wilcoxon rank sum test showing differences between  
621 pH groups; Bonferroni adjusted  $p$ -values. Significant results in bold ( $p < 0,05$ ).

622

623 **Supplementary Figure S1. A.** Cross-polarized microscopy image of abalone larvae (48  
624 hpf) showing the three regions of interest (ROI) selected for the analysis of  
625 birefringence intensity. **B.** Quantification of grey-scale levels (in pixels) in the three  
626 ROI. The values were averaged to provide a global mean grey level for each larval shell  
627 (n = 40 larvae per pH condition).

628

630 **References**

- 631 Auzoux-Bordenave, S., Badou, A., Gaume, B., Berland, S., Helléouet, M.-N., Milet, C.,  
632 and Huchette, S. 2010. Ultrastructure, chemistry and mineralogy of the growing  
633 shell of the European abalone *Haliotis tuberculata*. *J. Struct. Biol.* 171: 277-290.
- 634 Byrne, M. 2012. Global change ecotoxicology: Identification of early life history  
635 bottlenecks in marine invertebrates, variable species responses and variable  
636 experimental approaches. *Mar. Environ. Res.* 76: 3-15.
- 637 Byrne, M., Ho, M., Wong, E., Soars, N. A., Selvakumaraswamy, P., Shepard-Brennan,  
638 H., Dworjanyn, S. A., *et al.* 2011. Unshelled abalone and corrupted urchins:  
639 development of marine calcifiers in a changing ocean. *Proc. R. Soc. B: Biological*  
640 *Sciences*, 278: 2376-2383.
- 641 Comeau, S., Gorsky, G., Alliouane, S., and Gattuso, J.- P. 2010. Larvae of the pteropod  
642 *Cavolinia inflexa* exposed to aragonite undersaturation are viable but shell-less.  
643 *Mar. Biol.* 157: 2341-2345.
- 644 Cook, P. A. 2014. The Worldwide Abalone Industry. *Modern Economy*, 05: 1181-1186.
- 645 Courtois de Viçose, G., Viera, M. P., Bilbao, A., and Izquierdo, M. S. 2007. Embryonic  
646 and larval development of *Haliotis tuberculata coccinea* Reeve: an indexed micro-  
647 photographic sequence. *J. Shellfish Res.* 26: 847-854.
- 648 Crim, R. N., Sunday, J. M., and Harley, C. D. G. 2011. Elevated seawater CO<sub>2</sub>  
649 concentrations impair larval development and reduce larval survival in endangered  
650 northern abalone (*Haliotis kamtschatkana*). *J. Exp. Mar. Biol. Ecol.* 400: 272-277.
- 651 Dickson, A.G., Sabine, C.L., Christian, J.R. (Eds), 2007. Guide to best practices for  
652 ocean CO<sub>2</sub> measurements. PICES Special Publication, 3: 191 pp.
- 653 Dickson, A.G. 2010. The carbon dioxide system in seawater: equilibrium chemistry and  
654 measurements. In *Guide to Best Practices for Ocean Acidification Research and*

655 Data Reporting pp.17-40. Ed. By U. Riebesell, V. J. Fabry, L. Hansson and J.-P.  
656 Gattuso). Luxembourg: Publications Office of the European Union.

657 Dickson, A.G., and Millero, F.J. 1987. A comparison of the equilibrium constants for  
658 the dissociation of carbonic acid in seawater media. *Deep Sea Res.* 34: 1733–1743.

659 Dineshram, R., Quan, Q., Sharma, R., Chandramouli, K., Yalamanchili, H.K., Chu, I.  
660 and Thiyagarajan, V., 2015. Comparative and quantitative proteomics reveal the  
661 adaptive strategies of oyster larvae to ocean acidification. *Proteomics* 15, 4120–  
662 4134.

663 Doncaster, C. P. and Davey, A. J. H. 2007. *Analysis of Variance and Covariance: How*  
664 *to Choose and Construct Models for the Life Sciences*. Cambridge: Cambridge  
665 University Press.

666 Ekstrom, J., Suatoni, L., Cooley, S., Pendleton, L., Waldbusser, G., Cinner, J., Ritter, J.,  
667 Langdon, C., van Hoodonk, R., Gledhill, D., Wellman, K., Beck, M., Brander, L.,  
668 Rittschof, D., Doherty, C., Edwards, P. & Portela, R. 2015. Vulnerability and  
669 adaptation of US shellfisheries to ocean acidification. *Nature Climate Change*, 5:  
670 207–214.

671 Ellis, R. P., Bersey, J., Rundle, S. D., Hall-Spencer, J. M., and Spicer, J. I. 2009. Subtle  
672 but significant effects of CO<sub>2</sub> acidified seawater on embryos of the intertidal snail,  
673 *Littorina obtusata*. *Aquat. Biol.* 5: 41-48.

674 Eyster, LS. 1986. Shell inorganic composition and onset of shell mineralization during  
675 bivalve and gastropod embryogenesis. *Biol. Bull.* 170:211–231

676 Fitzer, S.C., Cusack, M., Phoenix, V.R., Kamenos, N.A. 2014. Ocean acidification  
677 reduces the crystallographic control in juvenile mussel shells. *J. Struct. Biol.* 188 :  
678 39-45.

679 Fitzer, S.C., Phoenix, V.R., Cusack, M., Kamenos, N.A. 2014b. Ocean acidification  
680 impacts mussel control on biomineralisation. *Sci. Rep.* 4, 6218.  
681 (doi:10.1038/srep06218).

682

683 Frieder, C.A., Applebaum, S.L., Pan, T.-C.F., Hedgecock, D. and Manahan, D.T., 2016.  
684 Metabolic cost of calcification in bivalve larvae under experimental ocean  
685 acidification. *ICES J. Mar. Sci.* 74: 941–954.

686 Gattuso, J. P., Magnan, A., Bille, R., Cheung, W. W. L., Howes, E. L., Joos, F.,  
687 Allemand, D., *et al.* 2015. Contrasting futures for ocean and society from different  
688 anthropogenic CO<sub>2</sub> emissions scenarios. *Science* 349: aac4722.

689 Gaume, B., Fouchereau-Peron, M., Badou, A., Helléouet, M.-N., Huchette, S., and  
690 Auzoux-Bordenave, S. 2011. Biomineralization markers during early shell  
691 formation in the European abalone *Haliotis tuberculata*, Linnaeus. *Mar. Biol.* 158:  
692 341-353.

693 Gazeau, F., Gattuso, J. P., Dawber, C., Pronker, A. E., Peene, F., Peene, J., Heip, C. H.  
694 R., *et al.* 2010. Effect of ocean acidification on the early life stages of the blue  
695 mussel *Mytilus edulis*. *Biogeosciences* 7: 2051-2060.

696 Gazeau, F., Parker, L. M., Comeau, S., Gattuso, J.-P., O'Connor, W. A., Martin, S.,  
697 Pörtner, H.-O., *et al.* 2013. Impacts of ocean acidification on marine shelled  
698 mollusks. *Mar. Biol.* 160: 2207-2245.

699 Gazeau, F., Quiblier, C., Jansen, J. M., Gattuso, J.-P., Middelburg, J. J., and Heip, C. H.  
700 R. 2007. Impact of elevated CO<sub>2</sub> on shellfish calcification. *Geophys. Res. Lett.* 34.

701 Guo, X., Huang, M., Pu, F., You, W., and Ke, C. 2015. Effects of ocean acidification  
702 caused by rising CO<sub>2</sub> on the early development of three mollusks. *Aquat. Biol.* 23:  
703 147-157.

704 Harney, E., Artigaud, S., Le Souchu, P., Miner, P., Corporeau, C., Essid, H., Pichereau,  
705 V., *et al.* 2016. Non-additive effects of ocean acidification in combination with  
706 warming on the larval proteome of the Pacific oyster, *Crassostrea gigas*. *J*  
707 *Proteomics* 135: 151-161.

708 Hendriks, I. E., Duarte, C. M., and Álvarez, M. 2010. Vulnerability of marine  
709 biodiversity to ocean acidification: A meta-analysis. *Estuar. Coast. Shelf. Sci.* 86:  
710 157-164.

711 Hettinger, A., Sanford, E., Hill, T. M., Lenz, E. A., Russell, A. D., and Gaylord, B.  
712 2013. Larval carry-over effects from ocean acidification persist in the natural  
713 environment. *Glob. Change Biol.* 19, 3317–3326, doi: 10.1111/gcb.12307

714 Hofmann, G.E., Barry, J.P., Edmunds, P.J., Gates, R.D., Hutchins D.A., Klinger, T., and  
715 Sewell, M.A. 2010. The effect of ocean acidification on calcifying organisms in  
716 marine ecosystems: An organism-to-ecosystem perspective. *Annu. Rev. Ecol. Evol.*  
717 *Syst.* 41: 127-147.

718 Huchette, S., and Clavier, J. 2004. Status of the ormer (*Haliotis tuberculata* L.) industry  
719 in Europe. *J. Shellfish Res.* 23 : 951-955.

720 Hüning, A. K., Melzner, F., Thomsen, J. , Gutowska, M. A. , Krämer, L. , Frickenhaus,  
721 S. , Rosenstiel, P. , Pörtner, H. O. , Philipp, E. E. R. and Lucassen, M. 2012.  
722 Impacts of seawater acidification on mantle gene expression patterns of the Baltic  
723 Sea blue mussel: implications for shell formation and energy supply. *Marine*  
724 *Biology.* doi: 10.1007/s00227-012-1930-9

725 IPCC, 2014. Summary for Policymakers. In: *Climate Change 2014: Impacts,*  
726 *Adaptation, and Vulnerability. Part A: Global and Sectoral Aspects. Contribution of*  
727 *Working Group II to the Fifth Assessment Report of the Intergovernmental Panel on*  
728 *Climate Change.* Cambridge University Press, Cambridge, United Kingdom and New  
729 York, NY, USA, pp. 1-32.

730 Jardillier E., Rousseau M., Gendron-Badou A., Fröhlich F., Smith D.C., Martin M.,  
731 Helléouet M.-N., Huchette S., Doumenc D., Auzoux-Bordenave S., 2008. A  
732 morphological and structural study of the larval shell from the abalone *Haliotis*  
733 *tuberculata*. Mar. Biol. 154 (4): 735-744.

734 Kimura, R. Y. O., Takami, H., Ono, T., Onitsuka, T., and Nojiri, Y. 2011. Effects of  
735 elevated pCO<sub>2</sub> on the early development of the commercially important gastropod,  
736 Ezo abalone *Haliotis discus hannai*. Fish. Oceanogr. 20: 357-366.

737 Ko, G., Dineshram, R., Campanati, C., Vera, S., Havenhand, J. & Thiyagarajan, V.  
738 2014. Interactive effects of ocean acidification, elevated temperature and reduced  
739 salinity on early-life stages of the Pacific oyster. Environmental Science &  
740 Technology, 48: 10079–10088.

741 Kroeker, K. J., Kordas, R. L., Crim, R. N., and Singh, G. G. 2010. Meta-analysis reveals  
742 negative yet variable effects of ocean acidification on marine organisms. Ecol. Lett.  
743 13: 1419-1434.

744 Kurihara, H. 2008. Effects of CO<sub>2</sub>-driven ocean acidification on the early  
745 developmental stages of invertebrates. Mar. Ecol. Prog. Ser. 373: 275-284.

746 Kurihara, H., Asai, T., Kato, S., and Ishimatsu, A. 2008. Effects of elevated pCO<sub>2</sub> on  
747 early development in the mussel *Mytilus galloprovincialis*. Aquat. Biol. 4: 225-233.

748 Kurihara, H., Kato, S., and Ishimatsu, A. 2007. Effects of increased seawater pCO<sub>2</sub> on  
749 early development of the oyster *Crassostrea gigas*. Aquat. Biol. 1: 91-98.

750 Lewis, E., and Wallace, D.W.R. 1998. Program developed for CO<sub>2</sub> system calculations.  
751 Carbon Dioxide Information Analysis Center, Oak Ridge National Laboratory, U.S.  
752 Department of Energy

753 Martin, S., Richier, S., Pedrotti, M. L., Dupont, S., Castejon, C., Gerakis, Y., Kerros, M.  
754 E., *et al.* 2011. Early development and molecular plasticity in the Mediterranean sea

755 urchin *Paracentrotus lividus* exposed to CO<sub>2</sub>-driven acidification. J. Exp. Biol. 214:  
756 1357-1368.

757 Medakovic, D., 2000. Carbonic anhydrase activity and biomineralization process in  
758 embryos, larvae and adult blue mussels *Mytilus edulis* L. Helgol Mar Res 54:1–6.  
759 doi:10.1007/s101520050030

760 Mehrbach, C., Culberso, C., Hawley, J.E., and Pytkowic, R.M. 1973. Measurement of  
761 apparent dissociation-constants of carbonic-acid in seawater at atmospheric pressure.  
762 Limnol. Oceanogr. 18: 897–907.

763 Miller, A.W., Reynolds, A.C., Sobrino, C., and Riedel, G.F. 2009. Shellfish Face  
764 Uncertain Future in High CO<sub>2</sub> World: Influence of Acidification on Oyster Larvae  
765 Calcification and Growth in Estuaries. PLoS ONE 4(5): e5661.  
766 doi:10.1371/journal.pone.0005661

767 Morash, A. J., and Alter K., 2015. Effects of environmental and farm stress on abalone  
768 physiology: perspectives for abalone aquaculture in the face of global climate  
769 change. Reviews in Aquaculture, (2015) 7, 1–27.

770 Noisette, F., Comtet, T., Legrand, E., Bordeyne, F., Davoult, D., and Martin, S. 2014.  
771 Does Encapsulation Protect Embryos from the Effects of Ocean Acidification? The  
772 example of *Crepidula fornicata*. PLoS ONE, 9: e93021.

773 Orr, J. C., Fabry, V. J., Aumont, O., Bopp, L., Doney, S. C., Feely, R. A.,  
774 Gnanadesikan, A., *et al.* 2005. Anthropogenic ocean acidification over the twenty-  
775 first century and its impact on calcifying organisms. Nature, 437: 681-686.

776 Pan, T.-C.F., Applebaum, S.L. and Manahan, D.T., 2015. Experimental ocean  
777 acidification alters the allocation of metabolic energy. Proc. Natl. Acad. Sci. 112:  
778 4696–4701.



779 Parker, L., Ross, P., O'Connor, W., Borysko, L., Raftos, D. & Pörtner, H. 2012. Adult  
780 exposure influences offspring response to ocean acidification in oysters. *Glob.*  
781 *Change Biol.*18: 82–92.

782 Parker, L., Ross, P., O'Connor, W., Pörtner, H., Scanes, E., and Wright, J. 2013.  
783 Predicting the Response of Mollusks to the Impact of Ocean Acidification. *Biology*,  
784 2: 651-692.

785 Parker, L. M., Ross, P. M., and O'Connor, W. A. 2009. The effect of ocean acidification  
786 and temperature on the fertilization and embryonic development of the Sydney rock  
787 oyster *Saccostrea glomerata* (Gould 1850). *Glob. Change Biol.* 15: 2123-2136.

788 Parker, L. M., Ross, P. M., and O'Connor, W. A. 2010. Comparing the effect of  
789 elevated  $p\text{CO}_2$  and temperature on the fertilization and early development of two  
790 species of oysters. *Mar. Biol.* 157: 2435-2452.

791 Przeslawski, R., Byrne, M., and Mellin, C. 2015. A review and meta-analysis of the  
792 effects of multiple abiotic stressors on marine embryos and larvae. *Glob. Change*  
793 *Biol.* 21: 2122-2140.

794 R Development Core Team. 2014. R: A language and environment for statistical  
795 computing. R Foundation for Statistical Computing, Vienna, Austria.

796 Ramesh, K., Hu, M.Y., Thomsen, J., Bleich, M. and Melzner, F., 2017. Mussel larvae  
797 modify calcifying fluid carbonate chemistry to promote calcification. *Nat.*  
798 *Commun.* 8: 1–8.

799 Riebesell, U., Fabry, V. J., Hansson, L., and Gattuso, J.-P. 2010. Guide to Best Practices  
800 for Ocean Acidification Research and Data Reporting (Publications Office of the  
801 European Union).

802 Ross, P. M., Parker, L., O'Connor, W. A., and Bailey, E. A. 2011. The Impact of Ocean  
803 Acidification on Reproduction, Early Development and Settlement of Marine  
804 Organisms. *Water*, 3: 1005-1030.

805 Rühl, S., Calosi, P., Faulwetter, S., Keklikoglou, K., Widdicombe, S. and Queirós,  
806 A.M., 2017. Long-term exposure to elevated pCO<sub>2</sub> more than warming modifies  
807 early-life shell growth in a temperate gastropod. ICES J. Mar. Sci. 74: 1113–1124.

808 Talmage, S.C., and Gobler, C.J., 2009. The effects of elevated carbon dioxide  
809 concentrations on the metamorphosis, size, and survival of larval hard clams  
810 (*Mercenaria mercenaria*), bay scallops (*Argopecten irradians*), and Eastern oysters  
811 (*Crassostrea virginica*). Limnol. Oceanogr. 54:2072–2080

812 Talmage, S. C., and Gobler, C.J. 2010. Effects of past, present and future ocean carbon  
813 dioxide concentrations on the growth and survival of larval shellfish. Proc. Natl.  
814 Acad. Sci. USA 107: 17246–172.

815 Thomsen, J., Haynert, K., Wegner, K. M., and Melzner, F., 2015. Impact of seawater  
816 carbonate chemistry on the calcification of marine bivalves. Biogeosciences, 12,  
817 4209–4220,.

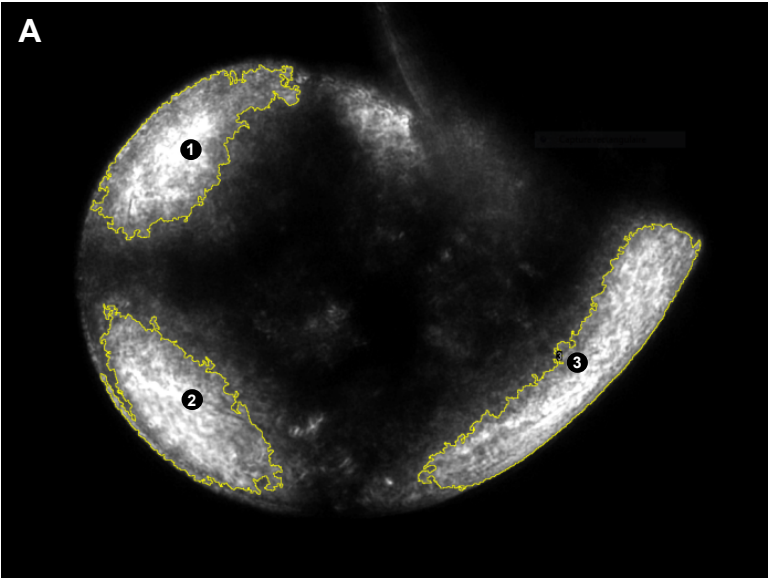
818 Timmins-Schiffman, E., O'Donnell, M. J., Friedman, C.S., and Roberts, S. B. 2013.  
819 Elevated pCO<sub>2</sub> causes developmental delay in early larval Pacific oysters,  
820 *Crassostrea gigas*. Mar. Biol. 160: 1973-1982.

821 Travers, M.-A., Basuyaux, O., Le Goïc, N., Huchette, S., Nicolas, J.-L., Koken, M., and  
822 Paillard, C. 2009. Influence of temperature and spawning effort on *Haliotis*  
823 *tuberculata* mortalities caused by *Vibrio harveyi*: an example of emerging vibriosis  
824 linked to global warming. Glob. Change Biol. 15: 1365-1376.

825 Waldbusser, G.G., Hales, B., Langdon, C. J., Haley, B. A., Schrader, P., Brunner, E. L.,  
826 Gray, W. P., *et al.* 2015. Saturation-state sensitivity of marine bivalve larvae to  
827 ocean acidification. Nat. Clim. Change 5:273–280.

- 828 Watson, S.-A., Southgate, P. C., Tyler, P. A., and Peck, L. S. 2009. Early Larval  
829 Development of the Sydney Rock Oyster *Saccostrea glomerata* Under Near-Future  
830 Predictions of CO<sub>2</sub>-Driven Ocean Acidification. J. Shellfish Res. 28: 431-437.
- 831 Weiss, I.M., Lüke, F., Eichner, N., Guth, C and Clausen-Schaumann, H., 2013. On the  
832 function of chitin synthase extracellular domains in biomineralization. J. Struct.  
833 Biol. 183 (2): 216-225.
- 834 Widdicombe, S., and Spicer, J. I. 2008. Predicting the impact of ocean acidification on  
835 benthic biodiversity: What can animal physiology tell us? J. Exp. Mar. Biol. Ecol.  
836 366: 187-197.
- 837 Wittmann, A.C., and Pörtner, H-O. 2013. Sensitivities of extant animal taxa to ocean  
838 Acidification. Nat. Clim. Change 3: 995–1001.
- 839 Zippay, M.L., and Hofmann, G.E. 2010. Effect of pH on gene expression and thermal  
840 tolerance of early life history stages of red abalone *Haliotis rufescens*. J. Shellfish  
841 Res. 29: 429-439.

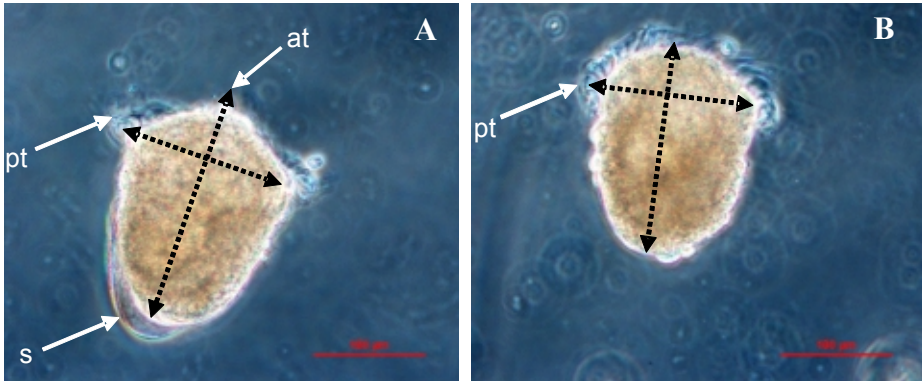
Supplementary Figure S1:



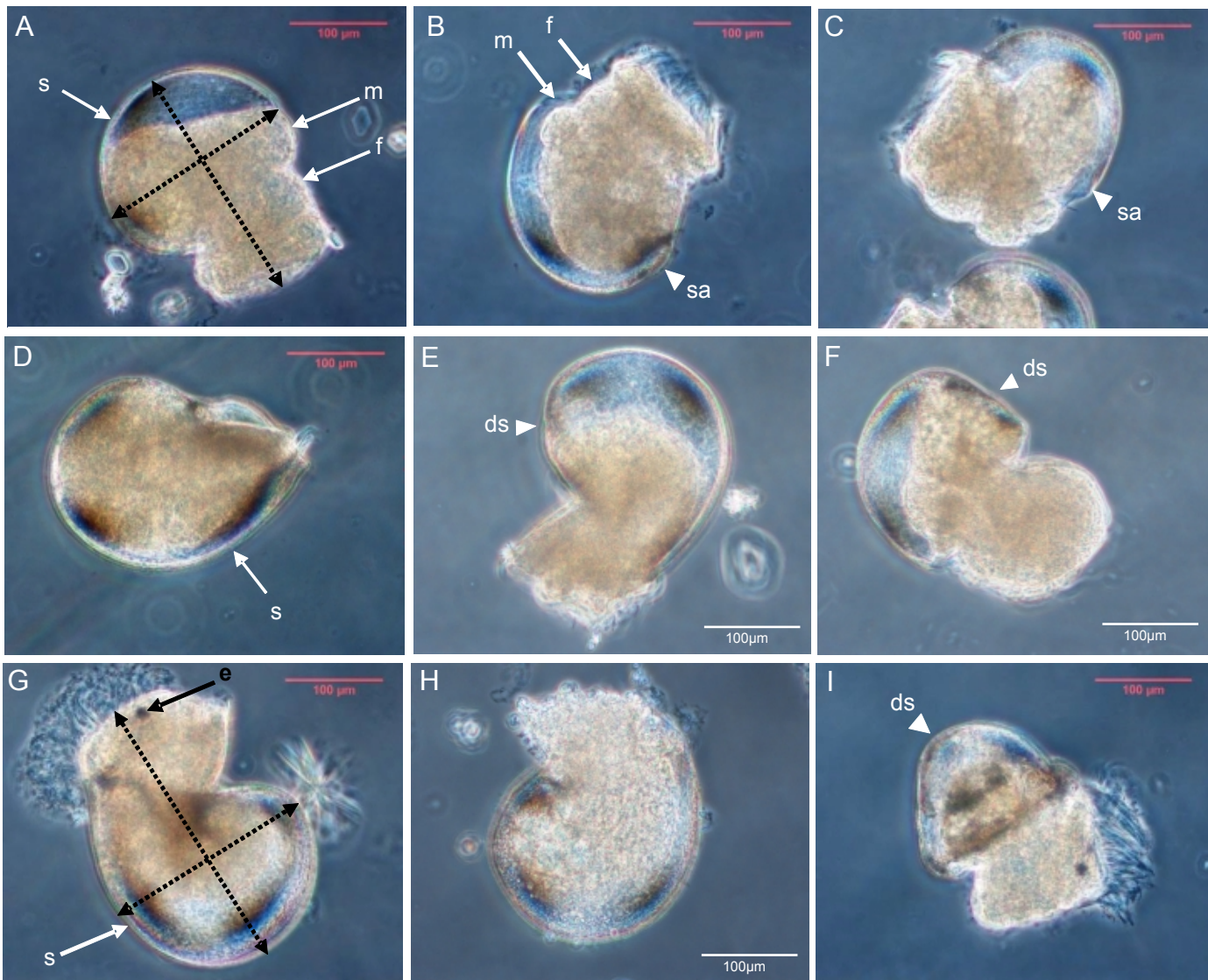
**B**

ROI	Grey-scale level
1	97,977
2	98,022
3	100,713
<b>Mean</b>	<b>98,90</b>
SD	1,28

**Figure 1**



**Figure 2**



**Figure 3**

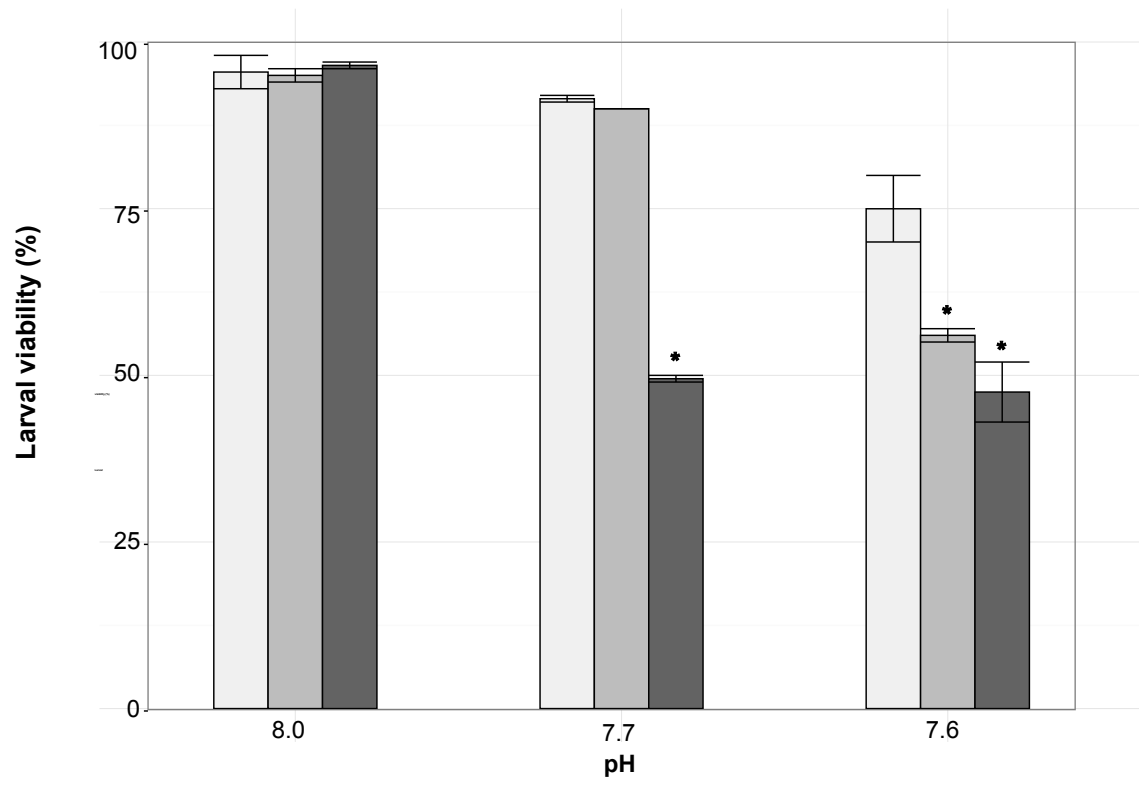
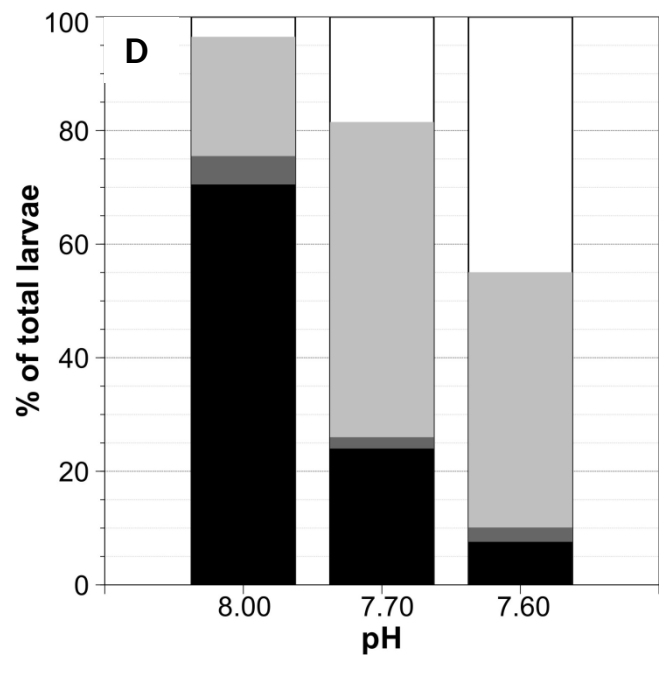
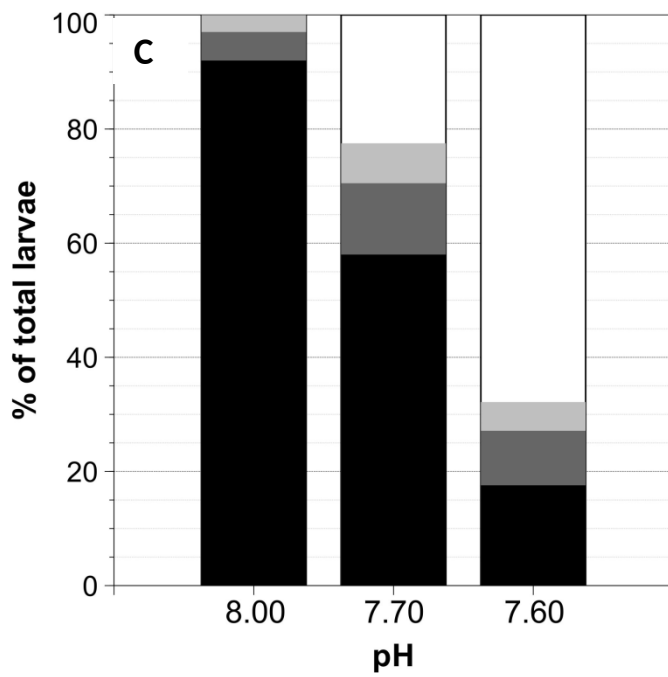
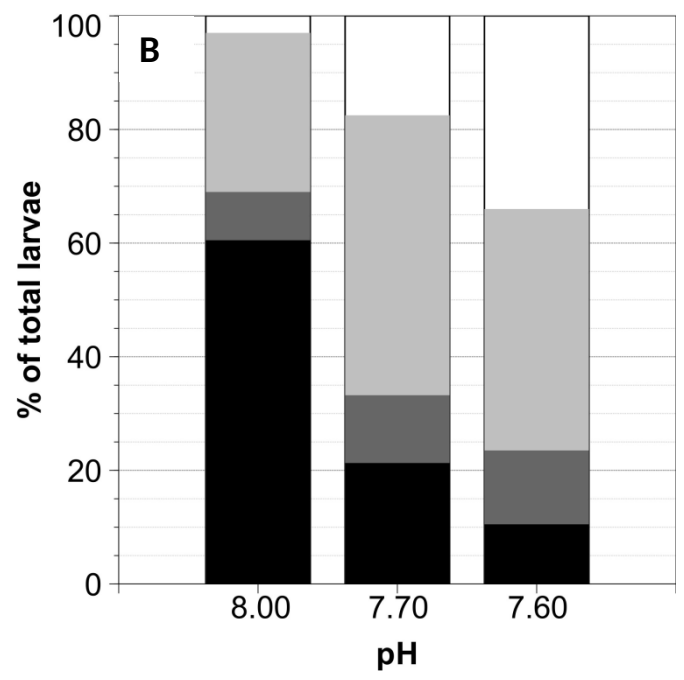
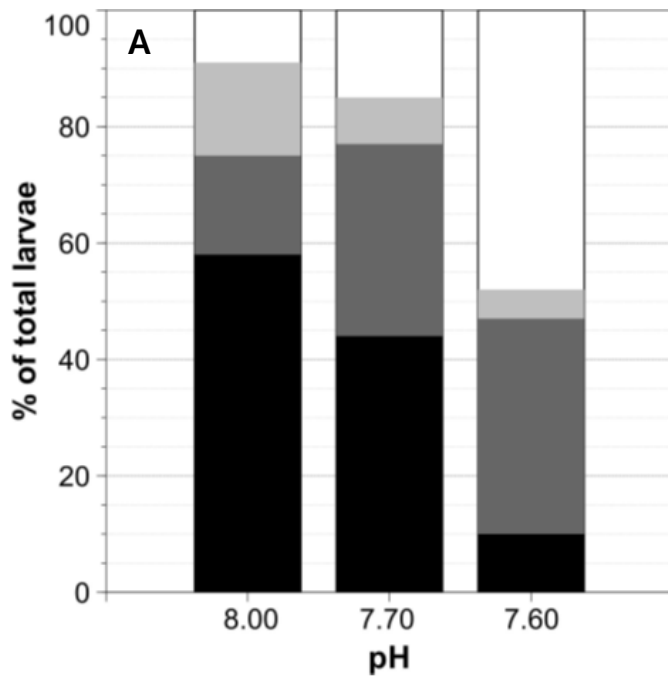


Figure 4





**Figure 5**

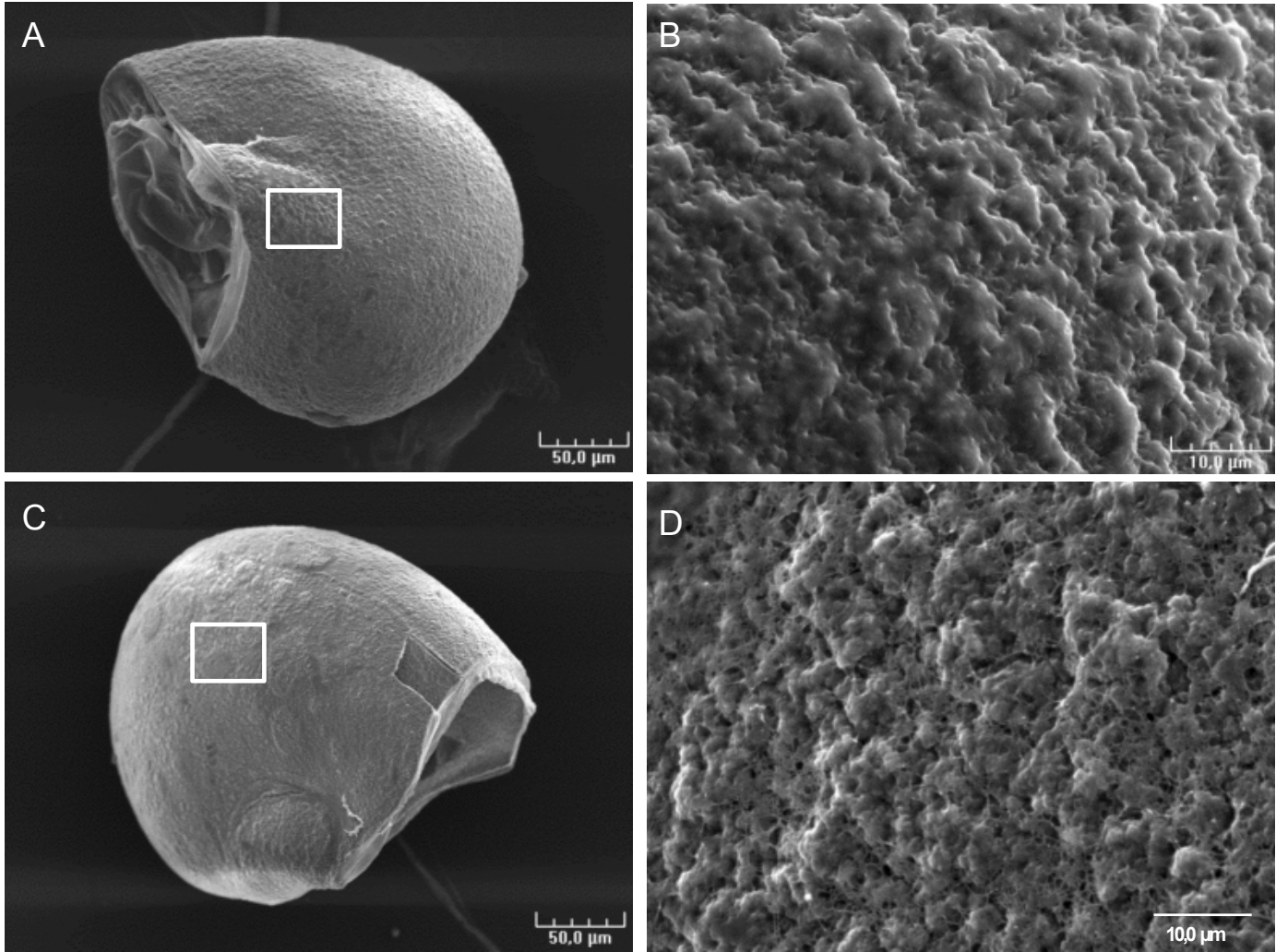
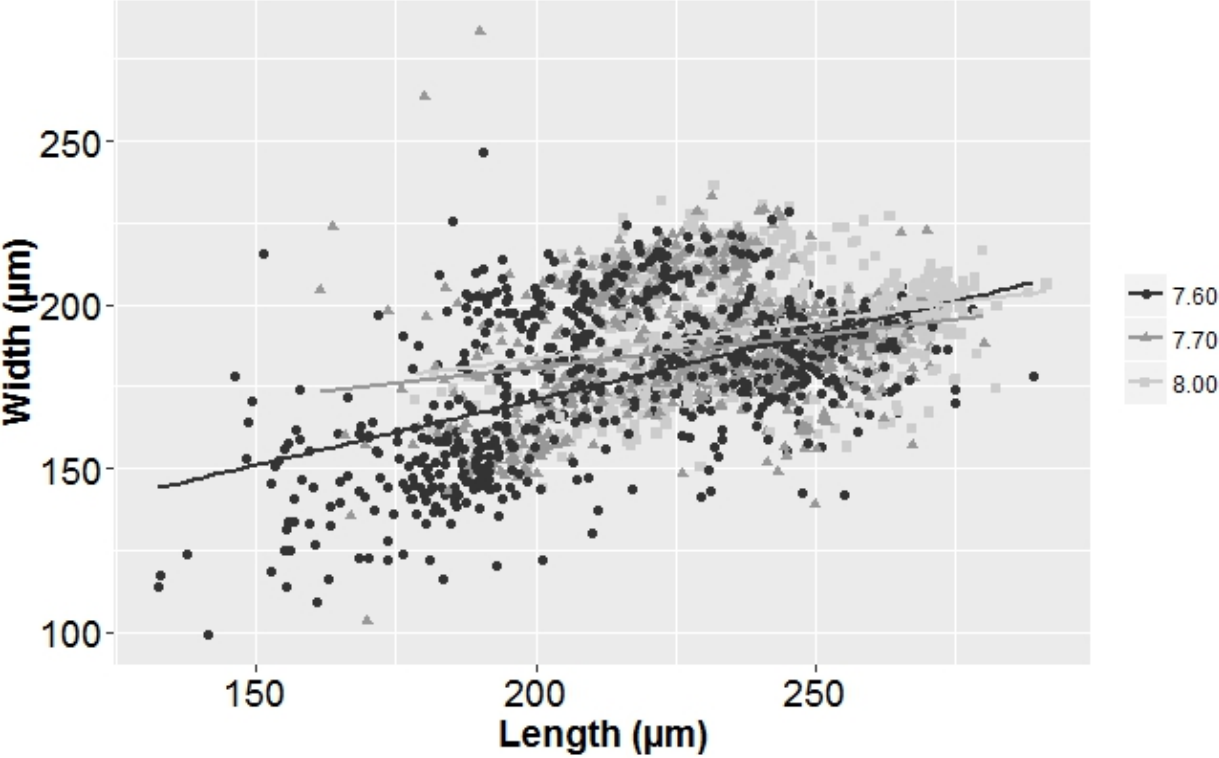
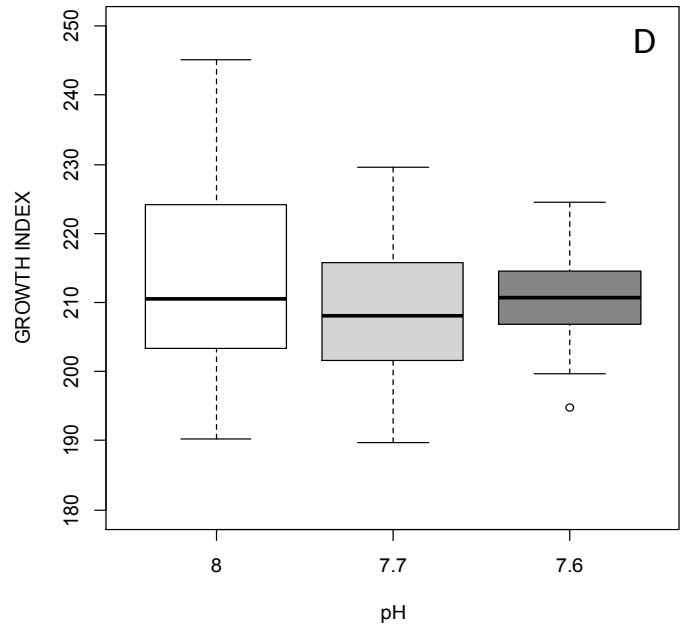
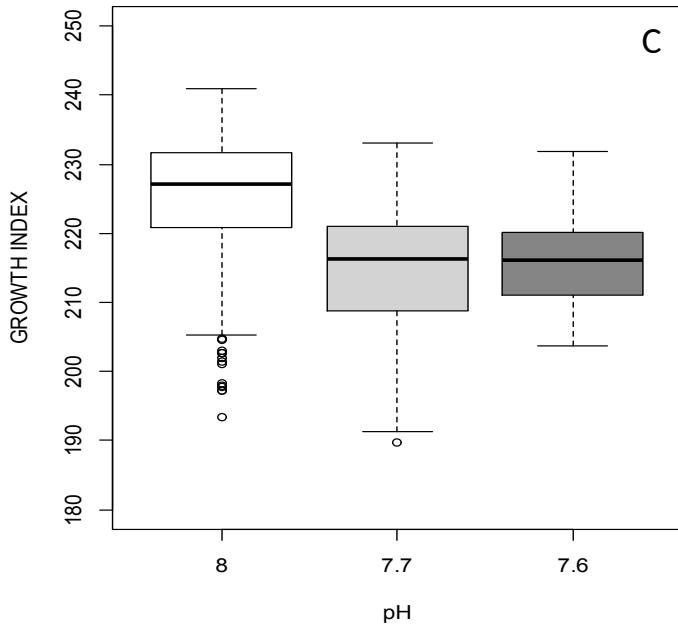
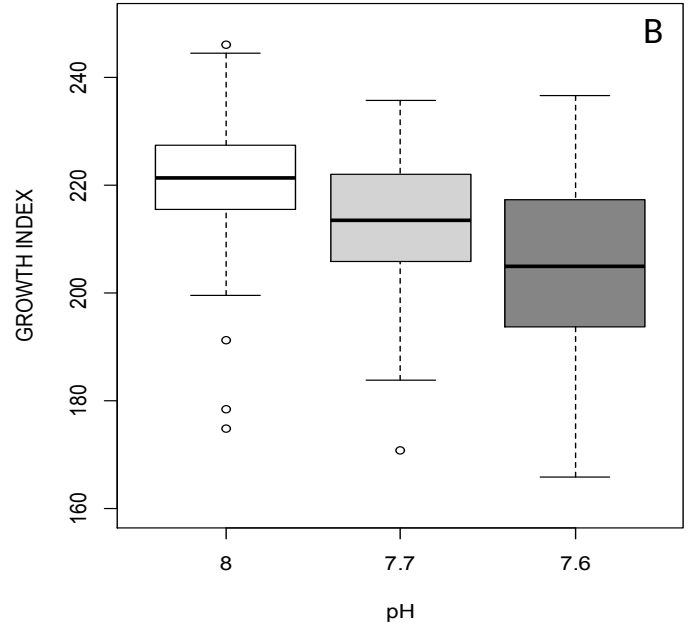
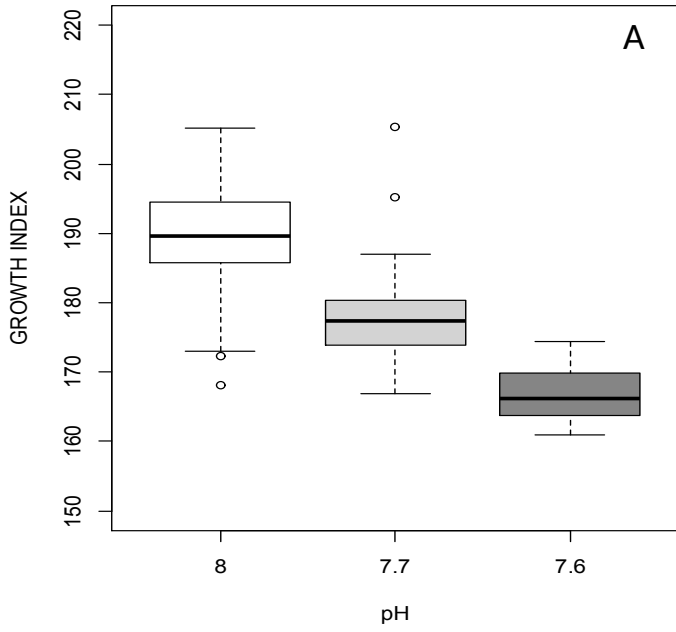


Figure 6



**Figure 7**



**Figure 8**

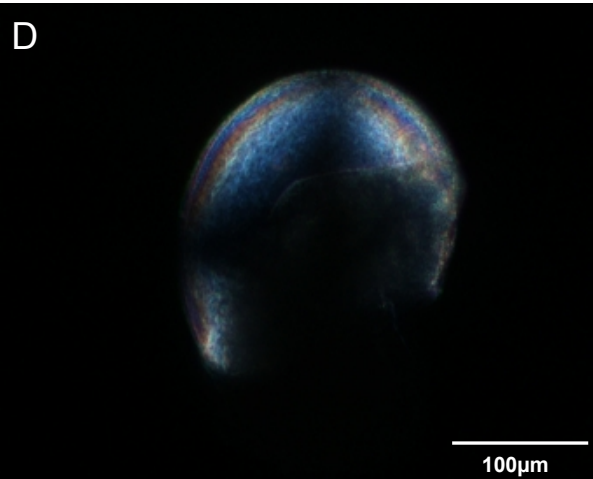
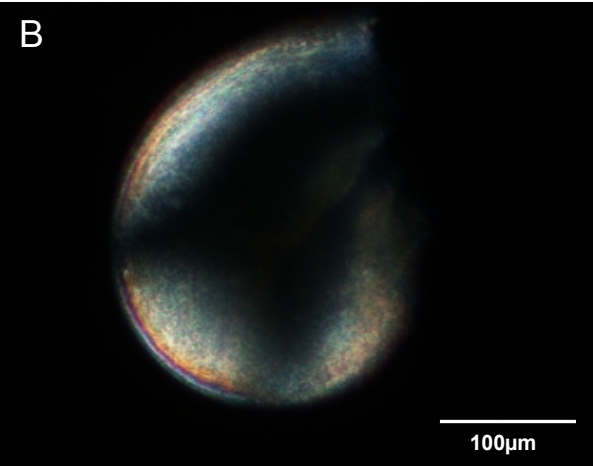
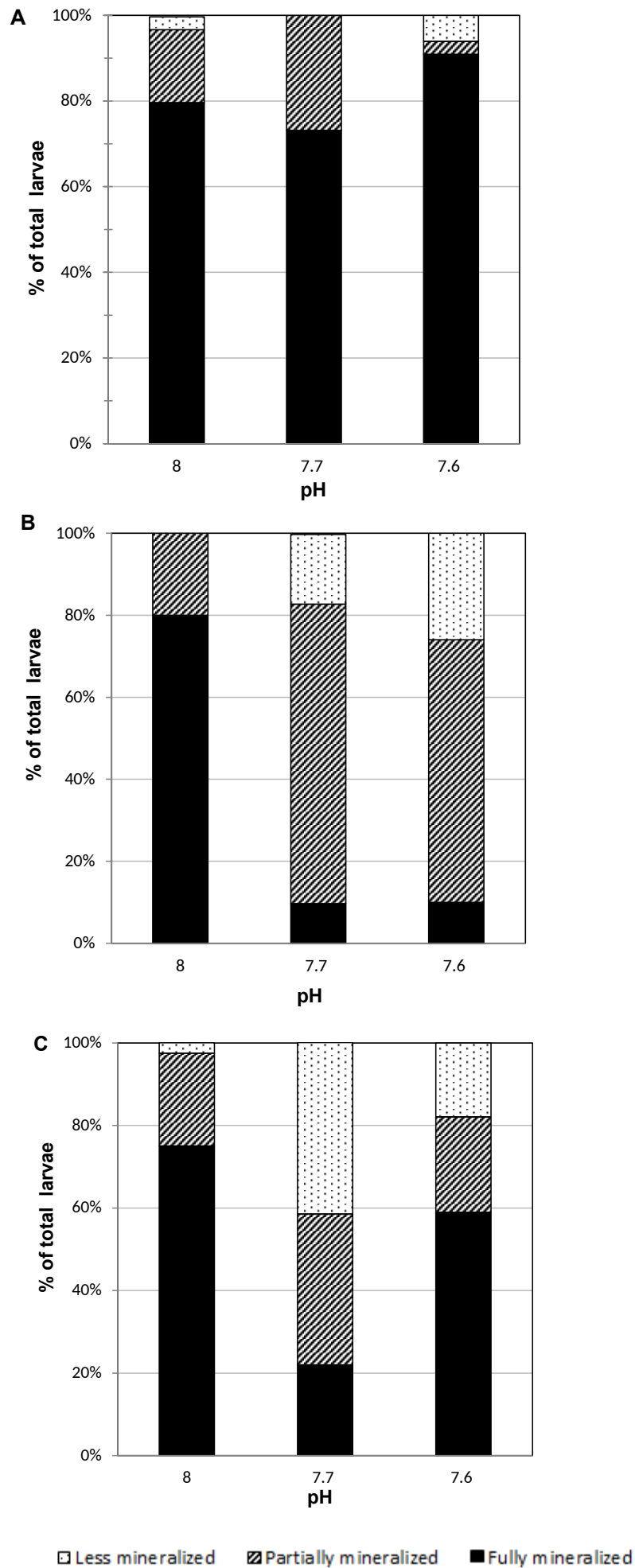


Figure 9



**Table 1.**

Nominal pH	pHT	pCO <sub>2</sub> (µatm)	DIC (µmol/kg <sup>-1</sup> )	HCO <sub>3</sub> <sup>-</sup> (µmol/kg <sup>-1</sup> )	CO <sub>3</sub> <sup>2-</sup> (µmol/kg <sup>-1</sup> )	Ω ar	Ω calc
8.1	8.00 ± 0.002	460 ± 3	2119 ± 2	1940 ± 3	163 ± 1	2,47 ± 0,01	3,8 ± 0,02
7.7	7.68 ± 0,001	1055 ± 3	2254 ± 1	2132 ± 0,05	85,4 ± 0,3	1,30 ± 0,01	2 ± 0,01
7.6	7.58 ± 0,003	1331 ± 10	2286 ± 2	2170 ± 2	70,1 ± 0,6	1,06 ± 0,01	1,6 ± 0,01

**Table 2.**

Treatment (pH)	Rho	p-value
8.0	0.2842896	<0.001
7.7	0.1919181	<0.001
7.6	0.4593034	<0.001

**Table 3.****A. Repeated measures ANOVA**

Effect between subjects	df	MS	F-ratio	<i>p</i> -value
pH	2	357.3	33.1	<b>0.009</b>
Error	3	10.8		

Effect within subjects	df	MS	F-ratio	<i>p</i> -value
Time	3	1998	141.6	<b>&lt;0.001</b>
Time * pH	6	38.0	2.7	0.088
Error	9	14.1		

**B. Multiple comparisons**on factor time: *p*-value

20h vs 30h	<b>0.002</b>
20h vs 48h	<b>&lt;0.001</b>
20h vs 96h	<b>&lt;0.001</b>
30h vs 48h	0.159
30h vs 96h	0.418
48h vs 96h	<b>0.009</b>

on factor pH: *p*-value

7.6 vs 7.7	0.105
7.6 vs 8.0	<b>0.008</b>
7.7 vs 8.0	<b>0.031</b>



**Table 4.****A. Homogeneity  $\chi^2$  test**

<b>Development Time</b>	<b>Chi-sq</b>	<b>df</b>	<b><i>p</i>-value</b>
30h	9.28	4	0.055
48h	67.53	4	<b>&lt;0.001</b>
96h	28.48	4	<b>&lt; 0.001</b>

**B. Pair wise Wilcoxon rank sum test**

	48h	96h
<b>pH factor</b>	<b><i>p</i>-value</b>	<b><i>p</i>-value</b>
7.7 vs 8.0	<b>&lt;0.001</b>	<b>&lt;0.001</b>
7.6 vs 8.0	<b>&lt;0.001</b>	0.073
7.6 vs 7.7	0.49	<b>0.0016</b>