Morphological changes in branchial mitochondria-rich cells of the teleost *Paralichthys olivaceus* as a potential indicator of CO₂ impacts

Masahiro Hayashi¹³, Takashi Kikkawa²⁴ and Atsushi Ishimatsu¹*

¹Institute for East China Sea Research, Nagasaki University, 1551-7 Tairamachi, Nagasaki 851-2213, Japan

²Central Laboratory, Marine Ecology Research Institute, Onjuku, Chiba 299-5105, Japan

*Corresponding author. Tel: +81-95-850-7312; Fax: +81-95-840-1881.

E-mail address: a-ishima@nagasaki-u.ac.jp (A. Ishimatsu).

Present addresses

³Demonstration Laboratory, Marine Ecology Research Institute, 4-7-17 Arahama, Kashiwazaki, Niigata 945–0017, Japan

⁴Head Office, Marine Ecology Research Institute, 347 Yamabuki-cho Shinjuku-ku, Tokyo 162-0801, Japan
Abstract

We studied the morphological and biochemical changes of mitochondria-rich cells (MRCs) of a demersal teleost, *Paralichthys olivaceus*, during exposure to 0.98, 2.97 and 4.95 kPa pCO₂. The apical opening area of MRCs increased 2.2 and 4.1 times by 24 hr exposure to 2.97 and 4.95 kPa pCO₂, respectively, while the cross-sectional area or density of MRCs did not change. Gill Na⁺/K⁺-ATPase activity more than doubled at 72 hr and then returned to the pre-exposure level at 168 hr in 0.98 kPa pCO₂, while it increased 1.7 times at 24 hr at 4.95 kPa. These results indicate that the apical opening area of MRCs and the gill Na⁺/K⁺-ATPase activity may be used as an indicator of acute (up to 72 hr), but not chronic, impacts of high (> 1 kPa) seawater CO₂ conditions in *P. olivacues*. Limitations of those parameters as indices of CO₂ impacts are discussed.

Keywords: carbon dioxide; CO₂ leakage; environmental hypercapnia; mitochondria-rich cell; Na⁺/K⁺-ATPase activity; *Paralichthys olivaceus*
1. Introduction

Geological storage of CO₂ has received greater attention as a potential mitigation measure against global warming, with which CO₂ is injected into different types of geological formations such as deep saline-water saturated formations, depleted oil and gas fields, and un-mineable coals (CSLF, 2011; IPCC, 2005; Pires et al., 2011). Several projects of CO₂ geological storage have already been operating and many more are being planned (CSLF, 2011). The CO₂ is being monitored after injection in the storage sites, but potential leakage of CO₂ and its ecological and environmental consequences have always been a concern against large-scale implementation (Upham and Roberts, 2011), even though the probability of the leakage from the CO₂ stored in a geological formation is considered to be extremely low (IPCC, 2005).

Monitoring CO₂ leakage is more problematic when CO₂ is stored under the seabed, and novel sensors and devices are being developed to improve the precision of detecting changes in seawater CO₂ and pH (Shitashima et al., 2012). Biological monitoring could be used to complement those engineering approaches. Among different marine
organisms, fishes have been most extensively investigated with respect to biological responses to environmental perturbations including elevations in water CO₂ concentrations, making them candidates for biological monitoring of CO₂ leakage. On the other hand, drawbacks of using fishes for this purpose include the relatively high mobility (particularly pelagic species) and thus possible avoidance from a leakage site and their higher tolerance to CO₂ than most marine invertebrates (Ishimatsu et al., 2005, 2008).

Fishes have well developed pH-regulating mechanisms, which largely rely upon ion transfer processes across the gills by mitochondria-rich cells (MRCs, Evans et al., 2005; Heisler, 1986; Perry and Gilmour, 2006). When fishes are exposed to environmental hypercapnia (elevated CO₂ conditions), CO₂ will diffuse into the body fluids along pCO₂ gradient mainly through the gills, arterial blood pH decreases rapidly (within minutes) but it then recovers towards pre-exposure levels within several hours (in marine species) or longer (in many freshwater fishes). During the pH recovery, plasma Cl⁻ decreases with an almost equimolar increase of plasma HCO₃⁻ in both seawater and
freshwater teleosts (Brauner and Baker, 2009; Fivelstad, 2012; Hayashi et al., 2004; Larsen and Jansen, 1997; Toews et al., 1983; Elasmobranchs appear to employ different acid-base regulatory mechanisms, because plasma Cl⁻ ion concentrations usually does not or only slightly decrease during pH compensation from hypercapnic acidosis. See Ishimatsu et al., 2005). The mechanisms for ion regulation in fishes have been intensively investigated and are discussed in many extensive review papers (Dymowska et al., 2012; Evans et al., 2005; Hwang et al., 2011; Kaneko et al., 2008). In comparison, relatively few studies examined morphological changes in MRCs in response to CO₂ exposure; Cameron and Iwama (1987) found a 30% increase in number and a 75% increase in the apical opening area of MRCs after a 4-day exposure up to 8% CO₂ (7.84 kPa pCO₂) in the freshwater channel catfish. In contrast, Goss et al. (1992) demonstrated a 90% reduction in the apical opening area of MRCs and a 50% reduction in cell density, which was in concert responsible for a 95% reduction in MRC fractional surface area (% in a unit epithelial area), in the congeneric brown bullhead exposed to 2% CO₂ (1.96 kPa) for 2 days. Goss and Perry (1993) reported a similar, but much less
pronounced decrease in MRC fractional area in hypercapnia rainbow trout (1% CO₂, 4 days). More recently, Baker et al. (2009) reported a significant decrease in MRC apical opening area, density and fractional surface area in white sturgeon exposed to 1.53% CO₂ (1.50 kPa). Thus, there is a possibility that morphological, biochemical and/or molecular characteristics of MRCs are useful indicators of CO₂ impacts on fishes.

The present study was aimed to examine this possibility by investigating the effects of elevated seawater CO₂ levels on the morphology of branchial MRCs and the activity of gill Na⁺/K⁺-ATPase, in the Japanese flounder, Paralichthys olivaceus. The Na⁺/K⁺-ATPase is the enzyme that provides an electrochemical gradient that drives ion fluxes through other transport pathways in MRC (Evans, 2005; Hwang et al., 2011) and therefore commonly used as an index of MRC activity.

2. Materials and methods

2.1 Experimental fish
Japanese flounder with mean (± S.D.) weight of 116 ± 16 g (n = 84) were purchased from two local aquaculture farms, and kept in an indoor fiberglass tank (1,000 l capacity) with filtered, recirculating well-aerated natural seawater at 20°C. Seawater was pumped at 20 m depth from the coast near the Institute for East China Sea Research, Nagasaki University, where all experiments were carried out. The fish would not accept food for extended periods under captivity and therefore were used within 2 weeks after purchase.

2.2 Experimental protocol

The experimental setup consisted of two fish chambers (internal dimension; 475 × 240 × 45 (H) mm), a water reservoir (ca. 50 l capacity) and two gas equilibration columns (90 (internal diameter) × 795 (H) mm). The total volume of the recirculating water was ca. 65 l. Six individuals were placed in each chamber supplied with continuous flow of well-aerated seawater (20°C, salinity 35 ppt) at a rate of 4 l min⁻¹. Fresh seawater was continuously supplied into the reservoir at a rate of 0.5-1 l min⁻¹ to
avoid buildup of waste substances in water. After 1 day acclimation, the fish were
exposed to normocapnia (water equilibrated with air) or different levels of hypercapnia
(water equilibrated with a gas mixture containing 1%, 3% or 5% CO₂ in air; 1% CO₂ =
0.98 kPa pCO₂ under the barometric pressure of 101.3 kPa, at 20°C). The gas mixtures
(flow rates, 6 l min⁻¹) were prepared with a gas-mixing flowmeter (GF-3/MP, Cameron
Instruments, USA). Hayashi et al. (2004) have shown that seawater pH would attain
new stable levels within 1 hour of gas bubbling at the same flow rates of gas and at the
same recirculation rate of seawater as used in this study. The measured seawater pH
values (7.01 at 0.98 kPa pCO₂ (1% CO₂), 6.41 at 2.97 kPa (3% CO₂), and 6.18 at 4.95
kPa (5% CO₂), control seawater pH 8.18) were nearly identical with the value predicted
from the pCO₂ of the gas mixture used, temperature, salinity, and alkalinity. Thus, we
presumed that nearly complete equilibration was attained between partial pressures of
the gas mixtures and seawater in this study. Tissue samples were taken at 0, 24, 72 and
168 hr (except in the 4.95 kPa pCO₂ treatment in which survival rates were 100% at 24
hr but 0% at 72 hr) at each exposure level. No mortality occurred in the other treatments.
Due to the limited number of fish chambers, exposure periods of each fish group were staggered.

2.3 Tissue sampling for morphological observations

The gills on the eyed (top) side were excised under benzocaine anesthesia (ethyl-m-aminobenzoate) at a final concentration of 0.1%. The excised gills were washed rapidly three times in cold saline (0.9% NaCl solution). For scanning-electron microscopy (SEM), the first gill arch was fixed in 4% paraformaldehyde (PFA)-2% glutaraldehyde in 0.1 M phosphate buffer (PB) solution (pH 7.4) for 1 day, then washed overnight in 0.1 M PB (containing 10% sucrose) solution, and preserved in 70% ethanol. Fixation, washing and preservation were all done at 4°C. Fish exposed to the 2.97 kPa pCO₂ were used only for SEM analysis. For confocal laser-scanning microscopy (LSM), the second gill arch was fixed in 4% PFA in 0.1 M PB (pH 7.4) for 1 day at 4°C and treated in the same way as for the SEM observations. For the measurement of gill Na⁺/K⁺-ATPase activity, the third gill arches were stored in 1 ml homogenizing solution.
(250 mM sucrose, 6 mM EDTA-2Na and 20 mM imidazole) of pH 6.8 at –80°C until use.

2.4 SEM determination of the apical opening area of MRCs

A portion of the first arch (containing several gill filaments) was severed from the central section of the trisected first gill arches, dehydrated in ethanol, and immersed in 2-methyl-2-propanol. The gill filaments were carefully excised, freeze-dried (JFD-310, JEOL, Japan), mounted on specimen stubs, and coated with gold in an ion sputter (JFC-1200, JEOL, Japan). Then, the apical opening area of MRCs from three randomly selected filaments was determined with a scanning electron microscope (JSM-5310LV, JEOL, Japan) at a magnification of 3500×. Observation sites were two non-contiguous fields in a filament randomly selected from a flat region of the afferent-vascular edge, which lacked secondary gill lamellae. A total of six micrographs per individual was taken and saved as TIFF files. The apical opening area of MRCs was determined using a graphics tablet system (Intuos2 i-420, Wacom, Japan) with an image analyzing software.
Mean apical opening area (µm²) of individual fish (n = 6) was obtained from all the apical openings in the six micrographs.

2.5 LSM determination of the cross-sectional area and density of MRCs

Tissues of the second gill arches were processed according to the method of Katoh et al. (2000), and the cross-sectional area and density of MRCs were observed with a confocal laser-scanning microscope. For the detection of MRCs in the whole-mount preparations of the gill filaments, the tissues were immunocytochemically stained with an antibody specific for Na⁺/K⁺-ATPase (Ura et al., 1996). The specific antibody was affinity-purified and labeled with Alexa fluor 488 (A-11034, Molecular Probes, USA) as a fluorescent marker. Before the whole-mount immunocytochemistry, a portion of the second gill arch was severed as described for SEM, and was treated as follows: (1) washed in distilled water and then in 0.01 M phosphate-buffered saline (PBS, pH 7.2); (2) incubated overnight at 4°C with anti-Na⁺/K⁺-ATPase serum diluted 1:1000 v/v with T-PBS (PBS containing 0.05% Triton X-100, 10% normal goat serum, 0.1% bovine...
serum albumin, 0.02% keyhole limpet hemocyanin and 0.01% sodium azide); (3) rinsed in 0.01 M PBS; (4) incubated overnight at 4°C with Alexa fluor diluted 1:1000 v/v with T-PBS; (5) rinsed in 0.01 M PBS; (6) incubated with Propidium iodide (P-3566, Molecular Probes, USA) diluted 1:20 v/v with T-PBS for 20 min at room temperature (for staining nucleic acid); and (7) rinsed in 0.01 M PBS. Subsequently, the gill filaments were incised from a segment, placed in a chamber slide with a cover slip over, and observed with a confocal laser-scanning microscope (LSM 510 META-Ver. 3.0, Carl Zeiss, Germany). The 488 nm line of an argon-ion laser and the 543 nm line of a helium/neon-ion laser were used as the excitation wavelength. Observations were made from three non-contiguous fields randomly selected from the surface of the trailing edge of each of the three excised filaments (nine digital images per individual). The obtained images were saved as above. The cross-sectional area and density of immunopositive MRCs were measured using an image-processing software (HGK-ST, HOGA, Japan). Mean cross-sectional area ($\mu m^2$) of individual fish ($n = 6$) was obtained from all the cells in the nine digital images. For determination of MRC density, an area was
randomly selected from a digital image and all the MRCs present were counted except
for the uncompleted cell outlines found at the edge of the predefined boundary. Mean
cell density of individual fish (n = 6) was obtained from nine digital images and was
expressed as cells per mm².

2.6 Measurement of gill Na⁺/K⁺-ATPase activity

Na⁺/K⁺-ATPase activity was measured as described by Soyano et al. (1988). The
stored gill arches were rapidly thawed and several gill filaments were removed. The
filaments were placed in a microtube containing fresh homogenizing solution,
homogenized on ice, and centrifuged at 12,000 × g for 20 min at 4°C. The supernatant
was assayed for Na⁺/K⁺-ATPase activity and protein content. The gill homogenate (40
µl) was added into the tubes containing 160 µl of the assay buffer (160 µl; 250 mM
imidazole, 12.5 mM Na₂ATP, 337.5 mM NaCl, 162.5 mM KCl and 50 mM MgCl₂) with or
without 2.5 mM ouabain, and incubated at 37°C for 20 min. The reaction was terminated
by addition of 4 ml of ice-cold Iron-TCA (100 g l⁻¹ TCA, 10 g l⁻¹ Thiourea and 30 g l⁻¹
Mohr’s salt. Finally, 500 µl of a coloring reagent (90 ml l⁻¹ H₂SO₄ and 44 g l⁻¹ \((\text{NH₄})₆\text{Mo}_7\text{O}_{24}·4\text{H}_2\text{O})\) was added to the tubes to determine the concentration of inorganic phosphate of the supernatant using the method of Goldenberg and Fernandoz (1966). The protein content of the sample was determined with a commercial kit (Bio-Rad Protein Assay, Bio-Rad, USA).

2.7 Statistics

All values are expressed as means ± standard deviation (S.D.). Statistical comparisons among sampling time were made using one-way analysis of variance (ANOVA) followed by Tukey’s test (Sigmastat 2.0, Jandel, USA).

3. Results

The scanning electron micrographs of gill filaments in the Japanese flounder exposed to hypercapnic seawater are shown in Fig. 1 (A-D, 2.97 kPa pCO₂; E and F, 4.95 kPa).
The apical opening area of MRCs did not change significantly in control (normocapnia) and at 0.98 kPa pCO₂, but increased 2.2 (p < 0.05; Tukey’s test) and 4.1 (p < 0.05; Tukey’s test) times by 24 hr exposure to 2.97 and 4.95 kPa pCO₂, respectively (Fig. 2). The area subsequently decreased but still remained significantly high than the 0 hr value by 72 hr at 2.97 kPa (Fig. 2). Neither the cross-sectional area nor density of MRCs changed significantly at any CO₂ treatment until 168 hr of exposure (Fig. 3). Gill Na⁺/K⁺-ATPase activity more than doubled (p < 0.05; Tukey’s test) at 72 hr and then decreased at 0.98 kPa pCO₂, while it increased 1.7 times (p < 0.05; Tukey’s test) at 24 hr during exposure to 4.95 kPa pCO₂ (Fig. 4).

4. Discussion

The results demonstrated that the surface morphology of branchial MRCs and the gill Na⁺/K⁺-ATPase responded significantly to the elevations of seawater CO₂ in *Paralichthys olivaceus*. To our knowledge, this is the first demonstration of the MRC
morphology in response to environmental hypercapnia in a marine teleost. In comparison, two previous studies reported conflicting results on the effect of environmental hypercapnia on the apical opening area of MRCs in two closely related freshwater teleosts. Namely, Cameron and Iwama (1987) found a 30% increase in number and a 75% increase in the apical opening area of MRCs after a 4-day stepwise exposure up to 8% CO₂ (7.84 kPa pCO₂) in the channel catfish *Ictalurus punctatus*, whereas Goss et al. (1992) demonstrated a 90% reduction in the apical opening area of MRCs in the congeneric brown bullhead *I. nebulosus* exposed to 2% CO₂ (1.96 kPa) for 2 days. In agreement with the findings by Goss et al. (1992), Baker et al. (2009) recently reported significant reductions in the apical opening area, density and fractional area (%) of a unit epithelial area) of MRCs in the freshwater white sturgeon *Acipenser transmontanus* exposed to 1.53% CO₂ (1.50 kPa). Goss and Perry (1993) also reported a similar, but much less pronounced decrease in MRC fractional area in rainbow trout *Oncorhynchus mykiss* exposed to 1% CO₂ for 4 days. Morphological changes in the apical opening area of MRCs can occur rapidly (in response to salinity change) as
shown for the mudskipper (*Periophthalmus modestus*) (within 30 min, Sakamoto and Ando, 2002) and the silver sea bream (*Sparus sarba*) (in 6 hr, Kelly and Woo, 1999). Thus, there is a possibility that our 24-hr sampling interval failed to detect rapid MRC changes possibly occurred within 24 hr of hypercapnia.

In contrast to the possibly variable morphological responses of the MRCs to elevated ambient CO$_2$, changes in plasma ion concentration during hypercapnia appear to be highly consistent in teleosts irrespective of environmental salinities (freshwater or seawater): the plasma Cl$^-$ concentration decreases in a CO$_2$-concentration-dependent manner with often equimolar increases in HCO$_3^-$, whereas plasma Na$^+$ concentration is only marginally affected (Baker et al., 2009; Larsen and Jansen, 1997; Toews et al., 1983; Truchot, 1987) unless ambient pCO$_2$ increases to lethal levels (Hayashi et al., 2004). These results on plasma ionic changes lend support to the supposition that a Cl$^-$-modulating mechanism plays a predominant role in acid-base regulation during hypercapnic pH compensation in both marine and freshwater teleosts, which also agrees with the data from conventional flux studies (freshwater rainbow trout, Perry et al.,
The observed reduction of MRC apical opening area in the freshwater brown bullhead was interpreted to be a mechanism to suppress Cl⁻ uptake/HCO₃⁻ extrusion by restricting the functional surface area with overlying adjacent pavement cells, which would lead to a buildup of HCO₃⁻ and a decrease in Cl⁻ in the body fluids (Goss et al., 1992, 1995). If ion fluxes through MRCs are also modulated through an extension/retreat of adjacent pavement cells in a marine teleost, then one would predict that the apical opening areas of MRCs are enlarged when a marine teleost is subjected to an elevation of seawater pCO₂, because marine teleosts must actively extrude Cl⁻ (and Na⁺) against electrochemical gradients (Evans et al., 2005). The present results agree with this prediction. The HCO₃⁻ substitution study by Esbaugh et al. (2012) indicated that the seawater concentration of HCO₃⁻ ions significantly influenced pH recovery of fish under hypercapnia, again suggesting a predominant role of the Cl⁻/HCO₃⁻ exchange mechanism in acid-base regulation in marine teleosts. On the other hand, Claiborne et al. (2002) proposed that enhanced acid secretion during acidosis in marine teleosts was mostly due to Na⁺/H⁺ exchangers in the
MRCs and that apically located Cl⁻/HCO₃⁻ exchangers may be responsible for base excretion, on the basis of cellular and molecular data. The proposed predominance of Na⁺/H⁺ exchangers in acid-base regulation in marine teleosts is however in conflict with the plasma ion data described above, and the apparent discrepancy between the mechanisms inferred from plasma ion and flux data and from molecular localization of transporters proteins must be resolved by further investigations.

The current model for the extrusion of NaCl by MRCs in marine teleosts include secondary active transport of Cl⁻ through Cl⁻ channels residing in the apical membrane driven by basolateral Na⁺/K⁺ ATPase, and passive Na⁺ transport through the leaky tight junctions between the MRCs and adjacent accessory cells (see Evans et al., 2005, Hwang et al., 2011, and Marshall and Grosell, 2006, for review). Although Na⁺ and Cl⁻ ions are generally considered to be transported in 1:1 ratio to maintain electroneutrality of the body fluids, the strong ion difference theory predicts that an excess extrusion of anions over cations should cause alkalinization of the fluid, without any “actual” movement of acid-base relevant ions (such as H⁺ and HCO₃⁻) across the body surface.
Therefore, it is theoretically possible for the MRCs to restore acid-base disturbance of marine teleosts by extruding $\text{Na}^+$ and $\text{Cl}^-$ at different rates.

Effects of environmental conditions on the gill MRC morphology in freshwater teleosts have been extensively investigated (see Goss et al., 1995). For example, both proliferation of MRCs and increased apical opening area have been reported in rainbow trout exposed to ion-poor water (Greco et al., 1996) or hyperoxia (Goss et al., 1994), in rainbow trout injected with NaHCO$_3$ or HCl to induce metabolic alkalosis or acidosis (Goss et al., 1994), in parr and smolt Atlantic salmon *Salmo salar* exposed to cadmium ions (Devos et al., 1998), and in snakehead *Channa punctata* exposed to a mixture of four trace metals (Cu, Cd, Fe and Ni, Pandey et al., 2008). Also, a fourfold increase in the fractional surface area of branchial MRC was observed when rainbow trout was exposed to alkaline water (pH 9.5) for 3 days (Wilkie and Wood, 1994). Goss et al. (1995) summarized effects of acid and alkaline water pH on MRC morphology of freshwater fishes. In comparison, effects of environmental conditions on MRC morphology are only poorly known for marine fishes.
Table 1 summarizes the data on morphological, biochemical and molecular responses in the gills of marine teleosts in response to environmental hypercapnia. Apparently, no clear pattern emerges from this data set. For example, the activity of Na$^+$/K$^+$ ATPase remained elevated for 42 days during hypercapnia exposure to 1.0 kPa pCO$_2$ in *Zoarces viviparus* (Deigweiher et al., 2008). Similarly, the activity and protein abundance of Na$^+$/K$^+$ ATPase were found significantly higher than control levels when *Gadus morhua* was exposed to 0.6 kPa for 12 months (but no change under 3 kPa after 4 months, Melzner et al., 2009). The Na$^+$/K$^+$ ATPase activity transiently increased or decreased in *Paralichthys olivaceus* (this study) and *Opsanus beta* (Esbaugh et al., 2012), respectively, and the activity was found to be depressed after 2 days of CO$_2$ exposure in *Porichthys notatus* (Perry et al., 2010). Carbonic anhydrase showed opposite changes in mRNA expression between *Opsanus beta* (a decrease, Esbaugh et al., 2012) and *Porichthys notatus* (an increase, Perry et al., 2010). With respect to those proteins related to Cl$^-$ fluxes, SLC4A2 decreased in mRNA expression, while SLC26A3 or SLC26A6 did not show any change in *Opsanus beta* (Esbaugh et al., 2012). Similarly,
mRNA expression of zoarcid Cl/HCO$_3^-$ exchanger did not change in CO$_2$-exposed $Z$. $viviparus$ (Deigweiher et al., 2008). These studies employed different exposure protocols with the seawater pCO$_2$ ranging from 0.2 to 5.2 kPa, the exposure period from only 1 hr to 12 months, and various sampling schedules. Therefore comparison of these data cannot be made with certainty.

As shown in Table 1, some parameters changed only transiently in response to high CO$_2$ exposure, and subsequently returned to pre-exposure, normocapnic levels. These time-dependent variables may not be very useful as biological indices of CO$_2$ impacts because the power of detection depends on the time between the onset of a hypercapnic period and sampling, and because temporal patterns of responses are likely to vary with test species, CO$_2$ levels, and other biotic and abiotic factors. Rather, it would be ideal if a parameter remains deviated from its baseline level as long as environmental CO$_2$ levels are elevated or even after the ambient CO$_2$ concentration has returned to normal levels of a locality. The opening area of the gill MRCs and the activity of branchial Na$^+$/K$^+$ ATPase in $P. olivaceus$ were both affected significantly within 72 hr but then
returned to pre-exposure levels (Figs. 2 and 4), which limits the usefulness of those parameters as indices of CO₂ impact. Further, consideration must also be given to the detection limits of environmental CO₂ by those biological indices. The threshold levels at which these parameters showed significant changes were 3 and 1 kPa for apical opening area (Fig. 2) and Na⁺/K⁺ ATPase activity (Fig. 4), respectively. At 1 kPa, seawater pH dropped to 7.0, a drop of 1.2 pH units, which is easily detectable by pH sensors (Shitashima et al., 2012). Nevertheless, considering the ecological and economical importance of fishes, careful investigations must be continued to examine effects of CO₂ on marine fishes, particularly using long-term (months or longer) exposure protocols with pCO₂ levels lower than those used in the present study. Tentatively, the present data would rank both of the parameters we studied as relatively poor indicators of CO₂ impacts, but given the highly variable nature of biological responses to CO₂ (Kroeker et al., 2010), more fish species must be tested for their responses of MRC morphology and enzymatic activity before we can arrive at a solid conclusion about the usefulness of MRC morphology and Na⁺/K⁺ ATPase activity as
indicators of CO₂ impact.

In the present study, we chose *P. olivaceu* because the fish was easily available from aquaculture farms but also inhabits the depth range down to 200 m, the depth at which subsea CCS may be implemented in the continental shelves. The fish is demersal and less mobile than pelagic fish species (Kawabe et al., 2004), and may therefore be more prone to be impacted by CO₂ leakage from a CCS site. Although demersal, inactive fishes such as *P. olivaceus* could be useful model animals for monitoring CO₂ conditions around an injection site, it would be desirable also to explore suitable indices of CO₂ impacts in sessile or poorly mobile invertebrates. Although biological information is much less available than for fishes, marine invertebrates, particularly those forming calcareous shells and skeletons (many of them have much less mobility than fishes), are considered to be more vulnerable to CO₂ than fishes (Hofmann et al., 2010). Some marine invertebrates such as sea urchins have been preferably used as models in some fields of marine biology, which might facilitate search for suitable biological indicators of CO₂ impacts (Ernst 1997; Lawrence, 2007). In addition, a rapid
expansion of ocean acidification research in recent years is predictive of the identification of reliable biomarkers for CO$_2$ impacts in marine fishes and invertebrates in the near future (Hardege et al., 2011).

Acknowledgements

This study was partly supported by the New Energy and Industrial Technology Development Organization (NEDO) and the Research Institute of Innovative Technology for the Earth (RITE).

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http://dx.doi.org/10.1016/j.aquaeng.2012.11.006


Upham, P., Roberts, T., 2011. Public perceptions of CCS in context: results of NearCO₂ focus groups in the UK, Belgium, the Netherlands, Germany, Spain and Poland.

Figure Legends

Fig. 1 Scanning electron micrographs of gill filaments in the Japanese flounder exposed to hypercapnic seawater (2.97 kPa (A-D) and 4.95 kPa (E, F) pCO$_2$) at 0 (A, E), 24 (B, F), 72 (C) and 168 hr (D). Arrowheads, apical opening of MRCs. pc, pavement cell.

Scale bars, 5 µm.

Fig. 2 The apical opening area of mitochondria-rich cells (MRCs) in the Japanese flounder exposed to normocapnic (white bars) and hypercapnic (0.98 kPa (hatched bars), 2.97 kPa (cross-hatched bars) and 4.95 kPa (black bars) pCO$_2$) seawater. Data are expressed as means ± S.D. (n = 6). Data at the same CO$_2$ levels with same letters are not significantly different from each other (p < 0.05; Tukey’s test).

Fig. 3 The cross-sectional area (A) and cell density (B) of MRCs in the Japanese flounder exposed to normocapnic (white bars) and hypercapnic (0.98 kPa (hatched bars) and 4.95 kPa (black bars) pCO$_2$) seawater. Data are expressed as means ± S.D. (n = 6).
Fig. 4  Na⁺/K⁺-ATPase activity in the gills of the Japanese flounder exposed to normocapnic (white bars) and hypercapnic (0.98 kPa (hatched bars) and 4.95 kPa (black bars) pCO₂) seawater. Data are expressed as means ± S.D. (n = 6). Symbols are the same as in Fig. 2.
Fig. 2

The bar chart illustrates the apical opening area (μm²) over different time points (0, 24, 72, 168 hours). The chart shows significant changes in apical opening area with time, with peak values at 72 hours.
<table>
<thead>
<tr>
<th>Species</th>
<th>Responses</th>
<th>pCO₂ (kPa)</th>
<th>Time</th>
<th>Ref</th>
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<tbody>
<tr>
<td><em>Dicentrarchus labrax</em></td>
<td>Na⁺/H⁺ exchanger (NHE1): mRNA expression decreased at 5.2 kPa (1 hr) but no change at 3.5 kPa (96 hr)</td>
<td>3.5/5.2*</td>
<td>96/1 hr</td>
<td>(1)</td>
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<tr>
<td><em>Gadus morhua</em></td>
<td>Na⁺/K⁺ ATPase: activity and protein abundance both increased at 0.6 kPa after 12 months but not at 0.3 kPa after 4 months</td>
<td>0.3/0.6</td>
<td>4/12 months</td>
<td>(2)</td>
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<td><em>Opsanus beta</em></td>
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<td>0.2</td>
<td>3 days</td>
<td>(3)</td>
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<td>Carbonic anhydrase: mRNA expression depressed</td>
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<td>V-type H⁺-ATPase: a non-significant drop in activity at 24 hr</td>
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<td>SLC4A2: mRNA expression decreased</td>
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<td>V-type H⁺-ATPase: a non-significant drop in activity at 24 hr</td>
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<td>SLC26A3, SLC26A6: mRNA expression did not change</td>
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<td>Na⁺/HCO₃⁻ cotransporter: mRNA expression did not change</td>
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<td><em>Paralichthys olivaceus</em></td>
<td>Na⁺/K⁺ ATPase: a transient increase in activity above 1.0 kPa</td>
<td>&lt; 4.9</td>
<td>7 days</td>
<td>(4)</td>
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<td>MRC apical opening area: a transient increase above 2.9 kPa</td>
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<td>MRC cross sectional area: no change</td>
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<td>MRC density: no change</td>
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<tr>
<td><em>Porichthys notatus</em></td>
<td>Na⁺/K⁺ ATPase: no change in activity</td>
<td>5.0</td>
<td>2 days</td>
<td>(5)</td>
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<td>Carbonic anhydrase: activity and protein expression both increased</td>
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<td><em>Zoarces viviparus</em></td>
<td>Na⁺/K⁺ ATPase: mRNA expression, protein abundance, and activity all increased</td>
<td>1.0</td>
<td>42 days</td>
<td>(6)</td>
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<td>NHE1: a transient drop in mRNA expression with nearly no change in protein abundance</td>
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<td>Cl⁻/HCO₃⁻ exchanger: no change in mRNA expression</td>
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</table>
Na⁺/HCO₃⁻ cotransporter: mRNA expression increased only after 42 days

(1) Rimoldi et al., 2009; (2) Melzner et al., 2009; (3) Esbaugh et al., 2012; (4) this study; (5) Perry et al., 2010; (6) Deigweiher et al., 2008. *estimated from the given concentration of dissolved CO₂ and temperature. SLC4 and SLC26 families are various types of anion exchangers (see Romero et al., 2004 and Mount and Romero, 2004 for details)