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Excess post-hypoxic oxygen consumption is independent from lactate accumulation in two cyprinid fishes

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ABSTRACT

Carassius carassius responds to hypoxic conditions by conversion of lactate into ethanol, which is excreted over the gills. However, a closely related species, *Cyprinus carpio*, does not possess the ability to produce ethanol and would be expected to accumulate lactate during hypoxic exposure. While the increase in oxygen consumption in fish required following strenuous exercise or low environmental oxygen availability has been frequently considered, the primary contributing mechanism remains unknown. This study utilized the close relationship but strongly divergent physiology between *C. carpio* and *C. carassius* to examine the possible correlation between excess post-hypoxic oxygen consumption (EPHOC) and lactate accumulation. No difference in the EPHOC:O_2 deficit ratio was observed between the two species after 2.5 h anoxia, with ratios of 2.0 ± 0.6 (*C. carpio*) and 1.3 ± 0.3 (*C. carassius*). As predicted, lactate accumulation dynamics did significantly differ between the species in both plasma and white muscle following anoxic exposure. Significant lactate accumulation was seen in both plasma and muscle in *C. carpio*, but there was no accumulation of lactate in white muscle tissue of *C. carassius*. These findings indicate that lactate accumulated as a consequence of 2.5 h anoxic exposure is not a major determinant of the resulting EPHOC.

1. Introduction

Three cyprinid teleosts, the crucian carp (*Carassius carassius* (L)), goldfish (*Carassius auratus* (L)), and bitterling (*Cyprinus amarus* (Bloch)), are unique among vertebrates for their ability to convert lactate into ethanol as the end product of anaerobic metabolism (Shoubridge and Hochachka, 1980; Johnston and Bernard, 1983; Wissing and Zebe, 1988). The produced ethanol is freely diffusible over the cell membrane and is excreted from the fish via the gills (Shoubridge and Hochachka, 1980; van den Thillart et al., 1983; Stecyk et al., 2004). This rare adaptation is instrumental in a greatly enhanced tolerance to hypoxic conditions. Indeed, *C. carassius* can survive more than 24 h of anoxia at room temperature, and at least 4.5 months at near-zero temperatures (Holopainen and Hyvärinen, 1985; Piironen and Holopainen, 1986; Nilsson and Renshaw, 2004). In contrast, the common carp (*Cyprinus carpio*), a cyprinid species closely related to *C. carassius*, does not possess the ability to produce ethanol (Nilsson, 1988), yet is regarded as a good anaerobe tolerating anoxic exposure of at least 1 h at 20 °C (van Waarde et al., 1990; van Raaij et al., 1996), and surviving less severe hypoxia (0.5 mg O_2 L^{-1}) for at least 7 days at 22–23 °C (Zhou et al., 2000).

The comparison of the hypoxia tolerance strategies between these two species is based on the distinct differences in metabolic responses to oxygen limitation each species employs. Standard metabolic rate (MO_{2std}) is the minimum oxygen requirement for the maintenance of unimpaired physiological reactions in postprandial unstressed animals at rest. When the oxygen saturation (O_{2sat} (%)) in the water is too low to support these basal requirements by aerobic metabolism, phosphocreatine (PCr) acts as an "energy buffer", stabilizing the [ATP] by rapidly regenerating ATP from ADP. The capacity to maintain the [ATP] by PCr hydrolysis is limited (van Ginneken et al., 1995; Dalla Via et al., 1997) and anaerobic glycolysis is therefore the principal ATP-generating pathway that can function during long periods of anoxia (Bickler and Buck, 2007). Due to the low ATP yield from anaerobic glycolysis, cells compensate for the diminished aerobic energy production by a substantial rise in glucose consumption rates resulting in lactate accumulation (Hochachka, 1986). For every mole of glucosyl-units used to support

Abbreviations/symbols: ADP, adenosine diphosphate; AMS, aerobic metabolic scope (MO_{2max}/MO_{2std}); ATP, adenosine triphosphate; EPHOC, excess post-hypoxic oxygen consumption (mg O_2 kg^{-1}); MO_{2max}, maximal oxygen consumption rate (mg O_2 kg^{-1} h^{-1}); $MO_{2post\text{-}anoxia}$, oxygen consumption rate (mg O_2 kg^{-1} h^{-1}) following anoxic exposure; MO_{2std}, standard metabolic rate (mg O_2 kg^{-1} h^{-1}); O_{2sat}, oxygen saturation (%); PCr, phosphocreatine; S_{crit}, critical oxygen saturation.

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anaerobic glycolysis, 2 mol of lactate is produced; deviations from a 2:1 ratio therefore indicate sources additional to glycogen depletion. This is the response to low oxygen saturation observed in most teleost fish species, including *C. carpio*. In conjunction with this process, *C. carassius* converts lactate into ethanol, which addresses the problem of acidification by ATP hydrolysis associated with lactate production (Hochachka and Mommsen, 1983) and, combined with one of the largest known glycogen stores of any vertebrate (Hyvärinen et al., 1985) allows *C. carassius* to maintain a relatively high glycolytic rate for extended periods (Nilsson, 1990). The conversion of lactate to ethanol in *C. carassius* happens exclusively in muscle tissue and lactate produced in tissues other than the muscle is transported via the blood to the muscle tissue for fermentation (Johnston and Bernard, 1983; Nilsson, 1988).

Fish can increase their oxygen consumption rate by several folds compared to MO_{2std} until reaching their maximum capacity (MO_{2max}) wherein all aerobic activities are undertaken. During recovery from anoxia, oxygen consumption increases above MO_{2std} for an extended period of time, but studies that quantify the total metabolic cost of recovery from severe hypoxia are rare (van den Thillart and Verbeek, 1991; Maxime et al., 2000; Svendsen et al., 2012). The accumulated excess post hypoxic oxygen consumption (EPHOC, mg O_2 kg^{-1}) has classically been attributed to the lactate load, but evidence suggests that EPHOC is only partially related to the lactate load, and that resynthesis of glycogen from lactate during recovery is not the major component of the increased O_2 consumption. Instead, the EPHOC has been attributed to re-synthesis of ATP and PCr in addition to glycogen, and also the buffering of protons generated from ATP utilization (van den Thillart and Verbeek, 1991; Virani and Rees, 2000; Mandic et al., 2008). However, the relative contributions of these processes to EPHOC in fish, and in particular the role of lactate, remain an area of ongoing investigation.

The present study examines the hypothesis that EPHOC associated with acute exposure to anoxia (≤1% O_{2sat}) is positively correlated to lactate accumulation. Utilizing the close phylogenetic relationship, yet distinct difference in hypoxia tolerance physiology between *C. carpio* and *C. carassius* this study investigates the link between lactate load and EPHOC. Because lactate is converted to ethanol in *C. carassius*, but not in *C. carpio*, it was hypothesized that 1) acute exposure to anoxia would cause substantial lactate accumulation in *C. carpio*, while it would be limited in *C. carassius*; and 2) the lactate accumulation would result in greater EPHOC relative to the produced O_2 deficit in *C. carpio*, compared to *C. carassius*. In this study, we therefore quantified 1) concentration of lactate in muscle and plasma during exposure to anoxia in juvenile *C. carassius* and *C. carpio*, and 2) EPHOC (mg O_2 kg^{-1}) after exposure to 2.5 h anoxia.

2. Materials and methods

2.1. Experimental animals

A total of 34 juvenile *C. carpio* and 33 *C. carassius* (110–130 mm) were collected from a pond near Slagelse, Denmark (55°17′58 N 11°27′47 E) in April 2009. At capture water temperature was 12.5–14.0 °C. Fish were transferred to the University of Copenhagen, Marine Biological Laboratory, Helsingør, Denmark and kept indoors in a 400 L tank supplied with a continuous flow of unchlorinated tap water. Water was filtered using a mechanical filter pump (1100 L h^{-1}) connected to the tank, and water temperature was kept at 15 ± 0.1 °C and continually aerated to maintain normoxic conditions. The fish were kept in a 12L:12D light regime and were fed to satiation 2–4 times per week with commercial fish pellets (Ecolife 3 mm, Biomar, Denmark). Prior to experimentation, fish were acclimated to these conditions for 4 months. No fish was used more than once. All methods applied in the present study were in agreement with current Danish regulations for the treatment and welfare of experimental animals.

2.2. Respirometry

2.2.1. Equipment setup

The setup consisted of a static respirometer and a mixing pump submerged in a 50 L opaque tank on a wet table, filled with unchlorinated tap water maintained at 15 ± 0.1 °C. The respirometer was made of transparent Perspex tubing and was fitted with two outlet and two inlet ports. The mouth of the outlet tube, through which water left the respirometer, was elevated slightly above the water surface level to prevent the ambient water from entering the respirometer. Inside the respirometer, a plate positioned 5 mm from the ports propagated water mixing and prevented the fish from disturbing the inflow and outflow. A perforated tube was inserted into the respirometer to minimize spontaneous activity associated with exposure to decreased O_{2sat} levels, a behavior that has been previously observed in *C. carpio* (Vianen et al., 2001). The tank was positioned behind a black curtain to minimize stressful stimuli.

Measurements of O_2 consumption rate (MO_2; mg O_2 kg^{-1} h^{-1}) were carried out every 7 min 50 s using computerized intermittent-flow respirometry allowing long term (>48 h) repeated measurements as previously described (Steffensen et al., 1984; Steffensen, 1989). The repeated respirometric loops consisted of a 3 min 20 s flushing phase during which a pump flushed the respirometer with ambient water through one set of ports. The second set of ports and a pump allowed the water in the respirometer to be re-circulated in a closed circuit phase for 4 min 30 s, divided into a waiting period (2 min) and a measurement period (2 min 30 s).

Oxygen partial pressure was measured at 1 s^{-1} by a fiber optic sensor (Fibox 3 connected to a dipping probe; PreSens, Regensburg, Germany) located in the recirculated loop. The flush pump was controlled by AutoResp software (Loligo Systems Aps, Tjele, Denmark) that also calculated the oxygen consumption rate in the measuring phase using the oxygen partial pressure and standard equations (Schurmann and Steffensen, 1997). Preliminary testing demonstrated that the duration of the measurement period (2 min 30 s) in combination with the mass of the experimental fish (19.5 ± 0.7 g) and the volume of the respirometer and re-circulated loop (0.335 L) ensured that the coefficient of determination (r^2) associated with the MO_2 measurements was always >0.90 as in previous studies (Behrens and Steffensen, 2007; Campbell et al., 2008). Moreover, in normoxia the respiration of the fish never reduced the O_{2sat} to less than 84% (approx. 17.5 kPa).

Water for the flush pump was supplied from one of two different tanks containing either normoxic or hypoxic water maintained at 15 ± 0.1 °C. Adequate water quality in the system was maintained by an internal filter pump and an ultraviolet light sterilizer running continually. Prior to initiation of an experiment the adjustable tank was reduced to ≤2.5% O_{2sat} (approx. 0.5 kPa) by circulating water from the tank through a vertical cylinder (0.25 m in diameter, 1 m high) where the water was exposed to a stream of nitrogen bubbles (Behrens and Steffensen, 2007). To minimize diffusion of O_2 from the ambient air, water surfaces were covered by floating bubblewrap. The O_{2sat} in the adjustable tank was measured using a Mini DO probe (Loligo Systems Aps., Tjele, Denmark) connected to a relay that controlled the O_{2sat} in the tank via a solenoid valve regulating nitrogen gas delivery to the cylinder similar to the procedure described by (Jordan and Steffensen, 2007). The O_{2sat} in the normoxic tank was maintained at a constant high normoxic level (≥95% O_{2sat}, approx. 19.8 kPa) using air stones. The desired O_{2sat} in the hypoxic tank was adjusted and stabilized before the flush pump started supplying water from this tank. In this way, the experiment was not influenced by any delays caused by the time required to reduce the O_{2sat} in the hypoxic tank. The shift from normoxic to hypoxic water was made by manually closing the valve regulating outflow from the normoxic tank and opening the valve from the adjustable tank, which had been previously brought to ≤2.5% O_{2sat} as described above. Both

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valves were situated outside the tank to eliminate disturbance of the fish, and preliminary tests confirmed that the procedure did not influence the metabolic rate of the fish. During the flush phase, the flush pump exchanged greater than 8 times the volume of water in the respirometer, which is sufficient to replace >99% of the water (Steffensen, 1989). Using this arrangement, the O_{2sat} inside the respirometer reached the designated O_{2sat} level in <3.5 min and was immediately followed by MO_2 measurements.

2.2.2. Experimental protocol of MO_2 measurements

EPHOC following anoxia was determined in two size-matched groups of 9 *C. carpio* (19.5 ± 1.1 g) and 8 *C. carassius* (19.5 ± 0.7 g). Fish were fasted for 24 h prior to experimentation. Individual MO_{2max} was tested in normoxia by transfer of the fish from the holding tank to a bucket and chasing to exhaustion, according to Richards et al. (2002). This protocol has been used to induce MO_{2max} in several teleost species as an alternative to swimming the fish in the respirometer (Peake and Farrell, 2006; Jordan and Steffensen, 2007; Killen et al., 2007). Upon exhaustion, identified by no further response to manual stimulation (after 5–6 min), fish were transferred to the respirometer where MO_2 measurements were started immediately. After the $MO_{2\ max}$ measurements, fish were acclimated to the respirometer for 24–48 h.

Preliminary testing confirmed previous work that indicated the maximum survival time for *C. carpio* exposed to anoxia at 15 °C was approx. 2.5 h (Stecyk and Farrell, 2002), and 2.5 h was consequently used as the duration of anoxic exposure. Tests with the two different species were carried out in random order. It was not possible to reduce the O_{2sat} in the hypoxic tank to less than 2.5% (0.5 kPa). Therefore, to induce anoxia in the respirometer, the flush pump was turned off after the first flush period of the experiment. Shutting off the water exchange caused the fish to induce anoxia (≤1% O_{2sat}, approx. 0.2 kPa) in the respirometer in ≤15 min. After the anoxic exposure the flush pump was engaged and the respirometer flushed with normoxic water. Due to the lag time of the fiber optic sensor adjusting from ≤0.1 to >95% O_{2sat}, the flush period of the first respirometric loop was extended by 3 min and the measurement discarded. Collection of MO_2 data every 7 min and 50 s continued for >12 h after the exposure to anoxia.

2.2.3. Acquisition and analysis of respirometry data

Because of the rapid turnover of water, both the exact rate of change of the O_{2sat} and the response time of the O_2 consumption rate of the fish were unknown during the flush periods; because of these uncertainties the flush periods used to modify the O_{2sat} inside the respirometer were not included in the calculations. MO_{2std} was defined as the mean of the last seven measurements (54 min 50 s) (Fig. 1) before onset of hypoxia, similar to previously employed procedures (Scarabello et al., 1991; Svendsen et al., 2010). The EPHOC protocol involved rapid changes of the O_{2sat} inside the respirometer during single flush periods (from normoxia to anoxia and vice versa). The oxygen deficit (mg O_2 kg^{-1}) accumulated during the anoxic period was quantified as the MO_{2std} during the 2.5 h. Individual recovery periods were regarded as completed when the first MO_2 datum in the post anoxia recovery period ($MO_{2\text{-}post\text{-}anoxia}$) was within a 95% confidence interval (CI) of the MO_{2std} (Fig. 2) as previously described (Bushnell et al., 1994; Svendsen et al., 2010). The metabolic cost of recovery (mg O_2 kg^{-1}) was determined by subtracting the MO_{2std} from $MO_{2\ post\text{-}anoxia}$, following Jordan and Steffensen (2007). Aerobic metabolic scope was calculated as the difference between MO_{2max} and $MO_{2\ std}$, following Farrell and Richards (2009).

2.3. Measurements of plasma and muscle lactate

2.3.1. Equipment setup

Two groups of 25 size matched *C. carpio* and *C. carassius* (20.9 ± 0.5 g) were used for the time series measurements of lactate development in plasma and white muscle. A 180 L aquarium was fitted with black plastic on all sides to prevent visual disturbance, filled with unchlorinated tap water, and fitted with an internal filter pump to ensure adequate mixing. The temperature was kept at 15 ± 0.1 °C and the water was maintained normoxic by continuous aeration by air stones. The O_{2sat} was monitored using a Mini DO probe (Oxyguard International, Birkerød, Denmark) connected to a relay controlling the O_{2sat} in the tank via a solenoid valve that regulated nitrogen gas delivery to multiple air stones on the bottom of the aquarium. All holes around tubes and cables into the aquarium were covered with plastic film. The sealed container facilitated precise regulation of O_{2sat} from ≥95% to 1%. To allow individual sampling with a minimum of disturbance of the remaining fish in the aquarium, each fish was inserted in a small cage made from plastic mesh tube (40 mm diameter). A nylon string was fitted to each cage and a small weight kept the cage on the bottom and made it impossible for the fish to move the cage.

2.3.2. Experimental protocol of lactate sampling

Fish were starved for 24 h before being transferred from the holding tank to the aquarium and inserted in the cages. Acclimation to the aquarium under normoxia lasted for 36 h, and fish were not fed during this time. Five fish of each species were sampled immediately before the onset of hypoxia as a normoxic baseline. Within 1 h anoxia was reached (1 ± 0.2% O_{2sat}, approx. 0.2 kPa) by nitrogen bubbling. Subsequently, a fish was sampled every 4 min. Alternating between

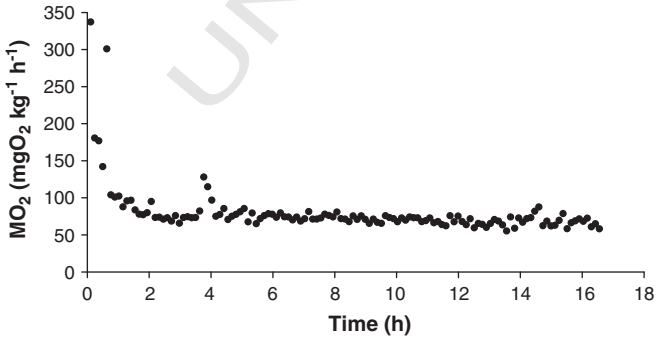

Fig. 1. Representative trace of the time course of MO_2 measurements (mg O_2 kg^{-1} h^{-1}) during acclimation in a static respirometer. Data were collected using a 23.7 g common carp (*Cyprinus carpio*) at 15 °C. Each datum represents a 7 min 50 s period. MO_2 is corrected for background respiration.

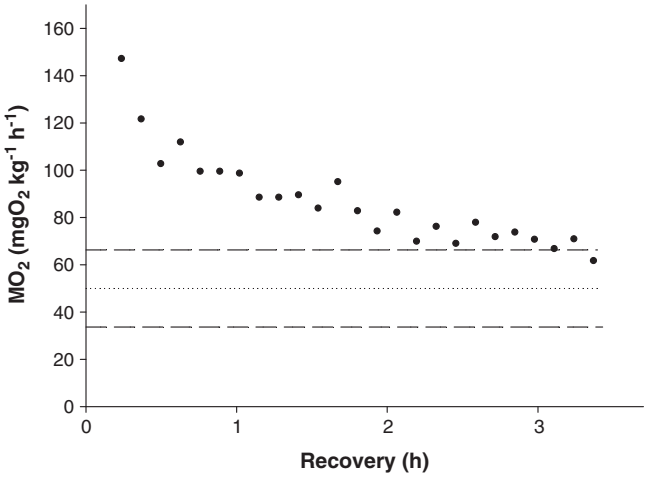

Fig. 2. Representative trace of $MO_{2post\text{-}anoxia}$ in a 19.2 g crucian carp (*Carassius carassius*) after 2.5 h anoxia exposure. MO_{2std} ± 95% CI are illustrated using a dotted and two dashed lines, respectively. MO_2 is corrected for background respiration. The first MO_2 datum in the post anoxia recovery period within the 95% confidence interval (CI) of the MO_{2std} was used as the marker indicating completion of the recovery period.

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each species, a total of 20 *C. carpio* and 20 *C. carassius* were sampled, giving a total anoxic exposure period of 2 h 40 min.

At sampling, the lid was lifted slightly and a cage retracted from the tank by the nylon string. The cage was immediately transferred to a 2.5% benzocaine solution (Sigma-Aldrich Chemicals, USA) made from a 4% ethanol stock solution. At complete anesthesia (≤1 min), the fish were removed from the cages, patted dry and weighed to the nearest 0.1 g. Blood samples were collected by severing the tail from the body and collecting the blood flowing from the caudal vein with a heparinized 1 mL syringe (LEO Pharma A/S, Ballerup, Denmark). The blood sample was transferred to a 0.5 mL centrifuge tube and centrifuged at 2000 *g* for 30 s to isolate the plasma. A tissue sample was taken as a cross section of the trunk musculature posterior to the dorsal fin and wrapped in an aluminum foil. Both the tissue and plasma samples were flash frozen in liquid N_2 and stored at −80 °C until analysis.

2.3.3. Determination of lactate concentration

Extraction of lactate from the tissue samples was carried out following procedures previously described (Viant et al., 2003; Lin et al., 2007). The frozen muscle samples were ground to a fine powder in a N_2-cooled mortar. The frozen, powdered tissue (100 mg) was weighed in a N_2-cooled 1.5 mL centrifuge tube and extracted using 5 mL g^{-1} (wet mass) ice cold 6% perchloric acid. Samples were kept on ice throughout the extraction procedure. Samples were vortexed for 15 s three times, centrifuged (10,000 *g*, 10 min, 4 °C), and the supernatant was removed and neutralized to pH 7.5 with 2 M K_2CO_3, testing pH using small drops of sample on pH paper (pH paper range: 5.5–9.0). Samples were kept on ice for an additional 30 min to facilitate complete precipitation. Following centrifugation (10,000 *g*, 10 min, 4 °C), the supernatant was removed and stored at −80 °C. Muscle extracts and plasma were analyzed for lactate using a commercial kit (Biomedical Research Service, NY, USA). The measurements were corrected using internal lactate standards in samples from fish of both species sampled in normoxic conditions.

2.4. Statistical analysis

All values are reported as mean ± standard error of the mean (SEM). Means were compared using Student's *t*-test (two-tailed) after testing the assumptions of normal distribution of data and homogeneity of variance. Means of data found not to be normally distributed were compared using the Mann–Whitney test. Least square linear regression analysis was performed using SigmaPlot 10.0 (Systat Software Inc. San Jose, CA, USA), and regression line slopes were compared using analysis of covariance. Statistical analyses were carried out using SPSS 15.0 (IBM SPSS, Armonk, NY, USA). Means were considered significantly different when $P<0.05$.

3. Results

3.1. Oxygen consumption rates

$MO_{2\ std}$ differed significantly between *C. carassius* and *C. carpio* (Table 1, $P<0.02$). Similarly, MO_{2max} in *C. carassius* was significantly lower than the MO_{2max} measured in *C. carpio* ($P<0.0001$). Despite the overall greater oxygen consumption seen in *C. carpio*, aerobic metabolic scope (AMS, MO_{2max}/MO_{2std}) did not differ between these two species ($P>0.8$). As a consequence of the different MO_{2std} the accumulated O_2 deficit during the anoxic period was greater in *C. carpio* than *C. carassius* ($P<0.02$), and there was also a significant difference in EPHOC ($P<0.02$). Although both EPHOC and O_2 deficit were lower in *C. carassius* than *C. carpio*, the ratio of EPHOC:O_2 deficit did not differ between species ($P>0.48$). The average time to complete metabolic recovery was longer for *C. carpio* (7.0 ± 1.4 h) than for *C. carassius* (3.8 ± 0.7 h) ($P<0.034$). As was the case with the AMS, the utilized metabolic scope was similar for the two species, and during the recovery phase neither of the species utilized their full metabolic scope, with the highest measurements of MO_2 representing 65.4 ± 8.3% of MO_{2max} in *C. carassius* and 61.6 ± 7.6% in *C. carpio* ($P>0.7$).

Table 1
Observations of metabolic parameters in normoxia and during recovery from 2.5 h acute anoxic exposure in crucian carp (*Carassius carassius*, n = 8, 19.5 ± 0.6 g) and common carp (*Cyprinus carpio*, n = 9, 19.5 ± 1.1 g) at 15 °C. Asterisks indicate significant differences between species using two tailed Student's *t*-test, * $P<0.05$; and *** $P<0.0001$; NS, not significant.

	C. carassius	*C. carpio*	P
$MO_{2standard}$ (mg O_2 kg^{-1} h^{-1})	43.7 ± 5.3	66.5 ± 6.2	*
MO_{2max} (mg O_2 kg^{-1} h^{-1})	213.7 ± 7.3	329.5 ± 10.3	***
AMS (MO_{2max}/MO_{2std})	5.2 ± 0.4	5.4 ± 0.6	NS
O_2 deficit (mg O_2 kg^{-1})	108.5 ± 13.1	164.9 ± 15.5	*
EPHOC (mg O_2 kg^{-1})	124.4 ± 18.9	281.1 ± 53.5	*
EPHOC:O_2 deficit	1.3 ± 0.3	2.0 ± 0.6	NS
Time to recovery (h)	3.8 ± 0.7	7.0 ± 1.4	*
% of MO_{2max}	65.4 ± 8.3	61.6 ± 7.6	NS

3.2. Lactate accumulation

Parameters describing the production and accumulation of lactate during anoxic exposure are summarized in Table 2. Concentrations of lactate prior to anoxic exposure did not differ between species in plasma (3.8 ± 0.5 in *C. carassius* vs. 3.3 ± 0.4 mM in *C. carpio*, $P>0.48$) nor muscle (2.1 ± 0.1 vs. 1.7 ± 0.3 μmol g^{-1}; $P>0.26$). While the lactate concentration in the plasma rose significantly in both species, in the muscle tissue the concentration of lactate increased only in *C. carpio* (Fig. 3). In consequence the accumulation of lactate in *C. carassius* was significantly higher in plasma than in muscle ($P<0.0001$), with plasma [lactate] increasing 3 fold to 12 mmol L^{-1} (Table 2, Fig. 3A). *C. carpio* plasma [lactate] increased 6 fold to 21 mmol L^{-1}, and in contrast to *C. carassius* the lactate accumulation in plasma was significantly higher than in muscle ($P<0.0001$; Table 2), with muscle [lactate] increasing 5 fold to a final concentration of 8.97 μmol g^{-1} (Table 2, Fig. 3B). The lactate accumulation was faster in *C. carpio* than in *C. carassius* in both plasma (mmol L^{-1} h^{-1}, $P<0.01$) and muscle (μmol g^{-1} h^{-1}, $P<0.01$).

4. Discussion

4.1. Respirometry

4.1.1. Extent of EPHOC in various species

To our knowledge, EPHOC following exposure to oxygen levels below S_{crit} has been quantified for only three other fish species: *C. auratus* (van den Thillart and Verbeek, 1991) *Scophthalmus maximus* (Maxime et al., 2000), and *Oncorhynchus mykiss* (Svendsen et al., 2012). Several species of flatfish are moderately hypoxia tolerant (Dalla Via et al., 1994; Pichavant et al., 2002), and in hypoxia trials on *S. maximus*, a benthic flatfish found in temperate seas, the EPHOC:O_2 deficit ratio was 16:1 (Maxime et al., 2000), which is only half of the ratio of up to 35:1 observed in the hypoxia intolerant rainbow trout (*O. mykiss*) (Svendsen et

Table 2
Lactate development during 2.7 h acute anoxic exposure at 15 °C in crucian carp (*Carassius carassius*) and common carp (*Cyprinus carpio*) (20.8 ± 0.5 g combined mean body mass). Concentrations at 2.5 h anoxia were calculated from the linear regression (see Fig. 3). Asterisks (*) indicate statistical differences between species ($P<0.01$).

Lactate parameters	Plasma (mmol L^{-1})			Muscle (μmol g^{-1})		
	C. carassius	*C. carpio*	P	*C. carassius*	*C. carpio*	P
Normoxia	3.8 ± 0.5	3.3 ± 0.4	NS	2.1 ± 0.1	1.7 ± 0.3	NS
2.5 h anoxia	12.0	20.9	–	2.6	9.0	–
Increase (fold)	3.16	6.33	–	1.23	5.28	–
Slope (h^{-1})	1.45 ± 0.45	3.92 ± 0.82	*	0.05 ± 0.18	1.20 ± 0.41	*

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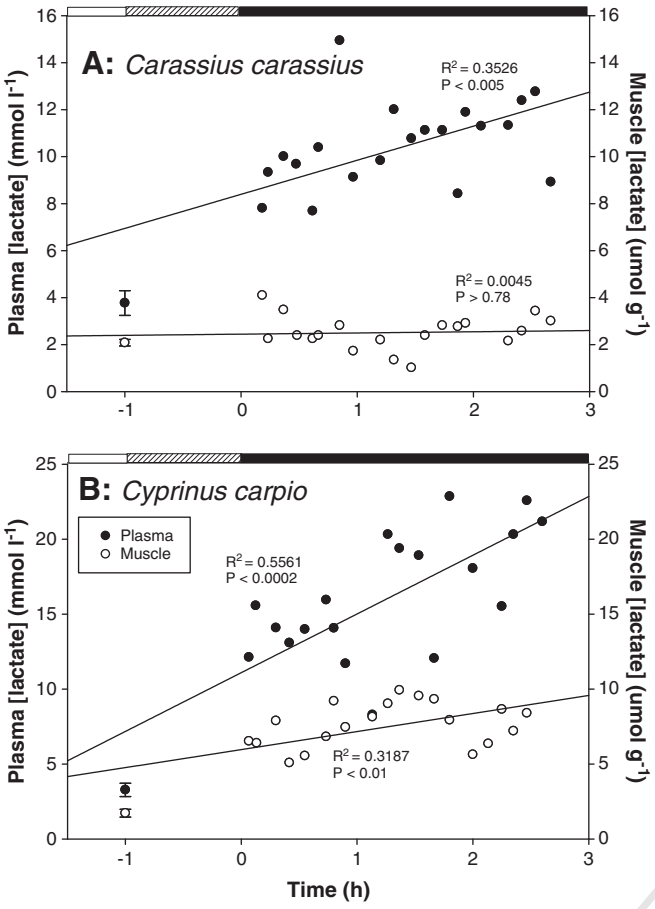

Fig. 3. Linear accumulation of lactate in plasma (mmol L^{-1}, black) and muscle (μmol g^{-1}, white) in A: crucian carp (*Carassius carassius*, 20.3 ± 0.6 g) and B: common carp (*Cyprinus carpio*, 21.4 ± 0.8 g) prior to and during 2.5 h of anoxia. The white bar indicates the normoxia (>95% O_{2sat}) period, the hatched bar indicates the period with decreasing O_{2sat}, and the black bar indicates the anoxia period (≤1% O_{2sat}), beginning at 0 h. Each datum represents a measurement on one fish in the anoxia period, except at t = −1, which is the mean ± SEM of fish sampled in normoxia (n = 5). Linear regressions are represented by the following equations: *C. carassius* plasma y = 1.4499x + 8.3973, and muscle y = 0.0511x + 2.4467; *C. carpio* plasma y = 3.9176x + 11.0967, and muscle y = 1.2026x + 5.9680.

al., 2012), but still an EPHOC:O_2 deficit ratio far greater than the ratios determined for both *C. carassius* and *C. carpio* in the present study (1.3:1 and 2.0:1, respectively), and of approximately 1.5:1 observed in *C. auratus* (van den Thillart and Verbeek, 1991). Of the fish species so far investigated for EPHOC, it is interesting to note that the species that accumulate only minimal oxygen debt are all members of the *Cyprinidae*. Of even greater interest, this capability does not seem to depend entirely on the ability to produce ethanol, as *C. carpio* demonstrates substantially lower EPHOC:O_2 deficit than other non-ethanol-producing species, but an approximately equivalent deficit to ethanol-producing *C. carassius* and *C. auratus*.

The observed EPHOC:O_2 debt ratio in *C. carassius* of 1.3:1 following 2.5 h anoxia at 15 °C (Table 1) is quite similar to the results obtained for the closely related species *C. auratus* (van den Thillart and Verbeek, 1991), which showed an EPHOC:O_2 deficit ratio of 1.5:1 after 12 h of anoxia at 20 °C. However EPHOC in the goldfish (*C. auratus*) was not observed following 3 h of anoxia, (van den Thillart and Verbeek, 1991) requiring greater time and temperature than those needed for EPHOC to be recorded in *C. carassius*. The fact that we did observe an EPHOC in *C. carassius* after the relatively short 2.5 h anoxia exposure could be attributed to the following factors: 1) species-specific physiological differences, despite the fish belonging to the same genus, 2) difference in the timescale of changing O_{2sat} levels and hence time for adjusting ventilatory and cardiac response as well as for the initiation of metabolic depression, 3) an overestimate of the resting metabolic rate in the previous study, thereby "hiding" the EPHOC, or 4) metabolic suppression continuing after reestablishment of normoxia. In addition to the conversion of lactate to ethanol, *C. carassius*, unlike *C. carpio*, can also depress its metabolism; van Ginneken and van den Thillart (2009) demonstrated that metabolic depression in *C. auratus* was initiated within 20–30 min after reduction of environmental O_{2sat}, and additionally that 1–2 h was needed to accomplish the full metabolic depression (by approximately 70% from MO_{2std}). In van den Thillart and Verbeek's (1991) study discussed above, in which EPHOC did not occur following 3 h anoxia, complete anoxia was not reached until after approximately 2.5 h, giving the goldfish sufficient time to reach full metabolic depression before anoxia was reached. In the present study, anoxia was reached in <30 min, and in consequence, *C. carassius* would only have been able to take full advantage of the ability to depress metabolism for approximately the last hour of the exposure. Regardless of the reason for this difference between our results and the observations by van den Thillart and Verbeek (1991), a very small EPHOC in *C. carassius* was observed in the present study, indicating comparatively higher hypoxia tolerance than is observed in *C. carpio* and other fish species. Q2

4.1.2. Small and uniform EPHOC in Cyprinidae

The oxygen deprivation utilized in this study was at a near lethal level for *C. carpio* (Johnston and Bernard, 1983; van der Linden et al., 2001; Stecyk and Farrell, 2002) but should be easily tolerated by *C. carassius*, yet no difference in the ratio of EPHOC:O_2 was found between the two species (Table 1). Interestingly, both species only increased metabolic rate to approximately 60% of their MO_{2max} in the recovery period and for a relatively short period of time (4–7 h), given the length of the exposure.

During anoxia, ATP levels in the brain of *C. carpio* slowly decrease (van Ginneken et al., 1996) and a significant swelling of the brain is seen over time due to the inactivation of the ATP dependent pumps regulating cell volume (Nilsson, 2001; van der Linden et al., 2001). These physiological responses to anoxia cause *C. carpio* to in essence slowly die during anoxia, while *C. carassius* is protected from such effects. Hallman et al. (2008) showed that *C. carpio* have a fairly large capacity for maintaining ATP levels using PCr as a buffer during O_2 levels below S_{crit} (approx. 13% O_{2sat} or 2.7 kPa). During this exposure it took approximately 2 h to reduce the [PCr] by half. Over the same timespan only a minor rise in plasma lactate took place in white muscle, indicating that *C. carpio* preferentially uses its PCr reserves before initiating the fermentation pathway for ATP resynthesis, presumably as an attempt to reduce metabolic acidification (Hochachka and Mommsen, 1983; van den Thillart and van Waarde, 1993).

In *C. carpio* (Hallman et al., 2008) as well as *C. auratus* (Mandic et al., 2008) both pH and PCr are completely recovered before lactate recovers. Despite high lactate loads remaining during recovery from exercise, fish can perform strenuous exercise at pre-fatigue levels when excess post-exercise oxygen consumption is repaid (Brett, 1964). This suggests that the acidification from the fermentation of glucose is likely of greater importance for the EPHOC than the lactate load itself. Indeed, in *C. auratus* (van den Thillart and Verbeek, 1991) and *C. carassius* (present study) the accumulation of lactate per se does not appear to burden the fish, and seems only to have a limited impact on the EPHOC in the two species at shorter timescales. A lactate-independent EPHOC could also indicate that *C. carpio* may have evolved to be able to cope with high lactate loads through residence in eutrophic habitats that experience regular hypoxic events (e.g. during the night). High amounts of stored lactate could subsequently be converted to glucose for aerobic respiration. Lactate is an excellent substrate for oxidation, and lactate in the blood can be metabolized by the heart, kidney and gills during period of high oxygen levels, or used for glyconeogenesis in situ. By not having to produce glycogen from the accumulated lactate, *C. carpio* would only have to

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repay an EPHOC corresponding to the required regeneration of ATP, PCr and internal O_2 stores (Scarabello et al., 1991).

Unique to ethanol producing species is the extensive loss of carbonic molecules due to anaerobic metabolism. *C. auratus* excrete 80% of the ethanol produced (van den Thillart and Verbeek, 1991) during anoxia, and continues to excrete significant amounts of ethanol for several hours after return to normoxic conditions (Mandic et al., 2008), indicating that lactate is preferentially converted to ethanol, even under normoxic conditions. If similar processes occur in *C. carassius*, accumulated lactate would have a minor influence on the EPHOC and it follows that the observed EPHOC from the duration of anoxia examined here (2.5 h) would mainly consist of regeneration of ATP, PCr and internal O_2 stores in a similar way as *C. carpio*, with limited remaining substrate for either Cori cycle or in situ glyconeogenesis. This may, at least in part, explain the observed similarity of the EPHOC despite significantly different lactate loads and diverse physiology between *C. carpio* and *C. carassius*. Further investigation of pH, lactate, ethanol, PCr and ATP dynamics during anoxia and recovery is needed, in combination with MO_2 measurements, to shed light on the cause of this unexpected observation of small and uniform EPHOC:O_2 deficit in *C. carassius* and *C. carpio*.

4.2. Lactate

4.2.1. Diverse lactate accumulation

As predicted, there was a difference in the pattern of lactate accumulation between *C. carpio* and *C. carassius*. Both plasma and muscle [lactate] rose significantly in *C. carpio*, but in *C. carassius* only plasma [lactate] increased (Table 2). Our measurements in muscle of *C. carassius* (Table 2) indicate no accumulation over normoxic values, which can be attributed to the short duration of anoxic exposure. In this species [lactate] the muscle increases approximately 4 fold following 6 h anoxia, yet no accumulation is seen following 3 h progressive hypoxia (Johnston and Bernard, 1983). The magnitude of lactate accumulation in the plasma also differed between species, with *C. carpio* accumulating almost twice as much lactate in plasma (Table 2), indicating a larger glycolytic flux in *C. carpio*.

4.2.2. High plasma lactate concentrations

C. carpio exerting moderate levels of exercise maintain levels of plasma [lactate] of approximately 1.5 mmol L^{-1} (van Ginneken et al., 2004a), which is similar to that measured in the present study (Table 2). However, both *C. carpio* and *C. carassius* completely at rest in normoxia have only 0.2–0.5 mmol L^{-1} lactate in the plasma (Holopainen et al., 1986; Vianen et al., 2001) at 20 °C and 18 °C, respectively, indicating that the fish in this study (at 15 °C) were most likely exhibiting some spontaneous activity prior to sacrifice for lactate quantification, despite efforts to limit this activity. Following anoxic exposure, the accumulated plasma [lactate] in *C. carpio* (20.9 mmol L^{-1}) is also higher than reported in other studies of carp exposed to hypoxia. Vianen et al. (2001) measured 6–13 mmol L^{-1} in plasma of cannulated *C. carpio* after 6 h progressive severe hypoxia. In *C. carassius*, plasma [lactate] increased approximately 3-fold to 12 mmol L^{-1}, demonstrating a similar qualitative response to anoxia as in previous studies where plasma [lactate] doubled following anoxic exposure (Holopainen et al., 1986).

There are two probable explanations for the high plasma [lactate] after exposure to anoxia. Firstly, the quick entry into anoxia (~1 h) directly from normoxia, compared to a gradual transition that allows for metabolic depression before entry into hypoxia. Change in O_{2sat} over only 1 h might be too fast to ensure sufficient time to initiate metabolic depression (van Ginneken and van den Thillart, 2009) or adequate ventilatory and cardiac responses (van Ginneken et al., 2004b; Wilkie et al., 2008), creating a higher S_{crit}, and forcing initiation of anaerobic metabolism earlier than if extraction capacity was able to be adjusted during the O_{2sat} decrease. Second, the metabolic stress during anoxia caused by relying exclusively on anaerobic metabolism may produce additional lactate accumulation, compared to the scenario in hypoxia where some aerobic metabolism can be maintained. In an Amazonian cichlid, *Astronotus ocellatus*, the lactate accumulation was 5 fold higher at 6% O_{2sat} than at 10% O_{2sat} (Muusze et al., 1998) and in *Solea solea* a 4–5 fold higher accumulation at 6% O_{2sat} than at 12% O_{2sat} was observed (Dalla Via et al., 1994). This illustrates how the shift to complete reliance on anaerobic metabolism happens relatively swiftly when anoxia is approached, and why data obtained in different levels of hypoxia remain difficult to compare.

Both species considered in this study demonstrated higher [lactate] in plasma compared to muscle. This may be a distinguishing factor for lactate accumulation due to hypoxia. For example the response of *S. solea* to severe hypoxia is qualitatively similar to our observations in *C. carpio* (Dalla Via et al., 1994), however, during exercise in *S. solea* the pattern is quite different, with the majority of lactate being produced and subsequently retained in the working muscles, resulting in lactate concentrations in muscle that are several folds higher in muscle than in plasma (Dalla Via et al., 1997). This is an advantage in normoxia due to the higher buffer capacity of the muscle tissue and because any acidification of the blood will lead to lowering of the hemoglobin binding affinity reducing O_2 extraction capacity, which is likely to prolong the duration of recovery. Indeed, accumulation of lactate in both the plasma and muscle tissue of *C. carpio*, but not *C. carassius*, coincides with significantly longer metabolic recovery (Table 1).

4.2.3. Impact of ethanol production on lactate accumulation

The ethanol production in *C. carassius* is well described (Johnston and Bernard, 1983) and is evident in the present study by the complete absence of accumulation of lactate in muscle tissue of *C. carassius*. Unlike in *C. carpio*, ATP levels in *C. carassius* are not primarily maintained by PCr stores. Mandic et al. (2008) measured a significant excretion of ethanol by *C. auratus* to the surrounding water within 2 h of initiation of anoxia but found only a 50% reduction in [PCr] after 10 h of anoxia at 15 °C. These results, considering the time needed for lactate production, conversion to ethanol and diffusion into the water, and the absence of any initial rise in lactate concentration, suggest an immediate activation of ethanol production.

When *C. carassius* is exposed to anoxia, lactate is shuttled to the muscles for conversion to ethanol. The continuous rise in plasma [lactate] but constant low muscle concentration indicates either that 1) the lactate shuttling from blood to muscle is quite slow, or 2) that the lactate shuttle is tightly regulated in a way that no more than the lactate that can be instantly converted to ethanol is transported into the tissue. The first option seems most plausible since Mandic et al. (2008) measured 7 μmol lactate g^{-1} in white muscle of *C. auratus* after 10 h anoxia, indicating higher transport of lactate into the tissue than can be quickly converted. The presence of lactate accumulation in the study by Mandic et al. (2008) but not in the present study is potentially a species-specific difference, but a slow shuttling mechanism combined with the relatively short exposure period may have prevented detection of any lactate accumulation in the muscle tissue in the present study.

5. Conclusions

Despite the significant difference in lactate accumulation, no difference in EPHOC:O_2 deficit ratio could be detected between *C. carassius* and *C. carpio*. As discussed above, the measured EPHOC for *C. carassius* is in agreement with previous studies by being small compared to less hypoxia tolerant and non-ethanol-producing species, but how *C. carpio* achieves such a small EPHOC after near lethal anoxia exposure, without depressing its metabolism or converting lactate into ethanol, is not easily explained. Despite its inability to produce ethanol in response to oxygen deprivation, the metabolic profile of *C. carpio* is more similar to the ethanol-producing members of *Cyprinidae* than other taxa that cannot produce ethanol. *C. carpio* accumulates a greater EPHOC and requires longer recovery time than *C. carassius*, but this is likely related to the

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severity of the anoxic exposure used in this study relative to the overall anoxia survival capability of each species. No lactate accumulation in white muscle of *C. carassius* and less severe accumulation in plasma, in comparison to *C. carpio*, probably indicates rapid implementation of the ethanol production pathway upon exposure to anoxia, but a slow shuttling mechanism from plasma to muscle. The results of the present study emphasize the importance of metabolic depression to *C. carassius* and PCr buffering capacity to *C. carpio*, and thus factors other than ability to produce ethanol are suggested to contribute in large part to EPHOC development in fishes.

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References

Behrens, J., Steffensen, J., 2007. The effect of hypoxia on behavioural and physiological aspects of lesser sandeel, *Ammodytes tobianus* (Linnaeus, 1785). Mar. Biol. 150, 1365–1377.

Bickler, P.E., Buck, L.T., 2007. Hypoxia tolerance in reptiles, amphibians, and fishes: life with variable oxygen availability. Annu. Rev. Physiol. 69, 145–170.

Brett, J.R., 1964. The respiratory metabolism and swimming performance of young sockeye salmon. J. Fish. Res. Board Can. 21, 1183–1226.

Bushnell, P.G., Steffensen, J.F., Schurmann, H., Jones, D.R., 1994. Exercise metabolism in two species of cod in arctic waters. Polar Biol. 14, 43–48.

Campbell, H.A., Fraser, K.P.P., Bishop, C.M., Peck, L.S., Egginton, S., 2008. Hibernation in an Antarctic fish: on ice for winter. PLoS One 3, e1743.

Dalla Via, J., Van den Thillart, G., Cattani, O., de Zwaan, A., 1994. Influence of long-term hypoxia exposure on the energy metabolism of *Solea solea*. 11. Intermediary metabolism in blood, liver and muscle. Mar. Ecol. Prog. Ser. 111, 17–27.

Dalla Via, J., van den Thillart, G., Cattani, O., Cortesi, P., 1997. Environmental versus functional hypoxia/anoxia in sole *Solea solea*: the lactate paradox revisited. Mar. Ecol. Prog. Ser. 154, 79–90.

Farrell, A.P., Richards, J.G., 2009. Defining hypoxia: an integrative synthesis of the responses of fish to hypoxia. Fish Physiol. 27, 487–503.

Hallman, T.M., Rojas-Vargas, A.C., Jones, D.R., Richards, J.G., 2008. Differential recovery from exercise and hypoxia exposure measured using 31P- and 1H-NMR in white muscle of the common carp *Cyprinus carpio*. J. Exp. Biol. 211, 3237–3248.

Hochachka, P.W., 1986. Defense strategies against hypoxia and hypothermia. Science 231, 234–241.

Hochachka, P.W., Mommsen, T.P., 1983. Protons and anaerobiosis. Science 219, 1391.

Holopainen, I.J., Hyvärinen, H., 1985. Ecology and physiology of crucian carp [*Carassius carassius* (L.)] in small Finnish ponds with anoxic conditions in winter. Verh. Int. Ver. Theor. Angew. Limnol. 22, 2566–2570.

Holopainen, I.J., Hyvärinen, H., Piironen, J., 1986. Anaerobic wintering of crucian carp (*Carassius carassius* L.) — II. Metabolic products. Comp. Biochem. Physiol. A: Physiol. 83, 239–242.

Hyvärinen, H., Holopainen, I.J., Piironen, J., 1985. Anaerobic wintering of crucian carp (*Carassius carassius* L.) I. Annual dynamics of glycogen reserves in nature. Comp. Biochem. Physiol. A: Physiol. 82, 797–803.

Johnston, I.A., Bernard, L.M., 1983. Utilization of the ethanol pathway in carp following exposure to anoxia. J. Exp. Biol. 104, 73–78.

Jordan, A.D., Steffensen, J.F., 2007. Effects of ration size and hypoxia on specific dynamic action in the cod. Physiol. Biochem. Zool. 80, 178–185.

Killen, S.S., Costa, I., Brown, J.A., Gamperl, A.K., 2007. Little left in the tank: metabolic scaling in marine teleosts and its implications for aerobic scope. Proc. R. Soc. B 274, 431–438.

Lin, C., Wu, H., Tjeerdema, R., Viant, M., 2007. Evaluation of metabolite extraction strategies from tissue samples using NMR metabolomics. Metabolomics 3, 55–67.

Mandic, M., Lau, G.Y., Nijjar, M.M.S., Richards, J.G., 2008. Metabolic recovery in goldfish: a comparison of recovery from severe hypoxia exposure and exhaustive exercise. Comp. Biochem. Physiol. C 148, 332–338.

Maxime, V., Pichavant, K., Boeuf, G., Nonnotte, G., 2000. Effects of hypoxia on respiratory physiology of turbot, *Scophthalmus maximus*. Fish Physiol. Biochem. 22, 51–59.

Muusze, B., Marcon, J., van den Thillart, G., Almeida-Val, V., 1998. Hypoxia tolerance of Amazon fish: respirometry and energy metabolism of the cichlid *Astronotus Ocellatus*. Comp. Biochem. Physiol. A 120, 151–156.

Nilsson, G., 1988. A comparative study of aldehyde dehydrogenase and alcohol dehydrogenase activities in crucian carp and three other vertebrates: apparent adaptations to ethanol production. J. Comp. Physiol. B 158, 479–485.

Nilsson, G.E., 1990. Long-term anoxia in crucian carp: changes in the levels of amino acid and monoamine neurotransmitters in the brain, catecholamines in chromaffin tissue, and liver glycogen. J. Exp. Biol. 150, 295–320.

Nilsson, G.E., 2001. Surviving anoxia with the brain turned on. News Physiol. Sci. 16, 217–221.

Nilsson, G.E., Renshaw, G.M.C., 2004. Hypoxic survival strategies in two fishes: extreme anoxia tolerance in the North European crucian carp and natural hypoxic preconditioning in a coral-reef shark. J. Exp. Biol. 207, 3131–3139.

Peake, S.J., Farrell, A.P., 2006. Fatigue is a behavioural response in respirometer-confined smallmouth bass. J. Fish Biol. 68, 1742–1755.

Pichavant, K., Maxime, V., Thebault, M.T., Ollivier, H., Garnier, J.P., Bousquet, B., Diouris, M., Boeuf, G., Nonnotte, G., 2002. Effects of hypoxia and subsequent recovery on turbot *Scophthalmus maximus*: hormonal changes and anaerobic metabolism. Mar. Ecol. Prog. Ser. 225, 275–285.

Piironen, J., Holopainen, I.J., 1986. A note on seasonality in anoxia tolerance of Crucian carp (*Carassius carassius* (L)) in the laboratory. Ann. Zool. Fenn. 23, 335–338.

Richards, J.G., Heigenhauser, G.J.F., Wood, C.M., 2002. Glycogen phosphorylase and pyruvate dehydrogenase transformation in white muscle of trout during high-intensity exercise. Am. J. Physiol. Reg. Int. Comp. Physiol. 282, R828–R836.

Scarabello, M., Heigenhauser, G.J.F., Wood, C.M., 1991. The oxygen debt hypothesis in juvenile rainbow trout after exhaustive exercise. Respir. Physiol. 84, 245–259.

Schurmann, H., Steffensen, J.F., 1997. Effects of temperature, hypoxia and activity on the metabolism of juvenile Atlantic cod. J. Fish Biol. 50, 1166–1180.

Shoubridge, E.A., Hochachka, P.W., 1980. Ethanol: Novel end product of vertebrate anaerobic metabolism. Science 209, 308–309.

Stecyk, J.A.W., Farrell, A.P., 2002. Cardiorespiratory responses of the common carp (*Cyprinus carpio*) to severe hypoxia at three acclimation temperatures. J. Exp. Biol. 205, 759–768.

Stecyk, J.A.W., Stenslökken, K.O., Farrell, A.P., Nilsson, G.E., 2004. Maintained cardiac pumping in anoxic crucian carp. Science 306, 77.

Steffensen, J., 1989. Some errors in respirometry of aquatic breathers: how to avoid and correct for them. Fish Physiol. Biochem. 6, 49–59.

Steffensen, J.F., Johansen, K., Bushnell, P.G., 1984. An automated swimming respirometer. Comp. Biochem. Physiol. A: Physiol. 79, 437–440.

Svendsen, J.C., Tudorache, C., Jordan, A.D., Steffensen, J.F., Aarestrup, K., Domenici, P., 2010. Partition of aerobic and anaerobic swimming costs related to gait transitions in a labriform swimmer. J. Exp. Biol. 213, 2177–2183.

Svendsen, J.C., Steffensen, J.F., Aarestrup, K., Frisk, M., Etzerodt, A., Jyde, M., 2012. Excess post hypoxic oxygen consumption in rainbow trout *Oncorhynchus mykiss*: recovery in normoxia and hypoxia. Can. J. Zool. 90, 1–11.

van den Thillart, G., van Waarde, A., 1993. The role of metabolic acidosis in the buffering of ATP by phosphagen stores in fish: an *in vivo* NMR study. In: Hochachka, P.W., Lutz, P.L., Sick, T., Rosenthal, M., van den Thillart, G. (Eds.), Surviving Hypoxia: Mechanisms of Control and Adaptation. CRC Press, pp. 237–252.

van den Thillart, G., Verbeek, R., 1991. Anoxia-induced oxygen debt of goldfish (*Carassius auratus* L.). Physiol. Zool. 64, 525–540.

van den Thillart, G., van Berge-Henegouwen, M., Kesbeke, F., 1983. Anaerobic metabolism of goldfish, *Carassius auratus* (L.): ethanol and CO_2 excretion rates and anoxia tolerance at 20, 10 and 5 °C. Comp. Biochem. Physiol. A: Physiol. 76, 295–300.

van der Linden, A., Verhoye, M., Nilsson, G.E., 2001. Does anoxia induce cell swelling in carp brains? In vivo MRI measurements in crucian carp and common carp. J. Neurophysiol. 85, 125–133.

van Ginneken, V., van den Thillart, G., 2009. Metabolic depression in fish measured by direct calorimetry: a review. Thermochim. Acta 483, 1–7.

van Ginneken, V., van den Thillart, G., Addink, A., Erkelens, C., 1995. Fish muscle energy metabolism measured during hypoxia and recovery: an in vivo 31P-NMR study. Am. J. Physiol. Reg. Int. Comp. Physiol. 268, R1178–R1187.

van Ginneken, V., Nieveen, M., Van Eersel, R., Van den Thillart, G., Addink, A., 1996. Neurotransmitter levels and energy status in brain of fish species with and without the survival strategy of metabolic depression. Comp. Biochem. Physiol. A 114, 189–196.

van Ginneken, V., Boot, R., Murk, T., van den Thillart, G., Balm, P., 2004a. Blood plasma substrates and muscle lactic-acid response after exhaustive exercise in common carp and trout: indications for a limited lactate-shuttle. Anim. Biol. 54, 119–130.

van Ginneken, V.J.T., Snelderwaard, P., van der Linden, A., van der Reijden, R., van den Thillart, G., Kramer, K., 2004b. Coupling of heart rate with metabolic depression in fish: a radiotelemetric and calorimetric study. Thermochim. Acta 414, 1–10.

van Raaij, M.T.M., Pit, D.S.S., Balm, P.H.M., Anton, B., van den Thillart, G., 1996. Behavioral strategy and the physiological stress response in rainbow trout exposed to severe hypoxia. Horm. Behav. 30, 85–92.

van Waarde, A., van den Thillart, G., Erkelens, C., Addink, A., Lugtenburg, J., 1990. Functional coupling of glycolysis and phosphocreatine utilization in anoxic fish muscle. An in vivo 31P NMR study. J. Biol. Chem. 265, 914–923.

Vianen, G.J., Thillart, G.E.E.J., Van Kampen, M., Van Heel, T.I., Steffens, A.B., 2001. Plasma lactate and stress hormones in common carp (*Cyprinus carpio*) and rainbow trout (*Oncorhynchus mykiss*) during stepwise decreasing oxygen levels. Neth. J. Zool. 51, 33–50.

Viant, M.R., Rosenblum, E.S., Tjeerdema, R.S., 2003. NMR-based metabolomics: a powerful approach for characterizing the effects of environmental stressors on organism health. Environ. Sci. Technol. 37, 4982–4989.

Virani, N.A., Rees, B.B., 2000. Oxygen consumption, blood lactate and inter-individual variation in the gulf killifish, *Fundulus grandis*, during hypoxia and recovery. Comp. Biochem. Physiol. A 126, 397–405.

Wilkie, M.P., Pamenter, M.E., Alkabie, S., Carapic, D., Shin, D.S.H., Buck, L.T., 2008. Evidence of anoxia-induced channel arrest in the brain of the goldfish (*Carassius auratus*). Comp. Biochem. Physiol. C 148, 355–362.

Wissing, J., Zebe, E., 1988. The anaerobic metabolism of the bitterling *Rhodeus amarus* (cyprinidaf, teleostei). Comp. Biochem. Physiol. B 89, 299–303.

Zhou, B.S., Wu, R.S.S., Randall, D.J., Lam, P.K.S., Ip, Y.K., Chew, S.F., 2000. Metabolic adjustments in the common carp during prolonged hypoxia. J. Fish Biol. 57, 1160–1171.

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