Aquaculture 580 (2024) 740280

Contents lists available at ScienceDirect

Aquaculture

journal homepage: www.elsevier.com/locate/aquaculture

The relationship among acceleration, metabolism, and tail beat frequency of channel catfish (*Ictalurus punctatus*) during critical swimming speed tests

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ARTICLE INFO

Keywords:
Accelerometry
Aquaculture
Bioenergetics
Biologging
Physiology
Swim flume

ABSTRACT

Accelerometers collect real-time, high-resolution data over extended time periods providing insight into behavior, movement, and physiology of free-swimming animals in captive or natural environments. In aquaculture settings, acceleration can also be used as a proxy for metabolism, providing useful information to inform bioenergetic models and indices of fish welfare. However, defining the acceleration-metabolic relationship for species of interest is a critical first step before information can be applied. Because catfish aquaculture has a large economic impact in the USA, examining the use of acceleration as a proxy for continuous measures of metabolism could prove beneficial. Here, accelerometers were used in channel catfish (*Ictalurus punctatus*) to examine the relationship between acceleration as measured by overall dynamic body acceleration (ODBA) and metabolism as measured by oxygen consumption (MO_2), and how these variables changed with increasing water velocity during critical swimming speed tests. Tail beat frequency was used to validate ODBA measurements. There was a clear relationship between ODBA and water velocity, and metabolism and water velocity, with ODBA increasing 3.2-fold and metabolism increasing 2-fold as water velocity increased. Tail beat frequency also increased 1.5-fold with each increase in water velocity, validating our acceleration results. There was also a clear relationship between ODBA and metabolism, with metabolism increasing 1.1-fold as the ODBA values increased. Results in this study show accelerometers can act as a proxy for metabolic measures in channel catfish and provide the basis for further applications in bioenergetics or operational welfare to benefit the aquaculture industry.

1. Introduction

Technological advancements in biologging have greatly improved our ability to understand animal responses to changing environments through the monitoring of behavior, movement, and physiology. Providing high-resolution data in real-time over extended periods (Thorsteinsson, 2002), measures of acceleration, heart rate, depth, and temperature can be used to determine behaviors and physiological changes for a variety of species. For instance, time budgets in various behaviors have been documented using accelerometers in a variety of mammalian species (e.g., European badgers, *Meles meles*, McClune et al., 2015; snowshoe hares, *Lepus americanus*, Studd et al., 2019), livestock (e.g., yearling steers, Brennan et al., 2021; Hereford X Angus cattle, Sprinkle et al., 2021), and avian species (e.g., cormorants, *Phalcrocorax carbo*; Wilson et al., 2006; little penguins, *Eudyptula minor*, Zimmer et al., 2011). Today, new age biologging technology allows fine-scale glimpses into the lives of free-ranging animals under changing environments.

Recording and obtaining fine-scale information about the lives of free-swimming animals has proved more difficult. Beginning with a capillary tube to record the depth of a diving harpooned fin whale (*Balaenoptera physalus*; Kooyman, 2004), data collection has progressed toward the use of small accelerometers that can be attached to or surgically implanted into the animal's body (Hockersmith and Beeman, 2012; Kooyman, 2004; Thorsteinsson, 2002). Yet, the use of these types of bio-loggers is disproportionately skewed among taxa, with only 11.2% usage in fishes (Brown et al., 2013). Despite limited use in fish species, accelerometers and other biologging tools (e.g., heart rate monitors, depth loggers) are becoming of increasing interest, particularly in aquaculture as they have potential to benefit production. They provide an important tool to determine metabolic rates and develop daily and seasonal energy budgets in production settings, useful for fine-

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https://doi.org/10.1016/j.aquaculture.2023.740280
Received 27 April 2023; Received in revised form 17 October 2023; Accepted 24 October 2023

tuning management and feeding practices, particularly where visibility limits direct observation such as in earthen catfish ponds (Boyd, 2004). In addition, accelerometers can serve as operational welfare indicators; depth and temperature loggers have been used on farm raised Atlantic salmon (*Salmo salar*) to assess competition for preferred thermal spaces in thermally stratified production cages (Johansson et al., 2009). More recently, various biologgers including accelerometers have been used to link activity to stress levels in farmed Atlantic salmon (Svendsen et al., 2020), opening the possibility of remote stress monitoring in commercial production. These examples indicate biologging can be a valuable tool to aquaculture producers, and that applications of such technologies could be transformative to the field.

Changes in acceleration can also be related to an animal's physiological responses to changing environments, as acceleration can be a proxy for oxygen consumption, a measure of aerobic metabolism, which can be used to assess energy expenditure. For instance, Wilson et al. (2006) found both oxygen consumption and carbon dioxide production rates to be positively associated with overall dynamic body acceleration (ODBA) in great cormorants (*Phalacrocorax carbo*) while exercising on a treadmill. Similarly, Halsey et al. (2008) concluded ODBA and heart rate were the most powerful predictors of oxygen consumption in humans (*Homo sapiens*) on a treadmill. Both Halsey et al. (2008) and Wilson et al. (2006) suggested ODBA can provide accurate estimates of metabolism for various terrestrial animals in their natural environments. These same concepts are also commonly supported with swimming speed and metabolism in fishes, where increasing acceleration is correlated with increasing swimming speeds and metabolic rates (Brownscombe et al., 2018; Clark et al., 2010; Gleiss et al., 2010; Wilson et al., 2013; Wright et al., 2014; Zrini and Gamperl, 2021). Such knowledge can be beneficial in improving bioenergetic models to ultimately improve productivity within aquaculture practices.

Since its inception in the 1960s, the catfish aquaculture industry has rapidly expanded and is now the largest finfish aquaculture industry in the United States (Quintero et al., 2009; Reichley et al., 2018; United States Department of Agriculture-National Agricultural Statistics Service, 2021). Although a multitude of species have been evaluated for culture suitability, channel catfish (*Ictalurus punctatus*) have been the historically cultivated species and remain popular within the industry (Reichley et al., 2018; Tucker and Hargreaves, 2004). Previous studies have investigated metabolic rates and swimming performance of channel catfish, particularly at warm temperatures (Beecham, 2004; Gerhart, B.J. & Allen, P.J., *unpublished results*; Hocutt, 1973; Nepal et al., 2021). The relationships among acceleration, metabolism and swimming speed have been investigated in Atlantic salmon and other fishes; however, there is limited understanding of these relationships in channel catfish. Past bioenergetic models have relied on laboratory-derived data (Cuenco et al., 1985), which hold value, however, in-pond measurements incorporating daily or seasonal environmental conditions would be directly applicable to commercial production practices. A further understanding of these relationships through accelerometry can provide insight into energy expenditure throughout the production cycle and environmental changes while also determining optimal production conditions (Brijs et al., 2021). Therefore, the objective of this study was to examine the relationship between acceleration and metabolic rates during swimming performance tests of channel catfish, and to evaluate whether these movement metrics could be used to increase production. Based on similar studies, we predicted that acceleration and metabolic rates would increase with increasing water velocities, and that acceleration and metabolism would be related.

2. Materials and methods

2.1. Ethical statement

All experimental work and procedures were conducted in accordance with approved Mississippi State University Institutional Animal Care and Use Committee animal welfare laws, guidelines and policies (Protocol # 20-526).

2.2. Fish rearing

Juvenile Delta Select channel catfish were spawned and reared by the (USDA) Agricultural Research Service (ARS) at the Thad Cochran National Warmwater Center in Stoneville, Mississippi. The Delta Select strain was established in 2006 and derived from 10 commercial catfish farms for selecting improved growth and carcass yield by the USDA's Warmwater Aquaculture Research Unit (Bosworth et al., 2020). Fish were subsequently transported to Mississippi State University's South Farm Aquaculture Facility and gradually acclimated for two weeks from 27 °C (temperature they were at in the Thad Cochran National Warmwater Center) to 20 °C, a temperature representative of conditions that may occur through a broad portion of the year (i.e., spring, fall, and winter (Bly and Clem, 1992; Burger et al., 2018; Tucker and van der Ploeg, 1993)), and at which active feeding and growth occurs (Hargreaves et al., 2004). Initial fish size was (mean ± SE) 36.08 ± 1.68 grams and 174.1 ± 2.7 mm in total length. Fish were held in three, 355-L tanks, with 50 catfish per tank across an independent recirculating aquaculture system supplied with well water and forced air via air stones. Fish were held at a 12-h light: 12-h dark photoperiod. Temperature, salinity, and dissolved oxygen were measured with a dissolved oxygen meter (Pro 2030, YSI Inc., Yellow Springs, OH, USA) and pH with a pH probe (EcoSense pH100A, YSI Inc.). Water quality conditions maintained for the duration of the study included: temperature 20.1 ± 0.0 °C, dissolved oxygen 7.6 ± 0.0 mg/L, salinity 0.8 ± 0.0, and pH 7.2 ± 0.02, representing levels appropriate for channel catfish (Tucker and Robinson, 1990). Fish were fed daily to satiation with a commercial diet (Fry/Fingerling Catfish Food; Fishbelt Feeds Inc., Moorhead, MS, USA) consisting of 35% crude protein, 2.5% crude fat, 7% crude fiber, and 0.4% phosphorus. Tanks were monitored during a 30-min feeding period in the morning (between 07:00 and 12:00); if all pellets were consumed, additional pellets were added until feed consumption ceased, and pellets remained at the surface. After feeding, remaining pellets were netted and removed from the tanks. Biofilters were backwashed daily, and tanks were siphoned and/or scrubbed weekly, and any dead fish were immediately removed upon discovery.

2.3. Surgical procedure

To measure catfish acceleration, Star-Oddi micro-ACT tags (Garðabær, Iceland; length: 25.4 mm, diameter: 8.3 mm, weight in air: 3.3 grams) were surgically implanted into channel catfish ($n = 8$, ranging from 267.25 to 315.06 grams in wet weight, and 310 to 331 mm in total length). The tags did not exceed 2% of the fish's body mass. Prior to all surgeries, each tag was inserted into the tag-computer interface (communication box) and the start date, start time (here, 0900), sampling intervals (here, 2 min), and sampling frequency (here, 10 Hz) were programmed with Mercury software (version 5.99, Star-Oddi, Garrard, Iceland). To ensure a post-absorptive state prior to the surgical procedure, individual fish were placed in a separate 355-L holding tank where food was withheld for 36 h. All catfish were anaesthetized in a buffered solution (150 mg/L MS-222, 400 mg/L $NaHCO_3$) and weighed (nearest 0.01 gram). Fish were then placed on a wet surface in the supine position and gills were irrigated with aerated water containing a buffered anesthetic (75–100 mg/L, MS-222, 400 mg/L $NaHCO_3$) throughout the entire surgical procedure. An incision was made in the mid-ventral region of the fish in the sagittal plane. A piece of non-absorbable silk suture (3–0) was tied around the tag, similar to Zrini and Gamperl (2021), and the tag was inserted with the blunt end in the posterior direction. The tag was then guided forward with the attached suture, past the incision site, to orient it closer to the pericardial cavity. Once the tag was oriented, the sutures around the tag were passed through the body wall cavity with a curved needle and tied into place (Suppl. Fig. 1

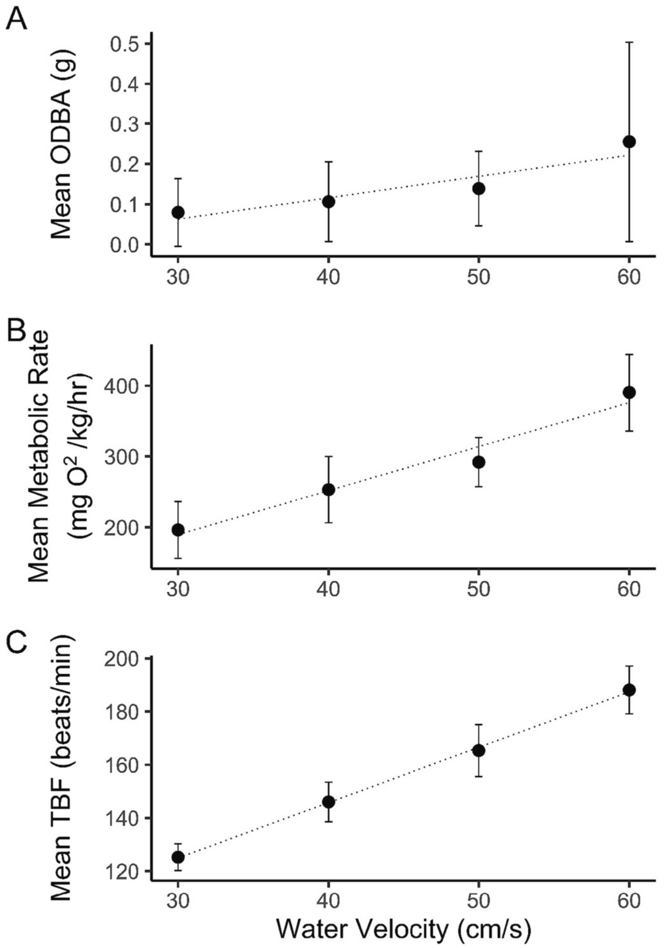

Fig. 1. Mean ± SD of A) overall dynamic body acceleration (ODBA), B) metabolic rate, and C) tail beat frequency (TBF) for channel catfish (*Ictalurus punctatus*) at each water velocity.

for tag orientation). The incision was then closed with 2–3 non-absorbable sutures. Following the surgical procedure, and similar to Zrini and Gamperl (2021), the fish recovered in the holding tank for 5 days with several other catfish to encourage feeding during the recovery period. While unable to verify food consumption by individual fish, most fish appeared to eat without incident as pellet quantity decreased throughout the feeding time. Fish surgeries across the 8 individuals occurred over 8 successive days to ensure only one flume test occurred per day.

2.4. *Critical swimming speed test*

To examine the relationship between acceleration and metabolism, we used a 98-L Blazka-style swim flume with a cylindrical swimming chamber (100.3 cm length, 15.2 cm diameter). The swim flume was covered on the observer's side with a dark, plastic covering to reduce external stimuli. A flow meter and probe (Flo-Mate 2000, Marsh McBirney Inc., Frederick, Maryland, USA) were used to determine the water velocity (cm/s) and calibrate to revolutions per minute (RPM) in a tachometer display box. The RPM measurements for each water velocity were an average of measurements at the bottom, middle, and top of the swim flume fish chamber. During the swimming tests, oxygen consumption was measured with a fiber optic oxygen probe (Oxygen Dipping Probe, PreSens), a connected meter (OXY-1 SMA, PreSens), and associated computer software (Measurement Studio 2, 3.0.3.1703, PreSens). The oxygen probe was calibrated at 0% and 100% oxygen saturation prior to placement of each fish into the swim flume.

On its 6th day of recovery, each fish was relocated to the swim flume to acclimate overnight at 10 cm/s. To ensure a post-absorptive state, food was withheld for 36 h before placing a fish in the swim flume and 48 h before the swimming test. During acclimation, water was continuously recirculated to the flume from an adjacent circular 200-L tank where temperature was maintained at 20 °C by a chiller (Cyclone Drop-In ½ Hp, Aqua Logic, Inc.) and oxygen by air stones. After overnight acclimation (15–16 h) and before oxygen measurements, the water velocity was increased by 10 cm/s to 20 cm/s for 5 min. After 5-min, the water velocity was increased again to 30 cm/s. At this point oxygen measurements began and the recirculating pump connecting the swim flume to the adjacent 200-L tank was turned off to prevent oxygenated water from flowing into the swim flume during the 30-min measurement period. After the 30-min measurement period, an 11-min flush followed, allowing oxygenated water from the 200-L adjacent tank to pump into the swim flume at 9 L/min to complete one water turnover. After the 11-min flush, this 41-min procedure was repeated with increasing water velocities of 10 cm/s. The swim test was concluded once the fish fatigued, which was determined by three uninterrupted 10-s impingements of the fish on the back screen of the swim flume following Allen et al. (2021). After each impingement, the timer was paused, and water velocity was reduced to 0 cm/s to allow the fish to remove itself from the back screen. Immediately after the fish removed itself from the back screen, the water velocity was increased to the designated velocity and the timer was resumed. To validate accelerometer readings during the swim test, tail beat frequency (TBF) was recorded via visual observation for 30 s every 5, 15, and 25 min during each measurement period. Counts were then extrapolated to 1 min and the averages were used for each water velocity.

After completing the swimming test, the fish was anaesthetized in a buffered solution (150 mg/L MS-222, 400 mg/L $NaHCO_3$), axis lengths were obtained from the widest points of the body with calipers to the nearest mm, and the tag was removed from the fish's body cavity. The tag was immediately inserted into the communication box for data retrieval. We determined critical swimming velocity (U_{crit}) using Brett (1964):

$$U_{crit} = V_f + V_i\left(T_f/T_i\right)$$

where V_f is the final water velocity at which the fish swam the entire 41-min period (2460 s); V_i is the increment of water velocity increase (10 cm/s); T_f is the time swam at the water velocity of fatigue; and T_i is the time increment for each water velocity (i.e., 2460 s). To determine if a solid-blocking correction was needed for U_{crit} calculations, cross-sectional areas of fish were compared to the flume. For the fish, cross-sectional area was determined using an equation for the area (A) (mm^2) of an ellipse:

$$A = \pi r_v r_h$$

where r_v is the radius (mm) of the vertical axis and r_h is the radius (mm) of the horizontal axis. Because the maximum cross-sections of fish did not exceed 10% of the cross-sectional flume area, no solid blocking corrections were required for critical swimming speed (Bell and Terhune, 1970).

Using measures of oxygen consumption and fish volume and weights, we calculated metabolic rates (MO_2) for each fish at each water velocity similar to Cech (1990):

$$MO_2 = (msV)/M$$

where m is the slope derived from the oxygen consumption rate over the 30-min measurement period; s is the solubility coefficient of oxygen in water (0.439 mg O_2/L/kPa across trials); V is the flume volume (i.e., the difference between the full flume volume (98 L) and the fish volume (converted from grams to mL); and M is the fish's mass (nearest 0.01 gram). The highest metabolic rate before fatigue for each fish served as the maximum metabolic rate for that sample, following Messina-

Henriquez et al. (2022). Standard metabolic rate was extrapolated from the y-intercept (i.e., 0 cm/s) based on the above relationship (Allen et al., 2021; Clark and Seymour, 2006; Cucco et al., 2012; Parsons and Sylvester Jr., 1992). The difference between the maximum metabolic rate and standard metabolic rate was used to determine the metabolic scope of each fish, following Clark et al. (2013).

$$Metabolic\ scope = maximum\ MO_2 - standard\ MO_2$$

2.5. Statical analyses

Total body acceleration (i.e., the vector sum of acceleration across three axes) is typically calculated from raw tri-axial accelerometry data as either ODBA or the vector of the dynamic body acceleration (VeDBA). To obtain ODBA or VeDBA, static acceleration from gravity must first be removed, and average static acceleration was calculated with a running median smoothing window of 51 samples (Shepard et al., 2008, Studd et al., 2019; see Suppl. Fig. 2 for window size selection). Static acceleration was removed from total acceleration to retain only acceleration generated from the movement of the fish. In this case, since ODBA and VeDBA were highly correlated (Pearson's correlation coefficient = 0.99; Suppl. Fig. 3), the acceleration-metabolic-water velocity relationship was examined with ODBA.

Of the 8 fish, only one fish completed the swim test up to 70 cm/s; therefore, data were only analyzed up to 60 cm/s across fish. As measures of ODBA, metabolism, and TBF followed an exponential relationship (Fig. 1), data were natural-log transformed to better fit a linear mixed effect model with the nlme package in R version 1.3.1093 (R Core Team, 2019). We first examined ODBA, metabolic rate, and TBF separately (response variables) in relation to water velocity (predictor variable), and then examined metabolic rate (response variable) in relation to ODBA (predictor variable); in each model, the individual fish were a random effect to account for the variability between fish. Beta coefficient and intercept values from each model were used to calculate predicted relationships and the level of statistical significance for all analyses was considered as $P \leq 0.05$.

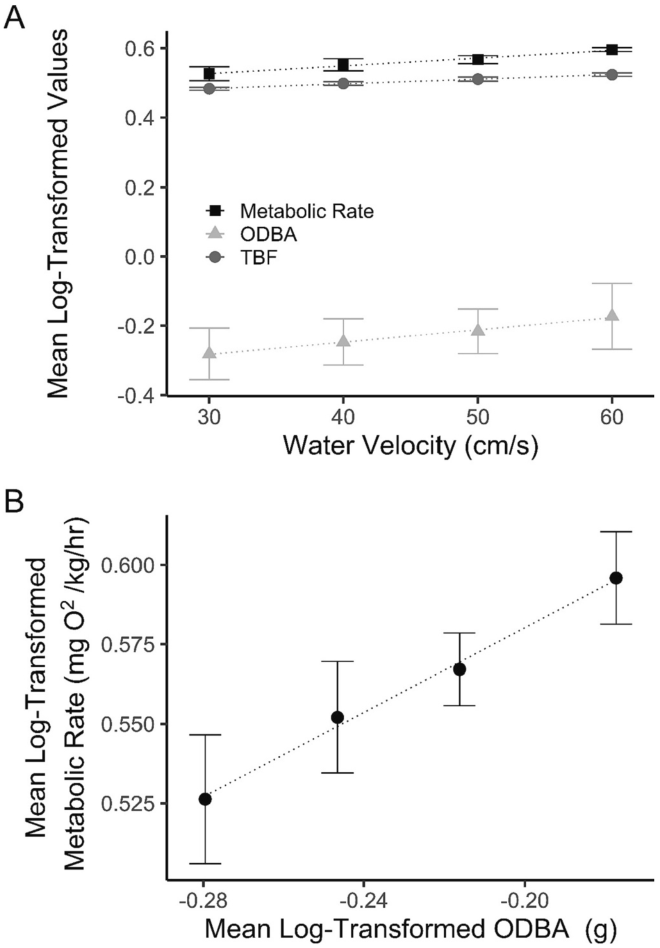

Fig. 2. Mean ± SD natural-log transformed A) metabolic rate, overall dynamic body acceleration (ODBA), and tail beat frequency (TBF) counts related to water velocity and B) metabolic rate related to transformed overall dynamic body acceleration (ODBA) for channel catfish (*Ictalurus punctatus*). Panels also have a model predicted lines.

3. Results

Fish critical swimming speed, U_{crit}, ranged from 53.7 to 61.4 cm/s (mean ± SE: 57.2 ± 1.0). Across the 8 sampled fish, a sum of 369,012 ODBA measurements, 28 metabolism measurements, and 32 TBF measurements were recorded. Extrapolated standard metabolic rates ranged from 60.1 to 152.0 mg O_2/kg/h (98.5 ± 12.8), maximum metabolic rates ranged from 296.6 to 461.2 mg O_2/kg/h (390.5 ± 20.6), and metabolic scopes ranged from 144.6 to 378.7 mg O_2/kg/h (292.1 ± 27.8).

Overall dynamic body acceleration ranged from 0 to 7.6 g, TBF from 117 to 199 beats/min, and metabolic rates from 152.2 to 461.2 mg O_2/kg/h across increasing water velocities. There was a relationship between water velocity and fish acceleration, with ODBA increasing 3.2-fold from water velocities of 30 cm/s to 60 cm/s (ODBA; Table 1, Fig. 1A). This acceleration was validated by observations of TBF, with TBF increasing 1.5-fold from water velocity of 30 cm/s to 60 cm/s (Table 1, Fig. 1B). A similar relationship was observed for metabolism, with metabolic rates increasing about 2-fold from water velocity of 30 cm/s to 60 cm/s (Table 1, Fig. 1C). Based on the model outputs for the three response variables, for every unit change in velocity ODBA was 1.6 times greater than metabolic rate while metabolic rate was 1.7 times greater than TBF (Fig. 2A). Also, ODBA was 2.7 times greater than TBF for every unit change in velocity (Fig. 2A). Finally, there was a relationship between ODBA and metabolism with metabolic rates increasing about 1-fold from increasing ODBA values throughout the swimming performance tests (Table 1, Fig. 2B).

4. Discussion

Biologging tools have allowed scientists to study multiple realms of biology and the responses to various conditions of free-ranging and free-swimming animals in either their natural or captive environments (Cooke et al., 2004). These loggers establish relationships between various biological processes – here, movement and metabolic processes - which provides critical information regarding the animals' bioenergetic demands under changing conditions. As predicted, we showed acceleration (measured by ODBA), TBF, and metabolism increased in channel catfish as water velocity increased during swimming tests. Furthermore, the three variables were related to each other in different ways, with ODBA increasing at a greater rate over metabolic rate and TBF over increasing increments of water velocity. Through a better understanding

Table 1
Beta coefficients, intercepts, *P*-values, and model fit (R^2) for linear mixed effect models relating water velocity to natural-log transformed variables overall dynamic body acceleration (ODBA), metabolism, and tail beat frequency (TBF), as well as a model relating metabolism to ODBA.

Model	Beta coefficients	Intercept	*P*-value	R^2
ODBA ~ Water Velocity	0.0035	−0.39	< 0.001	0.21
Metabolism ~ Water Velocity	0.0022	0.46	< 0.001	0.84
TBF ~ Water Velocity	0.0013	0.44	< 0.001	0.97
Metabolism ~ ODBA	0.6647	0.71	< 0.001	0.90

of these relationships relative to swimming speed and environmental conditions, these values provide the fundamental knowledge for the parametrization of developing bioenergetic models to improve aquaculture production. For instance, accelerometers provide insight into swimming activity and metabolic rates, parameters that can be used to estimate energy budgets of cultured fish in relation to feed conversion rates (Brijs et al., 2021). Moreover, these energetic expenditure estimates can determine the metabolic costs required for sustaining life and the remaining scope for processes like feeding, swimming, and growth (Brijs et al., 2021).

In the current study, ODBA had an exponential relationship with increasing water velocity (here, a surrogate for swimming speed). This has also been documented in sockeye salmon (*Oncorhynchus nerka;* Clark et al., 2010), European sea bass (*Dicentrarchus labrax;* Wright et al., 2014), and Atlantic salmon (Zrini and Gamperl, 2021). While the relationship between ODBA and water velocity was weak, potentially due to high amounts of variability in ODBA values across water velocities and fish, there was still enough information for a relationship to be discerned. This weak relationship between ODBA and water velocity is not typical as stronger relationships have been found in sockeye salmon ($R^2 = 0.85$; Clark et al., 2010) and European sea bass ($R^2 = 0.87$; Wright et al., 2014). It is also notable that this relationship is not always exponential. For example, Brownscombe et al. (2018) reported a positive linear relationship between ODBA and swimming speed in bonefish (*Albula vulpes*). Therefore, we contend that this relationship should be evaluated on a species-by-species basis and treated as a hypothesis to be tested, as these relationships are likely not generalizable across taxa.

We also detected an exponential relationship between TBF and increasing water velocity, an expected result given fish increase swimming speed under faster water velocities. One benefit of working with aquatic species is acceleration is commonly tied to observations of body undulations. For example, TBF has been shown to have a direct relationship with water velocity in a wide variety of species including Atlantic cod (*Gadus morhua*; Lurman et al., 2007), cojinoba (*Seriolella violacea*; Allen et al., 2021), jack mackerel (*Trachurus symmetricus*; Hunter and Zweifel, 1971), and various catfish species (Gerhart, B.J. & Allen, P.J., *unpublished results*). Moreover, Zrini and Gamperl (2021) concluded acceleration values could serve as estimates of swimming speed and TBF from Star Oddi centi-HRT ACT tags (the manufacturer of the tags used here). Because body undulations are a good proxy for how body acceleration should change, and TBF and ODBA both increased in relation to water velocity, we have confidence in our acceleration results given this validation.

Similar to TBF, metabolism also had a strong exponential relationship with water velocity, a relationship observed in many other fish studies (Allen et al., 2021; Brett, 1964; Claireaux et al., 2006; Clark and Seymour, 2006; Cucco et al., 2012; Rubio-Gracia et al., 2020). Moreover, standard metabolic rates of channel catfish in this study were within similar ranges to previous studies. For instance, Moss and Scott (1961) found channel catfish across 25, 30, and 35 °C had standard metabolic rates ranging from 77 to 193 mg O_2/kg/h, while Gerhart, B.J., & Allen, P.J. (unpublished results) reported standard metabolic rates across 23 and 33 °C ranged from 83 to 195 mg O_2/kg/h. Although our maximum metabolic rates were lower than those reported elsewhere, this could be due to the cooler maintained water temperature and larger fish being sampled, compared to previous studies.

We also observed a strong relationship between ODBA and metabolism, which is expected considering both values increased over changing water velocities and exhaustive exercise requires fish to use both aerobic and anaerobic metabolic processes (Kieffer, 2000; Lurman et al., 2007). Similar relationships are also shown in other species, with bonefish ODBA values related to metabolism (Brownscombe et al., 2018), and changes in speed and trajectory increasing metabolic demands of lemon sharks (*Negaprion brevirostris*) and hammerhead sharks (*Sphyrna lewini*) (Gleiss et al., 2009, Gleiss et al., 2010). Moreover, Lear et al. (2017) explained temperature and ODBA were the two most important factors in estimating metabolic rates in nurse sharks (*Ginglymostoma cirratum*), lemon sharks, and blacktip sharks (*Carcharhinus limbatus*). Although not problematic in flume tests, long-term studies should also consider the potential influence of sampling frequency, as the most appropriate sampling frequency can vary depending on study objectives. While we used 10 Hz, Brownscombe et al. (2018) showed 1 Hz was sufficient to establish acceleration, swimming speed, and metabolic relationships in bonefish, and this lower sampling frequency can increase the longevity of tag battery life. Tag retention should also be considered in longer term work as various species such as channel catfish and blue catfish (*Ictalurus furcatus*) have been documented to expel internal transmitters through the original incision or encapsulate the transmitter in either the stomach or intestine to later pass through the digestive tract (Bodine and Fleming, 2013; Marty and Summerfelt, 1986). While there may be challenges in implementing this work at a greater scale, we provide a basis for the acceleration-metabolic relationship in catfish that is beneficial for subsequent studies of fine-scale bioenergetics of free-swimming catfish.

Biologging tools have already proven to be beneficial, as they have served as operational welfare indicators in aquaculture settings (Svendsen et al., 2020), and acceleration measurements have been used as predictors of metabolism and energy expenditure in fishes (Clark et al., 2010; Gleiss et al., 2009, Gleiss et al., 2010; Halsey et al., 2008). Here, the relationships between ODBA and metabolism in controlled environments are developed as a necessary first step for further applications of accelerometry to study the metabolic demands of cultured channel catfish. Further refinement of the technique holds promise for use in production ponds where insight into metabolism is otherwise limited by turbid conditions preventing direct observation. As a general application for aquaculture, fine scale biologging information can be used to inform holistic bioenergetic models providing a better understanding of metabolic processes and energy expenditure, in a wide variety of environments. These advancements provide new tools for helping maximize aquaculture production.

Author contributions

A.J.V and P.J.A. conceptualized the project, A.J.V. curated and analyzed the data and led manuscript writing, and M.R.B. and G.M.S. contributed to data analysis. All authors contributed to drafts and gave final publication approval.

Declaration of Competing Interest

The authors declare that they have no known competing financial interests or personal relationships that could have appeared to influence the work reported in this paper.

The authors declare the following financial interests/personal relationships which may be considered as potential competing interests:

Peter J. Allen reports financial support was provided by USDA Agricultural Research Service. Peter J. Allen reports financial support was provided by USDA National Institute of Food and Agriculture.

Data availability

Data will be made available on request.

Acknowledgements

We thank the USDA ARS (grant #: 58-6066-5-042), the Mississippi Agricultural and Forestry Experiment Station, and the USDA National Institute of Food and Agriculture (Hatch project #: 1006942) for funding. We also thank Mack Fondren, Troy Lindsey, Rodney Reed, and Darrel Taylor for facility assistance, Brian Bosworth for providing the catfish, and Asgeir Bjarnason for Star-Oddi tag insight. Finally, we thank Brandon J. Gerhart, Manuel E. Coffill-Rivera, Joshua J. Neary, Jacob A.

Moreland, and Shannon A. Santiago, for technical assistance.

Appendix A. Supplementary data

Supplementary data to this article can be found online at https://doi.org/10.1016/j.aquaculture.2023.740280.

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