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Development, nutrition, and rearing practices of relevant catfish species (Siluriformes) at early stages

Enric Gisbert[1*], Ronald Kennedy Luz[2], Ignacio Fernández[3], Pravata K. Pradhan[4], Maria Salhi[5], Mansour T. Mozanzadeh[6], Aditya Kumar[4], Yannis Kotzamanis[7], Diana Castro-Ruiz[8], Martin Bessonart[5], Maria J. Darias[9,*]

[1] IRTA, Centre de Sant Carles de la Ràpita (IRTA-SCR), Aquaculture Program, Crta. Poble Nou, km 5.5 43540 Sant Carles de la Ràpita, Spain.
[2] Laboratório de Aquacultura da Escola de Veterinária da Universidade Federal de Minas Gerais, Av. Antônio Carlos, 6627 Belo Horizonte, MG, Brazil.
[3] Aquaculture Research Center, Agro Technological Institute of Castilla y León (ITACyL), Ctra. Arévalo, s/n, 40196 Zamarramala, Segovia, Spain.
[4] ICAR-National Bureau of Fish Genetic Resources, Canal Ring Road, Dilkusha, Lucknow - 226002, Uttar Pradesh, India.
[5] Laboratorio de Recursos Naturales, Instituto de Ecología y Ciencias Ambientales, Facultad de Ciencias, Universidad de la República, Iguá 4225, 11400 Montevideo, Uruguay.
[6] South Iran Aquaculture Research Centre, Iranian Fisheries Science Institute (IFSRI), Agricultural Research Education and Extension organization (AREEO), 6148140003 Ahwaz, Iran.
[7] Hellenic Centre for Marine Research (HCMR), Institute of Marine Biology, Biotechnology and Aquaculture, Fish Nutrition and Pathology Lab, Agios Kosmas, Hellinikon, 16777, Athens, Greece.
[8] Instituto de Investigaciones de la Amazonía Peruana (IIAP), Dirección de Investigación en Ecosistemas Acuáticos Amazónicos (AQUAREC), Iquitos, Peru.
[9] MARBEC, Univ Montpellier, CNRS, Ifremer, IRD, Montpellier, France.

[*]Corresponding authors: enric.gisbert@irta.cat (E. Gisbert); maria.darias@ird.fr (M.J. Darias).

Abstract

Catfish (Siluriformes) are important species for aquaculture worldwide, with an annual production in 2018 of *ca*. 6 million t. This review focuses on reassessing larval development, first feeding, and early rearing practices of the most important farmed catfish species, along with some candidates species for aquaculture diversification: *Pangasianodon hypophthalmus* (Pangasiidae), *Clarias gariepinus* (Clariidae), *Ictalurus punctatus* (Ictaluridae), *Pseudoplatystoma* spp. (Pimelodidae), *Heteropneustes fossilis* (Heteropneustidae), *Rhamdia quelen* (Heptapteridae), *Ompok bimaculatus* (Siluridae), and *Lophiosilurus alexandri* (Pseudopimelodidade). These species are initially reared indoors from one day to two weeks and are then transferred to fertilised outdoor ponds where they either feed on natural zooplankton or compound feeds. With the exception of *C. gariepinus*, *I. punctatus*, *R. quelen* and *P. hypophthalmus*, consistent and reliable fry production is a bottleneck that limits the expansion of farming of other species, such as *Pseudoplatystoma* spp., *H. fossilis*, ~~*R. quelen*~~, *O. bimaculatus*, and *L. alexandri*. Rearing systems (extensive, semi-extensive, intensive) and feeding protocols vary with species and geographical regions. Cannibalism and size heterogeneity are common, and these features create problems for larval and fry rearing of catfish species. Information about their nutritional requirements is required for the formulation of compound feeds that can guarantee high survival and good growth of catfish fries. However, such knowledge for most species is scarce, although some data are available for *I. punctatus.* Further genomic resources might allow fine-tuning rearing success. This review describes some successes in this field, and also highlights gaps in knowledge to guide future research that can promote the development of catfish aquaculture.

Key words: first feeding, live prey, feed formulation, feeding practices, hatchery, omics.

Introduction

Catfish (order Siluriformes) are a highly diverse clade of ray-finned fish species with a worldwide distribution. They dwell primarily in freshwater, but also in coastal regions of continents and nearby islands. Catfish are majorly distributed in the tropics of South America, Africa, and Asia [1]. Siluriformes, composed of over 3,000 living species and estimated 1,750 undescribed ones, is one of the largest orders of Teleostei, representing *ca.* 12% of all teleosts [2]. Catfish are named after the characteristic whisker-like barbels located around the mouth, which contain numerous taste buds for detecting food and navigating in turbid waters. Moreover, most catfish have a sub-cylindrical body with a flattened ventrum, dorsoventral flattened head, and sharp spines on their dorsal and pectoral fins [3,4]. Interestingly, the size range within this group (*ca.* 14 mm to 5 m) is probably the greatest in Osteichthyes[3]. Catfish have a scale-less skin covered with protective mucus; however, in families such as Callichthyidae and Loricariidae, the skin is covered with bony dermal plates[4].

Catfish have an exceptional importance for commercial, subsistent and recreational fisheries, ornamental fish trade, and aquacultural production. With regard to the latter, catfish possess a wide repertoire of characteristics that make them especially suitable for aquacultural purposes, such as high potential for domestication and adaptation to intensive rearing conditions, high fecundity, nocturnal foraging habits or capacity to live in turbid waters, relatively high resistance against infectious diseases, efficient feed conversion and no intramuscular bones, which greatly facilitates fillet processing [5-7]. Moreover, they are highly tolerant to low dissolved-oxygen levels, as some species are capable of air-breathing such as *Clarias* spp. and *Heterobranchus bidorsalis* (Clariidae), *Heteropneustes fossilis* (Heteropneustidae), *Pangasianodon hypophthalmus* and *Pangasius* spp. (Pangasiidae) [8].

The availability of high-quality fingerlings for the grow-out phase is one of the most critical factors affecting commercial prosperity in aquaculture. Successful larval production depends

on a wide range of biotic and abiotic factors as well as on the development of the zootechnical conditions for optimal rearing (e.g., larval density, feeding protocol, health management). Among the over 30 catfish species farmed worldwide, the present review is focused on the early culture of the most produced species in the different continents: the striped catfish, *Pangasianodon hypophthalmus* (Sauvage, 1878) (Pangasiidae, Asia); African sharptooth catfish, *Clarias gariepinus* (Burchell, 1822) (Clariidae, Africa and Europe); channel catfish, *Ictalurus punctatus* (Rafinesque, 1818) (Ictaluridae, North America); and species of the genus *Pseudoplatystoma* spp. Bleeker, 1862 (Pimelodidae, South America). In addition, four other species were added, as they are either relatively important at a local scale or are candidate species of interest for aquaculture diversification or conservation: the stinging catfish, *Heteropneustes fossilis* (Bloch, 1794) (Heteropneustidae) and butter catfish, *Ompok bimaculatus* (Bloch 1794) (Siluridae) in Asia; and the silver catfish, *Rhamdia quelen* (Quoy & Gaimard, 1824) (Heptapteridae) and the pacamã catfish, *Lophiosilurus alexandri* Steindachner 1876 (Pseudopimelodidae) in South America (production values for each species and contry of production are shown in the Supplementary file 1).

This review briefly introduces the importance of catfish in aquaculture and presents the selected catfish species. Further, it provides an overview of the rearing practices during early stages of these species, including information on the ontogeny of the digestive system, first feeding and early rearing, nutrition, cannibalism, and available molecular resources.

The importance of catfish in aquaculture

According to FAO's aquaculture statistics [9], a total of 5,781,235.1 t catfish were produced worldwide in 2018 with the exception of Oceania, for which no data were available (Table 1). Catfish were mainly produced in freshwater and represented 10.6% of the global freshwater fish aquaculture production (54,270,001.6 t), whereas a small production in brackish waters

(1,886 t; 0.03% of total world catfish production) was reported in African and South American countries [9]. The full list of countries and the species produced is shown in Supplementary File 1.

The major catfish producer in 2018 was Asia (5,333,195 t; 92.3% of total world catfish production; Table 1), with several cultured species of six different families (Pangasiidae, Clariidae, Bagridae, Siluridae, Ictaluridae, and Heteropneustidae). Three Asian countries accounted for 73.6% of the Asian catfish production, i.e., Indonesia (1,405,269 t; 26.4% of the Asian production), Vietnam (1,382,000 t; 26.0%), and China (1,127,252 t; 21.2%) (Supplementary File 1). Pangasiidae and Clariidae families accounted for 78.4% of the total Asian catfish production. Particularly, species from the Pangasiidae family (*P. hypophthalmus* and *Pangasius* spp.) were the most produced (2,826,068 t; 53.0%), followed by Clariidae (mainly *Clarias* spp., *C. gariepinus* × *C. macrocephalus*, and *C. batrachus*; 1,352,494 t; 25.4%). The other catfish families in terms of importance were Bagridae (mainly *Pelteobagrus fulvidraco*, *Leiocassis longirostris*, and *Hemibagrus nemurus*; 537,958 t; 10.1%), Siluridae (mainly *Silurus asotus*, *Wallago attu*, and *Silurus glanis*; 372,439 t; 7.0%), Ictaluridae (*I. punctatus*; 230,442 t; 4.3%), and Heteropneustidae (*Heteropneustes fossilis*; 373 t; 0.01%).

Africa was the second continent in terms of catfish production (251,333 t; 4.3% of total world catfish production; Table 1), with most species produced in freshwater [Clariidae: *C. gariepinus* (218,478 t; 87%), *Clarias* spp. (28,241 t; 11.2%), and *Heterobranchus longifilis* (8 t; 0.003%); Mochokidae: *Synodontis* spp. (5,510 t; 1.8%); Siluridae: *Silurus glanis* (44 t; 0.02%); and Bagridae: *Bagrus bajad* (2 t; 0.001%)] and a few species in brackish environments [Clariidae: *C. gariepinus* and Bagridae: *Chrysichthys nigrodigitatus* (50 t; 0.02%)]. Up to 35 African countries produced catfish in 2018; Nigeria was the main producer

(192,851 t; 76.7% of the African production), and Uganda, the second (33,454 t; 13.3%). The production in the remaining countries was limited (25,028 t; 10%) (Supplementary File 1).

In 2018, America (South, North, and Central) was the third most important region in catfish production (183,221 t; 3.2% of total world catfish production). According to FAO [9], the most produced species were ictalurids (*I. punctatus* and *Ictalurus* spp.; 161,271 t, 88.02%) and non-specified freshwater siluroids (13,950 t, 7.61%), the latter produced in Brazil. The national aquaculture production statistics of Brazil from 2019 were, however, more specific than those reported by FAO and showed that the Brazilian catfish production relied on *Pseudoplatystoma* spp. (Pimelodidae) and their interspecific and intergeneric hybrids (10,918 t) [10]. The remaining American catfish production, as indicated by FAO[9], was based on *C. gariepinus* (6,286 t; 3.43%), *P. hypophthalmus* (1,020 t; 0.56%), species from the Pimelodidae family (*Pseudoplatystoma* spp., and *Pimelodus* spp.; 660 t; 0.36%), *Hoplosternum littorale* (Callichthyidae; 22 t; 0.01%), *R. quelen* (Heptapteridae; 6 t; 0.003%), and *Pterygoplichthys pardalis* (Loricariidae; 6 t; 0.003%). The main catfish producing countries were the United States (159,423 t; 87.01%), Brazil (13,950 t; 7.61%), Cuba (6,286 t; 3.43%), and Mexico (1848 t; 1.01%), followed by a limited production in 11 other countries (0.6–580 t) (Supplementary File 1).

The aquaculture of catfish in Europe in 2018 (13,487 t; 0.2% of total world catfish production; Table 1) was focused on *C. gariepinus*, the hybrid *H. longifilis* × *C. gariepinus*, *I. punctatus*, *S. glanis*, and *Ameiurus melas* (Ictaluridae), which represented 49.6% (6,689 t), 24.7% (3,333 t), 14.5% (1953 t), 10.6% (1,425 t), and 0.6% (87 t) of the European production, respectively. This production was distributed among 19 countries, and the Netherlands (4,000 t; 29.7%), Hungary (3,585 t; 26.6%), and Russia (1,879 t; 13.9%) were the main producers (Supplementary File 1).

Selected catfish species

Pangasianodon hypophthalmus

Species of the Pangasiidae family are native to Southern Asia, and most are distributed in the Mekong and Chao Phraya river basins and in Indonesia [11]. The aquaculture of pangasiids has grown and expanded dramatically in the past 25 years. Among the existent 28 species, *P. hypophthalmus* —previously known as *Pangasius sutchi* or *Pangasius hypophthalmus*— is today the most produced catfish worldwide, accounting for over 40% of the world's catfish production. Vietnam is, by far, the biggest producer of pangasiids in Asia (48.1%), followed by India (18.5%) and Bangladesh (15.6%) [9].

The striped catfish *P. hypophthalmus*, reaching 1.3 m in total length (TL) and a maximum weight of 44 kg [12], is listed as endangered by the IUCN. It has been introduced in the South-East Asia, Indian subcontinent, Brazil and in the Caribbean (Dominican Republic, Jamaica, Haiti, and Puerto Rico) for aquaculture purposes [12]. Its farming success is mainly based on its high fecundity, omnivorous feeding habits and tolerance to low dissolved-oxygen levels in water, which is due to the presence of a well-vascularised swim bladder that enables this species to breathe atmospheric oxygen; consequently, pangasiids can be reared in ponds at high densities and with low water renewal [7]. Interspecific and intergeneric hybrids have also been produced between *P. hypophthalmus* and other cultured pangasiid species, such as *Pangasianodon gigas*, *Pangasius bocourti*, *P. larnaudii*, or *P. djambal*, for their high growth, survival rate, or flesh quality [7,13].

Clarias gariepinus

The African sharptooth catfish essentially has a pan-African distribution, although it is naturally absent from Maghreb, Upper and Lower Guinea, and Cape provinces. This species is also naturally present in Jordan, Lebanon, Syria, Israel, and Turkey, and it has been

introduced in over 25 countries in Europe, Asia, and Latin America [9]. In natural environments, *C. gariepinus* is found in lakes, streams, rivers, swamps, and floodplains, many of which are subjected to seasonal drought. In such habitats, it can survive during the dry season because of the air-breathing dendretic organ located in the suprabranchial chamber. *Clarias gariepinus* reaches an average adult size of 1–1.5 m (maximum size = 1.7 m TL) and 60 kg in weight. Because of its high growth rate at high stocking densities, high feed conversion rates, good flesh quality, and year-round production [14], this is the most cultured catfish species not only in Africa but also in Europe [9]. In Asia, *C. gariepinus* is also cultured in many countries, where it is also hybridised with native *Clarias* species, such as *C. batrachus* and *C. macrocephalus*. Interestingly, these hybrids present higher growth rates than the local *Clarias* species and better flesh quality and taste than the Asian species [15].

Ictalurus punctatus

The channel catfish is a North American freshwater carnivorous species native to the central drainages of the United States and into southern Canada [5]. Because of its large size (maximum size = 1.3 m TL and 26.3 kg in weight) [12] and excellent taste, it has been extensively introduced for recreational fisheries and aquaculture throughout the United States and northern regions of Mexico, as well as in over 40 countries worldwide [9]. China is the biggest producer, followed by the United States, Russia, Mexico, and Italy. Propagation of *I. punctatus* began in the United States in 1914 for stocking lakes, reservoirs, and farm ponds [16]; while currently, *I. punctatus* accounts for 60% of the US freshwater aquaculture production [9]. However, the channel catfish has been replaced by the interspecific hybrid catfish *I. punctatus* ♀× *I. furcatus* ♂, which represents 70% of the current US catfish production. The hybrid has a superior

performance because of its high growth rates, bacterial disease resistance, tolerance to hypoxic conditions, and carcass yield [17].

Pseudoplatystoma spp.

The species of the genus *Pseudoplatystoma* are distributed in the major river basins of South America and are highly prized for human consumption because of their flesh quality and absence of intra-muscular bones (pin-bones) [18,19]. *Pseudoplatystoma* spp. are piscivorous and reach maximum sizes of 1.40 TL and 25 kg in weight [18,20]. The genus currently consists of eight species (*P. punctifer*, *P. reticulatum*, *P. orinocoense*, *P. fasciatum*, *P. magdaleniatum*, *P. tigrinum*, *P. metaense*, and *P. corruscans*) (Supplementary File 2). However, there are inconsistencies in the taxonomy of the genus proposed by Buitrago-Suárez & Burr [18] as subsequent molecular and morphological studies revealed [21,22], which highlight the need to re-evaluate the classification within the genus. The high commercial value of *Pseudoplatystoma* spp. in comparison to other local native fish has motivated the development of its commercial rearing, and Brazil is the biggest producer in the region [19]. However, its production has mostly relied on interspecific hybrids, which have better growth performance than that of pure species [23,24]. Currently, the most produced hybrids are intergeneric between *Pseudoplatystoma* spp. and the omnivorous pimelodid catfish *Leiarius marmoratus* or *Phractocephalus hemioliopterus*, which show less cannibalistic behaviour, readily accept compound diets, as well as more omnivorous feeding habits at juvenile and adult stages [25-27]. The production of hybrids has been identified as a serious threat to the industry despite their widespread use. Studies based on molecular markers have shown that fish farmers are in some cases mistakenly using interspecific hybrids as broodsfish, which, in the case of post-F1 hybrids, reduces the viability of the offspring due to their high mortality rates [25]. Considering also the threat that hybrids escaped from fish farms represent to natural populations due to their

potential introgressive hybridization that may have negative impacts on biodiversity [28], research efforts should be focused in developing breeding programs and technologies for pure *Pseudoplatystoma* species in order to promote profitable and environmentally safe alternatives to the production of hybrids [27]. A list of *Pseudoplatystoma* hybrids and their characteristics is shown in Supplementary File 2.

Heteropneustes fossilis

The stinging catfish is a commercially important and popular species, particularly in countries such as India, Thailand, Bangladesh, Pakistan, Nepal, Sri Lanka, Myanmar, Indonesia, and Cambodia [29-30]. This omnivorous species dwells in ponds, ditches, swamps, and marshes, but sometimes it is also found in muddy rivers. *Heteropneustes fossilis* can survive in oxygen-depleted waters by utilising atmospheric oxygen for respiration due to the presence of a respiratory air sac. This species grows up to 0.3 m TL and 0.2 kg and is mostly preferred for its tender flesh, taste, and low-fat content. In addition, its flesh is recommended to people with anaemia because of its high iron content [31].

Heteropneustes fossilis is appreciated for aquaculture for its tolerance to crowding stress, air-breathing capacity, and acceptance of pelleted feeds [32]; it is also considered an interesting ornamental fish [12]. Currently, *H. fossilis* is commercially reared exclusively in Bangladesh (13,421 t) and Myanmar (373 t) [9]. However, it is considered a highly promising candidate for the diversification of freshwater aquaculture in India [32,33]. Successful intergeneric hybridization has been achieved between *H. fossilis* ♀ and *Clarias batrachus* ♂; however, hybrids performed worse in terms of growth when compared to *C. batrachus*, but better with regard to *H. fossilis* conspecifics [34].

Rhamdia quelen

The silver catfish *R. quelen*, also known as South American catfish, black catfish or jundiá, is an omnivorous freshwater species that grows fast during druing the first years of life and successfully reproduces in captivity. These characteristics, associated with a high acceptance by the consumer markets of Brazil, Argentina, and Uruguay, encouraged its aquaculture production. The genus Rhamdia includes several species, most of them with great similarities in body shape, color patterns and habitat use, being 49 of them synonymized as *R. quelen*, with a wide geographical distribution from central regions of Argentina to southern Mexico [12]. However, according to several studies [35-37], the taxonomy of this genus needs to be re-evaluated by means of molecular tools in order to clarify current synonymies. Particularly, *R. branneri* and *R. voulezi*, which were initially were considered as synonyms of *R. quelen* [38], have been recently confirmed as valid species [35,37]. Since most information compiled in this review is based on data from Argentina, Brazil and Uruguay obtained before the recognition of various species previously considered as *R. quelen*, the possibility that species such as *R. branneri* and *R. voulezi* may have been included in this review under *R. quelen* denomination must be taken into consideration. Brazil is the main country producing this species [19]. Since 1980, several studies have been conducted to develop production technologies for this species [39]. In nature, *R. quelen* males grow faster than females up to the third or fourth year of life, when this condition is reversed. The maximum size for *R. quelen* in nature is approximately 0.7 and 0.5 m TL for females and males, respectively, reaching a maximum weight of 4 kg; this size is attained at the ages of 18 and 12 years, respectively [39]. This species can live in a wide range of temperatures, although a better growth performance is displayed at temperatures around 24°C [40]. As it tolerates much lower temperatures than other fish within its distribution area, *R. quelen* is a particularly promising aquaculture species in subtropical regions, where

temperatures drop during the winter [41]. No hybrids with other catfish species have been reported.

Ombok bimaculatus

The butter catfish is distributed in the Indian subcontinent and Myanmar. This freshwater species is found in quiet, shallow, often muddy waters, in sandy streams, rivers, canals, beels, and inundated fields. *Ombok bimaculatus* is an omnivorous species, mainly feeding on vegetable matter, fish, and occasionally on crustacean and planktonic organisms, reaching a maximum size of 0.5 m TL and 0.2 kg in weight. The butter catfish is considered a delicacy in many parts of India, particularly in the North-eastern states, because of its good taste, excellent nutritional profile, and soft bony structure [42]; it is one of the most expensive fish species in this country. Recently, it has been also introduced in ornamental fish markets of India owing to its moderate market demand among hobbyists. Because of its increasing demand, this species is categorised as near threatened by the IUCN. Considering the consumer acceptance, *O. bimaculatus* has been considered an important candidate for the diversification of freshwater aquaculture in India and neighbouring countries [43]. No hybrids with other catfish species have been reported for *O. bimaculatus*.

Lophiosilurus alexandri

The pacamã, *L. alexandri* is a freshwater carnivorous fish endemic to the São Francisco River in Brazil [44], reaching a maximum size of 0.5 m TL and 5 kg of weight. This species is highly appreciated for human consumption because of the quality of its flesh. It is considered as an emerging aquaculture species within its range of natural distribution due to its high demand for consumption and use as ornamental fish [45]. *Lophiosilurus alexandri* is a sedentary species, prefers lentic environments, and reproduces by batch spawning with the release of eggs on

sandy substrate with male parental care [46]. This species is considered vulnerable to extinction [47], although it has not yet been classified by the IUCN. In this context, efforts have been made to improve the production of fingerlings for restocking programmes in the Rio São Francisco basin [48-51], as well as for human consumption [52,53]. No hybrids with other catfish species have been reported.

Ontogeny of the gastrointestinal tract and digestive capacity

To survive and grow, fish must be able to capture, ingest, and digest food and absorb nutrients. Although fish larvae may be morphologically capable of capturing different food items (e.g. zooplanktonic organisms and microdiets), their digestive system undergoes a series of developmental changes before being fully functional shortly after hatching [54]. In this regard, knowledge about the ontogeny of the digestive system may contribute to the development of efficient larval feeding protocols. For example, the morphology and functionality (e.g., activity of digestive enzymes) of the digestive system are often used to assess the nutritional condition of fish larvae reared under different conditions [55]. Although there are similarities in the ontogenic development among fish species, there are also interspecific differences with regard to the timing of differentiation, development, and functionality of the digestive system in relation to the physiological ecology of the species. The chronology of developmental events expressed solely in terms of time does not provide a reliable basis when comparing fish that have been reared at different water temperatures. Therefore, in this review larval development has been described in relation to larval size in length or accumulated degree-days (ADD).

Morphoanatomical development of the digestive system

A summary of the main morphoanatomical changes of the digestive system in the catfish species considered within this review is presented in Table 2. In particular, *I. punctatus* and *R. quelen* are precocial species, whereas *C. gariepinus, P. hypophthalmus, P. punctifer, H. fossilis, O. bimaculatus,* and *L. alexandri* exhibit altricial development. *Rhamdia quelen* is the species that develops faster, showing an open mouth at only 4 ADD (4 hours post hatching, hph at 24.6°C; *ca*. 5 mm TL) and a differentiated stomach at 17 ADD (16 hph at 24.6 °C; *ca*. 6 mm TL) that is completely formed and functional at 49 ADD (2 days post hatching, dph at 24.6°C; *ca*. 8 mm TL). The next fastest developing species are *C. gariepinus* and *H. hypophthalmus,* although key digestive structures appear much later in these species than in *R. quelen.* Mouth opening in *C. gariepinus* occurs at 50 ADD (2 dph at 25°C; *ca*. 9 mm TL) and the stomach is formed at 114 ADD (4 dph at 28.5°C; *ca*. 11 mm TL). *Pseudoplatystoma punctifer, H. fossilis, and O. bimaculatus* present similar but delayed developmental patterns. Although mouth opening occurs earlier in *H. fossilis* (29 ADD, 1 dph at 29°C; *ca*. 3 mm SL) than in *P. punctifer* (56 ADD, 2 dph at 28°C; *ca*. 5 mm TL) and *O. bimaculatus* (54 ADD, 2 dph at 27°C; *ca*. 3 mm TL), the timing of most anatomical and histological events is similar between the three species (Table 2). For instance, the stomach is formed at 252 ADD (9 dph at 28°C; *ca*. 10 mm TL) in *P. punctifer,* at 290 ADD (10 dph at 29°C; *ca*. 7 mm TL) in *H. fossilis*, and at 297 ADD (11 dph at 27°C; *ca*. 14 mm TL) in *O. bimaculatus. Lophiosilurus alexandri* shows a different developmental pattern, characterised by an already opened mouth at hatching (0 dph at 27°C; *ca*. 3 mm TL) and a delayed first exogenous feeding as well as pancreas and intestine differentiation compared with the other species (Table 2). However, the complete histological development of the digestive system, marked by the formation of the stomach, is achieved approximately at the same time as that in other catfish species. A mixed feeding period exists in all these species, which lasts between 1 and 4 days. Yolk-sac resorption is particularly long in *L. alexandri*, which occurs almost in synchrony with the

formation of the stomach (Table 2). Despite being a species extensively studied and reared, we could not find any detailed description of the digestive system ontogeny of *I. punctatus*. However, we present here some information on the development of this species as a guideline. The incubation time of channel catfish eggs averages 5 days at 27–28°C, and larvae have an average size of 10.6 mm TL at hatching. The period from hatching to first feeding lasts from 5 to 9 days, depending on water temperature; the onset of exogenous feeding occurs at 13–14 mm TL [66]. *Ictalurus punctatus* has a long yolk-sac resorption period of 5 to 10 days [67]. However, the juvenile period is considered to start from the onset of exogenous feeding [68].

Functional development of the digestive system

The ontogenic development of the digestive enzymes of altricial species may be divided in three different phases: 1) from hatching to the onset of exogenous feeding; 2) exogenous feeding phase, based on alkaline proteolytic enzymes produced by the exocrine pancreas; and 3) commencement of acidic protein digestion to supplement alkaline proteases caused by the development of a functional stomach, and transition from larval to juvenile/adult digestion mode [54].

During the endogenous feeding phase, catfish possess pancreatic digestive enzymes such as alkaline proteases, lipases/esterases, and carbohydrases. These enzymes are involved in the digestion and reabsorption of the yolk sac by the syncytium that surrounds it, as well as the accumulation of zymogens in the exocrine pancreas [54]. Nevertheless, it should be highlighted that the biochemical detection of certain enzymes in newly hatched larvae may also be attributed to other factors rather than the development of accessory digestive organs. For instance, high activity levels of trypsin-like proteases just after hatching are generally associated with the lysis of the chorion during the hatching process [69]. In addition, detecting bile salt-activated lipases at hatching, when the exocrine pancreas is not yet fully

differentiated, does not mean that catfish larvae utilise such lipases to digest lipids contained in their yolk-sac reserves. In fact, it indicates that the spectrophotometric method for assessing this enzyme, in which lipase activity is enhanced by means of bile salts (sodium cholate), is not specific [70] and it may also detect other lipases hydrolysing triglycerides and wax esters in the yolk [71].

Among the species selected in this review, the functional development of the digestive system has only been reported for *C. gariepinus* [57,72], *P. hypophthalmus* [73], *O. bimaculatus* [61], *R. quelen* [63], and *P. punctifer* [74,75]. Unlike the other catfish species, *I. punctatus* and *R. quelen* larvae present functional stomachs before changing from endogenous to exogenous feeding [63]. Regarding the other species, after the onset of exogenous feeding and before the development of a functional stomach, proteins are digested by alkaline proteases, principally trypsin and chymotrypsin, in combination with intestinal cytosolic peptidases (*i.e.*, leucine-alanine peptidase). During this period, larvae display limited capacity of digesting macromolecules that are absorbed by enterocytes [76]. Comparatively, in *R. quelen*, a sharp increase in the specific activity of digestive alkaline proteases was detected at the onset of exogenous feeding (49 ADD, 2 dph at 24.6°C; *ca*. 8 mm TL) [63]; this increase was observed several days after first feeding in the other catfish species [61,73,74]. The combination of histological and biochemical tools revealed that an increase in the production of pancreatic alkaline proteases was observed after the completion of the exocrine pancreas development [61,63,74]. Similar patterns regarding lipase and α-amylase have been also described [61,74], although profiles in activity along larval ontogeny varied according to the species. These results may be attributed to different developmental patterns, rearing protocols, and analytical methods for quantifying enzymatic activity. In this context, pepsin-like activity was detected in newly hatched larvae of *P. punctifer* [74]. However, the presence of pepsin-like activity in hatchling homogenates cannot be attributed to the presence of a functional stomach, as this organ is not

developed yet; thus, pepsin-like activity is due to the presence of lysosomal proteases involved in the intracellular digestion of yolk proteins. This finding was further confirmed in a recent study on the ontogeny of the main digestive enzyme precursors during the larval development of *P. punctifer*, in which pepsinogen expression was detected as early as 56 ADD (2 dph at 28°C, 5 mm TL) [75]. This is due to the fact that acidic (aspartic) proteases are homologous entirely in terms of amino acid sequences, particularly around the active site residues. The sharp increase in pepsinogen expression detected at 252 ADD (9 dph at 28°C; *ca.* 10 mm TL) is certainly attributed to the pepsin-coding gene expression, as at this age the stomach is formed and full of gastric glands [59].

In gastric species, the acquisition of a functional stomach is widely considered the end of the larval stage [54]. The onset of acidic digestion is also generally considered an optimal point for larval weaning onto microdiets, when the adult-like mode of digestion becomes fully functional and dietary complex proteins are easily digested. However, this is not a universal rule, as some species can be weaned onto dry feed before acidic digestion begins [77]. In the reviewed catfish species, pepsin activity was detected at *ca.* 49 ADD (2 dph at 24.6°C; *ca*. 8 mm TL) in *R. quelen* [63], 114 ADD (4 dph at 28.5°C; *ca*. 11 mm TL) in *C. gariepinus* [72], 252 ADD (9 dph at 28°C; *ca*. 10 mm TL) in *P. punctifer* [74,75], and 413 ADD (15 dph at 27°C; *ca.* 25 mm TL) in *O. bimaculatus* [61]. Thus, the histological and functional formation of the stomach is synchronised in these species with the exception of *O. bimaculatus*, in which a gap of several days existed between the stomach differentiation and pepsin secretion [61]. These differences may be attributed to different reproductive and developmental guilds as well as differences in growth and developmental rates in response to different environmental pressures (*e.g.*, food availability, habitat seasonal modifications). These results highlight the need to conduct both histological and biochemical studies for each species to accurately assess

422 the shift between the larval- and adult-like modes of digestion. When only histological data
423 are available, conclusions should be made with care.

424 Besides serving to characterise the digestive capacities of developing fish, the activities of
425 pepsin and other digestive enzymes may also serve as biomarkers for evaluating hatchery
426 practices [77]. In particular, pepsin activity may act as an indicator of the population's
427 heterogeneity during the process of adaptation to new diets during weaning. Particularly, a
428 high coefficient of variation in pepsin activity at a single age or stage of development or
429 activity fluctuations along several days after a shift in diet may be used for the above-
430 mentioned purposes. Similarly, digestive enzyme activities may provide insights into the
431 larval ability to modulate their digestive enzyme production, depending on the nutritional
432 composition of the diet. This has been demonstrated in *P. punctifer* at both larval and early
433 juvenile stages. For instance, gene expression of amylase, phospholipase, and lipoprotein
434 lipase were differentially regulated in *P. punctifer* in response to the dietary DHA content in
435 *Artemia* during the larval phase (Diana Castro-Ruiz, unpublished data). Similarly, gene
436 expression of the main digestive enzymes, as well as their enzymatic activity, can also be
437 modulated in response to dietary composition. Diets containing 45% protein induced an
438 increase in *trypsin* and *pepsinogen* expression and a decrease in *amylase* in *P. punctifer*
439 compared with that in individuals fed diets containing 30% protein. Changes in gene
440 expression were associated with changes in their corresponding enzyme activity; the
441 regulation could be at transcriptional or translational levels, depending on the digestive
442 enzyme analysed [78].

443 Changes in enzyme activities over circadian rhythms as well as their postprandial
444 modifications after a single meal are also important for understanding larval digestive
445 capacities and adjusting feeding practices (*i.e.*, number of meals per day). In this context, it
446 has been demonstrated that postprandial changes in proteolytic enzymes were observed in *C.*

gariepinus larvae aged 3 and 7 dph [5.4 and 29.7–33.1 mg body weight (BW), respectively] within 30 min after feeding [79]. In particular, proteolytic activity in the gut decreased significantly because of the immediate utilisation of enzymes present in the gut, whereas *ca.* 1 h later, when larvae had completely filled their gut, protease activity started to increase and a maximum of enzyme activity was recorded 12 h after the intake of one single meal. Thus, decapsulated *Artemia* cysts were completely digested *ca*. 9 h after ingestion, whereas other types of food with higher protein digestibility, e.g., *Artemia* nauplii, were digested faster because the peak of proteolytic activity occurred earlier. In addition, no change in enzymatic activity was verified in starved larvae when evaluating the activity of proteolytic enzymes in *C. gariepinus* along a 24-h cycle. However, total protease activity in larvae fed every 4 h showed small significant differences during the same 24-h period. Seemingly, enzyme production did not occur in a rhythmic cycle and was not affected by the light regime either [79]. The last but not the least, further research must be focused on the appetite-regulating hormones and their role in the physiological regulation of appetite and prey ingestion considering species-specific feeding habits, feeding protocols and diet composition [54], as well as their potential relationship with the cannibalistic behaviour in this group of species.

Rearing practices for early life stages

The development of a reliable protocol for rearing fish larvae and fries is a necessary step to guarantee its culture at a commercial scale. The establishment of reliable rearing protocols is difficult, as larval and fry culture is a complex process that relies on multiple factors, such as larval and fry development, behaviour, growth, and survival. The above-mentioned processes are modulated by many factors that may be classified into four categories: species-specific reproductive guilds, environmental factors (i.e., temperature, light intensity, photoperiod, water quality, and tank cleaning), feeding factors (i.e., food composition, feeding frequency

and ratio, meal distribution timing, and weaning period), and population factors (i.e., fish density, strain, and domestication level) [80]. A wide range of larval rearing practices have been developed in the last decades for different catfish species, protocols that vary mainly depending on the geographical area, level of initial economic investment, main production purpose (i.e., subsistence, commercial, or restocking), among other factors. Thus, this section is devoted to review this species-specific state of the art regarding different rearing systems and feeding practices.

Pangasianodon hypophthalmus

The striped catfish has been farmed for decades in the Mekong Delta relying on wild-caught seed. However, the explosive growth of its commercial production started after the optimisation of induced breeding in the late 1990s, with larval production increasing 18-fold between 2002 and 2011 [81,82]. *Pangasianodon hypophthalmus* larvae are obtained by hormonally-induced spawning. Eggs and milt of hormonally-treated broodfish are collected, and eggs are fertilized with milt by gently mixing. For removing the adhesiveness of the fertilized eggs, they are then washed with 1% tannic acid solution for 5–10s [83,84]. Fertilized eggs are distributed on steel trays or hatching jars (Zoug, Weiss or McDonald jars) for incubation with a continuous freshwater flow. Hatching occurs between 23 and 34 hours post fertilization (hpf) in incubation temperatures ranging from 26°C to 30°C. The onset of exogenous feeding of larvae occurs at 2 dph (6.2 mm TL) [83,85].

Regarding rearing procedures, in the commercial hatcheries from the Mekong Delta (Vietnam), larvae are generally reared in indoor tanks with volumes ranging from 0.2 to 4.7 m^3 in flow-through water systems with constant aeration. Stocking densities vary between 200 and 7,000 larvae m^{-3}. Most hatcheries sell larvae to nursery farms before the onset of exogenous feeding [83,85]. In the nursery farms, *P. hypophthalmus* larvae are cultured in earthen

ponds (1,000–5,000 m^2 and 1.5–2-m depth) using high-quality screened, chlorine-treated water (pH: 6.4–8.5; dissolved oxygen ≥3 ppm) (Table 3). *Pangasianodon hypophthalmus* larvae present cannibalistic behaviour from the onset of exogenous feeding until 8 dph [85,86]. The impact of this cannibalistic behaviour can be reduced with low stocking densities and the presence of natural zooplankton in the ponds. Before transferring the larvae, ponds are cleaned from sludge, treated with lime (10–15 kg 100 m^{-2}) and often also with salt, and dried for 3 to 5 days. Ponds are then fertilised with fish powder or fish meal (2–3 kg 1,000 m^{-2}), soybean meal (2–3 kg 1,000 m^{-2}) or blood powder (1 kg 1000 m^{-2}) and zeolite (4 kg 1,000 m^{-2}), and probiotics (0.3 kg 1,000 m^{-3}) and 1–2 kg of live prey (i.e., *Moina* sp.) to promote their growth and proliferation and serve as food for larvae. Once ponds are prepared, 1 dph-old *P. hypophthalmus* larvae are stocked at densities of 500 to 800 larvae m^{-2} and reared for 20 to 45 days depending on the farm, until fry are transferred to fingerling nursing ponds [82,83]. The above-mentioned rearing practices generally result in survival rates ranging from 30 to 50%.

Larval growth, size heterogeneity, mortality, and cannibalistic behaviour in *P. hypophthalmus* are profoundly affected by water temperature [86]. In experimental conditions in an indoor recirculating system, mortality rates showed an inverse correlation with water temperature during the first 4 days after hatching, whereas cannibalistic rates were higher in cold than in warm water temperatures (23°C vs. 33°C). In addition, size heterogeneity decreased with an increase in water temperature, evidencing that choosing an optimum thermal temperature for larval rearing in *P. hypophthalmus* promoted growth, reduced cannibalism and early mortality, and decreased size heterogeneity. Thus, the optimal temperature for somatic growth in *P. hypophthalmus* larvae is 31°C at the onset of exogenous feeding, increases to 32.7°C when larvae weigh 8 mg BW, and then decreases progressively in larger fish, at a rate of *ca.* 0.7°C for each 10-fold increase of BW.

Regarding feeding practices, from the onset of exogenous feeding (2 dph, 6.2 mm TL), *P. hypophthalmus* larvae feed on wild zooplankton and stocked zooplankton and zoobenthos such as *Moina* sp., *Artemia* sp., or *Tubifex* sp. Additionally, larvae may be fed five times a day during the first week of rearing in the ponds; the farm-made feed is basically composed of soybean meal or fishmeal, soybean milk, egg or yeasts. During the second week, larvae are fed a concentrated powder (40% protein) 4 times a day, and from the third week, early juveniles are fed commercial pellets (30–35% protein) 3 to 4 times a day. For further details on the feeding protocol used in the nurseries of the Mekong Delta, readers are invited to consult Nguyen *et al.* [82]. One-day-old larvae have been stocked at low densities (60 larvae m^{-2}) in rotifer-enriched nursery ponds and fed custard egg and soya powder during the first days and a carp fry diet subsequently [84]. These authors reported a larval survival rate of 18.3% and a growth rate of 0.2 g day^{-1} after 45 days of culture.

In experimental conditions, the striped catfish larvae have been reared in indoor recirculating systems and fed 36 h post hatching (hph) *Artemia* nauplii eight times a day until 8 dph. Under these conditions, survival rates ranged from 20% to 60%, depending on prey and fish densities [87]. Authors observed that at 8 dph, survival depended on feeding level rates rather than on prey density. The survival rate of *P. hypophthalmus* that was fed high levels of *Artemia* nauplii was higher in lower densities (10 and 30 larvae L^{-1}) than in higher densities (90 larvae L^{-1}). Similarly, a higher feeding level promoted larval growth at 8 dph, which was not influenced by larval density. A model of maximal food intake showed that during the early feeding stages, the maximal meal size of *P. hypophthalmus* larvae was small (12% BW at 5.5 mm TL and 0.72 mg BW), but it increased quickly at 6 mm TL (22% BW, 1.2 mg BW) and at 6.5 mm TL (26% BW, 1.6 mg BW). From 7 mm TL onwards, meal size decreased curvilinearly to 10% BW at 15 mm TL (25 mg BW) [87]. The best first-feeding time for *P. hypophthalmus* is recommended between 30 and 36 hph using rotifers (*Brachionus. angularis*)

during 3 days followed by cladocerans (*Moina macrocopa*) for the subsequent 7 days [88]. These authors also reported that the best live prey density and feeding frequency in terms of growth and survival were between 8 to 11 individuals mL^{-1} and six times per day, respectively.

When comparing feeding behaviour between light and dark rearing conditions, ingestion rates of *A. nauplii* in 4- and 7-dph-old larvae reared in darkness were higher than those of larvae under light conditions [89], which may be due to the higher swimming activity at night than in the day. These results indicate that the feeding behaviour in this species depends on chemo-sense rather than visual sense because of the presence of free neuromasts that respond to mechanical stimuli and the numerous taste buds on the barbels, head surface, buccal cavity, and gills [90]. Concerning weaning under experimental conditions, *Artemia* nauplii could be fed to larvae until they attained 100 mg BW (at 11 dph with the optimal rearing temperature), then larvae may be weaned onto a commercial feed (Nippai SeaBream, Nippai, Yokohama, Japan; 55% protein, no data on lipid content provided by authors) within 6 days, and fed another commercial feed (BioMar BioOptimal Start, Nersac, France; 52% protein) after attaining 300 mg BW [86].

Clarias gariepinus

Clarias gariepinus larvae and fingerlings may be produced using three different systems [91]: 1) in nursery ponds, where larvae are extensively on-grown to fingerling size before being stocked into larger grow-out ponds; 2) in a hatchery for a period of up to 14 days and then grown to the fingerling size for a further 30 days in nursery ponds, after which they are stocked into larger on-growing ponds; and 3) larvae are intensively reared to the fingerling size in a hatchery, after which they are on-grown under pond or high-density tank culture conditions (Table 3). Generally, when extensive pond systems are used for larval rearing, the most critical factor for success is the availability of zooplankton during the first days. This naturally

571 growing zooplankton is mainly formed by cladocerans (*Moina* sp., *Chidorus* sp.,
572 *Diaphanosoma* sp., *Bosmina* sp., and *Daphnia* sp.), copepods (different Cyclopoidea species)
573 and rotifers (*Keratella* sp., *Brachionus* sp., *Synchaeta* sp., among others) [6].

574 Under extensive rearing conditions, ponds are prepared to assure abundance of
575 zooplanktonic prey for larvae. This generally occurs up to 14 days before stocking 3-dph
576 larvae and consists of liming and fertilisation. Several manuals recommend adding 100–150
577 kg ha^{-1} of quicklime to the damp pond bottom to eliminate pathogens and potential
578 invertebrate predators. Then, ponds are left for 7–14 days and filled with water to a depth of
579 30 cm, and the pH is adjusted by adding lime. Afterwards, farmers promote the proliferation
580 of zooplanktonic blooms by adding inorganic or organic fertilisers, which are selected
581 depending on the economic resources of the farmer. Only then, larvae are introduced into the
582 rearing ponds. Readers are encouraged to consult Hecht [91] for further details about different
583 strategies for chemical pond fertilisation. Regardless of the procedure employed, it is
584 recommended to maintain soluble nitrogen and orthophosphate at 0.95 mg N L^{-1} and 0.1–0.5
585 mg P L^{-1}, respectively [92]. The most commonly used organic pond fertilisers, are poultry, pig,
586 and bovine manure [93]. The following rates of manure application (kg 100 m^{-2}) may be applied:
587 an initial quantity of 25 kg of poultry manure followed by 3 to 5 kg every 10 days; 7 kg of pig
588 manure every two days; or 10 kg of bovine manure every two days. However, the success of
589 these procedures may change depending on local environmental conditions; if an adequate
590 phytoplankton bloom is not achieved within six to eight sunny days, more manure should be
591 added into the ponds. As ponds can only assimilate a certain amount of manure per day, it
592 should be added frequently on a daily basis [94]. Finally, a combination of organic and inorganic
593 fertilisers can also be applied to promote zooplankton growth in ponds. In particular, a mixture
594 of dry poultry manure (10–20 kg), urea (0.4–0.8 kg), and triple superphosphate (0.1–0.2 kg)
595 per 100 m^2 per week is advisable. In addition, periphyton can also be successfully used for the

596 rearing of *Clarias* larvae. In this context, it has been reported the beneficial combined effect
597 of pond fertilisation (20 kg pig manure per 100 m^2 at initial fertilisation rate followed by 10
598 kg every two weeks) and the use of bamboo poles (4 per m^2) for the development of periphyton
599 [95].

600 Regarding feeding practices, before the onset of exogenous feeding (*ca*. 80 hph, depending
601 on temperature), larvae aged 3 dph are moved from the hatchery facilities and stocked into
602 rearing ponds (100–250 m^2) at a density of 100–250 larvae m^{-2}. At lower larval rearing
603 densities (100 larvae m^{-2}), the feeding strategy consists of adding 1 kg rice or wheat bran and
604 1 kg 100 m^{-2} of crumbled formulated feeds into the ponds during the first three weeks. For the
605 following two weeks, bran quantities should be maintained stable, but formulated feed may
606 increase up to 2 kg 100 m^{-2} day^{-1} (divided in two meals per day). Size grading is advisable
607 after three weeks to homogenise size classes and reduce fry cannibalism. Survival rates of
608 40% and 3-g fries (BW) could be obtained along a rearing cycle of 50 days when proper
609 feeding and management practices are employed [96].

610 Rearing *C. gariepinus* under intensive hatchery conditions generally lasts from 12 to 14
611 days at 28°C, which is considered as the optimal growth temperature for this catfish species.
612 After the onset of exogenous feeding (3 dph at 28°C), different types of live prey (*Artemia*
613 nauplii and metanauplii, *Daphnia* sp., *Moina* sp., or other zooplanktonic species of suitable
614 size) can be used for first-feeding larvae during the first week in hatcheries [91]. The earliest
615 weaning time to maximise growth rate of *C. gariepinus* larvae was after 4 days of feeding
616 with *Artemia*, when larvae weighed *ca.* 18 mg BW at 27.5°C, although weaning may be
617 achieved at 7.1 mg BW without any effect on the survival rate [97]. Among different weaning
618 strategies, the most commonly used protocols are summarised in Table 4. After 12–14 days,
619 early juveniles are stocked into nursery ponds at densities ranging from 65 to 2,000 specimens
620 m^{-2} (100,104). Under pond-farming conditions, it is recommended to feed the fry at 25% BW

per day (divided in three meals), using a 38–40% protein diet [100]. If the larvae and early juveniles are reared in tanks, the feed should have a protein content of around 50%. The nursery period ends when fries reach 1–2 g BW and are ready to be stocked into ponds or tanks for the on-growing phase.

Ictalurus punctatus

The aquaculture of *I. punctatus* was developed at state and federal fish hatcheries of the USA during the 1950s for stocking reservoirs and sport fishing ponds. Many of the techniques developed at those hatcheries are still used to produce fry and fingerlings for large-scale commercial culture [106]. Larvae of *I. puncatus* are generally obtained by natural spawning of broodfish in ponds when egg masses are adhered to artificially made cavities. Spawning can also be induced by hormonal treatments when needed [107]. Then, egg masses are transported to the hatchery where they are incubated using well or surface water in rectangular troughs at 25ºC to 28ºC. Hatching normally occurs after 6 days of incubation [108].

Similar to *R. quelen*, larvae of *I. punctatus* at the onset of exogenous feeding (13–14 mm TL) present an external and internal anatomy similar to that of adult channel catfish, except for the reproductive system [68]. After hatching, *I. punctatus* fries are typically kept indoors under hatchery conditions (*i.e.*, good quality water supply, controlled rearing conditions, etc.) up to 8 days. During this period, fries are kept in rectangular troughs (2–4 m long) at a density of 150,000 to 200,000 fries per trough (45 specimens mL^{-1}) [106] (Table 5).

To reduce operational costs (*i.e.*, labour and feed), some hatcheries may stock yolk-sac fries at 2 dph (14.4–18.8 mg BW) in nursery ponds, before the onset of exogenous feeding. However, this practice results in reduced fingerling survival rates because of the reduced mobility of yolk-sac fries compared with that of older specimens. Moreover, yolk-sac fries are highly vulnerable to predators (i.e., aquatic insects, sunfish, and congeners not removed

646 from previous harvests). In contrast, stocking *I. punctatus* at the onset of exogenous feeding
647 or at 7 dph (22.8–29.1 mg BW) was shown to result in no deleterious effects on fingerling
648 production [106,109]. Regardless of the chosen age for fry stocking into nursery ponds, these
649 should be fertilised to ensure that adequate levels of feed are available. Zooplankton
650 populations are important in *I. punctatus* fry culture during the first 3–4 weeks, but their
651 importance diminishes as fish grow and are able to forage compound diets. Thus, the main
652 goal of fertilising fry ponds is to promote zooplankton growth while establishing a
653 phytoplankton bloom as quickly as possible to shade the pond bottom and prevent aquatic
654 plant growth between 3 and 4 weeks before stocking fish. Fries prefer large cladocerans (e.g.,
655 *Daphnia* sp., *Moina* sp., *Sida* sp.) to all other zooplanktonic organisms like small cladocerans,
656 copepods, and rotifers [110]. Thus, emphasis should be placed on fertilisation strategies that
657 increase cladoceran density; recommendations for fertilisation of channel catfish nursery
658 ponds may vary widely [111]. Regardless of the fertilisation strategy adopted, the
659 recommendation is to use high-nitrogen fertilisers rather than high phosphorous fertilisers.
660 Particularly, it is advisable to apply only inorganic fertiliser at an initial rate of *ca.* 20 kg N
661 ha^{-1} and 2 kg P ha^{-1}, followed by subsequent applications of 10 kg N ha^{-1} and 1 kg P ha^{-1} twice
662 a week for 3–4 weeks or until fries are stocked and commercial diets are administered. The
663 use of high-nitrogen fertilisers (i.e., the least expensive source of N available or urea if costs
664 are similar) results in shifting phytoplankton population to desirable algal groups, as well as
665 preventing macrophyte growth, promoting the growth of zooplanktonic organisms of large
666 size. After a few weeks, fries fed a combination of zooplankton and starter feeds will have
667 grown to fingerlings of 2.5 to 5 cm TL.
668 Regarding feeding practices, under common rearing conditions (26–28°C), yolk resorption
669 is completed at 4–5 dph, when the onset of exogenous feeding occurs. The digestive system
670 in *I. punctatus* fries is complete and functional at the onset of exogenous feeding. Thus, at 4–

671 5 dph, this species is able to ingest and efficiently digest compound feeds (i.e., starter diets).
672 At first feeding, *I. punctatus* fries are fed a compound diet (45–50% crude protein) at a ratio
673 of 25% stocked biomass (SB) (8–10 meals per day) until they are reared in nursery ponds.
674 Salmon and trout starter feeds may be used [106]; however, currently several starter feeds
675 especially formulated for *I. punctatus* fries are available. These starter diets are considered
676 nutritionally complete and may be used for 2 to 10 days before fish are stocked into grow-out
677 nursery ponds [112]; however, a number of dietary supplements might be used for partially
678 replacing traditional starter diets to increase fish growth rates and produce larger fries with
679 greater chances of surviving the critical transition from hatchery to nursery ponds. For
680 instance, Weirich *et al.* [113] recommended supplementing starter feeds with *Artemia*
681 decapsulated cysts (ADC), a feeding strategy previously tested in *C. gariepinus* [99]. Although
682 the particle size of ADC (200–250 µm) [99] is smaller than that recommended for *I. punctatus*
683 fries (420–560 µm) [114], feeding first-feeding fries for 10 days with a starter diet supplemented
684 with ADC promoted higher growth rates than those fed with just the compound feed.
685 Particularly, fries fed ADC were 61–98% heavier than their congeners fed the starter diet.
686 Traditionally, channel catfish farmers have used krill-based products as a dietary supplement
687 because of the well-balanced amino acid and fatty acid profile of such products; they contain
688 high levels of n-3 polyunsaturated fatty acids (PUFAs) including docosahexaenoic acid
689 (DHA) and eicosapentaenoic acid (EPA). However, feeding *I. punctatus* fries with krill-based
690 supplemented diets, contrary to what was traditionally thought, did not increase the growth or
691 survival rates of fries [109]. Zooplankton may also serve as a sustainable and reliable supplement
692 during *I. punctatus* hatchery production [115]. However, fries fed live or dried zooplankton
693 (copepods, cladocerans, and ostracods) performed worse in terms of somatic growth than fries
694 fed only a compound starter diet (Finfish Starter; Ziegler Brother, USA) or a combination of
695 the commercial diet with zooplankton. Although zooplankton from nursery ponds contain

65% crude protein and 9% fat [116], the dietary energy–protein ratio (digestible energy) of zooplankton may be too low for fry optimal growth when zooplankton is the only food source, and it is recommended to provide it in combination with compound diets [115]. In particular, fries fed dry zooplankton or live zooplankton combined with the commercial diet were 40% (292 mg BW) and 50% (312 mg BW) heavier, respectively, than fish fed only the commercial diet (209 mg BW). These results may be attributed to either an enhanced ingestion and digestion of feeds or the presence of micronutrients or trace elements in zooplanktonic organisms [117].

Pseudoplatystoma spp.

Pseudoplatystoma spp. larvae are obtained by hormonally induced spawning (26); after fertilisation, spawned eggs are generally incubated in cylindroconical tanks connected to a freshwater recirculating system [118]. In *Pseudoplatystoma* spp. and their interspecific and intergeneric hybrids, hatching occurs between 13 and 18 hpf in temperatures ranging from 26°C to 29°C, whereas the onset of exogenous feeding occurs at 2–3 dph (4.5–6 mm TL) [119-122]. *Pseudoplatystoma* spp. and interspecific hybrids are reared similarly. Larvae are fed *Artemia* nauplii during the first 7 to 10 days until they develop skin pigmentation [123,124]. Feeding ratios consist of 500 *Artemia* nauplii per larva per day during the first 5 days, and 1,000 nauplii per larva per day from day 6 to 10, divided in 6 to 10 rations distributed along 24 h [26,123]. In addition to *Artemia* nauplii, larvae can also be fed rotifers and egg yolk [26]. In experimental conditions, *P. punctifer* larvae have been successfully fed five times a day (only during daytime) with *Artemia* nauplii in slight excess from 4 to 12 days post fertilisation (0.6–9 nauplii mL^{-1}) [119]. Larvae may be also reared in complete darkness and kept in the incubation tanks up to 12 days [26] or transferred to rectangular or circular tanks, generally connected to a freshwater recirculating system, at 2–3 dph when they start swimming horizontally [123]. During

this phase, 15 larvae L^{-1} has been suggested as the best rearing density [120], although higher densities such as 30, 40, and 50 larvae L^{-1} have been successfully used in experimental conditions [119,125,126]. After this initial period when body pigmentation is developed, larvae can be reared indistinctly in tanks connected to a recirculating system or in outdoor fertilised ponds, a choice that depends on available facilities (Table 5) [26,123,124].

In recirculating systems, larvae are reared in darkness at a density of 5,000 to 10,000 larvae m^{-3} and fed naturally produced zooplankton (cladocerans and copepods) that are collected from fertilised ponds [26,120,123,124]. Larvae can be additionally fed minced fish or meat [26]. The transition from *Artemia* sp. to cladocerans and copepods is made in 10 days or more [123]. Fingerlings are fed at least eight times a day (including night-time) for 30 to 40 days until they reach 4 cm to 5 cm TL, and are continuously graded by size to avoid cannibalism [26].

Alternatively, larvae can be reared in fertilised ponds at a density of 100 to 150 larvae m^{-2} [26,124]. Before the transfer of larvae, ponds are sun-dried for 3 to 5 days to reduce the presence of predators and then limed. One week after liming, ponds can be fertilised with 0.1 kg m^{-2} rice bran and 0.01 kg m^{-2} urea or bovine (0.5 kg m^{-2}) or poultry (0.12 kg m^{-2}) manure with ammonium sulphate (0.02–0.05 kg m^{-2}) and single superphosphate (0.01-0.02 kg m^{-2}). The start of the zooplankton production takes 3 to 5 days, depending on temperature and sunlight [123]. Next, larvae are stocked into ponds during the first zooplankton bloom, and cladocerans are the optimal food at this period [26]. Phytoplankton blooms and the zooplankton production are regularly evaluated, and new fertilisations are undertaken if necessary. Ponds can also be stocked with forage fish larvae such as *Prochilodus lineatus*, *Leporinus* sp., or *Piaractus* sp. (forage fish to *Pseudoplatystoma* spp. larvae proportion 10:1) [26,123]. After 30 days, 4 cm to 5 cm TL fingerlings are harvested, preferably at night. Survival during this period is highly variable, depending on zooplankton abundance, weather conditions, or insect predation [26].

Pseudoplatystoma spp. fingerlings, 4 to 5 cm TL, are transferred to self-cleaning tanks with constant water renewal at a density of 1,500 to 6,000 juveniles m^{-3}, depending on the capacity of the system [26]; here, fingerlings are kept until they are sold for on-growing purposes (11–13 cm TL). Live feed is gradually eliminated, and fingerlings are progressively weaned over a period of 4 to 6 weeks onto formulated diets. These diets are based on moist feeds, including ingredients such as sardines, beef heart and lungs, frozen plankton, or minced fish gonads [26,123]. During this period, fingerlings continue to be periodically graded by size to reduce cannibalism. However, proper nutrition during early life stages is key to reduce the incidence of cannibalism and to significantly advance weaning [119]. Fernández-Méndez *et al.* [126] weaned *P. punctifer* at 18 dph within 3 and 6 days using moist and dry compound diets, respectively. The use of moist feeds resulted in better growth and survival, which may be linked to the taste and smell associated with the attractants released. In another study, *P. punctifer* larvae were successfully weaned at 12 dph from *A. nauplii* onto compound feed (45% proteins, including protein hydrolysate, and 15% lipids, rich in phospholipids) within 3 days [119], increasing survival and growth 2- and 6-fold, respectively, compared with *P. punctifer* larvae fed following preceding protocols under similar rearing conditions [59]. Indeed, as the digestive system of *P. punctifer* is completely functional at 9 dph (10.9 ± 0.18 mm TL), this species can be weaned at least from 9 dph onwards [59,74,75]. Moreover, recent nutritional studies with this species have accomplished weaning at 4 dph (Diana Castro-Ruiz, unpublished data), showing that significant advances in larviculture are possible using feeding protocols adapted to the digestive capacities and nutritional needs of this species during development. Nevertheless, the rather long procedures to achieve weaning used in commercial farming of *Pseudoplatystoma* spp. or their interspecific hybrids have encouraged producers to increasingly focus on intergeneric hybrids that readily accept formulated feeds, are omnivorous, and show lower cannibalism rates (Supplementary file 2), thus reducing

production costs and having high productivity [27]. However, even if their commercial production has rapidly increased, scientific data on the early culture of these hybrids are currently scarce.

Heteropneustes fossilis

Larvae of *H. fossilis* are either collected from the wild or are produced by artificial breeding (hormonally-induced spawning of sexually mature fish) in hatcheries. After fertilization, eggs are generally incubated in fibre-reinforced plastic (FRP) tray incubators at 26-30°C in an open-flow water system for 2 days [60,127]. The onset of exogenous feeding takes place at 2 dph at 29°C to 30°C when larvae measure 2.7-3.3 mm TL [60]. Newly hatched larvae are generally reared in indoor tanks (FRP or concrete) for an initial period of 10–12 days. When indoor tanks are used, larvae are stocked at a density of 3,000 to 5,000 larvae m^{-2} and fed 4–6 times a day with zooplankton (small rotifers and ciliates), *Artemia* nauplii, and egg custard (Table 6). After 12 days at 25–30°C, larvae measure 10–12 mm TL [128] and are transferred either to outdoor rearing tanks (*ca*. 2,000 L) or to small earthen ponds (50 m^2). Before larval stocking, outdoor tanks are provided with a 5–8-cm-thick layer of soil on the bottom and filled with water up to 25- to 30-cm height. Thereafter, the tanks are fertilised with superphosphate (*ca*. 100 g) and filtered bovine manure suspension (*ca.* 2 kg) for promoting zooplankton growth for a week. Then, larvae are stocked at 200 larvae m^{-2} and fed *ad libitum* with *Tubifex* sp., finely ground trash fish, rice bran, and chopped mollusc meat. Within a rearing period of one month, early juveniles reach 4–5 cm TL, when they are ready for stocking in grow-out ponds [129]. In earthen ponds, 12-dph larvae are stocked at 300–500 larvae per m^2. Ponds are prepared in advance for guaranteeing abundant zooplankton in order to promote larval survival [130]. Ponds are generally emptied, aquatic vegetation removed, and the soil exposed to sunlight for 15 days. Then, lime (300–1,500 kg ha^{-1}) is applied, and the pond filled with ground water.

After 5 or 6 days, ponds are fertilised with bovine manure (10,000 kg ha^{-1}), urea (300 kg ha^{-1}), and superphosphate (150–250 kg ha^{-1}) [131], although these quantities may vary according to local practices [132].

Regarding foraging behaviour, larvae can feed voraciously on zooplanktonic organisms and show preference for benthonic or substratum-associated prey such as ciliates, rotifers, copepod nauplii, small cladocerans, and ostracods [133,134]. In addition to zooplankton, larvae can feed on any kind of compound feed [133]. Larvae are also provided with supplementary feeds consisting of powdered rice bran, mustard oil cake, or granulated egg yolk [135]. Other authors recommend feeding *H. fossilis* larvae at a ratio of 5–10% BW with either finely minced trash fish and mollusc meat and rice bran (1:1) or a mixture of fishmeal, rice bran, groundnut oilcake/mustard oilcake, soybean, and wheat flour (2:2:3:1:2) [131]. Early juveniles are reared for 30–40 days in nursery ponds before they are stocked in grow-out ponds.

Different studies have been conducted to evauate better weaning strategy for *H. fossilis*, approaches that varied depending on the level of aquaculture development and geographic area considered. In India, Kumar *et al*. [136] evaluated different food items and their combination for first feeding at 2 dph to 22 dph (water temperature: 28.0–29.1°C; feeding rate: at apparent satiation and food distributed at 08:00, 12:00 and 16:00 h). Particularly, the following diets administered throughout the study were evaluated: 1) *Artemia* nauplii, 2) mixed pond-produced zooplankton (copepods and cladocerans), and 3) a commercial microdiet (Micro Elite 50, LuckyStar®, Singapore). In addition, the following dietary regimens were also tested: 4) non-enriched *Artemia* nauplii (2–8 dph), zooplankton (6–12 dph), and the microdiet (10–22 dph); 5) zooplankton (2–7 dph) and the microdiet (5–22 dph); and 6) zooplankton (2–12 dph) and the microdiet (9–22 dph). At the end of the trial, larvae fed with live feed showed better performance in terms of growth and survival, whereas no differences were observed in the development of the digestive system among the different dietary regimes. Therefore, it is

feasible to rear stinging catfish larvae without *Artemia* nauplii, and larvae may be weaned onto microdiets after 7 dph, as survival was the highest after this age (>65.6%; survival rate of larvae only fed the microdiet was 41.1%). Similarly, another study in Bangladesh evaluated different diets containing powdered milk, hen egg, boiled potato, and raw fish muscle (basal diet), and only differing in the inclusion of fish skin, viscera, and bones (rearing conditions: 26–29°C, 0.4–0.7 larvae L^{-1}, and feeding ratio: 10% SB) [137]. These authors found that first-feeding larvae (5.8 mm TL; 4 dph) fed a basal diet containing powdered milk, egg and boiled potatoes supplemented with boiled fish with skin, viscera and bones showed the best results in terms of growth (12.6 mm TL) and survival (60%) in comparison with larvae fed the basal diet incorporating just raw fish muscle with skin (12.0 mm TL; survival: 50%) and those fed the basal diet with raw fish muscle without skin (11.5 mm TL; survival: 50%). In addition, a study conducted in India focused on evaluating different food items (zooplankton, *Artemia* nauplii, snail meat, fish meat, and rice bran) on larval performance (rearing conditions: 25°C, 20 larvae L^{-1}, feeding ratio: 20% SB) [131]. Similar to other catfish species, the best results in terms of growth were found in larvae fed wild zooplankton (37 mg BW) followed by *Artemia* nauplii (*ca.* 24 mg BW), whereas other feed types resulted in low growth performance (snail meat: *ca.* 19 mg BW; fish meat: *ca.* 15 mg BW; rice bran: *ca*. 4 mg BW). However, this study did not include results on survival rates or the analysis of the proximate composition of the evaluated food items. In another study, *H. fossilis* larvae were fed with a mixture of zooplankton, egg custard, and *Artemia* nauplii for two weeks at 26–28°C. At the end of larval rearing in a circular cement cistern (2 m diameter), survival rate was 70% and larvae reached 10–20 mm TL [138].

Heteropneustes fossilis larvae can feed in darkness, showing prey selectivity patterns similar to those exhibited under light conditions because of the involvement of mechanoreception and chemoreception in prey detection [134]. This is not relevant when *H.*

fossilis larvae are reared in ponds, where there is generally no limitation in zooplankton availability; in contrast, when rearing *H. fossilis* larvae in tanks, special attention is needed to guarantee the presence of live prey at night time.

Rhamdia quelen

Although *R. quelen* can naturally spawn in captivity, hormonal induction is commonly used. Fertilized eggs are preferably incubated in Zoug-type incubators in continuous aerated freshwater flow [139]. Depending on water temperature, hatching takes place between 19 and 43 hpf at incubating temperatures of 30 ºC and 21ºC, respectively. Successful embryonic and larval development was found at different temperatures ranging from 21ºC to 30ºC, although some malformations (heart oedema) were found at 30ºC. According to these authors, the optimal water temperature for egg incubation is 26ºC, whereas larval size at hatching is inversely correlated to water temperature, even though this pattern is reversed after hatching. Fish size at hatching changes depending on the study with values ranging from 2.8 to 4.9 mm TL [62,140]. Such variability has been correlated to differences in spawning season, egg size and broodstock nutrition [141,142].

Silver catfish early culture can be conducted in indoor facilities under controlled conditions (intensive) for three weeks or, alternatively, directly in earthen ponds from the onset of exogenous feeding (2 dph) or after a short period of indoor culture (Table 6) [139,143]. In the latter case, according to Baldisserotto *et al.* [39], results are quite satisfactory, even better than those obtained in indoor tanks with clear water. Although the nursery stage for this species begins with fish weighting 1–3 g, it has been suggested to prolong the hatchery period until reaching a size of 5–6 g to improve survival rates [144]. Regardless of the rearing strategy chosen, recommended values of water pH and dissolved oxygen for silver catfish early culture

869 are 8.0–8.5 [145] and 6–8 mg O_2 L^{-1}, respectively [139]. Additionally, larval rearing of *R. quelen*
870 may be conducted in slightly brackish waters (up to 2 g NaCl L^{-1}) using feeding rates of 700
871 *Artemia* nauplii per larva and day [146].

872 Similar to other catfish species cultured in ponds, early culture of *R. quelen* in ponds
873 requires their careful preparation to assure the availability of live preys in adequate quantities.
874 In this context, the most common prey found in the stomach of *R. quelen* fry reared in
875 experimental fertilised ponds were ostracods, chironomid larvae, cladocerans, and calanoid
876 and cyclopoid copepods, whereas smaller prey such as rotifers and copepod nauplii were
877 seldom found [147]. If benthic prey become scarce, larvae may consume a higher proportion of
878 planktonic prey [148]. Before the introduction of fish, the ponds must be drained, limed, fertilised
879 (2,000 kg organic fertiliser ha^{-1}), and filled with filtered water (50–60 cm depth) 5 or 6 days
880 before the fish transfer [149,150]. Under pond rearing conditions, insects (Odonata, Hemiptera,
881 and Coleoptera) can prey on small *R. quelen* fries. The use of sieves or filtering nets in water
882 inlets is crucial to avoid the entrance of these predators [149]. Concerning the duration of the
883 period of intensive rearing before their transfer to ponds, Agüero *et al*. [151] recommend rearing
884 larvae indoors up to 8–10 dph (4.8 mg BW) (rearing conditions: 25.9°C; 25 larvae L^{-1}; feeding
885 larvae four times a day with an experimental dry diet) rather than directly stocking them at the
886 onset of exogenous feeding at 2 dph or at older ages (5 or 15 dph; 1.68 and 15.29 mg BW,
887 respectively). Santinón *et al*. [152] recommend transferring early juveniles to cages at 10 dph to
888 reduce feeding and operating costs. The authors verified that fish performed similarly after 65
889 days in net cages when transferring indoor reared larvae fed different experimental dry diets
890 (35% fish roe, fish silage or raw chicken liver) to outdoor net cages at the age of 10 dph (11.3–
891 26.7 mg BW) or 15 dph (23.5–115.1 mg BW, depending on the diet tested). Moreover, the
892 longer the period of indoor intensive culture, the higher the risks associated with the

893 appearance of pathologies or skeletal deformities [151,152]. These results might be attributed to a
894 lack of standardised rearing protocols and knowledge gaps on larval nutritional requirements
895 in this species [153].

896 In ponds, stocking density can reach 200 specimens m^{-2}, although this value should be
897 adjusted according to the food availability and the need of food supplementation. This can be
898 achieved using commercial feeds (≥40% protein) dispersed on the surface or placed on trays
899 that are then submerged *ca.* 15 cm from the bottom of the pond [149]. According to different
900 experimental studies, survival rates of 20–30% may be achieved after 40–45 days of rearing
901 in ponds at temperatures 23–26°C when supplementary food was offered (*I. punctatus*
902 compound feed or commercial dry food >28% crude protein) [151,154]. During this period, *R.*
903 *quelen* can grow to 3.2–3.3 g BW (*ca.* 6 cm TL). A common practice in ponds is the use of
904 lime to increase water hardness or pH. However, lime may contain different Ca^{2+} and Mg^{2+}
905 ratios that may substantially vary among ponds, directly affecting fish performance and the
906 regulation of their hydromineral balance. In this regard, water hardness affects silver catfish
907 performance [155,156]. In particular, when early juveniles were reared from first feeding during
908 three weeks in water containing 30 and 70 mg $CaCO_3$ L^{-1}, they grew (11.8 and 12.3 mm TL,
909 respectively) and survived (80.4% and 62.0%, respectively) better than at ≥150 mg $CaCO_3$ L^{-1}
910 (<10 mm TL, <9% survival) (156). Different levels of Ca^{2+} and Mg^{2+} were also studied by
911 Silva *et al.* [155]. The best survival (94.1–92.5%) and growth rates (19.6–18.7 mm TL) were
912 observed with 5.2 mg Ca^{2+} L^{-1} and 0.95 mg Mg^{2+} L^{-1} (water hardness: 20 mg $CaCO_3$ L^{-1}) and
913 20.3 mg Ca^{2+} L^{-1} and 2. 9 mg Mg^{2+} L^{-1} (water hardness: 70 mg $CaCO_3$ L^{-1}) compared with 150
914 mg $CaCO_3$ L^{-1}, regardless of Ca^{2+} and Mg^{2+} concentrations.

915 Biofloc technology has also been experimentally tested for *R. quelen* early culture [41]. These
916 authors tested different biofloc concentrations (as total suspended solids, TSS) obtained from

an intensive culture of tilapia (*Oreochromis niloticus*) in small experimental units (microcosms) where *R. quelen* larvae (2 dph) were fed exclusively on *Artemia* nauplii. After 21 days of culture, survival rates (38.1–54.4%) were significantly higher in all biofloc treatments than in the control group (10.2%), which was negatively affected by the protozoan *Ichthyophthirius multifiliis*. These results were attributed to the probiotic effect of the biofloc community. The best growth performance (21.1 mm TL; 88.6 mg BW) was obtained with TSS concentrations of 150–200 mg L^{-1} compared with higher TSS concentrations (400–600 or 800–1,000 mg L^{-1}) (16.2 and 15.9 mm TL; 45.7 and 44.5 mg BW; respectively) [41].

Intensive indoor early culture of silver catfish can be conducted under controlled conditions in tanks, using clear water, controlled water temperature (21–26°C) [157], and protection from predators. In these systems, cannibalism was observed from first feeding at 2 dph and became more frequent after 6 dph. As cannibalism was more prominent when fish were stocked at low densities, a stocking density of 10 specimens L^{-1} is recommended [139]. Although *R. quelen* can be fed exclusively on dry formulated diets from the onset of exogenous feeding, the best growth and survival rates are usually obtained by feeding larvae live prey (*Artemia* nauplii or collected zooplankton) alone or in combination with commercial or experimental dry feeds (45-56% crude protein, 10-18% crude lipid, <6% fibre, and <14% ash) [158-160]. In addition, other authors have shown that ADC were less effective than *Artemia* nauplii [161]. Comparatively, Luchini and Avendaño-Salas [154] found that rearing silver catfish larvae for 10 days using *Artemia* nauplii or a filtered mixture of cooked egg custard resulted in similar survival rates (76–82%), but *R. quelen* fed the egg custard grew better than those fed only *Artemia* (1.1 vs. 0.8 cm TL). When comparing live prey (*Artemia* nauplii, cysts or metanauplii) with dry diets (experimental diets based on yeast and raw bovine liver, and commercial diets such as Bio-Camaronina® or Anhami®; Anhami Nutrição Animal, PR, Brazil), these always performed worse than live preys alone [158,161,162], even if fish were fed for

the first five days with *Artemia* nauplii before weaning onto a dry diet [140]. However, Hernández *et al*. [163], comparing larval performance of two *R. quelen* biotypes from Argentina, one from the Pampean area (PA), and another from the North-eastern area (NE)—two lines from different geographical origin presenting different morphological and productive particularities [164] — reported that fish grew equally when fed live *Artemia* nauplii for 21 days from the onset of exogenous feeding or a dry formulated diet based on baker's yeast, fish meal and 2% soybean lecithin (53% protein). However, specimens from the PA showed the highest survival rates (>90%), producing the highest final biomass when fed on the dry diet. In this study, the NE biotype was more affected by skeletal deformities when fish were fed the dry diet in comparison to the PA biotype. These authors concluded that both biotic and abiotic factors (biotype and diet) must be considered when rearing *R. quelen* at early life stages in terms of skeletal development and quality. Growth and survival may be improved by co-feeding *R. quelen* fries with compound microdiets (>45% crude protein, >10% crude lipid, <6% fibre, and <14% ash) and *Artemia* nauplii [158,160]. Regarding the weaning time, Behr *et al.* [161] found similar results when feeding the silver catfish *Artemia* for 3 or 7 days before weaning. The best results in terms of weaning were achieved when fry were fed *Artemia* nauplii for 15 days before weaning, whereas the extension of this period to 20 days did not improve performance [165]. Comparing feeding frequencies, Lazzari *et al*. [166] did not find significant differences by feeding fries for 21 days on a dry diet hourly or every two hours. However, testing lower but larger ranges of feeding frequencies on fries fed a dry diet supplemented with *Artemia* nauplii, concluded that growth can be improved by feeding them three to seven times a day compared with feeding twice a day [167]. Besides, since no significant differences were found by increasing feeding frequency from three to seven times, the authors recommend feeding *R. quelen* three times a day, thus avoiding increasing production costs. Low light intensity (1.2 lx) is beneficial for silver catfish early culture fed live food during the

first week before weaning to a dry diet [168]. Improved larval specific growth rate (SGR) obtained under these conditions compared to 17 and 20 lx were in accordance with the nocturnal habits of this species [169]. To summarise, under adequate controlled conditions, 80–95% survival rates can be reached after 21 days of rearing. During this period, silver catfish fries can grow from about 2 mg and 5 mm TL to 118 mg and 20 mm TL [139].

Ompok bimaculatus

Eggs of *O. bimaculatus* are obtained by hormonally-induced spawning of a mature female and fertilized with the milt of a pool of males [61]. After fertilization, eggs are generally incubated between 27°C and 30°C in concrete or FRP incubators with continuous water-flow for 3 days. After hatching (23 ± 1 hpf), newly hatched larvae are kept in the incubators until the age of 3 dph. Larval rearing protocols for the butter catfish were summarised by Chakrabarti *et al.* [170]. In brief, *O. bimaculatus* larvae are generally reared in FRP tanks or cement cisterns for 40–45 days at 27–30°C (Table 7). When larvae reach a fingerling size of 5.0–6.0 cm TL and 3.0–4.5 g BW, they are then stocked in grow-out ponds. The mouth opens at 2 dph (3.3 mm TL), and first feeding occurs at 3 dph (4.2 mm TL), when larvae are fed finely sieved zooplankton (i.e., copepods, cladocerans) two times a day (early morning and evening). *Ompok bimaculatus* larvae are, then, fed finely chopped *Tubifex* sp. worms (food ratio: 25% stocked biomass, SB) from 7 dph (8–9 mm TL) until 15 dph. To avoid cannibalistic behaviour, grading of larvae of different sizes is recommended, in addition to providing shelters and hiding places when larvae are reared at 10 to 20 larvae L^{-1}. After 15 days, larvae are fed formulated diets based on egg custard, fishmeal, and silkworm pupae powder (feed ratio: 3–5% SB; distributed 2–3 times a day). Additionally, some hatchery managers also fed larvae with boiled and finely chopped chicken viscera or low-cost trash fish, or both. Other authors have reported feeding

991 larvae for 10 days at 27°C with wild zooplankton (no data on composition provided by
992 authors) and boiled egg yolk, resulting in 10.4% survival, whereas high mortality rates were
993 observed between 5 (10 mm TL) and 10 (25 mm TL) days associated with cannibalism and
994 the non-acceptance of food [171]. In another study, 3-dph larvae (2.3 ± 0.07 mm TL) were reared
995 in glass aquaria for 12 days with freshly hatched *Artemia* nauplii and wild zooplankton
996 (copepods, rotifers, and cladocerans). Both types of live prey were administered *ad libitum*
997 and twice a day (07:00 h and 16:00 h) [172]. Larvae of *O. bimaculatus* fed *Artemia* nauplii were
998 heavier than those fed live zooplankton (112 ± 8.1 vs. 94 ± 6.5 mg BW), and survival rates
999 were also higher (62.7 ± 5.2 vs. 47.3 ± 5.9%). After that period, fish were transferred to a
1000 cement cistern for 30 days and fed a mixture of rice bran, mustard oil cake, and dry fish powder
1001 (feed ratio not provided) daily. At the end of the study, fish were 7.5 cm TL, 5.5 g BW, and
1002 survival rate was 90%. Recent studies have focused on refining the feeding protocols for *O.*
1003 *bimaculatus.* In this context, first-fed larvae have been fed with a mixture of zooplankton [173]
1004 and at 7 dph (11 mm TL, 0.7 g BW) larvae were shifted to five different diets for a period of
1005 27 d (until 35 dph). The following diets were tested: 1) wild zooplankton, mainly composed
1006 of copepods, rotifers and cladocerans; 2) *Tubifex* sp. worms (64.8% crude protein, 14.0%
1007 crude fat, and 6.0% ash); 3) wild zooplankton + *Tubifex* sp.; 4) egg custards (whole, 2 g
1008 *Spirulina* sp. powder, 6 g corn flour, 4 g *Artemia* flakes, 2 g yeast, 6 g milk powder, and 10
1009 mL cod liver oil); and 5) compound feed (Gold Coin Biotechnologies, Singapore). At the trial,
1010 SGR values were higher in larvae fed a mixture of zooplankton and *Tubifex* sp. worms (SGR
1011 = 4.8 ± 0.6 % BW day^{-1}) than in larvae from the other treatments. Larvae fed *Tubifex* sp.
1012 worms (SGR = 4.1 ± 0.5 % BW day^{-1}) or wild zooplankton (SGR = 3.9 ± 0.1% BW day^{-1})
1013 showed intermediate values, and the lowest growth performance was found in larvae fed egg
1014 custards (SGR = 3.46 ± 0.31% BW day^{-1}) and the compound feed (SGR = 2.93 ± 0.24 % BW
1015 day^{-1}). A similar trend was observed when survival rates were considered; the highest survival

rates were recorded in larvae fed wild zooplankton + *Tubifex* sp. (66.50 ± 2.14%) and in those fed only *Tubifex* sp. (61.75 ± 2.02%), whereas the lowest, in fish fed the compound feed (45.8 ± 1.03%) (173). Similarly, Pradhan *et al.* [172] evaluated different weaning strategies based on the type of food (i.e., *Artemia* nauplii, wild zooplankton, and microdiet, Frippak Fresh CAR #1, INVE®, Dendermonde, Belgium) and co-feeding regimes on 2-dph larvae (3.3 ± 0.5 mm TL). In particular, diets were provided to apparent satiation four times a day (08:00, 12:00, 16:00, and 20:00 h). The authors concluded that weaning should not take place in *O. bimaculatus* earlier than at 7 dph (10.8 ± 0.1 mm TL). Moreover, larvae fed a co-feeding regime based on wild zooplankton or *Artemia* nauplii combined with the microdiet for 5 days showed good results in term of survival (65.0–78.7%) and growth performance (3.0–3.2 mm TL). In contrast, survival and size of larvae fed the compound diet from the onset of exogenous feeding were 48.7% and 2.6 ± 0.6 cm TL, respectively. In addition, early weaning of *O. bimaculatus* resulted in the delay of gut and pancreas development, impairing digestion and nutrient absorption, and ultimately, affecting larval performance. However, these results need to be considered with caution as the tested compound diet was formulated for larval and post-larval penaeid shrimps and not for freshwater fish larvae. In this context, it remains uncertain whether the nutritional requirements of *O. bimaculatus* larvae were met. For instance, Biswas *et al*. [174] fed weaned *O. bimaculatus* specimens for 30 days with five purified diets (49% crude protein and 8.2% crude fat) containing 2% of different attractants (betaine, DL-alanine, L-tryptophan, and inosine monophosphate) and found that dietary L-tryptophan and betadine promoted fry survival (48.7 and 41.3%, respectively) in comparison with the control diet (33%). The increase in survival in *O. bimaculatus* fed the diet containing 2% L-tryptophan was associated with a reduction in aggressive and cannibalistic behaviour among conspecifics. Regarding growth, the highest size was observed in the group fed the diet supplemented with 2% ionosine monophosphate (3.1 ± 0.05 cm TL) in comparison with the control group (2.8 ±

0.02 cm TL). These results may be attributed to the promotion of gut development due to dietary nucleotides. Weaning *O. bimaculatus* larvae (15 dph, 2.1 ± 0.1 cm TL) fed compound diets (49.8% protein, 8.2% lipid, 4.2% fibre, and 8.4% ash) supplemented with freeze-dried *Tubifex* sp. at 5% of stocked biomass resulted in higher survival rates when compared with those of the control group (43% vs. 28%), which was due to the presence of L-tryptophan in *Tubifex* sp. Regardless of the results from the above-mentioned studies, there is an urgent need of formulating specific compound diets for *O. bimaculatus* to promote high survival rates, growth, and larval quality.

<u>*Lophiosilurus alexandri*</u>

Early culture of *L. alexandri* in Brazil is mainly conducted under intensive conditions. Larvae are obtained from natural spawning in tanks with sand in the bottom, where fertilized eggs adhere [175]. Egg masses are collected and incubated in a box (40-150 L of functional volume) with aeration and an internal biological filter. The eggs are generally maintained in a 25-cm diameter sieve (0.5 mm mesh) fixed to floats, and hatching occurs between 24 and 48 h at 27-28°C [175,176]. Intensive culture of *L. alexandri* produces larvae during several months of the year, as spawning naturally occurs during 5 to 6 months.

The onset of exogenous feeding in *L. alexandri* occurs between 7 and 9 dph (12.0–15.5 mm TL) at 26–28°C. *Lophiosilurus alexandri* early culture is successfully performed in fresh water with *Artemia* nauplii (Table 7) [49,53,177,178,179]. However, the use of NaCl in the water is an interesting management technique during the initial phases of *L. alexandri* larviculture. In particular, larvae exhibit a $CL_{50–96h}$ of 8.9 g NaCl L^{-1} at 8 dph and tolerate up to 10 g NaCl L^{-1} at 12 dph (four days after first feeding) (14.1–15.5 mm TL) [180]. NaCl in the water increases *Artemia* nauplii survival, influences larvae physiology, and prevents sanitary problems, such as the occurrence of the protozoan *I. multifiliis*. Thus, larviculture of *L. alexandri* can be

conducted in slightly brackish waters (up to 2 g NaCl L^{-1}) at stocking densities of 20 to 60 larvae L^{-1} using *Artemia* nauplii as food [181]. The authors reported that survival reached 100% when using a density of 20 larvae L^{-1} and salinity of 2 g NaCl L^{-1}; survival was 93% when fresh water was used. However, SGR values were reduced when using a rearing density of 60 larvae L^{-1} and 4 g NaCl L^{-1}. These results [181] and those of Santos & Luz [50] indicate that rearing of *L. alexandri* larvae should be conducted at lower salinities of 4 g NaCl L^{-1}. A recent study using water with low salinity (2 g NaCl L^{-1}) under water recirculation conditions have shown that is feasible to rear *L. alexandri* larvae fed *Artemia* nauplii at densities ranging from 60 to 300 larvae L^{-1} without affecting growth performance (23–24 mm TL) or survival (>95%) after 15 days of trial [48]. From this perspective, it is important to highlight that this is the first study on the larviculture of freshwater, carnivorous species, reporting such good results using the above-mentioned high stocking rearing densities. In addition, laboratory studies have shown that *Artemia* nauplii density (300, 600 or 900 nauplii $larva^{-1}$ day^{-1}) was directly correlated to larval growth in TL and BW [50]; the increase in prey densities was linked to an increase in the levels of nitrogen compounds in the water (1.7 mg L^{-1} of un-ionised ammonia), but without negative effects on larval performance.

Regarding the optimal larval rearing temperature, no differences in survival rates were found among larvae reared between 23°C and 32°C (>90% after 15 days of larval rearing) (Table 7). However, larvae reared at 29°C and 32°C showed the highest size (27.2 mm); no differences in BW were found among larvae reared temperatures >26°C. Regardless of the rearing temperature considered, larvae fed high live prey densities (700 vs. 1,300 nauplii $larva^{-1}$ day^{-1}) presented better growth rates than those fed low live prey densities [51]. Santos *et al.* [178] found that during the first 15 days of feeding, the optimal live prey density in terms of BW and SL were 1,600 and 1,000–1,600 *Artemia* nauplii $larva^{-1}$ day^{-1}, respectively. However, no differences in survival were found when testing live prey densities ranging from 100 to 1,600

1091 nauplii larva^{-1} day^{-1}. Regarding feeding frequency, *L. alexandri* larvae can be fed *Artemia*
1092 nauplii two (at 8 and 17 h or at 8 and 12:30 h); three (at 8, 12:30 and 17 h); or four (at 8, 11,
1093 14 and 17 h) times a day, without differences in performance and survival (>89%) [179].
1094 Therefore, the final choice of feeding regimens will depend on the hatchery operators. Natural
1095 zooplankton [182,183] and the fairy shrimp (*Dendrocephalus brasiliensis*, Brachiopoda) [184] can
1096 also be used with positive outcomes. When wild zooplankton was offered to larvae stocked at
1097 densities of 150, 250 and 500 larvae per channel (0.43 m^2) in a continuous flow system for 20
1098 days, only survival was affected. Thus, survival in *L. alexandri* was inversely related to larval
1099 densities (lower densities, 60%; higher densities, 37%). These results were mainly associated
1100 with an increase in cannibalism [183]. In contrast, cannibalistic behaviour was not reported in
1101 larvae stocked at 300 larvae L^{-1} when fed *Artemia* [48].

1102 *Lophiosilurus alexandri* is generally weaned after 15 days using *Artemia* nauplii as live
1103 food in specimens with more than 20 mm TL at 27°C and 28.7°C. The transition to compound
1104 diets for early juveniles described by Luz *et al.* [53] is summarised in Table 8 (73% survival at
1105 the end of this period). Instead of using bovine heart, some authors have successfully used
1106 commercial gelatine powder (Gelita®, Eberbach, Germany) [185]. In addition, salinity values of
1107 4 g NaCl L^{-1} during this step should be avoided, as it reduces larval survival. Stocking density
1108 should also be considered during this period. When weaning was performed at 5, 10, 15, 30,
1109 and 40 fish L^{-1} (23.9 ± 1.2 mm TL and 0.12 ± 0.01 g BW) in recirculation aquaculture system
1110 (RAS), survival was lower than expected at high stocking densities (26% and 28% for
1111 densities of 30 and 40 fish L^{-1}, respectively), as a result of cannibalism. The highest survival
1112 rate (54%) was for the density of 5 juveniles L^{-1} [48].

1113 Regarding larviculture in RAS, the use of different biofiltration systems (biofilters internal
1114 or external to breeding tanks) and substrates (gravel and calcareous shell) led to similar
1115 performance in terms of growth and survival [182]. The comparison of different water flow rates

(one, four, and eight changes of total tank volume h^{-1}) revealed that the highest flow of water tested impaired larval growth because of their intense swimming. However, lower flow rates (0.3, 1, 2, and 4 total changes in tank volume h^{-1}) in a continuous water exchange system did not affect survival (values ranging from 71 to 76%) or growth (<23 mm TL; Luz *et al*. 2011). Recently, Melillo-Filho *et al*. [49] tested two tank drainage systems in RAS, one with water exiting from the surface and another from the water column, and the authors concluded that water surface drainage increased BW and survival. These results were associated to the greater retention of *Artemia* nauplii in the water column, increasing their chance of being consumed by larvae. When evaluating the two above-mentioned drainage systems in RAS units during the weaning period (feeding rate: 100% SB), survival was 61% and 72%, respectively. However, the high feeding rates significantly reduced water quality, hindering daily operations. Thus, the authors recommended weaning fish (7 specimens L^{-1}) by feeding them three times a day (at 9, 13, and 17 h) at a daily feeding rate of 50% SB (survival rates: 56–67%). This feeding strategy in RAS contributed to reducing labour costs associated with tank cleaning and maintenance, and minimised water quality problems [52].

Nutritional requirements during early life stages

Proper knowledge of the nutritional requirements throughout early development is important to optimise diets and feeding protocols and, thereby, improve larval and juvenile quality. The provision of high-quality, palatable, nutritive, and well-balanced diets is essential for promoting the growth, health, and well-being of fish throughout their life cycle [117]. Feed quality is of special importance during the larval stage, as larval nutritional requirements differ both qualitatively and quantitatively from those of juveniles or adults, as fish undergo dramatic morphological and physiological changes that are coupled with high growth rates (i.e., SGR in *C. gariepinus* larvae range from 15 to 141% BW day^{-1}) [186,187]. Thus, larvae have

to feed continuously and digest efficiently to support high growth rates [117]. Such food (live prey or compound feeds) must adequately provide larvae all the necessary macro- and micronutrients to support growth and health. Furthermore, technical characteristics of compound larval feeds, e.g., particle size, buoyancy/density, shape, consistency, texture, and colour as well as feeding regimen, are fundamental factors to be considered for meeting the feeding requirements of fish larvae [93]. Despite that, there are extremely limited data on larval nutritional requirements of different catfish species. Gaps in knowledge and bottlenecks exist not only in the design and formulation of compound diets but also in the use of live food for catfish larvae. This lack of knowledge hinders, in varying degrees, the early culture of several catfish species.

Proteins and essential amino acids

The reviewed catfish species are generally fed live prey during early culture. This type of food is easily produced and widely considered a reliable source of adequate nutrients, thus supporting efficiently fish survival, growth, and health [188-191]. In the absence of knowledge about the nutritional requirements during early life stages, the composition of the live food can generally be used as the starting point for approaching and establishing the qualitative and quantitative nutritional requirements of larvae [117]. For instance, Bwala *et al.* [189] provided information on the proximate composition and amino acid profile of three different *Artemia* types used as food for *C. gariepinus* larvae [*Artemia nauplii* developing either oviparously (55.9% crude protein, 11% crude lipid) or ovoviviparously (41% crude protein, data on lipid content not provided) and ADC (54.0% crude protein)]. The authors found that feeding *C. gariepinus* larvae with oviparous nauplii resulted in higher survival and protein efficiency ratio. Interestingly, oviparous nauplii had the lowest protein levels among the food items. These results were attributed to the protein quality and its digestibility rather than to their

content levels [192], although other factors such as larval foraging behaviour may also have affected the performance of larvae fed decapsulated cysts. Larvae and early juveniles (*ca*. <5 g) of *C. gariepinus* larvae have a high protein demand of 50–55% and a lipid requirement of 9%, whereas dietary carbohydrate content may be as high as 21% [103]. Regarding *I. punctatus* fries, a dietary protein level of 52% and 48% for fries from 0.02 to 0.25 g and from 0.25 to 1.5 g BW, respectively, as well as 3,650 kcal kg^{-1} digestible energy [193]. Furthermore, Robinson *et al*. [194] verified that *I. punctatus* fries fed salmon or trout starter feeds (protein: 51.5% and 55.7%; lipid: 14.8% and 11.5%, respectively) showed 50–75% weight gain and better feed conversion than fish fed a catfish starter feed (49.2 protein and 10.2% lipids). In the same context, Kelly *et al.* [195] compared three isocaloric practical diets (45% or 50% protein), including 50, 65 or 75% menhaden meal, and reported no differences in growth performance of *I. punctatus* fries among experimental diets or in comparison with a commercial salmonid starter diet (55% protein). Finally, these authors suggested that it is feasible to reduce dietary protein to 45%, although recent studies recommend 48% protein and 9% lipids for first-feeding fries [196]. These protein requirements were higher than those reported by Degani *et al*. [197] for this species (40%), as well as for other catfish species. For instance, in *Clarias* sp. hybrids (*C. batrachus* ♀ × *C. gariepinus* ♂ and *C. macrocephalus* ♀× *C. gariepinus* ♂), larval protein requirements for maximal growth were estimated at 35–40% [198]. Regarding *P. punctifer*, individuals performed best when weaned with a diet containing 45% protein and 15% lipids [from 8 mg at weaning (12 mm TL) to 600 mg 14 days later (50 mm TL)] [78,119]. However, *P. hypophthalmus* fries (0.2 g) showed better growth, survival rate, and feed conversion ratio when fed a diet containing 25% protein (and 5% lipids); no significant growth advantage was observed by increasing the dietary protein levels above 25% [199]. Major reasons for these differences in varying dietary protein percentages are owing to species-specific

feeding habits, variation in fish sizes, level of non-protein energy in the diets, protein quality, water temperature, and amount of natural food available in ponds when these requirements were established under a co-feeding or weaning conditions.

Although the protein requirements in different catfish species have been explored in an uneven way depending on the species considered, there is limited information about their requirements in terms of dietary amino acids (AA). Most of the available information has been obtained from larval AA profiles and AA utilisation in *C. gariepinus* at different stages of development and fed different diets [200]. Nevertheless, there are no particular nutritional studies focused on evaluating the essential AA requirements in this group of species at early life stages. To our knowledge, the only exception is the study from Khan & Abidi [201], which reported that the histidine requirements in *C. gariepinus* were 0.40–0.42% dry diet, corresponding to 1.0–1.05% of dietary protein.

Lipids and polyunsaturated fatty acids (PUFA)

Compared to studies on proteins, there are fewer studies evaluating the lipid nutritional requirements in larvae from the reviewed catfish species, and most of the available literature deals with juveniles, which is out of the scope of the present review. The total lipid level as well as the content of polar and neutral lipids and their fatty acid profile are important components affecting larval performance [202]. Larvae of several catfish species are able to synthetize arachidonic acid (20:4 *n*-6) and docosahexaenoic acid (22:6 *n*-3) from their C18 fatty acid precursors e.g., *P. punctifer* [78], *I. punctatus* [202] and *C. gariepinus* [204].

Feed nutrient richness can affect larval performance. *Artemia* nauplii does not satisfy the nutritional needs of 11-dph larvae of *P. punctifer* (12 mm TL), which is when cannibalism begins, coinciding with the end of the larval stage [59,74]. Enriching *Artemia* nauplii with a commercial enriching product high in DHA (*ca*. 43% total fatty acids, TFA, Algamac 3050®

1215 Aquafauna, Biomarine Inc., Hawthorne, CA, USA; ca. 4% TFA in enriched *Artemia*) from 3
1216 to 14 dph did not have any effect on *P. punctifer* growth compared with non-enriched *Artemia*.
1217 However, larvae fed enriched *Artemia* presented less fat in the liver but similar lipid deposits
1218 in the intestine [205]. In the same context, *P. punctifer* early juveniles were fed enriched *Artemia*
1219 as described by Darias *et al*. [205] and weaned from 14 dph onto a compound diet (10% lipids,
1220 38% proteins) showed improved growth and survival and a reduced incidence of cannibalism
1221 compared with those fed a non-enriched compound diet [206]. Moreover, early juvenile
1222 specimens fed both non-enriched *Artemia* and non-enriched compound diet showed a
1223 significant accumulation of lipids in the posterior intestine (steatosis) compared with that in
1224 the liver, contrary to specimens fed enriched-*Artemia* or enriched-compound diet. The latter
1225 presented similar amounts of lipids in both organs, indicating a more balanced digestive
1226 physiology [205]. Differences in dietary DHA/EPA and PUFA n-3/n-6 ratios between the two
1227 compound diets were responsible for differences in lipid accumulation. Furthermore, feeding
1228 *P. hypophthalmus* different dietary phospholipid levels (1, 2, 3 and 4%) revealed that
1229 increased dietary phospholipids is necessary for maintaining cellular membranes and even
1230 improving their normal physiological activities, supporting the idea that early life stages have
1231 higher nutritional requirements in phospholipids that juvenile stages due to their limited
1232 biosynthesis capacity [207].

1233 Although the dietary lipid requirements have not been determined for *R. quelen* during
1234 early life stages, Salhi *et al.* [208] revealed that increasing lipid levels (8 vs. 14%) improved fish
1235 performance. These authors recommended a diet with 38% crude protein and 14% crude fat.
1236 Similarly, *P. punctifer* early juveniles weaned from 12 dph a compound diet with 45% protein
1237 and 15% lipid levels, including hydrolysed fishmeal and phospholipids, showed significantly
1238 higher TL, BW, SGR and survival, and lower incidence of cannibalism than specimens fed
1239 diets containing 45:10, 30:15 and 30:10 protein:lipid levels. Histological and enzymatic

analyses of the digestive system unveiled a more developed digestive function in individuals fed the 45:15 diet, which indicated that a more balanced diet for *P. punctifer* early juveniles promoted a faster digestive system development and a better growth [78], compared with other diets.

Vitamins and minerals

Although few studies evaluated the vitamin requirements in different catfish species, Uys & Hecht [103] formulated and successfully tested a compound diet for first feeding *C. gariepinus* larvae. The following vitamin composition was recommended for this species and may be used as a guide for other catfish species, although little is known about the vitamin requirements for different species: vitamin A (65,000 IU kg^{-1}), vitamin D (12,000 IU kg^{-1}), vitamin E (943 IU kg^{-1}), vitamin K (100 IU kg^{-1}), thiamine (0.036 mg kg^{-1}), riboflavin (0.071 mg kg^{-1}), pyridoxine (0.019 mg kg^{-1}), pantothenic acid (0.445 mg kg^{-1}), biotin (0.611 mg kg^{-1}), choline (8,500 mg kg^{-1}), vitamin B12 (0.200 mg kg^{-1}), niacin (0.590 mg kg^{-1}), ascorbic acid (1,500 mg kg^{-1}), folic acid (0.013 mg kg^{-1}), and inositol (2,860 mg kg^{-1}). Other studies have addressed the requirements of particular vitamins. For instance, Merchie *et al.* [209] reported that the addition of ascorbyl palmitate (10%) into an emulsion for enriching *Artemia* metanauplii increased by 50% their vitamin C levels (500 µg g^{-1} DW), whereas 20 or 30% addition increased vitamin C in *Artemia* three- and six-fold. *Clarias gariepinus* fed vitamin-C-enriched *Artemia* nauplii resulted in high growth rates and stress tolerance. Moreover, Bardócz *et al.* [210] reported that ADC enriched with vitamin C (255 µg g^{-1} BW) increased SGR values in *C. gariepinus* compared with freshly decapsulated cysts. In *R. quelen* fries, the optimal dietary vitamin A levels in terms of growth performance and survival was found at 3,000 IU kg^{-1} in diets containing 56% crude protein and 10% crude fat [211]. Other vitamin-mix formulations for *I. punctatus* may be found in El-Saidy *et al.* [212] and Kelly *et al.* [195]. Regarding mineral

requirements for catfish larvae, there is even less information. Scarpa & Gatlin [213] revealed that the dietary zinc requirements of *I. punctatus* varied depending on water hardness; in particular, fries required 20 mg Zn kg^{-1} and 20–40 mg Zn kg^{-1} diet when reared in soft and hard waters, respectively. Furthermore, the recommended level of mineral mix inclusion in *I. punctatus* fry diets is 2% [211] and its mineral content (g kg^{-1} of dry diet) should be as follows: $CaHPO_4$ $2H_2O$ (3.75), $CaCO_3$ (4.25), KH_2PO_4 (3.5), Na_2CO_3 (2.0), $MnSO_4$ H_2O (0.088), FeCl $6H_2O$ (0.125), $MgSO_4$ (1.5), KIO_3 (0.0025), $CuSO_4$ $5H_2O$ (0.0075), $ZnCl_2$ (0.0375), $CoCl_2$ $6H_2O$ (0.0005), Na_3SeO_3 (0.0005), and Na_2MoO_4 $2H_2O$ (0.002).

Cannibalism in early life stages

One of the main bottlenecks in early culture of the reviewed catfish species, except for *I. punctatus*, is their high rates of intracohort cannibalism [86,119,214-216]. Cannibalistic behaviour may be affected by rearing density, feeding frequency, food availability, food composition, light intensity, and photoperiod.

Several studies have evaluated how manipulating illumination conditions (i.e., light intensity or wavelength, λ) reduced cannibalism during early life stages. In this context, Mukai [217] found that *P. hypophthalmus* larvae showed higher survival and growth rates when reared under 0.1 lx (1.40×10^{-3} μmol m^{-2} s^{-1}) of white fluorescent light compared with those reared under other light intensities (1, 10, and 100 lx). Moreover, Mukai *et al.* [218] demonstrated that *P. hypophthalmus* larvae showed more aggressive behaviour at higher light intensities (10 and 100 lx) than those under low light intensity (0 and 0.1 lx); these results corroborate those reported for *C. gariepinus* [214]. Furthermore, when *C. gariepinus* larvae were reared under normal photoperiod (600-1,000 lx during light hours) or continuous dark (<0.01 lx) conditions from hatching up to 20 dph, no differences in larval size were found, even though larvae reared under dark conditions had higher survival rates than those under normal photoperiod [89].

Yellow (λ = 570–590 nm) and red (λ = 620–750 nm) wavelengths at a light intensity of 1.40 × 10^{-3} μmol m^{-2} s^{-1} improved growth performance and survival rates in *P. hypophthalmus* [219]; in particular, larvae reared under red wavelength conditions showed higher SGR values than those under different wave lengths. In fact, when larvae were reared under dark conditions and low stocking density (10 larvae L^{-1}), their survival rates were higher than those of larvae reared in light conditions and at higher stocking densities (20 or 40 larvae L^{-1}). These results were associated with a reduction in cannibalism [220]. Regarding *P. fasciatum*, Nuñez *et al*. [221] showed that survival was higher in larvae reared under dark conditions (<0.01 lx) than in larvae reared under other light intensities (1 or 10 lx) and a 12:12 L:D photoperiod.

Behavioural studies revealed that *C. gariepinus* larval activity increased under dim light conditions, whereas the number of fish resting on the bottom of the aquaria decreased. These changes resulted in fewer larvae bitten by other individuals in comparison to light conditions, thus reducing cannibalism rates [89,215]. These authors recommended manipulating different rearing variables such as feeding frequency, food availability, and light intensity to reduce swimming activity to a minimum. Thus, it is generally recommended to rear *C. gariepinus* larvae and fingerlings at low light intensity values (<15 lx), assuring continuous food availability in tanks or ponds by feeding them every two hours when reared under intensive conditions [91]. Similarly to *R. quelen* [222], the incidence of cannibalism in *Pseudoplatystoma* spp. is generally reduced through size grading, rearing density, photoperiod and feeding frequency [123,216,221]. *Pseudoplatystoma punctifer* size is inversely related to prey size [216]. Thus, specimens of increasing size preferred increasingly smaller prey relative to their own size, which highlights the importance of size grading. Besides, the authors suggested that cannibalism could be reduced when feeding *P. punctifer* at least six times a day. Other authors have recommended frequent grading of larvae of different sizes in *P. punctifer* [123], in addition

to providing shelters and hiding places in *O. bimaculatus* to reduce or avoid cannibalistic behaviour [171].

Significant advances in reducing cannibalism have been achieved with the nutritional composition of feeds. In this regard, a low incidence of cannibalism in *P. punctifer* early juveniles associated with a high dietary phospholipid content (41% TFA) [119] was observed when compared with dietary regimes previously used [59]. The inclusion of phospholipids could have induced a reduction of aggressiveness and activity, as observed in humans and rats. Additionally, diets supplemented with tryptophan or ingredients rich in this non-polar aromatic amino acid have also been recommended for reducing intracohort cannibalism in different catfish species [174,223,224]. Finally, another issue to be considered when dealing with cannibalism during catfish larval rearing is their personality. In this context, Torres *et al.* [225] showed that *L. alexandri*, during the first 15 days of exogenous feeding with *Artemia* nauplii, tanks that had only "shy" or "bold" larvae exhibited higher survival rates than those with both personalities combined. This finding was due to the higher occurrence of cannibalism when "shy" and "bold" larvae were present in the same tank, whereas differences in BW might be related to their lower swimming activity. Other strategies for reducing cannibalistic behaviour in catfish species have been related to triploidy, as it has been described in *R. quelen* [226].

Cannibalism in *P. hypophthalmus* is largely independent from aggressiveness or feeding. However, it is a consequence of morphological traits, such as long sharp oral bones, which overhang from the mouth and prevent its closure at the start of exogenous feeding, and also an initially limited manoeuvrability caused by the late development of the pectoral fins [85]. As these morphological characteristics change during development, the associated risk is considered critical from 60 to 96 hpf and present until 129 hpf. Aggressiveness can be reduced with lower stocking densities, reducing the probability of contact between specimens. The significant mortality rate observed in this species during the first week of life is basically a

consequence of pathogenic infections of the wounds resulting from the encounters between larvae. In this context, survival and growth rates of *P. hypophthalmus* larvae were significantly improved when adding oxytetracycline (5 to 20 mg L^{-1}) or chloramine-T (2.5 mg L^{-1}) to the water; the use of the disinfectant is recommended in commercial hatcheries over the antibiotic to reduce the risk of bacterial resistance if applied incorrectly [227].

Omic approaches for improving catfish aquaculture

This section reviews the approaches conducted with omic technologies on the selected catfish species that have resulted in remarkable advances in the state of the art of catfish rearing. Nowadays, the omic analytical techniques (genomics, transcriptomics, regulomics, metabolomics and proteomics) may greatly contribute to the establishment of breeding programmes in aquaculture towards improving fish efficiency, production, quality and health [228-231]. Omics enable the understanding of the molecular basis underlying the influence of rearing conditions, nutrition, genetic background, or any other factor on survival, development, growth potential, immune resistance, and fish quality, among other parameters [232,233]. All genomic, transcript, and protein sources available for the reviewed catfish species are presented in Supplementary File 3. Although there is scarce transcriptomic and proteomic information related to early life stages of the different catfish species —*I. punctatus* is by far the most studied catfish species at the omics level—, these resources might open new avenues for improving early culture protocols, diets and feeding regimens, evaluating the impact of each factor on larval performance, quality, and health.

Mitochondrial DNA resources

The knowledge on mitochondrial DNA (mtDNA) is older than that on nuclear DNA. This sequence has been studied in *P. hypoththalmus* [234], *C. gariepinus* [235], *I. punctatus* [236], *O.*

bimaculatus [237], *P. reticulatum* [238], *H. fossilis* [239], *R. quelen* [240] and *L. alexandri* [241]. This deeper knowledge on mtDNA is due to its historical use as a source of information for phylogenetic, molecular evolution, and population genetic studies [242]. There is a large number of studies associating variations of mtDNA sequences with different populations of the same species; these data may provide useful information for conservation, breeding, and management programmes [243]. However, regardless of the wide use of full or partial mtDNA sequences for genetic analysis due to the higher mutation rate than in the nuclear genome, some particular features of the mtDNA may limit the power of these analyses. Unlike nuclear DNA, mtDNA resides in multiple cellular copies and may vary in sequence (heteroplasmy) and quantity among tissues [243]. In addition, when also considering the environment, mitochondria-encoded traits are influenced by interactions between the two genomes and a variety of environments and physiological conditions. Furthermore, the expanded use and relevance of mtDNA sequences in fish species are limited by the implementation of forward and reverse genetic studies to understand how sequence variation determines commercial traits [243]. It is expected that new approaches to address these issues will be available in the near future. In parallel, the broader implementation of third-generation sequencing technologies may help to fulfil one of the most relevant knowledge gaps in catfish species, including the identification and characterisation of single nucleotide polymorphisms (SNPs) for selecting genetic lineages in breeding programmes. In this regard, only deep and detailed association studies have been performed in *I. punctatus* [244]. An association of polymorphisms in prolactin gene and growth traits has been recently published in *I. punctatus* [245]. Furthermore, several genotyping efforts have been conducted to construct a high-density SNP array [246-249], which will certainly allow rapid advances on aquaculture selection programmes in ictalurid species and/or their hybrids.

Nuclear genomic resources

The nuclear genome sequence knowledge through NGS technologies in *I. punctatus* and *P. hypophthalmus* increased the set of known genes, transcripts, and proteins from these species [234,236]. In this context, the whole genome of these species is available and may be used as an initial platform for molecular breeding programmes to obtain novel catfish varieties using genomic approaches [244]. These approaches have served to identify the genetic basis of *I. punctatus* skull morphology, which has an enormous economic relevance because of its direct impact on fillet yield [250]. Additionally, sex determination mechanisms have been also unveiled in this species [251], which have important implications towards rearing monosex populations for improving growth and reproductive performances. However, the information for other catfish species remains limited to that obtained using the classical cloning methodologies. In this regard, Ju *et al.* [252] conducted an EST analysis of a cDNA library from the brain mRNA of *I. punctatus*. The number of available ESTs were increased through the analysis of different cDNA libraries from several fish tissues [253-255]. Based on previous cDNA libraries from *I. punctatus*, Ju *et al.* [256] used a low-density microarray to identify 61 differentially expressed genes in fish maintained at 12°C and 24°C. Key genes (including genes encoding chaperones and transcription factors, genes involved in lipid metabolism, and genes encoding translational machinery such as ribosomal proteins) involved in fish growth were identified; and how catfish rearing might be influenced under different rearing temperatures was also evaluated. Certainly, assessing the expression of those genes under early life stages might provide a molecular approach to determine the optimal rearing temperature during early life stages and how it might affect the performance at later growing phases. Furthermore, Li and Waldbieser [257] explored the altered transcriptome in *I. punctatus* spleen in a time-course from 2 h to 24 h after injection of lipopolysaccharide. The authors identified up to 138 differentially expressed genes, information that provided insights into the immune response of fish against a bacterial infection. Other studies have increased our understanding on how fish respond to particular

bacterial infections, such *Edwardsiella ictaluri*, and allowed the identification of the mechanisms of resistance to this gram-negative bacterium [258-260]. Nevertheless, the molecular knowledge on any biological response (e.g. immune system or heat stress response) in *I. punctatus* was soon further revolutionised with RNA-seq approaches [261,262]. These approaches, at the molecular level, lay the groundwork for further studies to be specifically conducted in developmental stages that are more sensitive to different stressors, such as the larval stages, and for predicting egg and embryo quality. In this context, a recent study evaluated a specific set of nine genes through quantitative PCR as potential markers of egg and embryo quality for hybrid catfish species [263], a promising approach to select high-quality egg batches and reducing the associated problems of rearing poor-quality eggs (e.g., low survival, reduced growth, and high incidence of skeletal deformities).

Non-coding RNA studies

In farmed catfish species, there is a limited but increasing number of studies identifying and characterising the role of non-coding RNAs (ncRNAs). There is increasing evidence of the critical control exerted by the tightly transcribed ncRNA genes in multi-cellular organisms through epigenetic changes and the control of post-transcriptional processes [264]. The number and type of ncRNAs known in each species compiled in the RNA central database are also shown in Supplementary File 3. As for the genomic, transcriptomic, and proteomic resources, most information regarding ncRNAs is specific to *I. punctatus*. In particular, Barozai [265] conducted the first approach of computational search for novel miRNA homologs and their targets along with their characterisation. At that time, 60 novel precursor miRNAs belonging to 45 families, including the bioinformatic prediction of the 341 proteins targeted by them, were identified and characterised. Instead, only 16 miRNAs (representing 12 miRNA families) and one mRNA target were reported by Xu *et al.* [266]. Just one year later, the use of

Solexa sequencing technology helped identify 237 conserved miRNAs and 45 novel miRNAs in *I. punctatus*, and the tissue expression pattern of some of them was reported [267]. Furthermore, the characterisation of the expression profile of miRNAs and the identification of potentially targeted mRNAs open new avenues to unveil the underlying mechanisms by which some biological features occur and those by which they might be transmitted to the future progenies and/or induce epigenetic imprinting [268].

Proteomic analyses

In parallel, as the high-throughput proteomic technologies were also developed and improved, the application of different methodologies, from the simplest polyacrylamide gel electrophoresis (PAGE) to the more complex isobaric tags for relative and absolute quantification (iTRAQ), further increased our understanding of the link between the genotype and phenotype [269]. One of the earliest applications of proteomic analysis was to characterise *I. punctatus* muscle, which is characterised by a pale/white colour with greyish to a slightly red tint, but stress may induce an undesirable reddish colour in fillets. In this context, Desai *et al.* [270] profiled the muscle proteomes employing two-dimensional electrophoresis and mass spectrometry and revealed over-abundant beta subunit of haemoglobin in reddish fillets. Further insights on how channel catfish fillet quality might be impacted by environmental and handling stress were obtained by these approaches [271]. Changes in the abundance of structural proteins and those involved in protein regulation and energy metabolism were identified, suggesting that increased proteolytic activity could be responsible for the alterations in colour and texture. A label-free quantitative proteomics workflow was also used to study how salinity affects the proteome of the kidney in *P. hypophthalmus* challenged with *Edwardsiella ictaluri* [272]. Among the 2,024 protein spots identified, 496 proteins were differentially expressed; most of them were related to cell metabolism, response to stress, cell structure, immunity and ion

homeostasis pathways, and functional categories. Furthermore, two-dimensional proteomic and mass spectrometry analysis of intestine and liver samples from *C. gariepinus* infected with *Aeromonas hydrophila* provided insight into host-pathogen interactions [273]. Unequivocally, further development of proteomic technologies and its wider implementation will certainly help address the current and future challenges in catfish species biology and domestication research.

Metabolomic studies

Metabolomics might be one of the last frontiers to gain an integrative understanding of fish physiology. Although, until now, only two studies have applied this technology in catfish species, these studies have already proved how metabolomic studies offer relevant information to evaluate the impact and to solve one of the persistent problems in *I. punctatus* aquaculture, anaemia. Using 1-D ^{1}H and 2-D ^{1}H J-resolved NMR analysis in healthy and anaemic *I. punctatus* kidney and liver tissues, the study revealed depleted energy sources, changes in metabolites associated with anaerobic metabolism or alternative energy pathways, as well as reduced taurine and inosine levels and protein synthesis [274]. Furthermore, a condition of oxidative stress was identified with an increase in valine, leucine, and isoleucine and a decrease in glutathione concomitant with a decreased respiratory gas transport capability through reductions in erythrocytes and haemoglobin markers in blood. Thus, this study clearly improved our understanding of anaemia symptoms and suggested useful biomarkers to identify fish status under farming conditions. A comparative analysis of brain nutritional metabolites showed how they are different depending the fish species considered, *Cyprinus carpio* vs. *I. punctatus*, and provided comprehensive information for the utilisation of fish heads in fish processing and dietary nutrition guidance [275].

The development of bioinformatic platforms would definitively provide optimal tools to address any biological question relevant to catfish aquaculture. In this context, specific educational programmes established by next-generation researchers would benefit the popularisation of such bioinformatic platforms. As global aquaculture relies on environmental conditions, an inherent vulnerability to climate change is evident. Climate change will be a major driver of aquaculture research needs in the future. A thorough understanding of how stressors affect fish physiology and how fish epigenetically adapt to new aquaculture conditions is of utmost importance. Research focused on these issues will help determine new engineering and management solutions to reduce the exposure to these stressors or mitigate their impact, or both. A combination of different approaches (i.e., genomics, transcriptomics, proteomics, metagenomics, metabolomics, and epigenomic) is recommended to gain a comprehensive, integrative, and clear understanding of any biological process occurring in catfish aquaculture under a climate change scenario. Such knowledge would allow to identify, validate, and apply potential biomarkers with predictive or diagnosis purposes. Thus, stressor-resistant traits can be genetically selected, and an adequate population variability maintained to improve resilience and overall fitness.

Microbiome studies

Recently, microbiome analyses have also been applied in catfish species. In this regard, both gut and skin microbiomes benefit the host species, probably by hindering the invasion of opportunistic pathogens, stimulating the immune system, or taking advantage of more nutritional metabolites available from the intestinal lumen [276]. These microbial communities can be disrupted or altered by different factors. Through ribosomal intergenic spacer analysis (RISA) and pyrosequencing, it was demonstrated that potassium permanganate exposure disturbed the external microbiomes in the skin and gills of *I. punctatus* and increased fish

mortality after a bacterial challenge with *Flavobacterium columnare* [277]. Similarly, feed containing florfenicol altered the *I. punctatus* gut microbiome, resulting in an increased relative abundance of potential opportunistic pathogens [278]. Both studies demonstrated that we need to beware of potent surface-acting disinfectants and antibiotics, when these are applied to avoid detrimental impacts on fish health. Furthermore, the gut microbiota is dynamic and adapts throughout fish development [279]. In this context, differences in microbiome communities were found along different larval developmental stages (i.e., egg, swim-up, 1 day of pond stocking, 24-h post stocking, and 21-d post stocking) in *I. punctatus*, indicating that the aquatic rearing environment and diet are important factors influencing the transfer of microbes from water (or food) into the gut [280,281]. These studies also indicated that even though probiotic treatments may be possible, gut community manipulation would require concurrent manipulation of pond environments. Indeed, fertilising ponds with livestock manure in catfish aquaculture is a common procedure; it might affect the microbial community and induce a primarily prebiotic effect on the pond ecosystem rather than a direct probiotic effect on fish [282]. Moreover, the sediment microbiome of catfish ponds responds to production practices; thus, monitoring the microbial community might be beneficial as a potential biomarker/predictor of catfish ponds productivity or fish physiological conditions, particularly during rearing at early life stages.

Future directions and conclusions

A constant and reliable source of fingerlings is required for a successful aquaculture industry and profitable farm operations, regardless of the final objective (i.e., human consumption, aquariology, or restocking). From this perspective, contrary to *C. gariepinus*, *I. punctatus*, R. *quelen* and *P. hypophthalmus*, whose hatchery procedures have been developed for sustaining a commercial large-scale production; consistent, reliable, satisfactory fry production is the

main bottleneck limiting the aquaculture of *Pseudoplatystoma* spp., *H. fossilis*, *O. bimaculatus*, and *L. alexandri*. In this context, matching the stage of development with zootechnology (e.g., optimal rearing conditions, first-feeding and weaning diets, weaning time) is essential to develop or optimise rearing procedures during catfish early life stages; it is also important for monitoring the success and failure of these protocols when implemented under local conditions. For this purpose, it is critical to optimise larval rearing protocols, considering species-specific developmental patterns and their nutritional requirements, to synchronise development with rearing procedures under controlled conditions. These approaches, regardless of the fry production system considered (i.e., tanks or ponds), may contribute to produce more robust larvae. Robust larvae may lead to reducing the potential losses derived from the transfer of larvae or fries from indoor conditions to ponds and the dependence of larvae on zooplankton.

Information on the nutritional requirements of catfish during early life stages is still scarce and fragmentary on some of the species considered. Few commercial compound diets specifically formulated for selected catfish species like *I. punctatus*, *C. gariepinus*, and *Pangasius* spp. are available in the market. However, in most cases, microdiets and starter diets for other aquatic species are used to feed early catfish larvae, a choice based on larval performance and production costs. In this context, the development of compound diets with locally available ingredients could improve rearing practices and their sustainability, as well as promote the aquaculture value chain and its stakeholders. From this point of view, most South American, Asian, and African countries where these species are cultured have adequate technological resources to manufacture appropriate feeds; however, the availability and cost of protein and oil ingredients may be major constraints. In most cases, the general paucity of good quality aquafeeds locally is a factor of scale. To properly foster the development of not only compound diets for early weaning for the different catfish species but also most efficient

and sustainable practices, future experiments should be designed. Therefore, the development of other strategies for enhancing larval health and welfare is needed. A crucial strategy would be the use of functional feeds that both promote and sustain somatic growth and enhance immune response. With this focus, a more holistic approach with different variables (i.e., levels of macro- or micronutrients, additives, and immunostimulants) and high-throughput technologies like omic tools under different rearing conditions (i.e., tank and pond larval rearing, stocking densities, feeding rates, water temperatures, and oxygen levels) may be tested to provide a more robust and realistic outcome. This approach may be conducted according to the level of technological development and research needs for each species at the local level. In addition, for reducing larval cannibalism and maximising larval performance and quality, nutritional and husbandry practices need to be further explored. In this regard, taking advantage of omic technologies, better breeding selection programmes, quality monitoring of eggs, embryos, and rearing water in ponds, as well as the formulation of highly balanced diets for each species might be possible in the nearest future. The implementation and further development of these tools might warrant a successful achievement of these high-priority goals in catfish aquaculture.

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References

1. Armbruster JW. Global catfish biodiversity. *Am Fish Soc Symp.* 2011; 77:15–37.
2. Sullivan JP, Lundberg JG, Hardman M. A phylogenetic analysis of the major groups of catfishes (Teleostei: Siluriformes) using *rag1* and *rag2* nuclear gene sequences. *Mol Phylogenet Evol.* 2006; 41:636–662.
3. Bruton MN. Alternative life-history strategies of catfishes. *Aquat Liv Resour.* 1996; 9:35–41.
4. Teugels GG. Taxonomy, phylogeny and biogeography of catfishes (Ostariophysi, Siluroidei): an overview. *Aquat Liv Resour.* 1996; 9:9–34.
5. Tucker CS, Hargreaves JA. *Biology and culture of channel catfish*. Elsevier, Amsterdam; 2004.
6. Potongkam K, Miller J. *Manual on catfish hatchery and production. A guide for small to medium scale hatchery and farm producers in Nigeria*. FAO, Rome; 2006.
7. Lazard J, Cacot P, Slembrouck J, Legendre M. La pisciculture des Pangasiidae. *Cahiers Agricultures* 2009; 18:164–173.
8. Lefevre S, Wang T, Jensen A, *et al*. Air-breathing fishes in aquaculture. What can we learn from physiology? *J Fish Biol.* 2014; 84:705-731.

9. FAO. Fishery and Aquaculture Statistics. Global production by production source 1950–2018 (FishstatJ). In: *FAO Fisheries and Aquaculture Department* [online]. Rome. Updated 2020. www.fao.org/fishery/statistics/software/fishstatj/en

10. IBGE. Pesquisa Pecuária Municipal: Produção da aquicultura. Available at: https://sidra.ibge.gov.br/Tabela/3940, accessed on 20 April 2021; 2021.

11. Ferraris CJ. Checklist of catfishes, recent and fossil (Osteichthyes: Siluriformes), and catalogue of siluriform primary types. *Zootaxa* 2007; 1418:1–628.

12. FishBase. FishBase, a global information system on fishes. Froese R, Pauly D eds. https://www.fishbase.se/home.htm; 2020.

13. Sriphairoj K, Na-Nakorn U, Klinbunga S. Species identification of non-hybrid and hybrid Pangasiid catfish using polymerase chain reaction-restriction fragment length polymorphism. *Agric Nat Resour.* 2018; 52:99–105.

14. Akinwole AO, Faturoti EO. Biological performance of African catfish (*Clarias gariepinus*) cultured in recirculating system in Ibadan. *Aquac Eng.* 2007; 36:18–23.

15. Na-Nakorn U, Brummett RE. Use and exchange of aquatic genetic resources for food and aquaculture: *Clarias* catfish. *Rev Aquacult.* 2009; 1:214–223.

16. Hargreaves J, Tucker CS. Industry development. *Dev Aquac Fish Sci.* 2004; 34:1–14.

17. Dunham RA, Elaswad A (2018) Catfish biology and farming. *Annu Rev Anim Biosci.* 2018; 6:305–325.

18. Buitrago–Suárez UA, Burr BM. Taxonomy of the catfish genus Pseudoplatystoma Bleeker (Siluriformes: Pimelodidae) with recognition of eight species. *Zootaxa* 2007; 1512:1–38.

19. Valladão GM, Gallani SU, Pilarski F. South American fish for continental aquaculture. *Rev Aquacult*. 2018; 10:351–369.

20. García-Dávila C, Sánchez H, Flores M, *et al*. Peces de consumo de la Amazonía Peruana. Instituto de Investigaciones de la Amazonía Peruana, Iquitos, Perú; 2018.

21. Carvalho-Costa LF, Piorski NM, Willis SC, Galetti PM, Ortí G. Molecular systematics of the neotropical shovelnose catfish genus *Pseudoplatystoma* Bleeker 1862 based on nuclear and mtDNA markers. *Mol Phylogenet Evol.* 2011; 59:177–194.

22. Estivals G, García-Dávila C, Darias MJ. Description of the skeletal anatomy of reared juveniles of *Pseudoplatystoma punctifer* (Castelnau, 1855) with notes on skeletal anomalies. *J Appl Ichthyol.* 2015; 31:88–97.

23. Crepaldi DV, Faria PMC, Teixeira EDA, *et al.* O surubim na aquacultura do Brasil. *Rev Bras Reprod Anim.* 2006; 30:150–158.

24. Oliveira AMS, Oliveira CAL, Rodrigues RA, *et al.* (2014) Crescimento de juvenis de *Pseudoplatystoma reticulatum* e *Pseudoplatystoma* spp. em viveiro. *Semina Ciências Agrárias* 2014; 35:1091–1098

25. Hashimoto DT, Senhorini JA, Foresti F, Porto-Foresti F. Interspecific fish 29. in Brazil: management of genetic resources for sustainable use. *Rev Aquacult.* 2012; 4:108–118.

26. Campos JL. O cultivo do pintado, *Pseudoplatystoma corruscans* (Spix e Agassiz, 1829), outras espécies do gênero *Pseudoplatystoma* e seus híbridos. In: Baldisserotto B, Gomes LC eds. *Espécies nativas para piscicultura no Brasil*, Editora UFSM, Santa Maria; 2013:335–361.

27. Alves AL, Varela ES, Moro GV, Kirschnik LNG. *Riscos genéticos da produção de híbridos de peixes nativos*. Palmas, Brazil: Embrapa Pesca e Aquicultura; 2014.

28. Hashimoto DT, Prado FD, Senhorini JA, Foresti F, Porto-Foresti F. Aquaculture of catfish hybrids: genetic strategies for conservation and management. In: Regan B, ed. Carp and Catfish: Biology, Behavior and Conservation Strategies. Nova Science Publishers, New York, 2015:1-30.

29. Burgess WE. *An atlas of freshwater and marine catfishes a preliminary survey of the Siluriformes*. TFH Publications, Neptune City; 1989.

30. Hossain MY, Islam R, Ahmed ZF, Rahman MM, Hossen MA, Naser SMA, Rasel RI (2015) Threatened fishes of the world: *Heteropneustes fossilis* (Bloch, 1794) (Siluriformes: Heteropneustidae). *Croatian Journal of Fisheries* 73:77–79.

31. Chakraborty BK, Nur NN. Growth and yield performance of shingi, *Heteropneustes fossilis* and koi, *Anabas testudineus* in Bangladesh under semi-intensive culture systems. *Int. J Agric Res Innov Technol.* 2012; 2:15–24.

32. Haniffa MA, Jafar SS, Bhat AA (2017) Seed production an urgent need for singhi (*Heteropneustes fossilis*) farming – a review. *Annals of Aquaculture and Research* 2017; 4:1038–1045.

33. Vijayakumar C, Sridhar S, Haniffa MA. Low cost breeding and hatching techniques of the catfish (*Heteropneustes fossilis*) for small-scale farmers. *Naga, ICLARMQ* 1998; 21:15–17.

34. Jothilakshmanan N, Marx KK. Hybridization between Indian catfish, ♀ *Heteropneustes fossilis* (Bloch) and Asian catfish, *Clarias batrachus* ♂ (Linn.). *Afr J Biotechnol.* 2013; 12:976–981.

35. Garavello JC, Shibatta OA. Reappraisal of *Rhamdia branneri* Haseman, 1911 and *R. voulezi* Haseman 1911 (Siluriformes: Hepatapteridae) from the rio Iguaçu with notes on their morphometry and karyotype. *Neotrop Ichthyol.* 2016; 14:e140111.

36. Perdices A, Bermingham E, Montilla A, Doadrio I. Evolutionary history of the genus *Rhamdia* (Teleostei: Pimelodidae) in Central America. *Mol Phylogenet Evol*. 2002; 15:172-189.

37. Ribolli J, Scaranto BM, Shibatta OA, Bombardelli RA, Zaniboni-Filho E (2017) DNA barcoding confirms the occurrence of *Rhamdia branneri* and *Rhamdia voulezi* (Siluriformes: Heptapteridae) in the Iguaçu River Basin. *Neotrop Ichthyol.* 2017; 15:e160147.

38. Silfvergrip AMC. *A systematic revision of the neotropical catfish genus Rhamdia* (Teleostei, Pimelodidae). PhD thesis, Stockholm University, Stockholm; 1996.

39. Baldisserotto B, Barcellos LG, Fracalossi DM, Kreutz L. Jundiá (*Rhamdia* sp.). In: Baldisserotto B, ed. Espécies nativas para piscicultura no Brasil, 3rd ed. Editora da UFSM, Santa Maria, 2020:245–288.

40. Garcia LDO, Coppatti CE, Wachholz F, Pereira-Filho W, Baldisserotto B. Freshwater temperature in the state of Rio Grande do Sul, Southern Brazil, and its implication for fish culture. *Neotrop Ichthyol*. 2008; 6:275–281.

41. Poli MA, Schveitzer R, de Olivera Nuñer AP. The use of biofloc technology in a South American catfish (*Rhamdia quelen*) hatchery: Effect of suspended solids in the performance of larvae. *Aquac Eng.* 2015; 66:17–21.

42. Banik S, Goswami P, Acharjee T, Malla S. *Ompok pabda* (Hamilton-Buchanan, 1822): an endangered catfish of Tripura, India: reproductive physiology related to freshwater lotic environment. *J Environ.* 2012; 1:45–55.

43. NBFGR. *Proceedings of national consultation on species prioritization for ex situ conservation and freshwater aquaculture*. September 17–18, 2011. NBFGR, Lucknow; 2011.

44. Tenório RA, Santos AJG, Lopes JP, Nogueira EMS. Crescimento do niquim (*Lophiosilurus alexandri* Steindachner 1876), em diferentes condições de luminosidade e tipos de alimento. *Acta Sci Biol Sci.* 2006; 28:305–309.

45. Campeche DFB, Balzana L, Figueiredo RCR, Barbalho MRS, Reis FJS, Melo JFB. Peixes nativos do Rio São Francisco adaptados para cultivo. Petrolina: Embrapa Semiárido, PE, Brazil; 2011.

46. Costa DCC, Silva WS, Melillo-Filho R, Filho KMC, Santos JCES, Luz RK. Capture, adaptation and artificial control of reproduction of *Lophiosilurus alexandri*: a carnivorous freshwater species. *Anim Reprod Sci.* 2015; 159:148–154.

47. Brasil. Lista Nacional Oficial de Espécies da Fauna Ameaçadas de Extinção – Peixes e Invertebrados Aquáticos. Ministério do Meio Ambiente. Portaria MMA n◦ 445, de 17 de dezembro de 2014; 2014.

48. Cordeiro NIS, Costa DC, Silva WSS, Takata R, Miranda-Filho KC, Luz RK. High stocking density during larviculture and effect of size and diet on production of juvenile *Lophiosilurus alexandri* Steindachner, 1876 (Siluriformes: Pseudopimelodidae). *J Appl Ichthyol.* 2016; 32:61–66.

49. Melillo-Filho R, Takata R, Santos AEH, *et al.* Draining system and feeding rate during the initial development of *Lophiosilurus alexandri* (Steindachner, 1877), a carnivorous freshwater fish. *Aquac Res.* 2014; 45:1913–1920.

50. Santos JCE, Luz RK. Effect of salinity and prey concentrations on *Pseudoplatystoma corruscans*, *Prochilodus costatus* and *Lophiosilurus alexandri* larviculture. *Aquaculture* 2009; 287:324–328.

51. Takata R, Silva WDSE, Costa DC, Melillo-Filho R, Luz RK. Effect of water temperature and prey concentrations on initial development of *Lophiosilurus alexandri* Steindachner, 1876 (Siluriformes: Pseudopimelodidae), a freshwater fish. *Neotrop Ichthyol.* 2014; 12:853–859.

52. Silva WS, Cordeiro NIS, Costa DC, Takata R, Luz RK. Frequência alimentar e taxa de arraçoamento durante o condicionamento alimentar de juvenis de pacamã. *Pesquisa Agropecuária Brasileira* 2014; 49:648–651.

53. Luz RK, Santos JCE, Pedreira MM, Teixeira EA. Effect of water flow rate and feed training on “pacamã” (Siluriforme: Pseudopimelodidae) juvenile production. *Arq Bras Med Vet Zootec.* 2011; 63:973–979.

54. Rønnestad I, Yúfera M, Ueberschär B, Ribeiro L, Sæle Ø, Boglione C. Feeding behaviour and digestive physiology in larval fish: current knowledge, and gaps and bottlenecks in research. *Rev Aquacult.* 2013; 5:S59–S98.

55. Gisbert E, Ortiz-Delgado JB, Sarasquete C. Nutritional cellular biomarkers in early life stages of fish. *Histol Histopathol.* 2008; 23:1525–1539.

56. Islam A. Embryonic and larval development of Thai Pangas (*Pangasius sutchi* Fowler, 1937). *Dev Growth Differ.* 2005; 47:1–6.

57. Verreth J, Toreele E, Spazier E, *et al.* Development of a functional digestive system in the African catfish *Clarias gariepinus* (Burchell). *J World Aquacult Soc.* 1992; 23:286–298.

58. Reyes RC. Descriptions of the early life stages of three common Ictalurids from the Sacramento-San Joaquin River Delta, California. Tracy Technical Bulletin 2010; 2.

59. Gisbert E, Moreira C, Castro-Ruiz D, Ozturk S, *et al*. Histological development of the digestive system of the Amazonian pimelodid catfish *Pseudoplatystoma punctifer*. *Animal* 2014; 8:1765–1776.

60. Kumar A, Pradhan PK, Chadha NK, *et al.* Ontogeny of the digestive tract in stinging catfish, *Heteropneustes fossilis* (Bloch) larvae. *Fish Physiol Biochem.* 2019; 45:667–679.

61. Pradhan PK, Jena JK, Mitra G, Sood N, Gisbert E. Ontogeny of the digestive tract in butter catfish *Ompok bimaculatus* (Bloch) larvae. *Fish Physiol Biochem.* 2012; 38:1601–1617.

62. de Amorim PM, Campos Gomes BV, Simoes Martins Y, Sato Y, Rizzo E, Bazzoli N. Early development of the silver catfish *Rhamdia quelen* (Quoy & Gaimard, 1824) (Pisces: Heptapteridae) from the Sao Francisco River Basin, Brazil. *Aquac Res.* 2009; 40:172–180.

63. Silveira J, Silva CP, Cargnin-Ferreira E, Alexandre D, Elias MA, Fracalossi DM. Freshwater catfish jundiá (*Rhamdia quelen*) larvae are prepared to digest inert feed at the exogenous feeding onset: physiological and histological assessments. *Fish Physiol Biochem.* 2013; 39:1581–1591.

64. Rocha MS, Silva RC, Santos JC, Schorer M, Nascimento MP, Pedreira MM. Comparative larval ontogeny of two fish species (Characiformes and Siluriformes) endemic to the São Francisco River in Brazil. *J Fish Biol.* 2020; 96:49–58.

65. Guimarães-Cruz RJ, Santos JE, Sato Y, Veloso-Júnior VC. Early development stages of the catfish *Lophiosilurus alexandri* Steindachner, 1877 (Pisces: Pseudopimelodidae) from the São Francisco River basin, Brazil. *J Appl Ichthyol.* 2009; 25:321–327.

66. Silverstein JT, Small BC. Reproductive physiology. *Dev Aquacult Fish Sci.* 2004; 34:69–94.

67. Hecht T. An alternative life history approach to the nutrition and feeding of Siluroidei larvae and early juveniles. *Aquat Liv Resour.* 1996; 9:121–133.

68. Grizzle JM. Reproductive biology. In: Tucker CS ed. *Channel Catfish Culture*. Elsevier, Amsterdam; 1985:229–282.

69. Yamagami K. Mechanisms of hatching in fish. *Fish Physiology* 1988; 11A:447–499.

70. Nolasco-Soria H, Moyano-López F, Vega-Villasante F, *et al*. Lipase and phospholipase activity methods for marine organisms. In: Sandoval G ed., *Lipases and Phospholipases*. Humana Press, Springer, New York, 2018:139–167.

71. Heming TA, Buddington RK. Yolk absorption in embryonic and larval fishes. *Fish Physiol.* 1988; 11A:407–446.

72. Nattabi JK. *Aspects of the digestive physiology of larvae of the North African catfish, Clarias gariepinus (Burchell 1822), during early development*. Doctoral Thesis, University of Stirling, Stirling; 2018.

73. Rangsin W, Areechon, N, Yoonpundh R. Digestive enzyme activities during larval development of striped catfish, *Pangasianodon hypophthalmus* (Sauvage, 1878). *Kasetsart J.* 2012; 46:217–228.

74. Castro-Ruiz D, Mozanzadeh MT, Fernández-Méndez C, *et al.* Ontogeny of the digestive enzyme activity of the Amazonian pimelodid catfish *Pseudoplatystoma punctifer* (Castelnau, 1855). *Aquaculture* 2019; 504:210–218.

75. Castro-Ruiz D, Andree KB, Fernández-Méndez C, García-Dávila C, Gisbert E, Darias MJ. Perfil de expresión de genes implicados en la digestión durante el desarrollo larvario y juvenil de la doncella *Pseudoplatystoma punctifer*. In: *IV Congreso Peruano de Biotecnología y Bioingeniería. Libro de Resúmenes*. Colegio de Biólogos del Perú - Consejo Regional IV La Libertad y Sociedad Peruana de Biotecnología, Trujillo; 2019:97–101.

76. Cahu C, Zambonino-Infante J. Substitution of live food by formulated diets in marine fish larvae. *Aquaculture* 2001; 200:161–180.

77. Nolasco-Soria H, Nolasco-Alzaga HR, Gisbert E. The importance of pepsin-like acid protease quantification in aquaculture studies: a revision of available procedures and presentation of a new protocol for its assessment. *Rev Aquacult.* 2020; 12:1928–1943.

78. Castro-Ruiz D, Andree KB, Solovyev MM, *et al*. The digestive function of *Pseudoplatystoma punctifer* early juveniles is differentially modulated by dietary protein, lipid and carbohydrate content and their ratios. *Animals* 2021; 11:369.

79. García-Ortega A, Verreth J, Segner H. Post-prandial protease activity in the digestive tract of African catfish *Clarias gariepinus* larvae fed decapsulated cysts of *Artemia*. *Fish Physiol Biochem.* 2000; 22:237–244.

80. Colchen T, Gisbert E, Krauss D, Ledoré Y, Pasquet A, Fontaine P. Improving pikeperch larviculture by combining environmental, feeding and populational factors. *Aquac Rep.*

1807 2020; 17:100337.
1808 81. De Silva SS, Nguyen PT. Striped catfish farming in the Mekong Delta: a tumultuous path
1809 to a global success. *Rev Aquacult.* 2011; 3:45–73.
1810 82. Nguyen PT, Bui TM, Nguyen TA, De Silva S. Developments in hatchery technology for
1811 striped catfish (*Pangasianodon hypophthalmus*). In: Allan G, Burnell G eds. *Advances in*
1812 *Aquaculture Hatchery Technology*. Woodhead Publishing Limited, Cambridge, 2013:
1813 498–518.
1814 83. Bui TM, Phan LT, Ingram BA, *et al*. Seed production practices of striped catfish,
1815 *Pangasianodon hypophthalmus* in the Mekong Delta region, Vietnam. *Aquaculture* 2010;
1816 306:92–100.
1817 84. Sah U, Wagle SK, Mehta SN, Mukhiya YK. Preliminary observations on breeding and fry
1818 rearing of pangas (*Pangasius hypophthalmus*) in eastern terai region of Nepal. *Int J Fish*
1819 *Aquat Res.* 2018; 3: 14–16.
1820 85. Baras E, Slembrouck J, Cochet C, Caruso D, Legendre M. Morphological factors behind
1821 the early mortality of cultured larvae of the Asian catfish, *Pangasianodon hypophthalmus*.
1822 *Aquaculture* 2010; 298:211–219.
1823 86. Baras E, Raynaud T, Slembrouck J, Caruso D, Cochet C, Legendre M. Interactions
1824 between temperature and size on the growth, size heterogeneity, mortality and cannibalism
1825 in cultured larvae and juveniles of the Asian catfish, *Pangasianodon hypophthalmus*
1826 (Sauvage). *Aquac Res.* 2011; 42:260–276.
1827 87. Slembrouck J, Baras E, Subagja J, Hung LT, Legendre M. Survival, growth and food
1828 conversion of cultured larvae of *Pangasianodon hypophthalmus*, depending on feeding
1829 level, prey density and fish density. *Aquaculture* 2009; 294:52–59.
1830 88. Vu N-U, Huynh T-G. Optimized live feed regime significantly improves growth
1831 performance and survival rate for early life history stages of Pangasius catfish

(*Pangasianodon hypophthalmus*). *Fishes* 2020; 5:20.

89. Mukai Y, Lim LS. Larval rearing and feeding behavior of African catfish, *Clarias gariepinus* under dark conditions. *J Fish Aquat Sci.* 2011; 6:272–278.

90. Mukai Y, Tuzan AD, Lim LS, Yahaya S. Feeding behavior under dark conditions in larvae of sutchi catfish *Pangasianodon hypophthalmus*. *Fish Sci.* 2010; 76:457–461.

91. Hecht T. A review of on-farm feed management practices for North African catfish (*Clarias gariepinus*) in sub-Saharan Africa. In: Hasan MR, New MB eds. *On-farm feeding and feed management in aquaculture*. FAO Fisheries and Aquaculture Technical Paper No. 583, FAO, Rome, 2013:463–479.

92. Boyd CE. Water quality management for pond fish culture. Developments in Aquaculture and Fisheries Science, 9. Elsevier Scientific Publishing Co., Amsterdam; 1982.

93. Tacon AGJ. *The nutrition and feeding of farmed fish and shrimp – a training manual*. 3. Feeding methods. FAO Field Document No. 7, Brasilia; 1998.

94. Hepher B, Pruginin Y. *Commercial fish farming*. John Wiley & Sons Inc., New York; 1981.

95. Amisah S, Adjei-Boateng D, Afianu DD. Effects of bamboo substrate and supplementary feed on growth and production of the African catfish, *Clarias gariepinus*. *Journal of Applied Sciences and Environmental Management* 2008; 12:25–28.

96. De Graaf G, Janssen H. Artificial reproduction and pond rearing of African catfish, *Clarias gariepinus* in Sub-Saharan Africa. FAO Fisheries Technical Paper No. 362. Rome, FAO; 1996.

97. Verreth J, Van Tongeren M. Weaning time in *Clarias gariepinus* (Burchell) larvae. *Aquaculture* 1989; 83:81–88.

98. Janssen JAL. Elevage du poisson-chat africain *Clarias lazera* (C&V) en République Centraficaine. III. Alevinage et grossissement en étangs. FAO projet GCD/CAF/007/NET. Document Technique No. 22, FAO, Rome; 1985.

99. Verreth J, Storch V, Segner H. A comparative study on the nutritional quality of decapsulated *Artemia* cysts, micro-encapsulated egg diets and enriched dry feeds for *Clarias gariepinus* (Burchell) larvae. *Aquaculture* 1987; 63:269–282.

100. Hecht T, Uys W, Britz PJ. The culture of sharptooth catfish, *Clarias gariepinus* in southern Africa. South African National Scientific Programmes Report No. 153. Council for Scientific and Industrial Research, Pretoria; 1988.

101. Oellermann LK. *A comparison of the aquaculture potential of Clarias gariepinus (Burchell, 1922) and its hybrid with Heterobranchus longifilis Valenciennes, 1840 in Southern Africa.* Ph.D. Thesis, Rhodes University, Grahamstown; 1995.

102. Chepkirui-Boit V, Ngugi CC, Bowman J, *et al.* Growth performance, survival, feed utilization and nutrient utilization of African catfish (*Clarias gariepinus*) larvae co- fed *Artemia* and a micro- diet containing freshwater atyid shrimp (*Caridina nilotica*) during weaning. *Aquac Nutr.* 2011; 17:82–89.

103. Uys W, Hecht T. Evaluation and preparation of a suitable dry feed and optimal feeding frequency for the primary nursing of *Clarias gariepinus* larvae (Pisces: *Clariidae*). *Aquaculture* 1985; 47:173–183.

104. Viveen WJAR, Richter CJJ, Van Oordt PGWJ, Janssen JAL, Huisman EA (1985) Practical manual for the culture of the African catfish (*Clarias gariepinus*). The Netherlands Ministry for Development Cooperation, Section for Research and Technology, The Hague; 1985.

105. Tucker CS, Robinson EH. *Channel catfish farming handbook*. Springer Science & Business Media, New York; 1990.

106. Avery JL, Steeby JA. Hatchery management. In: Tucker C & Hargreaves J, eds. *Biology and Culture of Channel Catfish*, Elsevier Press, Amsterdam, 2004:145–165.

107. Busch RL, Steeby JA. An evaluation of a leuteinizing hormone-releasing hormone analog to induce spawning of Channel catfish *Ictalurus punctatus*. *J World Aquacult Soc.* 1990; 21:10–15.

108. Steeby J, Avery J. Channel catfish broodfish and hatchery management. *SRAC Publication* 2005; 1803:1–8.

109. Weirich CR, O'neal CC, Belhadjali K. Growth, body composition, and survival of channel catfish, *Ictalurus punctatus* fry fed hatchery diets supplemented with krill meal. *J Appl Aquacult.* 2005; 17:21–35.

110. Mischke CC, Wise DJ, Lane RL. Zooplankton size and taxonomic selectivity of channel catfish fry. *N Am J Aquac.* 2003a; 65:141–146.

111. Mischke CC. Channel Catfish Pond Fertilization. In: Mischke CC ed. *Aquaculture pond fertilization, impacts of nutrient input on production*, Wiley-Blackwell, Ames; 2012:137–146.

112. NRC, National Research Council. *Nutrient requirements of fish and shrimp*. National Academies Press, Washington DC; 2011.

113. Weirich CR, Reigh RC, Glenn III DW. Evaluation of decapsulated *Artemia* cysts in hatchery diets for channel catfish *Ictalurus punctatus* fry and effects on subsequent fingerling production. *J World Aquacult Soc.*2000; 31:609–617.

114. Dupree HK, Huner JV (1984) Nutrition, feeds, and feeding practices. In: Dupree HK, Huner JV eds. *Third report to the Fish Farmers*. US Department of the Interior, Fish and Wildlife Service, Washington DC; 1984:141–157.

115. Mischke CC, Wise DJ, Byars TS. Evaluation of zooplankton in hatchery diets for channel catfish fry. *N Am J Aquac.* 2009; 71:312–314.

116. Mischke CC, Li MH, Zimba PV. Pond fertilization does not affect nutritional value of zooplankton in channel catfish nursery ponds. *N Am J Aquac.* 2003; 65:248–254.

117. Hamre K, Yúfera M, Rønnestad I, Boglione C, Conceição LE, Izquierdo M. Fish larval nutrition and feed formulation: knowledge gaps and bottlenecks for advances in larval rearing. *Rev Aquacult.* 2013; 5:S26–S58.

118. Fondepes. *Protocolo de Reproducción de Doncella (Pseudoplatystoma punctifer)*. Fondo Nacional de Desarrollo Pesquero – Fondepes, Lima; 2015.

119. Darias MJ, Castro-Ruiz D, Estivals G, *et al.* Influence of dietary protein and lipid levels on growth performance and the incidence of cannibalism in *Pseudoplatystoma punctifer* (Castelnau, 1855) larvae and early juveniles. *J Appl Ichthyol.* 2015; 31:74–82.

120. Inoue LAKA, Ceccarelli PS, Senhorini JA. A larvicultura e a alevinagem do Pintado e do Cachara. *Revista Panorama da Aqüicultura* 2003; 13:15–21.

121. Moreno-Guerra YA, Mira-Lopez TM, Rodriguez-Pulido JA, Medina-Robles VM. Desarrollo embrionario de híbridos de *Pseudoplatystoma metaense* Suarez, 2007 x *Leiarius marmoratus* Gill, 1870 (Siluriformes: Pimelodidae). *Orinoquia* 2016; 20:78–85.

122. Oliveira D. *Fase embrionário e larval do híbrido Pseudoplatystoma reticulatum x Leiarius marmoratus e do parental Leiarius marmoratus.* MSc Thesis, Universidade Federal do Rio Grande do Sul, Porto Alegre; 2013.

123. Inoue LAKA, Hisano H, Ishkawa MM, Rotta MA, Senhorini JA. *Princípios básicos para produção de alevinos de surubins (Pintado e Cachara).* Embrapa Agropecuária Oeste, Dourados, Brazil; 2009.

124. Silva AFL, Russo MR, Ramos LDA, Rocha AS. Feeding of larvae of the hybrid surubim *Pseudoplatystoma* sp. under two conditions of food management. *Acta Sci Biol Sci.* 2013; 35:149–155.

125. Núñez J, Castro D, Fernández C *et al*. Hatching rate and larval growth variations in *Pseudoplatystoma punctifer*: maternal and paternal effects. *Aquac Res.* 2011; 42:764-775.

126. Fernández-Méndez C, David F, Darias MJ, Castro-Ruiz D, Núñez-Rodríguez J. Rearing of the Amazon catfish *Pseudoplatystoma punctifer* (Castelnau, 1855): weaning with dry and moist diets. *J Appl Ichthyol.* 2015; 31:83–87.

127. Puvaneswari S, Marimuthu K, Karuppasamy R, Haniffa MA. Early embryonic and larval development of Indian catfish, *Heteropneustes fossilis*. *EurAsian J Biosci.* 2009; 3:84-96.

128. Nayak PK, Pandey AK, Singh BN, Mishra J, Das RC, Ayyappan S. Breeding, larval rearing and seed production of the Asian catfish, *Heteropneustes fossilis* (Bloch). Central Institute of Freshwater Aquaculture, Bhubabeswar; 2000.

129. Nayak PK, Sahoo SK, Ferosekhan S (2018) Breeding and seed production of Singhi, *Heteropneustes fossilis*. In: *Package of practices for breeding and culture of commercially important freshwater fish species*, National Fisheries Development Board, Hyderabad, 2018:17–19.

130. Devaraj KV. Culture of air-breathing fishes. *Seafood Export Journal* 1975; **7**:35–41.

131. Nayak PK, Mishra J, Kumar K, Sahoo S, Satpathy BB, Ayyappan S. Live food for the early larval growth of catfish *Heteropneustes fossilis* (Bloch). *Indian J Fish.* 2003; 50:333–338.

132. Rahman MA, Habib KA, Hossain MA, Azad SO, Rayhan MZ. Impacts of stocking density and economic returns on the cage culture of stinging catfish, *Heteropneustes fossilis*. *Int. J Fish Aquat Stud.* 2017; 5:198–201.

133. Thakur NK, Das P. Synopsis of biological data on singhi *Heteropneustes fossilis* (Bloch 1794). *Bul Cent Inland Fish Res Inst.* 1985; 39:1–32.

134. Mookerji N, Rao TR. Prey capture success, feeding frequency and daily food intake rates in rohu, *Labeo rohita* (Ham.) and singhi, *Heteropneustes fossilis* (Bloch) larvae. *J Appl Ichthyol.* 1995*;* 11:37–49.

135. Jhingran VG. *Fish and Fisheries of India*, 3rd edn. Hindustan Publishing Corporation, New Delhi; 1991.

136. Kumar A, Pradhan PK, Chadha NK, Mohindra V, Tiwari VK, Sood N (2018) Effect of dietary regimes on development of digestive system of stinging catfish, *Heteropneustes fossilis* (Bloch) larvae. *Int J Curr Microbiol Appl Sci.* 2018; 7:413–421.

137. Saha JK, Islam MA, Das M, Rahamatullah SM, Islam MS. Studies on the induced breeding and post-larval rearing of shing (*Heteropneustes fossilis* Bloch). *Bangladesh J Fish Res.* 1998; 2:139–144.

138. Chaturvedi CS, Singh RK, Raju KD, Ambulkar RS, Pandey AK. Induced breeding and larval rearing of stinging catfish, *Heteropneustes fossilis* (Bloch), under controlled conditions in Raipur, Chhattisgarh (India). *J Exp Zool.* 2015; 18:645–649.

139. Silva LVF (2004) Incubação e larvicultura. In: Baldisserotto B, Radünz Neto J, eds. *Criação de Jundiá*. Editora da UFSM, Santa Maria, Brasil; 2004:107–116.

140. Hernández DR, Domitrovic HA, Sánchez, S. Evaluación de diferentes dietas en la alimentación del bagre sudamericano (*Rhamdia quelen*). In: *IV Congreso Iberoamericano Virtual de Acuicultura* (*www.civa2006.org*), 2006:1151–1155.

141. Parra JEG, Radünz Neto J, Veiverberg CA, *et al*. 2008. Alimentação de fêmeas de jundiá com fontes lipídicas e sua relação com o desenvolvimento embrionário e larval. *Ciencia Rural* 2008; 38:2011–2017.

142. Parra JEG, Radünz Neto J, Veiverberg CA, *et al*. Desempenho reprodutivo de fêmeas de jundiá alimentadas com diferentes fontes protéicas. *Arch. de Zootec.* 2010; 59:255–265.

143. Luchini L. *Manual para el cultivo de bagre sudamericano (Rhamdia sapo)*. FAO RLAC/90/16-PES-20, Santiago; 1990.

144. Barcellos LJG, Kreutz LC, Quevedo RM, *et al.* Nursery rearing of jundiá, *Rhamdia quelen* (Quoy & Gaimard) in cages: cage type, stocking density and stress response to confinement. *Aquaculture* 2004; 231:383–394.

145. Lopes JM, Silva LVF, Baldisserotto B. Survival and growth of *Rhamdia quelen* (Pimelodidae) larvae exposed to different water pH. *Aquac Int.* 2001; 9:73–80.

146. Fabregat TEHP, Damian J, Fialho NS, *et al*. Acute toxicity of common salt and intensive larviculture of silver catfish Rhamdia quelen in brackish water. *Arq Bras Med Vet Zootec*. 2015; 67:547-554.

147. Zagarese HE. Rearing fry of South American catfish (*Rhamdia sapo*) on natural zooplankton populations. *Aquaculture* 1998; 70:323–331.

148. Zagarese HE. Effect of selective planktivory of fry of *Rhamdia sapo* (Pimelodidae: Pisces) on zooplankton community structure. *Freshw Biol.* 1990; 24:557–562.

149. Radünz Neto J (2004) Manejo alimentar-Nutrição. In: Baldisserotto B, Radünz Neto J, eds. *Criação de Jundiá*, Editora da UFSM, Santa Maria, Brasil; 2004:143–160.

150. Chediak G, Varela Z. Manejo de estanques para la cria de semilla de bagre negro. *Anales III Congreso Nacional de Veterinaria*. Montevideo; 1982:933–950.

151. Agüero CH, Hernández DR, Roux JP, Sánchez S, Santinón JJ. Crecimiento y supervivencia de larvas de *Rhamdia quelen* criadas en estanques luego de diferentes períodos de larvicultura intensiva. *Rev Vet.* 2014; 25:34–39.

152. Santinón JJ, Hernández DR, Sánchez S, Domitrovic HA. Duração da larvicultura sobre o desempenho posterior de juvenis de jundiá, *Rhamdia quelen*, recriados em tanques-rede. *Ciência Rural* 2010; 40:1180–1185.

153. Boglione C, Gisbert E, Gavaia P *et al.*. Skeletal anomalies in reared E uropean fish larvae and juveniles. Part 2: main typologies, occurrences and causative factors. *Rev Aquac.* 2013; 5:S121-S167.

154. Luchini L, Avendaño-Salas T. Cría de larvas de *Rhamdia sapo* (Val.) Eig. en estanques Primeros ensayos. *Revista de la Asociación de Ciencias Naturales del Litoral* 1983; 16:137–147.

155. Silva LVF, Golombieski JI, Baldisserotto, B. Growth and survival of silver catfish larvae, *Rhamdia quelen*, (Hepatpteridae), at different calcium and magnesium concentrations. *Neotrop Ichthyol.* 2005; 3:299–304.

156. Townsend CR, Silva LVF, Baldisserotto B. Growth and survival of *Rhamdia quelen* (Siluriformes, Pimelodidae) larvae exposed to different levels of water hardness. *Aquaculture* 2003; 215:103–108.

157. Chippari-Gomes AR, Gomes LC, Baldisserotto B. Lethal temperatures for *Rhamdia quelen* larvae (Pimelodidae). *Ciência Rural* 2000; 30:1069–1071.

158. Carneiro PCF, Mikos JD, Schorer M, Oliveira-Filho PRC, Bendhack F. Live and formulated diet evaluation through initial growth and survival of jundiá larvae, *Rhamdia quelen*. *Sci Agric.* 2003; 60:615–619.

159. Castañeda G, Esquivel J, Muelbert B, Vásquez-Torres W, Fracalossi DM. Larvicultura de *Rhamdia quelen* (Pisces, Pimelodidae) con proteína vegetal y animal, suplementada con plancton. *Revista MVZ Córdoba* 2011; 16:2678–2685.

160. Salhi M, Bessonart M. Growth, survival and fatty acid composition of *Rhamdia quelen* (Quoy & Gaimard, 1824) larvae fed on artificial diet alone o in combination with *Artemia* nauplii. *Aquac Res.* 2013; 44:41–49.

161. Behr ER, Tronco AP, Radünz Neto J. Ação do tempo e da forma de suplementação alimentar com *Artemia franciscana* sobre a sobreivência e o crescimento de larvas de jundiá. *Ciência Rural* 2000; 30:503–507.

162. Neto PGB, Dutra FM, Ballester ELC, Portz L. Crescimento e sobrevivência de larvas do jundiá, *Rhamdia quelen*, alimentadas com alimento vivo enriquecido e dieta artificial. *Revista Brasileira de Ciência Veterinária* 2013; 20 216–221.

163. Hernández DR, Santinón JJ, Sánchez S, Domitrovic HA. Crecimiento, supervivencia e incidencia de malformaciones óseas en distintos biotipos de *Rhamdia quelen* durante la larvicultura. *Lat Am J Aquat Res.* 2014; 41:877–887.

164. Wicki G, Rossi F, Martín S, Luchini L. Cría de bagre randiá en Argentina: crecimiento comparado entre dos líneas de distinto origen silvestre. *Infopesca Internacional* 2006; 26:33–39.

165. Diemer O, Neu DH, Sary C, Finkler JK, Boscolo WR, Feiden A. *Artemia sp*. na alimentação de larvas de jundiá (*Rhamdia quelen*). *Ciênc. Anim Bras.* 2012; 13:175–179.

166. Lazzari R, Radünz Neto J, Lima RL, Pedron FA, Losekan ME. Efeito da frequência de arraçoamento e da troca do tamanho de partícula alimentar no desenvolvimento de pós-larvas de jundiá (*Rhamdia quelen*). *Revista Brasileira de Agrociência* 2004; 20:231–234.

167. Gomes ACL, Fosse PJ, Rodrigues MF, Lengruber ELS, Lengruber EO, do Amaral AA. Efeito da frequência alimentar na sobrevivência e no desenvolvimento de larvas de jundiá (*Rhamdia quelen*) em condições experimentais. *Revista Ifes Ciência* 2019; 5:198–207.

168. Behr ER, Radünz Neto J, Tronco AP, Fontana AP. Influência de diferentes níveis de luminosidade sobre o desempenho de larvas de Jundiá (*Rhamdia quelen*) (Quoy & Gaimard, 1824) (Pisces: Pimelodidae). *Acta Sci Biol Sci.* 1999; 21:325–330.

169. Gomes LC, Golombieski JI, Chippari-Gomes AR, Baldisserotto B. Biologia do jundiá *Rhamdia quelen* (Teleostei, Pimelodidae). *Ciência Rural* 2000; 30:179–185.

170. Chakrabarti PP, Mandal SC, Chattopadhaya DN, Mandal RN, Paul BN, Jayasankar P. *Pabda- Seed Production & Culture*. Central Institute of Freshwater Aquaculture, Bhubaneswar, India; 2012.

171. Raizada S, Lal KK, Sarkar UK, *et al.* (2013) Captive breeding and embryonic development of butter catfish (*Ompok bimaculatus*, Bloch 1794), a threatened fish of Indian sub-continent in Northern India. *Proc Natl Acad Sci India Sect B Biol Sci.* 2013; 83:333–339.

172. Pradhan PK, Jena JK, Mitra G, Sood N, Gisbert E. Effects of different weaning strategies on survival, growth and digestive system development in butter catfish *Ompok bimaculatus* (Bloch) larvae. *Aquaculture* 2014; 424–425:120–130.

173. Malla S, Banik S. Larval rearing of an endangered catfish, *Ompok bimaculatus* (Bloch, 1794) with live and artificial diets: A preliminary study in Tripura, India. *International Journal of Fauna and Biological Studies* 2015; 2:1621.

174. Biswas PRP, Patel AB, Saha H. Effect of dietary incorporation of chemo-attractants on growth and survival during seed rearing of *Ompok bimaculatus* (Bloch). *Turkish J Fish Aquat Sci.* 2018; 18:491–499.

175. Costa DC, Takata R, Silva WS, *et al.* Description of amino acid and fatty acid content during initial development of *Lophiosilurus alexandri* (Siluriformes: Pseudopimelodidae), a carnivorous freshwater catfish. *Neotrop Ichthyol.* 2018; 16:e180014.

176. Goncalves Junior LP, Mattioli CC, Martins EFF, *et al.* (2019) Temperature-induced changes in reproductive variables in the teleost fish *Lophiosilurus alexandri*. *J Therm Biol.* 2019; 80:133–140.

177. Pedreira MM, Luz RK, Santos JCE, Sampaio EV, Silva RSF. Biofiltração da água e tipos de substrato na larvicultura do pacamã. *Pesqui Agropecu Bras.* 2009; 44:511–518.

178. Santos JCE, Correia ED, Luz RK. Effect of daily *Artemia* nauplii concentrations during juvenile production of *Lophiosilurus alexandri. Boletim do Instituto de Pesca* 2015; 41:771–776.

179. Santos JCE, Pedreira MM, Luz RK. Feeding frequency in pacamã larviculture. *Revista Caatinga* 2016; 29: 512–518.

180. Luz RK, Santos JCE. Avaliação da tolerância de larvas do pacamã *Lophiosilurus alexandri* Steindachner, 1877 (Pisces: Siluriformes) a diferentes salinidades. *Acta Sci Biol Sci*. 2008; 30:345–350.

181. Luz RK, Santos JCE. Densidade de estocagem e salinidade de água na larvicultura de pacamã. *Pesquisa Agropecuária Brasileira* 2008b; 43:903–909.

182. Pedreira MM, Santos JCE, Sampaio EV, Ferreira FN, Silva JDL. Efeito do tamanho da presa e do acréscimo de ração na larvicultura de pacamã. *R. Bras Zootec.* 2008; 37:144–150.

183. Lopes CM, Sampaio EV. Sobrevivência e crescimento larval do pacamã *Lophiosilurus alexandri* Steindachner 1876 (Siluriformes, Pimelodidae), em função de três densidades de estocagem em laboratório. *Acta Scientiarum* 2000; 22:491–494.

184. Lopes JP. *Considerações sobre a branchoneta, Dendrocephalus brasiliensis, (Crustacea, Anostraca, Thamnocephalidae) como fonte alternativa na alimentação de alevinos de espécies carnívoras*. Monografia (Especialização em Aquicultura). Universidade Federal Rural de Pernambuco, Recife; 1998.

185. Salaro AL, Oliveira Junior JC, Lima FW, *et al.* Gelatin in replacement of bovine heart in feed training of *Lophiosilurus alexandri* in different water salinities. *An Acad Bras Cienc.* 2015; 87:2281–2287.

186. Enyidi U, Onuoha JU. Use of probiotics as first feed of larval African catfish *Clarias gariepinus* (Burchell 1822). *Annu Res Rev Biol.* 2016; 9:1–9.

187. Conceição LEC, Dersjant-Li Y, Verreth JAJ. Cost of growth in larval and juvenile African catfish (*Clarias gariepinus*) in relation to growth rate, food intake and oxygen consumption. *Aquaculture* 1998; 161:95–106.

188. Verreth J, Eding EH, Rao GRM, Huskens F, Segner H. A review of feeding practices, growth and nutritional physiology in larvae of the catfishes *Clarias gariepinus* and *Clarias batrachus*. *J World Aquacult Soc.*1993; 24:135–144.

189. Bwala R, Salie K, Van Stappen G. Ovoviviparously produced *Artemia* nauplii are a suitable live food source for the larvae of the African catfish (*Clarias gariepinus*: Burchell, 1822). *Aquac Res.* 2018; 49:3319–3328.

190. Adewolu MA, Akintola SL, Akinwunmi OO. Growth performance and survival of hybrid African catfish larvae (*Clarias gariepinus x Heterobranchus bidorsalis*) fed on different diets. *Zoologist* 2009; 7:45–51.

191. Adeyemo AA, Oladosu GA, Ayinla AO. Growth and survival of fry of African catfish species, *Clarias gariepinus* Burchell, *Heterobranchus bidorsalis* Geoffery and *Heteroclarias* reared on *Moina dubia* in comparison with other first feed sources. *Aquaculture* 1994; 119:41–45.

192. Hoornyck V. Heat treatment affects protein quality and protease activity in decapsulated cysts of *Artemia* when used as starter food for larvae of African catfish *Clarias gariepinus* (Burchell). *Aquac Nutr.* 2000; 6:25–31.

193. Winfree RA, Stickney RR. Formulation and processing of hatchery diets for channel catfish. *Aquaculture* 1984; 41:311–323.

194. Robinson EH, Steeby J, Brent JR. Evaluation of three feeds for hatchery rearing channel catfish fry. *J World Aquac Soc.* 1989; 20:256–260.

195. Kelly AM, Kohler CC, Ayala CE. Menhaden meal in practical diets for channel catfish fry and fingerlings reared in intensive systems. *North Am J Aquac.* 2002; 64:290–293.

196. Sink T D, Lochmann R T, Kinsey NR. Growth and survival of channel catfish, *Ictalurus punctatus,* fry fed diets with 36 or 45 percent total protein and all plant or animal protein sources. *J World Aquac. Soc.* 2010; 41:124–129.

197. Degani G, Ben-Zvi Y, Levanon D. The effect of different protein levels and temperatures on feed utilization, growth and body composition of *Clarias gariepinus* (Burchell 1822). *Aquaculture* 1989; 76:293–301.

198. Giri SS, Sahoo SK, Sahu AK, Meher PK. Effect of dietary protein level on growth, survival, feed utilisation and body composition of hybrid Clarias catfish (*Clarias batrachus* × *Clarias gariepinus*). *Anim. Feed Sci. Technol.* 2003; 104:169–178.

199. Chuapoehuk W, Pothisoong K. Protein requirements of catfish fry, *Pangasius sutchi*, Fowler. In: Cho CY, Cowey CB, Watanabe T eds. *Finfish nutrition in Asia: methodological approaches to research and development.* IDRC, Ottawa; 1985:103–105.

200. Conceição LEC, Ozório ROA, Suurd EA, Verreth JAJ. Amino acid profiles and amino acid utilization in larval African catfish (*Clarias gariepinus*): effects of ontogeny and temperature. *Fish Physiol Biochem.* 1998; 19:43–58.

201. Khan MA, Abidi SF. Optimum histidine requirement of fry African catfish, *Clarias gariepinus* (Burchell). *Aquac Res.* 2009; 40:1000–1010.

202. Gisbert E., Villeneuve L, Zambonino-Infante JL, Quazuguel P, Cahu CL. Dietary phospholipids are more efficient than neutral lipids for long-chain polyunsaturated fatty acid supply in European sea bass *Dicentrarchus labrax* larval development. *Lipids* 2005; 40:609–618.

203. Satoh S, Poe WE, Wilson RP. Studies on the essential fatty acid requirement of channel catfish, *Ictalurus punctatus*. Aquaculture 1989; 79:121–128.

204. Verreth J, Coppoolse J, Segner H. The effect of low HUFA-and high HUFA-enriched *Artemia*, fed at different feeding levels, on growth, survival, tissue fatty acids and liver histology of *Clarias gariepinus* larvae. *Aquaculture* 1994; 126:137–150.

205. Darias MJ, Castro-Ruiz D, García-Dávila C, Gisbert E. Effects of *Artemia* and inert diet enrichment with DHA on lipid deposition in the intestine and liver of *Pseudoplatystoma punctifer* larvae and early juveniles. In: FENACAM & LACQUA/SARA (WAS)'15: Abstract book. LACQUA, ABCC, WAS, Fortaleza; 2015:140.

206. Magris J, Sánchez FJ, Sylvain G, *et al.* Improving larval feeding protocols for Doncella, *Pseudoplatystoma punctifer*, by enriching *Artemia* and compound diets. In: IV Conferencia Latinoamericana sobre Cultivo de Peces Nativos: Latin American and Caribbean Aquaculture 2013. Villavicencio, Universidad de los Llanos; 2013.

207. Tocher DR, Bendikse EA, Campbell PJ, Bell JG. The role of phospholipids in nutrition and metabolism of teleost fish. *Aquaculture* 2008; 280:21–34.

208. Salhi M, Bessonart M, Chediak G, Bellagamba M, Carnevia D. Growth, feed utilization and body composition of black catfish, *Rhamdia quelen*, fry fed diets containing different protein and energy levels. *Aquaculture* 2004; 231:435–444.

209. Merchie G, Lavens P, Verreth J, *et al.* The effect of supplemental ascorbic acid in enriched live food for *Clarias gariepinus* larvae at startfeeding. *Aquaculture* 1997; 151:245–258.

210. Bardócz T, Kovacs E, Radics F, Sandor Z. Experiments for the improved use of decapsulated *Artemia cysts* in intensive culture of African catfish larvae. *J Fish Biol.* 1999; 55:227–232.

211. Peil SQ, Pouey JLOF, Lopes PRS, Martins CR, Timm G. Addition of vitamin a in the diet of post-larvae of silver catfish. *Biodiversidade Pampeana* 2007; 5:9–15.

212. El-Saidy DMSD, Dabrowski K, Bai SC. Nutritional effects of protein source in starter diets for channel catfish (*Ictalurus punctatus* Rafinesque) in suboptimal water temperature. *Aquac Res.* 2000; 31:885–892.

213. Scarpa J, Gatlin III DM. Dietary zinc requirements of channel catfish, *Ictalurus punctatus*, swim-up fry in soft and hard water. *Aquaculture* 1992; 106: 311–322.

214. Appelbaum S, Kamler E. Survival, growth, metabolism and behavior of *Clarias gaeriepinus* (Burchell 1822) early stages under different light conditions. *Aquac Eng.* 2000; 22:269–287.

215. Almazán-Rueda P, Schrama JW, Verreth JA. Behavioural responses under different feeding methods and light regimes of the African catfish (*Clarias gariepinus*) juveniles. *Aquaculture* 2004; 231:347–359.

216. Baras E, Silva del Aguila DV, Montalvan Naranjos GV, *et al.* How many meals a day to minimize cannibalism when rearing larvae of the Amazonian catfish *Pseudoplatystoma punctifer*? The cannibal's point of view. *Aquat Liv Resour.* 2011; 24:379–390.

217. Mukai Y. High survival rates of sutchi catfish, *Pangasianodon hypophthalmus*, larvae reared under dark conditions. *J Fish Aquat Sci.* 2011a; 6:285–290.

218. Mukai Y, Tan NH, Lim LS. Why is cannibalism less frequent when larvae of sutchi catfish *Pangasianodon hypophthalmus* are reared under dim light? *Aquac Res.* 2013; 46:1958–1964.

219. Tan NH, Yusoff NH, Ismail KM, Sallehudin MF, Mukai Y. Influence of light wavelength and intensity on the survival and somatic growth of the early larval stage of sutchi catfish *Pangasianodon hypophthalmus*. *Int J Aquatic Sci.* 2017; 8:113–119.

220. Mukai Y. Remarkably high survival rates under dim light conditions in sutchi catfish *Pangasianodon hypophthalmus* larvae. *Fish Sci.* 2011b; 77:107–111.

221. Nuñez J, Dugué R, Corcuy Arana N, *et al.* (2008) Induced breeding and larval rearing of Surubí, *Pseudoplatystoma fasciatum* (Linnaeus, 1766), from the Bolivian Amazon. *Aquac Res.* 2008; 39:764–776.

222. Costenaro-Ferreira C, Oliveira RRB, Oliveira PLS, *et al*. Cannibalism management of jundiá fry, *Rhamdia quelen*: behavior in heterogeneous batches fed on food with different particle sizes. *Appl Anim Behav Sci.* 2016; 185:146–151.

223. Haetami K, Zidni I, Rostika R, Ginanjar W. Effect of addition of banana peel extract on commercial feed as an effort to reduce patin cannibalism (*Pangasius hypophthalmus*) larval stage. *Asian J Fish Aquat Res. 2*019; 4:1–9.

224. Rawat P, Biswas P, Jena AK, Patel AB, Pandey PK. Effect of dietary incorporation of natural attractants on growth and survival during seed rearing of Indian butter catfish, *Ompok bimaculatus*. *J Environ Biol.* 2019; 40:661–667.

225. Torres IFA, Júlio GSDC, Figueiredo LG, Lima NLC, Soares APN, Luz RK. Larviculture of a carnivorous freshwater catfish, *Lophiosilurus alexandri*, screened by personality type. *Behav Process.* 2017; 145:44–47.

226. Morón-Alcain E, Mendia AC, Muñoz LH, *et al*. (2017) Effects of heat and cold shock-induced triploidy on productive parameters of silver catfish (*Rhamdia quelen*) late-hatched in the reproductive season. Aquaculture 2017; 473:303–309.

227. Subagja J, Slembrouck J, Hung LT, Legendre M. Larval rearing of an Asian catfish *Pangasius hypophthalmus* (Siluroidei, Pangasiidae): analysis of precocious mortality and proposition of appropriate treatments. *Aquat Liv Resour.* 1999; 12:37–44.

228. Robledo D, Palaiokostas C, Bargelloni L, Martínez P, Houston R. Applications of genotyping by sequencing in aquaculture breeding and genetics. *Rev Aquacult.* 2017; 10:670–682.

229. Raposo de Magalhães CSF, Cerqueira MAC, Schrama D, Moreira M J V, Boonanuntanasarn, S, Rodrigues PMLA. Proteomics and other Omics approach in the context of farmed fish welfare and biomarker discovery. *Rev Aquacult.* 2020; 12:122–144.

230. Alfaro AC, Young T. Showcasing metabolomic applications in aquaculture: a review. *Rev Aquacult.* 2018; 10:135–152.

231. Abdelrahman H, El Hady M, Alcivar-Warren A, *et al*. (2017) Aquaculture genomics, genetics and breeding in the United States: current status, challenges, and priorities for future research. *BMC Genomics* 2017; 18:191.

232. Germain PL, Ratti E, Boem F. Junk or functional DNA? ENCODE and the function controversy. *Biol Philos.* 2014; 29:807–831.

233. Saroglia M, Zhanjiang L. *Functional Genomics in Aquaculture*. John Wiley & Sons, Oxford; 2012.

234. Kim OTP, Nguyen PT, Shoguchi E, *et al*. A draft genome of the striped catfish, *Pangasianodon hypophthalmus*, for comparative analysis of genes relevant to development and a resource for aquaculture improvement. *BMC Genomics* 2018; 19:733.

235. Han C, Li Q, Xu J, Li X, Huang J. Characterization of *Clarias gariepinus* mitochondrial genome sequence and a comparative analysis with other catfishes. *Biologia (Poland)* 2015; 70:1245–1253.

236. Liu Z, Liu S, Yao J, *et al*. (2016) The channel catfish genome sequence provides insights into the evolution of scale formation in teleosts. *Nat Commun.* 2016; 7:11757.

237. Barman AS, Singh M, Pandey PK. Complete mitochondrial genome of near threatened butter catfish *Ompok bimaculatus* (Siluriformes: Siluridae). *Mitochondrial DNA B Resour*. 2017; 2:313–314.

238. Villela LCV, Alves AL, Varela ES, *et al*. Complete mitochondrial genome from South American catfish *Pseudoplatystoma reticulatum* (Eigenmann & Eigenmann) and its impact

in Siluriformes phylogenetic tree. *Genetica* 2017; 145:51–66.

239. Sahoo L, Kumar S, Das SP, *et al*. Complete mitochondrial genome sequence of *Heteropneustes fossilis* obtained by paired end next generation sequencing. *Mitochondrial DNA* 2016; 27:2485–2486.

240. Waldbieser GC, Bilodeau AL, Nonneman DJ. Complete sequence and characterization of the channel catfish mitochondrial genome. *DNA Seq.* 2003; 14:265–277.

241. Carvalho DC, Perini VDR, Bastos AS, *et al.* The complete mitochondrial genome of the threatened neotropical catfish *Lophiosilurus alexandri* (Silurifomes: Pseudopimelodidae) and phylogenomic analysis indicate monophyly of pimelodoidea. *Genet Mol Biol.* 2016; 39:674–677.

242. Moritz C. Applications of mitochondrial DNA analysis in conservation: a critical review. *Molecular Ecology* 1994; 3:401–411.

243. Shtolz N, Mishmar D. The mitochondrial genome–on selective constraints and signatures at the organism, cell, and single mitochondrion levels. *Front Ecol Evol.* 2019; 7:342.

244. Chen X, Zhong L, Bian C, *et al*. High-quality genome assembly of channel catfish, *Ictalurus punctatus*. *GigaScience* 2016; 5:39.

245. Zhang S, Li X, Chen X, Pan J, *et al*. Significant associations between prolactin gene polymorphisms and growth traits in the channel catfish (*Ictalurus punctatus* Rafinesque, 1818) core breeding population. *Meta Gene* 2019; 19:32–36.

246. Zeng Q, Fu Q, Li Y, Waldbieser G, *et al*. Development of a 690 K SNP array in catfish and its application for genetic mapping and validation of the reference genome sequence. *Sci Rep.* 2017; 7: 40347.

247. Wang S, Sha Z, Sonstegard TS, *et al*. Quality assessment parameters for EST-derived SNPs from catfish. *BMC Genomics* 2008; 9:450.

248. Sun L, Liu S, Wang R, *et al*. Identification and analysis of genome–wide SNPs provide

insight into signatures of selection and domestication in channel catfish (*Ictalurus punctatus*). *PLoS ONE* 2014; 9: e109666.249. Liu S, Wang X, Sun F, Zhang J, *et al*. Generation of genome-scale gene-associated SNPs in catfish for the construction of a high-density SNP array. *BMC Genomics* 2011; 12:53.

249. Carvalho DC, Perini VDR, Bastos AS, *et al.* The complete mitochondrial genome of the threatened Neotropical catfish *Lophiosilurus alexandri* (Silurifomes: Pseudopimelodidae) and phylogenomic analysis indicate monophyly of Pimelodoidea. *Genet Mol Biol.* 2016; 39:674–677.

250. Geng X, Liu S, Yao J, Bao L, *et al*. A genome-wide association study identifies multiple regions associated with head size in catfish. *G3* 2016; 6:3389–3398.

251. Bao L, Tian C, Liu S, Zhang Y, Elaswad A, Yuan Z, *et al.* The Y chromosome sequence of the channel catfish suggests novel sex determination mechanisms in teleost fish. *BMC Biol.* 2019; 7:1.

252. Ju Z, Dunham R, Liu Z. Transcriptome analysis of channel catfish (*Ictalurus punctatus*): Genes and expression profile from the brain. *Gene* 2000; 261:373–382.

253. Cao D, Kocabas A, Ju Z, *et al*. Transcriptome of channel catfish (*Ictalurus punctatus*): Initial analysis of genes and expression profiles of the head kidney. *Anim Genet.* 2001; 32:169–188.

254. Li P, Peatman E, Wang S, *et al*. Towards the ictalurid catfish transcriptome: Generation and analysis of 31,215 catfish ESTs. *BMC Genomics* 2007; 8:177.

255. Wang S, Peatman E, Abernathy J, *et al*. Assembly of 500,000 inter-specific catfish expressed sequence tags and large scale gene-associated marker development for whole genome association studies. *Genome Biol.* 2010; 11:R8.

256. Ju Z, Dunham R, Liu Z. Differential gene expression in the brain of channel catfish (*Ictalurus punctatus*) in response to cold acclimation. *Mol Genet Genom.* 2002; 268:87–

95.

257. Li RW, Waldbieser GC. Production and utilization of a high-density oligonucleotide microarray in channel catfish, *Ictalurus punctatus*. *BMC Genomics* 2006; 7:134.

258. Li C, Zhang Y, Wang R, *et al*. RNA-seq analysis of mucosal immune responses reveals signatures of intestinal barrier disruption and pathogen entry following *Edwardsiella ictaluri* infection in channel catfish, *Ictalurus punctatus*. *Fish Shellfish Immunol.* 2012; 32:816–827.

259. Peatman E, Baoprasertkul P, Terhune J, *et al*. (2007) Expression analysis of the acute phase response in channel catfish (*Ictalurus punctatus*) after infection with a Gram-negative bacterium. *Dev Comp Immunol.* 2007; 31:1183–1196.

260. Pridgeon JW, Yeh HY, Shoemaker CA, Klesius PH. Global transcription analysis of vaccinated channel catfish following challenge with virulent *Edwardsiella ictaluri*. *Vet Immunol Immunopathol.* 2012; 146:53–61.

261. Sun F, Peatman E, Li C, *et al*. Transcriptomic signatures of attachment, NF–κB suppression and IFN stimulation in the catfish gill following columnaris bacterial infection. *Dev Comp Immunol.* 2012; 38:169–180.

262. Liu S, Zhou Z, Lu J, *et al*. RNA-Seq reveals expression signatures of genes involved in oxygen transport, protein synthesis, folding, and degradation in response to heat stress in catfish. *Physiol Genomics* 2013; 45:462–476.

263. Myers JN, Dyce PW, Chatakondi NG, *et al*. (2020). Analysis of specific mRNA gene expression profiles as markers of egg and embryo quality for hybrid catfish aquaculture. *Comp Biochem Physiol.* 2020; 243A:110675.

264. Lozada-Chávez I, Stadler PF, Prohaska SJ. Hypothesis for the modern RNA world: a pervasive non-coding RNA-based genetic regulation is a prerequisite for the emergence of multicellular complexity. *Orig Life Evol Biosph.* 2011; 41:587–607.

265. Barozai K. The microRNAs and their targets in the channel catfish (*Ictalurus punctatus*). *Mol Biol Rep.* 2012; 39:8867–8872.

266. Xu Z, Qin Q, Ge J, Pan J, Xu X. Bioinformatic identification and validation of conservative microRNAs in *Ictalurus punctatus*. *Mol Biol Rep.* 2012; 39:10395–10405.

267. Xu Z, Chen J, Li X, Ge J, Pan J, Xu X. Identification and characterization of microRNAs in channel catfish (*Ictalurus punctatus*) by using Solexa sequencing technology. *PLoS ONE* 2013; 8:e54174.

268. Riesco MF, Valcarce, DG, Martínez-Vázquez, JM, Robles V. Effect of low sperm quality on progeny: a study on zebrafish as model species. *Sci Rep.* 2019; 9: 1–10.

269. Coyne VE. Proteomics: applications and advances. In: Kim SK ed. *Springer Handbook of Marine Biotechnology.* Springer, Berlin; 2015:475–495.

270. Desai MA, Joseph P, Suman SP, Silva JL, Kim T, Schilling MW. Proteome basis of red color defect in channel catfish (*Ictalurus punctatus*) fillets. *LWT - Food Sci Technol.* 2014; 57:141–148.

271. Ciaramella MA, Nair MN, Suman SP, Allen PJ, Schilling MW. Differential abundance of muscle proteome in cultured channel catfish (*Ictalurus punctatus*) subjected to ante-mortem stressors and its impact on fillet quality. *Comp Biochem Physiol.* 2016; 20D:10–18.

272. Schmitz M, Mandiki SNM, Douxfils J, Ziv T, Admon A, Kestemont P. Synergic stress in striped catfish (*Pangasianodon hypophthalmus*, S.) exposed to chronic salinity and bacterial infection: Effects on kidney protein expression profile. *J Proteom.* 2016; 142:91–101.

273. Sellegounder D, Gupta YR, Murugananthkumar R, Senthilkumaran B. Enterotoxic effects of *Aeromonas hydrophila* infection in the catfish, *Clarias gariepinus*: Biochemical, histological and proteome analyses. *Vet Immunol Immunopathol.* 2018; 204:1–10.

274. Allen PJ, Wise D, Greenway T, Khoo L, Griffin MJ, Jablonsky M. Using 1–D 1H and 2–D 1H J–resolved NMR metabolomics to understand the effects of anemia in channel catfish (*Ictalurus punctatus*). *Metabolomics* 2015; 11:1131–1143.

275. Du H, Fu J, Wang S, *et al*. 1H-NMR metabolomics analysis of nutritional components from two kinds of freshwater fish brain extracts. *RSC Advances* 2018; 8:19470–19478.

276. Bledsoe JW, Waldbieser GC, Swanson KS, Peterson BC, Small BC. Comparison of channel catfish and blue catfish gut microbiota assemblages shows minimal effects of host genetics on microbial structure and inferred function. *Front Microbiol.* 2018; 9:1073.

277. Mohammed HH, Arias CR. Potassium permanganate elicits a shift of the external fish microbiome and increases host susceptibility to columnaris disease. *Vet Res.* 2015; 46:82.

278. Wang E, Yuan Z, Wang K, Gao D, Liu Z, Liles MR. Consumption of florfenicol-medicated feed alters the composition of the channel catfish intestinal microbiota including enriching the relative abundance of opportunistic pathogens. *Aquaculture* 2019; 501:111–118.

279. Zhang Z, Li D, Refaey MM, Xu W. High spatial and temporal variations of microbial community along the southern catfish gastrointestinal tract: Insights into dynamic food digestion. *Front Microbiol.* 2017; 8:1531.

280. Burgos FA, Ray CL, Arias CR. Bacterial diversity and community structure of the intestinal microbiome of Channel catfish (*Ictalurus punctatus*) during ontogenesis. *Syst. Appl. Microbiol.* 2018; 41:494–505.

281. Abdul-Razak S, Griffin MJ, Mischke CC, *et al*. Biotic and abiotic factors influencing channel catfish egg and gut microbiome dynamics during early life stages. *Aquaculture* 2019; 498:556–567.

282. Minich JJ, Zhu Q, Xu ZZ, *et al*. Microbial effects of livestock manure fertilization on freshwater aquaculture ponds rearing tilapia (*Oreochromis shiranus*) and North African

2367 catfish (*Clarias gariepinus*). *Microbiology Open* 2018; 7:e716.

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Table 1. Worldwide catfish production and by continent in terms of production (t) and economic value (USD) in 2018. Data is presented in terms of family taxonomical level. Geographical units are ordered in terms of catfish production relevance. Data per geographical region expressed as production and value percentages were calculated with relation to the region and not to overall worldwide values. Data were retrieved from FAO [9].

	Production (t)	Production (%)	Value (USD 000)	Value (%)
World	5,781,235.1	100	9,489,861.120	100
Asia	**5,333,194.55**	**92.25**	**8,318,614.530**	**87.66**
Bagridae; FW	53,7957.80	10.1	1,291,932.150	15.53
Clariidae; FW	1,352,494.05	25.4	1,853,994.520	22.29
Heteropneustidae; FW	13,793.76	0.3	73,476.310	0.88
Siluridae; FW	372,438.47	7.0	887,809.870	6.57
Pangasiidae; BW	...	…	…	…
Pangasiidae; FW	2,826,068.47	53.0	3,664,792.260	44.06
Ictaluridae; FW	230,442.00	4.3	546,608.420	10.67
Others; FW	...	...	…	…
Africa	**251,332.47**	**4.35**	**731,432.060**	**7.71**
Bagridae; BW	50.00	0.02	179.950	0.03
Bagridae; FW	2.00	0.001	2.250	<0.001
Clariidae; BW	1,836.00	0.73	1,343.370	0.18
Clariidae; FW	244,890.47	97.44	719,508.560	98.37
Siluridae; FW	44.00	0.02	24.930	<0.001
Mochokidae; FW	4,510.00	1.79	10,3730	1.42
Others; BW	...	…	…	…
North America	**168,579.08**	**2.92**	**355,880.850**	**3.75**
Callichthyidae; FW	2.00	0.001	6.000	0.002
Clariidae; FW	6,286.00	3.73	6,286.000	1.77
Ictaluridae; FW	161,271.08	95.66	34,6213.170	97.28
Pangasiidae; FW	1,020.00	0.61	3,375.680	0.95
South America	**14,642.01**	**0.25**	**44,973.760**	**0.47**
Clariidae; FW	…	…	…	…
Callichthyidae; BW	19.92	0.14	95.9	0.21
Callichthyidae; FW	...	….	….	…
Ictaluridae; FW	...	….	….	…
Loricariidae; FW	6.15	0.04	18.71	0.04
Pimelodidae; FW	665.94	4.55	4,771.03	10.61
Others; FW	13,950.00	95.27	40,088.12	89.14
Europe	**13,486.44**	**0.23**	**38,960.920**	**0.41**
Clariidae, FW	10,022.08	74.3	25,478.25	65.39
Ictaluridae; BW	...	…	…	
Ictaluridae; FW	2,039.69	15.1	6,559.81	16.84
Siluridae; FW	1,424.67	10.6	6,922.86	17.77
Others; FW	...	...	…	
Oceania	…	…	…	…
Pangasiidae	…	…	…	…
Clariidae	…	…	…	…

Abbreviations: FW, freshwater; BW, brackish water; "..." = Data not available; unobtainable; data not separately available but included in another category [9]. "Others" refer to species classified as Siluriformes (catfish), but not further taxonomically identified [9].

Table 2. Comparison of the main developmental events of the digestive system ontogeny between the catfish species presented in this review. For comparative purposes among catfish species, larval development was scaled using thermal units (cumulative degree-days post hatch). This unit is calculated as the average temperature (ºC) over the period of development and it is the product of the value of the average temperature multiplied by the number of days.

	Developmental events						
Catfish species	Mouth opening	First feeding	Yolk-sac resorption	Intestine differentiation	Pancreas differentiation	Zymogen granules in pancreas	Fully formed stomach
P. hypophthalmus [56]	52	52	104	-	-	-	-
C. gariepinus [57]	50	55	114	27	29	-	114
I. punctatus [58]	-	294-336*	210-231*	-	-	-	-
P. punctifer [59]	56	112	168	112	28	28	252
H. fossilis [60]	29	58	145	58	58	87	290
O. bimaculatus [61]	54	54	135	54	27	27	297
R. quelen [62,63]	4	49	74	72	17	39	49
L. alexandri [64,65]	0	162	270	189	108	-	288

* Data retrieved from natural populations not from aquaculture studies.

Table 3. Summary of rearing practices for *Pangasiodon hypoththalmus* and *Clarias gariepinus* during early life stages.

	P. hypoththalmus		*C. gariepinus*	
	Intensive	**Extensive**	**Intensive**	**Extensive**
Rearing system	Hatchery tanks ($0.2–4.7$ m^3)	Earthen ponds ($1{,}000–5{,}000$ m^2)	Hatchery tanks (100–1,000 L)	Earthen ponds ($100–250$ m^2)
Treatment	Surface chlorinated water, open-flow	Pond liming and fertilization	Surface water, open-flow, RAS	Pond liming and fertilization
Food	Natural zooplankton (cladocerans, rotifers), *Artemia*, compound feeds	Natural zooplankton (cladocerans), *Artemia*, *Tubifex* sp., custard egg and soya powder, compound feeds	*Artemia*, cladocerans	Natural zooplankton (cladocerans, copepods, rotifers), crumbled formulated feeds
Water quality				
Temperature	26–28°C	26–32°C	28°C	28–32°C
pH	7.4–7.5	6.4–8.5	7.0	6.0–9.0
Oxygen	≥5 mg L^{-1}	≥3 mg L^{-1}	≥5 mg L^{-1}	≥3 mg L^{-1}
Fish density	10,000 fish m^{-3}	500–800 fish m^{-2}	6 larvae L^{-1}	100–250 larvae m^{-2}
Stocking age	1-2 dph (6.2 mm TL)	1-2 dph (6.2 mm TL)	1 dph (3.5-4.0 mm TL)	3 dph (4.8-5.0 mmTL)

Abbreviations: dph, days post hatching; RAS, recirculating aquatic system; TL, total length.

Table 4. Different weaning protocols recommended for *Clarias garepinus*.

Protocol / Reference	Days post hatching															
	1	2	3	4	5	6	7	8	9	10	11	12	13	14	15	16	17-32
Janssen [98]																	
Artemia																	
Compound diet																	
Verreth *et al.* [99]																	
Artemia																	
Weaning compound diet																	
Hecht *et al.* [100]																	
Live prey																	
Weaning compound diet																	
Compound diet																	
Oellermann [101]																	
Artemia																	
Weaning compound diet																	
Compound diet																	
Chepkirui-Boit *et al.* [102]																	
Artemia																	
Weaning compound diet																	

Details of feeding protocols: [1]*Artemia* nauplii was distributed *ad libitum*; the grow-out feed was supplemented with wheat bran; [2]*Artemia* nauplii was distributed four times per day; *Artemia* nauplii were progressively replaced by the growth-out feed (commercial trout pelleted feed) from 10 to 15 days four times per day; the similar protocol may be used but using *Artemia* dry cysts instead of nauplii [97]; [3]*Artemia* nauplii or live prey (*Daphnia* sp.) was distributed once per day; the replacement of the live feed by the compound diet was progressive and weaning diet should have 38-40% crude protein; [4]*Artemia* nauplii was distributed four times per day; weaning diet is described in Uys & Hecht [103]; [5]the experiment only lasted until 21 days post hatching; *Artemia* nauplii were distributed four to six times per day; the weaning diet contained the freshwater atyid shrimp (*Caridina nilotica*) at 75.5%; during weaning, *Artemia* nauplii and the dry feed were administered at equal parts.

Table 5. Summary of rearing practices for *Ictalurus punctatus* and *Pseudoplatystoma spp.* during early life stages.

	I. punctatus		*Pseudoplatystoma* spp.	
	Intensive	**Extensive**	**Intensive**	**Extensive**
Rearing system	Rectangular troughs (380–450 L)	Earthen ponds (4,000–20,000 m^2)	Cylindroconical tanks (60–200 L) / Rectangular or circular tanks	Earthen ponds
Water treatment	Well or surface water, open-flow, RAS	Pond liming and fertilization (organic and inorganic)	Well or surface water, RAS	Pond liming and fertilization (organic and inorganic)
Food	Starter diets; *Artemia* decapsulated cysts or zooplankton supplementation	Large cladocerans	*Artemia* from 2 to 12 dph, then cladocerans and copepods, optionally minced fish or meat	Cladocerans and copepods, optionally forage fish
Water quality				
Temperature	25.5–27.5ºC	26.0–30.0ºC	26–28°C	26–28°C
pH	7.0-8.5	7.0-8.5	*ca.* 7.0	*ca.* 7.0
Oxygen	≥4 ppm	≥3-4 ppm	≥6 ppm	≥6 ppm
Fish density	150,000–200,000 fry $trough^{-1}$	12–50 fry m^{-2}	15–50 larvae L^{-1} / 5,000–10,000 larvae m^{-3}	100–150 larvae m^{-2}
Stocking age	2 dph (14.4–18.8 mg BW)	2 dph (14.4–18.8 mg BW) – 7 dph (22.8–29.1 mg BW)	1 dph (< 3 mm TL) / 12 dph (13–15 mm TL)	12 dph (13–15 mm TL)

Abbreviations: BW, body weight; dph, days post hatching; RAS, recirculating aquatic system; TL, total length.

Table 6. Summary of rearing practices for *Heteropneustes fossilis* and *Rhamdia quelen* during early life stages.

	H. fossilis		*R. quelen*	
	Intensive	**Extensive**	**Intensive**	**Extensive**
Rearing system	FRP or concrete tanks (30 m^2)	Earthen ponds (100 - 400 m^2)	Indoor tank	Earthen ponds (up to 300-400 m^2)
Water treatment	Well or surface water, open-flow	Liming and fertilization (organic and inorganic)	Well or surface water, open-flow, RAS	Liming and fertilization (organic and inorganic)
Food	Zooplankton, rotifers, ciliates *Artemia* nauplii, egg custard, snail meat, fish meat, rice bran and commercial starter feed / microdiet	Zooplankton (ostracods, cladocerans, rotifers, copepod nauplii), *Tubifex* sp., finely ground trash fish, rice bran, mustard oil cake and chopped mollusc meat	Live prey (*Artemia* nauplii or pond-collected zooplankton) alone or in combination with dry feeds	Natural zooplankton (ostracods, chironomid larvae, cladocerans and copepods); natural zooplankton plus compound diet; bioflocs
Water quality				
Temperature	28.0–29.1ºC	26.0–29.0ºC	21.0–26.0ºC	17.0–27.0ºC
pH	6.8–7.6	7.2–7.6	8.0–8.5	8.0–8.5
Oxygen	6–8 mg L^{-1}	5.3–5.8 mg L^{-1}	6–8 mg L^{-1}	6–8 mg L^{-1}
Hardness	-	-	-	20–70 mg $CaCO_3$ L^{-1}
Fish density	3,000–5,000 larvae m^{-2}	300–500 larvae m^{-2}	10 fish L^{-1}	Up to 200 fish m^{-2} / 25 fry L^{-1}
Stocking age	1 dph (3 mm TL)	12 dph (10–12 mm TL)	2 dph (5 mm TL)	2 dph / 8-10 dph (7.5-8 mm TL)

Abbreviations: BW, body weight; dph, days post hatching; FRP, fibre-reinforced plastic; RAS, recirculating aquatic system; TL, total length.

Table 7. Summary of rearing practices for *Ompok bimaculatus* and *Lophiosilurus alexandri* during early life stages.

	O. bimaculatus		*L. alexandri*	
	Intensive	**Extensive**	**Intensive**	**Extensive**
Rearing system	Cement cistern, FRP tanks (4 m^2)	Earthen ponds (100–400 m^2)	Hatchery tanks (10–100 L)	-
Water treatment	Well or surface water, open-flow	Liming and fertilization (organic, and inorganic)	Well or surface water, open-flow, RAS	-
Food	*Artemia* nauplii, pond-collected zooplankton (copepods, cladocerans), *Tubifex*, trash fish, formulated diets, chicken viscera	Zooplankton (copepods, cladocerans), rice bran, mustard oil cake, and dry fish powder	*Artemia*	-
Water quality				
Temperature	27.0–28.1°C	27–28°C	26–32ºC	
pH	6.8–7.6	7.2–7.7	6.5–8.5	
Oxygen	6–8 mg L^{-1}	5–6 mg L^{-1}	>4 mg L^{-1}	
Hardness	-	-	2 g NaCl L^{-1}	-
Fish density	3,000–4,000 larvae m^{-2}	100–200 larvae m^{-2}	Up to 300 larvae L^{-1}	-
Stocking age	2 dph (3.3 mm TL)	2–3 dph (3–4 mm TL)	1 dph (8 mm TL)	-

Abbreviations: dph, days post hatching; FRP, fibre-reinforced plastic; RAS, recirculating aquatic system.

Table 8. Weaning protocol recommended for *Lophiosilurus alexandri* onto compound diets.

Days of feeding	Feeding protocol[1]
1-3 days	80% OH + 20% CD + 10 g *Artemia* nauplii
4-6 days	60% OH + 40% CD + 10 g *Artemia* nauplii
7-9 days	40% OH + 60% CD + 5 g *Artemia* nauplii
10-12 days	20% OH + 80% CD
13-15 days	100% CD

[1] Ingredient percentages for the weaning protocol are indicated considering the preparation of 100g feed. *Abbreviations*: OH, ox heart; CD, compound diet.

Supplementary file 1. Catfish production (t) per country, species and environment in 2018. Data retrieved from FAO [9].

ASIA

Country (Country)	Catfish species	Environment	Production (t)	
Azerbaijan	*Silurus glanis*	Freshwater	...	
Bangladesh	*Wallago attu*	Freshwater	1,022	
Bangladesh	*Heteropneustes fossilis*	Freshwater		
Bangladesh	*Clarias batrachus*	Freshwater	13,969	
Bangladesh	*Pangasianodon hypophthalmus*	Freshwater	441,929	
Brunei Darussalam	*Pangasius spp*	Freshwater	...	
Brunei Darussalam	*Clarias spp*	Freshwater	...	
Cambodia	*Siluroidei*	Freshwater	...	
Cambodia	*Clarias batrachus*	Freshwater	...	
Cambodia	*Clarias spp*	Freshwater	6,000	
Cambodia	*Pangasius spp*	Freshwater	75,850	
China	*Leiocassis longirostris*	Freshwater	21,610	
China	*Ictalurus punctatus*	Freshwater	230,442	
China	*Silurus asotus*	Freshwater	365,590	f
China	*Pelteobagrus fulvidraco*	Freshwater	509,610	
China, Hong Kong SAR	*Clarias fuscus*	Freshwater	0	f
Georgia	*Silurus glanis*	Freshwater	5	
Georgia	*Clarias gariepinus*	Freshwater	8	f
India	*Clarias spp*	Freshwater	114,000	
India	*Pangasianodon hypophthalmus*	Freshwater	523,000	f
Indonesia	*Hemibagrus nemurus*	Freshwater	4,811	
Indonesia	*Pangasius spp*	Freshwater	373,263	f
Indonesia	*Clarias spp*	Freshwater	1,027,195	f
Kazakhstan	*Silurus glanis*	Freshwater	...	f
Korea, Dem. People's Rep	*Clarias gariepinus*	Freshwater	7,000	
Korea, Republic of	*Ictalurus punctatus*	Freshwater	0	
Korea, Republic of	*Bagridae*	Freshwater	400	f
Korea, Republic of	*Silurus asotus*	Freshwater	4,800	
Lebanon	*Clarias gariepinus*	Freshwater	...	f
Malaysia	*Wallago spp*	Freshwater	0	
Malaysia	*Hemibagrus nemurus*	Freshwater	1,526.8	f
Malaysia	*Pangasius pangasius*	Freshwater	18,454	f
Malaysia	*Clarias spp*	Freshwater	33,420	f
Myanmar	*Heteropneustes fossilis*	Freshwater	372.76	
Myanmar	*Clarias spp*	Freshwater	10,000	
Myanmar	*Pangasianodon hypophthalmus*	Freshwater	18,920	
Nepal	*Pangasianodon hypophthalmus*	Freshwater	750	f
Nepal	*Clarias gariepinus*	Freshwater	1,800	
Philippines	*Clarias spp*	Freshwater	4,397.8	

Saudi Arabia	*Clarias gariepinus*	Freshwater	100	
Singapore	*Clarias batrachus*	Freshwater	3.25	f
Singapore	*Pangasianodon hypophthalmus*	Freshwater	10.47	
Sri Lanka	*Pangasianodon hypophthalmus*	Freshwater	3	f
Syrian Arab Republic	*Clarias gariepinus*	Freshwater	500	
Taiwan Province of China	*Silurus asotus*	Freshwater	469.48	
Thailand	*Pangasianodon hypophthalmus*	Freshwater	13,889	
Thailand	*Clarias gariepinus x C. macrocephalus*	Freshwater	112,101	
Turkey	*Silurus glanis*	Freshwater	5	
Uzbekistan	*Silurus glanis*	Freshwater	547	f
Viet Nam	*Clarias spp*	Freshwater	22,000	
Viet Nam	*Pangasianodon hypophthalmus*	Freshwater	13,60,000	

AFRICA

Country (Country)	Catfish species	Environment	Production (t)	
Algeria	*Clarias gariepinus*	Freshwater	310.5	
Angola	*Clarias spp*	Freshwater	0	
Benin	*Chrysichthys spp*	Brackishwater	...	
Benin	*Clarias gariepinus*	Freshwater	2,310	
Burkina Faso	*Clarias anguillaris*	Freshwater	0	
Burkina Faso	*Clarias gariepinus*	Freshwater	119	
Burundi	*Clarias gariepinus*	Freshwater	80	f
Cameroon	*Heterobranchus longifilis*	Freshwater	7	f
Cameroon	*Clarias gariepinus*	Freshwater	1,150	f
Central African Republic	*Clarias gariepinus*	Freshwater	10	f
Chad	*Bagrus bajad*	Freshwater	...	
Chad	*Clarias gariepinus*	Freshwater	150	
Congo	*Clarias gariepinus*	Freshwater	5	f
Congo, Dem. Rep. of the	*Clarias gariepinus*	Freshwater	15	f
Côte d'Ivorie	*Siluroidei*	Brackishwater	...	
Côte d'Ivorie	*Chrysichthys spp*	Brackishwater	0	
Côte d'Ivorie	*Clarias spp*	Freshwater	...	
Côte d'Ivorie	*Clarias spp*	Brackishwater	...	
Côte d'Ivorie	*Chrysichthys nigrodigitatus*	Brackishwater	50	f
Côte d'Ivorie	*Clarias gariepinus*	Freshwater	180	f
Egypt	*Bagrus bajad*	Freshwater	2	
Egypt	*Clarias gariepinus*	Brackishwater	1,836	f
Egypt	*Clarias gariepinus*	Freshwater	5,000	f
Equatorial Guinea	*Clarias gariepinus*	Freshwater	2	f
Eswatini	*Clarias gariepinus*	Freshwater	...	
Gabon	*Clarias gariepinus*	Freshwater	5	f
Gambia	*Clarias spp*	Freshwater	2	f
Ghana	*Clarias gariepinus*	Freshwater	4,657	f
Guinea	*Clarias gariepinus*	Freshwater	58.6	

Kenya	*Clarias gariepinus*	Freshwater	1,960	f
Lesotho	*Clarias gariepinus*	Freshwater	...	
Liberia	*Heterobranchus bidorsalis*	Freshwater	...	
Liberia	*Heterobranchus longifilis*	Freshwater	1	f
Liberia	*Clarias gariepinus*	Freshwater	14	f
Malawi	*Clarias gariepinus*	Freshwater	364	
Mali	*Clarias gariepinus*	Freshwater	392	
Namibia	*Clarias gariepinus*	Freshwater	6.19	
Niger	*Clarias gariepinus*	Freshwater	120	
Nigeria	*Chrysichthys nigrodigitatus*	Brackishwater	0	
Nigeria	*Chrysichthys nigrodigitatus*	Freshwater	...	
Nigeria	*Bagrus spp*	Freshwater	...	
Nigeria	*Clarias spp*	Brackishwater	...	
Nigeria	*Synodontis spp*	Freshwater	4,510	
Nigeria	*Clarias spp*	Freshwater	28,227	
Nigeria	*Clarias gariepinus*	Freshwater	160,114	
Rwanda	*Clarias gariepinus*	Freshwater	300	f
Senegal	*Clarias spp*	Freshwater	...	
Senegal	*Clarias gariepinus*	Freshwater	25	
Sierra Leone	*Clarias gariepinus*	Freshwater	5	f
South Africa	*Clarias gariepinus*	Freshwater	20	
Sudan	*Clarias gariepinus*	Freshwater	2,000	
Sudan (former)	*Bagrus bajad*	Freshwater	...	
Tanzania, United Rep. of	*Clarias gariepinus*	Freshwater	3,800	
Togo	*Clarias spp*	Freshwater	12	
Tunisia	*Silurus glanis*	Freshwater	44	
Uganda	*Clarias gariepinus*	Freshwater	33,454	
Zambia	*Clarias gariepinus*	Freshwater	10	f
Zimbabwe	*Clarias gariepinus*	Freshwater	5	f

NORTH AMERICA

Country (Country)	Catfish species	Environment	Production (t)	
Costa Rica	*Ictalurus punctatus*	Freshwater	...	
Cuba	*Ictalurus punctatus*	Freshwater	...	
Cuba	*Clarias gariepinus*	Freshwater	6,286	
Dominican Republic	*Pangasianodon hypophthalmus*	Freshwater	550	f
Guatemala	*Ictalurus punctatus*	Freshwater	...	
Haiti	*Pangasianodon hypophthalmus*	Freshwater	70	f
Jamaica	*Pangasianodon hypophthalmus*	Freshwater	399	
Mexico	*Rhamdia quelen*	Freshwater	0	
Mexico	*Ictalurus punctatus*	Freshwater	634.68	
Mexico	*Ictalurus spp*	Freshwater	1,213.4	
Puerto Rico	*Ictalurus punctatus*	Freshwater	...	
Puerto Rico	*Pangasianodon hypophthalmus*	Freshwater	1	f
Trinidad and Tobago	*Hoplosternum littorale*	Freshwater	2	
United States of America	*Ictalurus punctatus*	Freshwater	159,423	

SOUTH AMERICA

Country (Country)	Catfish species	Environment	Production (t)	
Argentina	*Rhamdia quelen*	Freshwater	...	
Argentina	*Pseudoplatystoma* spp.	Freshwater	79.34	*
Brazil	*Ictalurus punctatus*	Freshwater	...	
Brazil	*Clarias gariepinus*	Freshwater	...	
Brazil	*Pseudoplatystoma corruscans*	Freshwater	...	
Brazil	*Rhamdia quelen*	Freshwater	...	
Brazil	*Hypostomus plecostomus*	Freshwater	...	
Brazil	*Siluroidei*	Freshwater	13,950	f
Colombia	*Pseudoplatystoma fasciatum*	Freshwater	...	
Colombia	*Sorubim lima*	Freshwater	...	
Colombia	*Pimelodus* spp.	Freshwater	0.6	
French Guiana	*Hoplosternum littorale*	Freshwater	...	
Guyana	*Hoplosternum littorale*	Freshwater	...	
Guyana	*Hoplosternum littorale*	Brackishwater	19.92	
Paraguay	*Ictalurus punctatus*	Freshwater	...	
Paraguay	*Pseudoplatystoma corruscans*	Freshwater	580	
Peru	*Pseudoplatystoma spp*	Freshwater	...	
Peru	*Siluroidei*	Freshwater	...	
Peru	*Pterygoplichthys pardalis*	Freshwater	6.15	
Suriname	*Hoplosternum littorale*	Freshwater	...	
Uruguay	*Rhamdia quelen*	Freshwater	6	f
Venezuela	*Pseudoplatystoma fasciatum*	Freshwater	...	
Venezuela	*Siluroidei*	Freshwater	...	

EUROPE

Country (Country)	Catfish species	Environment	Production (t)	
Austria	*Ictalurus* spp.	Freshwater	...	
Austria	*Silurus glanis*	Freshwater	5	
Austria	*Clarias gariepinus*	Freshwater	421	
Belarus	*Clarias gariepinus*	Freshwater	2	
Belarus	*Silurus glanis*	Freshwater	18	
Belgium	*Clarias gariepinus*	Freshwater	...	
Bosnia and Herzegovina	*Silurus glanis*	Freshwater	0	
Bulgaria	*Ictalurus punctatus*	Freshwater	23	
Bulgaria	*Silurus glanis*	Freshwater	246	
Bulgaria	*Clarias gariepinus*	Freshwater	281	
Croatia	*Clarias gariepinus*	Freshwater	20	
Croatia	*Silurus glanis*	Freshwater	23	
Czechia	*Silurus glanis*	Freshwater	91	
Denmark	*Siluroidei*	Freshwater	...	
France	*Silurus glanis*	Freshwater	200	f
Germany	*Silurus glanis*	Freshwater	110	

Supplementary file 2.

Table S1. Species of the genus *Pseudoplatystoma* before and after the taxonomic revision of Buitrago-Suárez & Burr [18] and their geographic distribution.

Species		**Geographic distribution**	
Before	**After**	**River basins**	**Countries**
P. fasciatum	*P. punctifer*	Amazon	Bolivia, Brazil, Colombia, Ecuador, Peru, Venezuela
	P. reticulatum	Central Amazon, Paraná	Argentina, Bolivia, Brazil, Paraguay, Uruguay
	P. orinocoense	Orinoco	Venezuela
	P. fasciatum	Guyana region	Guyana, Suriname, French Guiana
	P. magdaleniatum	Magdalena, Cauca	Colombia
P. tigrinum	*P. tigrinum*	Amazon	Brazil, Colombia, Ecuador, Peru, Venezuela
	P. metaense	Orinoco	Colombia, Venezuela
P. corruscans	*P. corruscans*	Paraná, São Francisco	Argentina, Brazil, Paraguay, Uruguay

Buitrago–Suárez UA, Burr BM. Taxonomy of the catfish genus *Pseudoplatystoma* Bleeker (Siluriformes: Pimelodidae) with recognition of eight species. *Zootaxa* 2007; 1512:1-38.

Table S2. *Pseudoplatystoma* hybrids in South American aquaculture.

Parent species ♀	♂	**Characteristics**	**References**
P. reticulatum	*P. corruscans*	Longer spawning period of *P. reticulatum* ♀; better growth performance; hardiness; hybrids and backcrosses are fertile.	1, 2, 3
P. corruscans	*P. reticulatum*	Better growth performance; hardiness; hybrids and backcrosses are fertile.	3
P. reticulatum	*Leiarius marmoratus*	Better acceptance of formulated feed; omnivorous; lower cannibalism; faster growth rate; unknown fertility.	1, 3
P. corruscans	*Leiarius marmoratus*	Better acceptance of formulated feed; omnivorous; lower cannibalism; faster growth rate; unknown fertility.	3
P. punctifer	*Leiarius marmoratus*	Better acceptance of formulated feed; omnivorous; lower cannibalism; faster growth rate; unknown fertility.	3
P. metaense	*Leiarius marmoratus*	np	4
P. reticulatum	*Phractocephalus hemioliopterus*	Prized as ornamental and sport fishing species; good growth rate; flesh quality; unknown fertility.	1
P. corruscans	*Phractocephalus hemioliopterus*	np	5
P. reticulatum x P. corruscans	*P. reticulatum*	np	5
P. reticulatum x P. corruscans	*P. corruscans*	np	5
P. reticulatum x P. corruscans	*P. reticulatum x P. corruscans*	np	5
P. reticulatum x P. corruscans	*Leiarius marmoratus*	np	5
P. reticulatum x P. corruscans	*Phractocephalus hemioliopterus*	np	5
Phractocephalus hemioliopterus	*P. reticulatum x P. corruscans*	np	5

Abbreviation : np, data not provided.

References

1 Hashimoto DT, Senhorini JA, Foresti F, Porto-Foresti F (2012) Interspecific fish hybrids in Brazil: management of genetic resources for sustainable use. *Reviews in Aquaculture* **4**: 108-118.

2 Campos JL (2013) O cultivo do pintado, *Pseudoplatystoma corruscans* (Spix e Agassiz, 1829), outras espécies do gênero *Pseudoplatystoma* e seus híbridos. In: Baldisserotto B, Gomes LC (eds.) *Espécies nativas para piscicultura no Brasil*, 2nd edn, pp. 335-361. Editora UFSM, Santa Maria, Brazil.

3 Alves AL, Varela ES, Moro GV, Kirschnik LNG (2014) *Riscos genéticos da produção de híbridos de peixes nativos*. Embrapa Pesca e Aquicultura, Palmas, Brazil.

4 Porras-Rivera G, Rodríguez-Pulido JA (2019) Comparación y Caracterización Morfométrica del Híbrido (*Pseudoplatystoma metaense* x *Leiarius marmoratus*) y sus Parentales (Siluriformes: Pimelodidae). *International Journal of Morphology* **37**: 1409-1415.

5 Hashimoto DT, Prado FD, Senhorini JA, Foresti F, Porto-Foresti F (2015) Aquaculture of neotropical catfish hybrids: genetic strategies for conservation and management. In: Regan B (ed.) *Carp and Catfish: Biology, Behavior and Conservation Strategies*, pp. 1-30. Nova Science Publishers, New York.

Supplementary file 3. List of references in the literature and the amount of genomic, transcript, protein and non-coding resources available for each catfish species considered in this review.

Species	PubMed Central[#]	Nuclear Genome					Mitochondrial genome	
		Assembly	BioProject	BioSample	SRA	Genome	Known	Reference
P. hypophthalmus	201	2	9	31	65	1[a]	Yes	1
C. gariepinus	1,010		6	52	71		Yes	2
I. punctatus	2,299	2	60	417	372	1[b]	Yes	3
P. punctifer	9						No	
H. fossilis	272		3	2	2		Yes	4, 5
R. quelen	248						No	
O. bimaculatus	24	1	2	1		1[c]	Yes	6
L. alexandri	22		2	1				

Species	Genes	Transcripts	Proteins		
			NCBI	Uniprot	
				Reviewed	Unreviewed
P. hypophthalmus	27,514	50,954	66,920		21,421
C. gariepinus	37	16,694	2,901	5	288
I. punctatus	27,984	892,266	125,991	89	43,724
P. punctifer		231	91		17
H. fossilis	13	508	381	2	196
R. quelen		474	452		190
O. bimaculatus	37	393	354		93
L. alexandri	37	99	45		22

Species	Non-coding RNAs				
	tRNA	rRNA	miRNA	snoRNA	snRNA
P. hypophthalmus	1,713	39			
C. gariepinus	41	86			
I. punctatus	37	1,058	204	176	115
P. punctifer					
H. fossilis	24	24			
R. quelen	2	2			
O. bimaculatus	22	17			
L. alexandri	28	6			

Data retrieved the 27 of June 2020 from www.ncbi.nlm.nih.gov, www.ensembl.org, https://www.uniprot.org/ and https://rnacentral.org/ databases.

[a] Institute of Genome Research, Vietnam Academy of Science and Technology, 2018; [b] Auburn University, 2016; [c] All India Institute of Medical Sciences (AIIMS), 2019.

References:

[1] Kim OTP, Nguyen PT, Shoguchi E, Hisata K, Vo TTB, Inoue J *et al*. A draft genome of the striped catfish, *Pangasianodon hypophthalmus*, for comparative analysis of genes relevant to development and a resource for aquaculture improvement. *BMC Genomics* 2018; 19:733.

[2] Han C, Li Q, Xu J, Li X, Huang J. Characterization of *Clarias gariepinus* mitochondrial genome sequence and a comparative analysis with other catfishes. *Biologia (Poland)* 2015; 70:1245–1253.

[3] Waldbieser GC, Bilodeau AL, Nonneman DJ. Complete sequence and characterization of the channel catfish mitochondrial genome. *DNA Sequence - Journal of DNA Sequencing and Mapping* 2003; 14:265–277.

[4] Sahoo L, Kumar S, Das SP, Patnaik S, Bit A, Sundaray JK *et al*. Complete mitochondrial genome sequence of *Heteropneustes fossilis* obtained by paired end next generation sequencing. *Mitochondrial DNA* 2016; 27:2485–2486.

[5] Behera BK, Baisvar VS, Kumari K, Rout AK, Pakrashi S, Paria P *et al*. The complete mitochondrial genome of the Asian stinging catfish, *Heteropneustes fossilis* (Siluriformes, Heteropneustidae) and its comparison with other related fish species. *Mitochondrial DNA Part B: Resources* 2016; 1:804–805.

[6] Barman AS, Singh M, Pandey PK. Complete mitochondrial genome of near threatened butter Catfish

Ompok bimaculatus (Siluriformes: Siluridae). *Mitochondrial DNA Part B: Resources* 2017; 2:313–314.

[7] Carvalho DC, Perini VDR, Bastos AS, Costa IRD, Luz RK, Furtado C *et al.* The complete mitochondrial genome of the threatened neotropical catfish *Lophiosilurus alexandri* (Silurifomes: Pseudopimelodidae) and phylogenomic analysis indicate monophyly of pimelodoidea. *Genetics and Molecular Biology* 2016; 39:674–677.

Germany	*Clarias gariepinus*	Freshwater	780	
Greece	*Clarias gariepinus*	Freshwater	...	
Hungary	*Clarias gariepinus*	Freshwater	...	
Hungary	*Silurus glanis*	Freshwater	252	
Hungary	*H. longifilis x C. gariepinus*	Freshwater	3,333	
Italy	*Ameiurus melas*	Brackishwater	...	
Italy	*Ictalurus spp*	Freshwater	...	
Italy	*Clarias gariepinus*	Freshwater	...	
Italy	*Ictalurus punctatus*	Freshwater	51	
Italy	*Ameiurus melas*	Freshwater	87	
Latvia	*Clarias gariepinus*	Freshwater	...	
Latvia	*Silurus glanis*	Freshwater	...	
Lithuania	*Silurus glanis*	Freshwater	7	
Lithuania	*Clarias gariepinus*	Freshwater	214	
Moldova, Republic of	*Silurus glanis*	Freshwater	2	
Netherlands	*Clarias gariepinus*	Freshwater	4,000	f
Poland	*Clarias gariepinus*	Freshwater	150	
Poland	*Silurus glanis*	Freshwater	365	
Romania	*Clarias batrachus*	Freshwater	...	
Romania	*Clarias gariepinus*	Freshwater	...	
Romania	*Ictalurus* spp.	Freshwater	...	
Romania	*Silurus glanis*	Freshwater	28	
Russian Federation	*Ictalurus punctatus*	Freshwater	1,879	
Serbia	*Silurus glanis*	Freshwater	18	
Slovakia	*Silurus glanis*	Freshwater	1	
Slovakia	*Clarias gariepinus*	Freshwater	822	
Slovenia	*Silurus glanis*	Freshwater	...	
Ukraine	*Ictalurus punctatus*	Freshwater	...	
Ukraine	*Silurus glanis*	Freshwater	58	
United Kingdom	*Clarias gariepinus*	Freshwater	...	

OCEANIA

Country (Country)	Catfish species	Environment	Production (t)
Guam	*Clarias batrachus*	Freshwater	...
Vanuatu	*Pangasianodon hypophthalmus*	Freshwater	...

* Brazilian aquaculture statistics showed some divergences with regard to data provided by FAO [9]. In particular, the Brazilian catfish production relies on *Pseudoplatystoma* spp. (Pimelodidae) and their interspecific and intergeneric hybrids (11,505 t in 2018) [10].

Symbols used: "..." = data not available; unobtainable; data not separately available but included in another category; "0" = more than zero but less than half the unit used; "f" = FAO estimation from available sources of information.