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Cover image: *Spatuloricaria puganensis* egg, ca. 11 days. Photo: M. Walters

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- The JCSG relies on the contribution of content from its members and other parties. No fees or honoraria are paid in exchange for content and all proceeds from advertising and subscriptions are used to support CSG events and activities. At the end of the financial year, any remaining funds generated from subscriptions to JCSG are transferred to the CSG Science Fund. The Editor and other CSG personnel involved in the production of the journal do so voluntarily and without payment.

- Please support the CSG by contributing original articles concerning catfishes that might be of general interest to JCSG subscribers. Word processor files (e.g., .doc, .docx) and high-resolution images (at least 300 dpi and 8.5 cms wide) can be sent as email attachments to editor@catfishstudygroup.org. If necessary, the Editor will work with contributors to structure and polish text and format images for inclusion in the journal. The Editor reserves the right to edit submitted text and images to improve flow and comply with the layout and page limits of the journal. Any manuscripts that propose taxonomic or nomenclatural changes will be forwarded to a qualified professional for evaluation.

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Editorial

Welcome to the first issue of the 2016 journal. Many thanks to all our sponsors, subscribers and, especially contributors.

This issue marks my first anniversary as Editor, and I agreed to continue generating the journal at the 2016 AGM.

The Spring brings the promise of new life, and this issue of the journal focuses on an aspect of the hobby that motivates many of us to convert outbuildings, harvest rainwater, collect or culture live food and spend months encouraging our catfishes to breed. For me, trying to spawn catfishes is the least I can do and should guarantee that I provide the best conditions for the live animals in my care.

I am delighted to see several new contributors to this issue, with articles from regulars such as myself and Mark Walters being bolstered by Colin Eveson, Elko Kinlechner, Pete Liptrot and Daniel Konn-Vetterlein. I am especially pleased to have a report from Pete on what is possibly the first confirmed UK spawning of a banjo catfish!

Welcome to our new subscribers and members. I hope you find something new and interesting in these pages, and that you recommend us to your friends and colleagues.

Thanks,

Michael

editor@catfishstudygroup.org

From the Chair

The first edition of the 2016 Journal provides an opportunity to reflect on another successful 2015 for the CSG. I see the club delivering its main objectives, to further the study of catfishes, through four core activities – the journal, the convention, social media and our regular meetings. The committee spends most of its time managing these products and events on behalf of CSG members.

As I look forward to our annual convention, I realize how much its success relies on the input of members who attend and I am looking forward to meeting and greeting lots of new conventioneers this year! During the last year the group has increased its membership significantly. Membership had been hovering between 60 and 80 for at least the ten years of my involvement. In the last twelve months since we introduced the new constitution and began modernizing our structure and administration, membership has grown to over 300 with roughly half of that number subscribing to the journal and attending the convention!

The introduction of free membership has helped push us in this positive direction, but other factors have contributed, including opening up our Facebook group to other interested catfish keepers and promoting membership through this and other forums. The introduction of digital subscription for the journal has also helped the group reach and serve a wider audience, making it easy and affordable to be a part of the global catfish-keeping community.

Numerous technological improvements to the group have been made, thanks in no small part to our General Secretary and IT guru Julian Dignall. Jools has revamped the website to become a slick one-stop-shop for CSG information, ticketing and subscriptions. We have also shifted our forum activity fully to our Facebook group, where you can learn about upcoming events and the latest news from CSG members. The facebook site is now generating hundreds of posts a month and some of those posts have become full articles in this issue of the journal.

Group meetings are also receiving our attention and this year sees the CSG broadening its geographical reach with meetings being planned in other locations. A full list of diary dates has been posted on the website.

The first event of 2016 was the Annual General Meeting, which gave the committee and members in attendance time to consider the results of our actions in 2015 and prepare for those we will make this year. The membership voted on a number of constitutional amendments and all were carried. This included the transfer of the President's duties to the Chairman and retirement of the post. Ian Fuller decided to step down from his role as President late last year so he could focus on his role as convention manager. The committee thanks him for the time and energy he has given to the difficult job of CSG President.

The club has made significant progress in the last year to further the study of catfishes through its core activities. With a strong and supportive committee I am sure I can help to drive the club forward to even greater heights in 2016.

Mark

chairman@catfishstudygroup.org

An introduction to catfish reproductive strategies and behaviour

By Michael Hardman

Fig. 1. Spawning group of *Scleromystax barbatus*. Photo by M. Hardman.

We love animals - particularly fishes. Why? I've studied and kept catfishes for over 20 years and have never considered a second of it wasted. The secrets that biology has to reveal to us are among the greatest rewards in life, and I would be wandering aimlessly if I were anything other than an ichthyologist. I've tried other things, but I'm no good at them.

The things that continually amaze me about catfishes are their diversity and distribution around the globe. Of all the vertebrate species on Earth today, 1 in 20 of them are catfishes. They are found from coral reefs to torrential mountain streams, and on every landmass except Antarctica - where they were up until around 30 million years ago. Catfishes range in size from inch-and-a-half dwarves to 6-foot leviathans and feed on everything from microscopic algae to the blood of other fishes.

Biologists are continually trying to understand the story of life and how it has evolved. They try to answer questions such as why certain kinds of plants and animals thrive and become diverse over time whereas others dwindle and become extinct. Catfishes are on the winners' podium when it comes to evolution, but we have few explanations for what has given them their competitive advantage.

The knife-edge of evolution often runs straight through how a species reproduces. Of the mixture of characteristics that make up a species, for individuals to maintain their species they have to get reproduction right. If environmental conditions change and no longer suit a strategy that was previously successful, local species will have to move, adapt or risk extinction.

GENERAL PATTERN

As far as they exist, we look for general patterns in the universe of diversity and hope that they reflect real phenomena that cause them; like the general pattern that objects drop to the ground as a result of gravity. General patterns give investigators a starting point when dealing with an unknown system. Given the importance of reproduction, I wanted to know more about its variation among catfishes and if any general patterns were to be found. I wanted to know this to consider what role, if any, reproductive strategy has played in their evolution.

I collected information from books and technical articles written by ecologists and fisheries biologists. I only included information if it concerned catfishes in their natural habitat so as to avoid any problems that might arise from unnatural behaviours stemming from life in captivity. This article highlights the incredible range of strategies that catfishes use to reproduce and what any patterns might imply about species that have yet to be spawned in captivity.

Secondary sexual dimorphism

Males and females are different in many ways. Their most important or *primary* differences are the sex organs that produce eggs (*ovaries*) or sperm (*testes*). Their maturity and ripening is usually tied to seasonal changes, especially at temperate latitudes where day length varies considerably during the year. Environmental cues trigger the pituitary gland to release hormones that stimulate the ovaries or testes. Once activated, the sex organs begin to release other hormones that cause changes elsewhere in the body. These changes are the *secondary* sexual characteristics and can involve structural, coloration or behavioural differences that potential mates or competing males can use to evaluate their readiness or quality. As such, they are usually obvious and external.

Many catfishes are sexually dimorphic and remain so throughout the year. Some express them only seasonally whereas others have only slight differences in the genital papillae. Amblycipitids (Torrent catfishes), some amphiliids (Loach catfishes), ictalurids (North American catfishes) and claroteids have males with expanded mouths and swollen cheek muscles that establish and defend a nest of eggs from would-be predators (Fig. 2). Mature male loricariids and callichthyines (*Hoplosternum, Callichthys* and relatives) have longer pectoral spines and more of the skin teeth (*odontodes*) that give them a “hairy” appearance. While most *Corydoras* have smaller males, species of the closely related *Scleromystax* have males with elaborate patterns, fins and cheek patches of odontodes (Fig. 1).

Fig. 2. Male (above) and female (below) *Chrysichthys auratus*. Notice the swollen cheek muscles and swollen mouth of the male in this nest-defending species.

Internal or external fertilization?

The vast majority of fishes fertilize their eggs externally, as do most catfishes. Typically, the female and male release their eggs or sperm simultaneously in an embrace. Much like the familiar “T-position” commonly seen in spawning *Corydoras*, catfishes such as *Auchenipterichthys* (Auchenipteridae), *Pseudobagrus* (Bagridae), *Clarias* (Clariidae), *Noturus* (Ictaluridae), *Synodontis* (Mochokidae) and *Tandanus* (Plotosidae) all have these spawning embraces.

Astroblepids are not known to form embraces but males develop an extended genital papilla that presumably allows him to release sperm directly on the egg in their torrential habitat of mountain streams where sperm would otherwise be washed away. Driftwood catfishes (Auchenipteridae) have gone one step further and developed a form of internal fertilization. Male auchenipterids have a reinforced and elongated genital pore that is used to inseminate females who store the modified sperm-packets in a special pouch until they are needed to fertilize the eggs.

Although still technically external fertilization, scientists in Japan have determined that female *Corydoras* catfishes actually drink sperm from the male, pass it through their intestines and release it onto several eggs held between their pelvic fins. In this way, the female increases the fertility rate by creating a sperm-rich environment for her eggs. While not as complex as the sperm packets of auchenipterids, *Corydoras* sperm are released in smears of protective mucus, presumably so they can reach the egg without becoming dinner! The sperm-swallowing strategy is, at present, a unique feature of *Corydoras* in the entire animal kingdom.

Where do catfishes place their eggs?

Fishes are often beautifully adapted to their environments. Because catfishes are found in so many different habitats their reproductive strategies are equally variable. One of the main reproductive decisions facing fishes is where to release the eggs. Catfishes either scatter or place their eggs in open water, on a substrate or inside a cavity.

Relatively few catfishes scatter or *broadcast* their eggs in open water. Two families are known to do this. The first are the shovel-nosed pimelodids that migrate up the flooded streams of South America to spawn. Females, when ready, invert at the surface and allow several attendant males to swim over her and release their sperm while she expels many thousands of buoyant eggs. The eggs ride the stream and eventually hatch, stopping along the way to feed before ultimately arriving in the nutrient-rich waters of the Amazon or Orinoco delta. In Africa, schooling schilbeids release thousands of small eggs into the water column and, similar to pimelodids, never experience the joys of parenthood.

Several catfish families are slightly more responsible and, while they provide no parental care, at least try to give the eggs some protection by broadcasting them over a porous substrate or inside a cavity. Heptapterids such as *Rhamdia* show no obvious sexual dimorphism and gather in large spawning groups to scatter their eggs over clean gravel in slow-flowing streams. Clariids follow the rising floodwater into the surrounding fields to spawn in soil among vegetation. Interestingly, aquaculturists using pituitary extract to artificially spawn *Clarias* have noted that the 10-15 hour period it takes for females to ovulate is similar to the amount of time the river takes to spill over its banks after the onset of heavy rains.

Mochokids are perhaps best known for the cuckoo-style spawning of *Synodontis multipunctatus* that parasitizes the mouth-brooding instincts of Lake Tanganyika cichlids. Non-Tanganyikan *Synodontis* are known to spawn in crevices and cavities but, until recently, were not known to show any sexual dimorphism. Detailed anatomical studies of *Synodontis*, *Microsynodontis,* *Mochokiella* and *Acanthocleithron* have all revealed dimorphisms of the skin, gill cover, shoulder bones and fins, although it's not clear how the species reproduce or how the structures might be involved.

Fig. 3. Male *Loricaria similima* with egg raft. An example of a lip-brooding loricariine, Photo: M. Hardman

Stay put or keep moving?

Placing the eggs somewhere protected improves their chance of survival to hatching, but only in so far as they are not discovered. Many catfishes remain with the eggs until they hatch to keep predators at bay as well as keep them clean and well ventilated. Catfishes that protect the eggs do so either in a fixed or mobile location. Based on what my review, it seems that most catfishes hide their eggs and remain with them until they hatch but there are several fascinating exceptions.

Ariids and some dwarf claroteids in Lake Tanganyika incubate eggs in their mouths rather than nests. In the marine Ariidae, having a mobile nest allows the parent to follow a changing salinity in a lagoon that receives variable amounts of freshwater input. This is important because while the adults can tolerate salt the developing embryos cannot. In freshwater Lake Tanganyika, being small means that lots of larger fishes can eat you, so evacuating yourself and your family in the instance of being discovered clearly has a benefit.

Other catfishes have adopted the mobile family lifestyle but use different structures to move the eggs around. The males of certain loricariids (*Loricaria, Pseudohemiodon, Planiloricaria* and *Loricariichthys*) have seasonally-expanded lower lips to hold and transport a raft of adhesive eggs (Fig. 3). In contrast, female Banjo catfishes of the genus *Aspredo* develop special structures for holding individual eggs on their bellies. These *cotylephores* are filled with blood vessels that might supply oxygen as well as nutrients to the developing embryo. In the estuaries of southern Asia, *Mystus gulio* (Bagridae) similarly transports and possibly nourishes a large egg mass with specially developed belly skin.

But, as I mentioned, most catfishes establish a nest and defend it. Some occupy existing holes such as the dens of other animals, e.g., auchenipterids, ictalurids and loricariids (Fig. 4). Some dig their own holes, e.g., claroteids, malapterurids, pseudopimelodids and silurids. Some build their own nests of plant debris and saliva-coated bubbles (callichthyines), gravel (plotosids), or shells (auchenoglanidids). Quite commonly, catfishes will spawn underneath a

Fig. 4. Male *Panaqolus* sp. L002 ushering a female into a potential spawning cavity. Photo: M. Hardman

large boulder or ledge, e.g., amblyciptids and bagrids.

Spending time with the kids

While defending nests of eggs is common, it seems that once catfish embryos hatch, they are on their own. Protecting the embryos and juveniles has only been observed in a few species from a three families. Parents of both the dwarf (*Lophiobagrus*) and giant (*Chrysichthys*) claroteids of Lake Tanganyika have been found guarding hundreds of free-swimming juveniles.

Similarly, both parents of *Ameiurus* species (Ictaluridae) are known to protect their young well beyond the free-swimming stage. Additionally, young *Ameiurus* form revolving masses that, if juveniles from different parents are mixed together, they will re-segregate and form two separate masses according to their family.

Tough love for Cichlids

The award for "most impressive reproductive behaviour among catfishes" must surely go to *Bagrus meridionalis* (Bagridae). This 20-lb endemic of Lake Nyasa nests under large rocks and defends both the eggs and juveniles. But it doesn't stop there. The female regularly releases unfertilized eggs for the family to feed on and, as they grow, the male helps them find invertebrates by disturbing local sediments.

These excellent parents also operate something of a foster-home for orphaned cichlids, although they earn their keep by occupying the outermost positions in the shoal where they are most likely to become a meal in place of a much loved baby catfish.

But the part of their reproductive behavior that I find most amazing is that while the male performs all the threat postures to potential predators, it is the female that strikes.

Clues to spawning success

From this review, I've learned several things about reproduction in catfishes. The first is that they reproduce in a remarkable number of ways and have achieved a level of sophistication that rivals many of the more advanced fishes. Most catfishes have sexual dimorphism, fertilize their eggs externally during brief embraces and protect them in nests until they hatch. In the tropics, reproductive activity peaks during periods of high rainfall and increased temperature, whereas daylength (*photoperiod*) appears to be more important trigger in temperate species.

Unfortunately, the categories that I tried to correlate with reproductive strategy (phylogeny, maximum body size and habitat) showed a poor or no relationship. This means that these easily measured features do not provide clues as to how we might trigger the reproduction of those groups that are suitable for life in captivity but for which we know little of their reproduction, e.g., Akysidae, Chacidae, and Sisoridae. However, when it was reported, all tropical catfishes timed their reproduction with seasonal rains and when annual temperatures were highest. So, you could do worse than increase both the flow rate and the setting on your heater-thermostat when your catfishes look ripe with eggs.


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Spawning and raising *Ancistomus* sp. L208 (Loricariidae: Hypostominae)

By Elko Kinlechner

Fig.1. 5 month-old *Ancistomus* sp. L208. Photo: E. Kinlechner

A few years ago, I bought two males and one female of an undescribed pleco known as *Ancistomus* sp. L 208 (Fig. 1). I am delighted to report that within a few months of receiving them, I managed to spawn and raise this rare and handsome species. This is an account of the spawning and the technique I used to raise the larvae and juveniles.

Ancistomus sp. L208 was introduced to hobbyists by a familiar face to many CSG conventioneers; Hans-Georg Evers in March 1996. Hans designated the L208 for what was a newly-imported and previously unknown species in DATZ magazine. The fish was brightly patterned and was reportedly from Rondônia in Brazil.

In the original article, the photos for L207 and L208 were incorrectly labeled. The article text clearly states that L208 is from Rondônia and has a high-contrast pattern, but the corresponding image was labeled "L207 aus Rondônia" [L207 from Rondônia].

Four years after it first appeared in DATZ, Hans collected *Ancistomus* sp. L208 in the río Jaciparaná. This stream is a right-bank tributary of the mighty río Madeira and lies a little more than 100km southwest of Porto Velho. The Jaciparaná is a white-water river and when he was there in October 2000, Hans measured the following parameters: 28 °C; conductivity 20–40 µS/cm; pH 6.9.

The Jaciparaná is not often visited by ornamental fishermen – especially those looking for plecos – as it is considered to be of low productivity and too far away from more lucrative areas. As such, European imports from the Jaciparaná are extremely rare… but always fascinating!

Spawning aquarium

Before I was due to collect my new plecos, I prepared a 240L aquarium (120 x 40 x 50 cm). As in my other pleco tanks, I added a coarse sand substrate (1–3 mm diameter) and installed an Eheim Aquaball 2212 (max. 650 l/h), a Sera

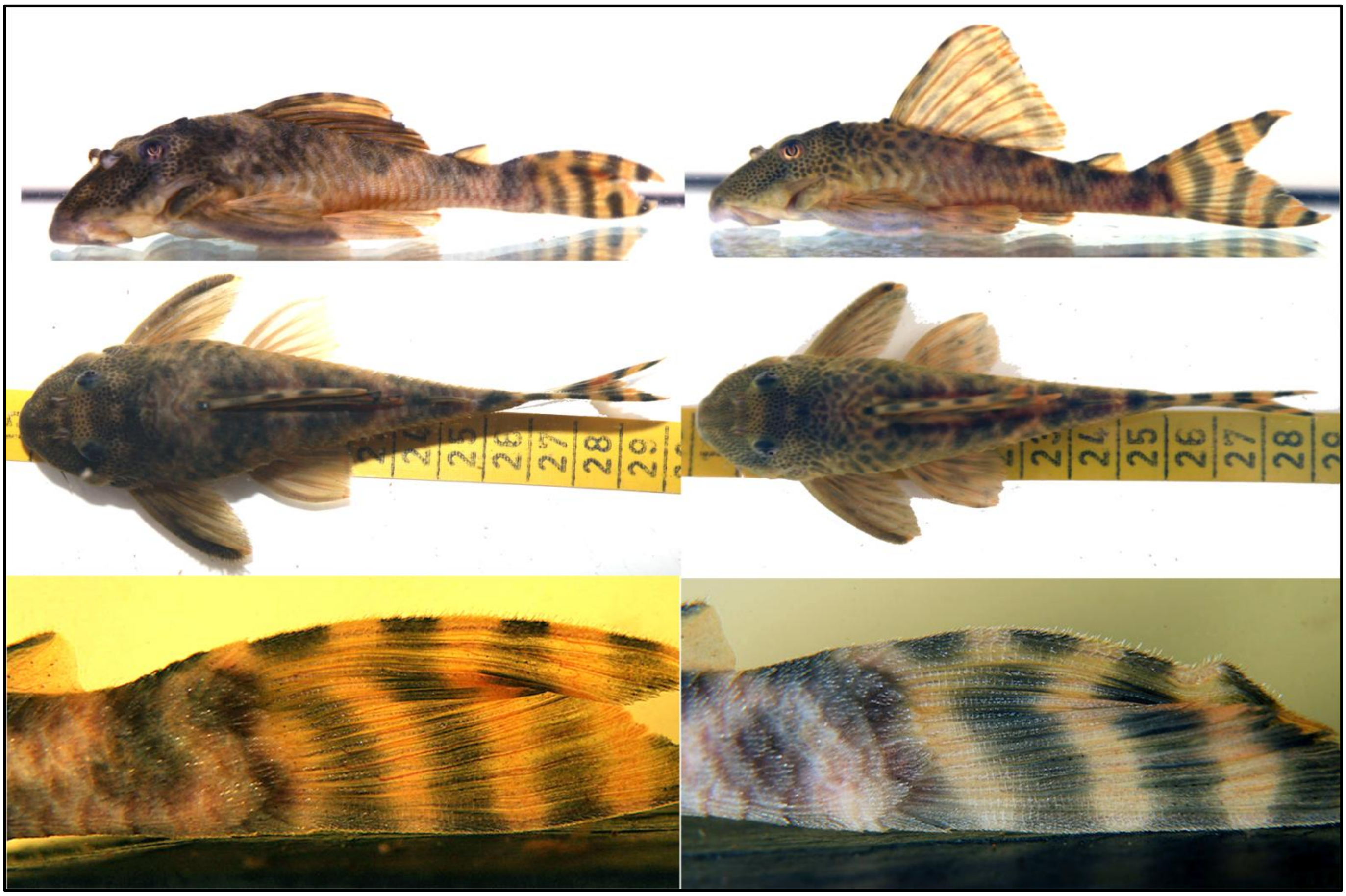

Fig. 2. Comparison of adult female (left) and male (right) *Ancistomus* sp. L208. Photos: E. Kinlechner.

air filter L150 (max. 150 l/h) and two 75W heaters. I always double-up on filters and heaters in case of any failures.

To this basic pleco system, I added five standard caves (~18 cm long) and a few seasoned roots to add some natural structure and shelter. I do not have aquatic plants and the tank receives ambient lighting.

I feed my *Ancistomus* sp. L208 mainly with and 2:1 mix of DuplaRin G and XL. These are high-quality dry foods designed for carnivores and can be found in German online retailers. Occasionally, I also provide catfish tablets and twice a week all my fish receive frozen foods. Although I offered fresh vegetables, they were never eaten.

With respect to water chemistry, I tried to simulate the field measurements provided by Hans. When they spawned, the temperature in the aquarium was 28 °C, pH 6.9 GH <1° dH, KH <1° dH, nitrite <0.025 mg/l, nitrate <10 mg/l, conductivity 112 µS/cm. I changed approximately 50% of the water each week, with rainwater or RO water.

Spawning group

The adults were approximately 13 cm when they spawned, and have grown a few cms since. The male can be distinguished from the female on the basis of body shape in that females have wider or more rounded bodies, especially in dorsal view (Fig. 2). Additionally, the genital papilla of males is more pointed. Unlike many other ancistrines, the odontodes on the interopercle, pectoral-fin spines and unbranched rays of the caudal fin are actually quite similar in both sexes.

Spawning

On the 3rd of February back in 2009, I was surprised to find a few loose eggs in front of a cave. I had not made any attempt to spawn them, but during the 10 days prior to spawning the atmospheric air pressure fluctuated strongly (Fig. 3) and I suspect this may have provided the cue and trigger for oviposition in my group.

I carefully removed the eggs and transferred them to an in-tank incubator. To discourage fungal infection of the eggs, I used alder cones,

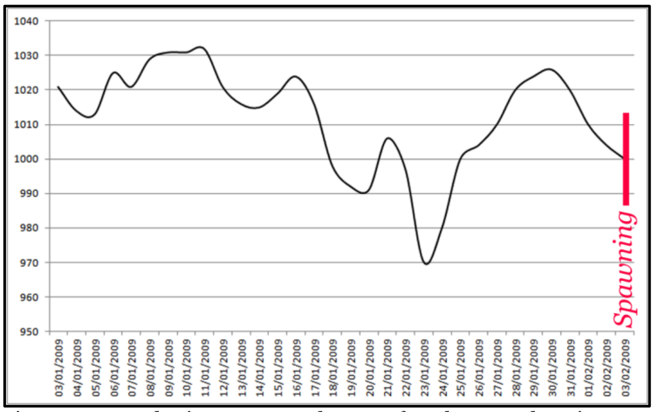

Fig. 3. Atmospheric pressure changes for the month prior to spawning. Note the large drop 17–23 January, and the second drop beginning 30 January. My group spawned 2–3 February.

catappa leaves and Sera Ectopur (according to the instructions). A few Malaysian trumpet snails (*Melanoides tuberculata*) were also added to gently scrape any biofilm that might accumulate on the sides of the incubator or surface of the eggs. I removed a single egg that became whitish and opaque with a clean scalpel.

I could see that there were plenty more eggs in the cave with the brooding male, and propped the open end slightly upwards so that the eggs would tend to stay in the cave rather than detach from the mass and make their way out onto the sand. Seemingly, the brooding males objected to this and, using his tail fin, had swept away the substrate so that the cave was again horizontal.

After six days or so, the eggs hatched and some of the larvae could be seen emerging from the cave. At this point, I agitated the cave until the remaining fry had come out. There was a total of 50 eggs spawned, six of which died during the incubation period (days 0–6), 12 hatched in the incubator and 32 were in the cave.

Fig. 4. Several of the fry displayed a malformation of the spine, creating a truncated or kinked fish. Photo: E. Kinlechner

Seven of the 44 fry had clear developmental problems in that their spines producing kinked or truncated fish that were otherwise healthy. All of these were of fry that had developed and hatched in the cave. I allowed these individuals to survive and the malformation became more pronounced at larger sizes (Fig. 4). While malformations of this kind are quite common in captive-bred plecos, the cause was not identified.

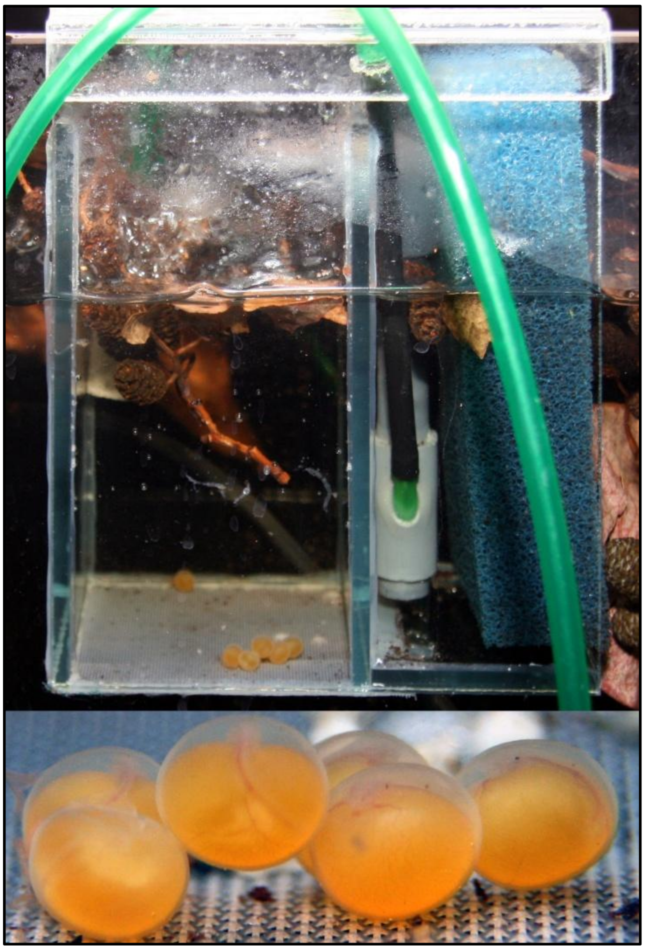

Fig. 5. Refugium incubator and close-up of the eggs discovered outside the cave. Photos: E. Kinlechner.

I housed the growing juveniles in a refugium (Fig. 5) placed within the spawning tank. I added a thin layer of sand and a few trumpet snails to consume any uneaten food and scrape all surfaces clean. A refugium receives a constant input of water from the main tank which leaves with any waste through a sponge filter in a similar way to a Hamburg Matten filter. In this way, the juveniles are kept in a food-rich environment with excellent water quality.

The yolk sac had been absorbed by roughly the 12th day. On day 10, I had begun offering small quantities of JBL Nobil Fluid, Cyklop Eeze, JBL Novo Tom and crushed JBL Tabis several times a day. Prior to this, the young animals were a

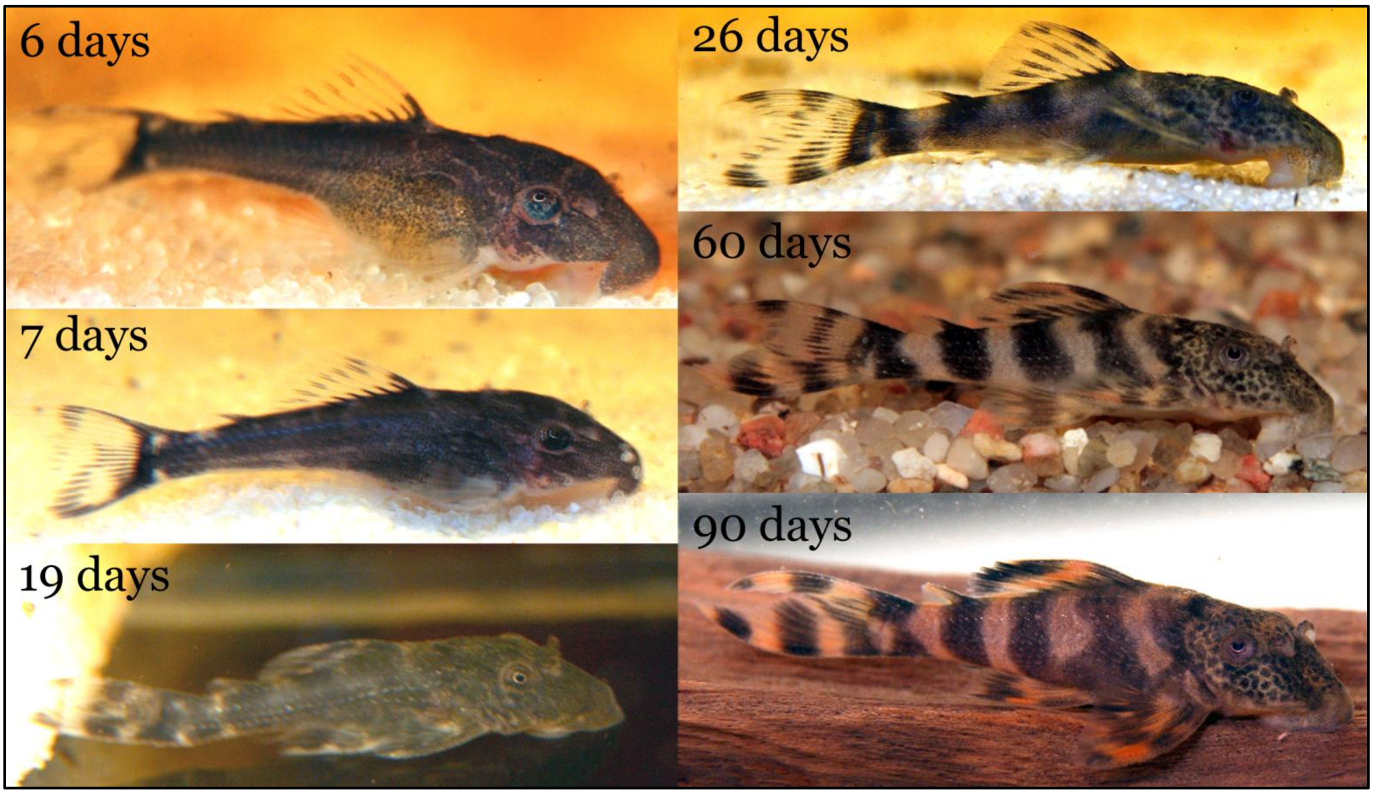

Fig. 6. Developmental sequence of *Ancistomus* sp. L208. Notice that the adult pattern is basically achieved approximately three weeks after hatching, but that fry are initially dark grey. Photos: E. Kinlechner.

dark grey and showed little resemblance to the adult pattern. However, after the yolk sac was absorbed (day 12), I noticed the beginning of the color change on the caudal peduncle. Within the next nine days, the animals gradually transformed and were eventually similar to their parents (Fig. 6).

After two months in the refugium, I moved the young plecos to their own 60L aquarium. From this point forward, they received the same food and husbandry as the adults. By November 2009, most were at least 9.5 cm (including the caudal fin), and were growing as much as 1 cm per month!

I have since learned that other aquarists have managed to spawn *Ancistomus* sp. L208 but my group or their offspring have not shown any signs of spawning since February 2009. Perhaps the weather has not been quite right…

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– (2002) Naturschutz mit und für den Menschen, *Aquaristik Fachmagazin* 163: 50–56.

Tatia dunni (Fowler 1945) – A great woodcat for the smaller tank (Auchenipteridae: Centromochlinae).

By Daniel Konn-Vetterlein

Fig. 1. *Tatia dunni* (Fowler 1945). Photo: D. Konn-Vetterlein

Woodcats (Auchenipteridae) are widely known to be painfully-shy creatures. They rarely venture out during the day, preferring to hide in the darkest and most-secluded space in your tank until the lights have gone out and the darkness of night descends. *Tatia dunni* (Fig. 1) is quite typical in this respect, but recently I was able to spawn this attractive species and wanted to share my observations on woodcat reproductive behaviour to that which is so far mainly based on *Centromochlus perugiae*.

Tatia dunni grows up to 10–12 cm and has a body shape that is fairly standard for woodcats; sort of like a squashed cigar with very short paired fins – perfect for hiding in crevices, holes and places most other fish wouldn't fit into.

Dunn's Tatia should be offered live and frozen foods even if they accept standard dried foods and tablets. In nature, *Tatia* and other centromochlines are known to feed nocturnally on insects that become caught in the water surface. I had the opportunity to observe this myself using a torch to animate different flying insects and trust me, it's great to see all those mouths roaming about just beneath the water surface. They seem happy with all the live foods I've offered and it's satisfying to watch how it excites these surprisingly agile and fast-

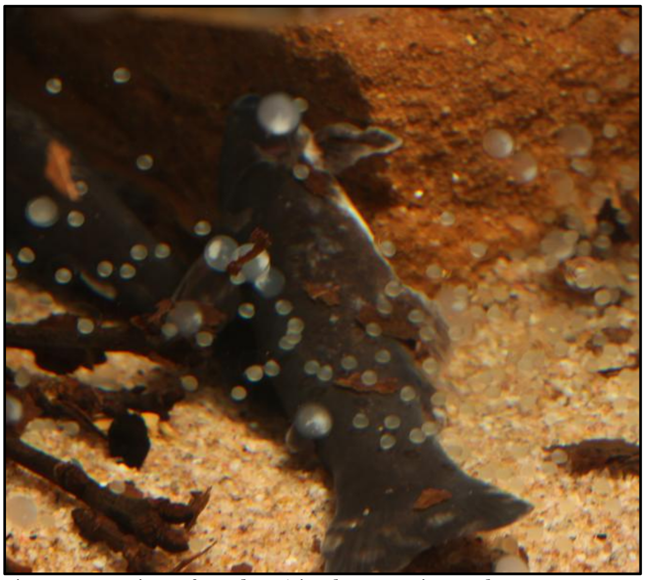

Fig. 2. Spawning of *T. dunni* in the aquarium. Photo: D. Konn-Vetterlein.

swimming predators.

Like other woodcats, male *Tatia* have highly-modified anal fins (*intromittent organs*) that transfer a parcel of sperm to a storage organ in the female. In this way, the female can save the males sperm and release it to fertilize her eggs when she decides it's time to do so. In aquaria, eggs can be placed in or on the substrate or sometimes in a secure cavity within the rock structure or bogwood.

Normally, the female tries to keep the eggs together in a single clutch, but on one occasion when *T. dunni* spawned, it appeared that the current was too strong and the eggs were scattered all over the aquarium (Fig. 2).

The first spawning resulted in almost 150 eggs, the second 93 eggs and the third around 180. The eggs are quite large (ca. 4mm in diameter) and it was straightforward to remove them from the spawning aquarium and incubate them in a smaller tank containing the same water and a small filter. After 30-35 hours (h), it was possible to see the larvae beginning to develop in the egg, and after 60h they started to hatch. The larvae have a huge yolk sac to feed on for the first three days. At first, one could be forgiven for thinking they had hatched too early

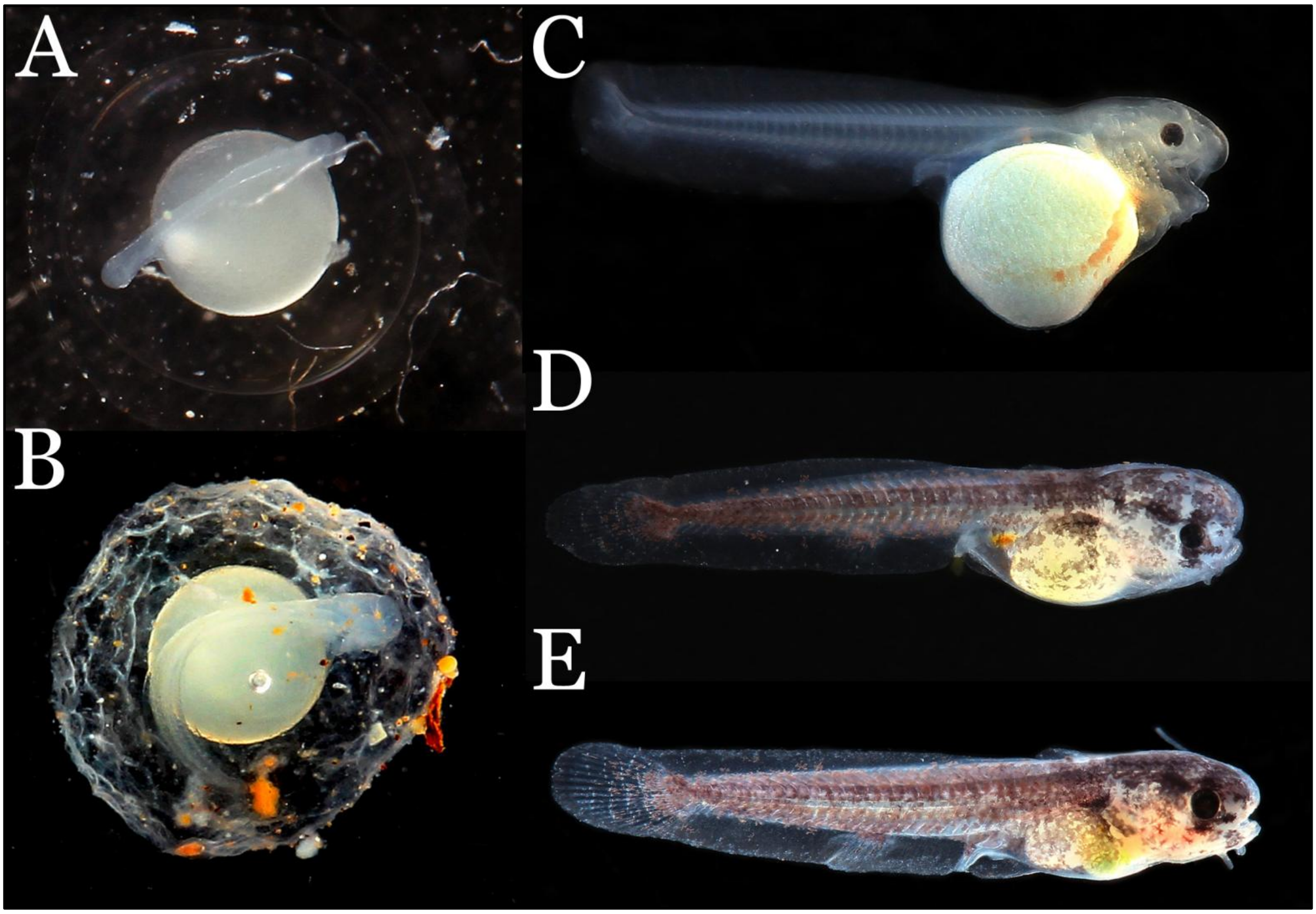

Fig. 3 (A–E). Developmental sequence of *T. dunni*. A: Egg at ca. 20 hours; B: Egg at ca. 40h; C: Newly-hatched larva at 60h; D: larva at 7 days, ca. 8mm; E: Larva at 9 days, ca. 10mm. Photos: D. Konn-Vetterlein.

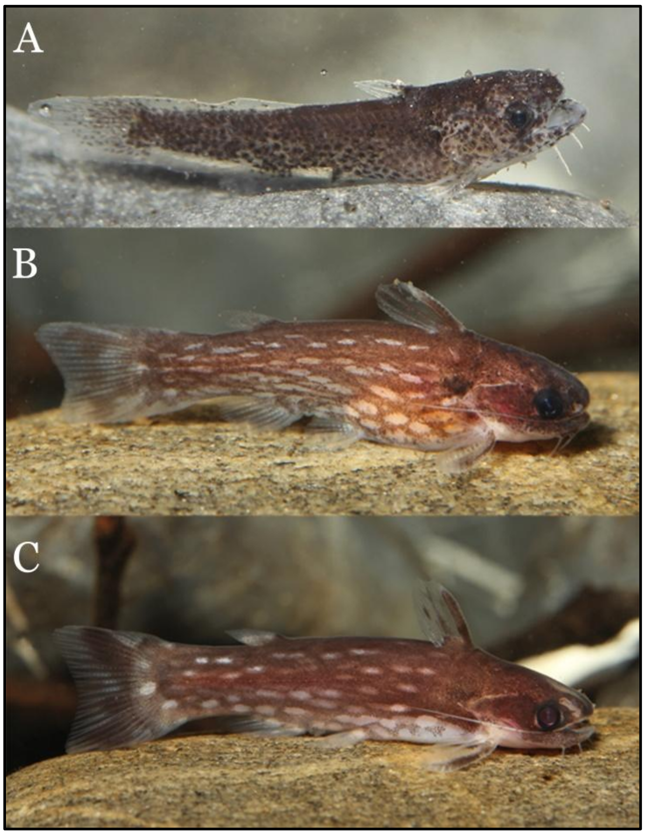

Fig. 4 (A–C). Growth sequence of *T. dunni* illustrating the development of pattern and shift to adult colouration. A. Juvenile at 3 weeks, ca. 18 mm; B: Juvenile at ca. 40 mm. C. Sub-adult at ca. 55 mm. Photos: D. Konn-Vetterlein.

because the egg membrane is so large and far from the larva, it can easily be overlooked in the early stages of development unless some debris attaches to it or it suffers some kind of damage. In comparison, loricariid eggs can be large but the membrane is more tightly-fitting around the developing larva.

The newly-hatched larvae are relatively easy to care for. They grow quickly so the main concern is providing adequate food while maintaining water quality. During the first few days after hatching, I feed them a protein-rich powder mixed with *Chlorella* algae and *Anabaena* bacteria twice a day. After three weeks they have already reached 8–10 mm and start to get very active during feeding time. This is possibly the best period of all, because it is the most exciting and it looks great when a hundred or more small *Tatia* are swimming around the aquarium in search of food. Once full, they return to their hiding place and the tank looks empty again.

After only two months the juveniles are already 3–4 cm long, after that they add roughly 1 cm per month – an impressive growth rate for a fish with an adult size of only 10–12 cm. Juveniles have a striking pattern of horizontal white stripes covering the whole body, but unfortunately lose this pattern as they become adults. Full-grown fish still show the same basic pattern, but the contrast is less strong. The brilliant white stripes of juveniles eventually fade to an attractive light brown.

Like many species we see in the hobby, there seems to be some confusion about the name of this fish. Shops and even private breeders mostly offer them as *Tatia intermedia*, but I think this is incorrect and that our fish should more accurately be identified as *Tatia* cf. *dunni*. This confusion can be traced back to a revision of the genus *Tatia* by Saramento-Soares and Martins-Pinheiro (2004), in which all known species of *Tatia* were listed and described. In that paper, two specimens were shown and identified as *Tatia dunni*; the holotype from the Río Caquetá in Colombia and another specimen from the Río Conambo in Ecuador. In my opinion, the second specimen is not conspecific with *T.* dunni and may represent a separate species. The pattern on the holotype of *T. dunni* is quite similar to the pattern of an illustrated specimen of *T. intermedia* from Suriname, and I suspect this is where the confusion may have began.

Fig. 5 Life stages of *T. dunni* illustrating the development of pattern and shift to adult colouration. Photo: D. Konn-Vetterlein.

Furthermore, *T. dunni* is restricted to the upper Amazon basin in Peru, Colombia and possibly western Brazil. This is where they are imported from and often labeled as *T. intermedia*. Since *T. intermedia* is superficially similar to the holotype of *T. dunni*, at first sight that seems okay but *T. intermedia* is found in tributaries much further downstream (e.g., Xingu, Tocantins north to Suriname). Woodcats labelled as *T. intermedia* coming from Peru are likely to be *T. dunni*, there is no overlap in the distributions. There are also morphological differences mentioned in the paper. The most useful of which when identifying a live specimen is: "a male modified anal fin (in *T. intermedia*) with 3–5 elongate antrorse curved segments (vs. 1-3 short antrorse curved segments in *T. dunni*)." (Saramento-Soares & Martins-Pinheiro, 2008).

To sum up, *T.* cf. *dunni* is an easy to keep and easy to breed woodcat, with an especially beautiful juvenile pattern. It is not expensive and can be comfortably housed in smaller the (<250L) tanks. It might get difficult to distribute juveniles of the subsequent spawnings, but it's worth doing it once to see the armada of young *Tatia* buzzing around for a few minutes each day in one of your "empty" tanks.

Literature:

Sarmento-Soares, L.M. and R.F. Martins-Pinheiro (2008). A systematic revision of *Tatia* (Siluriformes: Auchenipteridae: Centromochlinae). *Neotropical Ichthyology* 6(3):495–542.

An unexpected and very rare spawning of the banjo catfish *Bunocephalus colombianus* Eigenmann 1912 (Aspredinidae: Bunocephalinae).

By Pete Liptrot

Fig. 1. *Bunocephalus colombianus* Eigenmann 1912. Photo: S. Grant

Recently at [Bolton Aquarium](), we had an unexpected spawning in one of our display tanks containing a small group of banjo catfish, *Bunocephalus colombianus* Eigenmann 1912 (Fig. 1).

We have had banjos for most of the last two decades as they have proven popular with our visitors and help us to explain crypsis. Plus, we find them fascinating in their own right and are a group of catfishes about which we still have a lot to learn.

Given our special interest in banjos, we have tried to spawn them a few times in the past. Many of us are familiar with the idea that banjos spawn in nests of sunken leaf litter and other organic debris. Earlier, we provided what we thought were ideal conditions, a suitable substrate, and gave them extra rations containing lots of well-cleaned live foods. They grew fat and looked happy enough, but they never showed any signs of pre-spawning activity or interest in each other. They did what banjos do so well; nothing. For me, they've always been something I really wanted to spawn because of their mystery, but I could say that about so many fish...

The group we currently have is a mix of individuals sourced from Pier Aquatics (Wigan) and Aqualife (Leyland). We have tentatively identified them as *Bunocephalus colombianus*, and they were purchased to supplement our small – and shrinking – group composed of older fish, some of which were at least 10 years old!

The new fish were not fully grown when we received them and we estimated that they were 1-2 years old. They are housed in one of our 400L Neotropical displays that they share with dwarf cichlids, geophagines, characoids and other small and peaceful fish. The aquarium is heavily planted with *Echinodorus* spp. and various stem plants, and which receives a ca.50% water change with conditioned tap-water each week. We use a Fluval 405 external filter (max 1500 l/h) containing mature biological media, and the water has a pH of 6.5–7.0, is soft and the temperature varies between 24–27 °C depending on ambient.

We mainly feed a staple diet of Aquarian and API dry foods, KB Discus granules, Vitalis pellets, Repashy and NLS gel foods, supplemented with a daily dose of live *Artemia* nauplii, frozen foods and an occasional treat of well-rinsed *Tubifex* worms.

Fig. 2 Ventral view of *B. colombianus*, female. Photo: S. Grant.

On the 21st January 2016, my colleague Paul Dixon noticed that the banjos seemed agitated. in particular, one of our plump females was swimming almost constantly, only stopping occasionally to land on or next to a much darker and more slender male. After which, their bodies would rock forwards, there was some increased ventilation, and the male would then lazily follow the female briefly before returning to the substrate and leaving the female to continue her activity alone.

We had done nothing different in terms of their husbandry, but the early part of 2016 has seen an unusually high number of low pressure systems moving across the UK. We have no evidence this was important, but experienced breeders often cite the importance of such weather patterns when encouraging reluctant species to spawn.

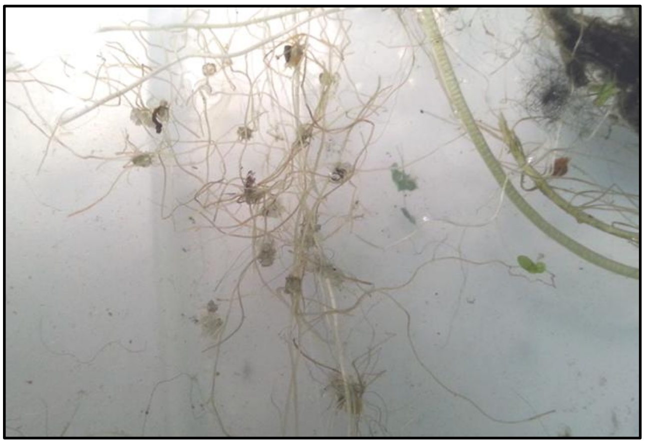

Fig. 3 Eggs of *B. colombianus*, as we recovered them attached to plant roots and other debris. Photo: P. Liptrot

We allowed the banjos to continue their activity undisturbed, and early next morning were excited to discover a large number of small green eggs scattered over the plants growing near the surface roughly at the centre of the aquarium, with a smaller number attached to roots and other debris directly beneath this area (Fig. 3). We removed the eggs and placed them into three containers with varying depths and amounts of aeration to see which would produce the best results.

On the 24th Jan the eggs began to hatch, producing very small fry which were negatively phototactic. Over the next 24 hours all the eggs hatched. The fry clustered under any debris within the hatching containers. Then came the wait for them to start feeding...

I'll write a separate article concerning our experiences rearing the larvae and provide an update to their progress in the near future.

Thanks to Eric Thomas for having previously shared his own observations on PlanetCatfish, and everyone from the CSG Facebook group for their enthusiastic response to our good news!

**** Editor's note – video of the spawning behavior, larvae and juveniles can be seen on the CSG Facebook group ****

Keeping, spawning and raising *Corydoras ourastigma* Nijssen 1972 (Callichthyidae: Corydoradinae).

By Colin Eveson

Fig. 1 *Corydoras ourastigma* Nijssen 1972, female. Photo: S. Grant.

I have to admit that *Corydoras ourastigma* wasn't at the top of my cory wish list but, like most catfish enthusiasts, I'm always on the lookout for new, rare or otherwise interesting species.

However, during a visit to my local fish shop in March 2013 I spotted a group of recently imported *C. ourastigma* (Fig. 1). By the looks of them they were young adults and in good condition. A quick check was made to ensure there was a suitable number of each sex before I struck a deal and made my way home with four males and two females of this rarely seen saddle-nosed cory.

I placed the bags in a vacant aquarium in my fish-house and left them to acclimate while I looked into them. I was surprised to find very little on keeping - let alone breeding - them, so I looked for clues for where to start in my notes on two other species of saddle-nosed corys I'd kept before; *C. septentrionalis* and *C.* sp. C115/116. My notes revealed I'd bred *C. septentrionalis* in a standard 60l aquarium with an air-powered sponge filter and the usual decor of sand, java fern (*Microsorium pteropus*), java moss (*Vesicularia dubyana*) and *Anubias* sp. Because they are closely related, it's fair to assume the conditions I provided for *C. septentrionalis* would be suitable for *C. ourastigma* too, and the tank they were currently floating in was already set up that way.

An hour or so later, the fish were released into a 60 x 30 x 30 cm 54L tank in the middle tier of tanks which usually ran with a temperature of 24 °C. A large and mature air-driven sponge filter provided excellent water quality and ample cover was provided by large clumps of java fern, java moss and *Anubias*.

The fish settled in quickly and little conditioning was needed due to them already being in good health. I feed a wide variety of dry, frozen and live foods and they soon became lively fish that were generally seen out and about most of the time I was in the fish house.

Over the next nine months, in spite of good feeding and regular waterchanges, no spawning activity was seen. As has been reported for other saddle-nosed corys, all six *C. ourastigma* regularly engaged in aggressive contests similar to the sparring of male *Scleromystax barbatus*! I never saw any broken fins or wounds and it wasn't possible to determine which, if any, was the dominant fish in the group.

Unfortunately, I lost four of the original six fish I bought to unknown causes. There were no obvious symptoms of disease or infection, so I just put it down to natural death or bad luck. Although the remaining fish appeared to be a pair, I felt my chance to breed them had gone and so I moved them to another tank which contained a small breeding group of *Ancistrus claro* along with some of their youngsters.

Again, the tank was the same size but on the top row where the temperature can be slightly higher. Furthermore, the tank was also equipped with a small internal power filter in addition to a large air-driven sponge filter, and furnished with a mix of terracotta caves, bogwood, java fern and *Anubias*.

Good feeding and waterchanges were kept up and the fish continued to stay in good condition, and I noticed that the aggression had died down. I was therefore surprised to enter the fish house one morning and find a good number of eggs spread around the tank and that the remaining pair of *C. ourastigma* were still spawning! The next half hour was spent watching the typical cory spawning routine, with the male chasing the female before forming the t-position in a corner or otherwise restricted space. The female then released between 1 and 3 small (ca. 1 mm) eggs into her ventral-fin pouch before placing them on the tank sides, the undersides of *Anubias* leaves as well as on the bogwood where small amounts of java moss had become attached.

At the time of spawning, the parameters in the tank were pH 6.5, TDS 115 and 26 °C. A 25% waterchange had been done on the tank the day before spawning using a 1:1 mix of rain and tapwater which lowered the temperature by around 4 °C.

I left the fish to complete their spawning in peace and returned later in the afternoon when all seemed quiet. I removed over 100 eggs from the spawning tank and placed them into a 1 l plastic tub containing tank water and an airline was added to create a steady flow over the eggs but not as much to move them around the tub. I then added four alder cones to soak for two hours before being removed; my preferred method for discouraging fungus.

Approximately 24 hours later, ca. 99% of the water in the hatching tub was replaced with water from the spawning tank. This routine was repeated daily over the next three days with infertile eggs being removed as they were discovered. Four days after spawning, the 50 or

Fig. 2. Eggs and 4–5 day old fry of *C. ourastigma*. Photos: C. Eveson.

so remaining eggs began to hatch - resulting in a mass of the tiniest fry I've seen among the 50 or so cory species I've bred!

Daily waterchanges continued to be carried out using tank water from the spawning tank but they were reduced to 50% volume and, as per my usual routine, I added some java moss to the tub to provide some cover and something for the fry to forage on.

I began feeding the fry two days after hatching, which consisted of a small amount of microworm (*Panagrellus redivivus*) three times a day. This was continued until they were 10 days old when they were transferred to a 3l tub with a small air-powered sponge filter where they received freshly-hatched *Artemia* (once a day), microworms (once a day) and finely crushed flake food before lights out.

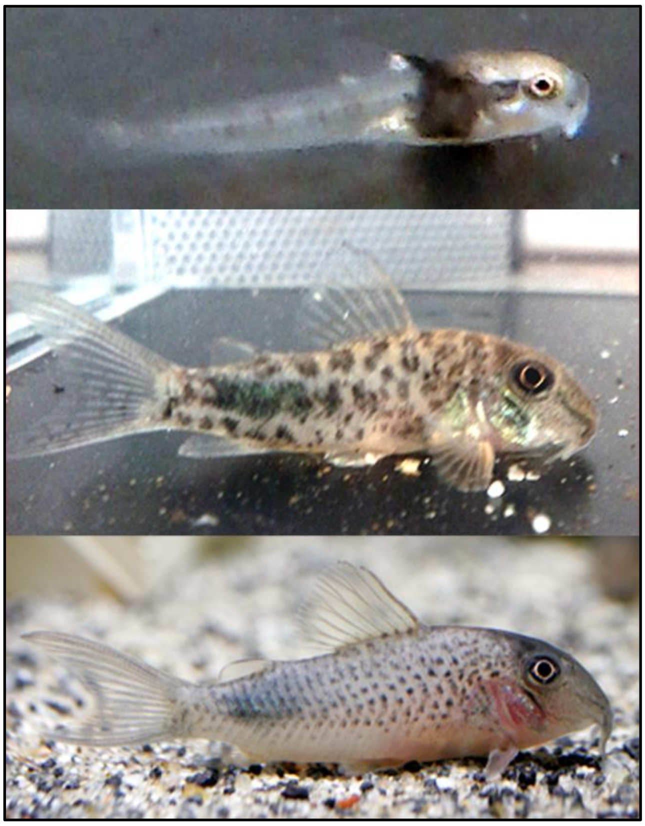

Fig. 3. Developmental sequence of *C. ourastigma* colouration in the aquarium. Upper: ca. 2 weeks. Middle: ca. 2.5 months. Lower: ca. 12 months Photos: C. Eveson.

The fry began to grow steadily but I lost approximately a third of the fry in the first two weeks. Approximately six weeks after hatching, the surviving fry were transferred to a 45x25x25 cm (28L) aquarium. After 8-9 weeks the youngsters began to develop the adult colouration with the tail spot staring to show on the larger fish in the group (Fig. 3).

Though not the quickest growing cory I've been lucky enough to raise, they did grow well enough for me to sell or exchange some of them with fellow aquarists when they were around 10 months old.

As I always do with any of the fish I breed, I kept six youngsters for myself and these fish bred when they were 19 months old, again moderate to strong turnover was important along with well-filtered warm water.

The original pair continued to breed roughly once every 2–3 weeks. I never noticed a particular trigger, and it seemed that they just needed clean, warm water with a good flow coupled with plenty of high quality live foods.

I'm still keeping *C. ourastigma* and am now growing on the grandchildren of the original pair. I would like to add somedifferent bloodlines to the spawning group I have, so if anyone sees any wild-caught fish, please give me a shout through facebook.com/colin-eveson or email editor@catfishstudygroup.org for my details.

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Spatuloricaria puganensis (Pearson 1937) (Loricariidae: Loricariinae) – a spawning report.

By Mark Walters

Fig. 1. *Spatuloricaria puganensis* (Pearson 1937). Photo: M. Hardman

Over the past few years, importers have made several species of the whiptail genus *Spatuloricaria* available to UK hobbyists, including the marbled *S.* sp. "caquete". In late 2014, Pier Aquatics imported several *Spatuloricaria* including a rather plain species from Peru. I noticed them in the shop because they were active and priced such that I could afford to buy a small group, which is how I like to buy catfishes I'm interested in. At the time, tank space and some other projects mean that they would have to wait.

During the CSG Convention in March 2015, Michael Hardman bought a few of the Peruvian species and posted some video footage of their foraging behaviour in the CSG Facebook group. The video showed how the fish picked up small stones (15–25 mm diameter) and cast them aside, in what looked to be a natural foraging behaviour – perhaps they were searching for aquatic insect larvae (e.g., trichopterans and ephemeropteans). Most loricariines tend to scrape surfaces (e.g., *Sturisoma, Harttia*) or sift soft sediments (e.g., *Loricaria, Pseudohemiodon*, etc.).

I was intrigued by the behaviour, and decided it was time to make room in my fish house for a group of *Spatuloricaria*. During my next visit to Pier Aquatics, I had the staff select four specimens on the basis of their genital papillae and body proportions. Given their extensively frilled lips, I assumed *Spatuloricaria* to be a lip-brooder similar to *Loricaria*, *Pseudohemiodon* and other sifting loricariines (see page 10 of this issue). However, I could not see any obvious differences among my group in terms of their lip morphology, so it seemed off if they were.

Of the remaining specimens, roughly half were >18 cm and the rest ca. 12–16 cm. I decided on two large and two small and, back in Boston Spa, they were acclimated to a 100 cm tank in a 250l quarantine system. I have been sharing notes with Michael about our experiences with this species, and he explained that he had been looking into the taxonomy of the group and that our Peruvian fish were most likely *Spatuloricaria puganensis* (Pearson 1937) (Fig. 1), although unclear diagnoses meant that he couldn't be sure.

After 6 weeks, I was happy they were clean so I moved the group to an 80 cm tank in my main 150ol system. I did not intend this to be their permanent home, realising that large whiptails require a tank with a large footprint to give them lots of room to roam, forage and (hopefully) breed.

They shared the 80cm with a small group of *Baryancistrus beggini*. Given that their dietary and habitat use do not overlap, I assumed they would not have a problem with each other. The tank contained a thin layer of sand for the *Spatuloricaria* and lots of seasoned bogwood to keep the *B. beggini* happy. A strong circulation pump added good flow which the *Spatuloricaria* settled happily into and eagerly awaited their next feed.

I find these whiptails to be quite flexible in their feeding preferences - greedily devouring all proprietary aquarium foods, although meatier foods such as gel-based preparations and frozen chironimid larvae caused special excitement in the tank.

Fig. 2. Cheek odontodes expressed by the male *Spatuloricaria puganensis*. Photo: M. Walters.

After 6 months, one of the larger specimens developed a brush of odontodes along the edges of his cheeks (Fig. 2) – revealing him to be a mature male ready to spawn. I collect and use warmed rainwater for my water changes, and October–November 2015 was especially wet in the UK. Consequently, I had increased water changes on the 150ol system because of the copious rainfall we received in west Yorkshire. I did not anticipate any further activity from the group, and wasn't deliberately trying to trigger spawning activity, although the two large fish were seen to be interacting and one of the larger fish without odontodes – assumed to be female - had become heavier and looked to have mature ovaries.

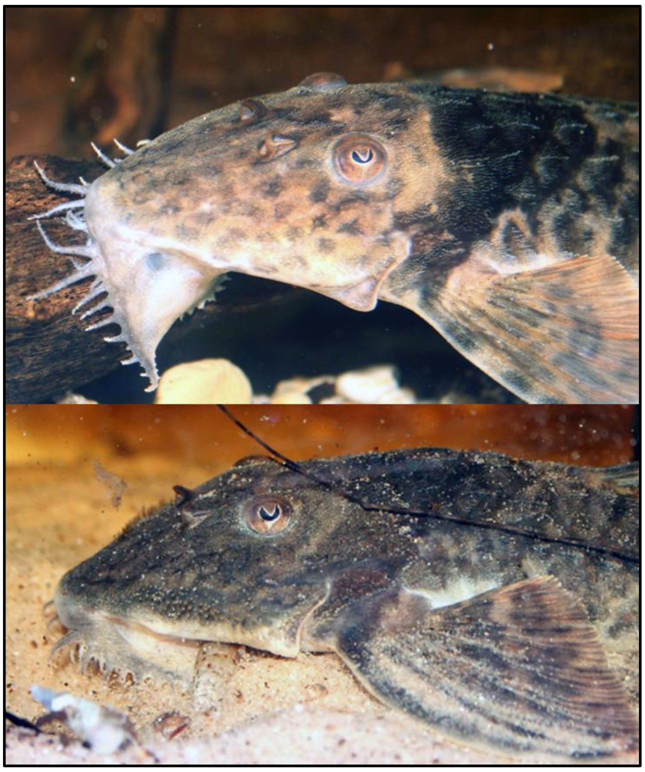

Fig. 3. Comparison of the heads of female (above) and male (below) *Spatuloricaria puganensis*. Note the cheek odontodes of the male. Photos: M. Walters.

A few weeks after witnessing their increased activity (Fig. 4), I added a few litres of fine gravel infested with Malaysian trumpet snails (MTS: *Melanoides tuberculata*) to their tank. I had noticed the population of MTS in the *Spatuloricaria* tank had crashed for some reason, and assumed that they had been eaten by the large whiptails. Some of the other tanks in the main system are also lacking MTS and these house *Leporacanthicus*, large *Peckoltia* and *Callichthys*. Perhaps they are also eating young MTS and keeping their numbers in check.

Fig. 2. Posteroventral view of a mature female just prior to spawning.. Photo: M. Walters.

A day later, I noticed how the *Spatuloricaria* had obviously given the new substrate a

Fig. 4. Combative pre-spawning behavior of *Spatuloricaria puganensis*. Male in the foreground. Photo: M. Walters.

thorough cleaning and, on closer inspection, I started to spot small spheres rolling across the surface with a similar size to the fine gravel. I was stunned to realise that these might be eggs, although them being loose on the gravel didn't make much sense.

To date, no loricariine is known to broadcast eggs and within the family Loricariidae, only the primitive genera *Rhinelepis* and *Pseudorhinelepis* are believed to spawn their eggs into vegetation, over a substrate or into the water column.

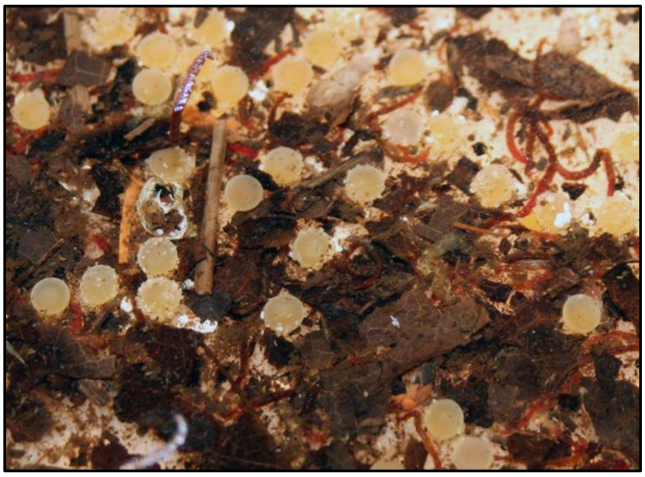

Fig. 5. Newly-laid eggs of *S. puganensis* recovered from the spawning tank.. Photo: M. Walters.

I posted photos on the CSG Facebook group page and Michael mentioned he had recently come across two articles in Amazonas magazine (Hemmann, 2007a,b) that detailed the spawning of two other species of *Spatuloricaria*. Both species attached an egg plaque to the underside of a flat stone and the male protected them, similar to other surface-spawning loricariines with sexual dimorphism.

On the basis of this information given in Hemmann (2007a,b), I was less-than-optimistic that the eggs were the result of a successful spawn. However, I carefully siphoned over 60 large eggs from the substrate (Fig. 5). The eggs were slightly adhesive and had pieces of sand and gravel attached to their surface. I handled a few of the eggs and noticed how tough the egg membrane was. After taking a few photographs, the eggs were placed in a tub of clean water filled from the outlet of a UV steriliser and I added a drop of methylene blue to prevent microbial growth.

Over the next eight days, the water was changed twice a day, adding fresh methylene blue each time. To my delight, the eggs appeared to be viable and were developing! On the 8th day, a few larvae hatched but were unfortunately dead and seemed underdeveloped (Fig. 7A). Based on my experience with other loricariids and Hemmann (2007a,b), I expected the eggs to remain developing for 10–12 days, so I was a little concerned that they had hatched prematurely and feared the loss of the clutch, perhaps due to the artificial incubation technique I used.

I stopped adding methylene blue when the water was replaced and checked on their progress more frequently, removing any eggs that died and turned white as soon as I noticed them. On the 10th day, the eggs started hatching and a few hours later I intervened with a pair of

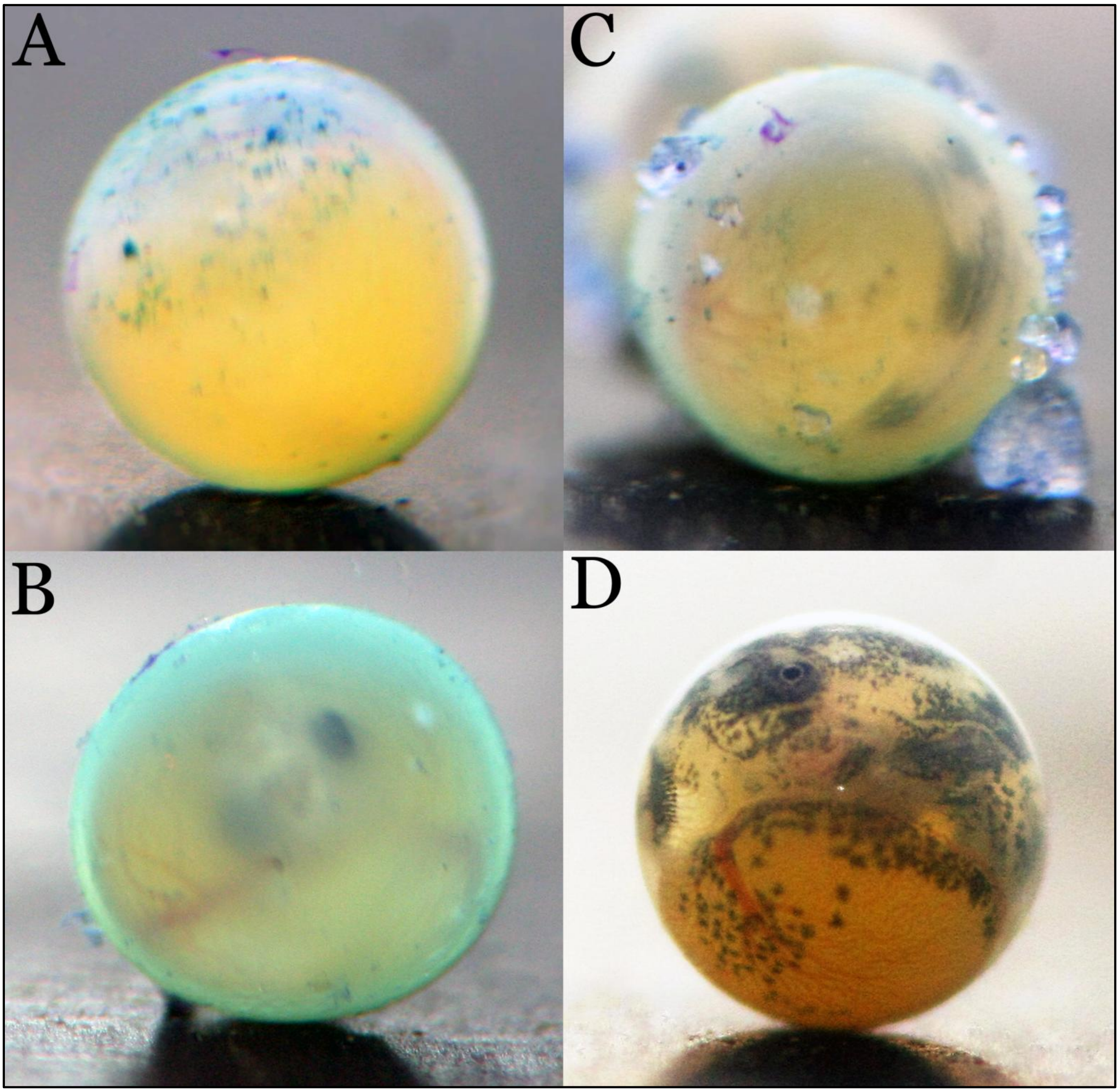

Fig. 6 (A–D). Developmental sequence of eggs in *S. puganensis*. A: ca. 24h. B. ca. 5 days. C: ca. 8 days. D: ca. 11. days. Photos: M. Walters.

pipettes to help the remainder out of their membranes. Of the original 60 eggs removed from the spawning, I managed a 25% hatch rate and 15 healthy fry from my first spawning of *S. puganensis*. After a further 5 days, the fry had consumed their yolk sacs and I offered first food in the form of powdered *Spirulina* and other fine fry foods. Like the adults, the fry of this species have healthy appetites and graduated to other foods (crushed tablets, chopped bloodworms, newly-hatched *Artemia,* etc.) and have been relatively trouble-free. I am pleased to report I have recently released 12 healthy juveniles into a grow-on tank where they receive the same foods as the adults.

A few weeks after the spawning event, I moved the adults to their own larger tank containing piles of flat rocks to see if they would spawn in the way reported for other species of *Spatuloricaria* (Hemmann, 2007a,b). In talking with Michael, we couldn't figure out why a species with sexual dimorphism (remember that males develop odontodes) would broadcast its eggs, and we wondered if the observation of loose eggs on the gravel was due to no suitable spawning site being available, or if they had spawned on the bogwood but the eggs had detached.

At the time of writing, my group has not shown any signs of spawning again, and the

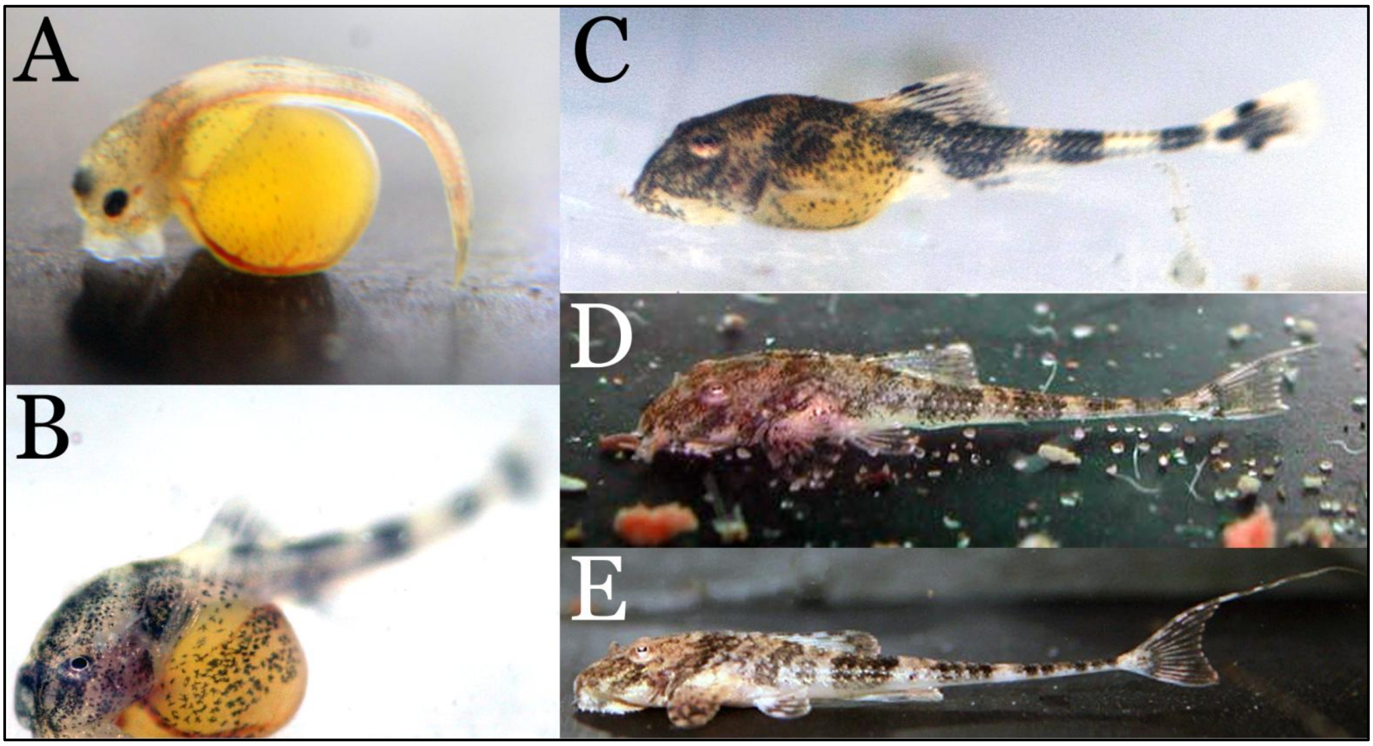

Fig. 7 (A–E). Larval developmental sequence in *S. puganensis*. A: Premature ly hatched larva (8 days) B.Newly.-hatched larva ca. 12 days. C: Larva ca. 16 days. D: Juvenile ca. 3 weeks. E: Juvenile ca. 7 weeks. Photos: M. Walters.

male has shed his odontodes. Incidentally, CSG member Richard Smith has also noticed how the cheek odontodes in another species of *Spatuloricaria* are often shed and re-grown at different times of the year. Temporary cheek odontodes might be a typical feature of the genus and something that distinguishes it from other sexually-dimorphic loricariines that tend to keep them year round (e.g., *Sturisoma*), at least in aquaria.

Literature

Hemmann, M. (2007a). Kammharnischwelse – *Spatuloricaria* sp. „Peru". *Amazonas* 12: 47–53.

– (2007b). Bärtige Kuscheltiere – *Spatuloricaria* sp. „Rio Nanay". *Amazonas* 12: 54–56.

CSG diary dates 2016

Date	**Event** *contact or presenter*	*Venue*
22 May	**Coryfest** *with Ian Fuller*	Derwent Hall, Darwen BB3 0DQ
19 June	**Summer lectures and sale** *with Richard Smith and Ralf Britz*	Derwent Hall, Darwen BB3 0DQ
10 July	**Away day: Castleford AS Catfish & Loach Show** *with Mark Walters*	Lock Lane Centre, Castleford WF10 2LW
18 September	**Annual Open Show** *with Brian Walsh*	Derwent Hall, Darwen BB3 0DQ
16 October	**Focus on Loricariidae** *with Mark Walters*	SW England, TBA
20 November	**Autumn auction** *with Mark Walters*	Derwent Hall, Darwen BB3 0DQ
11 December	**Christmas meeting** *with Brian Walsh*	Derwent Hall, Darwen BB3 0DQ

Futher information at our online events page and Facebook group!


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