Mammalian face as an evolutionary novelty

Higashiyama, Hiroki; Koyabu, Daisuke; Hirasawa, Tatsuya; Werneburg, Ingmar; Kuratani, Shigeru; Kurihara, Hiroki

Published in: Proceedings of the National Academy of Sciences of the United States of America

Published: 02/11/2021

Document Version: Final Published version, also known as Publisher's PDF, Publisher's Final version or Version of Record

License: CC BY-NC-ND

Publication record in CityU Scholars: Go to record

Published version (DOI): 10.1073/pnas.2111876118


Citing this paper: Please note that where the full-text provided on CityU Scholars is the Post-print version (also known as Accepted Author Manuscript, Peer-reviewed or Author Final version), it may differ from the Final Published version. When citing, ensure that you check and use the publisher's definitive version for pagination and other details.

General rights: Copyright for the publications made accessible via the CityU Scholars portal is retained by the author(s) and/or other copyright owners and it is a condition of accessing these publications that users recognise and abide by the legal requirements associated with these rights. Users may not further distribute the material or use it for any profit-making activity or commercial gain.

Publisher permission: Permission for previously published items are in accordance with publisher's copyright policies sourced from the SHERPA RoMEO database. Links to full text versions (either Published or Post-print) are only available if corresponding publishers allow open access.

Take down policy: Contact lbscholars@cityu.edu.hk if you believe that this document breaches copyright and provide us with details. We will remove access to the work immediately and investigate your claim.
Mammalian face as an evolutionary novelty

Hiroki Higashiyama,a,† Daisuke Koyabub,c, Tatsuya Hirasawad,e Ingmar Werneburgf,g, Shigeru Kuratanie,h, and Hiroki Kuriharaa

Abstract

The anterior end of the mammalian face is characterized composition of a semimotile nose, not the upper jaw as in other tetrapods. Thus, the therian nose is covered ventrolaterally by the "premaxilla," and the osteocranium possesses only a single nasal aperture because of the absence of medial bony elements. This stands in contrast to those in other tetrapods in whom the premaxilla covers the rostral terminus of the snout, providing a key to understanding the evolution of the mammalian face. Here, we show that the premaxilla in therian mammals (placentals and marsupials) is not entirely homologous to those in other amniotes; the therian premaxilla is a composite of the septomaxilla and the palatine remnant of the premaxilla of nontherian amniotes (including monotremes). By comparing topographical relationships of craniofacial primordia and nerve supplies in various tetrapod embryos, we found that the therian premaxilla is predominantly of the maxillary prominence origin and associated with mandibular arch. The rostral-most part of the upper jaw in nonmammalian tetrapods corresponds to the motile nose in therian mammals. During development, experimental inhibition of primordial growth demonstrated that the entire mammalian upper jaw mostly originates from the maxillary prominence, unlike other amniotes. Consistently, cell lineage tracing in transgenic mice revealed a mammalian-specific rostral growth of the maxillary prominence. We conclude that the mammalian-specific face, the muzzle, is an evolutionary novelty obtained by overriding ancestral developmental constraints to establish a novel topographical framework in craniofacial mesenchyme.

Significance

The anatomical framework of the jaw has traditionally been thought to be highly conserved among vertebrates. However, here we show that the therian-unique face (muzzle) evolved via a drastic alteration of the common pattern of the tetrapod jaw. Through comparative morphological and developmental analyses, we demonstrated that the therian mammal's premaxilla (rostral-most upper jawbone) is derived from the maxillary prominence of the mandibular arch. The developmental primordium that produces the premaxilla in nonmammalian tetrapods rarely contributes to the upper jaw in therian mammals but rather forms a motile nose. We propose that these previously unrecognized rearrangements allowed key innovations such as the highly sensitive tactile perception and olfactory function in mammalian evolution.


The authors declare no competing interest.

This article is a PNAS Direct Submission. This open access article is distributed under Creative Commons Attribution-NonCommercial-NoDerivatives License 4.0 (CC BY-NC-ND).

† To whom correspondence may be addressed. Email: h-hiroki@m.u-tokyo.ac.jp.

This article contains supporting information online at http://www.pnas.org/lookup/suppl/doi:10.1073/pnas.2111876118/-/DCSupplemental.

Published October 29, 2021.

PNAS 2021 Vol. 118 No. 44 e2111876118

https://doi.org/10.1073/pnas.2111876118 | 1 of 8
Fig. 1. Murine “premaxilla” develops differently from premaxillae of other tetrapods. (A) The anatomy of the therian mammal’s face. (B) General scheme of craniofacial development in amniotes (10, 11, 15). (C) Three-dimensional models of tetrapod embryos. The murine premaxilla ossifies in the same topographical position as the septomaxilla (orange) of other species. The infraorbital branch (nerve branch for vibrissae) of V2 was removed in 13.5 dpc mouse. The summary is shown in D. sn, solum nasi; nid, nasolacrimal duct; V1, ophthalmic nerve; V2, maxillary nerve. (Not to scale.)

the chondrocranium (Fig. 1C). In the frog and gecko, the premaxilla grows throughout development to form the rostral end of the upper jaw, whereas in the monotreme (echidna), this bone becomes relatively smaller, and the occupying region is gradually replaced by the septomaxilla (Fig. 1C and SI Appendix, Fig. S2). In the mouse, the therian premaxilla first ossifies laterally to the nasal capsules, at the caudal lateral edge of the cartilaginous narial fenestra, adjacent to the anterior opening of the nasolacrimal duct, which corresponds to features of the septomaxilla in nontherian animals. The nontherian premaxilla arises in the innervation domain of the ophthalmic nerve (V1), whereas the murine premaxilla develops in the domain of the maxillary nerve (V2). The rostral tip of the nasal cartilage, where the nontherian premaxilla occurs, does not produce any bony elements in the mouse. The V1 distribution domain does not contribute to any parts of the upper jaw in mice but rather forms the tip of the nose (SI Appendix, Fig. S3). Thus, the topographical position of the premaxilla conspicuously differs between the mouse and the nonmammalian tetrapods, implying that the homonymous bones may differ morphologically and/or developmentally. In particular, our comparative embryological analyses raise the possibility that the murine premaxilla more closely resembles the nontherian septomaxilla (Fig. 1D).

Maxillary Prominence Contributes to the Murine Premaxilla. Given that the distribution pattern of the trigeminal nerve reflects its embryonic origin, the therian premaxilla that develops in the domain of V2 may originate from the maxillary prominence of the mandibular arch. To assess this, we conducted experiments to trace the cell lineage of the maxillary prominence using Dlx1-CreER<sup>T2</sup> mice. The Dlx1 gene is expressed in the neural crest cells in the mandibular and posterior arches but not in the frontonasal domain, allowing the distinction of the maxillary prominence from the frontonasal domain at the pharyngula stage (SI Appendix, Fig. S4) (16). In both Dlx1-CreER<sup>T2</sup> R26RLacZ<sup>+</sup> and R26RYFP mice, the mesenchyme of the maxillary prominence was successfully labeled from 9.5 to 10.5 dpc (days postcoitum) (SI Appendix, Fig. S4). The labeled cell mass protruded rostrally, eventually giving rise to the rostral-most soft tissue of the upper jaw in older embryos (Fig. 2A and B and SI Appendix, Fig. S5). The distribution of the labeled cells clearly and exclusively overlapped the innervating area of V2 (Fig. 3C and SI Appendix, Fig. S5). Furthermore, in the fetal stage, the labeled cells were found in the murine premaxilla in Dlx1-CreER<sup>T2</sup> R26RYFP mice, the chemical transgenic mouse (18), whose maxillary prominence derivatives, including the main body of the premaxilla, partial transform into the lower jaw identity (SI Appendix, Fig. S6).

Inhibition of Primordial Growth. To compare which of the facial primordia gives rise to the premaxilla and septomaxilla in different amniotes, we impeded facial development. Using chicken (Gallus gallus) and mouse embryos, we artificially constructed cleft lip and cleft palate (cleft lip/palate) conditions with cyclopamine to inhibit the neural crest mesenchymal proliferation via the suppression of the Sonic hedgehog pathway following previously reported methods (19, 20). In the late pharyngula stages, the fusions of the frontonasal and maxillary prominences were successfully inhibited both in chicken and mouse, and clefts formed between these primordia. We also observed gecko embryos that spontaneously showed cleft lip/palate conditions. The clefts of these three animals persisted in the same position throughout development, dividing the whole snout into three parts: two lateral maxillary prominence parts and the middle frontonasal part (Fig. 3A). In the chicken and gecko,
the separation coincided with both the V1–V2 boundary and the premaxilla–maxilla suture, suggesting that the premaxilla is entirely derived from the frontonasal prominence, consistent with the generally accepted view (2, 4, 10, 11). Another dermal element, the septomaxilla, was found in the maxillary process domain, caudolateral to the cleft in the gecko (Fig. 3B and SI Appendix, Fig. S7). In the mouse, the upper jaw was also divided into the medial and lateral components, each coinciding with the nerve distributions. Specifically, the medial component, or the palatine process, arises in the frontonasal domain. However, unlike the gecko and chicken, the main body of the premaxilla was found on the lateral side of the cleft, the domain of V2 (Fig. 3B and SI Appendix, Fig. S7). Thus, the therian premaxilla is not entirely a frontonasal derivative; instead, its main body is likely derived from the maxillary prominence (Fig. 3C and SI Appendix, Fig. S7B), which is equivalent to the nontherian septomaxilla. Therefore, we conclude that the therian premaxilla is a uniquely composite structure, consisting of homologs of the nonmammalian premaxilla and possibly the septomaxilla and developed from the frontonasal prominence and maxillary prominence, respectively.

Comparison of Synapsid Fossils. To understand the evolutionary history of the therian premaxilla, we examined the fossil record in the stem group of mammals, or fossil synapsids. The septomaxilla is found inside of the nostrils in many tetrapods (21–23). Indeed, septomaxilla were observed inside the nostrils of juvenile geckos and frogs (Fig. 4A). We confirmed that the septomaxilla was a small bony element in the nasal opening, which was pierced by the nasolacrimal duct in the basal synapsids [e.g., †Dimetrodon (23) and †Dicynodontoides; Fig. 4B and C]. In the Gorgonopsia and Therocephalia, which are closer to the Mammalia than †Dimetrodon, the lateral part of the septomaxilla had begun to extend onto the lateral surface of the bony snout (Fig. 4C and SI Appendix, Fig. S8) (23–25). The premaxilla and septomaxilla remain separated in most cynodonts and mammaliform lineages (26–36) (Fig. 4D). A significant change occurs in the lineage toward the Theria: the medial part of the premaxilla is lost, and a single nasal aperture appears in the osteocranium (Fig. 4D). Given that modern marsupials exhibit complete “therian premaxilla” like those of eutherians, the fusion of the septomaxilla (main body) and premaxilla (palatal process) should have occurred in the common ancestor of...
therians. Thus, the typical mammalian muzzle should have arisen in a common ancestor of the Theria.

**Discussion**

One long-lasting enigma of comparative anatomy is that the rostral-most part of the upper jaw is innervated by different nerve branches in mammals and nonmammalian tetrapods (15). In the present study, we showed that the premaxilla of nonmammalian tetrapods is not represented in the rostral-most part of the upper jaw in therian mammals but rather forms a motile nose (Fig. 5A). Developmentally, the maxillary prominences on both sides circumscribe the entire upper jaw, leading to a novel configuration of the rostral-most jawbone and the rostral distribution of maxillary vibrissae accompanied by V2 (7, 37, 38) (SI Appendix, Fig. S9A). In vertebrate embryos, craniofacial primordia develop semiautonomously as developmental modules (39), each maintaining its morphological identity and topographical relationship (40, 41) with specific cranial nerve branches. Thus, the described enigma in the innervation pattern are attributable to the rearrangement of the developmental primordia and resultant reconfiguration of the premaxilla-related structures in the rostral-most part of the upper jaw in the lineage toward therian mammals.

The suggested evolutionary changes in developmental patterns can be traced in the gradual replacement of the premaxilla and septomaxilla in the series of fossil synapsids (Fig. 5B). This sequence is partially mirrored in the development of monotremes with the gradual loss of the prenasal part of the premaxilla and in the evolutionary fusing of septomaxilla and premaxilla in therians (Fig. 1C and SI Appendix, Fig. S2), suggesting a heterochronic change as a mode of morphological evolution. In parallel, the contribution of the other pharyngeal arch elements to the facial part of the cranium, such as the establishment of maxillary vibrissae and facial muscles, also occurred in derived synapsids (SI Appendix, Fig. S9) (37, 38). Thus, the fossil evidence indicates that the developmental pattern of the craniofacial primordia had uniquely changed in the evolutionary lineage toward modern mammals. Until now, the septomaxilla was regarded either to be lost in therian mammals or to remain as the os nasale only in the xenarthran mammals (e.g., armadillos) (Fig. 4D) (23, 24, 42, 43). A few classic histological studies, however, suggested that the septomaxilla had been fused into the therian premaxilla during evolution (44).

Our results revive this old hypothesis: the main body of the therian premaxilla should be homologous with the nonmammalian septomaxilla. Indeed, the therian premaxilla develops from two separate ossification centers—the facial part and the palatal process—in many species (7, 23, 45), which is consistent with our findings.

In conclusion, the mammalian muzzle can be assigned as an evolutionary novelty (46) explained by a disruption of the ancestral morphological network (i.e., morphological homologies) and the establishment of new connections of modules. Namely, the mammalian maxillary prominence has invaded into the teeth-forming field of the rostral-most region of the upper jaw (ancestrally occupied by the premandibular domain), resulting in the unique incisor-bearing septomaxilla, an element not found in any other tetrapods. This reduction of the ancestral premaxilla and recruitment of the septomaxilla in the
Fig. 4. Evolutionary transitions of upper jaw bones in the fossil synapsids. (A) Upper jaw skeleton of the frog and gecko. (B) The snout of †Dimetrodon, which was redrawn from the previous study (23). The septomaxilla is housed in the nostril. (C) The observation of fossil synapsids. The †Silphoictidoides fossil shows a large septomaxilla covering the rostral surface of the snout, the proportions of which resemble those of the embryonic echidna (Fig. 1C and SI Appendix, Fig. S2). The phylogenetic framework is based on a previous study (25). The schemes of cynodonts were redrawn from previous studies: †Galesaurus planiceps (flipped left to right from the original figure) (26), †Massetognathus ochagaviae (27). (D) The transition of upper jaw bones in the Mammaliaforms. Some old studies identified the os nasale of the xenarthran mammals as the septomaxilla homolog, but this bone is likely a neomorph of xenarthrans as discussed in recent studies (23, 28). Similarly, the bony element (asterisk) found in the snout of the stem-eutherian †Acristatherium (29) should not be homologous with the septomaxilla. Although the “premaxilla” of the Multituberculata (dark gray) is quite similar to the therian premaxilla, it is currently impossible to exclude the possibility that the multituberculates “premaxilla” incorporated a neomorphic bone rather than a homolog of the septomaxilla without minute analyses of topographical position of the soft tissues. The schemes were redrawn from previous studies: Perameles nasuta (30), †Sinoconodon sp (31), †Haidanaodon expectatus (32), †Docofossor brachydactylus (flipped left to right from the original figure) (33), †Kryptobaatar dashzevegi (34), †Anebodon luo (35), †Necrolestes patagoniensis (28), †Acristatherium yanensis (29), and †Zalambdalestes lechei (36). The phylogenetic framework is based on refs. 26 and 29. mx, maxilla; n, nasal; pmx, premaxilla; sc, septomaxillary canal; smf, septomaxillary foramen; smx, septomaxilla. (Scale bars: 2 cm.)
mammalian upper jaw could have served as a key innovation that permitted the evolution of the semimotile nose unique to therians, adding to the expansion of facial muscles that characterize all mammals. Thus, the embryonic reconstitution of the mammalian muzzle, including the upper jaw and nose, is another novel evolutionary feature, similar to classic examples such as the mammalian middle ear (4, 47–49) or the turtle shell (50). Our present study on the therian premaxilla, or incisive bone, gives a fresh perspective on the premaxilla, whose homology has not been questioned since the dawn of comparative morphology (9). This finding provides a basis for studies of the evolution of the mammalian muzzle as well as a reconsideration of the traditional framework of craniofacial morphology of jawed vertebrates.

Materials and Methods

Laboratory Animal Specimens. Procedures involving animals were approved by the University of Tokyo Animal Care and Use Committee (approval identification: P14-109, P16-071, P19-043, and P19-050) and the guidelines of the RIKEN Center for Developmental Biology. For the present study, we used the embryos of mice, chickens, geckos, and frogs.

For mice (Mus musculus), we used two wild-type strains (C57BL/6J and ICR) as well as Dlx1-CreERT2, Rosa26R<tm1(R26R) cre/fl >, and Rosa26R<tm1(R26R) py/ py > mice. All these strains were kept in the laboratory at the University of Tokyo. The mice were housed in light-, temperature- (25 °C), and humidity-controlled conditions, and food and water were available ad libitum. We also revalidated data of Ednar<tm1(R26R) mice that were obtained in our laboratory in 2008 (SI Appendix, Fig. S6). See the following sections for details of the genetic experiments.

For chicken (Gallus gallus), we incubated fertilized eggs of the Boris Brown chicken in a humidity chamber at 38 °C, and the embryos were staged according to Hamburger and Hamilton criteria (51). For the gecko (Madagascar ground gecko, Paroedura picta), we collected fertilized eggs of the Madagascar ground gecko in 2012 from the Laboratory for Animal Resources and Genetic Engineering, RIKEN Center for Developmental Biology. They were incubated in a humidity chamber at 38 °C. The embryos were staged according to Noro et al. (52).

For the frog (Japanese brown frog, Rana japonica), we obtained fertilized eggs legally from a rice field in Shibasaki, Tsukuba, Ibaraki Prefecture, Japan, in May 2018. The eggs were kept in a tank with fresh water. We fed snail leaves to the hatched tadpoles and fixed the animals during the metamorphosis stage.

Museum Specimens. For the study of the short-beaked echidna (Tachyglossus aculeatus), we observed and took high-resolution photos of three histological sections from the Hill Collection at the Berlin Museum of Natural History: 1) Tachyglossus aculeatus MOS5, 8 greatest length (gl) p egg 14.5 mm; 2) Tachyglossus aculeatus M158, 12.5 gl late-stage p egg 17 mm; and 3) Tachyglossus aculeatus M161, 24 gl p young stage.


We also used two legally collected, alcohol-preserved specimens: 1) armadillo (Dasypus hybridus), UMUT14772, CRL 80 mm from the University Museum of the University of Tokyo and 2) echidna, ZMB echidna 81, CRL 43 mm from the Museum für Naturkunde, Berlin, Germany.

The Dlx1-CreERT2 Mouse Experiment. The B6;129S4-Ddx1<tm1(cre/ERT2)Zjh/J(Embryonic stem cell) adult mice were purchased from The Jackson Laboratory through Charles River Laboratories Japan and maintained with a mixed (C57BL/6ICR) genetic background under heterozygous mating. For cell lineage tracing, we paired Dlx1-CreERT2 mice with R26R<tm1(R26R) cre/fl > or R26R<tm1(R26R) py/ py > mice. Cre recombinase activity was induced via oral administration of tamoxifen (Sigma-Aldrich, no. T5648) in corn oil (Sigma-Aldrich, no. CB267) at a dose of 0.1 mg/kg body weight. The administration was performed once at 8.5, 9.5, and 10.5 dpc, and mice administered at 9.5 dpc were used for the analysis (SI Appendix, Fig. S5). Embryos from stages 10.5 to 17.5 dpc were collected. We tested at least four mothers at each stage.

β-galactosidase Detection. Embryos were fixed in 4% paraformaldehyde (PFA) in 0.1 M phosphate-buffered saline (PBS) with 0.2% glutaraldehyde containing 5 mM ethylene glycol tetracetic acid and 2 mM MgCl2 at 4 °C for 15 min. They were then washed with a washing buffer (2 mM MgCl2, 0.02% Nonident P-40, and 0.1% sodium deoxycholate in PBS) three times for 30 min each time. Next, embryos were preincubated in a 0.2-M kallolite phenylphosphonate (KPP) solution (a mixture of 0.1 M KH2PO4 and 0.1 M K2HPO4 to which...
was added 2 mM MgCl$_2$, 0.02% Nonidet P-40 and 0.1% sodium deoxycholate immediately before use) for 10 min and then incubated in the stain solution of 10 mM K$_2$[Fe(CN)$_6$], 10 mM K$_4$[Fe(CN)$_6$], and 1 mg/ml X-gal (Cayman Chemical, no. 16495) in KPP solution in darkness overnight at 37 °C. After staining, the samples were washed with PBS and postfixed using 4% PFA/PBS.

**Cleft Lip/Palate Preparation.** For the mouse, as described by Lipinski et al. (20), we administered cyclophamine through implantation of micro-osmotic pump. The cyclophamine (LC Laboratories, no. C-8700) was dissolved in a sodium phosphate/citrate buffer containing 2-hydropropyl-b-cyclodextrin (HPBCD) (Sigma-Aldrich) shortly before use; the amount used was 1.5 mg cyclophamine/100 μl 30% HPBCD (percentage by weight). Then, the solution was injected into the micro-osmotic pump (Alzet, no. 2001D), and the implantation was conducted at 8.25 dpc. The same cyclophamine solution was used for the chicken. We injected 10 to 30 μl solution into the pericalvaral region of stage 15 chicken embryos following the method used by Abzhanov et al. (19). For the gecko, we occasionally obtained wild-type cleft lip/palate embryos.

**Whole-Mount Immunohistochemistry.** To visualize the peripheral nerve in mice, we used monoclonal antibody 2H3 (Developmental Studies Hybridoma Bank, University of Iowa). The embryos were fixed with 4% PFA/PBS, washed and dehydrated in a graded series of methanol (70%, 95%, and stored at −30 °C). Next, they were placed into a mixture of hydrogen peroxide and methanol (1:9) for several days for depigmentation and for blocking endogenous peroxidase activities. Then, 0.5 μl 10% Triton X-100 in distilled water was added, and the embryos were further incubated for 30 min at room temperature. After washing in Tris-HCl-buffered saline solution (TST: 20 mM Tris-HCl [pH 8.0], 150 mM NaCl, 0.01% Triton X-100), the samples were blocked with 5% nonfat dried milk in TST (TSTM). The embryos were then incubated in a primary antibody solution (diluted 1/100 in spin-cleaned TSTM containing 0.1% sodium azide) for 2 to 4 d at 37 °C while being gently agitated. The secondary antibody used was horseradish peroxidase (HRP)-conjugated polyclonal goat anti-mouse (Dako, no. P0447) diluted 1/400 in TSTM. After staining, the samples were washed with PBS and postfixed using 4% PFA/PBS.

**Skeletal Preparation.** The skeletal staining of embryos was conducted using Skeletal Preparation (35% aqueous solution) for 20 to 40 min. After the incubation, the embryos were washed with PBS and postfixed using 4% PFA/PBS.

**Frozen Sections and Fluorescent Immunohistochemistry.** We acquired grayscale images of museum fetal specimens (UMUT14772 and ZMB echidna 78) using a microscope (Eclipse Ti, Nikon). The fluorescent observations of the sections were conducted using a microscope (Olympus BX51, Japan) equipped with an Olympus DP70 camera and Olympus DP controller software (Olympus). On the digitized sections, each embryonic component was identified and reconstructed using the AMIRA 3D Visualization Framework (Thermo Fisher Scientific).

**Microcomputerized Tomography Scans.** We acquired grayscale images of the specimens in the Hill Collection. We are grateful to Yukiko Kurihara and Akiyasu Iwase (the Tokyo University of Technology) for technical advice with mouse experiments and for help with keeping mice; Kaoru Mizukami and Yuka Haneada (The University of Tokyo) for helping with frog and chicken experiments; Giovanni Levi (Muséum National d’Histoire Naturelle, Paris), Masaki Takechi (Tokyo Medical Dental University), Daichi G. Suzuki (The University of Tsukuba), Takao K. Suzuki (The University of Tokyo), and Miliko Tanaka (Tokyo Institute of Technology) for technical and scientific advice; Kai Ito (Tsurumi University) and Kengo Iwai (Musashino Art University) for helping with three-dimensional reconstructions; and Yuriko Kondo (The University of Tokyo) for secretarial assistance. We acknowledge financial support from Japan Society for the Promotion of Science (JSPS) Grants 17J11177, 20H04858, 20K02385, 21K24756 (H.L.), 18H04816, 18H02092, 18K19359, and 18K00207; JSPS Joint Research Program implemented in association with German Research Foundation (DFG-LeAD with DFG) Grant JPSJR20181608; and City University of Hong Kong Start-up Grant 9610466 (D.K.); and DFG no. WE 5440/6-1 (I.W., K.K.). This work was also supported by JSPS Grant 15H02416, Grant-in-Aid for Scientific Research on Innovative Areas (Research in a Proposed Research Area) 17H06385, and a Naito Grant for the Promotion of Focus Research (S.K.) and JSPS Grant 19H01048 and Core Research for Evolutional Science and Technology of the Japan Science and Technology Agency Grant JPMJCR13W2 (H.K.).

**Data Availability.** All study data and materials are available as described in the article and/or SI Appendix.

**ACKNOWLEDGMENTS.** We greatly appreciate Peter Giere (Museum für Naturkunde, Berlin) for supporting our observation of the historical eichria sections in the Hill Collection. We are also grateful to Yukiko Kurihara and Akiyasu Iwase (the Tokyo University of Technology) for technical advice with mouse experiments and for help with keeping mice; Kaoru Mizukami and Yuka Haneada (The University of Tokyo) for helping with frog and chicken experiments; Giovanni Levi (Muséum National d’Histoire Naturelle, Paris), Masaki Takechi (Tokyo Medical Dental University), Daichi G. Suzuki (The University of Tsukuba), Takao K. Suzuki (The University of Tokyo), and Miliko Tanaka (Tokyo Institute of Technology) for technical and scientific advice; Kai Ito (Tsurumi University) and Kengo Iwai (Musashino Art University) for helping with three-dimensional reconstructions; and Yuriko Kondo (The University of Tokyo) for secretarial assistance. We acknowledge financial support from Japan Society for the Promotion of Science (JSPS) Grants 17J11177, 20H04858, 20K02385, 21K24756 (H.L.), 18H04816, 18H02092, 18K19359, and 18K00207; JSPS Joint Research Program implemented in association with German Research Foundation (DFG-LeAD with DFG) Grant JPSJR20181608; and City University of Hong Kong Start-up Grant 9610466 (D.K.); and DFG no. WE 5440/6-1 (I.W., D.K.). This work was also supported by JSPS Grant 15H02416, Grant-in-Aid for Scientific Research on Innovative Areas (Research in a Proposed Research Area) 17H06385, and a Naito Grant for the Promotion of Focus Research (S.K.) and JSPS Grant 19H01048 and Core Research for Evolutional Science and Technology of the Japan Science and Technology Agency Grant JPMJCR13W2 (H.K.).

**Three-Dimensional Imaging.** For three-dimensional reconstruction, the stained sections of the embryos were digitized using an Olympus BX60 microscope equipped with an Olympus DP70 camera and Olympus DP controller software (Olympus). On the digitized sections, each embryonic component was identified and reconstructed using the AMIRA 3D Visualization Framework (Thermo Fisher Scientific).