

1 **Conserved molecular players involved in human nose**  
2 **morphogenesis underlie evolution of the exaggerated snout**  
3 **phenotype in cichlids**

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## 1 **Abstract**

2 Instances of repeated evolution of novel phenotypes can shed light on the conserved molecular  
3 mechanisms underlying morphological diversity. A rare example of an exaggerated soft tissue  
4 phenotype is the formation of a snout flap in fishes. This tissue flap develops from the upper  
5 lip and has evolved in one cichlid genus from Lake Malawi and one genus from Lake  
6 Tanganyika. To investigate the molecular basis of snout flap convergence, we used mRNA  
7 sequencing to compare two species with snout flap to their close relatives without snout flaps  
8 from each lake. Our analysis identified 201 genes that were repeatedly differentially expressed  
9 between species with and without snout flap in both lakes, suggesting shared pathways, even  
10 though the flaps serve different functions. Shared expressed genes are involved in proline and  
11 hydroxyproline metabolism, which have been linked to human skin and facial deformities.  
12 Additionally, we found enrichment for transcription factor binding sites at upstream regulatory  
13 sequences of differentially expressed genes. Among the enriched transcription factors were  
14 members of the FOX transcription factor family, especially *foxf1* and *foxa2*, which showed an  
15 increased expression in the flapped snout. Both of these factors are linked to nose  
16 morphogenesis in mammals. We also found *ap4* (*tfap4*), a transcription factor showing reduced  
17 expression in the flapped snout with an unknown role in craniofacial soft tissue development.  
18 As genes involved in cichlid snout flap development are associated with human mid-line facial  
19 dysmorphologies, our findings could hint at the conservation of genes involved in mid-line  
20 patterning across distant evolutionary lineages of vertebrates, although further functional  
21 studies are required to confirm this.

22  
23 **Key words:** RNA-seq, Lake Malawi, Lake Tanganyika, snout flap, cichlids, functional  
24 conservation

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## 2 **Significance statement**

3 The study of the evolution of similar physical traits across taxa can give insight into the  
4 molecular architecture underlying shared phenotypes. This has mostly been studied in bony  
5 structures, while soft tissue traits have been less intensely covered. We investigated the  
6 exaggerated snout in cichlid species from Lake Malawi and Lake Tanganyika and found that  
7 many genes involved in the development of the snout flap and are also associated with mid-  
8 line dysmorphologies in humans, implying a conservation across distant vertebrate lineages.

9

## 10 **Introduction**

11 The repeated evolution of phenotypes, reflecting particular ecological specializations, is a  
12 ubiquitous characteristic of adaptive radiations (Schluter & Nagel 1995; Losos et al. 1998;  
13 Rundle et al. 2000; Rüber et al. 1999). Cichlid adaptive radiations from the East African Great  
14 lakes display an impressive array of repeated morphological traits (Kocher et al. 1993),  
15 including a few dramatic examples of exaggerated phenotypes like the overgrowth of  
16 craniofacial soft tissues in various anatomical regions such as lips (Machado-Schiaffino et al.  
17 2014; Manousaki et al. 2013; Colombo et al. 2013; Baumgarten et al. 2015; Lecaudey et al.  
18 2019; Henning et al. 2017), the frontal head (nuchal hump) (Lecaudey et al. 2021) and the nose  
19 snout (or nose flap) (Concannon & Albertson 2015; Conith et al. 2018). Although there is  
20 increasing insight into the evolution of such phenotypic novelties, especially regarding  
21 hypertrophid lips, exaggerated soft tissue traits are less well studied than bony traits and the  
22 genetic mechanisms underlying these traits are not entirely understood. Comparative  
23 approaches can shed light on the genetic mechanisms that reconfigure the body plan and give  
24 rise to such complex traits. With examples of both parallel and non-parallel mechanism  
25 underlying cases of repeated evolution (e.g. Manousaki et al. 2013; Colombo et al. 2013) of

1 phenotypic novelties such comparisons can thus also help us to understand the molecular  
2 mechanisms that shape morphological diversity.

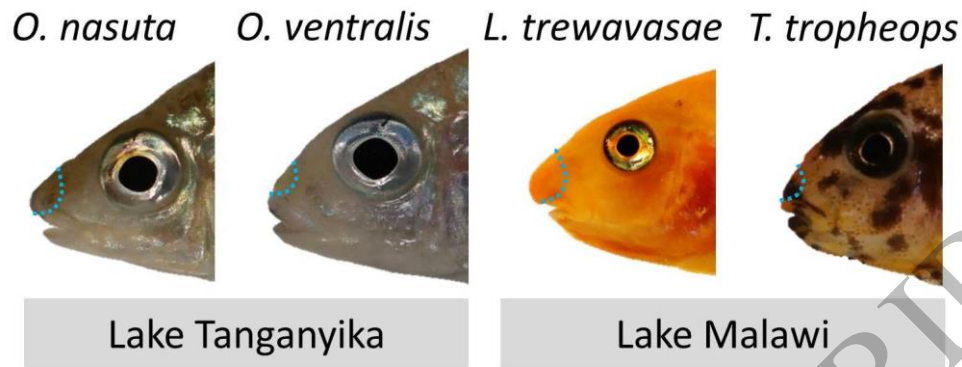
3 One of these repeated exaggerated phenotypes in cichlids is the snout flap, a  
4 pronounced projection that emanates from a flap of fibrous tissue just above the upper lip. It is  
5 a rare morphological innovation that has only evolved in two tribes of cichlid fishes from East  
6 Africa, the modern Haplochromines in Lake Malawi and the Ectodini in Lake Tanganyika  
7 (Concannon & Albertson 2015). When this snout is sexually monomorphic, it is thought to be  
8 a trophic adaptation that improves feeding efficiency (Konings 2007). When the snout is  
9 sexually dimorphic, it is hypothesised to be involved in sexual selection (Konings 2007;  
10 Concannon & Albertson 2015). The cichlid snout flap has been studied at the molecular level  
11 only in the genus *Labeotropheus* from Lake Malawi where it is sexually monomorphic and  
12 functions as a trophic adaptation to efficiently leverage algae from rocks (Concannon &  
13 Albertson 2015; Conith et al. 2018). A similar snout structure has also been described in two  
14 species from the Ectodini tribe (*Ophthalmotilapia nasuta* and *Asprotilapia leptura*) from Lake  
15 Tanganyika. In *A. leptura* it is sexually monomorphic and likely involved in increased foraging  
16 efficiency (similar to *Labeotropheus*), whereas in *O. nasuta* it is only found in mature males  
17 and is likely a secondary sexual character (Hanssens et al. 1999; Conith et al. 2019). Thus, the  
18 exaggerated snout is a convergent phenotype that evolved independently in two cichlid lineages  
19 that diverged  $> 9$  MYA (Irisarri et al. 2018; Conith et al. 2019).

20 In *Labeotropheus*, the snout is evident histologically by the time the yolk is absorbed  
21 and exogenous feeding occurs (~1 month post-fertilization) (Conith et al. 2018; Concannon &  
22 Albertson 2015), and the early formation and growth of the snout is linked to the transforming  
23 growth factor beta (TGF $\beta$ ) signalling pathway (Conith et al. 2018). However, it remains unclear  
24 which (1) genes and pathways contribute to the maintenance of this complex trait and if (2)  
25 these candidate genes and pathways can be linked to more conserved patterning of craniofacial

1 features. Furthermore, while previous research focused on the TGF $\beta$  signalling pathway, a  
2 more extensive molecular interaction map of the formation and maintenance of this  
3 exaggerated phenotype remains to be unravelled. A transcriptome-wide overview is  
4 particularly important since it is well-known that there is molecular cross-talk between the  
5 TGF $\beta$  signalling pathway and several other pathways which all play a pivotal role in  
6 craniofacial morphogenesis and adaptive evolutionary divergence in teleost fishes (Ahi 2016).

7 In this study, we set out to investigate the molecular mechanisms that underlie the  
8 formation and evolution of the exaggerated snout phenotype, in two non-sister cichlid lineages  
9 from lakes Tanganyika and Malawi (Figure 1) and link it to conserved molecular players in  
10 mid-line patterning. We compared two species that develop a snout; (1) *Labeotropheus*  
11 *trewavasae* (tribe Haplochromini) from Lake Malawi and (2) *Ophthalmotilapia nasuta* (tribe  
12 Ectodini) from Lake Tanganyika (Figure 1). As controls, we used two closely related species  
13 within each tribe that do not develop such a structure; (1) the Lake Malawi mbuna species  
14 *Tropheops tropheops* (Haplochromini) and (2) the Lake Tanganyika featherfin cichlid  
15 *Ophthalmotilapia ventralis* (Ectodini) (Figure 1). We used mRNA-sequencing to quantify gene  
16 expression differences between the exaggerated snout and non-snout tissues for each lake.  
17 Altogether, we identified parallel and non-parallel molecular mechanisms that underlie the  
18 evolution of the snout flap in Lake Malawi and Lake Tanganyika cichlids. Our study design  
19 provides valuable information on conserved regulatory mechanisms underlying the  
20 morphogenesis of a unique hypertrophic facial soft tissue in cichlids, which also exhibit  
21 striking similarity to those mechanisms driving craniofacial development and mid-line  
22 patterning in other vertebrates including humans. Notably, cichlids are already introduced as  
23 excellent models to study craniofacial skeletal deformities in humans (Powder & Albertson,  
24 2016), and our study can be one of the first indications that cichlids can be used as models to  
25 study deformities in facial soft tissues as well.

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3 **Fig.1. Convergent cases of snout flap evolution.** East African cichlid species used in this study. The  
4 area of the soft tissue that was dissected is depicted by blue dashed lines. (*O. nasuta*)  
5 *Ophthalmotilapia nasuta*, (*O. ventralis*) *Ophthalmotilapia ventralis*, (*L. trewavasae*) *Labeotropheus*  
6 *trewavasae*, (*T. tropheops*) *Tropheops tropheops*.

## 7 **Results**

8 To investigate molecular mechanisms underlying the formation of a snout flap in two distant  
9 lineages of cichlids, we dissected the snout tissue of five biological replicates per species,  
10 which entailed the area above the upper lip including the nostrils. These tissue samples  
11 consisted of epidermis, dermis and the underlying connective tissue (Figure 1). Subsequently  
12 we extracted RNA of these five samples per species to quantify gene expression differences.

13

### 14 **RNA-sequencing, gene expression and downstream analyses**

15 The RNA-sequencing resulted in between 6.7 and 15.8 million reads per sample and after  
16 filtering of low-quality reads, between 4.6 and 11.1 million reads were retained for each sample  
17 (Supplementary Table S1). The raw data of sequence reads have been deposited in the  
18 Sequencing Read Archive (SRA; Supplementary Table 1) of NCBI (accession number:  
19 PRJNA770252). The final annotation of all merged species included 33,251 genes. Through  
20 pairwise comparisons between species of each lake radiation we identified 832 of the 33,251

1 genes (2.4%) with significant differential expression (FDR cut-off at  $P < 0.05$ ) for the  
2 comparison of *O. nasuta* versus *O. ventralis*, while the comparison between *L. trawavasae*  
3 versus *T. tropheops* yielded 4,292 (12.7%) significant differentially expressed genes (FDR cut-  
4 off at  $P < 0.05$ ).

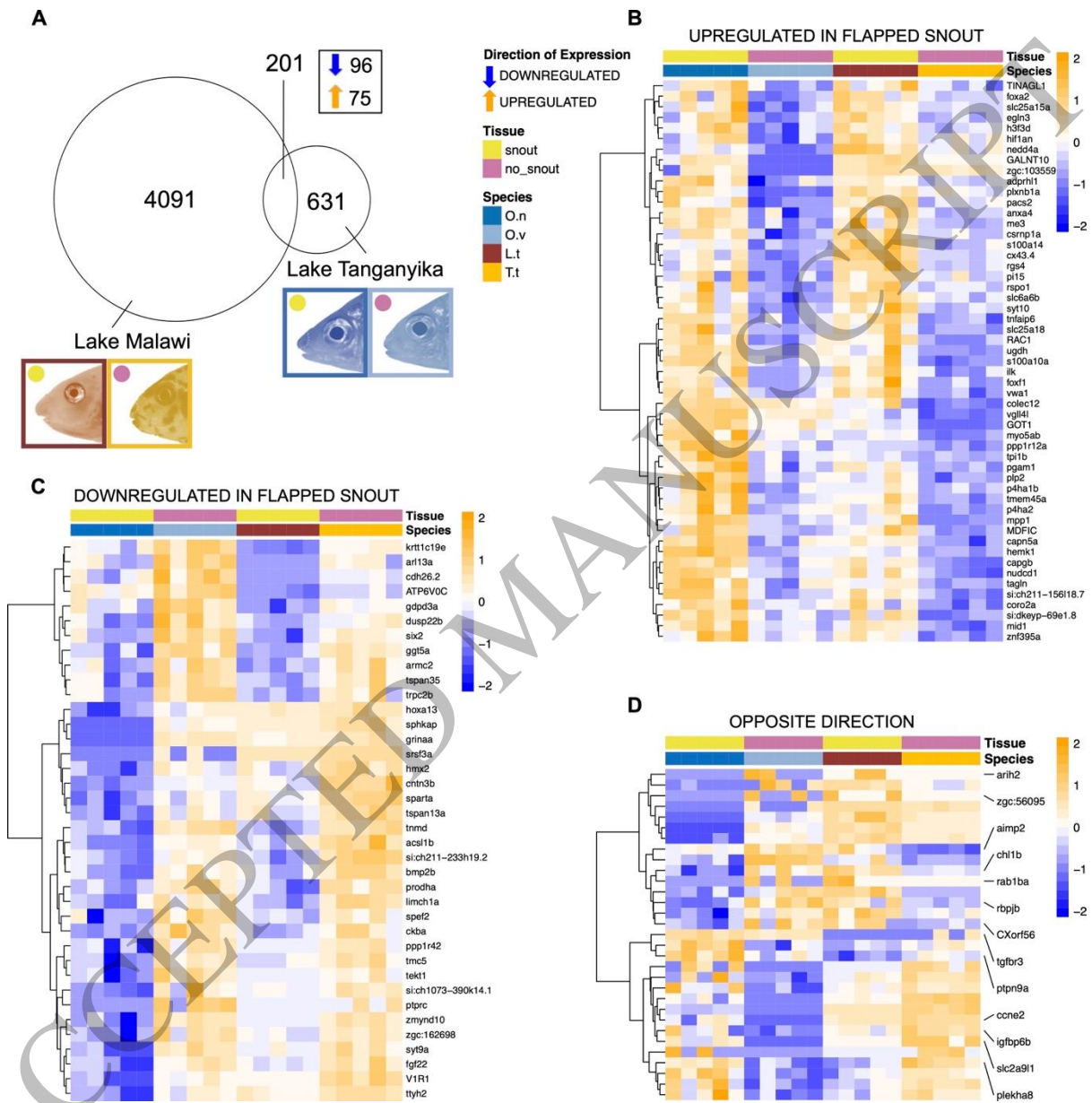
5 GO enrichment analysis conducted for differentially expressed genes within each species pair  
6 comparison for Lake Tanganyika and Lake Malawi respectively revealed the involvement in  
7 biological processes like ‘peptidyl-proline modification’, ‘tendon development’ and ‘cell  
8 adhesion’ for the comparison of the Lake Tanganyika species (*O. nasuta* versus *O. ventralis*),  
9 while the Lake Malawi comparison (*L. trewavasae* versus *T. tropheops*) showed terms like  
10 ‘cell matrix adhesion’, ‘apoptotic process involved in morphogenesis’ as well as ‘regulation of  
11 brown fat cell differentiation’ amongst more cell specific processes (Supplementary Table S2).

12 To understand if similar genes were involved in the formation of a snout across the two  
13 lakes, we investigated the intersection set of the two pairwise comparisons and could identify  
14 an overlapping list of 201 differentially expressed (DE) genes which were distinct between the  
15 flapped snout versus the non-flapped snout regions in both lakes (24.2% of the differentially  
16 expressed genes (DEG) for the Lake Tanganyika comparison and 4.8% for the Lake Malawi  
17 comparison) (Figure 2A) (Supplementary Table S3). Among the shared DE genes, 84.6%  
18 showed the same direction of expression with 74 genes being upregulated and 96 genes being  
19 downregulated in the flapped snout tissues in both comparisons which is a higher number of  
20 shared expression direction than one would expect by chance (Hypergeometric test,  $P < 0.05$ ),  
21 whereas 31 genes showed expression differences in opposite directions across the comparisons  
22 for each Lake (Figure 2B-D). The heatmap clustering of the DE genes showed that there are at  
23 least two major branches in each group of up- or down-regulated gene sets, while the clustering  
24 of the DE genes with opposite expression pattern also revealed the presence of two major  
25 branches (Figure 2B-D). These clustering structures indicate distinct transcriptional regulations



1 within each group which potentially originated from the effects of different upstream  
 2 regulators.

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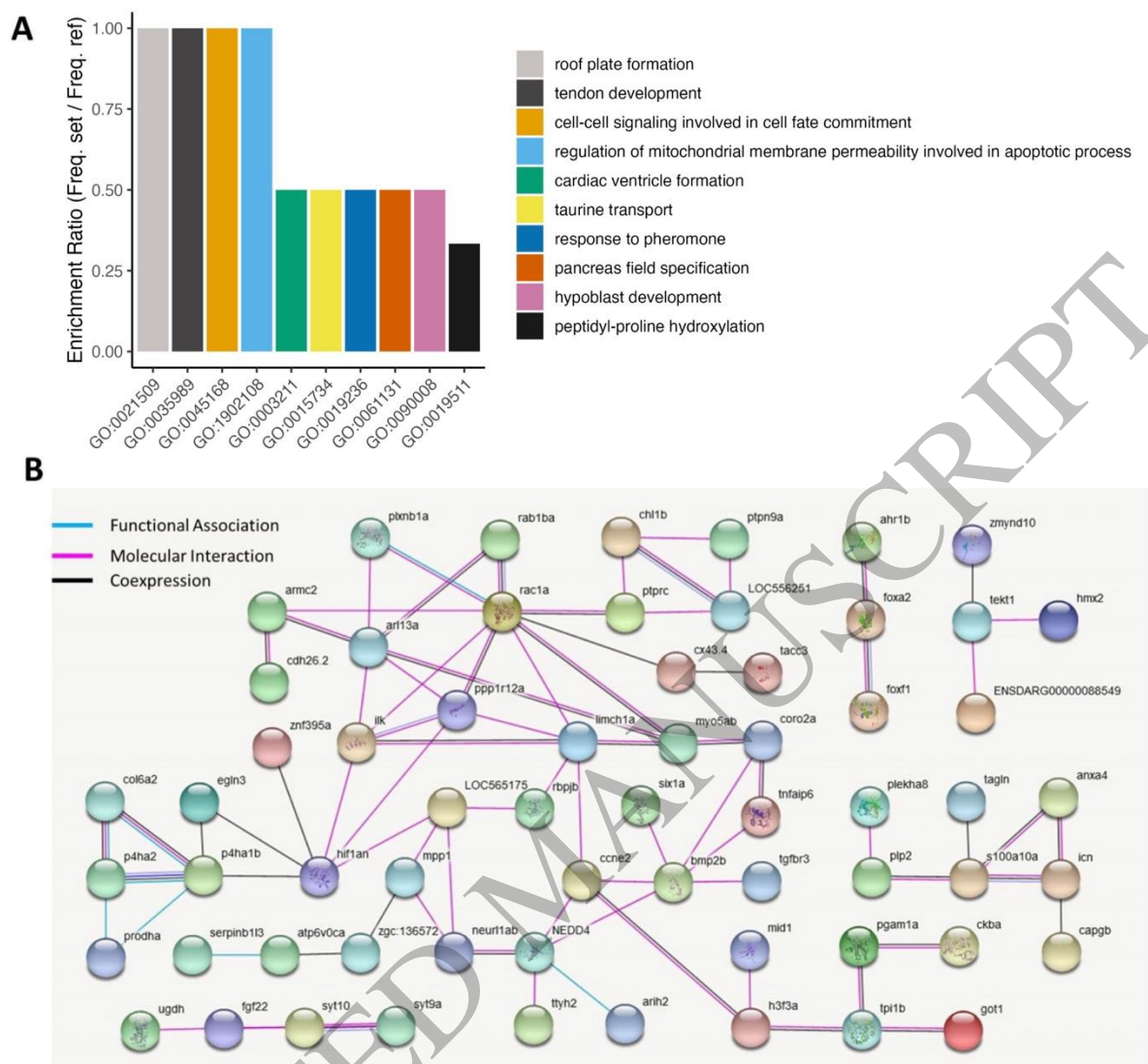
5 **Fig. 2. Differentially expressed genes in the snout regions.** (A) Venn diagram of genes with  
 6 differential expression between the snout regions ("snout" and "no snout") for Lake Malawi and Lake  
 7 Tanganyika and the overlap of 201 genes between the two comparisons of which 96 are downregulated  
 8 and 75 are upregulated in the flapped snout of both comparisons. Dendrogram clusters of the  
 9 overlapping annotated genes showing upregulation (B), and downregulation (C) in expression in the  
 10 flapped snout tissue, as well as those showing differential expression in both comparisons but in



1 opposing directions (including not annotated genes) (D). Orange and blue shadings indicate higher and  
2 lower relative expression respectively. Lake Tanganyika: *Ophthalmotilapia nasuta* (O.n, dark blue),  
3 *Ophthalmotilapia ventralis* (O.v; light blue); Lake Malawi: *Labeotropheus trewavasae* (L.t; red),  
4 *Tropheops tropheops* (T.t; orange).

5  
6 We performed gene ontology enrichment analysis using the list of the shared 201 DE  
7 genes as the input, and the result showed significant enrichment of GO terms for several  
8 biological processes such as amino acid metabolism (particularly proline related metabolic  
9 processes), ‘tendon development’, ‘positive regulation of BMP signaling pathway’ and cell  
10 adhesion and cell fate (Supplementary Table S2). When dividing the genes in their direction of  
11 expression in the snout flap, GO enrichment for upregulated genes was associated with  
12 ‘peptidyl-proline hydroxylation’, ‘tendon development’, ‘muscle attachment’, ‘endothelial cell  
13 development’, ‘negative regulation of Notch signaling pathway’ and although not significantly  
14 ‘positive regulation of Wnt signaling pathway’. The downregulated genes were involved in a  
15 lot of terms related to cell fate commitment and negative regulation of cell fate as well as  
16 ‘proline catabolic process’ and ‘positive regulation of BMP signaling pathway’  
17 (Supplementary Table S2).

18



1  
2 **Fig. 3. Functional analyses of the overlapping differentially expressed genes in the flapped**  
3 **snout.** (A) Enrichment for gene ontologies of biological processes using the shared 201 differentially  
4 expressed genes. (B) Functional interactions between the differentially expressed genes predicted  
5 based on zebrafish databases in STRING v10 (<http://string-db.org/>).

6  
7 We also applied the same list of the shared 201 DE genes for interactome analysis which  
8 demonstrated a large, interconnected network of genes with molecular and functional  
9 associations. Some of the genes in the network formed an interaction hub with the highest level  
10 of associations (based on the number of predicted interactions with other DE genes) with other  
11 genes such as *bmp2b*, *hif1an* and *rac1a*, suggesting their more pivotal role in the formation of

1 the flapped snout structure in cichlids (Figure 3B). Furthermore, we conducted TF binding  
 2 motif overrepresentation analysis on the upstream regulatory sequences of the DE genes  
 3 through MEME tool (Bailey et al. 2009). In total, seven motifs were enriched on the upstream  
 4 regulatory sequences of at least 40 out of 201 DE genes (Table 1). Next, we checked the  
 5 similarities of the enriched motifs with known TF binding sites in vertebrates and at least 11  
 6 TF candidates were identified to potentially bind to those motifs.

7  
 8 **Table 1. Predicted motifs and upstream regulators potentially binding to them.** Enriched motifs  
 9 on upstream regulatory sequences of the DE genes are presented in degenerated sequence format. PWD  
 10 IDs indicate positional weight matrix ID of predicted binding sites and E-values refer to matching  
 11 similarity between the predicted motif sequences and the PWD IDs. The count implies the number of  
 12 genes containing the predicted motif sequence on their regulatory region.

TF binding site	PWM ID	Count	Predicted motif sequence	E-value
FOXP1	M00987	71 / 201	AMAMACAMAMAMAMACACACAMAMACA	3.85E-12
FOXJ1	M00742			3.52E-08
RREB1	MA0073.1			1.87E-07
FOXJ1	M00742	47 / 201	AAAAASAAAMAAAMWMWCWKT	8.69E-10
FOX	M00809			9.15E-07
FOXD3	MA0041.1			3.95E-07
SP1	MA0079.2	41 / 201	CHCCYCCYCCYCCSCYCTCCY	1.12E-08
IRF9	M00258	61 / 201	KTTTTTYTTTTYYCWK	2.90E-09
MEF2	M00405	72 / 201	RTTAAAAAAA	4.28E-08
AP4	M00927	93 / 201	CWGCTGCWGCTKSTS	7.38E-08
HEB/tcf12	M00698	66 / 201	NYYCTGCTGD	1.03E-06

### 13 14 **Expression analysis by qPCR**

15 Validation of DE genes from RNA-seq was accomplished via quantitative RT-PCR (qPCR),  
 16 normalised to stably expressed reference genes (Kubista et al. 2006). In our previous studies  
 17 of East African cichlids, we found that validation of reference gene(s) is an essential step as  
 18 genes only selected from literature are not necessarily the best choice and can vary a lot  
 19 between different species and tissue types (Ahi, Singh, et al. 2019; EP. Ahi et al. 2020; E P.

1 Ahi et al. 2020; Pashay Ahi & Sefc 2018; Ahi, Richter, et al. 2019). We chose six candidate  
 2 reference genes with a small log<sub>2</sub> fold change and the lowest coefficient of variation (CV)  
 3 throughout all the samples (Supplementary Table S4). Based on the rankings by the three  
 4 software tools, BestKeeper, geNorm and NormFinder, only one of the candidate reference  
 5 genes, *pak2b*, showed consistent stability, i.e. always ranked among top two most stable  
 6 reference genes (Table 2). Thus, we used the C<sub>q</sub> value of *pak2b* in each sample to normalize  
 7 the relative gene expression levels of our target genes.

8  
 9 **Table 2. Ranking of reference genes in the nose tissue samples using three different algorithms.**

BestKeeper				geNorm		NormFinder	
Ranking	SD	Ranking	r	Ranking	M	Ranking	SV
<i>sp3</i>	0.461	<i>pak2b</i>	0.94	<i>pak2b</i>	0.369	<i>nup58</i>	0.310
<i>pak2b</i>	0.471	<i>pphln1</i>	0.931	<i>flot2a</i>	0.386	<i>pak2b</i>	0.408
<i>flot2a</i>	0.491	<i>flot2a</i>	0.926	<i>pphln1</i>	0.397	<i>pphln1</i>	0.443
<i>nup58</i>	0.509	<i>vps26a</i>	0.916	<i>sp3</i>	0.418	<i>flot2a</i>	0.498
<i>vps26a</i>	0.551	<i>nup58</i>	0.889	<i>nup58</i>	0.427	<i>sp3</i>	0.518
<i>pphln1</i>	0.587	<i>sp3</i>	0.887	<i>vps26a</i>	0.428	<i>vps26a</i>	0.646

10

11 Abbreviations: SD = Standard deviation, r = Pearson product-moment correlation coefficient, SV = stability  
 12 value, M = M value of stability.

13

14 Among the DE genes identified by RNA-seq, we chose 12 genes with a known role in  
 15 nose morphogenesis and/or other related functions in craniofacial development mainly based  
 16 on genetic studies in humans (Table 3), together with eight predicted upstream TFs (including  
 17 *ap4*, *foxd3*, *foxl1*, *foxp1*, *irf9*, *mef2a*, *rreb1a* and *sp1*) for qPCR analysis (Figure 4).

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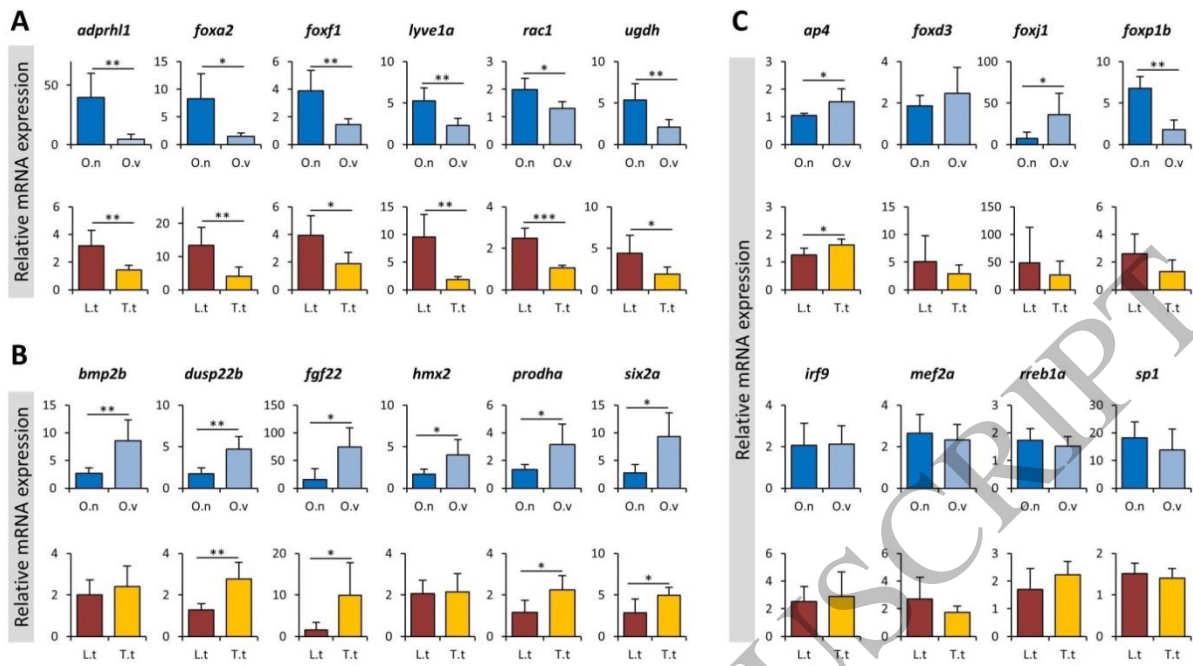
19 **Table 3. A selected set of differentially expressed genes in the flapped snout regions of studied**  
 20 **cichlids with known related functions in nose morphogenesis in mammalian models.**

Gene	Related function	Organism	References
<i>adprhl1</i>	Duplication of this gene is associated with prominent forehead, short and bulbous nose, and broad philtrum	Human	(De Pater et al. 2005)

<i>bmp2</i>	A ligand of the TGF $\beta$ signaling and its monoallelic deletion is associated with short upturned nose and long philtrum	Human	(Tan et al. 2017)
<i>dusp22</i>	Deletion at terminal end of this gene is associated with saddle shape nose morphogenesis	Human	(Hosono et al. 2020)
<i>fgf22</i>	Genomic rearrangement encompassing this gene is associated with elongation of nose with prominent nasal bridge	Human	(Quigley et al. 2004)
<i>foxa2</i>	Both deletion and missense variation in this gene causes hallow nasal bridge, short upturned nose and downturned nasolabial folds	Human	(Dines et al. 2019)
<i>foxf1</i>	Duplication and triplication causes bulbous nose and wide nasal bridge	Human	(Kucharczyk et al. 2014)
<i>hmx2</i>	Hemizygous deletion in this gene causes broad nasal bridge and prominent nose	Human	(Miller et al. 2009)
<i>lyve1</i>	Dysregulation of this gene is associated with cutaneous angiosarcoma on the nose	Human	(Mitteldorf et al. 2018)
<i>prodh</i>	Deletion and/or missense mutations in this gene causes frontal bossing, thin upper lip and short nose	Human	(Guilmatre et al. 2010)
<i>rac1</i>	Loss of function mutation in this gene causes failure in fusion of medial nasal processes and prominent nasal bridge	Human Mouse	(Thomas et al. 2010; Reijnders et al. 2017)
<i>six2</i>	Deletion in this gene causes frontonasal dysplasia syndrome in human with nasal clefting and broad nasal tip, and developmental deformities in nasal bridge in mouse	Human Mouse	(Hufnagel et al. 2016; Okello et al. 2017)
<i>ugdh</i>	Missense mutation in this gene causes bulbous nose and smooth philtrum	Human	(Alhamoudi et al. 2020)

1  
2 Based on the RNA-seq results, six of these candidate genes displayed upregulation in  
3 expression in the flapped snout (*adprhl1*, *foxa2*, *foxf1*, *lyve1a*, *rac1* and *ugdh*), while the six  
4 other candidate genes (*bmp2b*, *dusp22b*, *fgf22*, *hmx2*, *prodh* and *six2a*) showed a  
5 downregulation in expression in the flapped snout. The results of qPCR analysis confirmed that  
6 almost all of the genes showed expression patterns similar to RNA-seq results, except for  
7 *bmp2b* and *hmx2* which showed no significant difference between the snout regions of L.t and  
8 T.t. Among the predicted TFs only *ap4* showed consistent differences across both comparisons  
9 displaying a slightly reduced expression in both species with protruded snouts (O.n and L.t).  
10 This indicates potential transcriptional repressor effects of *ap4* on the downstream genes in the  
11 hypertrophic snout region. Two predicted members of FOX transcription factors, *foxj1* and  
12 *foxp1*, showed expression differences but only in one of the comparisons (O.n vs O.v), which  
13 makes them unlikely candidates for upstream regulators of shared DEGs in both comparisons.  
14 Altogether, the qPCR results demonstrate consistency between RNA-seq and qPCR results  
15 confirming the validity of our transcriptome data analysis.

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**Fig. 4. qPCR expression analysis of a selected set of candidate genes.** qPCR validation of

3

expression differences for selected sets of genes showing upregulation (A) or downregulation (B) in

4

snout tissues. (C) qPCR expression analysis of predicted transcription factors. The bars indicate mean

5

and standard deviation of RQ expression values for five biological replicates per species. The

6

asterisks above the bar represent significant expression differences (\* $P < 0.05$ ; \*\* $P < 0.01$ ;

7

\*\*\* $P < 0.001$ ). *Ophthalmotilapia nasuta* (O.n; dark blue), *Ophthalmotilapia ventralis* (O.v; light

8

blue), *Labeotropheus trewavasae* (L.t; red), *Tropheops tropheops* (T.t; orange).

## 9 Discussion

10

Cases of repeated morphological evolution in recurrent diversification events can contribute

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significantly to our understanding of the molecular architecture underlying shared phenotypes.

12

The snout flap of *L. trewavasae* is thought to have evolved under natural selection (Concannon

13

& Albertson 2015), as it plays a distinct role in the foraging efficiency for algal scraping

14

(Konings 2007; Conith et al. 2019). Additionally, no difference in snout flap size has been

15

detected between male and female of *Labeotropheus* and its formation has been shown to

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coincide with the time point of independent foraging, further supporting its function

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(Concannon & Albertson 2015). In contrast, only *O. nasuta* males show the distinct snout flap,



1 implying a role in mate choice (Concannon & Albertson 2015). Furthermore, both sexes of *O.*  
2 *nasuta* are planktivorous suction feeders, a feeding adaptation that is presumably not enhanced  
3 by a snout flap, although the snout of males continues to grow with increasing age (Hanssens  
4 et al. 1999). In a comparison of tissue types of the snout flap it has been found that the snout  
5 of *Labeotropheus fuelleborni* contains a lot more of intermaxillary ligament and loose  
6 connective tissue (80%) than the snout of *O. nasuta* (50%) (Conith et al. 2019). The  
7 morphological similarity of the snout flap across two cichlid radiations allows us to investigate  
8 if conserved molecular players are involved in the formation of snout, even if the morphologies  
9 possess different functions and differ in tissue composition and life-history.

10 We found quite differing numbers of DE genes between the comparisons within Lake  
11 Malawi and Lake Tanganyika, with roughly five times more differentially expressed genes  
12 between the chosen species pair from Lake Malawi over the species pair from Lake  
13 Tanganyika. This, most likely, can not be explained by the use of differing genera for the Lake  
14 Malawi comparison as the species flock shows a low sequence divergence of 0.1% - 0.25%  
15 probably due to the young age of the radiation (Malinsky et al. 2018) (Supplementary Fig. S1),  
16 but could be an indicator for the aforementioned difference in tissue composition of *L.*  
17 *fuelleborni* (Conith et al. 2019). Additionally, the GO enrichment analyses for the two within  
18 lake comparisons showed quite distinct enrichment terms. GO enrichment analysis for DE  
19 genes within the Lake Malawi comparison between *L. trewavasae* and *T. tropheops* yielded  
20 terms like ‘cell-matrix adhesion’, ‘regulation of brown fat cell differentiation’ and ‘apoptotic  
21 process involved in morphogenesis’ among terms involved in nerve development and different  
22 terms not readily connected to snout morphology (Supplementary Table S2). This could hint  
23 at a stunning difference in organization of connective tissue in *Labeotropheus* compared to  
24 *Tropheops*. Conith et al. 2018a found that the connective tissue (identified as the intermaxillary  
25 ligament) of *Labeotropheus*, which is high in collagen, invades the surrounding loose

1 connective tissue and anchors to the epithelium potentially helping with the stiffness of the  
2 snout to improve foraging. The GO enrichment for DE genes between *O. nasuta* and *O.*  
3 *ventralis* revealed terms linked to cell fate and cell shape regulation, ‘peptidyl-proline  
4 modification’ and ‘tendon development’. This suggests a difference in collagen/tendon  
5 development and cell adhesion and fate between the two Lake Tanganyika species, where the  
6 structure is not as unique as in *Labeotropheus*, and shows an overall increase in the proportion  
7 of skin and other tissue, much greater than in *Labeotropheus* (Conith et al. 2018a).

8 To understand if similar molecular mechanisms underly these seemingly similar  
9 phenotypes (yet different histologically) across both lakes, we looked at the intersection set of  
10 both comparisons and found many of DE genes, both upregulated and downregulated, that are  
11 associated with craniofacial development and involved in human dysmorphologies, many with  
12 mid-line facial defects including those that effect the nose in literature. Among the upregulated  
13 genes with related functions were *adprhl1* (De Pater et al. 2005), *angptl2* (Ehret et al. 2015),  
14 *colec12* (Zlotina et al. 2016), *cx43* (McLachlan et al. 2005), *foxa2* (Dines et al. 2019), *foxf1*  
15 (Kucharczyk et al. 2014), *galnt10* (Starkovich et al. 2016), *got1* (Tomkins et al. 1983), *lyve1*  
16 (Mitteldorf et al. 2018), *mdfic* (Kosho et al. 2008), *mid1* (Preiksaitiene et al. 2015)(Hüning et  
17 al. 2013), *nudcd1* (Selenti et al. 2015), *pacs2* (Holder et al. 2012), *plxnb1* (Haldeman-Englert  
18 et al. 2009), *rac1* (Thomas et al. 2010)(Reijnders et al. 2017), *rspo1* (Wieacker & Volleth  
19 2007), *s100a10* (Sawyer et al. 2007), *slc25a18* (Chen et al. 2013), *slc6a6* (Kariminejad et al.  
20 2015), *ugdh* (Alhamoudi et al. 2020), *vgl14* (Czeschik et al. 2014)(Barrionuevo et al. 2014),  
21 and *vwa1* (Giannikou et al. 2012). Among the downregulated genes we also found the  
22 following candidates to have such roles; *acs11* (Yakut et al. 2015), *adgb* (Alazami et al. 2016),  
23 *arl13* (Brugmann et al. 2010), *ATP6v0c* (Mucha et al. 2019; Tinker et al. 2021), *bmp2* (Tan et  
24 al. 2017), *cntn3* (Țuțulan-Cuniță et al. 2012), *dusp22* (Hosono et al. 2020)(Martinez-Glez et al.  
25 2007), *fgf22* (Quigley et al. 2004), *gdpd3* (Dell’Edera et al. 2018), *grina* (Bonaglia et al. 2005),

1 *hmx2* (Miller et al. 2009), *hoxa13* (Fryssira et al. 2011), *il23r* (Rivera-Pedroza et al. 2017),  
2 *ppp1r42* (Mordaunt et al. 2015), *prodh* (Guilmatre et al. 2010), *six2* (Hufnagel et al.  
3 2016)(Okello et al. 2017), *srsf3* (Pillai et al. 2019), *syt9* (Sofos et al. 2012), and *trpc2* (Sansone  
4 et al. 2014)(Zhang et al. 2010). Interestingly, one of the downregulated genes, *pi15*, is known  
5 as an important molecular player in beak formation in birds (Nimmagadda et al. 2015). Even  
6 among the overlapping DE genes which showed opposing expression patterns between the two  
7 comparisons, we still found at least four genes to have been associated with craniofacial mid-  
8 line defects in other vertebrates, including *ccne2* (Jain et al. 2010), *plekha8* (Schulz et al. 2008),  
9 *rab1b* (Alwadei et al. 2016), *RBPJ* (Nakayama et al. 2014) and *tgfbr3* (Lopes et al. 2019). The  
10 most likely explanation for opposing expression of these genes can be the existence of  
11 bimodality in their expression pattern. Bimodality of gene expression is a mechanism  
12 contributing to phenotypic diversity (Ochab-Marcinek & Tabaka 2010), and it can be reflected  
13 by up- or down-regulation of a gene during the same biological process. This regulatory  
14 bimodality can have various causes such as differential/opposing action of transcription factors  
15 (e.g. negative feedback loop), post-transcriptional factors (e.g. microRNA and circular RNA)  
16 and stochastic events. Interestingly, a highly conserved negative feedback loop in Notch  
17 signaling has already been shown to be mediated by opposing roles of RBPJ (Tanigaki & Honjo  
18 2010). This indicates a potential bimodality of *rbpj* expression through a negative feedback  
19 loop in regulation of Notch signaling, which plays a crucial role in the formation of the mid  
20 line structures including nasal structures (Tanigaki & Honjo 2010). Including developmental  
21 time series for expression profiling in future studies can help to identify whether the shared  
22 DEGs with opposing expressions also show bimodality in their expression.

23 These findings demonstrate that similar sets of genes are involved in mid-line patterning and  
24 growth across evolutionary distant vertebrates. Thus, functional studies investigating their  
25 specific role in divergent morphogenesis of snout structures in fish can provide valuable

1 information about the conserved molecular mechanisms underlying the formation of facial soft  
2 tissues (Powder & Albertson 2016). Moreover, future studies with developmental time series,  
3 histological analyses, species crossing (particularly for the species of Lake Malawi) and  
4 female *O. nasuta* are required to explore underlying mechanisms of potential convergent  
5 evolution, and to tease apart genes involved in snout development from those that only play a  
6 role in exaggeration of the snout.

7 Conducting GO enrichment analysis on the list of upregulated DE genes also revealed  
8 the involvement of several biological processes such as proline metabolism, ‘tendon  
9 development’, as well as Notch and Wnt signalling pathways (although Wnt signalling not  
10 significantly). Interestingly, a defective proline and hydroxyproline metabolisms has been  
11 already associated with a range of skin and facial deformities including abnormal nose  
12 morphogenesis in humans (Kiratli & Satilmiş 1998; Kretz et al. 2011; Zaki et al. 2016;  
13 Baumgartner et al. 2016). A defective proline metabolism is known to severely affect collagen  
14 formation and extracellular matrix integrity, and subsequently cell adhesion (Velez et al. 2019;  
15 Karna et al. 2020; Xinjie et al. 2001; Javitt et al. 2019; Noguchi et al. 2020). We found genes  
16 involved in ‘peptidyl-proline hydroxylation’ enriched in the upregulated genes as well as  
17 ‘proline catabolic process’ in the enrichment analysis of downregulated genes. In addition, it  
18 has been recently shown that the biosynthesis of proline is tightly regulated by transforming  
19 growth factor-beta (Tgfb) (Schwörer et al. 2020), a TF that also plays role in the early  
20 development of the flapped snout in cichlids (Conith et al. 2018). We also found components  
21 of this pathway (e.g., *tgfb3*) to be differentially expressed, and both of the enriched pathways,  
22 Wnt and Notch, have conserved crosstalk with Tgfb signal in regulation of various molecular,  
23 cellular and developmental events (Attisano & Labbé 2004; Chesnutt et al. 2004; Arnold et al.  
24 2019; Klüppel & Wrana 2005; Ahi 2016). In addition, both Wnt and Notch signalling pathways  
25 are known to play a pivotal role in craniofacial development and morphogenesis including the

1 formation of middle structures including nasal structures (Penton et al. 2012; Pakvasa et al.  
2 2020; Brugmann et al. 2007; Wang et al. 2011; Singh et al. 2021).

3 The induction of Tgfb signaling is required for the establishment of cell-cell contacts  
4 in different tissues, whereas later induction of Notch signal stabilizes the Tgfb mediated effects  
5 (Klüppel & Wrana 2005). In the context of the snout, it is possible that activation of Tgfb is  
6 required for early snout induction (Conith et al. 2018) and that continued snout growth is  
7 maintained via Notch signalling. This potential time dependent crosstalk may be mediated  
8 through the downstream targets of Notch and Tgfb signals, since it is shown that both signals  
9 can regulate similar target genes (Klüppel & Wrana 2005; De Jong et al. 2004), including *foxa2*,  
10 a member of the FOX family of transcription factors (both signals suppress *foxa2* expression)  
11 (Kondratyeva et al. 2016; Liu et al. 2012). In our study, we found upregulation of *foxa2* in the  
12 flapped snout region, and interestingly, a recent study in human shows that a deletion in *Foxa2*  
13 can cause a variety of nasal deformities (Dines et al. 2019). Moreover, we found *rbpjb*, a major  
14 transcription factor mediating canonical Notch signal (Tanigaki et al. 2002), to be differentially  
15 expressed in the flapped snout of both species. In mice, *Rbpj* is shown to regulate a receptor of  
16 Tgfb signal (*Tgfbr1*), thus making a reciprocal positive regulatory loop between the two  
17 pathways (Valdez et al. 2012). We also found another receptor of Tgfb signal (*tgfbr3*) to show  
18 a similar expression pattern as *rbpjb* raising the possibility of the existence of such a reciprocal  
19 regulatory loop in flapped snout cichlids. In human, a deletion in *RBPJ* gene has been linked  
20 to abnormal thickening of the nose and lip (Nakayama et al. 2014). On the other hand, Bmp2  
21 signal which is regarded as another molecular cross point between Tgfb and Notch pathways  
22 (De Jong et al. 2004), mediates its signal through *Tgfbr3* (Hill et al. 2012). Previous studies in  
23 cichlids had proposed variations in Bmp expression as a molecular player in adaptive  
24 morphological divergence in different skeletal structures (Gunter et al. 2013; Albertson et al.  
25 2005; Ahi et al. 2017; Hulsey et al. 2016). We found downregulation of *bmp2b* expression

1 suggesting that a key regulator linking both pathways is affected in the flapped snout region.  
2 Furthermore, deletion of *Bmp2* in human has been reported to cause a range of nose and lip  
3 deformities (Tan et al. 2017). Taken together, these findings suggest complex interactions  
4 between Notch and Tgfb $\beta$  signals in the formation and possibly the maintenance of the flapped  
5 snout structure in cichlids.

6 Finally, we also conducted enrichment for TF binding sites on regulatory sequences of  
7 DEGs and found several potential binding sites for TFs that may play a role in the formation  
8 of a flapped snout. The most represented TF binding sites belonged to members of FOX  
9 transcription factor family, e.g. *foxd3*, *foxf1* and *foxp1*, as well as a consensus binding site for  
10 the FOX family. In East African cichlids, both *foxd3* and *foxp1* were predicted to regulate a  
11 gene network involved in exaggerated fin elongation (Pashay Ahi & Sefc 2018; Ahi, Richter,  
12 et al. 2019). Additionally, *foxp1* was recently suggested as an upstream regulator of genes  
13 involved in the formation of the hypertrophic lip in another East African cichlid species  
14 (Lecaudey et al. 2021). None of the predicted FOX members (*foxd3*, *foxf1* and *foxp1*) displayed  
15 consistent differential expression across both comparisons. It is, therefore, possible that the two  
16 other FOX members identified by RNA-seq and qPCR, *foxf1* and *foxa2*, are the key regulators  
17 of the entire list of DEGs, since they might bind to the predicted consensus FOX binding site.  
18 In addition, both *foxf1* and *foxa2* displayed consistently increased expression in the flapped  
19 snout in both comparisons, and are also implicated in the nose morphogenesis in mammals  
20 (Dines et al. 2019; Kucharczyk et al. 2014). We have recently found *foxf1* among the  
21 differentially expressed genes in hypertrophied lips of an East African cichlid species as well  
22 (Lecaudey et al. 2021), suggesting a potential role of *foxf1* in soft tissues exaggeration in  
23 cichlids.

24 Among the other predicted TF binding site we found overrepresentation of binding motif  
25 for *tcf12*, a transcription factor with known roles in development and morphogenesis of the



1 frontal bone and cranial vault thickening in mammals (Piard et al. 2015; Sharma et al. 2013).  
2 Moreover, we have previously identified *tcfl2* as a potential key player in the formation of a  
3 nuchal hump in an East African cichlid (Lecaudey et al. 2019). In this study, we did not detect  
4 its differential expression in the snout region. However, there might be other types of potential  
5 variations in these TFs (for example alternative splicing (Singh & Ahi 2022)), which are not  
6 necessarily reflected in their overall expression differences, but still lead to changes in their  
7 regulatory effects. Interestingly, mutations causing missense, frame shift and splicing changes  
8 are already reported for *tcfl2*, which could lead to craniofacial deformities in humans (Sharma  
9 et al. 2013).

10 The only predicted TF with consistent expression difference in both comparisons was  
11 *ap4* (or *tfap4*), i.e. showing slight but significant reduced expression in the flapped snout. *ap4*  
12 encodes a member of the basic helix-loop-helix-zipper (bHLH-ZIP) family, and can act as a  
13 transcriptional activator or repressor on a variety of downstream target genes mediating cell  
14 fate decisions (Wong et al. 2021). We also found both up- and down-regulated genes among  
15 the predicted downstream target of *ap4* (i.e. 93 genes contained *ap4* binding site), which  
16 confirms its potential transcriptional activating or repressing roles. The exact role of *ap4* in  
17 craniofacial morphogenesis of soft tissues is unclear and although deletions in a genomic region  
18 containing this gene appeared to cause facial dysmorphisms in humans such as prominent  
19 beaked nose and micrognathia (Gervasini et al. 2007), these phenotypes are mainly thought to  
20 be linked to mutations in a neighboring gene (*CBP* or *CREBBP*) in this region. Future  
21 functional studies are required to verify the potential role of *ap4* in formation and  
22 morphogenesis of craniofacial soft tissues in fish.

23

## 24 **Conclusions**

1 The snout flap in *Labeotropheus trewavasae* and *Ophthalmotilapia nasuta* is a striking and  
2 rare example of an exaggerated soft tissue trait that has evolved recurrently in the cichlid  
3 radiations of Lake Malawi and Lake Tanganyika, albeit for different functions. Comparing the  
4 transcriptional landscape of the snout flap tissue of these two species with the snout of close  
5 relatives that do not develop such a structure, we identified 201 genes that were repeatedly  
6 recruited to give rise to the snout flap phenotype even after > 9 MYA of divergence. Our study  
7 provides support for a change in proline hydroxylation, a mechanism also linked to human  
8 facial deformations, to be a mechanism for metabolic changes involved in the formation of the  
9 snout flap in fish. Additionally, we found indications of complex interactions between the  
10 transforming growth factor-beta (Tgf $\beta$ ), regulating the biosynthesis of proline, and Notch  
11 signalling, associated with morphogenesis and craniofacial development, in the formation and  
12 maintenance of the snout flap. Upstream of the differentially expressed genes we identified  
13 transcription factors belonging to the FOX family (especially *foxf1* and *foxa2*) which are both  
14 linked to the morphogenesis of the nose in mammals and *ap4* a transcription factor that showed  
15 reduced expression in the species with snout flap, but with an unknown role in craniofacial soft  
16 tissue formation. We want to emphasise that the identification of genes involved in snout  
17 morphogenesis in fish can shed light on the conserved molecular mechanisms crucial for the  
18 development and shaping of facial soft tissue. In the future it would be important to build on  
19 these findings and confirm the reuse of these genes and pathways across more distant teleost  
20 groups.

## 22 **Methods**

### 23 **Fish rearing and tissue sampling**

24 Five captive bred males of each *O. nasuta*, *O. ventralis*, and five captive bred females of *L.*  
25 *trewavasae* and *T. tropheops* were raised and kept in a large tank (approximately 450 litres)

1 containing multiple stony shelters to decrease competition stress. All specimens were at the  
2 young adult stage and have been fed with the same diet, Tropical multi-ingredient flakes  
3 suitable for omnivorous cichlids. The two species in each comparison were sampled at the same  
4 time when the protrusion of the flapped snout had already appeared (Figure 1). To perform the  
5 dissections, we used a solution with 0.3 g MS222 per 1L water to euthanize the fish, and similar  
6 snout regions, an area above the upper lip encompassing the nostrils which includes epidermis,  
7 dermis and the underlying soft connective tissues, were sampled for each fish (Figure 1). The  
8 sampled snout tissues for each individual were placed into separate tubes containing RNAlater  
9 (Qiagen) and stored at -20 C°. The sacrificing of fish followed the guidelines of the Federal  
10 Ministry of Science, Research and Economy of Austria according to the regulations of the  
11 BMWF.

### 13 **RNA extraction and cDNA synthesis**

14 Total RNA was extracted from 20 dissected snout tissue samples (5 biological replicates per  
15 species) following the TRIzol method (Thermo Fischer Scientific). Each dissected sample  
16 included epidermis, dermis, and the underlying fibrous/connective tissues of the specified nose  
17 regions (Figure 1). Tissue samples were placed into tubes containing 1 ml of TRIzol with a  
18 ceramic bead (1.4mm) and homogenized using a FastPrep-24 Instrument (MP Biomedicals,  
19 CA, USA). RNA extraction followed the protocol of TRIzol RNA extraction from Thermo  
20 Fischer Scientific. A DNA removal step with DNase followed the extraction (Invitrogen). The  
21 total RNAs were dissolved in 50 µl nuclease-free water and their concentrations were  
22 quantified through a Nanophotometer (IMPLEN GmbH, Munich, Germany). We measured the  
23 quality of RNAs with the R6K ScreenTape System using an Agilent 2200 TapeStation (Agilent  
24 Technologies) and RNA integrity numbers (RIN) above 7 were aimed at for all samples. To  
25 synthesize cDNA for qPCR analysis, we used 500 ng of the total RNA per sample and followed

1 the manufacturer's protocol of the High Capacity cDNA Reverse Transcription kit (Applied  
2 Biosystems), and the resulted cDNAs were diluted 1:4 to be used for the qPCR reaction.

3

#### 4 **RNA-seq library preparation and gene expression quantification**

5 To attain transcriptome data of the snout tissues, we conducted RNA-seq library preparation  
6 with 1000 ng of total RNA per tissue sample as input and following the protocol of the Standard  
7 TruSeq Stranded mRNA Sample Prep Kit (Illumina) with indexing adapters. The library  
8 qualities were assessed using D1000 ScreenTape and reagents (Agilent) on a TapeStation 2200  
9 machine (Agilent). In order to reach an optimal quantity recommended for sequencing, we  
10 diluted the libraries and pooled them with equal molar concentration for each library. The  
11 RNA-sequencing was conducted in the NGS Facility at Vienna Biocenter Core Facilities  
12 (VBCF, Austria) on an Illumina HiSeq2500 and generated between 6.7 and 15.8 million paired-  
13 end reads with 125bp length per sample (Supplementary Table S1). Raw reads were de-  
14 multiplexed based on unique barcodes by the same facility. The quality of the reads was  
15 assessed with FastQC (v0.11.8) (Andrews 2012), and reads were filtered for a quality > 28 and  
16 a minimum length of 70 bp with Trimmomatic (v0.3.9) (Bolger et al. 2014). Reads were aligned  
17 to the *O. niloticus* reference genome (Conte et al. 2017) of the University of Maryland using  
18 RNAstar (v2.7.3.a) (Dobin et al. 2013). To check the mapping statistics, we used samtools  
19 idxstats (v1.9) (Danecek et al. 2021) (Supplementary Table S1) and further merged the single  
20 files for species and Lake with picard (v2.21.7) (Picard Toolkit. 2019. Broad Institute, GitHub  
21 Repository. <https://broadinstitute.github.io/picard/>). We used StringTie (v.2.0.6) (Pertea et al.  
22 2015) to assemble the alignments into potential transcripts without a reference. This step was  
23 conducted separately for single files (per biological replicate) and the merged files (per species  
24 and per Lake). The single files per biological replicate were further merged into species. This  
25 process of repeated merging steps was implemented to reduce the probability of false positives.

1 To assess the accuracy of the mapping we used gffcompare (v0.11.2) (Pertea & Pertea 2020)  
2 to compare our annotations to the reference annotation. Subsequently we filtered for  
3 monoexonic transcripts that were not contained in our reference and the transcripts assigned  
4 the class code 'possible polymerase run-on' by gffcompare. As the maximum intron length of  
5 the *O. niloticus* reference is 200000 bp, we also filtered for that in the produced annotation.  
6 The expression estimates for each transcript were based on these annotations and generated  
7 with StringTie (v.2.0.6) with no multimapping allowed. The final raw count matrices were  
8 produced from the expression estimates with a Perl script from the griffith lab  
9 ([https://github.com/griffithlab/rnaseq\\_tutorial/blob/master/scripts/stringtie\\_expression\\_matrix](https://github.com/griffithlab/rnaseq_tutorial/blob/master/scripts/stringtie_expression_matrix.pl)  
10 .pl) and the code used in this analysis is available at this github repository  
11 ([https://github.com/annaduenser/snout\\_flap\\_RNAseq](https://github.com/annaduenser/snout_flap_RNAseq)).

12 Differential expression analysis was conducted using DESeq2 (Love et al. 2014) in R  
13 (R Core Team 2017) running comparisons for each Lake separately. DESeq2 estimates  
14 variance-mean dependence based on a model using negative binomial distribution using the  
15 raw counts (Love et al. 2014). A FDR of  $P < 0.05$  was chosen as the cut-off for the adjusted  $P$ -  
16 value to determine differentially expressed genes.

17  
18 For the downstream analysis, an enrichment step for gene ontology (GO) terms of biological  
19 processes was conducted in R using topGO (v2.48.0) (Alexa & Rahnenfuhrer 2019) with the  
20 method *weight* and using Fisher's exact tests for the enrichment analysis, while GO terms for  
21 *Oreochromis niloticus* were acquired via the biomaRt package (v2.46.1) (Durinck et al. 2005,  
22 2009) from the Ensemble database. To further group functionally similar GO terms, we also  
23 used REVIGO (Supek et al. 2011) using simRel scores (Schlicker et al. 2006). To predict the  
24 potential upstream regulators of DE genes, we conducted motif overrepresentation analysis on  
25 4 kb upstream sequences (promoter and 5'-UTR) of these genes using the annotated genome of

1 the Nile tilapia (Zerbino et al. 2018) and MEME tool (Bailey et al. 2009). The motifs that were  
2 present in the promoters of at least one fifth of the total 201 DEGs were compared to position  
3 weight matrices (PWMs) in the TRANSFAC database (Matys et al. 2003) via STAMP  
4 (Mahony & Benos 2007) in order to identify matching TF binding sites. In addition, we  
5 investigated the functional interactions between the products of DE genes through STRING  
6 v10 (<http://string-db.org/>), a knowledge based interaction prediction tool, and zebrafish  
7 databases for protein interactomes (Szkarczyk et al. 2017).

8

### 9 **Primer design and qPCR**

10 We designed the qPCR primers on conserved regions of the selected candidate genes by  
11 aligning their assembled sequences to their already available homologous mRNA sequences  
12 from *Ophthalmotilapia ventralis* (Böhne et al. 2014), *Metriaclima zebra*, *Pundamilia nyererei*,  
13 *Neolamprologus brichardi* and *Astatotilapia burtoni* (Brawand et al. 2014), as well as  
14 *Oreochromis niloticus*. After aligning the conserved sequence regions across the  
15 abovementioned East African cichlids, we identified the exon junctions (using CLC Genomic  
16 Workbench, CLC Bio, Denmark, and annotated genome of *Astatotilapia burtoni* in the  
17 Ensembl database, <http://www.ensembl.org>). The primer designing steps were conducted as  
18 described previously (Pashay Ahi & Sefc 2018; Ahi, Richter, et al. 2019) using Primer Express  
19 3.0 (Applied Biosystems, CA, USA) (Supplementary Table S5). The qPCR was performed  
20 based on the protocol provided by Maxima SYBR Green/ROX qPCR Master Mix (2X)  
21 (Thermo Fisher Scientific, Germany) following the guidelines for optimal experimental set-up  
22 for each qPCR run (Hellemans et al. 2007). The qPCR program was set for 2 min at 50°C, 10  
23 min at 95°C, 40 cycles of 15 sec at 95°C and 1 min at 60°C, followed by an additional step of  
24 dissociation at 60°C – 95°C. The primer efficiency (E values) for each gene was calculated  
25 through standard curves generated by serial dilutions of pooled cDNA samples. The standard



1 curves were run in triplicates and calculated using the following formula:  $E = 10[-1/\text{slope}]$   
2 (Supplementary Table S5).

3 In order to select stably expressed candidate reference genes, we filtered for genes with  
4 a low log<sub>2</sub> fold change and subsequently ranked the remaining genes according to low  
5 coefficient of variation. The top six most stable genes shared across the transcriptome  
6 comparisons were selected as candidate reference genes (Supplementary Table S4). After  
7 qPCR expression analysis of the six genes across all samples, we ranked them based on their  
8 expression stability by three different algorithms: BestKeeper (Pfaffl et al. 2004), NormFinder  
9 (Andersen et al. 2004) and geNorm (Vandesompele et al. 2002). We used the C<sub>q</sub> values of the  
10 top most stable reference genes to normalize C<sub>q</sub> values of target genes in each sample ( $\Delta C_q$   
11 target = C<sub>q</sub> target – C<sub>q</sub> reference). The relative expression levels (RQ) were calculated by  
12  $2^{-\Delta\Delta C_q}$  method (Pfaffl 2001) and the log-transformed RQ values were used for independent-  
13 samples t-tests to calculate the statistical differences.

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## 21 **Author Contributions**

22 EPA, CA and CS conceived the project. WG contributed to fish husbandry and photography,  
23 and EPA and AD conducted the sampling and tissue dissection. AD, EPA and LL conducted  
24 the RNA lab work. AD, PS, LL, EPA contributed to the analyses and all authors to manuscript  
25 writing. CS and EPA contributed to funding. This work was supported by the Austrian Science

1 Fund [project number P29838] awarded to CS. All authors approved the final version of the  
2 manuscript.

3

#### 4 **Competing financial interests**

5 The authors declare no competing interests.

6

#### 7 **Ethical approval**

8 No experiments were conducted on the fish prior to sampling, so an ethics approval is not  
9 required according to the Austrian animal welfare law. Fish keeping and sacrifice was carried  
10 out in our certified aquarium facility in accordance with the Austrian animal welfare law.

11

#### 12 **Data availability**

13 The data underlying this article are available in the Sequencing Read Archive (SRA) of NCBI  
14 at <https://www.ncbi.nlm.nih.gov/> and can be accessed with PRJNA770252.

15

#### 16 **References**

- 17 Ahi E. 2016. Signalling pathways in trophic skeletal development and morphogenesis:  
18 Insights from studies on teleost fish. *Dev. Biol.* 420:11–31. doi:  
19 <http://dx.doi.org/10.1016/j.ydbio.2016.10.003>.
- 20 Ahi EP, Duenser A, Singh P, Gessl W, Sturmbauer C. 2020. Appetite regulating genes may  
21 contribute to herbivory versus carnivory trophic divergence in haplochromine cichlids. *PeerJ.*  
22 8:e8375. doi: 10.7717/peerj.8375.
- 23 Ahi EP et al. 2020. Expression levels of the tetratricopeptide repeat protein gene *ttc39b*  
24 covary with carotenoid-based skin colour in cichlid fish. *Biol. Lett.* 16:20200629. doi:  
25 [10.1098/rsbl.2020.0629](https://doi.org/10.1098/rsbl.2020.0629).
- 26 Ahi EP, Richter F, Lecaudey LA, Sefc KM. 2019. Gene expression profiling suggests  
27 differences in molecular mechanisms of fin elongation between cichlid species. *Sci. Rep.*  
28 9:9052. doi: 10.1038/s41598-019-45599-w.
- 29 Ahi EP, Richter F, Sefc KM. 2017. A gene expression study of ornamental fin shape in  
30 *Neolamprologus brichardi*, an African cichlid species. *Sci. Rep.* 7:17398. doi:  
31 [10.1038/s41598-017-17778-0](https://doi.org/10.1038/s41598-017-17778-0).
- 32 Ahi EP, Singh P, Duenser A, Gessl W, Sturmbauer C. 2019. Divergence in larval jaw gene

- 1 expression reflects differential trophic adaptation in haplochromine cichlids prior to foraging.  
2 BMC Evol. Biol. 19:150. doi: 10.1186/s12862-019-1483-3.
- 3 Alazami AM et al. 2016. Expanding the clinical and genetic heterogeneity of hereditary  
4 disorders of connective tissue. Hum. Genet. 135:525–540. doi: 10.1007/s00439-016-1660-z.
- 5 Albertson RC, Streelman JT, Kocher TD, Yelick PC. 2005. Integration and evolution of the  
6 cichlid mandible: the molecular basis of alternate feeding strategies. Proc. Natl. Acad. Sci. U.  
7 S. A. 102:16287–92. doi: 10.1073/pnas.0506649102.
- 8 Alexa A, Rahnenfuhrer J. 2019. topGO: Enrichment Analysis for Gene Ontology version  
9 2.42.0 from Bioconductor. R Packag. <https://rdrr.io/bioc/topGO/> (Accessed August 31, 2022).
- 10 Alhamoudi KM et al. 2020. A Missense Mutation in the UGDH Gene Is Associated With  
11 Developmental Delay and Axial Hypotonia. Front. Pediatr. 8. doi: 10.3389/fped.2020.00071.
- 12 Alwadei AH et al. 2016. Loss-of-function mutation in *RUSC2* causes intellectual disability  
13 and secondary microcephaly. Dev. Med. Child Neurol. 58:1317–1322. doi:  
14 10.1111/dmcn.13250.
- 15 Andersen CL, Jensen JL, Ørntoft TF. 2004. Normalization of real-time quantitative reverse  
16 transcription-PCR data: a model-based variance estimation approach to identify genes suited  
17 for normalization, applied to bladder and colon cancer data sets. Cancer Res. 64:5245–50.  
18 doi: 10.1158/0008-5472.CAN-04-0496.
- 19 Andrews S. 2012. FastQC: a quality control tool for high throughput sequence data.  
20 <http://www.bioinformatics.babraham.ac.uk/projects/fastqc>.
- 21 Arnold CP, Benham-Pyle BW, Lange JJ, Wood CJ, Sánchez Alvarado A. 2019. Wnt and  
22 TGFβ coordinate growth and patterning to regulate size-dependent behaviour. Nature.  
23 572:655–659. doi: 10.1038/s41586-019-1478-7.
- 24 Attisano L, Labbé E. 2004. TGFβ and Wnt pathway cross-talk. Cancer Metastasis Rev.  
25 23:53–61. doi: 10.1023/A:1025811012690.
- 26 Bailey TL et al. 2009. MEME SUITE: tools for motif discovery and searching. Nucleic Acids  
27 Res. 37:W202–8. doi: 10.1093/nar/gkp335.
- 28 Barrionuevo MG, Aybar MJ, Tríbulo C. 2014. Two different vestigial like 4 genes are  
29 differentially expressed during *Xenopus laevis* development. Int. J. Dev. Biol. 58:369–377.  
30 doi: 10.1387/ijdb.130353ct.
- 31 Baumgarten L, Machado-Schiaffino G, Henning F, Meyer A. 2015. What big lips are good  
32 for: On the adaptive function of repeatedly evolved hypertrophied lips of cichlid fishes. Biol.  
33 J. Linn. Soc. 115:448–455. doi: 10.1111/bij.12502.
- 34 Baumgartner MR, Valle D, Dionisi-Vici C. 2016. Disorders of Ornithine and Proline  
35 Metabolism. In: Inborn Metabolic Diseases. Springer Berlin Heidelberg pp. 321–331. doi:  
36 10.1007/978-3-662-49771-5\_21.
- 37 Böhne A, Sengstag T, Salzburger W. 2014. Comparative transcriptomics in East African  
38 cichlids reveals sex- and species-specific expression and new candidates for sex  
39 differentiation in fishes. Genome Biol. Evol. 6:2567–2585. doi: 10.1093/gbe/evu200.
- 40 Bolger AM, Lohse M, Usadel B. 2014. Trimmomatic: a flexible trimmer for Illumina  
41 sequence data. Bioinformatics. 30:2114–2120. doi: 10.1093/bioinformatics/btu170.
- 42 Bonaglia MC et al. 2005. A 2.3 Mb duplication of chromosome 8q24.3 associated with

- 1 severe mental retardation and epilepsy detected by standard karyotype. *Eur. J. Hum. Genet.*  
2 13:586–591. doi: 10.1038/sj.ejhg.5201369.
- 3 Brawand D et al. 2014. The genomic substrate for adaptive radiation in African cichlid fish.  
4 *Nature.* 513:375–381. doi: 10.1038/nature13726.
- 5 Brugmann SA et al. 2007. Wnt signaling mediates regional specification in the vertebrate  
6 face. *Development.* 134:3283–95. doi: 10.1242/dev.005132.
- 7 Brugmann SA, Cordero DR, Helms JA. 2010. Craniofacial ciliopathies: A new classification  
8 for craniofacial disorders. *Am. J. Med. Genet. Part A.* 152A:2995–3006. doi:  
9 10.1002/ajmg.a.33727.
- 10 Chen CP, Ko TM, Chen YY, Su JW, Wang W. 2013. Prenatal diagnosis and molecular  
11 cytogenetic characterization of mosaicism for a small supernumerary marker chromosome  
12 derived from chromosome 22 associated with cat eye syndrome. *Gene.* 527:384–388. doi:  
13 10.1016/j.gene.2013.05.061.
- 14 Chesnutt C, Burrus LW, Brown AMC, Niswander L. 2004. Coordinate regulation of neural  
15 tube patterning and proliferation by TGF $\beta$  and WNT activity. *Dev. Biol.* 274:334–347. doi:  
16 10.1016/j.ydbio.2004.07.019.
- 17 Colombo M et al. 2013. The ecological and genetic basis of convergent thick-lipped  
18 phenotypes in cichlid fishes. *Mol. Ecol.* 22:670–684. doi: 10.1111/mec.12029.
- 19 Concannon MR, Albertson RC. 2015. The genetic and developmental basis of an exaggerated  
20 craniofacial trait in East African cichlids. *J. Exp. Zool. B. Mol. Dev. Evol.* 324:662–70. doi:  
21 10.1002/jez.b.22641.
- 22 Conith MR et al. 2018. Genetic and developmental origins of a unique foraging adaptation in  
23 a Lake Malawi cichlid genus. *Proc. Natl. Acad. Sci.* 115:7063–7068. doi:  
24 10.1073/PNAS.1719798115.
- 25 Conith MR, Conith AJ, Albertson RC. 2019. Evolution of a soft-tissue foraging adaptation in  
26 African cichlids: Roles for novelty, convergence, and constraint. *Evolution (N. Y.)*. 73:2072–  
27 2084. doi: 10.1111/evo.13824.
- 28 Conte MA, Gammerdinger WJ, Bartie KL, Penman DJ, Kocher TD. 2017. A high quality  
29 assembly of the Nile Tilapia (*Oreochromis niloticus*) genome reveals the structure of two sex  
30 determination regions. *BMC Genomics.* 18:341. doi: 10.1186/s12864-017-3723-5.
- 31 Czeschik JC et al. 2014. A patient with a de-novo deletion 3p25.3 and features overlapping  
32 with Rubinstein–Taybi syndrome. *Clin. Dysmorphol.* 23:67–70. doi:  
33 10.1097/MCD.0000000000000035.
- 34 Danecek P et al. 2021. Twelve years of SAMtools and BCFtools. *Gigascience.* 10. doi:  
35 10.1093/gigascience/giab008.
- 36 Dell’Edera D et al. 2018. 16p11.2 microdeletion syndrome: A case report. *J. Med. Case Rep.*  
37 12:1–6. doi: 10.1186/s13256-018-1587-1.
- 38 Dines JN et al. 2019. Expanding phenotype with severe midline brain anomalies and  
39 missense variant supports a causal role for *FOXA2* in 20p11.2 deletion syndrome. *Am. J.*  
40 *Med. Genet. Part A.* 179:ajmg.a.61281. doi: 10.1002/ajmg.a.61281.
- 41 Dobin A et al. 2013. STAR: ultrafast universal RNA-seq aligner. *Bioinformatics.* 29:15–21.  
42 doi: 10.1093/bioinformatics/bts635.

- 1 Durinck S et al. 2005. BioMart and Bioconductor: a powerful link between biological  
2 databases and microarray data analysis. *Bioinformatics*. 21:3439–3440. doi:  
3 10.1093/bioinformatics/bti525.
- 4 Durinck S, Spellman PT, Birney E, Huber W. 2009. Mapping identifiers for the integration of  
5 genomic datasets with the R/ Bioconductor package biomaRt. *Nat. Protoc.* 4:1184–1191. doi:  
6 10.1038/nprot.2009.97.
- 7 Ehret JK et al. 2015. Microdeletions in 9q33.3-q34.11 in five patients with intellectual  
8 disability, microcephaly, and seizures of incomplete penetrance: Is STXBP1 not the only  
9 causative gene? *Mol. Cytogenet.* 8:1–14. doi: 10.1186/s13039-015-0178-8.
- 10 Fryssira H et al. 2011. Severe Developmental Delay in a Patient with 7p21.1–p14.3  
11 Microdeletion Spanning the *HOXA* Gene and the  
12 *HOXA* Gene Cluster. *Mol. Syndromol.* 2:45–49. doi: 10.1159/000334313.
- 13 Gervasini C et al. 2007. High frequency of mosaic CREBBP deletions in Rubinstein-Taybi  
14 syndrome patients and mapping of somatic and germ-line breakpoints. *Genomics*. 90:567–  
15 573. doi: 10.1016/j.ygeno.2007.07.012.
- 16 Giannikou K et al. 2012. Further delineation of novel 1p36 rearrangements by array-CGH  
17 analysis: Narrowing the breakpoints and clarifying the ‘extended’ phenotype. *Gene*. 506:360–  
18 368. doi: 10.1016/j.gene.2012.06.060.
- 19 Guilmatre A et al. 2010. Type I hyperprolinemia: genotype/phenotype correlations. *Hum.*  
20 *Mutat.* 31:961–965. doi: 10.1002/humu.21296.
- 21 Gunter HM et al. 2013. Shaping development through mechanical strain: the transcriptional  
22 basis of diet-induced phenotypic plasticity in a cichlid fish. *Mol. Ecol.* 22:4516–4531. doi:  
23 10.1111/mec.12417.
- 24 Haldeman-Englert CR et al. 2009. A 3.1-Mb microdeletion of 3p21.31 associated with  
25 cortical blindness, cleft lip, CNS abnormalities, and developmental delay. *Eur. J. Med. Genet.*  
26 52:265–268. doi: 10.1016/j.ejmg.2008.11.005.
- 27 Hanssens M, Snoeks J, Verheyen E. 1999. A morphometric revision of the genus  
28 *Ophthalmotilapia* (Teleostei, Cichlidae) from Lake Tanganyika (East Africa). *Zool. J. Linn.*  
29 *Soc.* 125:487–512. doi: 10.1111/j.1096-3642.1999.tb00602.x.
- 30 Hellemans J, Mortier G, De Paepe A, Speleman F, Vandesompele J. 2007. qBase relative  
31 quantification framework and software for management and automated analysis of real-time  
32 quantitative PCR data. *Genome Biol.* 8:R19. doi: 10.1186/gb-2007-8-2-r19.
- 33 Henning F, Machado-Schiaffino G, Baumgarten L, Meyer A. 2017. Genetic dissection of  
34 adaptive form and function in rapidly speciating cichlid fishes. *Evolution (N. Y.)*. 71:1297–  
35 1312. doi: 10.1111/evo.13206.
- 36 Hill CR et al. 2012. BMP2 signals loss of epithelial character in epicardial cells but requires  
37 the Type III TGF $\beta$  receptor to promote invasion. *Cell. Signal.* 24:1012–1022. doi:  
38 10.1016/j.cellsig.2011.12.022.
- 39 Holder JL, Lotze TE, Bacino C, Cheung S-W. 2012. A child with an inherited 0.31 Mb  
40 microdeletion of chromosome 14q32.33: Further delineation of a critical region for the 14q32  
41 deletion syndrome. *Am. J. Med. Genet. Part A.* 158A:1962–1966. doi:  
42 10.1002/ajmg.a.35289.
- 43 Hosono K et al. 2020. A case of childhood glaucoma with a combined partial monosomy



- 1 6p25 and partial trisomy 18p11 due to an unbalanced translocation. *Ophthalmic Genet.*  
2 41:175–182. doi: 10.1080/13816810.2020.1744019.
- 3 Hufnagel RB et al. 2016. A new frontonasal dysplasia syndrome associated with deletion of  
4 the *SIX2* gene. *Am. J. Med. Genet. Part A.* 170:487–491. doi: 10.1002/ajmg.a.37441.
- 5 Hulsey CD, Fraser GJ, Meyer A. 2016. Biting into the genome to phenome map:  
6 Developmental genetic modularity of cichlid fish dentitions. In: *Integrative and Comparative*  
7 *Biology*. Vol. 56 Oxford University Press pp. 373–388. doi: 10.1093/icb/icw059.
- 8 Hüning I et al. 2013. Exon 2 duplication of the *MID1* gene in a patient with a mild phenotype  
9 of Opitz G/BBB syndrome. *Eur. J. Med. Genet.* 56:188–191. doi:  
10 10.1016/j.ejmg.2013.01.004.
- 11 Irisarri I et al. 2018. Phylogenomics uncovers early hybridization and adaptive loci shaping  
12 the radiation of Lake Tanganyika cichlid fishes. *Nat. Commun.* 9:1–12. doi: 10.1038/s41467-  
13 018-05479-9.
- 14 Jain S, Yang P, Farrell SA. 2010. A case of 8q22.1 microdeletion without the Nablus mask-  
15 like facial syndrome phenotype. *Eur. J. Med. Genet.* 53:108–110. doi:  
16 10.1016/j.ejmg.2009.12.006.
- 17 Javitt G et al. 2019. *cis*-Proline mutants of quiescin sulphydryl oxidase 1 with altered redox  
18 properties undermine extracellular matrix integrity and cell adhesion in fibroblast cultures.  
19 *Protein Sci.* 28:228–238. doi: 10.1002/pro.3537.
- 20 De Jong DS et al. 2004. Regulation of Notch signaling genes during BMP2-induced  
21 differentiation of osteoblast precursor cells. *Biochem. Biophys. Res. Commun.* 320:100–107.  
22 doi: 10.1016/j.bbrc.2004.05.150.
- 23 Kariminejad A et al. 2015. Intellectual disability, muscle weakness and characteristic face in  
24 three siblings: A newly described recessive syndrome mapping to 3p24.3-p25.3. *Am. J. Med.*  
25 *Genet. Part A.* 167:2508–2515. doi: 10.1002/ajmg.a.37248.
- 26 Karna E, Szoka L, Huynh TYL, Palka JA. 2020. Proline-dependent regulation of collagen  
27 metabolism. *Cell. Mol. Life Sci.* 77:1911–1918. doi: 10.1007/s00018-019-03363-3.
- 28 Kiratli H, Satılmış M. 1998. Prolidase deficiency associated with pathologic myopia.  
29 *Ophthalmic Genet.* 19:49–53. doi: 10.1076/opge.19.1.49.2180.
- 30 Klüppel M, Wrana JL. 2005. Turning it up a Notch: cross-talk between TGF $\beta$  and Notch  
31 signaling. *BioEssays.* 27:115–118. doi: 10.1002/bies.20187.
- 32 Kocher TD, Conroy JA, McKaye KR, Stauffer JR. 1993. Similar morphologies of cichlid fish  
33 in lakes tanganyika and malawi are due to convergence. *Mol. Phylogenet. Evol.* 2:158–165.  
34 doi: 10.1006/mpev.1993.1016.
- 35 Kondratyeva LG et al. 2016. Downregulation of expression of mater genes *SOX9*, *FOXA2*,  
36 and *GATA4* in pancreatic cancer cells stimulated with TGF $\beta$ 1 epithelial–mesenchymal  
37 transition. *Dokl. Biochem. Biophys.* 469:257–259. doi: 10.1134/S1607672916040062.
- 38 Konings A. 2007. *Malawi cichlids in their natural habitat 4th Edition*. 4th ed. Cichlid Press.  
39 El Paso, Texas, USA.: El Paso, Texas, USA.
- 40 Kosho T et al. 2008. De-novo balanced translocation between 7q31 and 10p14 in a girl with  
41 central precocious puberty, moderate mental retardation, and severe speech impairment. *Clin.*  
42 *Dysmorphol.* 17:31–34. doi: 10.1097/MCD.0b013e3282f17688.



- 1 Kretz R et al. 2011. Defect in proline synthesis: Pyrroline-5-carboxylate reductase 1  
2 deficiency leads to a complex clinical phenotype with collagen and elastin abnormalities. *J.*  
3 *Inherit. Metab. Dis.* 34:731–739. doi: 10.1007/s10545-011-9319-3.
- 4 Kubista M et al. 2006. The real-time polymerase chain reaction. *Mol. Aspects Med.* 27:95–  
5 125. doi: 10.1016/j.mam.2005.12.007.
- 6 Kucharczyk M et al. 2014. The first case of a patient with de novo partial distal 16q  
7 tetrasomy and a data's review. *Am. J. Med. Genet. Part A.* 164:2541–2550. doi:  
8 10.1002/ajmg.a.36686.
- 9 Lecaudey LA et al. 2021. Transcriptomics unravels molecular players shaping dorsal lip  
10 hypertrophy in the vacuum cleaner cichlid, *Gnathochromis permaxillaris*. *BMC Genomics.*  
11 22:506. doi: 10.1186/s12864-021-07775-z.
- 12 Lecaudey LA, Sturbauer C, Singh P, Ahi EP. 2019. Molecular mechanisms underlying  
13 nuchal hump formation in dolphin cichlid, *Cyrtocara moorii*. *Sci. Rep.* doi: 10.1038/s41598-  
14 019-56771-7.
- 15 Liu M et al. 2012. IKK $\alpha$  Activation of NOTCH Links Tumorigenesis via FOXA2  
16 Suppression. *Mol. Cell.* 45:171–184. doi: 10.1016/j.molcel.2011.11.018.
- 17 Lopes F et al. 2019. Genomic imbalances defining novel intellectual disability associated  
18 loci. *Orphanet J. Rare Dis.* 14:1–13. doi: 10.1186/s13023-019-1135-0.
- 19 Losos JB, Jackman TR, Larson A, De Queiroz K, Rodríguez-Schettino L. 1998. Contingency  
20 and determinism in replicated adaptive radiations of island lizards. *Science (80- )*. 279:2115–  
21 2118. doi: 10.1126/science.279.5359.2115.
- 22 Love MI, Huber W, Anders S. 2014. Moderated estimation of fold change and dispersion for  
23 RNA-seq data with DESeq2. *Genome Biol.* 15:550. doi: 10.1186/s13059-014-0550-8.
- 24 Machado-Schiaffino G, Henning F, Meyer A. 2014. Species-specific differences in adaptive  
25 phenotypic plasticity in an ecologically relevant trophic trait: hypertrophic lips in Midas  
26 cichlid fishes. *Evolution (N. Y.)*. 68:2086–2091. doi: 10.1111/evo.12367.
- 27 Mahony S, Benos P V. 2007. STAMP: a web tool for exploring DNA-binding motif  
28 similarities. *Nucleic Acids Res.* 35:W253-8. doi: 10.1093/nar/gkm272.
- 29 Malinsky M et al. 2018. Whole-genome sequences of Malawi cichlids reveal multiple  
30 radiations interconnected by gene flow. *Nat. Ecol. Evol.* 2:1940–1955. doi: 10.1038/s41559-  
31 018-0717-x.
- 32 Manousaki T et al. 2013. Parsing parallel evolution: ecological divergence and differential  
33 gene expression in the adaptive radiations of thick-lipped Midas cichlid fishes from  
34 Nicaragua. *Mol. Ecol.* 22:650–69. doi: 10.1111/mec.12034.
- 35 Martinez-Glez V et al. 2007. Clinical presentation of a variant of Axenfeld-Rieger syndrome  
36 associated with subtelomeric 6p deletion. *Eur. J. Med. Genet.* 50:120–127. doi:  
37 10.1016/j.ejmg.2006.10.005.
- 38 Matys V et al. 2003. TRANSFAC®: Transcriptional regulation, from patterns to profiles.  
39 *Nucleic Acids Res.* 31:374–378. doi: 10.1093/nar/gkg108.
- 40 McLachlan E et al. 2005. Functional Characterization of Oculodentodigital Dysplasia-  
41 Associated Cx43 Mutants. *Cell Commun. Adhes.* 12:279–292. doi:  
42 10.1080/15419060500514143.

- 1 Miller ND et al. 2009. Molecular (SNP) analyses of overlapping hemizygous deletions of  
2 10q25.3 to 10qter in four patients: Evidence for HMX2 and HMX3 as candidate genes in  
3 hearing and vestibular function. *Am. J. Med. Genet. Part A.* 149A:669–680. doi:  
4 10.1002/ajmg.a.32705.
- 5 Mitteldorf C et al. 2018. Deceptively bland cutaneous angiosarcoma on the nose mimicking  
6 hemangioma-A clinicopathologic and immunohistochemical analysis. *J. Cutan. Pathol.*  
7 45:652–658. doi: 10.1111/cup.13275.
- 8 Mordaunt D et al. 2015. 8q13.1-q13.2 deletion associated with inferior cerebellar vermian  
9 hypoplasia and digital anomalies: A new syndrome? *Pediatr. Neurol.* 52:230-234.e1. doi:  
10 10.1016/j.pediatrneurol.2014.09.002.
- 11 Mucha BE et al. 2019. A new microdeletion syndrome involving TBC1D24, ATP6V0C, and  
12 PDPK1 causes epilepsy, microcephaly, and developmental delay. *Genet. Med.* 21:1058–  
13 1064. doi: 10.1038/s41436-018-0290-3.
- 14 Nakayama T et al. 2014. RBPJ is disrupted in a case of proximal 4p deletion syndrome with  
15 epilepsy. *Brain Dev.* 36:532–536. doi: 10.1016/j.braindev.2013.07.009.
- 16 Nimmagadda S et al. 2015. Identification and functional analysis of novel facial patterning  
17 genes in the duplicated beak chicken embryo. *Dev. Biol.* 407:275–288. doi:  
18 10.1016/j.ydbio.2015.09.007.
- 19 Noguchi Y, Iwasaki Y, Ueda M, Kakinoki S. 2020. Surfaces immobilized with oligo-prolines  
20 prevent protein adsorption and cell adhesion. *J. Mater. Chem. B.* 8:2233–2237. doi:  
21 10.1039/d0tb00051e.
- 22 Ochab-Marcinek A, Tabaka M. 2010. Bimodal gene expression in noncooperative regulatory  
23 systems. *Proc. Natl. Acad. Sci. U. S. A.* 107:22096–22101. doi: 10.1073/pnas.1008965107.
- 24 Okello DO et al. 2017. Six2 Plays an Intrinsic Role in Regulating Proliferation of  
25 Mesenchymal Cells in the Developing Palate. *Front. Physiol.* 8:955. doi:  
26 10.3389/fphys.2017.00955.
- 27 Pakvasa M et al. 2020. Notch signaling: Its essential roles in bone and craniofacial  
28 development. *Genes Dis.* doi: 10.1016/j.gendis.2020.04.006.
- 29 Pashay Ahi E, Sefc KM. 2018. Towards a gene regulatory network shaping the fins of the  
30 Princess cichlid. *Sci. Rep.* 8:9602. doi: 10.1038/s41598-018-27977-y.
- 31 De Pater JM et al. 2005. Striking Facial Dysmorphisms and Restricted Thymic Development  
32 in a Fetus with a 6-Megabase Deletion of Chromosome 14q. *Pediatr. Dev. Pathol.* 8:497–503.  
33 doi: 10.1007/s10024-005-0041-8.
- 34 Penton AL, Leonard LD, Spinner NB. 2012. Notch signaling in human development and  
35 disease. *Semin. Cell Dev. Biol.* 23:450–457. doi: 10.1016/j.semcd.2012.01.010.
- 36 Pertea G, Pertea M. 2020. GFF Utilities: GffRead and GffCompare. *F1000Research.* 9. doi:  
37 10.12688/f1000research.23297.2.
- 38 Pertea M et al. 2015. StringTie enables improved reconstruction of a transcriptome from  
39 RNA-seq reads. *Nat. Biotechnol.* 33:290–295. doi: 10.1038/nbt.3122.
- 40 Pfaffl MW. 2001. A new mathematical model for relative quantification in real-time RT-  
41 PCR. *Nucleic Acids Res.* 29:e45.
- 42 Pfaffl MW, Tichopad A, Prgomet C, Neuvians TP. 2004. Determination of stable

- 1 housekeeping genes, differentially regulated target genes and sample integrity: BestKeeper--  
2 Excel-based tool using pair-wise correlations. *Biotechnol. Lett.* 26:509–15.
- 3 Piard J et al. 2015. *TCF12* microdeletion in a 72-year-old woman with intellectual disability.  
4 *Am. J. Med. Genet. Part A.* 167:1897–1901. doi: 10.1002/ajmg.a.37083.
- 5 Pillai NR et al. 2019. Novel deletion of 6p21.31p21.1 associated with laryngeal cleft,  
6 developmental delay, dysmorphic features and vascular anomaly. *Eur. J. Med. Genet.*  
7 62:103531. doi: 10.1016/j.ejmg.2018.08.012.
- 8 Powder KE, Albertson RC. 2016. Cichlid fishes as a model to understand normal and clinical  
9 craniofacial variation. *Dev. Biol.* 415:338–346. doi: 10.1016/J.YDBIO.2015.12.018.
- 10 Preiksaitiene E et al. 2015. R368X mutation in *MID1* among recurrent mutations in patients  
11 with X-linked Opitz G/BBB syndrome. *Clin. Dysmorphol.* 24:7–12. doi:  
12 10.1097/MCD.000000000000059.
- 13 Quigley DI, Kaiser-Rogers K, Aylsworth AS, Rao KW. 2004. Submicroscopic deletion  
14 9(q34.4) and duplication 19(p13.3): Identified by subtelomere specific FISH probes. *Am. J.*  
15 *Med. Genet.* 125A:67–72. doi: 10.1002/ajmg.a.20457.
- 16 R Core Team. 2017. *R: A Language and Environment for Statistical Computing.* doi:  
17 10.1007/978-3-540-74686-7.
- 18 Reijnders MRF et al. 2017. *RAC1* Missense Mutations in Developmental Disorders with  
19 Diverse Phenotypes. *Am. J. Hum. Genet.* 101:466–477. doi: 10.1016/j.ajhg.2017.08.007.
- 20 Rivera-Pedroza CI et al. 2017. Chromosome 1p31.1p31.3 Deletion in a Patient with  
21 Craniosynostosis, Central Nervous System and Renal Malformation: Case Report and  
22 Review of the Literature. *Mol. Syndromol.* 8:30–35. doi: 10.1159/000452609.
- 23 Rüber L, Verheyen E, Meyer A. 1999. Replicated evolution of trophic specializations in an  
24 endemic cichlid fish lineage from Lake Tanganyika. *Proc. Natl. Acad. Sci. U. S. A.*  
25 96:10230–10235. doi: 10.1073/pnas.96.18.10230.
- 26 Rundle HD, Nagel L, Boughman JW, Schluter D. 2000. Natural selection and parallel  
27 speciation in sympatric sticklebacks. *Science (80- )*. 287:306–308. doi:  
28 10.1126/science.287.5451.306.
- 29 Sansone A, Syed AS, Tantalaki E, Korsching SI, Manzini I. 2014. *Trpc2* is expressed in two  
30 olfactory subsystems, the main and the vomeronasal system of larval *Xenopus laevis*. *J. Exp.*  
31 *Biol.* 217:2235–2238. doi: 10.1242/jeb.103465.
- 32 Sawyer JR, Binz RL, Swanson CM, Lim C. 2007. De novo proximal duplication of  
33 1(q12q22) in a female infant with multiple congenital anomalies. *Am. J. Med. Genet. Part A.*  
34 143A:338–342. doi: 10.1002/ajmg.a.31604.
- 35 Schlicker A, Domingues FS, Rahnenführer J, Lengauer T. 2006. A new measure for  
36 functional similarity of gene products based on gene ontology. *BMC Bioinformatics.* 7:302.  
37 doi: 10.1186/1471-2105-7-302.
- 38 Schluter D, Nagel LM. 1995. Parallel speciation by natural selection. *Am. Nat.* 146:292–301.  
39 doi: 10.1086/285799.
- 40 Schulz S, Volleth M, Muschke P, Wieland I, Wieacker P. 2008. Greig cephalopolysyndactyly  
41 (GCPS) contiguous gene syndrome in a boy with a 14 Mb deletion in region 7p13-14 caused  
42 by a paternal balanced insertion (5; 7). *Appl. Clin. Genet.* 1:19–22. doi: 10.2147/tacg.s4401.

- 1 Schwörer S et al. 2020. Proline biosynthesis is a vent for TGF $\beta$ -induced mitochondrial redox  
2 stress. *EMBO J.* 39. doi: 10.15252/embj.2019103334.
- 3 Selenti N et al. 2015. An interstitial deletion at 8q23.1-q24.12 associated with Langer-  
4 Giedion syndrome/Trichorhinophalangeal syndrome (TRPS) type II and Cornelia de Lange  
5 syndrome 4. *Mol. Cytogenet.* 8:64. doi: 10.1186/s13039-015-0169-9.
- 6 Sharma VP et al. 2013. Mutations in TCF12, encoding a basic helix-loop-helix partner of  
7 TWIST1, are a frequent cause of coronal craniosynostosis. *Nat. Genet.* 45:304–307. doi:  
8 10.1038/ng.2531.
- 9 Singh P, Ahi EP. 2022. The importance of alternative splicing in adaptive evolution. *Mol.*  
10 *Ecol.* 31:1928–1938. doi: 10.1111/mec.16377.
- 11 Singh P, Ahi EP, Sturmbauer C. 2021. Gene coexpression networks reveal molecular  
12 interactions underlying cichlid jaw modularity. *BMC Ecol. Evol.* 21:1–17. doi:  
13 10.1186/s12862-021-01787-9.
- 14 Sofos E et al. 2012. A novel familial 11p15.4 microduplication associated with intellectual  
15 disability, dysmorphic features, and obesity with involvement of the ZNF214 gene. *Am. J.*  
16 *Med. Genet. Part A.* 158A:50–58. doi: 10.1002/ajmg.a.34290.
- 17 Starkovich M, Lalani SR, Mercer CL, Scott DA. 2016. Chromosome 5q33 deletions  
18 associated with congenital heart defects. *Am. J. Med. Genet. Part A.* 170:3338–3342. doi:  
19 10.1002/ajmg.a.37957.
- 20 Supek F, Bošnjak M, Škunca N, Šmuc T. 2011. REVIGO Summarizes and Visualizes Long  
21 Lists of Gene Ontology Terms Gibas, C, editor. *PLoS One.* 6:e21800. doi:  
22 10.1371/journal.pone.0021800.
- 23 Szklarczyk D et al. 2017. The STRING database in 2017: quality-controlled protein–protein  
24 association networks, made broadly accessible. *Nucleic Acids Res.* 45:D362–D368. doi:  
25 10.1093/nar/gkw937.
- 26 Tan TY et al. 2017. Monoallelic BMP2 Variants Predicted to Result in Haploinsufficiency  
27 Cause Craniofacial, Skeletal, and Cardiac Features Overlapping Those of 20p12 Deletions.  
28 *Am. J. Hum. Genet.* 101:985–994. doi: 10.1016/j.ajhg.2017.10.006.
- 29 Tanigaki K et al. 2002. Notch-RBP-J signaling is involved in cell fate determination of  
30 marginal zone B cells. *Nat. Immunol.* 3:443–450. doi: 10.1038/ni793.
- 31 Tanigaki K, Honjo T. 2010. *Two Opposing roles of RBP-J in Notch signaling.* Academic  
32 Press doi: 10.1016/S0070-2153(10)92007-3.
- 33 Thomas PS, Kim J, Nunez S, Glogauer M, Kaartinen V. 2010. Neural crest cell-specific  
34 deletion of Rac1 results in defective cell-matrix interactions and severe craniofacial and  
35 cardiovascular malformations. *Dev. Biol.* 340:613–625. doi: 10.1016/j.ydbio.2010.02.021.
- 36 Tinker RJ et al. 2021. Haploinsufficiency of <scp>ATP6V0C </scp> possibly underlies  
37 16p13.3 deletions that cause microcephaly, seizures, and neurodevelopmental disorder. *Am.*  
38 *J. Med. Genet. Part A.* 185:196–202. doi: 10.1002/ajmg.a.61905.
- 39 Tomkins DJ, Gitelman BJ, Roberts MH. 1983. Confirmation of a de novo duplication,  
40 dup(10) (q24→q26), by GOT1 gene dosage studies. *Hum. Genet.* 63:369–373. doi:  
41 10.1007/BF00274764.
- 42 Țuțulan-Cuniță AC et al. 2012. 3p interstitial deletion: Novel case report and review. *J. Child*

- 1 Neurol. 27:1062–1066. doi: 10.1177/0883073811431016.
- 2 Valdez JM et al. 2012. Notch and TGF $\beta$  form a reciprocal positive regulatory loop that  
3 suppresses murine prostate basal stem/progenitor cell activity. *Cell Stem Cell*. 11:676–688.  
4 doi: 10.1016/j.stem.2012.07.003.
- 5 Vandesompele J et al. 2002. Accurate normalization of real-time quantitative RT-PCR data  
6 by geometric averaging of multiple internal control genes. *Genome Biol*.  
7 3:RESEARCH0034.
- 8 Velez DO et al. 2019. 3D collagen architecture regulates cell adhesion through degradability,  
9 thereby controlling metabolic and oxidative stress. *Integr. Biol*. 11:221–234. doi:  
10 10.1093/intbio/zyz019.
- 11 Wang Y, Song L, Zhou CJ. 2011. The canonical Wnt/ $\beta$ -catenin signaling pathway regulates  
12 Fgf signaling for early facial development. *Dev. Biol*. 349:250–260. doi:  
13 10.1016/j.ydbio.2010.11.004.
- 14 Wieacker P, Volleth M. 2007. *WNT4* and *RSPO1*: Are  
15 Not Involved in a Case of Male-to-Female Sex Reversal with Partial Duplication of 1p. *Sex*.  
16 *Dev*. 1:111–113. doi: 10.1159/000100032.
- 17 Wong MM-K, Joyson SM, Hermeking H, Chiu SK. 2021. Transcription Factor AP4 Mediates  
18 Cell Fate Decisions: To Divide, Age, or Die. *Cancers (Basel)*. 13:676. doi:  
19 10.3390/cancers13040676.
- 20 Xinjie L et al. 2001. Evaluation of the role of proline residues flanking the RGD motif of  
21 dendrospain, an inhibitor of platelet aggregation and cell adhesion. *Biochem. J*. 355:633–  
22 638. doi: 10.1042/bj3550633.
- 23 Yakut S et al. 2015. A familial interstitial 4q35 deletion with no discernible clinical effects.  
24 *Am. J. Med. Genet. Part A*. 167:1836–1841. doi: 10.1002/ajmg.a.37097.
- 25 Zaki MS et al. 2016. *PYCR2* Mutations cause a lethal syndrome of microcephaly and failure  
26 to thrive. *Ann. Neurol*. 80:59–70. doi: 10.1002/ana.24678.
- 27 Zerbino DR et al. 2018. Ensembl 2018. *Nucleic Acids Res*. 46:D754–D761. doi:  
28 10.1093/nar/gkx1098.
- 29 Zhang P, Yang C, Delay RJ. 2010. Odors activate dual pathways, a TRPC2 and a AA-  
30 dependent pathway, in mouse vomeronasal neurons. *Am. J. Physiol. Physiol*. 298:C1253–  
31 C1264. doi: 10.1152/ajpcell.00271.2009.
- 32 Zlotina A et al. 2016. Ring chromosome 18 in combination with 18q12.1 (DTNA) interstitial  
33 microdeletion in a patient with multiple congenital defects. *Mol. Cytogenet*. 9:1–7. doi:  
34 10.1186/s13039-016-0229-9.