Retinoic acid catabolizing enzyme CYP26C1 is a genetic modifier in SHOX deficiency

Antonino Montalbano1, Lonny Juergensen2, Ralph Roeth1, Birgit Weiss3, Maki Fukami3, Susanne Fricke-Otto4, Gerhard Binder5, Tsutomu Ogata6, Eva Decker7, Gudrun Nuernberg8,9, David Hassel2 & Gudrun A Rappold1,10,*

Abstract

Mutations in the homeobox gene SHOX cause SHOX deficiency, a condition with clinical manifestations ranging from short stature without dysmorphic signs to severe mesomelic skeletal dysplasia. In rare cases, individuals with SHOX deficiency are asymptomatic. To elucidate the factors that modify disease severity/penetration, we studied a three-generation family with SHOX deficiency. The variant p.Phe508Cys of the retinoic acid catabolizing enzyme CYP26C1 co-segregated with the SHOX variant p.Val161Ala in the affected individuals, while the SHOX mutant alone was present in asymptomatic individuals. Two further cases with SHOX deficiency and damaging CYP26C1 variants were identified in a cohort of 68 individuals with LWD. The identified CYP26C1 variants affected its catabolic activity, leading to an increased level of retinoic acid. High levels of retinoic acid significantly decrease SHOX expression in human primary chondrocytes and zebrafish embryos. Individual morpholino knockdown of either gene shortens the pectoral fins, whereas deletion of both genes leads to a more severe phenotype. Together, our findings describe CYP26C1 as the first genetic modifier for SHOX deficiency.

Keywords clinical variability; genetic modifiers; limb development; retinoic acid; skeletal dysplasia

Subject Categories Development & Differentiation; Genetics, Gene Therapy & Genetic Disease; Musculoskeletal System

DOI 10.15252/emmm.201606623 | Received 22 May 2016 | Revised 28 September 2016 | Accepted 10 October 2016 | Published online 14 November 2016

EMBO Mol Med (2016) 8: 1455–1469

Introduction

In the last decade, many genetic variations responsible for the pathogenesis of disease have been identified. Next-generation sequencing and high-throughput genotyping platforms have revolutionized the pursuit of disease-causing genetic variants. However, disease phenotypes vary—even among individuals carrying the same causative genetic variants within one family—and our understanding of the factors that modify disease severity/penetration remains incomplete.

Human height is a complex trait with a high heritability. Short stature affects approximately 3% of children worldwide and is diagnosed when height is significantly below the average of the general population for that person’s age and sex. More precisely, short stature is statistically defined as two standard deviations (SD) below the mean population height for age, sex, and ethnic group (less than the third percentile) or, when evaluating shortness in relation to family background, more than two SD below the mid-parental height (Ranke, 1994). To date, many different etiologies of short stature are known and more than two hundred genes underlying growth control have been identified (Marchini et al, 2007; Durand & Rappold, 2013; Baron et al, 2015).

Mutations in the short stature homeobox-containing (SHOX) gene cause SHOX deficiency, the most frequent monogenic cause of short stature (Marchini et al, 2016). The gene is localized in the pseudoautosomal region 1 (PAR1) shared between the X and Y chromosomes. Its varied clinical manifestations include idiopathic/isolated short stature (ISS), Léri-Weill dyschondrosteosis (LWD), and Langer mesomelic dysplasia (LD). Heterozygous mutations in its coding or regulatory regions have been identified in up to 10% of patients diagnosed with ISS and 70% of patients with LWD, while homozygous mutations cause LD (Zinn et al, 2002; Rosilio et al, 2012; Marchini et al, 2016). SHOX deficiency also contributes to the
short stature and skeletal features in Turner syndrome (Rao et al., 1997; Clement-Jones et al., 2000). Thus, SHOX deficiency is associated with a broad phenotypic spectrum ranging from short stature without dysmorphic signs to profound dysplasia (Rao et al., 1997; Belin et al., 1998; Shears et al., 1998). Patients with SHOX deficiency often present mesomorphic (disproportionate) short stature, a selective shortening of the lower arms/legs. The phenotype may also include Madelung deformity (MD) of the wrist, considered to be the archetypal sign of LWD, which is characterized by shortening and bowing of the radius together with distal hypoplasia of the ulna. Skeletal manifestations are usually more severe in females than in males (Binder, 2011).

The clinical severity of SHOX deficiency varies even in family members carrying the same SHOX mutation (Schiller et al., 2000; Binder, 2011). In rare cases, family members with identical mutation present with stature within the normal range (Huber et al., 2006; Benito-Sanz et al., 2012; Buyan et al., 2013). We postulated that SHOX deficiency provides a genetically sensitized background on which genetic modifiers may promote disease progression. To shed light on this phenomenon, we have analyzed a family with five affected individuals displaying LWD. All affected individuals carried a damaging missense mutation in the SHOX gene. Three family members with the same mutation were phenotypically unaffected. To explain this clinical variability, we hypothesized the presence of modifier gene(s). Using whole-genome linkage analysis and whole-exome sequencing, we identified a CYP26C1 variant to segregate solely in the affected individuals and carried out subsequent genetic and functional assays to provide evidence for a modifying role of this gene.

Results

SHOX deficiency and clinical variability

We have analyzed a three-generation German family with five affected individuals displaying LWD (family 1). According to the pattern of transmission of the trait, a dominant inheritance was hypothesized (Fig 1A). All female patients (I:2, I:3, II:7, and III:2, Fig 1A) presented with mesomelic short stature with a SD between −3.14 and −4.51 as well as MD, the archetypal sign of LWD. The father (II:3) of the index patient (II:2) presented with mesomelic short stature with an SD of −2.63 and a borderline MD, consistent with the fact that males are less severely affected than females (Table EV1). The mother (II:4), an aunt (II:8), and the stepsister (III:1) of the index patient (III:2) also presented with short stature, with the mother (II:4) and the stepsister (III:1) being diagnosed with the same mutation as the index patient (II:2). Variants in 57 genes were obtained (Dataset EV1). All variants were filtered with rare filtering as genetic modifier

Identification of CYP26C1 as genetic modifier

To address putative genetic causes of this intra-familiar clinical variability, whole-genome linkage analysis was performed. This analysis defined a 2.02 Mb interval in the PAR1 of chromosome X (chrX:706800-2735491; hg19) with a total LOD score of 1.7 (Figs EV1 and EV2). Moreover, a LOD score of 2.4 was obtained in a 19.2 Mb region of chromosome 10 (chr10:85477515-104681710; hg19) (Fig EV3). This region encompasses 263 genes, but could not be further refined. Subsequently, whole-exome sequencing analysis was performed on the index patient (III:2) and her father (II:3). Variant filtering was performed according to the hypothesized dominant transmission of the phenotype (see Materials and Methods); 98 variants in 97 genes were obtained (Dataset EV1). All variants were tested using the in silico mutation prediction tools PolyPhen-2, Mutation Taster, SIFT, and PROVEAN, and 36 variants were predicted as disease causing/damaging by at least one of these programs.

Sanger sequencing of the 36 identified variants was performed in the five affected and in the eight non-affected family members where DNA was available. Only variants in two genes, OPN4 (chr10:88419701-88419701; hg19) and CYP26C1 (chr10:94828408-94828408; hg19), segregated with the phenotype. Both genes reside within the chromosome 10 interval, which was previously defined by linkage analysis. OPN4 (melanopsin) is a known photopigment of photosensitive retinal ganglion cells and was not considered further (Panda et al., 2002). CYP26C1 (NG_007958.1) encodes for a member of the cytochrome P450 superfamily of enzymes involved in the catabolism of retinoic acid (RA) (Taimi et al., 2004; Uehara et al., 2006; Pennimpede et al., 2010). Loss-of-function analyses in mouse and zebrafish have shown that CYP26C1 RA catabolic activity is required for proper hindbrain development (Sirbu et al., 2005; Uehara et al., 2006; Hernandez et al., 2007). RA has been previously shown to play a key role in limb development (Cunningham & Duester, 2015) and CYP26B1, another member of the CYP26 family, is known to be involved in skeleton development (Laue et al., 2011). Moreover, RA has been previously shown to inhibit Shox expression in chicken limbs (Tiecke et al., 2006). Therefore, we decided to further investigate CYP26C1 as a candidate modifier in SHOX deficient LWD patients.

SHOX and CYP26C1 are members of the RA pathway

The heterozygous missense variant c.1523T>G (p.Phe508Cys) in CYP26C1 affected a highly conserved amino acid among vertebrates. p.Phe508Cys was predicted as damaging by all prediction tools that were applied (Dataset EV2). The variant has not yet been described in the three major public variants databases, Exome Aggregation Consortium (ExAC), Exome Variant Server (EVS), and 1000 Genomes Project (TGP). To assess whether this mutation affects the enzymatic activity of CYP26C1, we used the Cignal RARE system, a RA-responsive luciferase reporter assay, in U2OS cells treated with all-trans retinoic acid (ATRA) (Laue et al., 2011). Overexpression of wild-type CYP26C1 reduced the luciferase activity, confirming that it degrades ATRA. In contrast, the CYP26C1 p.Phe508Cys mutant was
not able to reduce luciferase activity, suggesting that this mutation impairs CYP26C1 enzymatic activity and consequently RA degradation (Fig 2B).

Next, we tested CYP26C1 expression in human primary chondrocytes, where SHOX is expressed. By performing RT–PCR and Western blot analysis, we could show that CYP26C1 is expressed in these cells (Fig 3A). We then asked whether RA affected SHOX expression in human primary chondrocytes (Appendix Fig S1). Treatment of primary chondrocytes with 100 nM ATRA led to a significant reduction of SHOX levels (Fig 3B). RA exerts its function by regulating the transcriptional activity of nuclear retinoic acid receptors (RARs) that bind as heterodimers with retinoic X receptors (RXRs) to RA response elements. Binding of RA to these receptors triggers activation or repression of their target genes (Weston et al., 2003). To verify whether RA directly or indirectly regulates SHOX expression, we tested a previously described SHOX promoter luciferase reporter assay in U2OS cells (Verdin et al., 2015). Treatment with ATRA led to a significant reduction of luciferase activity compared to mock control (Fig 3C). In silico analysis of the SHOX promoter predicted three putative RXRa binding sites (Fig 3C). However, disruption of these binding sites did not significantly influence the ATRA effect on the SHOX promoter, indicating an indirect effect of RA on SHOX expression (Fig 3C). Altogether, these results provide evidence that CYP26C1 and SHOX are members of the same pathway: CYP26C1 regulates SHOX expression by regulating intracellular levels of RA (Fig 3D).

Additional genetic evidence in SHOX deficiency patients presenting with LWD

We then investigated whether the co-occurrence of damaging SHOX and CYP26C1 variants in the severe phenotypes represents a unique finding specific to family 1. We screened CYP26C1 in a cohort of 68 LWD individuals with proven SHOX deficiency and in a cohort of...
350 control individuals with normal height. This analysis identified two further cases with co-occurrence of damaging variants in SHOX and CYP26C1 in the patient cohort (Fig 1B and Dataset EV2). No functional CYP26C1 damaging variants were found in the control individuals with normal height (Dataset EV3; Appendix Fig S2).

In family 2, the affected daughter (II:2) carried a heterozygous missense variant in SHOX, c.349C>G (p.Leu132Val). This variant has been shown to affect SHOX homodimerization and DNA binding (Schneider et al, 2005). The father (I:1) and the sister (II:1) also carried this SHOX variant, but they were reported to be unaffected without dysmorphic signs. Screening of CYP26C1 identified a heterozygous missense variant c.1133G>A (p.Arg378His) only in the daughter with LWD (II:2) (Fig 1B, Table EV1). DNA from the mother (I:2) was not available for the CYP26C1 gene analysis. The third case was an affected girl who carried a de novo heterozygous deletion of SHOX and a missense variant in CYP26C1, c.356A>C (p.Gln119Pro) (Table EV1). Both parents and the brother presented with normal stature and had no dysmorphic signs. The CYP26C1 variant was inherited from the father. Since the SHOX deletion is de novo, this family cannot demonstrate co-segregation, although it adds to the overall evidence that damaging variants in SHOX and CYP26C1 co-occur in individuals with severe LWD phenotypes.

We then performed luciferase analysis of all the SHOX and CYP26C1 mutations and could demonstrate their negative impact on protein activity (Fig 2B). We conclude that in addition to family 1, two out of 68 LWD patients with SHOX deficiency presented with functional damaging mutations in CYP26C1, while no damaging mutations with impact on protein activity were identified in 350 control individuals with normal height. In all families where clinical information was available, all individuals with damaging mutations in both SHOX and CYP26C1 presented with short stature and severe skeletal phenotypes.
Modeling SHOX deficiency in zebrafish embryos

Finally, to gain insight into the role of SHOX and CYP26C1 interaction on limb development, we performed antisense morpholino (MO) knockdown experiments in zebrafish embryos. Testing of each MO proofed efficacy (Appendix Figs S4–S6). We first assessed the effect of shox reduction on limb/fin development. Upon shox knockdown, the zebrafish embryos showed an overall delayed growth and a strong impairment of the developing pectoral fins, in concordance with a previous report (Sawada et al, 2015; Fig 4).
involved in limb development and acts by controlling RA levels. An in situ hybridization experiment showed that expression of *col2a1* was dramatically reduced in the pectoral fin buds upon shox reduction (Fig 4C). Overexpression experiments in zebrafish embryos of human SHOX wild type and the variants identified in families 1 and 2 corroborated the functional significance of SHOX p.Val161Ala (family 1) and p.Leu132Val (family 2) (Appendix Fig S7).

We then analyzed the effect of *cyp26c1* knockdown in zebrafish embryos. Expression of *cyp26c1* in the pectoral fins has been previously demonstrated by whole-mount *in situ* hybridization (Gu et al., 2005). Knockdown of *cyp26c1* resulted in a significant reduction of pectoral fin size, although less strikingly compared to the shox knockdown (Fig 5B–E). The embryos also showed an abnormal development of the otic vesicles and pharyngeal arches (Fig 5B). As reported previously, we also found signs of cardiac dysfunction as evident by the pericardial edema (Rydeen & Waxman, 2014) (Fig 5B). In the pectoral fins, we demonstrated that the expression of *col2a1* was strongly reduced or absent (Fig 5C). In addition, the expression of *shox* was shown to be decreased upon *cyp26c1* knockdown (Fig 5D and G). MO knockdown of *cyp26c1* led to a significant increase in RA levels compared to control MO, which is in line with its role in RA degradation (Fig 5F). We treated zebrafish embryos with RA and obtained a significant downregulation of *shox* expression, further corroborating the hypothesis of SHOX as a component of the RA pathway (Fig 5H). Finally, we tested the functional significance of the CYP26C1 variants identified in families 1–3 in zebrafish embryos and could show a striking effect on CYP26C1 activity (Appendix Fig S8). Together, these data indicate that CYP26C1 is involved in limb development and acts by controlling RA levels.

To test the hypothesis that CYP26C1 is a modifier of SHOX deficiency, we tested different concentration of shox and cyp26c1 MOs to determine subphenotypic dosages. While single knockdown of either shox or cyp26c1 using subphenotypic MO doses did not result in any obvious phenotype, double knockdown (simultaneous injection) of shox and cyp26c1 produced significantly smaller pectoral fins (Fig 6A–D). The relative expression of shox mRNA expression was also significantly reduced (Fig 6E). Staining for *col2a1* revealed impaired pectoral fin development (Fig 6B). These data further corroborate the hypothesis that CYP26C1 damaging mutations contribute to severe phenotypes in SHOX deficient individuals.

**Discussion**

Variable phenotypes can be attributed to genetic and environmental factors. Our study describes a Mendelian disorder with wide phenotypic variability, which provides a unique opportunity to identify the genetic causes of variability. Individuals with SHOX deficiency but normal stature in a three-generation family (Family 1) prompted us to postulate genetic modifiers as the possible reasons of this variability. Three individuals with height within the normal range and neither mesomelia nor MD had inherited the same SHOX variant p.Val161Ala as their five affected relatives with LWD. By combining whole-genome linkage and whole-exome sequencing analysis in family 1, we identified CYP26C1 as the gene co-segregating with the clinical phenotype and hypothesized a putative modifier function on SHOX expression. The mother (II:4), an aunt (II:8), and the stepsister (III:1) of the index patient (III:2) presented with short stature,
Figure 5. Pattern of defects in zebrafish embryos injected with anti-cyp26c1 morpholino.

Wild-type embryos injected with control MO or with cyp26c1 MO.

A, B (A) Lateral views of the embryos at 55 hours post-fertilization (hpf). (B) Dorsal view and magnification on the lateral view of the embryos. Dotted line, pectoral fins; *, otic vesicle. cyp26c1 morphants show smaller fins compared to controls (n = 30 embryos).

C, D Expression of col2a1 (C) and shox (D) at 55 hpf was examined by in situ hybridization in embryos injected with control MO or with cyp26c1 MO. (C) Dorsal view and magnification on the pectoral fins of col2a1 expression. Arrow and dotted line indicate the pectoral fin. (D) Dorsal view and magnification on the pectoral fins of shox expression. Arrow and dotted line indicate the pectoral fin.

E Pectoral fin area was measured by ImageJ (n = 30 embryos).

F Cignal-RARE system luciferase assay to test cyp26c1 MO knockdown effect on RA acid levels in zebrafish embryos (n = 5 replicates). In each replicate, 20–30 embryos per condition were assayed. RLU, relative light units.

G Relative shox mRNA expression normalized to reference genes eef1a and b-actin in zebrafish embryos injected with control MO or cyp26c1 MO (n = 4). RNA was extracted from 10–15 injected embryos at 55 hpf.

H Relative shox mRNA expression normalized to reference genes eef1a and b-actin in zebrafish embryos treated with 100 nM ATRA (n = 4). Embryos were collected at 24 hpf and treated with mock control or ATRA. RNA was extracted from 10–15 embryos after 6-h treatment.

Data information: The box represents the interquartile range. The whiskers represent Min to Max. (F) *P-value = 0.0006, (G) *P-value = 0.0119, (H) *P-value = 0.0286, (E) Two-tailed unpaired Student's t-test. (F–H) Two-tailed Mann–Whitney non-parametric t-test.
Figure 6. Co-injection of titrated subphenotypic doses of anti-shox and anti-cyp26c1 morpholinos impairs limb development.

Wild-type embryos injected with control MO, subphenotypic doses of shox MO, cyp26c1 MO, or a combination of shox + cyp26c1 MOs.

A Dorsal views of the embryos at 55 hours post-fertilization (hpf). Dotted line, pectoral fins. shox + cyp26c1 double morphants show smaller fins compared to control and single MOs (n = 30 embryos).

B, C Dorsal view and magnification on the pectoral fins of col2a1 expression at 55 hpf. Arrow and dotted line indicate the pectoral fin.

D Pectoral fin area was measured by ImageJ (n = 30 embryos). Data are shown as means ± SD. ***P = 0.0001 (one-way ANOVA with Bonferroni’s multiple comparison test).

E Relative expression of shox mRNA normalized to reference genes eef1a and b-actin in zebrafish embryos injected with control MO, shox MO, cyp26c1 MO, or shox + cyp26c1 MOs (n = 4). RNA was extracted from 10–15 injected embryos at 55 hpf. Data are shown as means ± SD. **P = 0.0048 (Kruskal–Wallis with Dunn’s multiple comparison test).

Data information: The box represents the interquartile range. The whiskers represent Min to Max. (D) ***P-value = 0.0001, (E) **P-value = 0.0048. (D) One-way ANOVA Bonferroni’s multiple comparison test. (E) Kruskal–Wallis Dunn’s multiple comparison test.
but dysmorphic skeletal signs were not diagnosed. These individuals did not carry damaging variants neither in SHOX nor in CYP26C1, suggesting that different factors contribute to their short stature phenotype. Mesomelia and MD were present only in those individuals carrying damaging variants in both SHOX and CYP26C1.

Defective CYP26C1 and SHOX were found to lead to a more severe phenotype in two further unrelated patients with LWD (Fig 1B). No functionally damaging variants in CYP26C1 were found in 350 control individuals with normal height, suggesting that this co-occurrence is not coincidental (P-value = 0.0261, two-tailed Fisher's exact test). In addition, we browsed TGP for genotypes bearing both SHOX and CYP26C1 damaging variants (predicted as damaging by at least one of the prediction tools used). A limitation of this approach is that not for all variants individual genotypes are available. We could not find any individual from the TGP database carrying damaging variants in both genes (1000 Genomes Project Consortium, 2015). Finally, we browsed the ExAC database and estimated the frequency of SHOX and CYP26C1 damaging variants (Lek et al., 2016). To estimate the frequencies, we summed the allele frequencies of each damaging variant (predicted as damaging by at least one of the prediction tools used) reported in the ExAC database. The estimated allele frequencies were 0.3 and 0.6% for SHOX and CYP26C1, respectively. With these estimated frequencies, the probability for an individual to bear a damaging variant in both genes is $1.8 \times 10^{-5}$.

CYP26C1 is an enzyme belonging to the cytochrome P450 superfamily and is involved in the oxidation of RA to generate polar retinoid species, which are readily excreted. Thus, CYP26C1 is involved in the catabolism of RA (Taimi et al., 2004; Uehara et al., 2006; Pennimpede et al., 2010). Accordingly, we found that damaging mutations in CYP26C1 reduce its RA catabolizing activity leading to a higher concentration of this retinoid in U2OS cells (Fig 2B).

Retinoic acid plays a key role in development, including formation of the body axis and skeleton (Pennimpede et al., 2010; Cunningham & Dueter, 2015). During skeletal development, RA coordinates the development of central body axis, limb axis, and cranium. Moreover, it controls chondroblast differentiation and coordinates maturation and replacement of bone tissue during endochondral ossification (Weston et al., 2003). RA also plays a role in the postnatal maintenance of bone (Green et al., 2015). An excess or deficiency of RA dysregulates the expression of the respective target genes, which has a dramatic effect on development (Weston et al., 2003). Excess or loss of RA has, for example, been shown to impair pectoral fin development in zebrafish and results in reduced forelimb size or complete loss of forelimb in mouse embryos (Uehara et al., 2006; Akimenko & Ekker, 1995; Begemann et al., 2001; Emoto et al., 2005; Cunningham et al., 2013). Hence, a tight regulation of RA metabolism is essential.

We demonstrated for the first time that CYP26C1 is expressed on RNA and protein level in human primary chondrocytes (Fig 3), suggesting a role for CYP26C1 in the fine regulation of RA in these cells. Damaging mutations in CYP26C1 that affect its RA oxidation activity may therefore lead to high levels of this retinoid. In support of this hypothesis, we found that knockdown of cyp26c1 in zebrafish embryos increased RA levels and reduced col2a1 expression (Fig 5F). Previous experiments on chicken limbs have shown that treatment with excess RA strongly reduces Shox expression (Tiecke et al., 2006). Consistent with a role of RA in SHOX regulation, we have shown that treatment of human primary chondrocytes and zebrafish embryos with 100 nM RA significantly reduced SHOX expression, while this was not the case at 10–50 nM RA (Fig 2B and 5H; Appendix Fig S1). Luciferase reporter analyses of the human SHOX promoter suggest an indirect effect of RA on SHOX expression (Fig 3C). RA could lead to SHOX downregulation independently of RAREs, for example, by controlling the activation of other transcription factors, by non-classical associations of receptors with other proteins, or other mechanisms (Balmer & Blomhoff, 2002). Finally, we showed that reduction of cyp26c1 in zebrafish embryos leads to the downregulation of shox expression (Fig 5D and G). Taken together, our results suggest that SHOX expression is not normally affected by endogenous RA in the nanomolar range (De Leeheer et al., 1995; Cunningham et al., 2013); however, loss of CYP26C1 results in excess RA that downregulates SHOX expression.

Height depends mostly on the longitudinal growth of the limbs. Limbs contain long bones that are formed through endochondral ossification. This process involves the aggregation of mesenchymal progenitors that differentiate into chondrocytes, which form a cartilage template that is then replaced by bone tissue. Endochondral ossification is orchestrated by a complex network of signaling pathways and transcription factors and is highly conserved in vertebrates. SHOX is one of the genes involved in this network (Blaschke & Rappold, 2001). In accordance with a role for SHOX in limb development, we could demonstrate that shox reduction impairs pectoral fin development in zebrafish embryos (Fig 4).

Gene dosage of the SHOX locus has previously been shown to determine height: SHOX deficiency causes short stature, whereas SHOX overdosage has been linked to tall stature in Klinefelter syndrome (Ogata et al., 2001). SHOX has been suggested to repress growth plate fusion and skeletal maturation in the distal limbs. Unlike SHOX deficiency, which is characterized by premature epiphyseal plate fusion and relatively advanced skeletal maturation, SHOX overdosage leads to a delayed growth plate fusion and consequently to longer limbs (Ogata et al., 2001). Thus, different dosages of the SHOX protein play a role in determining the adult height of an individual.

We have found that damaging mutations in CYP26C1 increase RA levels, which affect SHOX dosage and exacerbate SHOX deficiency phenotypes. To corroborate this hypothesis, we have demonstrated that the co-injection of subphenotypic dosages of shox and cyp26c1 MOs strongly impaired pectoral fin development (Fig 6). However, it is possible that damaging mutations in CYP26C1 modify SHOX deficiency not only by directly affecting its expression, but also by misregulating other genes involved in the formation of long bones.

Homozygous/compound heterozygous mutations in CYP26C1 have previously been associated with focal facial dermal dysplasia type IV (Slavotinek et al., 2013), a mild disorder of the skin. However, different CYP26C1 variants were reported and information on height in these individuals was not available. MO knockdown experiments of cyp26c1 in zebrafish embryos revealed abnormal pectoral fin development, but also an impairment of the otic vesicle and pharyngeal arches, structures that encompass the sites of the skin lesions observed in focal facial dermal dysplasia type IV (Slavotinek et al., 2013) (Fig 5B).
In 1963, Gruneberg (1963) defined modifiers as “genes capable of modifying the manifestation of a mutant gene without having an obvious effect on the normal condition”. It would therefore be interesting to find out whether damaging CYP26C1 variants have detectable phenotypes in the absence of SHOX deficiency. In family 3, the father (II:1), for example, carries a CYP26C1 damaging variant but presented height within the normal range and no obvious dysmorphic signs. A systematic analysis on a large cohort of patients with or without dysmorphic signs and short stature is therefore needed to resolve whether CYP26C1 variants alone can contribute to short stature phenotypes.

Taken together, this study represents an effort to elucidate the genetic causes of variability on the clinical manifestation in patients with SHOX deficiency. The RA pathway and one of its components, CYP26C1, were uncovered as biological triggers that we suggest to modify the severity of SHOX deficiency and alter the course of this disease. Only a few genetic modifiers have been reported in the literature so far (Oprea et al., 2008; Ebermann et al., 2010; Bećanović et al., 2015; Corvol et al., 2015; Guo et al., 2015; Lee et al., 2015), but recent technological advances are helping to broaden the understanding of the molecular basis of quantitative or discrete qualitative differences in phenotype. A primary reason to identify disease modifiers is to enable the accurate prediction of disease progression and improve therapeutic development. In the case of SHOX deficiency, manipulating the RA signaling pathway may be therapeutically beneficial.

Materials and Methods

Patients and controls

Family 1
Family 1 comprised 17 German individuals with five affected members diagnosed with LWD. DNA was available from 14 individuals (I:1, I:2, I:3, II:3, II:4, II:6, II:7, II:8, III:1, III:2, III:3, III:4, III:5, III:6). Four females (I:2, I:3, II:7, III:2) presented with mesomelia and MD. In the affected male (II:3), mesomelia and MD were borderline. Individuals (II:4, II:8, III:1) presented with short stature but no dysmorphic signs. The index patient (III:2) was diagnosed with LWD at the age of 8 years and treated with growth hormone from the age of 8.9 years onwards. Age of menarche was 12 years. Further details on the clinical data are given in Table EV1.

Cohort of patients with SHOX deficiency
The cohort comprised 68 unrelated cases with LWD and proven SHOX deficiency; it comprised 60 Europeans (28 Germans) and 9 Japanese. Two out of the 68 cases carried damaging mutations in CYP26C1 and all the available family information was retrieved (see below).

Family 2 comprised four French people with one daughter affected with LWD; parents and sister were unaffected without dysmorphic signs. The affected daughter (II:2) presented with short stature, mesomelia, and MD (available clinical data are given in Table EV1).

Family 3 comprised four Japanese individuals with one daughter presenting with LWD. Parents and brother were reported with normal stature and no dysmorphic signs (available clinical data are given in Table EV1).

Controls
As controls, we screened 350 (240 Germans, 110 Japanese) individuals with normal stature and no dysmorphic signs. We also compared frequencies to publicly available databases (ExAC, TGP, and EVS).

Linkage analysis
We genotyped DNA samples from 12 family members of family 1 using the Affymetrix GeneChip Human Mapping 50K Xba 240 Array (Affymetrix). Genotypes were called by the Affymetrix DNA Analysis Software (GDAS v3.0, Affymetrix). We verified sample genders by counting heterozygous SNPs on the X chromosome. Relationship errors were evaluated with the help of the program Graphical Relationship Representation (Abecasis et al., 2001). The program PedCheck was applied to detect Mendelian errors (O’Connell & Weeks, 1998) and data for SNPs with such errors were removed from the dataset. Non-Mendelian errors were identified by using the program MERLIN (Abecasis et al., 2002) and unlikely genotypes for related samples were deleted. Non-parametric linkage analysis using all genotypes of a chromosome simultaneously was carried out with MERLIN.

Whole-exome sequencing
Whole-exome sequencing was performed as previously described (Haack et al., 2012). Exomes were enriched in solution provided by the manufacturer and indexed with SureSelect, XT Human All Exon 50 Mb kits (Agilent). Samples were sequenced as 100-bp paired-end runs on a HiSeq2000 system (Illumina). Read alignment to the human genome assembly was performed with the Burrows-Wheeler Aligner (v.0.5.8.). SAMtools (v.0.1.7.) was used for detecting single nucleotide variants and small insertions and deletions. We generated 8.8 and 11.2 gigabases of sequence resulting in 89 and 93% of the target regions being covered at least 20 times for index patient (III:2) and her father (II:3), respectively. To identify putative candidate genes based on dominant inheritance, we filtered all variants that were present in both individuals, but absent in all except two of 1,297 in-house control exomes.
Sanger sequencing

Sanger sequencing was performed on MegaBACE sequencer using the DYEnamicTM ET Terminator Cycle Sequencing Kit (GE Healthcare Life Sciences) following the manufacturer’s protocol. Sequences were analyzed using the MegaBACE Sequence Analyzer (v3.0).

Analysis in human primary chondrocytes

Human primary chondrocytes (mycoplasma-free) were obtained as previously described (Marchini et al, 2004). Cells were cultured in DMEM (Gibco) containing 10% FBS (Gibco) and penicillin/streptomycin (Gibco) at 37°C, 5% CO₂, 95% humidity. To test CYP26C1 mRNA expression, cells were resuspended in Trizol® (Invitrogen) for RNA preparation according to standard protocols. cDNA synthesis was performed with SuperScript™ II (Invitrogen) according to manufacturer’s protocol. Specific primers for CYP26C1, SOX9, and SHOX transcripts were designed (Dataset EV4) and PCR performed. PCR products were confirmed by Sanger sequencing.

To test CYP26C1 protein expression, microsomal preparations were obtained as follows: Cells were washed three times with 2 ml cold solution containing 8 mM Na₂HPO₄, 1.5 mM KH₂PO₄, and 2.7 mM KCl and subsequently suspended in 1.5 ml of this buffer. Resuspension was centrifuged at 360 g for 2 min at 4°C. The pellet was resuspended in 1 ml of 100 mM potassium phosphate buffer pH 7.4 containing 10 mM EDTA. Resuspension was sonicated for 20 × 1 s. Lysed cells were then centrifuged at 10,700 g for 1 h at 4°C. The pellet was homogenized in 100 µl of 50 mM potassium phosphate buffer pH 7.4, containing 0.1 mM EDTA and 10% glyceral (Biomol). Protein concentration was determined using the BCA method. Western blotting experiments were performed following standard protocol with the LI-COR Odyssey™ system (LI-COR). hCyp26c1 antibody (PA5-15059, Thermo Fischer Scientific) was diluted as recommended (dilution 1:100) in LI-COR Blocking Buffer (LI-COR), 1 × TBS, Tween 0.1%.

SHOX expression analysis was performed as follows: Cells were plated in 6-well plates (1 × 10⁶ cells per well). After 24 h, cells were treated with ATRA (Sigma-Aldrich, stock solution 1 mM in ethanol absolute) for 6 h and then resuspended in Trizol for RNA extraction according to standard protocols. cDNA synthesis was performed with SuperScript™ II according to manufacturer’s protocol. SHOX expression was analyzed by quantitative PCR using gene-specific primers and normalized to HPRT and SDHA (Dataset EV4) with SensiFAST™ SYBR® Lo-ROX Kit (Bioline) in the 7500 Fast Real-Time PCR System (Applied Biosystems).

Luciferase assays

U2OS cells (human osteosarcoma cells, ATCC; mycoplasma-free) were cultured in DMEM containing 10% FBS and penicillin/streptomycin at 37°C, 5% CO₂, 95% humidity. Luciferase assays to test SHOX variants were performed as follows: Cells were seeded in 24-well plates at 1 × 10⁵ cells per well. After 24 h, cells were transfected with Lipofectamine 2000 (Invitrogen) according to standard protocols. For each well, cells were transfected with 300 ng of pcDNA4/TO empty vector (Life Technologies), pcDNA4/TO SHOX wild type or mutants; 300 ng of pGL3basic-FGFR3(-3,430/+464) (Decker et al, 2011) firefly luciferase reporter construct (Promega); 150 ng pRL-TK Renilla luciferase reporter construct (Promega). Mutations were introduced by site-directed mutagenesis (QuickChange Lightning Site-Directed Mutagenesis Kit). After 24 h of transfection, cells were lysed and luciferase activity measured with the Dual Luciferase Assay System (Promega) in a Berthold 96-microplate luminometer. The experiments were performed each time in triplicate.

Luciferase assays to test CYP26C1 variants were performed as follows: Cells were seeded in 96-well plates at 1 × 10⁵ cells per well. After 24 h, cells were transfected with Lipofectamine 2000 according to standard protocols. For each well, cells were transfected with 100 ng of pIRE2-EGFP empty vector (obtained from Dr. Thomas Boettger), pIRE2-EGFP-CYP26C1 wild type or mutants; 100 ng of Cignal RARE Reporter system plasmids (SA Biosciences). Mutations were introduced by site-directed mutagenesis. After 24-h transfection, cells were treated with 250 nM ATRA. After 24-h treatment, cells were lysed and assayed with the Dual Luciferase Assay System in a Berthold 96-microplate luminometer. The experiments were performed each time in triplicate.

Luciferase assays to test the SHOX promoter were performed as follows: Cells were seeded in 24-well plates at 1 × 10⁵ cells per well. After 24 h, cells were transfected with Lipofectamine 2000 according to standard protocols. For each well, cells were transfected with 500 ng of pGL3basic-empty vector (Promega) or pGL3basic-CNE3-SHOX promoter as previously described (Verdin et al, 2015); 50 ng of pRL-TK Renilla luciferase reporter construct. The CNE3 enhancer was chosen because it was reported with the highest activity on the SHOX promoter (Verdin et al, 2015). After 24-h transfection, cells were treated with 250 nM ATRA. After 24-h treatment, cells were lysed and assayed with the Dual Luciferase Assay System in a Berthold 96-microplate luminometer. The experiments were performed each time in triplicate.

Zebrafish experiments

Experiments were carried out in wild-type Danio rerio strains (AB, TL, Tübingen). Zebrafish were reared according to standard protocols. Morpholino were injected at one-cell stage embryos as previously described (Renz et al., 2015). The shox and cyp26c1 morpholinos were obtained from Gene Tools (Dataset EV4). Two splicing MOs were designed for shox knockdown: MO1 and MO2. In the main text, figures refer to MO1 (Figs 4 and 6). Specific phenotype was confirmed by MO2 (Appendix Fig S3). For cyp26c1 knockdown, we used a published MO (Liang et al, 2012). The standard control MO from Gene Tools was used as a control. Each embryo was injected with 1–2 ng of control, shox, or cyp26c1 MOs. Embryos for double knockdown experiments were injected with 100 pg of shox MO and/or 800 pg of cyp26c1 MO. RNA was extracted from 20–30 injected embryos at 36 h post-fertilization (hpf) with Trizol® according to standard protocols. cDNA synthesis was performed with SuperScript™ II according to manufacturer’s protocol. Effective splicing blocking was confirmed with test primers (Dataset EV4) and PCR products were cloned in pSTBlue1 vector (Novagen) and sequenced (Appendix Figs S4–S6). Sense-capped RNA of human SHOX and CYP26C1 wild type and mutants was synthesized using the mMESSAGE mMACHINE system (Ambion) from pCS2 (Hassel et al, 2009).

Expression analyses of shox after MO injection were performed as follows: RNA was extracted from 10–15 injected embryos at 55 h post-fertilization (hpf) with Trizol® according to standard protocols.
cDNA synthesis was performed with SuperScript™ II according to manufacturer’s protocol. shox expression was analyzed by quantitative PCR using gene-specific primers and normalized to eef1a and b-actin (Dataset EV4) with SensiFAST™ SYBR® Lo-ROX Kit (Bioline) in the 7500 Fast Real-Time PCR System (Applied Biosystems).

Luciferase experiments in zebrafish were performed injecting into one-cell stage embryos doses in the range of 1–2 nl of a 25 nM solution of Cignal RARE Reporter system plasmids and 1–2 ng of control MO or cyp26c1 MO. After 24-h injection, embryos were separated in groups of 20–30, lysed and assayed with a Dual Luciferase Assay System (Promega) in a Berthold 96-microplate luminometer.

Treatments of zebrafish embryos with RA were performed as follows: Wild-type embryos were left developing for 24 h post-fertilization. At 24 hpf, embryos were separated in groups of 10–15 and treated with mock control or ATRA for 6 h. Finally, RNA was extracted and used for shox expression analysis as described above.

Whole-mount in situ hybridization

Whole-mount in situ hybridization was performed as described previously (Jovett & Lettice, 1994). Gene-specific primers (Dataset EV4) were used to amplify col2a1 and shox cDNA. Amplicons were then cloned using the pSTBlue1 vector (Novagen). Digoxigenin-labeled RNA probes were synthesized using the DIG RNA Labeling Mix (Roche) with the MAXiScript® SP6/T7 Transcription Kit (Ambion). Images were taken with the microscope SZX16, Cell Imaging Software (Olympus). Pectoral fin area was measured with Fiji ImageJ (Schindelin et al., 2012).

Statistics

Samples for in vitro and zebrafish embryos experiments were randomly assigned to experimental groups, to processing order, and position in multi-well devices. No statistical method was used to predetermine sample size. Group sample sizes for experiments were chosen based on previous studies. Zebrafish embryos that died before analysis were excluded. Statistical analyses were performed using GraphPad Prism version 5 for Windows (GraphPad Software). Data were tested for normality using the D’Agostino and Pearson omnibus normality test. When data fitted normal distribution parameters, parametric tests were used; otherwise non-parametric tests were used. Specific tests are described for each group of data in the figure legends. Differences between two groups were analyzed by t-tested. Differences between three or more groups were analyzed by analysis of variance (ANOVA) test. For all experiments, data are expressed as the mean ± SD. P-values < 0.05 were considered significant.

Bioinformatics resources

Primers for cloning in situ hybridization probes, for sequencing, and for testing MO efficacy were designed using Primer3 (Untergasser et al., 2012). Primers for quantitative PCR were designed using Universal ProbeLibrary Assay Design Center (Roche). Protein and DNA schemes were drawn using Illustrator of Biological Sequences (IBS) (Liu et al., 2015). Mutations were tested with PolyPhen-2 (Adzhubei et al., 2013), Mutation Taster (Schwarz et al., 2010), SIFT (Vaslar et al., 2016), PROVEAN (Choi & Chan, 2015), and CADD (Kircher et al., 2014). Transcription factor binding sites were predicted using PROMO (Messeguer et al., 2002; Farré et al., 2003).

The paper explained

Problem

Mutations in the SHOX gene cause SHOX deficiency, the most frequent monogenic cause of short stature. The clinical severity of SHOX deficiency varies widely, ranging from short stature without skeletal signs to pronounced skeletal dysplasia. In rare cases, family members with identical mutations even have normal stature. So far, the underlying factors contributing to the phenotypic variability in individuals with SHOX deficiency are unknown. Genetic factors distinct from the SHOX gene may represent an important source of clinical variability in SHOX deficiency.

Genetic modifiers are genetic factors that influence the phenotypic expression of a disease-causing gene. Identifying genetic modifiers may result in a better understanding of the clinical variability of SHOX deficiency and enable a more accurate prediction of disease progression.

Results

We describe a large family where some individuals with a damaging SHOX mutation have normal stature, while other family members with the identical mutation have short stature and skeletal anomalies. To identify the genetic factors that could explain such phenotypic differences, a genetic analysis of this family was performed. This led to the identification of the retinoic acid-degrading enzyme CYP26C1 as a potential modifying genetic factor. Retinoic acid is the most active biological form of vitamin A and has been shown to play a role in skeleton formation. An excess of this molecule impairs limb development. Expression of CYP26C1 in human primary chondrocytes (cells that are involved in bone formation) could be demonstrated. CYP26C1 damaging mutations affect its ability to degrade retinoic acid, leading to higher levels of this vitamin A derivate. High levels of retinoic acid reduced SHOX expression in human primary chondrocytes, suggesting that CYP26C1 regulates SHOX expression within the retinoic acid signaling pathway. Two further families with SHOX and CYP26C1 mutations provided further evidence of the most severe phenotype. Zebrafish represents an animal model particularly suitable to study bone disorders. Decreasing both SHOX and CYP26C1 levels in zebrafish embryos led to smaller fins, further supporting that SHOX and CYP26C1 interact during skeletal development. Mutations in CYP26C1 result in increased retinoic acid levels which in turn decrease SHOX gene expression leading to more severe SHOX deficiency phenotypes.

Impact

This study represents an effort to understand the genetic causes of clinical variability. Unraveling factors which modify disease toward milder or more severe phenotypes may lead to novel therapeutic approaches. We provide evidence for CYP26C1 as a genetic factor influencing SHOX deficiency phenotypic outcomes through the retinoic acid signaling pathway. Manipulating vitamin A metabolism in SHOX deficiency patients may alleviate the skeletal abnormalities of this condition.

Study approval

Studies involving human material were approved by the Review Board Committee at the University Hospital Heidelberg and at the Hamamatsu University School of Medicine. Written informed consent was received from all participants prior to their inclusion in the study. The study was conducted in accordance with the
guidelines of the WMA Declaration of Helsinki and the Department of Health and Human Services Belmont Report. All animal experiments were conducted with approval of local Animal Care Committee and according to institutional guidelines.

**Acknowledgements**

We thank Tim Strom, Werner Blum, Miriam Rosilio, Dave Bunyan, Christine Fischer, Slavil Peykov, Herbert Steinbeisser, Thomas Holstein, and Christel Herold-Mende for support. We thank Nagarajan Paramasivam for calculating the CADD scores. Beate Niesler, Simone Berkel, and Antonio Marchini are acknowledged for comments on the manuscript. This study was supported by the DFG (Deutsche Forschungsgemeinschaft), the Medical Faculty of Heidelberg, and the DZHK Heidelberg (German Centre for Cardiovascular Research). AM is a member of the Hartmut Hoffmann-Berling International Graduate School of Molecular and Cellular Biology (HBIGS); GAR is a member of CellNetworks Cluster for Excellence of the University of Heidelberg, Germany. DH and GAR are members of the DZHK.

**Author contributions**

GAR designed the project. AM, GAR, and DH designed the experiments and analyzed the data. AM performed the cell culture experiments. LJ and AM performed the experiments in zebrafish embryos. SF-O, GB, MF, TO, and ED analyzed the data. AM performed the cell culture experiments. LJ and AM designed the experiments and contributed to the discussion and all authors commented the manuscript.

**Conflict of interest**

The authors declare that they have no conflict of interest.

**For more information**

ExAC
http://exac.broadinstitute.org
TGP
http://browser.1000genomes.org
EVS
http://evs.gs.washington.edu/EVS
PROMO
http://alggen.lsi.upc.es/cgi-bin/promo_v6/promo/promoinit.cgi?dirDB=TF_8.3
SHOX deficiency
http://www.ncbi.nlm.nih.gov/books/NBK1215/
X chromosome gene database
OMIM
http://www.omim.org/entry/312865

**References**


of retinal disease and a contributor to digenic Usher syndrome. J Clin Invest 120: 1812 – 1823


Grüneberg H (1963) The pathology of development; a study of inherited skeletal disorders in animals, pp. 309. New York, NY, USA: Wiley j


Rydehn AB, Waxman JS (2014) Cyp26 enzymes are required to balance the cardiac and vascular lineages within the anterior lateral plate mesoderm. Development 141: 1638 – 1648


open-source platform for biological-image analysis. Nat Methods 9: 676–682


**License:** This is an open access article under the terms of the Creative Commons Attribution 4.0 License, which permits use, distribution and reproduction in any medium, provided the original work is properly cited.