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Loss of BMPR2 Leads to High Bone Mass Due to Increased Osteoblast Activity

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RESEARCH ARTICLE

Loss of BMPR2 leads to high bone mass due to increased osteoblast activity

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ABSTRACT

Imbalances in the ratio of bone morphogenetic protein (BMP) versus activin and TGFβ signaling are increasingly associated with human diseases yet the mechanisms mediating this relationship remain unclear. The type 2 receptors ACVR2A and ACVR2B bind BMPs and activins but the type 2 receptor BMPR2 only binds BMPs, suggesting that type 2 receptor utilization might play a role in mediating the interaction of these pathways. We tested this hypothesis in the mouse skeleton, where bone mass is reciprocally regulated by BMP signaling and activin and TGFβ signaling. We found that deleting *Bmpr2* in mouse skeletal progenitor cells (*Bmpr2*-cKO mice) selectively impaired activin signaling but had no effect on BMP signaling, resulting in an increased bone formation rate and high bone mass. Additionally, activin sequestration had no effect on bone mass in *Bmpr2*-cKO mice but increased bone mass in wild-type mice. Our findings suggest a novel model whereby BMPR2 availability alleviates receptor-level competition between BMPs and activins and where utilization of ACVR2A and ACVR2B by BMPs comes at the expense of activins. As BMP and activin pathway modulation are of current therapeutic interest, our findings provide important mechanistic insight into the relationship between these pathways in human health.

KEY WORDS: BMP, Activin, BMPR2, Bone mass, Osteoblast, Osteoporosis

INTRODUCTION

All TGFβ superfamily cytokines [BMPs, GDFs, activin and activin-like ligands, such as GDF8 (also known as myostatin) and GDF11 (hereafter collectively referred to as 'activins' for simplicity), and TGFβ] can be categorized based on their activation of one of two distinct downstream pathways – either through SMAD1, SMAD5 and SMAD8 (hereafter denoted as SMAD1/5/8; note that SMAD8 is also known as SMAD9) or SMAD2 and SMAD3 (hereafter denoted as SMAD2/3), which are the canonical BMP signaling and activin and TGFβ signaling effectors, respectively (Lowery and de Caestecker, 2010). The canonical BMP pathway and activin and TGFβ pathway antagonize one another in numerous physiological contexts, including in early embryonic development, where SMAD2 antagonizes SMAD1 to establish body patterning (Yamamoto et al., 2009), in angiogenesis, where the balance of SMAD1/5/8 and SMAD2/3 establishes an angiogenic switch between activation and resolution phases (Goumans et al., 2003), in cell fate of type 2 alveolar epithelial cells, where trans-differentiation to a type 1 alveolar program is promoted by SMAD2/3, but restricted by SMAD1/5/8 (Zhao et al., 2013), during maintenance of epithelial cell polarity, where SMAD1/5/8 restricts the TGFβ-induced epithelial–mesenchymal transition (Saitoh et al., 2013), and during regulation of skeletal muscle mass, where SMAD1/5/8 and SMAD2/3 signaling inversely impact on muscle hypertrophy (Sartori et al., 2013; Winbanks et al., 2013). Moreover, imbalances in the ratio of TGFβ superfamily cytokines are increasingly associated with human diseases, including pulmonary and kidney fibrosis (Izumi et al., 2006; Nguyen and Goldschmeding, 2008), glaucoma (Wordinger et al., 2007; Zode et al., 2009), asthma (Stumm et al., 2014) and pulmonary arterial hypertension (Han et al., 2013; Morrell et al., 2001). However, the mechanisms that allow the BMP pathway and the activin and TGFβ pathway to antagonize one another remain unclear. For instance, competition for the shared transcription factor SMAD4 (Candia et al., 1997; Sartori et al., 2013) and regulation of downstream target genes (Oshimori and Fuchs, 2012) are likely insufficient general explanations because SMAD4, which is common to both pathways, is not always limiting (Piek et al., 1999) and target genes vary greatly by biological context (Massagué, 2012). Less attention has been paid, however, to potential regulation at the level of pathway activation via receptor utilization, which could influence the ratio of signaling upstream of any additional mechanisms.

TGFβ superfamily ligands signal through heteromeric combinations of type 1 and type 2 receptors, and both receptor types are essential to elicit downstream signaling. BMPs, activins and TGFβs generally utilize separate type 1 receptors (Hinck, 2012). In contrast, TGFβ has a unique type 2 receptor, whereas the type 2 receptors ACVR2A and ACVR2B (hereafter denoted ACVR2A/B) bind activins with high affinity but also bind BMPs with comparatively lower affinity. BMPs therefore can utilize ACVR2A/B and also the BMP-specific type 2 receptor, BMPR2, allowing for potential competition for ACVR2A/B by BMPs and activin based on the availability of BMPR2. We examined this hypothesis in the context of the mouse skeleton, taking advantage of the fact that postnatal bone formation and mineralization is under tight reciprocal regulation by BMPs and activins (Alves et al., 2013; Eijken et al., 2007; Fajardo et al., 2010; Ikenoue et al., 1999; Koncarevic et al., 2010; Li et al., 2013; Lotinun et al., 2010; Matsumoto et al., 2012; Mishina et al., 2004; Nicks et al.,

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2009; Pearsall et al., 2008; Perrien et al., 2007; Ruckle et al., 2009; Sakai et al., 2000; Sherman et al., 2013; Simic et al., 2006; Zhang et al., 2009; Zhao et al., 2002). Surprisingly, engineering specific deletion of BMPR2 in bone-forming cells (*Bmpr2*-cKO mice) selectively reduces activin pathway activation but has no apparent effect on BMP signaling. Additionally, *Bmpr2*-cKO mice develop substantially higher bone mass and bone mineral density by 9 weeks of age due to a higher bone formation rate, which is consistent with impaired activin signaling (Alves et al., 2013; Eijken et al., 2007; Ikenoue et al., 1999; Perrien et al., 2007). Our findings suggest a novel model whereby availability of BMPR2 in target cells permits partial segregation of ACVR2A/B to the activin pathway and increased BMP utilization of ACVR2A/B comes at the expense of activin signaling.

RESULTS

Type 2 BMP and activin receptor expression in bone cells

Each of the type 2 receptors *Bmpr2*, *Acvr2a* and *Acvr2b* are expressed in the postnatal skeleton (Fig. 1A; Liu et al., 2012). To remove BMPR2 from bone-forming cells, we crossed *Bmpr2*$^{floxed/floxed}$ mice to Paired-related homeobox gene 1 (*Prx1*)-Cre transgenic mice (Logan et al., 2002), which accomplishes widespread recombination and subsequent permanent loss of BMPR2 in osteoprogenitors, osteoblasts and osteocytes of the limb skeleton (Almeida et al., 2013; Logan et al., 2002). Loss of full-length BMPR2 in *Bmpr2*-cKO; *Prx1*-Cre mice (*Bmpr2*-cKO mice) was confirmed at the mRNA and protein levels (Gamer et al., 2011). Importantly, the bone-specific expression levels of *Acvr2a* and *Acvr2b* are unchanged in the bones of *Bmpr2*-cKO mice (Fig. 1B,C; Gamer et al., 2011).

Removal of BMPR2 selectively reduces activated SMAD2/3 in bone cells

We next performed western blots on marrow-free femora lysates to investigate whether signaling changes are associated with loss of BMPR2 expression and found that the level of phosphorylated (i.e. activated) SMAD1/5/8 was unchanged in *Bmpr2*-cKO mice at 4 weeks and 9 weeks of age (supplementary material Fig. S1A; Fig. 1D). Given that type 2 receptor occupancy is required for BMP signaling, ACVR2A/B appear to functionally compensate for loss of BMPR2 in bone cells *in vivo*. To examine whether this shift in receptor usage by BMPs has an effect on activin signaling, we next evaluated the level of phosphorylated (i.e. activated) SMAD2/3. Whereas activated SMAD2/3 was unaffected in *Bmpr2*-cKO mice at 4 weeks of age (supplementary material Fig. S1A), the level of activated SMAD2/3 was substantially reduced in *Bmpr2*-cKO mice at 9 weeks of age (Fig. 1E).

To examine, in a cell-specific manner, whether reduced SMAD2/3 activation is directly related to loss of BMPR2, we isolated primary osteoblasts from *Bmpr2*-floxed mice, which express each of the type 2 receptors *Bmpr2*, *Acvr2a* and *Acvr2b* (Fig. 2A), and induced recombination using adenoviral delivery of *Cre*. Loss of full-length BMPR2 was confirmed at the mRNA (Fig. 2B) and protein (Fig. 2C) levels. Consistent with our results in femora lysates, the basal level of canonical activin and TGFβ signaling was reduced, whereas canonical BMP signaling was unaffected in *Bmpr2*-cKO osteoblasts (Fig. 2C,D). This does not appear to be due to decreased activin ligand availability given that expression levels of the activin subunits *Inhba* and *Inhbb* and the antagonists inhibin α (*Inha*) and follistatin were unchanged in *Bmpr2*-cKO osteoblasts (supplementary material Fig. S2A). Note that because *Bmpr2*-cKO osteoblasts were responsive to exogenous activin A (supplementary material Fig. S2B), reduced basal SMAD2/3 activation in this cell population is not due to a requirement for BMPR2 in activin signaling.

High bone mass in *Bmpr2*-cKO mice due to an increased bone formation rate

We next examined the impact of loss of BMPR2 on postnatal bone physiology *in vivo*. Although limb skeletogenesis and expression of bone markers are unchanged in *Bmpr2*-cKO mice at birth (Gamer et al., 2011) and at 4 weeks of age (supplementary material Fig. S3A), both male and female *Bmpr2*-cKO mice display high trabecular bone mass and increased bone mineral

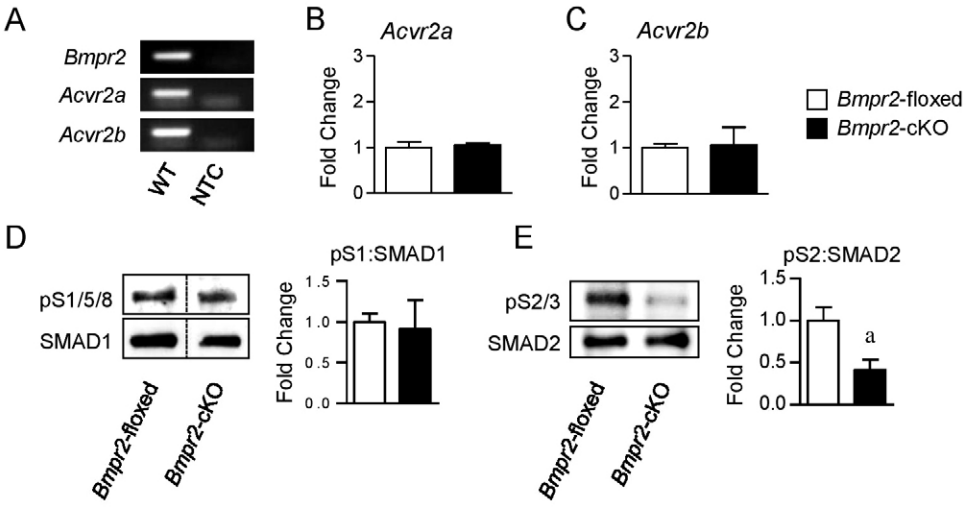

Fig. 1. Effects of removing *Bmpr2* in bone. (A) RT-PCR for *Bmpr2*, *Acvr2a*, and *Acvr2b* expression in wild-type (WT) 15-week-old mouse humerus. NTC, no DNA template control. (B,C) Expression of *Acvr2a* (B) and *Acvr2b* (C) in humerii of 4-week-old *Bmpr2*-cKO mice compared to *Bmpr2*-floxed littermates ($n \geq 4$ per genotype). (D,E) Western blots for phosphorylated isoforms of SMAD1/5/8 (pS1/5/8) and SMAD2/3 (pS2/3) relative to total SMAD1 or SMAD2. Lysates are from marrow-free femora of female mice at 9 weeks of age. Densitometry are normalized to *Bmpr2*-floxed from $n=3$ each genotype. $^{a}P<0.05$ compared with *Bmpr2*-floxed. The vertical bar in D indicates removal of intervening lane so that samples most representative of group mean are shown; the blot image with the intervening lane present is shown in supplementary material Fig. S1B.

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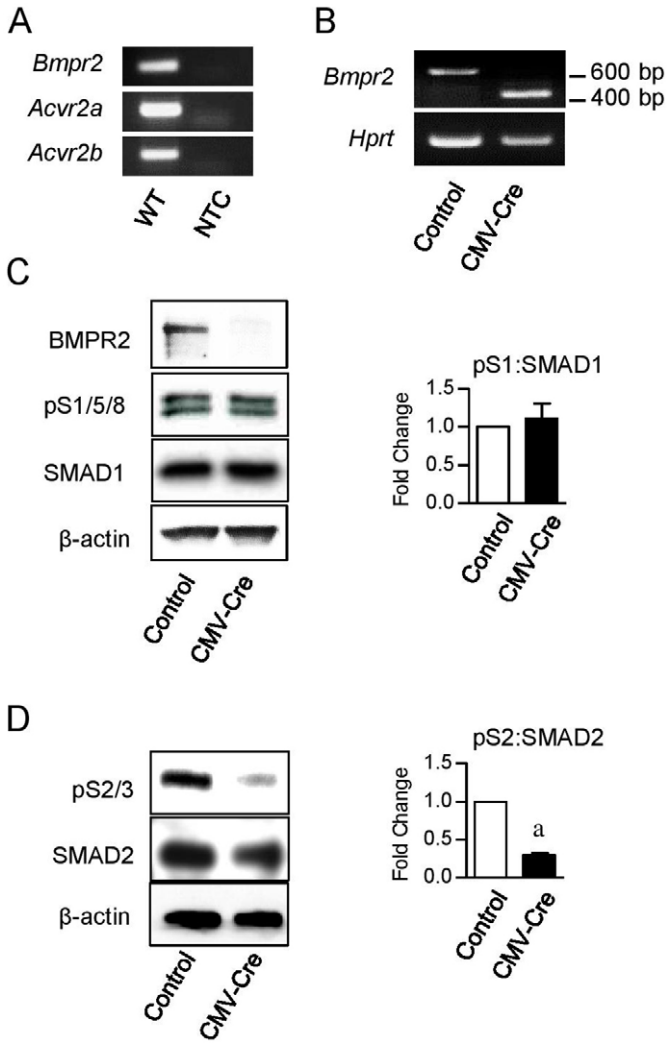

Fig. 2. Reduced activated SMAD2/3 in *Bmpr2*-cKO osteoblasts. (A) RT-PCR for *Bmpr2*, *Acvr2a* and *Acvr2b* expression in *Bmpr2*-floxed (WT) osteoblasts. NTC, no DNA template control. (B–E) Loss of full-length *Bmpr2* after adenoviral CMV-Cre delivery was confirmed at the mRNA level (B) by RT-PCR and protein level (C) by western blotting. Western blots for phosphorylated isoforms of SMAD1/5/8 (C, pS1/5/8) and SMAD2/3 (D, pS2/3) relative to total SMAD1 or SMAD2. Densitometry (graphs on the right) represents three independent isolation and transduction experiments normalized to control. *Hprt* and β-actin serve as loading controls in B, and C and D, respectively. [a]$P<0.002$ compared with control by paired Student's *t*-test.

density in the tibia by 9 weeks of age (Table 1; Fig. 3A). As an internal control, we examined the L5 vertebrae of *Bmpr2*-cKO mice (supplementary material Table S1), which is outside the *Prx1*-Cre expression domain. Bone mass is unaffected at this site, confirming that increased tibial bone mass is specifically due to Cre-dependent loss of *Bmpr2*.

We then performed detailed histomorphometric analyses (Dempster et al., 2013) to examine the cellular mechanism(s) responsible for the high bone mass in *Bmpr2*-cKO mice. Whereas osteoblast density and coverage of individual bone surfaces were unchanged in *Bmpr2*-cKO mice (Table 2), *in vivo* calcein labeling indicated that the percentage of bone surface undergoing mineralization tended to be higher in *Bmpr2*-cKO mice during the labeling period of 9 and 10 weeks of age. Bone mineral apposition rate was also significantly increased at this time (Table 2), resulting in a substantially elevated bone formation rate (Fig. 3B; Table 2). Although *Prx1*-Cre is not expressed in osteoclasts (Almeida et al., 2013; Kolanczyk et al., 2007), we examined whether *Bmpr2*-cKO mice displayed secondary defects in bone resorption. Osteoclast density, osteoclast coverage, and the percentage bone surface undergoing resorption were unchanged in *Bmpr2*-cKO mice (Table 2). Furthermore, the increase in bone volume in *Bmpr2*-cKO mice was proportional to the elevation in bone formation rate (Fig. 3C), indicating that bone resorption is unaffected by loss of BMPR2 in bone-forming cells.

Collectively, these findings indicate that the high bone mass we observe in in *Bmpr2*-cKO mice is due to increased individual osteoblast activity coincident with bone-specific reduction in activin signaling.

Activin sequestration has no effect on bone mass in *Bmpr2*-cKO mice

Our results led us to hypothesize that impaired activin signaling is responsible for the high bone mass seen in *Bmpr2*-cKO mice. We tested this idea by treating *Bmpr2*-floxed and *Bmpr2*-cKO mice with ACVR2B receptor decoy (ACVR2B-Fc), which sequesters activins with high affinity (Koncarevic et al., 2012; Sako et al., 2010), selectively reduces SMAD2/3 activation (supplementary material Fig. S4A,B; Ohsawa et al., 2006; Suragani et al., 2014; Zhou et al., 2010) and increases bone mass (Attie et al., 2013; Koncarevic et al., 2010). We reasoned that, if the reduced activin signaling underlies high bone mass in *Bmpr2*-cKO mice, then sequestration of activins with receptor decoy would have little or no additional effect on bone mass.

Table 1. Bone volume and mineral density of proximal tibiae from *Bmpr2*-floxed and *Bmpr2*-cKO mice

			Genotype			
Age of mice	Gender		*Bmpr2*-floxed	*Bmpr2*-cKO	Change (%)	*P* value
4 weeks	Male	BV/TV	0.0731±0.0028	0.0723±0.0036	−1.20	ns
		vBMD	71.11±7.531	72.90±6.541	2.50	ns
	Female	BV/TV	0.0673±0.0034	0.0769±0.0039	14.26	ns
		vBMD	68.20±6.040	84.31±6.613	23.62	ns
9 weeks	Male	BV/TV	0.1082±0.0092	0.1405±0.0054	29.85	0.0132
		vBMD	88.97±11.41	124.0±9.965	39.37	0.0432
	Female	BV/TV	0.0936±0.0073	0.1735±0.0108	85.36	<0.0001
		vBMD	75.28±7.357	164.5±10.87	118.51	<0.0001
15 weeks	Male	BV/TV	0.1341±0.0107	0.1835±0.0088	36.84	0.0024
		vBMD	128.4±12.33	185.3±10.29	44.31	0.0024
	Female	BV/TV	0.1004±0.0069	0.1522±0.0088	51.59	0.0009
		vBMD	95.52±8.975	155.9±11.25	63.21	0.0018

Change expressed as the percentage difference from gender-matched *Bmpr2*-floxed mice. n≥6 each group. BV/TV, bone volume/tissue volume; vBMD, volumetric bone mineral density expressed in mg hydroxyapatite per cubic centimeter; ns, not statistically significant.

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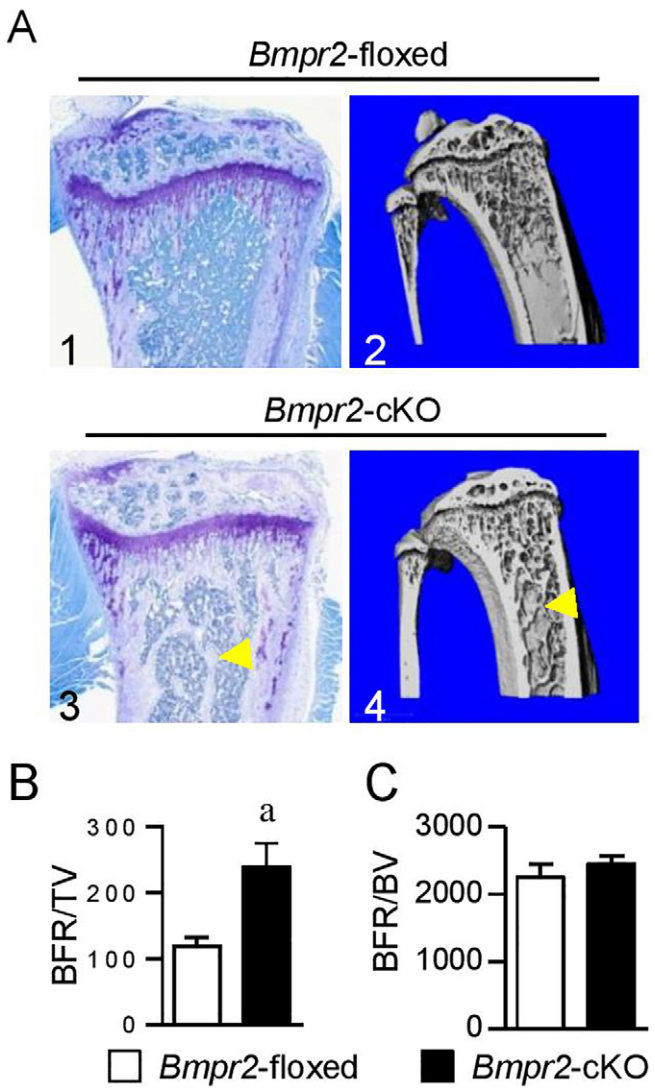

Fig. 3. High bone mass in *Bmpr2*-cKO mice due to increased bone formation rate. (A) Representative histological (1, 3) and μCT (2, 4) images of female 9-week-old *Bmpr2*-floxed (1, 2) and *Bmpr2*-cKO (3, 4) mice. The yellow arrowhead indicates increased trabecular bone volume in *Bmpr2*-cKO femur. (B,C) Histomorphometric analyses of 10-week-old *Bmpr2*-floxed and *Bmpr2*-cKO mice. $n \geq 8$ each group. BFR, bone formation rate; BV, bone volume; TV, tissue volume. $^{a}P<0.005$ compared with *Bmpr2*-floxed by Mann–Whitney test.

Consistent with previous reports where activin sequestration increased body weight gain and muscle mass (e.g. Cadena et al., 2010; Lee et al., 2012; Lee et al., 2005; Wang and McPherron, 2012), activin sequestration in *Bmpr2*-cKO mice increased the rate of body weight gain and the mass of the pectoralis major muscle (Fig. 4A,B), which are cellular targets outside the *Prx1*-Cre expression domain (Durland et al., 2008; Logan et al., 2002). In contrast, activin sequestration had a statistically indiscernible effect on trabecular bone mass and mineral density (Fig. 4C,D) in the tibiae of *Bmpr2*-cKO mice. This does not appear to be due to reduced activin ligand availability because the expression levels of activin ligands and their antagonists were unchanged in the bones of *Bmpr2*-cKO mice (supplementary material Fig. S3B); similarly, BMP ligand and antagonist expression levels were unaltered in *Bmpr2*-cKO bones (supplementary material Fig. S3B). The ability of the ACVR2B receptor decoy to substantially augment bone mass and mineral density as administered was simultaneously confirmed in a small cohort of *Bmpr2*-floxed mice (supplementary material Fig. S4E,F). The final bone mass and density attained after activin sequestration was the same in *Bmpr2*-floxed and *Bmpr2*-cKO mice, supporting the idea that impaired activin pathway activation underlies the bone phenotype we observe in *Bmpr2*-cKO mice.

DISCUSSION

Both BMPs and activins elicit pathway activation through interaction with heteromeric complexes containing type 1 and type 2 receptors (Zi et al., 2012). As represented in Fig. 5, structural differences mean that BMP ligands interact with BMP type 1 receptors (ALK1, ALK2, ALK3 and ALK6, also known as ACVRL1, ACVR1, BMPR1A and BMPR1B, respectively), whereas activin and related ligands (e.g. GDF8, GDF11) interact with activin type 1 receptors (ALK4 and ALK7, also known as ACVR1B and ACVR1C, respectively) (Hinck, 2012). This distinction stratifies the activation of SMAD1/5/8 versus SMAD2/3 downstream of BMPs and activins, respectively (Fig. 5A). Structural differences also govern interaction of ligands with type 2 receptors; only BMPs bind to the BMP-specific type 2 receptor BMPR2 and activate SMAD1/5/8 (Fig. 5A) (Hinck, 2012), whereas the type 2 receptors ACVR2A/B bind both BMPs and activins, engaging either SMAD1/5/8 or SMAD2/3, respectively (Fig. 5A) (Hinck, 2012). We hypothesized that BMPR2 is the preferred type 2 receptor for BMPs in bone, and that by removing BMPR2 from bone-forming cells we would create competition for a shared pool of ACVR2A/B molecules (Fig. 5B). Surprisingly, competition for ACVR2A/B led to a selective reduction in activin pathway signaling in bone with no apparent alteration in BMP signaling. The high bone mass phenotype we observed in *Bmpr2*-cKO mice is consistent with previous reports detailing the negative effect of activin on bone formation and matrix mineralization (Alves et al., 2013; Bialek et al., 2013; Eijken et al., 2007; Fajardo et al., 2010; Ikenoue et al., 1999; Koncarevic et al., 2010; Lotinun et al., 2010; Pearsall et al., 2008; Perrien et al., 2007; Ruckle et al., 2009; Sherman et al., 2013).

Collectively, our findings suggest that the availability of BMPR2 in bone-forming target cells provides a novel mechanism for establishing the relative amount of BMP to activin signaling. When BMPR2 is reduced or absent, increased BMP utilization of ACVR2A/B occurs at the expense of activin signaling. Although unexpected, this finding is supported by the fact that, in general, the affinity of BMPs for ACVR2A/B is in the same range as the affinity for BMPR2 (Allendorph et al., 2006; Berasi et al., 2011; Daly and Hearn, 2006; Greenwald et al., 2003; Greenwald et al., 2004; Heinecke et al., 2009; Hu et al., 2010; Isaacs et al., 2010; Kirsch et al., 2000; Knaus and Sebald, 2001; Koncarevic et al., 2010; Rosenzweig et al., 1995; Sako et al., 2010; Sengle et al., 2008), and BMPs also possess a flexible mode of receptor complex assembly, which might enhance their ability to compete with activins that have only a single mode of complex assembly (Hinck, 2012). Because bone cells have abundant type 1 BMP receptors (ALK2, ALK3 and ALK6, BioGPS Database), pre-formed receptor complexes might also be biased toward BMP binding. We do not believe our finding is only applicable to bone cells, as Piek et al. have reported that BMP7 can effectively compete with activin A for utilization of ACVR2A/B in human embryonic carcinoma cells (Piek et al., 1999), and direct competition for ACVR2A/B between BMP7 and the activin-like

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Table 2. Histomorphometric analysis of *Bmpr2*-floxed and *Bmpr2*-cKO mice

Parameter	Genotype		Change (%)	*P* value
	Bmpr2-floxed	*Bmpr2*-cKO		
Osteoblast density (N.Ob/B.Pm, per mm)	10.78±1.180	12.06±2.478	11.87	0.6839
Osteoblast coverage (Ob.S/BS, %)	13.26±1.481	15.41±3.270	16.21	0.6040
Surface undergoing mineralization (MS/BS, %)	32.39±3.103	38.75±1.676	19.64	0.0658
Trabecular mineral apposition rate (MAR, μm/day)	2.871±0.1994	3.571±0.1978	24.38	0.0278
Trabecular bone formation rate (BFR/BS, $\mu m^3/\mu m^2$/day)	327.1±23.33	498.4±25.89	52.37	0.0002
Endocortical mineral apposition rate (MAR, μm/day)	0.97±0.36	2.25±0.19	131.96	0.0070
Osteoclast density (N.Oc/B.Pm, per mm)	1.327±0.2539	1.372±0.2945	3.39	0.9126
Osteoclast coverage (Oc.S/BS, %)	3.425±0.5214	3.321±0.6657	−3.04	0.9091
Surface undergoing erosion (ES/BS, %)	1.803±0.3247	1.453±0.3062	−19.41	0.4521

Change expressed as the percentage difference from *Bmpr2*-floxed mice. n ≥ 8 for each group and parameter. B.Pm, bone perimeter; BFR, bone formation rate; BS, bone surface; ES, eroded surface; MAR, mineral apposition rate; MS, mineralizing surface; N.Ob, number of osteoblasts; N.Oc, number of osteoclasts; Ob.S, osteoblast surface; Oc.S, osteoclast surface.

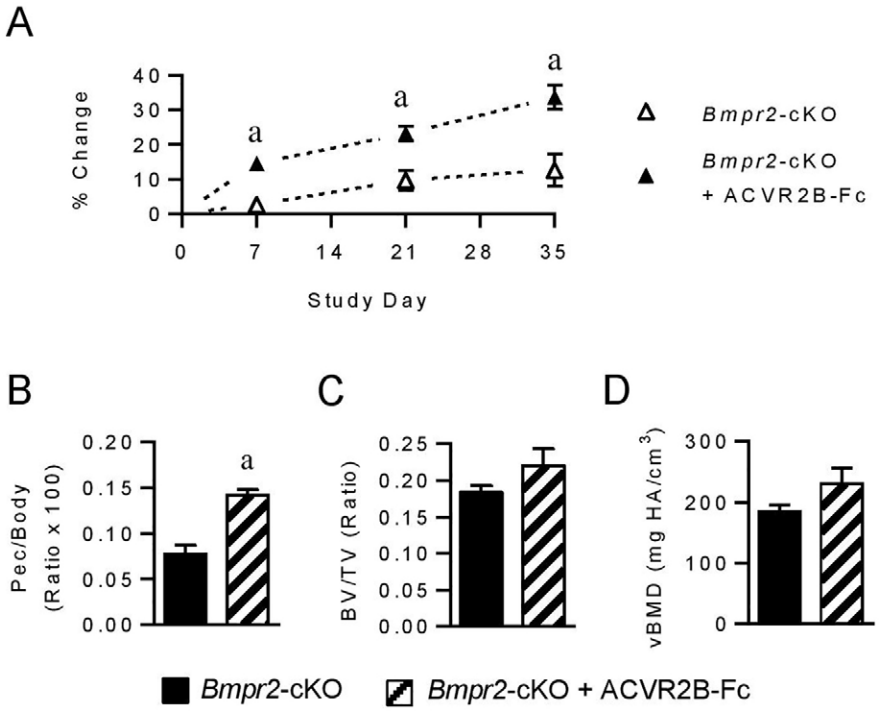

Fig. 4. Effects of activin receptor decoy administration in *Bmpr2*-cKO mice. (A) Rate of body weight gain over a 5-week ACVR2B-Fc treatment period in *Bmpr2*-cKO mice expressed as the mean percentage change from subject body weight on study day 0. (B–D) Effect of activin sequestration on dry weight of pectoralis major (B) and tibia trabecular bone volume (C) and bone mineral density (D) in *Bmpr2*-cKO mice. Data for untreated cohorts in C,D are also reported in Table 1. $n \geq 3$ for all groups. [a]$P<0.05$ compared with untreated *Bmpr2*-cKO.

ligand GDF8 has been reported by Rebbapragada et al. during adipogenesis (Rebbapragada et al., 2003).

Our model would predict that changes in either receptor or ligand availability (for instance, through altered expression or sequestration by extracellular antagonists) have the potential to modulate the balance of BMP versus activin signaling in a given cell. With respect to bone physiology, it is unknown whether the expression profile of type 2 BMP and activin receptors changes with age, but the ligands available to interact with type 2 BMP and activin receptors do change: BMP levels decline with age, which correlates with a decrease in ability to form new bone (Moerman et al., 2004; Syftestad and Urist, 1982), and there is a dramatic increase in circulating activin levels in adults of both genders, especially in the last decades of life (Baccarelli et al.,

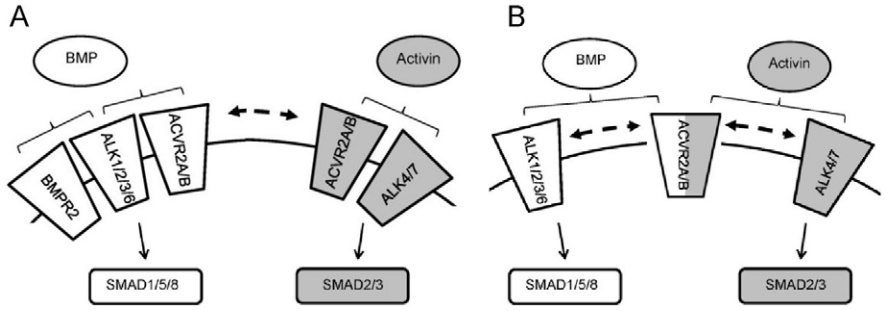

Fig. 5. Model of BMP and activin signaling. (A,B) Schematic of BMP and activin signaling in wild-type (A) and *Bmpr2*-cKO (B) cells. The type 2 receptors ACVR2A/B are shared between the BMP and activin pathways and elicit activation of either SMAD1/5/8 (in complex with the type 1 receptors ALK1, ALK2, ALK3 or ALK6) or SMAD2/3 (in complex with the type 1 receptors ALK4 or ALK7) in response to BMP or activin ligands, respectively (A). Removal of the type 2 receptor BMPR2 requires all BMP and activin signaling to utilize a common pool of ACVR2A/B molecules (B).

2001; Hurwitz and Santoro, 2004). In other contexts, serum immune-reactive activin is elevated further in patients with several diseases including hyperthyroidism, liver cirrhosis, chronic renal failure, advanced solid cancers, septicemia, acute inflammation and type 2 diabetes (de Kretser et al., 2011; Harada et al., 1996; Ueland et al., 2012). Our findings in bone raise the possibility that type 2 receptor segregation and/or competition could be a generalized mechanism by which BMP and activin signaling interact. Given that modulation of both BMP and activin pathways is of current clinical interest, a more detailed understanding of the molecular relationship between these pathways might provide insight into the development of new therapeutic strategies that are widely applicable.

MATERIALS AND METHODS

Mouse lines

The generation of the mouse lines used in this study, *Bmpr2*^{floxed} (Beppu et al., 2005), *Prx1*-Cre transgenic (Logan et al., 2002), and Rosa26$^{mT(mG)}$ Cre reporter mice (Muzumdar et al., 2007), have been described previously. For experimental study groups, female *Bmpr2*$^{floxed/floxed}$ mice were bred to male *Bmpr2*$^{floxed/floxed}$; *Prx1*-Cre mice to generate *Bmpr2* conditional knockout (*Bmpr2*-cKO) mice and Cre-negative *Bmpr2*$^{floxed/floxed}$ littermates (*Bmpr2*-floxed). Animals were killed by exposure to CO_2 followed by cervical dislocation. All animal experiments were performed under supervision of the Harvard Medical Area Standing Committee on Animals.

Osteoblast culture

Osteoblasts were isolated from the calvariae of newborn *Bmpr2*$^{floxed/floxed}$; *Rosa26*$^{mT(mG)}$ as described previously (Bakker and Klein-Nulend, 2003). Isolated cells were pooled to reduce technical variability, expanded and then split prior to delivery of adenovirus (Ad-CMV-Cre or Ad-CMV-Empty; Baylor College of Medicine Vector Development Lab, Houston, Texas, USA); recombination was monitored by expression of GFP from the *Rosa26*$^{mT(mG)}$ reporter. Cells were maintained in Dulbecco's modified Eagle's medium (DMEM) supplemented with 10% fetal bovine serum (FBS; Gibco, Life Technologies, Grand Island, New York, NY); for signaling studies, cells were serum-restricted in DMEM plus 2% FBS, treated with 100 ng/ml activin A (R&D Systems) for 1 hour, lysed and prepared for western blotting as described below.

Western blots

Western blotting was performed on protein isolates from primary osteoblasts and marrow-free femora lysed in RIPA lysis buffer supplemented with Halt Protease & Phosphatase Inhibitor Cocktail (Thermo, Waltham, MA). Prior to lysis, femora were cleaned of soft tissue, opened to expose the medullary cavity, centrifuged at 500 ***g*** for 2 minutes to separate marrow, and then homogenized using a Bullet Blender (Next Advance, Averill Park, New York, NY). Blotting was performed using antibodies against the following proteins: phosphorylated (phospho)-SMAD1/5/8 (Cell Signaling, Danvers, MA), phospho-SMAD2/3 (Cell Signaling), SMAD1 (Cell Signaling), SMAD2/3 (Cell Signaling), β-actin (Sigma-Aldrich, St. Louis, MO), mouse IgG conjugated to horseradish peroxidase (HRP; KPL, Gaithersburg, MD) and rabbit IgG conjugated to HRP (Cell Signaling). Blots were developed using SuperSignal West Femto Substrate (Thermo). For each time point, *Bmpr2*-floxed and *Bmpr2*-cKO samples were run on the same gels to avoid across-gel comparison and dashed lines are included to indicate where lanes are cropped to aid clarity of presentation. Signaling analyses were quantified by determining the ratio of the phosphorylated (active) SMAD isoform to the total pool. The validity of this approach is confirmed by the fact that total SMAD expression levels are unchanged relative to β-actin in *Bmpr2*-cKO mice (supplementary material Fig. S1C,D) or with ACVR2B-Fc treatment (supplementary material Fig. S1E). Band densities were quantified using ImageJ and normalized to *Bmpr2*-floxed control. Data are expressed as group mean±s.e.m. based on each individual ratio. Stripping of membranes was accomplished using Restore Western Blot Stripping Buffer (Thermo).

Physiological analysis of bones

Standard histomorphometric analyses were performed on the proximal tibiae of 10-week-old female mice. To examine mineral apposition rate, mice were injected intra-peritoneally with 20 µg calcein (Sigma-Aldrich) per gram body weight at 7 days and 2 days prior to killing. Tibiae were collected immediately following killing, fixed in 70% ethanol, embedded in methylmethacrylate without decalcification and sectioned. Parameters were examined by a blinded scorer. The reader is directed to an excellent guide to bone histomorphometry (Dempster et al., 2013).

For micro-computed tomography (µCT) analyses, bones were collected immediately after killing and fixed in 10% neutral buffered formalin (Millipore, Billerica, MA). Bones were exchanged into 70% ethanol, stored at 4℃, and analyzed at the HSDM µCT Core Facility on a SCANCO µCT 35 scanner. Bone volume and mineral density were determined using the following settings: FOV/diameter: 7168 µm; voxel size: 7.0 µm; slice increment: 7.0 µm; projections: 500; sample time: 600 ms; µ-scaling: 4096; energy: 55,000 V; intensity: 145 µA. The reader is directed to an excellent guide to µCT in rodents (Bouxsein et al., 2010).

Gene expression analyses

RNA was collected from isolated osteoblasts and marrow-free humerii from 4-week-old female or 15-week-old male mice using the RNEasy Plus Universal Kit (QIAGEN, Valencia, CA) after homogenization using a Bullet Blender (Next Advance). Reverse transcription was performed using an EcoDry Premix Kit (Clontech, Mountain View, CA). Quantitative PCR was performed using FastStart Universal SYBR Green Master Mix (Rox) (Roche, Nutley, NJ) on a StepOnePlus Real-Time PCR System (Applied Biosystems, Life Technologies) and primers as outlined in supplementary material Table S2. All primers were designed to cross exon boundaries and total RNA was treated with DNase. Data were analyzed by the comparative Ct method relative to cyclophilin B (*Ppib* Taqman probe, Mm00478295_m1, Life Technologies) using the equation $2^{-\Delta\Delta Ct}$.

ACVR2B receptor decoy administration

Recombinant human ACVR2B extracellular domain fused to human IgG1 Fc (ACVR2B-Fc) was obtained as a gift from Regeneron Pharmaceuticals (Tarrytown, New York, NY). Using the dosing scheme described by Koncarevic et al. (Koncarevic et al., 2010), male *Bmpr2*-floxed and *Bmpr2*-cKO littermates were treated with 10 µg ACVR2B-Fc in sterile PBS per gram body weight by intraperitoneal injection for time period indicated. Body weight was taken on day 0 and prior to each treatment. Upon killing, bones were processed for µCT as described above and the pectoralis major muscle was excised, dried at 37℃ for two days, then weighed.

Statistical analyses

All quantitative data are expressed as mean±s.e.m. except where indicated. Statistical significance was determined by unpaired Student's *t*-test for individual pairwise comparisons and one-way ANOVA with *post hoc* Newman–Keuls correction for multiple pairwise comparisons using GraphPad Prism (except where indicated). $P<0.05$ was considered significant.

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Competing interests

The authors declare no competing or financial interests.

Author contributions

J.W.L., G.I., L.G. and V.R. designed the study; J.W.L., G.I., S.L., V.S.S., S.O. and K.C. performed the experiments; all authors analyzed and interpreted the results; J.W.L. and V.R. wrote the manuscript; all authors read and approved the final version.

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