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ORIGINAL ARTICLE

Muscular contributions to dynamic dorsoventral lumbar spine stiffness

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Abstract Spinal musculature plays a major role in spine stability, but its importance to spinal stiffness is poorly understood. We studied the effects of graded trunk muscle stimulation on the in vivo dynamic dorsoventral (DV) lumbar spine stiffness of 15 adolescent Merino sheep. Constant voltage supramaximal electrical stimulation was administered to the L3–L4 interspinous space of the multifidus muscles using four stimulation frequencies (2.5, 5, 10, and 20 Hz). Dynamic stiffness was quantified at rest and during muscle stimulation using a computer-controlled testing apparatus that applied variable frequency (0.46–19.7 Hz) oscillatory DV forces (13-N preload to 48-N peak) to the L3 spinous process of the prone-lying sheep. Five mechanical excitation trials were randomly performed, including four muscle stimulation trials and an unstimulated or resting trial. The secant stiffness (k_y = DV force/L3 displacement, kN/m) and loss angle (phase angle, deg) were determined at 44 discrete mechanical excitation frequencies. Results indicated that the dynamic stiffness varied 3.7-fold over the range of mechanical excitation frequencies examined (minimum resting $k_y = 3.86 \pm 0.38$ N/mm at 4.0 Hz; maximum $k_y = 14.1 \pm 9.95$ N/mm at 19.7 Hz). Twenty hertz muscle stimulation resulted in a sustained supramaximal contraction that significantly ($P < 0.05$) increased k_y up to twofold compared to rest (mechanical excitation at 3.6 Hz). Compared to rest, k_y during the 20 Hz muscle stimulation was significantly increased for 34 of 44 mechanical excitation frequencies (mean increase = 55.1%, $P < 0.05$), but was most marked between 2.55 and 4.91 Hz (mean increase = 87.5%, $P < 0.05$). For lower frequency, sub-maximal muscle stimulation, there was a graded change in k_y, which was significantly increased for 32/44 mechanical excitation frequencies (mean increase = 40.4%, 10 Hz stimulus), 23/44 mechanical excitation frequencies (mean increase = 10.5%, 5 Hz stimulus), and 11/44 mechanical excitation frequencies (mean increase = 4.16%, 2.5 Hz stimulus) when compared to rest. These results indicate that the dynamic mechanical behavior of the ovine spine is modulated by muscle stimulation, and suggests that muscle contraction plays an important role in stabilizing the lumbar spine.

Keywords Biomechanics · Electromyography · Lumbar spine · Dynamic stiffness · Muscle stimulation

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Introduction

Maintenance of posture and performance of purposeful trunk motion are the result of coordinated load sharing between passive and active paraspinal tissues that act to balance the external loads [26]. Indeed, the spinal musculature plays a major role in spine stability [7, 8]. Physiologic deformation of the viscoelastic structures of the spine have also been shown to elicit active and reflexive muscular contraction of the multifidus and longissimus muscles, which in turn stiffen and stabilize the spine during movements [6]. Disturbances in the musculoskeletal system that result in excessive load sharing, abnormal motion and higher strains in the highly innervated lumbar spinal soft tissues have been suggested as possible causes of some low back disorders and chronic low back pain [13, 25]. Clinically, increased levels of muscle co-activation may also constitute an objective indicator of the dysfunction in the passive stabilizing system of the lumbar spine [2].

In vivo biomechanical assessments provide non-invasive estimates of spinal stiffness that, together with other objective tests and outcome measures, may help clinicians to discriminate and treat patients with spinal disorders [4]. Studies investigating posteroanterior (PA) spinal stiffness of the prone-lying human spine using mobilization assessment apparatuses have shown relationships between PA stiffness and a number of factors, including spinal level, body type, body position, and lumbar extensor muscle activity [1, 3, 19, 20, 28, 30]. Results from some in vivo studies have also reported an increased PA spine stiffness to voluntary contraction of the lumbar extensor muscles [3, 17, 28]. Spinal stiffness has been hypothesized to be an important parameter for clinicians to discriminate between patients with low back pain and asymptomatic subjects [4, 15, 22]. If so, then the role of muscular stabilization of the lumbar spine should assist in low back pain diagnosis and treatment.

To our knowledge, there have been no comprehensive biomechanical studies examining the dynamic mechanical response of the lumbar spine during graded muscle stimulation. The purpose of the current experimental animal study was to quantify the effects of muscle activation on in vivo ovine lumbar spine stiffness. Dorsoventral (DV) lumbar spine stiffness was characterized over a 0.46–19.7 Hz oscillatory mechanical excitation frequency range using a dynamic loading apparatus. Supramaximal muscle activation was performed using constant voltage, variable pulse frequency stimulation, and the resulting DV stiffness was compared to unstimulated (resting) stiffness.

Methods

Animal preparation

Fifteen healthy, adolescent Merino sheep (mean 47.7 kg, SD 4.9 kg) were examined using a protocol approved by an accredited animal ethics committee. A plain lateral X-ray film was taken to verify normal lumbar spine anatomy. The sheep were fasted for 24 h prior to surgery and anesthesia was induced with an intravenous injection of 1 g thiopentone. General anesthesia was maintained after endotracheal intubation by 2.5% halothane and monitored by pulse oximetry and end tidal CO_2 measurement. Animals were ventilated and the respiration rate was linked to the tidal volume keeping the monitored CO_2 between 40 and 60 mmHg. The anesthetized sheep were placed prone on a stainless steel operating table, which included a rigid (wood) support beneath the abdomen (just caudal to the ribcage). The support was designed to orient the long axis of the sheep spine parallel to the operating table and perpendicular to the load actuator and secondarily to stabilize the trunk. Foam blocks were also placed on either side of the sheep abdomen to further stabilize the trunk along the medial–lateral axis (Fig. 1). With the animals in this standardized prone-lying position, an adhesive earthing pad was applied to the groin, and a 1.5 cm region of the bony prominence of the L3 spinous process was exposed using electrocautery.

Muscle stimulation and electromyography

Two pairs of 28-gauge concentric monopolar electrical stimulation electrodes were placed bilaterally at the L3–L4 interspinous space of the multifidus muscle. These muscles are adjacent to the vertebrae, and play a key role in stabilizing the spine and controlling movement at the facet joints. The stimulation electrodes were spaced 2 cm apart and the leads were secured to the draping with clips and adhesive tape. Four 28-gauge concentric bipolar needle electromyographic (nEMG) recording electrodes (Model EL451, Biopac Systems, Inc. Santa Barbara, CA) were then inserted bilaterally into the multifidus musculature adjacent to L3 and L4. The nEMG electrodes were 460 μm in diameter and 3.0 cm long with a recording area of 0.06 mm^2. A monopolar ground needle electrode (Model EL452, Biopac Systems, Inc.) was also inserted into the fascia adjacent to the sheep trochanter.

Constant voltage (5 V), monophasic, symmetric pulse excitation was applied to each stimulating electrode using a programmable pulse stimulation module

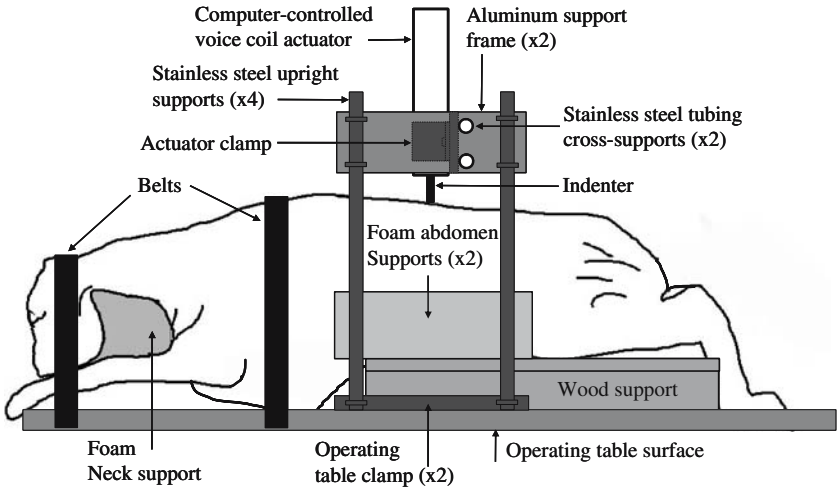

Fig. 1 Schematic diagram of the computer-controlled mechanical testing apparatus positioned over the ovine L3 spinous process of the prone-lying sheep. The apparatus consisted of an actuator assembly comprised a voice coil actuator, linear variable differential transformer, load cell, and stainless steel indenter. The actuator assembly was attached to a stainless steel and aluminum load frame that was rigidly mounted to the stainless steel operating table. Wood and foam supports were used to position and stabilize the sheep trunk and abdomen during the application of dorsoventral forces (Adapted from [10])

(STM100C, Biopac Systems, Inc, Goleta, CA) and an isolated voltage stimulus adaptor (STMISOD, Biopac Systems, Inc, Goleta, CA). The load resistance was 50 ohm, resulting in a 100 mA current applied to each stimulating electrode, which is equivalent to a supramaximal neuromuscular stimulation [29]. Stimulation intensity was modulated by varying the pulse duration, and four uniform pulse durations (25, 50, 100, and 200 ms) were applied resulting in muscle stimulation at 20, 10, 5, and 2.5 Hz.

Mechanical excitation

A custom, computer-controlled mechanical testing apparatus was used to generate mechanical excitation force–time profiles with varying frequency. The apparatus was comprised of a linear voice coil actuator (model LA25-42, BEI Technologies Inc., Ashford, Kent, UK) and a programmable, pulse width modulated servo amplifier, voice coil drive controller (model VCA100, BEI Kimko Magnetics, San Marcos, CA). The voice coil had a continuous stall force of 84 N and total stroke of 25.4 mm. A 665 N load cell (MLP-150, Transducer Techniques, Temecula, CA) and a ± 25 mm linear variable displacement transducer (model S1D, Instruments & Control, Inc., Branford, CT) were used to measure the actuator force and displacement signals, respectively. Force and displacement signals were amplified using a dual channel, digital programmable gain amplifier (model PGA204, gain = 1,000, Burr-Brown, Tucson, AZ).

DV forces were applied directly to the exposed L3 spinous process via a 12.7 mm-diameter stainless-steel indenter rod equipped with a slotted tip that cradled the spinous process. The slotted tip minimized problems associated with the indenter sliding off the sheep spinous processes, which are more slender than their human counterpart. DV forces (–13 N preload to –48 N peak) were applied using a swept-sine (harmonic) mechanical excitation protocol (0.46–19.7 Hz. This pseudo-chirp mechanical excitation protocol included a 1.3 s initial ramp loading phase and a 1.3 s final ramp unloading phase at 10 N/s. The peak DV forces were approximately 10% of the mean animal body weight, which is consistent with the magnitude of PA mobilization forces used by clinicians in the assessment of lumbar spine disorders [16]. Based on repeated measurements in 15 animals, the average coefficient of variation in the DV stiffness ranged from 4.4% (2 Hz mechanical excitation) to 7.3% (12 Hz mechanical excitation). Additional details of the mechanical testing apparatus can be found elsewhere [10].

Data collection and analysis

Muscle stimulation and unstimulated (rest) trials were applied in a randomly determined order for each animal. Mechanical excitation force and displacement were recorded for 22 s at a sampling frequency of 2,500 Hz using a 16-bit data acquisition system (Model MP150, Biopac Systems, Inc. Santa Barbara, CA). Muscle stimulation pulse and multifidus nEMG responses were

sampled at 1,250 Hz. The raw, needle nEMG was rectified and filtered to create an integrated electromyographic signal (iEMG).

Stiffness values were computed as the relative change in load and displacement using a custom Matlab® program (MathWorks, Natick, MA). DV stiffness (peak–peak force/peak–peak displacement, N/mm) was determined for 44 discrete time intervals over the 22 s pseudo-chirp excitation protocol (0.46–19.7 Hz). For each interval, the loss angle (phase angle, deg) between the peak force and peak displacement was determined using the relationship $360\Delta t/T$, where Δt (s) is the phase difference, and T (s) is the period. Descriptive statistics (mean, standard deviation, SD, min, max) were computed and differences between the unstimulated and muscle stimulated trials for each mechanical excitation frequency were assessed using a paired-observation's t test (two-tailed POTT). Statistical significance was $P < 0.05$.

Results

Typical force and displacement responses for the resting and muscle stimulation (20 Hz protocol) trials are shown in Fig. 2. Variations in displacement magnitude are evident throughout the swept-sine mechanical testing protocols. Muscle stimulation artifacts are also evident in the muscle stimulation protocol force–time histories. At 20 and 10 Hz, the supramaximal muscle stimulation resulted in a sustained muscle contraction, whereas the lower frequency electrical stimulation (5 and 2.5 Hz) produced rhythmic twitch responses indicative of wave summation below the threshold of tetany. In some animals, oscillatory iEMG activity was evident in the resting protocol, mirroring the mechanical oscillations. Mechanical excitation combined with muscle stimulation produced significantly (ANOVA, $P < 0.05$) greater peak iEMG responses in comparison to mechanical excitation without muscle stimulation (Fig. 3).

The stiffness–frequency response curves obtained for the unstimulated and four muscle stimulation protocols are summarized in Fig. 4. The in vivo DV dynamic stiffness k_y of the unstimulated ovine spine varied with mechanical excitation frequency, increasing approximately 3.7-fold over the 0.46–19.7 Hz mechanical excitation frequency range. The minimum and maximum resting k_y were 3.86 ± 0.38 and 14.1 ± 9.95 N/mm at 4.0 and 19.7 Hz, respectively. At the minimum stiffness (valley in the resting stiffness–frequency response curve, 4.0 Hz), the force and displacement were out of phase by 58.4° (SD 10.6). The maximum resting phase angle change was 85.6° (SD 6.2) at 6.28 Hz. Both resting and muscle stimulation trials showed a decreasing stiffness (up to 38.5%, 20 Hz muscle stimulation) for quasi-static mechanical excitation below 1 Hz (most likely due to creep deformation).

The 20 and 10 Hz muscle stimulation protocols produced the most dramatic increase in DV stiffness. Compared to rest, the sustained supramaximal 20 Hz muscle stimulation increased k_y up to twofold (3.6 Hz mechanical excitation frequency). k_y during the 20 Hz muscle stimulation was significantly increased (mean increase = 55.1%, SD 30.5%) for the majority of mechanical excitation frequencies examined (34 of 44), but was most marked between 2.55 and 4.91 Hz (mean increase = 87.5%). Table 1 summarizes the mean stiffness values (over the mechanical excitation range, 2.55–4.91 Hz) obtained for each animal. For lower frequency muscle stimulation, there was a graded change in k_y, which was significantly increased for 32/44 mechanical excitation frequencies (mean increase = 40.4%, SD 26.7%, 10 Hz muscle stimulation), 23/44 mechanical excitation frequencies (mean increase = 10.5%, SD 12.1%, 5 Hz muscle stimulation), and 11/44 mechanical excitation frequencies (mean increase = 4.16%, SD 9.71%, 2.5 Hz muscle stimulation) when compared to rest. Between 2.55 and 4.91 Hz, the mean stiffness increase (compared to rest) was 68.7, 20.4, and 11.1% for the 10, 5, and 2.5 Hz muscle stimulation trials, respectively (Fig. 5). Table 2 summarizes the pairwise statistical comparison (P values) of the dynamic DV stiffness response of the sheep spine (muscle stimulation vs. rest).

Muscle stimulation not only increased k_y, but also shifted (increased) the frequency corresponding to the minimum k_y. For the 20 and 10 Hz muscle stimulation protocols k_y was a minimum at 4.91 Hz (mean k_y = 7.24, SD 1.94 N/mm) and 4.91 Hz (mean k_y = 6.51, SD 3.78 N/mm), respectively. There was no appreciable frequency shift for the 5 Hz muscle stimulation protocol (mean k_y = 4.88, SD 0.92 N/mm at 4.00 Hz) or the 2.5 Hz muscle stimulation protocol (mean k_y = 4.32, SD 0.66 N/mm at 4.25 Hz). The phase angles corresponding to the minimum stiffness values were 39.9 (SD 21.2°), 45.9 (SD 24.9°), 41.2 (SD 26.0) and 51.8 (SD 25.1) for the 20, 10, 5.0 and 2.5 Hz muscle stimulation protocols, respectively. The maximum phase shift occurred at a mechanical excitation frequency of 7.0 Hz (85.6°, 76.3°, 74.5°, 76.5° for the 2.5, 5.0, 10, and 20 Hz muscle stimulation protocols, respectively).

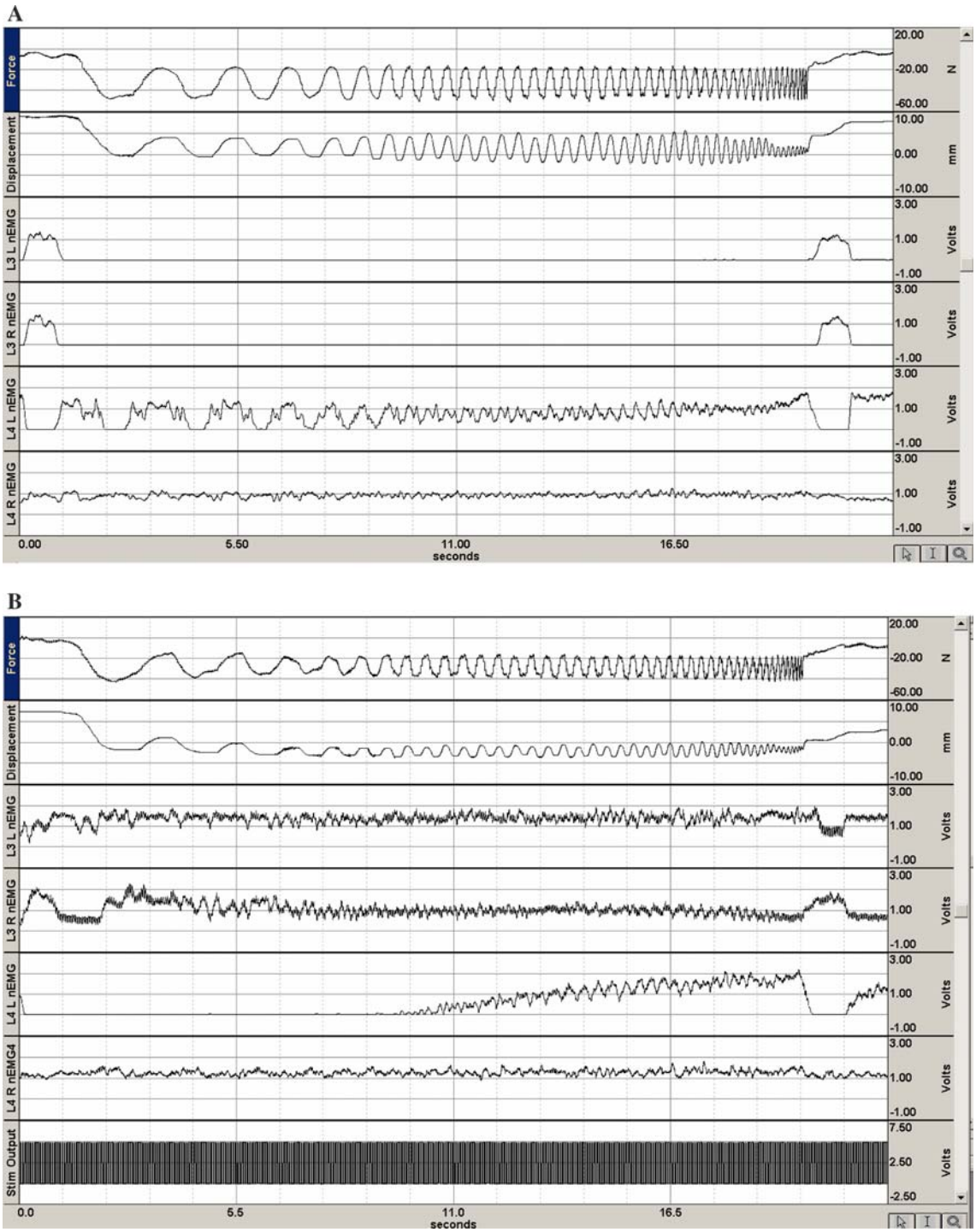

Fig. 2 Typical L3 force deformation and L3, L4 iEMG responses (rectified and integrated) during resting (**a**) and supramaximal stimulation at 20 Hz (**b**). A 5 V, uniform pulse 20 Hz electrical stimulus (*bottom graph* in **b**) was applied bilateral at the L3–L4 interspinous space. Periodic, lower frequency undulations every 3–4 s in the displacement responses corresponds to animal respiration. Experimental results for animal 8. Asymmetries are present in the iEMG responses due to localization of muscle activity picked up by the bipolar needle EMG electrodes

Discussion

The animal and human spine is modulated by intrinsic viscoelasticity of component tissues (ligaments, cartilage, bone, tendons, and muscle) and load sharing provided by adjacent structures (e.g., rib cage, sternum, and pelvis). When such factors are combined with other features such as spinal curvature, the net effect is

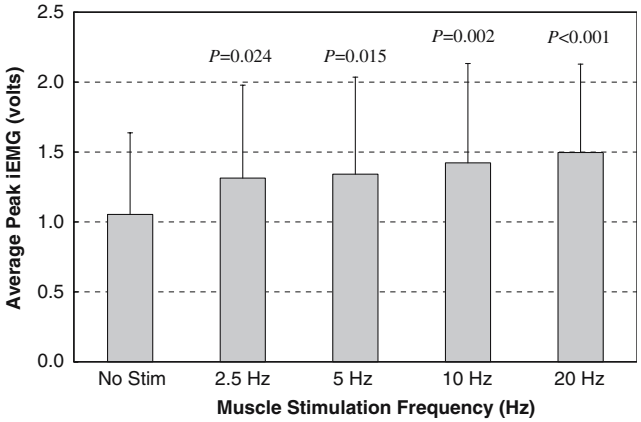

Fig. 3 Comparison of iEMG muscle activity (average peak responses of L3, L4 electrodes). *Error bars* are standard deviation. *P* values (ANOVA) indicate statistical comparison with respect to mechanical excitation without muscle stimulation

a complex structure-frequency-dependent mechanical behavior. In a previous study [10], employing a harmonic stiffness analysis, we reported that the DV stiffness behavior of resting ovine spine was modulated by mechanical excitation frequency. The current study builds upon this work and indicates that the oscillatory mechanical response of the ovine spine is also modulated by muscle stimulation.

Sustained bilateral, supramaximal muscle stimulation (10 and 20 Hz protocols) was found to significantly increase the stiffness of the ovine spine. At these stimulation frequencies, the multifidus muscles experienced a concentric tetanic contraction or maximum muscle response, which stabilizes the spine and facet joints, increasing spine stiffness. An important finding in the current study, therefore, is the dependence of dynamic load–displacement response on physiological processes such as muscle contraction. The musculature is a major contributor to spine stability [23–25], acting through voluntary and involuntary central nervous system control mechanisms. Quantifying frequency-dependent changes in spinal stiffness during varying degrees of muscle activation is important in understanding neuromuscular contributions to dynamic spinal stiffness. In the current study, muscle contraction stimulation intensity was modulated by varying the pulse duration of the supramaximal stimulus voltage. Future studies characterizing dynamic stiffness changes in response to other muscle stimulation modalities, such as submaximal muscle stimulation with varying stimulus voltage, will shed more light on the stabilizing role of the lumbar spine musculature.

During mechanical excitation without muscle stimulation, the prone posture ovine lumbar spine was most

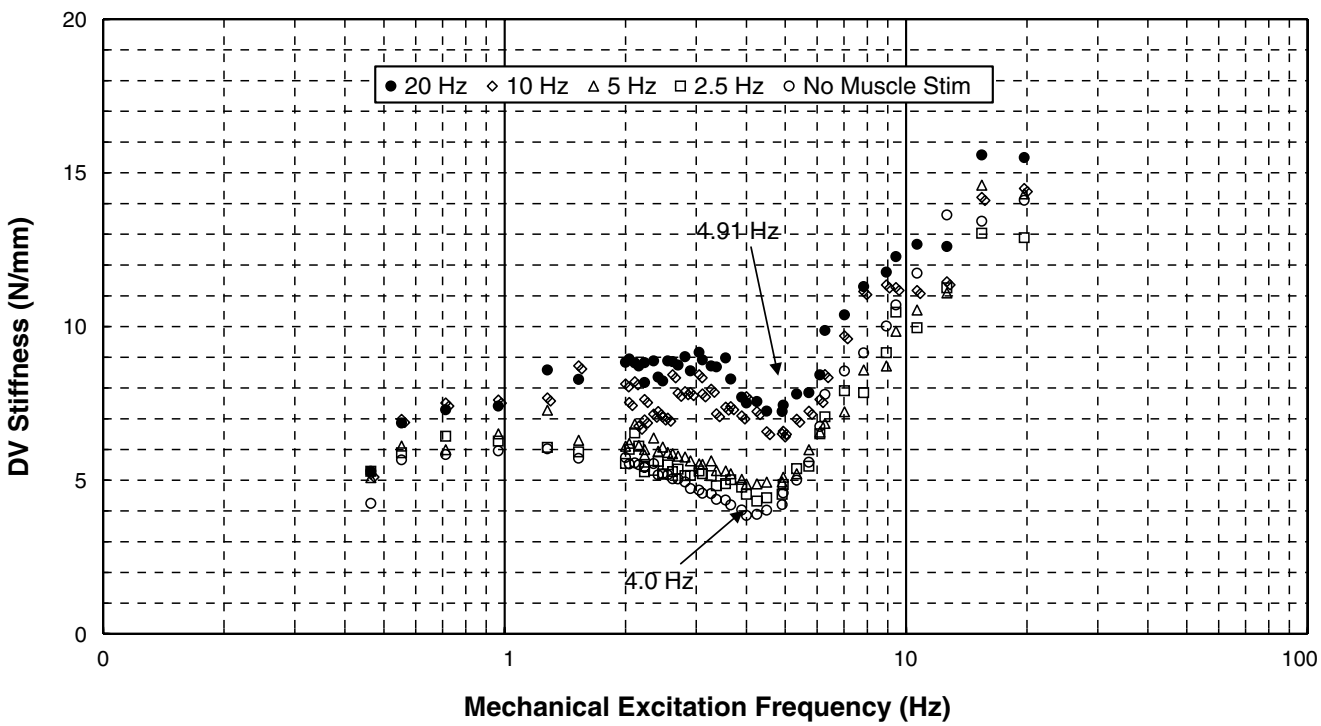

Fig. 4 Semilog plot illustrating the effects of mechanical excitation frequency on the DV stiffness of the ovine lumbar spine at rest (no muscle stim) and during 2.5, 5, 10, and 20 Hz supramaximal muscle stimulations. The natural (resonant) frequency is 4.0 and 4.91 Hz for rest and sustained muscle contraction (10 and 20 Hz muscle stimulation), respectively. *Symbols* are mean values (n = 15 animals)

Table 1 Summary of experimental results

Animal no.	Weight (kg)	Average DV stiffness (N/mm)				
		No stimulation	2.5 Hz stimulation	5 Hz stimulation	10 Hz stimulation	20 Hz stimulation
1	43.0	4.60	5.96	6.53	7.87	9.60
2	61.0	5.35	5.55	5.38	6.09	7.14
3	51.0	5.05	6.02	7.14	10.23	12.44
4	48.0	3.97	4.57	4.89	6.89	7.99
5	44.5	4.26	4.46	4.77	5.95	5.63
6	50.5	4.25	3.97	3.83	4.57	6.42
7	45.0	4.35	5.62	7.19	9.31	9.40
8	46.0	4.36	4.41	5.49	7.50	7.51
9	45.0	5.21	6.65	6.44	22.14	13.50
10	47.5	4.12	3.91	3.93	4.42	6.09
11	47.5	4.10	4.44	5.03	5.90	7.10
12	48.0	4.16	4.64	5.50	6.37	7.15
13	43.0	4.37	4.35	4.70	5.61	8.44
14	42.0	4.07	4.82	4.77	5.47	10.76
15	53.5	4.98	5.06	5.13	5.40	6.50
Mean	47.7	4.48	4.96	5.38	7.58	8.38
SD	4.9	0.45	0.81	1.04	4.34	2.34
Min	42.0	3.97	3.91	3.83	4.42	5.63
Max	61.0	5.35	6.65	7.19	22.14	13.50

Animal characteristics (weight), and average DV stiffness (N/mm, 2.55–4.91 Hz mechanical excitation) for the resting (no muscle stimulation) and four muscle stimulation trials (supramaximal stimulation at 2.5, 5, 10, and 20 Hz). Summary descriptive statistics (mean, SD, min, and max) are shown in the last four rows

mobile (least stiff) at 4.0 Hz. As there was a substantial force–displacement phase angle change at this frequency, 4 Hz most likely represents the natural frequency of the resting ovine lumbar spine subjected to DV forces. An increase in the natural frequency of oscillation is consistent with the increase in structural stiffness [11]. Using a simple mass-spring model, wherein the natural frequency is defined as $(k/m)^{1/2}$, k is the stiffness and m the mass, the predicted frequency shift associated with the 55% average increase in stiffness observed during 20 Hz muscle stimulation is 4.98 Hz. The latter is consistent with the natural frequency (4.91 Hz) obtained for the 20 Hz muscle stimulation protocol. This information is important for spine biomechanics researchers.

The stiffness response of the human and animal spine is dependent on other factors, including loading mode (DV in this study), loading magnitude and support or boundary conditions (e.g., pelvis and trunk stiffness). With the exception of our earlier work upon which this study was conceived [10], there are no previous in vivo studies examining the harmonic DV (or PA) stiffness response of the animal (or human) lumbar spine. However, Kaigle and associates studied the in vivo harmonic frequency response of the resting, prone-lying porcine lumbar spine subjected to axial (craniocaudal) forces [9]. Using a servo-controlled pneumatic actuator to apply axial harmonic forces (0.05–25 Hz) to a single lumbar motion segment (L2–L3 via intrapedicular screws), they reported that the axial stiffness varied approximately 1.5-fold and was a minimum (~150 N/mm) at 25 Hz. The frequency-dependent variation in axial stiffness is similar to that obtained in the current study, but both the natural (resonant) frequency and axial stiffness magnitude are substantially greater. Application of DV loads (current study) does not produce absolute vertical displacement of the mobilized segment. Rather DV (posteroanterior in humans) loading induces an extension moment and shear on the lumbar spine, analogous to three-point

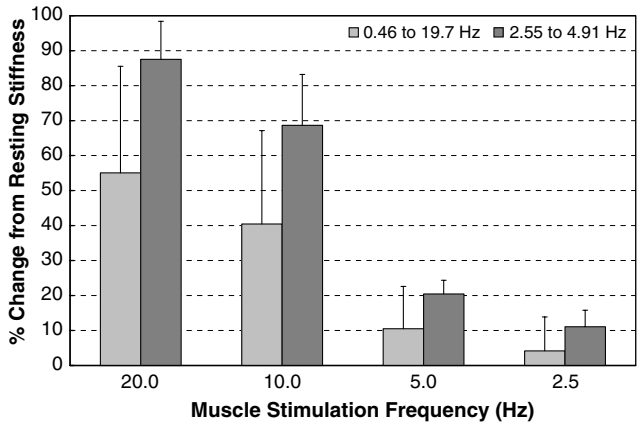

Fig. 5 Summary of the average DV stiffness increase (percent change from resting) following supramaximal muscle stimulation. *Light gray bars* represent average percent change for entire mechanical excitation frequency sweep (0.46–19.7 Hz). *Dark gray bars* show average percent changes for 2.55–4.91 Hz mechanical excitation frequency range. *Error bars* are standard deviation

Table 2 Statistical summary of the ovine DV lumbar spine dynamic stiffness (mean N/mm) obtained during supramaximal trunk muscle stimulation

Mechanical excitation frequency (Hz)	No stimulation	2.5 Hz stimulation	5 Hz stimulation	10 Hz stimulation	20 Hz stimulation
2.55	5.17	5.50	5.90*	7.02*	8.88*
2.62	5.05	5.28	5.87*	8.43*	8.86*
2.70	5.04	5.36	5.78*	7.83*	8.74*
2.81	4.94	5.15	5.75*	7.88*	9.01*
2.90	4.73	5.15*	5.63*	7.85*	8.56*
3.05	4.68	5.30*	5.54*	8.43*	9.16*
3.11	4.58	5.21*	5.51*	7.81*	8.92*
3.27	4.56	5.15	5.63*	7.96*	8.72*
3.37	4.37	4.82	5.31*	7.16*	8.69*
3.55	4.35	4.89*	5.30*	7.38*	8.98*
3.65	4.20	5.02*	5.21*	7.39*	8.29*
3.89	4.03	4.78*	5.03*	7.09*	7.71*
4.00	3.86	4.54*	4.88*	7.71*	7.51*
4.25	3.89	4.32*	4.88*	7.23*	7.56*
4.50	4.03	4.43	4.94*	6.58*	7.24*
4.91	4.20	4.54	4.94*	6.51*	7.24*
Mean	4.48	4.96	5.38	7.52	8.38
SD	0.43	0.36	0.37	0.57	0.68

*Significant change from no stimulation ($P < 0.05$, paired-observation's t test)

bending of beam structures [21]. Differences in spine stiffness between the current and aforementioned study, therefore, reflect the fact that there is significantly less resistance to deflection during bending in comparison to axial loading of beam-like structures. Consequently, decreased stiffness and concomitant lower natural frequency are expected during in vivo DV loading conditions in comparison to axial loading. Moreover, unlike axial loading, DV loading enables one to assess the stiffness of the spine using a non-invasive testing technique.

Non-linearities in the load-deformation characteristics of the spine result in variations in the measured displacement and stiffness that are dependent on the magnitude of the applied force. In their in vivo porcine axial loading study, Kaigle and associates noted that increasing force magnitude (100-N peak to 200-N peak) increased the mean axial stiffness by 28%. Previous human PA mobilization studies using servo-controlled indenter apparatuses have also reported a similar increase in oscillatory (0.5–2 Hz) PA stiffness when the peak applied force is increased [16, 18]. We did not examine the influence of oscillatory load magnitude on DV stiffness in the harmonic, muscle stimulation analysis performed in the current study. However, single cycle, variable amplitude, uniform pulse duration (100 ms) mechanical excitation tests indicate that the DV load–displacement response of the ovine spine is linear between 20 and 80 N [5]. In addition, the loading magnitudes used in the current study were low enough to avoid substantial influence from the facet joints. Nevertheless, when comparing results from in vivo mechanical testing studies in animals (and humans), caution should be taken to observe the load magnitudes employed.

Another important consideration for in vivo spine stiffness assessments is respiration, which has been shown to modulate the mechanical response of the animal spine during static load tests in animals [12]. In a recent study [27], respiratory efforts were found to modulate the oscillatory (1 Hz) PA lumbar spine stiffness of human subjects. These authors reported that the greatest changes in stiffness (67% increase) were seen for the measurements obtained during maximum expiration in comparison to the measurements obtained with the lung volume held at functional residual capacity. In the current study, respiration effects were evident in the displacement response to controlled DV forces (refer to Fig. 2). However, our previous fixed-frequency stiffness analysis [10] showed that ventilator-controlled respiration has only a minor effect (< 7.3%) on dynamic stiffness measurements. This variation is very small when compared to the variation in stiffness observed over the 0.46–19.7 Hz frequency range examined (e.g., 3.7-fold during the rest protocol), and indicates that respirator artifacts are minimal in ventilator-controlled experiments such as that performed in this study.

Clinically, spinal stiffness is determined according to the magnitude of movement detected (perceived) by clinicians. This simple approach is unsatisfactory because it neither takes into account the loads exerted on the spine, nor is the magnitude of the ensuing spine motion quantified. The harmonic testing approach used in the current study in contrast provides a very comprehensive stiffness assessment of a very complex

structure. One limitation is the fact that the swept-sine testing approach requires a lengthy data acquisition. The ability to non-invasively study and identify frequency-specific changes in spine stiffness, however, is appealing from the standpoint of better understanding of physiological processes (e.g., degree of muscle activity) and spinal pathology.

In clinical practice, spinal stiffness measurements may also be affected by factors such as the stiffness of table support padding and skin tissue rather than the intrinsic stiffness of the spine [14]. With regards to the manner in which the ovine spine was constrained during testing, the support conditions that we used in the current study were designed not only to orient and stabilize the ovine trunk during testing, but a rigid testing frame and stainless steel ensured that the measurements were isolated to the spine and trunk. Stabilization of the ovine spine in the transverse plane helped to minimize any artifacts created by off-axis motion of the ovine trunk, and we standardized the manner in which the ovine trunk was stabilized for all animals tested. As this was an animal model, we chose to avoid problems associated with skin deformation by mechanically coupling the load indenter directly to the spinous process. However, the presence of skin tissue would not be expected to have an appreciable effect on the stiffness measurements, since application of a preload causes the more compliant skin to compress. Nevertheless, the effects of skin tissue compliance on dynamic stiffness measurements deserve further attention.

Conclusions

Muscle stimulation increases DV stiffness and alters the frequency response of the ovine spine. Increased spine stiffness may function to enhance spinal stability and allow for more smooth controlled movement patterns. Knowledge of the muscular contributions to dynamic spinal stiffness assessments is also important for clinicians assessing patients with spinal disorders including low back pain. Harmonic stiffness assessment using an indenter over the spinous processes of prone-lying human subjects may assist clinicians in diagnosis and treatment of spinal disorders.

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References

1. Chansirinukor W, Lee M, Latimer J (2001) Contribution of pelvic rotation to lumbar posteroanterior movement. Man Ther 6:242–249
2. Cholewicki J, Panjabi MM, Khachatryan A (1997) Stabilizing function of trunk flexor–extensor muscles around a neutral spine posture. Spine 22:2207–2212
3. Colloca CJ, Keller TS (2004) Active trunk extensor contributions to dynamic posteroanterior lumbar spinal stiffness. J Manipulative Physiol Ther 27:229–237
4. Colloca CJ, Keller TS (2001) Stiffness and neuromuscular reflex response of the human spine to posteroanterior manipulative thrusts in patients with low back pain. J Manipulative Physiol Ther 24:489–500
5. Colloca CJ, Keller TS, Harrison DE, Moore RJ, Gunzburg R, Harrison DD (2006) Spinal manipulation force and duration affect vertebral movement and neuromuscular responses. Clin Biomech 21:254–262
6. Gedalia U, Solomonow M, Zhou BH, Baratta RV, Lu Y, Harris M (1999) Biomechanics of increased exposure to lumbar injury caused by cyclic loading. Part 2. Recovery of reflexive muscular stability with rest. Spine 24:2461–2467
7. Goel VK, Kong W, Han JS, Weinstein JN, Gilbertson LG (1993) A combined finite element and optimization investigation of lumbar spine mechanics with and without muscles. Spine 18:1531–1541
8. Granata KP, Marras WS (1995) The influence of trunk muscle coactivity on dynamic spinal loads. Spine 20:913–919
9. Kaigle A, Ekstrom L, Holm S, Rostedt M, Hansson T (1998) In vivo dynamic stiffness of the porcine lumbar spine exposed to cyclic loading: influence of load and degeneration. J Spinal Disord 11:65–70
10. Keller TS, Colloca CJ (2006) Dynamic dorsoventral stiffness assessment of the ovine lumbar spine. J Biomech (in press)
11. Keller TS, Colloca CJ, Beliveau JG (2002) Force-deformation response of the lumbar spine: a sagittal plane model of posteroanterior manipulation and mobilization. Clin Biomech 17:185–196
12. Keller TS, Holm SH, Hansson TH, Spengler DM (1990) 1990 Volvo Award in experimental studies. The dependence of intervertebral disc mechanical properties on physiologic conditions. Spine 15:751–761
13. Kong WZ, Goel VK, Gilbertson LG, Weinstein JN (1996) Effects of muscle dysfunction on lumbar spine mechanics. A finite element study based on a two motion segments model. Spine 21:2197–2206
14. Latimer J, Holland M, Lee M, Adams R (1997) Plinth padding and measures of posteroanterior lumbar stiffness. J Manipulative Physiol Ther 20:315–319
15. Latimer J, Lee M, Adams R, Moran CM (1996) An investigation of the relationship between low back pain and lumbar posteroanterior stiffness. J Manipulative Physiol Ther 19:587–591
16. Latimer J, Lee M, Adams RD (1998) The effects of high and low loading forces on measured values of lumbar stiffness. J Manipulative Physiol Ther 21:157–163
17. Lee M, Esler M-A, Mildren J (1993) Effect of extensor muscle activation on the response to lumbar posteroanterior forces. Clin Biomech 8:115–119
18. Lee M, Latimer J, Maher C (1997) Normal response to large posteroanterior lumbar loads—a case study approach. J Manipulative Physiol Ther 20:369–371
19. Lee M, Liversidge K (1994) Posteroanterior stiffness at three locations in the lumbar spine. J Manipulative Physiol Ther 17:511–516

20. Lee M, Svensson NL (1993) Effect of loading frequency on response of the spine to lumbar posteroanterior forces. J Manipulative Physiol Ther 16:439–446
21. Lee RY, Evans JH (2000) The role of spinal tissues in resisting posteroanterior forces applied to the lumbar spine. J Manipulative Physiol Ther 23:551–556
22. Lee RY, Tsung BY, Tong P, Evans J (2005) Bending stiffness of the lumbar spine subjected to posteroanterior manipulative force. J Rehabil Res Dev 42:167–174
23. Marras WS, Granata KP (1997) The development of an EMG-assisted model to assess spine loading during whole-body free-dynamic lifting. J Electromyogr Kinesiol 7:259–268
24. McGill SM, Norman RW (1986) Partitioning of the L4–L5 dynamic moment into disc, ligamentous, and muscular components during lifting. Spine 11:666–678
25. Panjabi MM (1992) The stabilizing system of the spine. Part I. Function, dysfunction, adaptation, and enhancement. J Spinal Disord 5:383–389
26. Quint U, Wilke HJ, Shirazi-Adl A, Parnianpour M, Loer F, Claes LE (1998) Importance of the intersegmental trunk muscles for the stability of the lumbar spine. A biomechanical study in vitro. Spine 23:1937–1945
27. Shirley D, Hodges PW, Eriksson AE, Gandevia SC (2003) Spinal stiffness changes throughout the respiratory cycle. J Appl Physiol 95:1467–1475
28. Shirley D, Lee M, Ellis E (1999) The relationship between submaximal activity of the lumbar extensor muscles and lumbar posteroanterior stiffness. Phys Ther 79:278–285
29. Viby-Mogensen J (2000) Neuromuscular monitoring, pp 1352–1366
30. Viner A, Lee M, Adams R (1997) Posteroanterior stiffness in the lumbosacral spine. The correlation between adjacent vertebral levels. Spine 22:2724–2729